

Chapter 4

Species Accounts

Organization of the Narrative Accounts

Narrative accounts for each species are presented by several authors in a consistent format to convey specific information relative to that mammal. The orders are arranged phylogenetically; families and species are arranged alphabetically to facilitate finding a particular species. Each account is organized as follows:

Content and Taxonomic Comments. This section provides a brief general summary of recent revisionary work, status of current taxonomic knowledge, and reference to authoritative reviews of the literature. To resolve taxonomic controversies, the *Revised Checklist of North American Mammals north of Mexico* (Baker et al. 2003) is relied upon, although in some instances departures from this authority occurred. Recent synonyms of the scientific name are also listed due to the dynamic nature of scientific nomenclature. Currently recognized subspecies are identified. Common names that are used locally or regionally are also included. The name of the original describer is given after the name of the species, thus *Sylvilagus palustris* (Bachman, 1837) for the marsh rabbit.

Distinguishing Characteristics. This segment provides information to assist identification based on size, shape, coloration, pelage, and cranial features. For each species, the following standard measurements are provided: total length (tip of snout to the tip of the last tail vertebra, not to the end of the hairs), tail length (base of tail to the tip of the last vertebra, the location of the base established by bending the tail up at right angles to the body), and hind foot length (ankle bend to the end of the longest claw or hoof), ear length (from the notch at the base of the ear to the tip of the ear), and weight. For bat species, the length of the forearm is also included. If sexual dimorphism occurs, additional detail is provided. Diagnostic characteristics include information on how to distinguish the mammal from related species.

To facilitate identification, an illustration of the skull is depicted along with dental formulae presented in the standard upper/lower incisor, canine, premolar, and molar numbers. The dental formula is characteristic for each species. For example, the formula for the coyote is $I\ 3/3, C\ 1/1, P\ 4/4, M\ 2/3 = 42$. This indicates that on each side of the skull there are 3 incisors, 1 canine, 4 premolars, and 2 molars above and 3 molars below, thus 21 teeth total. That number is then doubled (here 42) to account for the upper and lower jaws on both sides of the skull. As observed here, the numbers for the upper and lower jaws do not always coincide.

Conservation Status. This section provides a review of the current global and state conservation status based on Natural Heritage rankings and federal listing, if applicable. If a recovery plan is approved for the species, it is mentioned. Some states mirror federal efforts with state endangered species protection measures and these are often mentioned. Classification as a furbearer, game, or nuisance species by state agencies also is discussed.

Distribution. This segment provides a current range map indicating the areas of the South from which the species is known. Additional detail is provided in the text accompanied by references to the literature by each state. In some instances, species distributions were modified based on discussions with state natural resource agency, academicians, and other field research personnel. Currently recognized subspecies also are mapped with indication of their geographic distribution.

Abundance Status. This section provides available information on the regional and local abundance of the species. The reader is cautioned that abundance is highly variable over time, environmental conditions, and vegetation community. Some mammals are rare and limited in distribution, and others may

be so poorly known that it is difficult to estimate abundance status.

Primary Habitats. This section reviews the habitat preferences of each mammal, summarized with appropriate references. Differences in habitat use for hunting/foraging, denning, and dispersal are identified. Information on local and general variation in habitat is included, where applicable. Home-range size is also discussed in this section.

Reproduction. This segment provides an overview of peak breeding period, age at sexual maturity, length of gestation, average litter size, weaning and development of young, and age at dispersal. Particular reproductive strategies are also discussed, when warranted.

Food Habits. This section outlines the general diet composition, seasonal shifts in foods, and the references that provide further detailed assessments. Indication is given whether the species is herbivorous, carnivorous, or omnivorous, and whether the foods

consumed are influenced by availability or selectivity by the species. Feeding adaptations are also discussed.

Associated Species. This section summarizes the faunal associates often found with the species. Generally, these groupings are linked to commonalities in vegetative community or physiographic province. Those animals known to prey upon or compete with the mammal similarly are identified.

Vulnerability and Threats. This segment assesses factors that may imperil the species based on the current state of knowledge. Information on disease, parasites, environmental stressors, and other potential sources of mortality are identified.

Management Suggestions. This section offers general guidance for habitat enhancement and conservation. In some instances, management actions for population control through creation of less suitable habitat conditions is indicated for overabundant or nuisance species.

Virginia Opossum

Didelphis virginiana (Kerr, 1792)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

For many years, North American opossums were regarded as members of a single species, *Didelphis marsupialis* (Hershkovitz 1951). Gardner (1973) reviewed the systematics of North American *Didelphis* and defined two North American species. The species occurring in the United States was described as the Virginia opossum (*D. virginiana*), whereas the southern opossum inhabiting Central and South America was classified as *D. marsupialis*. The Virginia opossum is represented by four subspecies; two occur in the South (*D. v. pigra* and *D. v. virginiana*). McManus (1974) and Gardner (1982) review the literature.

DISTINGUISHING CHARACTERISTICS

The Virginia opossum is a robust-bodied, cat-sized mammal with a pointed snout, naked leathery ears, short legs, and a long, scaly prehensile tail. The medial digit on the hind foot is opposable and clawless. Measurements are: total length, 513–900 mm; tail, 220–380 mm; hind foot, 38–85 mm; ear, 40–60 mm; weight, 0.9–5.9 kg. Males are larger than females after breeding. There are two color phases: gray and black. The dorsal pelage is characterized by long dense fur that is white at the base and dark brown to black at the tip and interspersed with long guard hairs that are either white-tipped or black-tipped contributing to a grizzled appearance. The venter is darker. The head is white to pale gray and the ears are black with white tips. The lower legs and feet are black, but the toes are white. *Didelphis virginiana* is the only North American mammal with five pairs of upper incisors and 50 teeth. The dental formula is: I 5/4, C 1/1, P 3/3, M 4/4 = 50 (Figure 1).

CONSERVATION STATUS

The Virginia opossum has a global rank of Secure (NatureServe 2007). The species is also considered Secure in those states where it occurs within the region, except for Arkansas, where it is Apparently Secure. It is unranked in Florida. Game laws protecting the opossum vary by state; there can be open seasons for hunting or trapping.

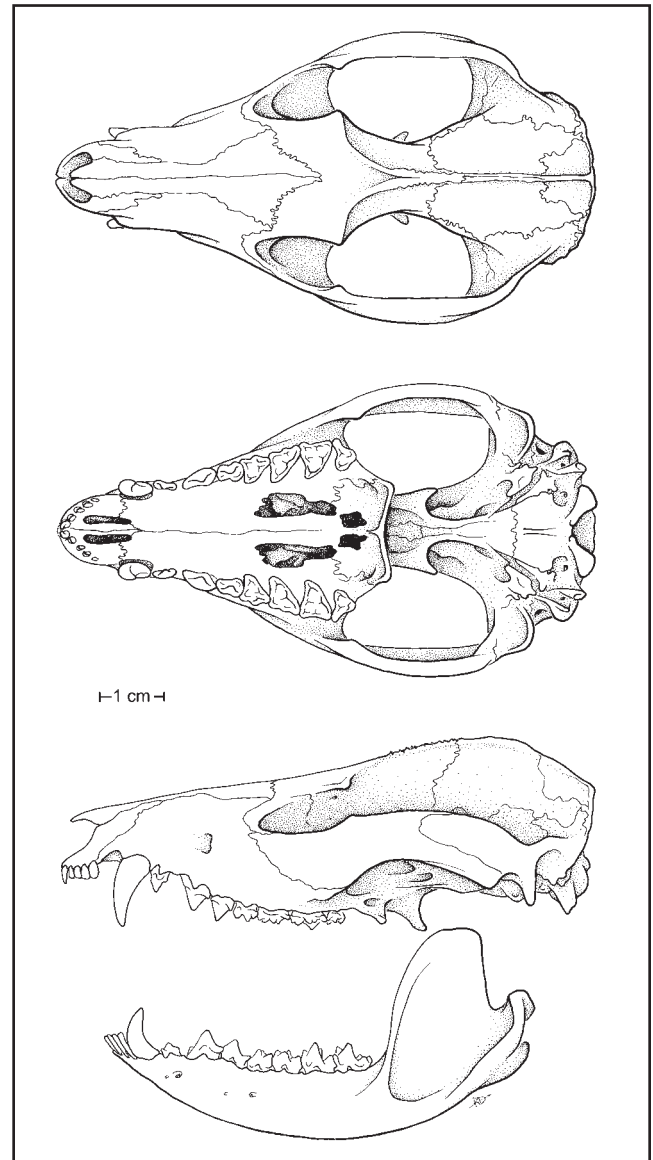


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Didelphis virginiana* from Talbot County, Maryland (USNM 560501, female).

DISTRIBUTION

The species is ubiquitous in the southern United States (Figure 2). It occurs throughout Virginia (Bailey 1946, Stout and Sonenshine 1974, Webster et al. 1985, Pagels et al. 1992, Linzey 1998), North Carolina (Lee et al. 1982, Webster et al. 1985, Handley 1992, Clark et al.

1993), South Carolina (Wood and Odom 1964, Golley 1966, Sanders 1978, Schacher and Pelton 1979, Smith et al. 1984, Webster et al. 1985, Cothran et al. 1991), Georgia (Harper 1927, McKeever 1958, Golley 1962, Neuhauser and Baker 1974, Laerm et al. 1982, Allen et al. 1985, Ruckdeschel and Shoop 1986), including most of the barrier islands of these states. It also inhabits Florida (Hamilton 1941, Rand and Host 1942, Moore 1946, Pournelle 1950, Schwartz 1952, McKeever 1958, Ivey 1959, Layne 1974, Connor et al. 1983, Ryser 1995), Alabama (Howell 1921, Holliman 1963, Linzey 1970, Summer and Hill 1980), Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989), Louisiana (Lowery 1974, Edmunds et al. 1978, Morrison et al. 1981, Nichols and Chabreck 1981, Martin et al. 1991), and most of Texas (Schmidly 1983, Jones and Jones 1992, Davis and Schmidly 1994). The range includes Tennessee (Calhoun 1941, Howell and Conaway 1952, Conaway and Howell 1953, Goodpaster and Hoffmeister 1952, Linzey and Linzey 1971, Smith et al. 1974, Leberg et al. 1983, Weckerly et al. 1987, Kennedy 1991, Kissell and Kennedy 1992), Kentucky (Barbour and Davis 1974, Fassler 1974, Lyons et al. 1995), Arkansas (Sealander and Heidt 1990), and Oklahoma (Caire et al. 1989).

ABUNDANCE STATUS

The Virginia opossum is one of the most common and widely distributed mammals in the region. Density estimates vary considerably depending upon habitat and season. Estimates range from 1/ha to as low as 1/50 ha (Lay 1942, McKeever 1958, Wood and Odom 1964, Stout and Sonenshine 1974, Conner et al. 1983, Leberg et al. 1983, Smith et al. 1984, Weckerly et al. 1987, Kissell and Kennedy 1992). Estimates near 1/25 ha are typical (Kissell and Kennedy 1992). Gardner (1982) provides additional references on opossum densities. Urbanization has altered considerable wildlife habitat throughout the South, resulting in an increase of opossum populations in many urban and suburban areas (Chamberlain and Leopold 2001).

PRIMARY HABITATS

Didelphis virginiana occurs in habitats ranging from relatively dry uplands to mesic bottoms and coastal marshes where sufficient food, water, and cover are available (Gardner 1982). The preferred habitat is deciduous woodland in association with surface water such as streams, ponds, or swamps; however, the species inhabits most forest cover types in the region. It often is found in croplands, residential areas, and ecotones between woodlands and grasslands. The Virginia opossum also is associated with brushy areas along ditches, fencerows, and transportation corridors. It dens in any available cavity or

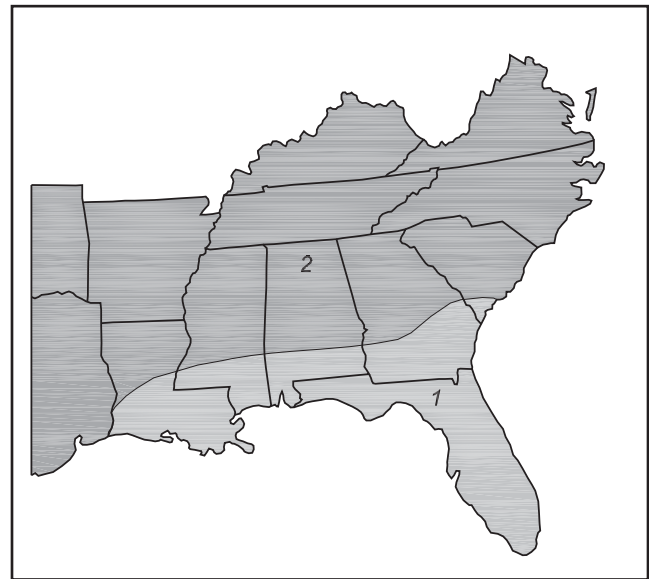


Figure 2. Distribution of *Didelphis virginiana* in the South: (1) *D. v. pigra*; (2) *D. v. virginiana*.

opening, including hollow trees, stumps, and logs, brush piles, refuse and trash heaps, rock outcrops, and buildings. The opossum also frequently uses the nests and burrows of other animals such as squirrels (*Sciurus* spp.), skunks, foxes, woodchucks (*Marmota monax*), nine-banded armadillo (*Dasypus novemcinctus*) and turtles (Gardner 1982).

REPRODUCTION

The reproductive biology of the Virginia opossum is discussed by Lay (1942), Reynolds (1945), Burns and Burns (1957), McKeever (1958), Llewellyn and Dale (1964), Stout and Sonenshine (1974), Edmunds et al. (1978), and Gardner (1982). In the region, breeding extends from January through late fall. Two distinct peaks occur, one in early winter (January–February) and another during May–June. Females are polyestrous with a cycle ranging from 22–38 days; gestation lasts about 12 days (Reynolds 1952). Newborns are altricial and further development occurs in the marsupium. Four to 35 are born initially, but because not all of the 13 teats are functional, the maximum litter size is limited to 12. Young are sexually mature as early as 6–8 months old.

FOOD HABITS

The diet of *Didelphis virginiana* consists of numerous invertebrate and vertebrate prey, including carrion. A wide spectrum of plant matter, including grasses, seeds, fruits, nuts, tubers, and agricultural crops is reported in the diet (Lay 1942, Reynolds 1945, Hamilton 1951, 1958; Llewellyn and Uhler 1952, Wood 1954, Whitaker et al. 1977, and Gardner 1982).

ASSOCIATED SPECIES

The Virginia opossum is so widely distributed within a diversity of habitats, it is associated with numerous mammals in the region.

VULNERABILITY AND THREATS

Within the region, there are no threats to the survival of the species. The mosaic of land use patterns associated with human development, particularly agricultural, has increased habitat diversity and edge effect to the benefit of the species.

MANAGEMENT SUGGESTIONS

Management is rarely directed at opossum populations (Chamberlain and Leopold 2001). Trapping is used to regulate population levels, if warranted. The species is not highly valued for its pelt; fur prices and market fluctuations are unlikely to affect population viability. Management usually is in the form of damage control or removal, particularly in urban settings. The removal of potential den sites is also effective when predation on ground-nesting game birds and waterfowl is problematic.

Habitat management centers on enhancement of soft mast production and understory diversity, and provision of fencerows as travel corridors (Allen 1987). Landscapes with an interspersed forest seral stages, openings, and fields near permanent water sources should be protected (Chamberlain and Leopold 2001).

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Northern Short-tailed Shrew

Blarina brevicauda (Say, 1823)

Joshua Laerm, W. Mark Ford and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Until recently, the short-tailed shrew (*Blarina brevicauda*) was the only recognized species in the genus *Blarina*. Revision of the *Blarina brevicauda* complex resulted in recognition of three species: the northern short-tailed shrew (*B. brevicauda*), the southern short-tailed shrew (*B. carolinensis*), and Elliot's short-tailed shrew (*B. hylophaga*; Genoways and Choate 1972, Tate et al. 1980, French 1981, George et al. 1981, George et al. 1982, Moncrief et al. 1982, Braun and Kennedy 1983, Jones et al. 1984, George et al. 1986). Twelve northern short-tailed shrew subspecies currently are recognized. Six of the subspecies, *B. b. churchi*, *B. b. kirtlandi*, *B. b. knoxjonesi*, *B. b. talpoides*, and *B. b. telmalestes* occur in the South (George et al. 1986, Webster 1996). Populations of *Blarina* in Florida, referred by some as *B. b. shermani*, are now regarded as subspecies of *B. carolinensis*. The literature on the northern short-tailed shrew was reviewed by George et al. (1986).

DISTINGUISHING CHARACTERISTICS

The northern short-tailed shrew is a large, short-tailed soricid and is the largest of the three *Blarina* species. Specimens vary in size throughout the range with larger forms occurring in the Appalachians and smaller forms occurring in the Piedmont and Interior Low Plateau. Measurements are: total length, 95–135 mm; tail, 13–32 mm; hind foot, 8–18 mm; weight, 20–30 g. The northern short-tailed shrew has small ears that are concealed in the pelage, minute eyes, and a long and pointed snout. The pelage color is variable, ranging from grayish brown to slate black dorsally and only slightly paler below. Specimens of *B. brevicauda* may be confused with the southern short-tailed shrew or Elliott's short-tailed shrew. Whereas distinction often can be made on the basis of distribution, in areas of near parapatry morphometric or genetic comparisons must be made (Tate et al. 1980, French 1981, George et al. 1981, George et al. 1982, Braun and Kennedy 1983). The dental formula of the northern short-tailed shrew is: I 3/1, C 1/1, P 3/1, M 3/3 = 32 (Figure 1). See keys for additional details.

CONSERVATION STATUS

The northern short-tailed shrew has a global rank of Secure (NatureServe 2007). The species is also

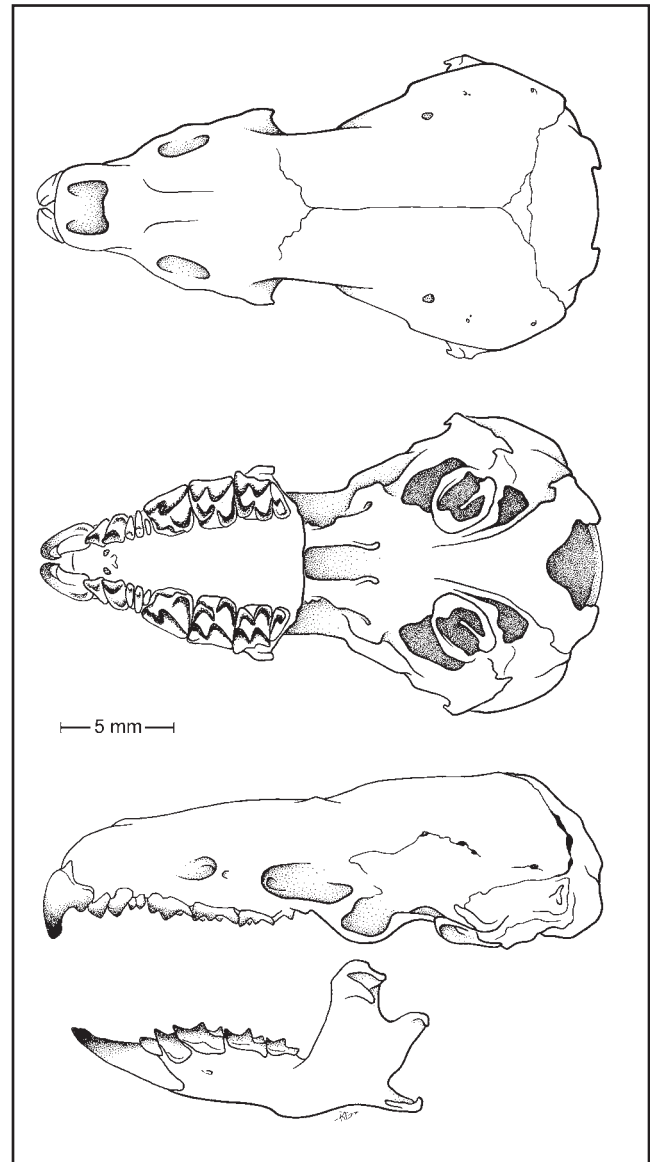


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Blarina brevicauda* from Macon County, North Carolina (USNM 291774, female).

considered Secure in those states where it occurs within the South. It is unranked in South Carolina.

DISTRIBUTION

The northern short-tailed shrew is distributed throughout south-central and southeast Canada

through northeastern and north-central United States and south through the Appalachians and Interior Low Plateau (Figure 2). Regionally, it exhibits a parapatric to limited sympatric distribution in the southern periphery with that of the southern short-tailed shrew, but precise distributional limits are uncertain (Tate et al. 1980, French 1981, Braun and Kennedy 1983, Webster et al. 1984, George et al. 1986, Pagels and French 1987). Except for the south-central and eastern parts of the state, it occurs throughout Virginia (Pagels and Tate 1976, Jackson et al. 1976, Dueser et al. 1979, Tate et al. 1980, Pagels and French 1987, Rose et al. 1990, Handley 1992, Pagels et al. 1992, Kalko and Handley 1993, Mitchell et al. 1993, 1997; Pagels and Baker 1997, Linzey 1998, Bellows et al. 2001). In North Carolina, populations are restricted to the upper Piedmont and Blue Ridge (Odom 1949, Johnston 1967, Gentry et al. 1968, Whitaker et al. 1975, Lee et al. 1982, Linzey 1995, Ford et al. 2006) and the lower Coastal Plain including the Outer Banks (Handley 1979, Webster et al. 1984, Adams et al. 1987, Webster 1988, Clark et al. 1993, Webster 1996). It is restricted to the upper Piedmont and Blue Ridge of South Carolina (Mengak et al. 1987, Laerm et al. 1999, Ford et al. 2006) and Georgia (Langley and Shure 1980, Ford et al. 1994, 1997; Parmley et al. 1997, Laerm et al. 1999, 2006) and the Piedmont of east-central Alabama (French 1981). The species occurs east of the Upper Coastal Plain in Kentucky and Tennessee (Barbour 1951, Howell and Conaway 1952, Conaway and Howell 1953, Fassler 1974, Dueser and Shugart 1978, Braun and Kennedy 1983, Seagle 1985a,b; Bryan 1991, Harvey et al. 1991, Copeland and Caldwell 1991, Harvey et al. 1992, McGehee-Marsh et al. 1992, Feldhamer et al. 1993, Kiser and Meade 1993, Linzey 1995). *Blarina b. telmalestes*, once believed confined to the Dismal Swamp, is now known to have a broader distribution in Virginia and North Carolina (Clark et al. 1985, Webster 1996) although Handley (1979) noted that this subspecies is poorly defined and individuals often are indistinguishable from those of surrounding populations of *B. b. kirtlandi*. The range of *B. b. churchi* also is poorly defined; the subspecies probably occurs only in the Blue Ridge (Bole and Moulthrop 1942). *Blarina b. knoxjonesi* is restricted to lowland riparian zones and pocosin-bay forests in east-central and southeastern North Carolina between the Pamlico and Cape Fear rivers (Webster 1996).

ABUNDANCE STATUS

The northern short-tailed shrew is common throughout much of the southern Appalachians and Interior Low Plateau, but less so in the Piedmont and Coastal Plain portions of its range. Population densities fluctuate monthly and yearly (Yahner 1982, 1983; Getz

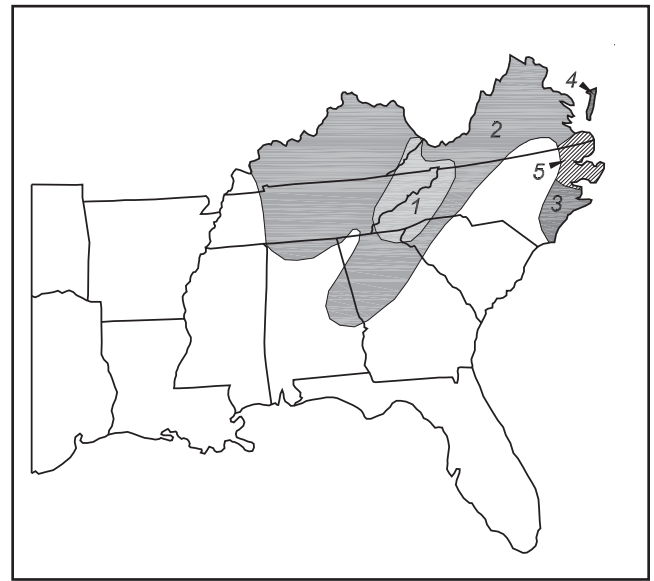


Figure 2. Distribution of *Blarina brevicauda* in the South: (1) *B. b. churchi*; (2) *B. b. kirtlandi*; (3) *B. b. knoxjonesi*; (4) *B. b. telmalestes*; (5) *B. b. talpoides*.

1989) and may be related to winter cold stress (Gottschang 1965, George et al. 1986) and precipitation (Getz 1989). Lima et al. (2002) suggest that population densities fluctuate due to intraspecific competition dynamics. Reported densities range from approximately 2–120/ha (George et al. 1986, Merritt 1986, Getz 1989) and may reach as high as 260/ha (Townsend 1935).

PRIMARY HABITATS

The northern short-tailed shrew is a habitat generalist, reported from virtually all cover types in the region (George et al. 1986, Bellows et al. 2001). In the Midwest and Northeast it is common in grasslands, various successional stages of oldfields, farmstead shelterbelts, all seral stages of coniferous and deciduous hardwood forests, and saturated wetlands (Getz 1961, Yahner 1982, 1983; George et al. 1986, Getz 1989). In the Ridge and Valley and Interior Low Plateaus, Dueser and Shugart (1978) and Seagle (1985b) indicated that the species exhibits narrow habitat specialization. In the Blue Ridge, Laerm et al. (1999) and Ford et al. (2006) suggest that the northern short-tailed shrew has broad habitat tolerances but tends to be associated with dense herbaceous cover and coarse woody debris in older, mixed mesophytic forests such as cove hardwoods. Similarly, Bellows et al. (2001) found this species closely associated with large diameter downed woody debris in the Coastal Plain. Because this species is semi-fossorial in habit, the northern short-tailed shrew often inhabits areas with a thick ground litter and a deep, well-developed soil profile (Pruitt 1953, 1959). Superficial and deep

burrow systems are constructed beneath litter, logs, and rocks. The burrows of other small mammals, such as the eastern mole (*Scalopus aquaticus*), probably are used as travel corridors and den sites as observed with southern short-tailed shrews (Hartman et al. 2001). Additionally, the northern short-tailed shrew, similar to many other soricids, can echolocate to aid in orientation and exploration (van Zyll de Jong 1983).

REPRODUCTION

The breeding season extends from February through November (Pearson 1944, Christian 1969) with peaks in spring and late summer-early fall. Gestation is 21–22 days. Litter size is 2–8 with an average of 4–5 young (Hamilton 1929, Blus 1971, Mumford and Whitaker 1982). Both sexes are capable of reproducing at approximately 60 days of age; young born in spring may reproduce in the first year (Dapson 1968, Blus 1971). Longevity in the wild is about 18 months; in captivity, 30–33 months (Pearson 1945).

FOOD HABITS

Annelids, gastropods, lepidopteran larvae, chilopods, arachnids, other invertebrates, and small quantities of plant material comprise most of the diet (Hamilton 1941, Whitaker and Ferraro 1963, Whitaker and Mumford 1972, Linzey and Linzey 1973, George et al. 1986). Northern short-tailed shrews also prey on small rodents, amphibians, and reptiles (Allen 1938, Hamilton 1941, Eadie 1944, 1952; Fulk 1972, George et al. 1986) in part facilitated by modified salivary glands that provide the northern short-tailed shrew with a venomous bite. The venom quickly immobilizes prey and enables the shrew to cache prey in a comatose state for a time, thereby ensuring an available fresh source of food (Merritt 1987).

ASSOCIATED SPECIES

Because it is a habitat generalist and has a ubiquitous distribution, the northern short-tailed shrew may be found in association with nearly all of the mammals in the region. The range of *Blarina brevicauda* exhibits zones of sympatry with the southern short-tailed shrew to the south and east (Jones et al. 1984, Choate et al. 1994), but they are seldom syntopic (Webster 1996, McCay et al. 2004).

VULNERABILITY AND THREATS

This species is considered numerous and stable within its range; there are no apparent population-level threats to its survival.

MANAGEMENT SUGGESTIONS

The northern short-tailed shrew appears tolerant of limited habitat disturbance from forest management activities such as timber harvest and prescribed burning (Ford et al. 1997, 1999, 2000; Ford and Rodrigue 2001). Activities that cause severe soil compaction should be avoided (Mitchell et al. 1997).

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Southern Short-tailed Shrew

Blarina carolinensis (Bachman, 1837)

Joshua Laerm, W. Mark Ford, Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Revision of the northern short-tailed shrew (*Blarina brevicauda*) complex resulted in the recognition of the southern short-tailed shrew (*B. carolinensis*) as a distinct species on the basis of morphological and karyological differences (Genoways and Choate 1972, Tate et al. 1980, French 1981, George et al. 1982, Braun and Kennedy 1983). Four subspecies, *B. c. carolinensis*, *B. c. minima*, *B. c. peninsulae*, and *B. c. shermani*, currently are recognized, but the taxonomic status is problematic. Hutterer (1993) incorrectly lists *B. c. minima* as a subspecies of Elliot's short-tailed shrew (*B. hylophaga*; see Easterla 1968, Lowery 1974, Schmidly 1983, Sealander and Heidt 1990). The taxonomy of the populations in lower peninsular Florida that are referred to *B. c. peninsulae* is uncertain (Layne 1992). George et al. (1982) recognized two distinct chromosomal groups of short-tailed shrews in Florida: *B. c. carolinensis* to the north and *B. c. peninsulae* in the south. The karyotype of *B. c. peninsulae* is sufficiently different from the northern short-tailed shrew (*B. brevicauda*) and southern short-tailed shrew to suggest that *B. c. peninsulae* may be a distinct species (Jones et al. 1984, George et al. 1986, Layne 1992). A population of short-tailed shrews known only from their type locality at Fort Myers, Florida, was described by Hamilton (1955) as *B. b. shermani*. Although this taxon was referred to *B. carolinensis* by George et al. (1982) and Layne (1992), the United States Fish and Wildlife Service (1991) referred them to *B. brevicauda*. Confusion regarding the status of *B. c. peninsulae* adds to the uncertain relationship of *B. c. shermani*, as *B. c. shermani* may be a subspecies of either *B. brevicauda* or *B. carolinensis* and it might be synonymous with *B. c. peninsulae* (Layne 1992). We follow Layne (1992) in the use of *B. c. shermani*. Furthermore, there is no consensus about the subspecific affinities of *B. carolinensis* in southern and eastern Arkansas, eastern Texas, and southeastern Oklahoma. Traditional maps (Hall 1981) suggest that two subspecies, *B. c. carolinensis* and *B. c. minima*, occur in that area. The status of the populations referable to *B. c. carolinensis* in southeastern Arkansas, southeastern Oklahoma, and eastern Texas is uncertain. They may represent isolates of *B. c. carolinensis*, intergrades between Elliot's short-tailed shrew (*B. hylophaga*) and *B. carolinensis*, or populations that should be referred to *B. c. minima*. The literature on the southern short-tailed shrew was reviewed by McCay (2001).

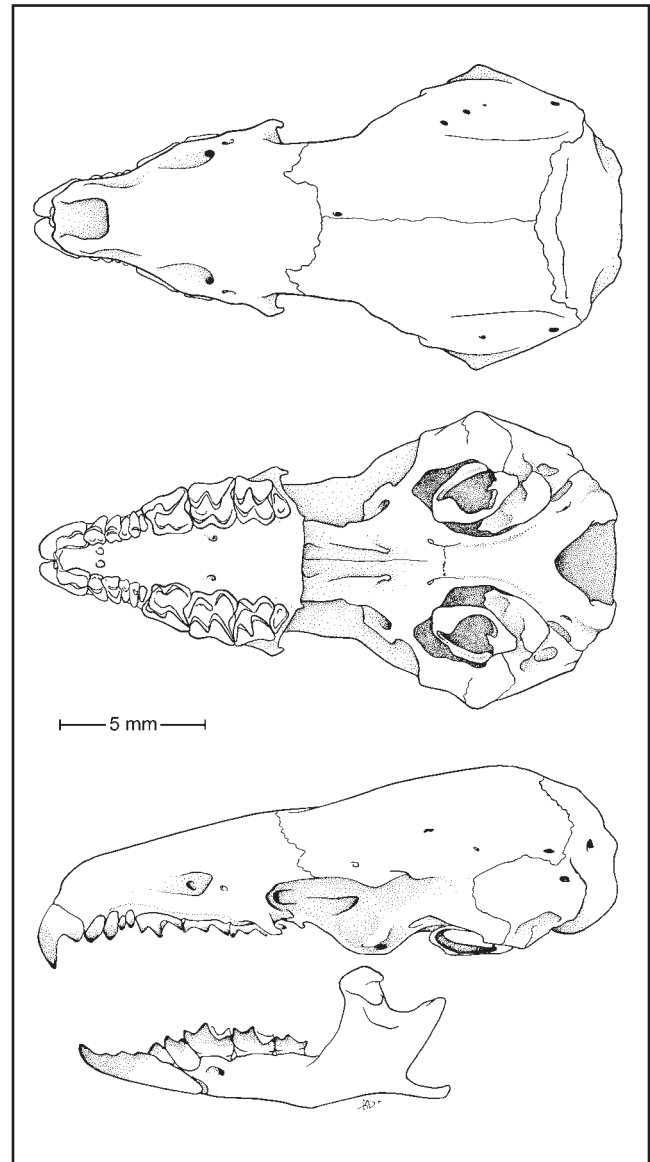


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Blarina carolinensis* from Charleston County, North Carolina (USNM 574241, male).

DISTINGUISHING CHARACTERISTICS

The southern short-tailed shrew is a large, short-tailed soricid and is the smallest of the three *Blarina* species in the region. Measurements are: total length, 79–123 mm; tail, 14–27 mm; hind foot, 10–17 mm; weight 8–15 g. The southern short-tailed shrew has small ears that

are concealed in the pelage, tiny eyes, and a long, pointed snout. The pelage color is variable, ranging from grayish brown to slate black dorsally and only slightly paler below. The species may be confused with the northern short-tailed shrew and Elliot's short-tailed shrew, and although distinction often can be made on the basis of distribution, in areas of parapatry or sympatry, morphometric or genetic comparisons must be made (Tate et al. 1980, French 1981, George et al. 1981, 1982; Braun and Kennedy 1983, Stangl and Carr 1997). The dental formula of the southern short-tailed shrew is: I 3/1, C 1/1, P 3/1, M 3/3 = 32 (Figure 1). See keys for additional details.

CONSERVATION STATUS

The southern short-tailed shrew has a global rank of Secure (NatureServe 2007). It is Secure in Alabama, Georgia, Kentucky, Mississippi, North Carolina, and Virginia. It is Apparently Secure in Arkansas, Louisiana, and Texas, but is Vulnerable in Oklahoma. It is unranked in Florida and South Carolina.

DISTRIBUTION

Figure 2 depicts the distribution of the short-tailed shrew, which occurs throughout much of the southeastern and south-central United States (Whitaker and Hamilton 1998). It ranges from south-central and eastern portions of Virginia (Tate et al. 1980, Pagels and French 1987, Linzey 1998) south throughout the Coastal Plain and lower Piedmont of North Carolina (Lee et al. 1982, Webster 1988, Mitchell et al. 1995), Coastal Plain and Piedmont of South Carolina (Schacher and Pelton 1979, French 1981, Mengak et al. 1989, Cothran et al. 1991), Coastal Plain, lower Piedmont and extreme northwestern portions of Georgia (Harper 1927, Neuhauser and Baker 1974, Wharton et al. 1981, Laerm et al. 1982, Thurmond and Miller 1994, Parmley et al. 1997, Laerm et al. 1999, Ford et al. 2006), all of Florida (Rand and Host 1942, Moore 1946, Pournelle 1950, Hamilton 1955, Ivey 1959, Layne 1992), and all but the Piedmont of east-central Alabama (Holliman 1963, Wolfe and Rodgers 1969, Linzey 1970, French 1981, Ford et al. 2006). It is found throughout Mississippi (Wolfe 1971, Kennedy et al. 1974, Wolfe and Esher 1981, Wolfe and Lohofener 1983, Jones and Carter 1989), all but the coastal marshes of Louisiana (Lowery 1974), and eastern Texas (Baumgardner et al. 1992, Jones and Jones 1992, Davis and Schmidly 1994). The southern short-tailed shrew is found west of the Tennessee River in western Tennessee (Goodpaster and Hoffmeister 1952, Braun and Kennedy 1983, Kennedy 1991) and western Kentucky (Rose and Seegert 1982, Bryan 1991), in all but northwestern Arkansas (Garland and Heidt 1989, Sealander and Heidt 1990,

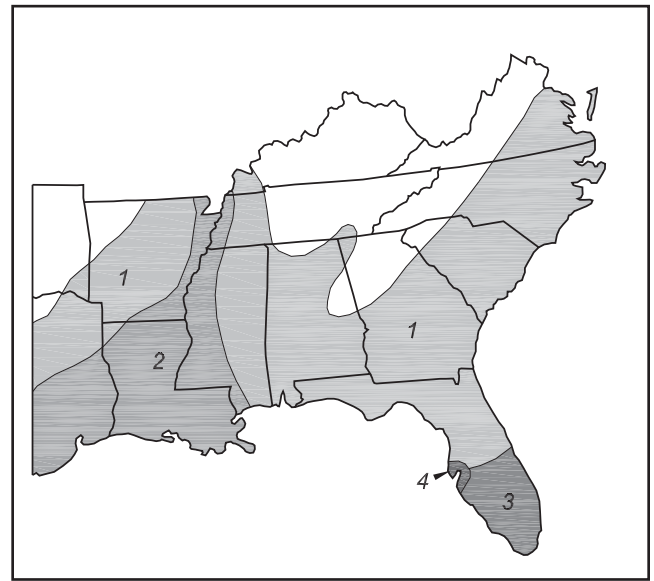


Figure 2. Distribution of *Blarina carolinensis* in the South: (1) *B. c. carolinensis*; (2) *B. c. minima*; (3) *B. c. peninsulae*; (4) *B. c. shermani*.

Tumlison et al. 1992), and into extreme southeastern Oklahoma (Caire et al. 1989).

ABUNDANCE STATUS

The southern short-tailed shrew is common in most of the lower Piedmont and Coastal Plain, except for Florida (Hamilton 1955, Layne 1992), eastern Texas (Schmidly 1983), and Oklahoma (Caire et al. 1989). Density estimates of 11 individuals/ha were reported in South Carolina (Smith et al. 1974). It is uncommon in the southern portions of the Ridge and Valley and Cumberland Plateau, along with the upper Piedmont (Ford et al. 2006). Populations may fluctuate dramatically (Briese and Smith 1974).

PRIMARY HABITATS

The southern short-tailed shrew is considered a habitat generalist; within its range, it is known from all forest cover types except mangrove (*Avicennia germinans*-*Rhizophora mangle*) swamps in Florida. It is common in oldfields (Golley et al. 1965), young loblolly pine (*Pinus taeda*) plantations (Perkins et al. 1989), pine flatwoods (Wolfe and Lohofener 1983, Mullin and Williams 1987), upland margins of pocosins and Carolina bays (Clark et al. 1985), and forested wetlands and bottomland hardwoods (Wharton et al. 1981, Menzel et al. 2005). The southern short-tailed shrew is most abundant in hardwood forests (Golley 1962, Lowery 1974, Kennedy 1991). In the upper Piedmont and southern Appalachians, this species only occurs at low elevations (Ford et al. 2006), utilizing the same cover types as the northern short-tailed

shrew. Hartman et al. (2001) found that southern short-tailed shrews utilize mole tunnels extensively, often preferring these tunnels during cold weather.

REPRODUCTION

The breeding season for southern short-tailed shrews extends from February through November with peaks in spring and late summer–early fall (Dapson 1968, O'Farrell et al. 1977). The litter size ranges from 2–6 and there are multiple litters per year (McCay 2001).

FOOD HABITS

The food habits of the southern short-tailed shrew probably are similar to those of the northern short-tailed shrew, for which more information is known. The diet likely includes annelids, gastropods, lepidopteran larvae, chilopods, arachnids, and other invertebrates. Small quantities of plant material and fungi are eaten, and occasionally small mammals are taken (Calhoun 1941, George et al. 1986, Whitaker et al. 1994). Similar to the northern short-tailed shrew, *B. carolinensis* has modified salivary glands that give the species a venomous bite (Lowery 1974, McCay 2001).

ASSOCIATED SPECIES

The southern short-tailed shrew associates with most other soricids and small rodents within its range. It occupies relatively narrow zones of sympatry with the northern short-tailed shrew to the north (Tate et al. 1980, French 1981, Ford et al. 2006) and Elliot's short-tailed shrew to the west (George et al. 1981, 1982; Jones et al. 1984, Baumgardner et al. 1992). It is broadly sympatric, but not syntopic, with the northern short-tailed shrew in eastern North Carolina where it occupies mineral-based soils and *B. brevicauda* occurs on organic soils (Webster 1996) or in the Blue Ridge (McCay et al. 2004).

VULNERABILITY AND THREATS

There are few threats to its survival in most of the southern region. *Blarina c. peninsulae* is widespread throughout southern peninsular Florida, but at low population densities (Layne 1992). The status of *B. c. shermani* is uncertain as only a few specimens are known from its type locality and subsequent efforts to document its presence have been unsuccessful, suggesting that the subspecies is either rare or extirpated (Layne 1992).

MANAGEMENT SUGGESTIONS

The southern short-tailed shrew is tolerant of the disturbance resulting from many forest management activities (Perkins et al. 1989, Menzel et al. 2005). Conditions that promote abundant coarse woody debris, thick leaf litter, and moist microhabitats may promote higher population densities (Thurmond and Miller 1994).

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Elliot's Short-tailed Shrew

Blarina hylophaga (Elliot, 1899)

Joshua Laerm, Brian R. Chapman, and W. Mark Ford

CONTENT AND TAXONOMIC COMMENTS

Elliot's short-tailed shrew (*Blarina hylophaga*) was described recently as distinct from the southern short-tailed shrew (*B. carolinensis*) and the northern short-tailed shrew (*B. brevicauda*) (Genoways and Choate 1972, George et al. 1981, George et al. 1982, Moncrief et al. 1982). Two subspecies are recognized. *Blarina h. plumbea*, described originally as *B. brevicauda plumbea* by Davis (1941) and referred to *B. carolinensis plumbea* by Schmidly and Brown (1979), is found only at its type locality at the Aransas National Wildlife Refuge, Aransas County, Texas (George et al. 1981, Schmidly 1983, Baumgardner et al. 1992). *Blarina h. hylophaga*, which is more widely distributed, is the only subspecies that occurs in the South.

DISTINGUISHING CHARACTERISTICS

Elliot's short-tailed shrew is intermediate in size between the other two *Blarina* species. Measurements are: total length, 103–120 mm; tail, 19–25 mm; hind foot, 12–16 mm; weight, 13–17 g. This species has small ears that are small and concealed in the pelage. The minute eyes are positioned in a long, pointed snout. The color of the pelage is similar to that of the southern short-tailed shrew, brownish gray to slate gray dorsally and only slightly paler below. Specimens of Elliot's short-tailed shrew may be confused with the southern short-tailed shrew to the south and east and with the northern short-tailed shrew to the north. Distinctions among the species sometimes can be made on the basis of distribution, but in areas of sympatry or parapatry, morphometric or genetic comparisons must be made (George et al. 1981, Moncrief et al. 1982). The dental formula of Elliot's short-tailed shrew is: $I\ 3/1, C\ 1/1, P\ 3/1, M\ 3/3 = 32$ (Figure 1). See keys for additional details.

CONSERVATION STATUS

Elliot's short-tailed shrew has a global rank of Secure (NatureServe 2007). It is Apparently Secure in Oklahoma and is unranked in Arkansas and Louisiana. However, it is classified as Critically Imperiled in Texas.

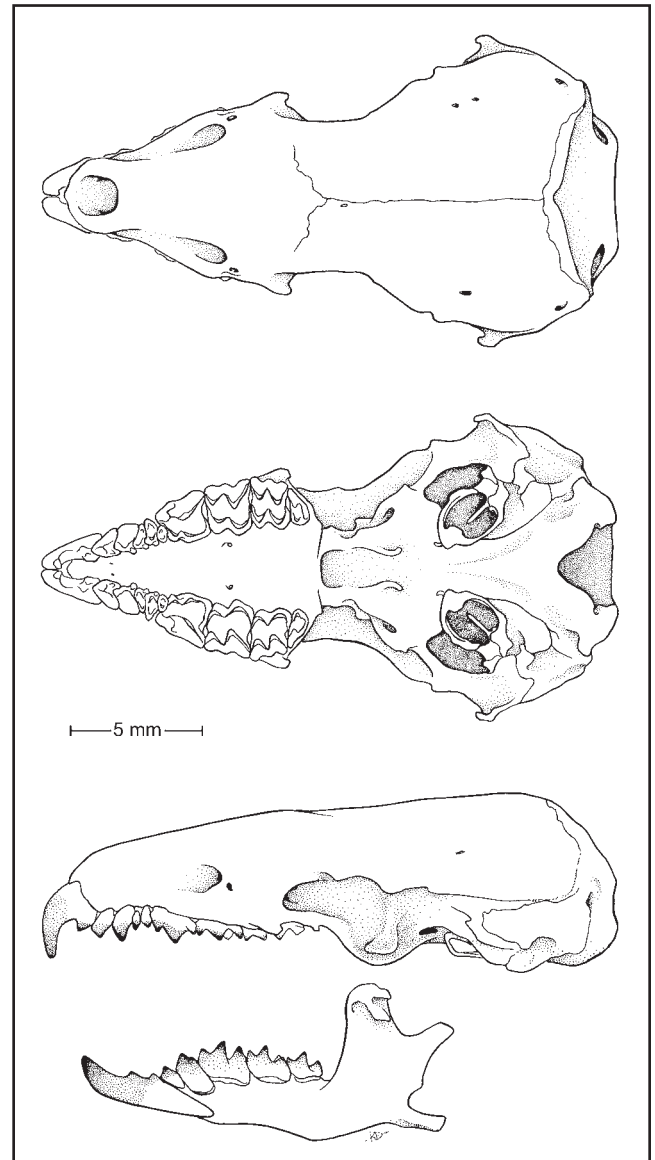


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Blarina hylophaga* from Jefferson County, Kansas (USNM 568207, female).

DISTRIBUTION

The distribution of Elliot's short-tailed shrew extends from east-central Colorado across southern Nebraska into southwestern Iowa and south into Texas and northwestern Arkansas (Jones et al. 1984). Figure 2 depicts the distribution of the short-tailed shrew in

the South. Elliot's short-tailed shrew occurs throughout the eastern two-thirds of Oklahoma except for the southeastern corner (Caire et al. 1989, Stangl and Carr 1997). In Arkansas, the species is restricted to the Ozark and Ouachita Highlands in the northwest third of the state (Garland and Heidt 1989, Sealander and Heidt 1990). There are limited records of this species in Caddo Parish, Louisiana (George et al. 1981). The precise geographic boundaries between Elliot's short-tailed shrew and the northern short-tailed shrew to the north and the southern short-tailed shrew to east and the south are defined poorly (Stangl and Carr 1997). Elliot's short-tailed shrew and the southern short-tailed shrew may be sympatric in northwestern Louisiana (George et al. 1982, Jones et al. 1984).

ABUNDANCE STATUS

Little specific information on abundance and densities of Elliot's short-tailed shrew is available. Similar to other species of *Blarina*, the species probably is uncommon to locally abundant depending on habitat conditions (Caire et al. 1989, Sealander and Heidt 1990, Baumgardner et al. 1992).

PRIMARY HABITATS

In Oklahoma, Elliot's short-tailed shrew is found in oak-elm (*Quercus-Ulmus*) forests and wooded floodplains (Elliot 1899, Caire et al. 1989), woodland-grassland ecotones (McCarley 1961), and areas of emergent rock in the Wichita Mountains (Glass and Halloran 1961). In Arkansas, the species is restricted primarily to the oak-hickory (*Quercus-Carya*) woodlands in the Ozark and Ouachita Highlands (Jones and Glass 1960, Sealander and Heidt 1990), but it also may occur in mixed pine-hardwood (*Pinus* spp.) and loblolly pine-shortleaf pine (*P. taeda-P. echinata*) cover types. Outside of the South, Elliot's short-tailed shrew occurs in tall grass prairie habitats with high vegetative cover, deep litter, and high ambient moisture (Sietman et al. 1994, Matlack et al. 2002).

REPRODUCTION

Information regarding reproduction of Elliot's short-tailed shrew is not available (Caire et al. 1989, Sealander and Heidt 1990). Presumably, it is similar to that of other *Blarina* species.

FOOD HABITS

The food habits of Elliot's short-tailed shrew are not well known (Caire et al. 1989, Sealander and Heidt 1990), but the diet likely includes annelids, gastropods, lepidopteran larvae, chilopods, arachnids,

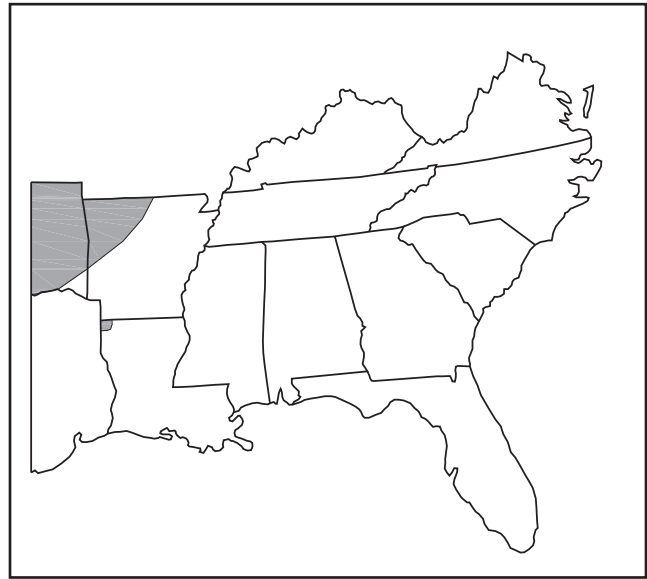


Figure 2. Distribution of *Blarina hylophaga* in the South.

other invertebrates, small vertebrates, and some plant material similar to *B. brevicauda* and *B. carolinensis*. Likewise, Elliot's short-tailed shrew has modified salivary glands that give the species a venomous bite (George 1999).

ASSOCIATED SPECIES

Elliot's short-tailed shrew occurs in communities that include other soricids and the rodents within its distributional range, including the least shrew (*Cryptotis parva*), white-footed mouse (*Peromyscus leucopus*), deer mouse (*P. maniculatus*), and golden mouse (*Ochrotomys nuttalli*). It occupies relatively narrow zones of sympatry with the southern short-tailed shrew in Arkansas (Sealander and Heidt 1990).

VULNERABILITY AND THREATS

There are no apparent threats to the survival of the species. Elliot's short-tailed shrew may be adversely affected by global climate change, long-term drought, or desertification in prairie and dry woodland habitats prone to moisture stress (Matlack et al. 2002).

MANAGEMENT SUGGESTIONS

Management activities that promote long-term site desiccation or habitat conversion are harmful. This species appears to prefer native prairie to hayfields (Sietman et al. 1994). Impacts from forest and range management activities, such as prescribed burning and livestock grazing, are unknown.

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Least Shrew

Cryptotis parva (Say, 1823)

Joshua Laerm, Brian R. Chapman, W. Mark Ford

CONTENT AND TAXONOMIC COMMENTS

The least shrew (*Cryptotis parva*) is distributed widely across the eastern United States. Only two of the nine recognized subspecies occur in the region (Whitaker 1974). *Cryptotis p. parva* is widespread in the region. *Cryptotis p. floridana* once was considered to be restricted to Florida (Whitaker 1974), but Handley and Varn (1994) suggest that populations extend along the Coastal Plain as far north as North Carolina. These coastal populations may represent a distinct species (Hutterer 1993). Choate (1970) revised the Middle American taxa of *Cryptotis*. The literature was reviewed by Whitaker (1974).

DISTINGUISHING CHARACTERISTICS

The least shrew is a small, short-tailed soricid with a long, pointed snout. Measurements are: total length, 70–92 mm; tail, 12–26 mm; hind foot, 9–13 mm; and weight, 3–6 g. This shrew has small ears concealed in the pelage and minute eyes. The pelage color is gray to brown dorsally, often with silver frosting, and light gray ventrally, with a distinctly short tail that is bicolored, dark above and light below. The skull of the least shrew is characterized by having only four upper unicuspid. Because the fourth unicuspid is tiny and difficult to see, the tooth row must be examined carefully. Specimens of the least shrew may be confused with small individuals in the genus *Blarina*, although specimens of *Blarina* have more uniformly lead-colored fur, five upper unicuspid, and are larger in size. The dental formula of the least shrew is: I 3/1, C 1/1, P 2/1, M 3/3 = 30 (Figure 1). See keys for additional details.

CONSERVATION STATUS

The least shrew has a global rank of Secure (NatureServe 2007). It is Secure in Alabama, Georgia, Kentucky, North Carolina, Oklahoma, Tennessee, and Virginia. Arkansas, Louisiana, and Texas classify the shrew as Apparently Secure. It is unranked in Florida, Mississippi, and South Carolina.

DISTRIBUTION

The least shrew is distributed from the Great Lakes south and west to the Great Plains and south into Central America to Panama (Hall 1981). Figure 2

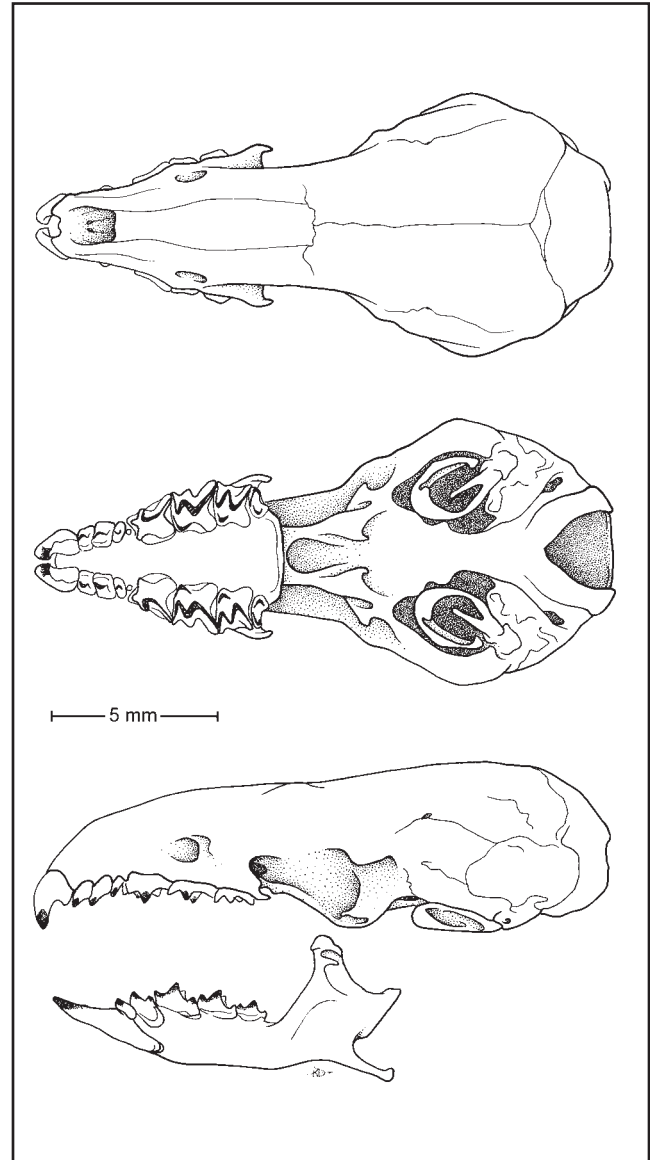


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Cryptotis parva* from Wakulla County, Florida (USNM 527294, male).

depicts the distribution of the least shrew in the South. It occurs throughout Virginia (Jackson et al. 1976, Handley 1979, Cranford and Maly 1990, Pagels 1991, Pagels et al. 1992, Linzey 1998), North Carolina (Lee et al. 1982, Clark et al. 1985, Webster 1988, Laerm et al. 1995, Mitchell et al. 1995), South Carolina (Sherman 1937, Golley 1966, Mengak et al. 1987, Cothran et al. 1991, Handley and Varn 1994), Georgia

(Harper 1927, Neuhauser and Baker 1974, Wharton et al. 1981, Laerm et al. 1982, Ford et al. 1994, Laerm et al. 1995, Ford et al. 1997, Cochran et al. 1999, Laerm et al. 1999a,b), Florida (Hamilton 1941, Moore 1943, Moore 1946, Quay 1949, Pournelle 1950, Ivey 1959, Layne 1974, Whitaker 1974), Alabama (Dusi 1959, Holliman 1963, Wolfe and Rodgers 1969, Laerm et al. 1995), Mississippi (Wolfe 1971, Kennedy et al. 1974, Wolfe and Esher 1981, Jones and Carter 1989), Tennessee (Howell 1954, Howell and Conaway 1952, Smith et al. 1974, Copeland and Caldwell 1991, Kennedy 1991, Harvey et al. 1992, Feldhamer et al. 1993, Linzey 1995), Kentucky (Welter and Sollberger 1939, Fassler 1974, Rose and Seegert 1982, Bryan 1991, Feldhamer et al. 1993), Arkansas (Garland and Heidt 1989, Sealander and Heidt 1990), Texas (Davis 1940, Schmidly 1983, Davis and Schmidly 1994), and Oklahoma (Caire et al. 1989). In Louisiana, it is absent from the coastal marshes in the southeastern portion of the state (Lowery 1974).

ABUNDANCE STATUS

Little information is available on population densities and dynamics of the least shrew, presumably because of difficulty in collection (Whitaker 1974). Getz (1962) indicated that the least shrew had a "spotty" distribution and was not particularly abundant in any location. Estimated densities range from 1.7–4.9 individuals/ha (Howell 1954, Kurta 1995) to over 25 individuals/ha (Hoffmeister and Mohr 1957, Kale 1972). This species may be more abundant than trapping records indicate because its skulls often are found in owl pellets (Davis 1938, Davis 1940, Lowery 1974, Whitaker 1974, Mumford and Whitaker 1982, Hoffmeister 1989, Copeland and Caldwell 1991).

PRIMARY HABITATS

Although it may be present in various seral stages, the least shrew most commonly is associated with oldfield and early successional habitats including grassy, weedy, and brushy fields (Hamilton 1934, Davis and Joeris 1945, Howell 1954, Whitaker 1974, Wolfe and Esher 1981, Bryan 1991, Pagels et al. 1992, Feldhamer et al. 1993, Handley and Varn 1994, Cochran et al. 1999, Bellows et al. 2001, Ford et al. 2006). It also is reported from forested wetlands (Rose and Seegart 1982, Mitchell et al. 1993), coastal islands (Schacher and Pelton 1979, Webster 1988, Laerm et al. 1999a), and brackish tidal salt marshes (Cranford and Maly 1990, Martin et al. 1991), though it apparently is absent from the interior Everglades and mangrove swamps (*Avicennia germinans*-*Rhizophora mangle*). The least shrew generally is absent or rare in mature hardwood and coniferous forests at middle to higher elevations in the southern Appalachians (Laerm et al.

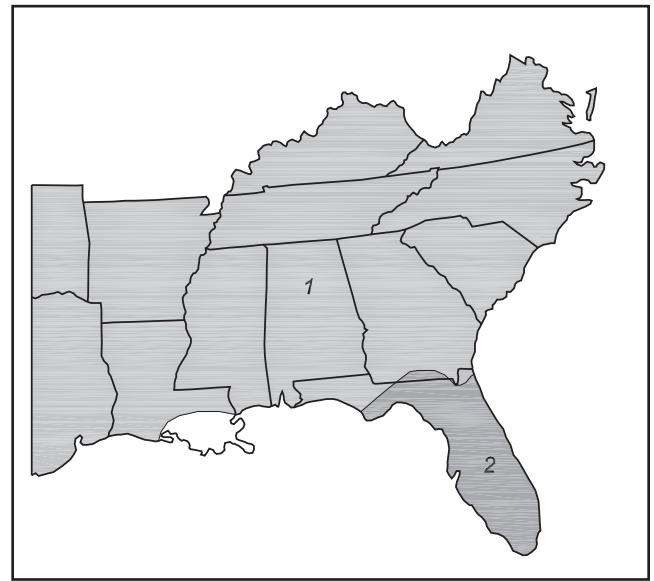


Figure 2. Distribution of *Cryptotis parva* in the South: (1) *C. p. parva*; (2) *C. p. floridana*.

1995, Ford et al. 1997, Laerm et al. 1999a,b; Ford et al. 2006). Although Whitaker (1974) noted that the least shrew was unknown above 905 m in the United States, Pagels (1991) reported it from a red spruce (*Picea rubens*) forest at 1,534 m on Mount Rogers in Grayson County, Virginia. The least shrew constructs burrows or utilizes those of other small mammals under logs, rocks, stumps, and other debris, where it constructs small (5–15 cm diameter) nests of dried grasses and shredded leaves (Smith 1938).

REPRODUCTION

Most breeding activities of the least shrew in the region probably occur between February and November (Hamilton 1944, Davis and Joeris 1945, Whitaker 1974), but some reproduction may occur at other times during the year. Conaway (1958) and Mock (1982) report that the gestation period is between 21–23 days. The litter size is 3–7 with a mean of 4.7 (Whitaker 1974). Adult size is reached at 30–40 days of age, and maximum longevity is approximately 21 months.

FOOD HABITS

The least shrew feeds on many kinds of invertebrates including annelids, gastropods, arachnids, chilopods, orthopterans, and lepidoptera and coleoptera larvae. It also eats small quantities of fungi and some green plant material (Hamilton 1944, Moore 1943, Whitaker 1974, Mumford and Whitaker 1982).

ASSOCIATED SPECIES

Because of its wide distribution, the least shrew may be found in association with nearly all of the mammals

in the region. It commonly occupies habitats with other soricids including the southern short-tailed shrew (*B. carolinensis*), Elliot's short-tailed shrew (*B. hylophaga*), and southeastern shrew (*Sorex longirostris*) in the Coastal Plain, Ozark and Ouachita Highlands and Cross Timbers. It also shares habitat with rodents such as the marsh rice rat (*Oryzomys palustris*), hispid cotton rat (*Sigmodon hispidus*), white-footed mouse (*Peromyscus leucopus*), cotton mouse (*P. gossypinus*), meadow vole (*Microtus pennsylvanicus*) and woodland vole (*M. pinetorum*).

VULNERABILITY AND THREATS

None.

MANAGEMENT SUGGESTIONS

The habitat of the least shrew is favored by the maintenance of oldfield and early successional habitats such as young pine (*Pinus* spp.) plantations prior to canopy closure (Atkeson and Johnson 1979, Yates et al. 1997). How the least shrew responds to site preparation intensity and herbaceous control in the South requires further investigation.

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Crawford's Desert Shrew

Notiosorex crawfordi (Coues, 1877)

Joshua Laerm and W. Mark Ford

CONTENT AND TAXONOMIC COMMENTS

Notiosorex is a monotypic genus. The Crawford's desert shrew (*Notiosorex crawfordi*) has two subspecies; one (*N. c. crawfordi*) occurs in the South. The literature on Crawford's desert shrew was reviewed by Armstrong and Jones (1972).

DISTINGUISHING CHARACTERISTICS

Crawford's desert shrew is a small, short-tailed soricid with small but conspicuous ears. Similar to most shrews, the tiny eyes are concealed although Crawford's desert shrews can see well (Sealander and Heidt 1990). Measurements are: total length, 77–98 mm; tail, 22–30 mm; hind foot, 9–13 mm; weight, 3–6 g. The snout is long and pointed. The pelage is silver-gray to brownish gray dorsally and pale gray to silver-gray ventrally, with a well-haired and indistinctly bicolored tail. In addition to its mouse-like appearance, the Crawford's desert shrew can be distinguished by dental comparison from three sympatric short-tailed shrews, Elliot's short-tailed shrew (*Blarina hylophaga*), the least shrew (*Cryptotis parva*) and the southeastern shrew (*Sorex longirostris*). The Crawford's desert shrew has 3 pairs of unicuspid whereas there are 4 in the least shrew and 5 in Elliot's short-tailed shrew and the southeastern shrew. The dental formula is: I 3/1, C 1/1, P 1/1, M 3/3 = 28 (Figure 1). See keys for additional details.

CONSERVATION STATUS

The Crawford's desert shrew has a global rank of Secure (NatureServe 2007). It is considered Apparently Secure in Texas, Vulnerable in Oklahoma, and Critically Imperiled in Arkansas.

DISTRIBUTION

The Crawford's desert shrew is distributed throughout west-central and northern Mexico into southwestern and south-central United States (Figure 2). Within the region, its range extends into western Oklahoma and eastward into the Ouachita and Ozark Highlands in western Arkansas (Sealander 1952, Clark 1953, Preston and Martin 1963, Preston and Sealander 1969, Tyler and Gilliland 1979, Steward et al. 1988, Caire et al. 1989, Garland and Heidt

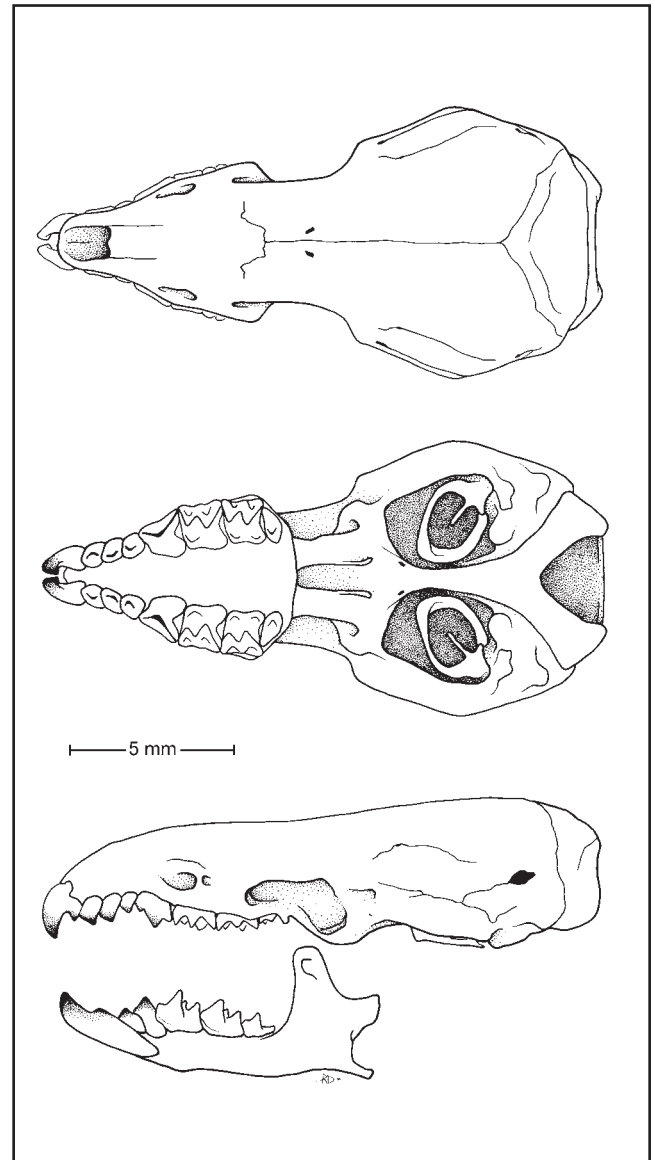


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Notiosorex crawfordi* from Baja California, Mexico (USNM 146933, male).

1989, Sealander and Heidt 1990). It is not reported from eastern Texas (Schmidly 1983).

ABUNDANCE STATUS

Little information is available on Crawford's desert shrew abundance in the region. Preston and Martin (1963) estimated densities elsewhere in Oklahoma at

1 individual/ha. However, in eastern Oklahoma it is known from only a single specimen (Clark 1953, Caire et al. 1989). At the extreme periphery of its range, Sealander and Heidt (1990) indicate that the species appears to be rare in Arkansas with only a few known locality records. However, Sealander and Heidt (1990) and Caire et al. (1989) note that it may be more abundant than available records indicate.

PRIMARY HABITATS

Crawford's desert shrew is reported from a diversity of habitats throughout its range (Armstrong and Jones 1972), but its associations with habitat types in the South are not well known. It is associated with semi-arid scrub communities containing mesquite (*Prosopis* spp.), agave (*Agave* spp.), or oaks (*Quercus* spp.; Armstrong and Jones 1972), and also occurs in riparian habitats containing cottonwood (*Populus* spp.) and juniper (*Juniperus* spp.), ponderosa pine (*Pinus ponderosa*), cultivated fields, and grassy washes (Blossom 1933, Blair 1947, Lindeborg 1960, Carothers 1968, Armstrong and Jones 1972). The Crawford's desert shrew utilizes leaf litter, shrub thickets, brush, and rubbish piles for cover, and has been reported using beehives (Blair 1954, Baker 1962, 1966; Hoffmeister and Goodpaster 1962, Armstrong and Jones 1972). In Oklahoma and Arkansas, it commonly is associated with eastern woodrat (*Neotoma floridana*) nests (Preston and Martin 1963, Caire et al. 1989).

REPRODUCTION

Baker and Spenser (1965) and Armstrong and Jones (1972) suggest that reproduction is restricted to warmer periods of the year, approximately April through November. Three to 5 young are produced. Adult pelage is complete at 90 days of age (Hoffmeister and Goodpaster 1962). Annual litter numbers and longevity are unknown.

FOOD HABITS

No information is available on food habits in the region. Huey (1936) and Hoffmeister and Goodpaster (1962) report that the Crawford's desert shrew feeds on many kinds of invertebrates including larval lepidoptera, larval and adult coleoptera, orthoptera, dicoptera, and chilopods. It also consumes vertebrate carrion.

ASSOCIATED SPECIES

Associated insectivores in the region include the southeastern shrew, least shrew, Elliot's short-tailed shrew, and eastern mole (*Scalopus aquaticus*), whereas associated rodents include the eastern woodrat,

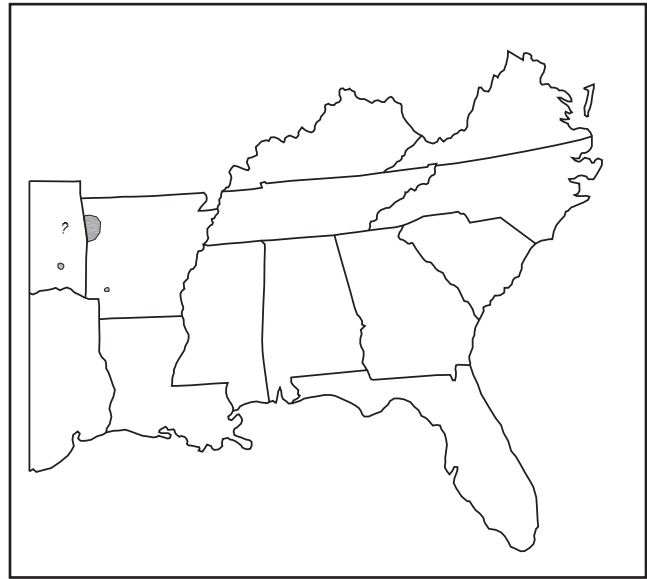


Figure 2. Distribution of *Notiosorex crawfordi* in the South.

white-footed mouse (*Peromyscus leucopus*), and deer mouse (*P. maniculatus*).

VULNERABILITY AND THREATS

Crawford's desert shrew is at the eastern limits of its range in the South representing a distributional vestige from a warmer, drier period in the late Pleistocene (Hibbard and Taylor 1960). Threats to the species viability in the region are unknown. Because its distribution and abundance status are uncertain, additional survey efforts are needed to ascertain population size and habitat preference of this unique species.

MANAGEMENT SUGGESTIONS

No specific management guidelines are known, although the close association of Crawford's desert shrews and eastern woodrats suggest that efforts favoring woodrats could be beneficial.

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Masked Shrew

Sorex cinereus (Kerr, 1792)

Joshua Laerm, W. Mark Ford, and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The subspecific taxonomy of the masked shrew (*Sorex cinereus*) is somewhat unsettled (van Zyll de Jong 1980, 1991; van Zyll de Jong and Kirkland 1989). Twelve subspecies are recognized by Hall (1981) and maps therein indicate that four subspecies occur in the South. The Maryland shrew (*S. c. fontinalis*) is regarded as a separate species by Jones et al. (1997), but van Zyll de Jong and Kirkland (1989) consider it to be a subspecies of *S. cinereus*. Although Handley (1982) believed that *S. c. fontinalis* did not occur in Virginia, Moncrief and Dueser (1998) recently confirmed its existence in the state. Another of the four potential subspecies, *Sorex c. ohioensis*, is no longer considered to be a distinct taxon (van Zyll de Jong and Kirkland 1989). Therefore, we recognize three subspecies in the region: *S. c. fontinalis*, which occurs in the Delmarva Peninsula of Virginia; *S. c. cinereus*, which ranges throughout the Appalachian Mountains, and *S. c. lesueurii*, which occurs through the lower Midwest including a small portion of northcentral Kentucky along the Ohio River.

DISTINGUISHING CHARACTERISTICS

The masked shrew is a small, long-tailed soricid with the following measurements: total length, 80–111 mm; tail, 34–48 mm; hind foot, 10–14 mm; weight, 3–5 g. The dental formula of the masked shrew is: I 3/1, C 1/1, P 3/1, M 3/3 = 32 (Figure 1). This species has small ears concealed in the pelage, minute eyes, and a long, pointed snout. The pelage is dark grayish-brown dorsally and light grayish-brown ventrally. Hair length and density is greater in the winter than summer months (Rinehart-Whitt and Pagels 2000). The tail is distinctly bicolored, dark above and pale below. The masked shrew may be confused with the southeastern shrew (*S. longirostris*) and pygmy shrew (*S. hoyi*). The ranges of the masked shrew and the southeastern shrew overlap in the southern Appalachians (Ford et al. 2001). The two species are segregated by altitude with *S. cinereus* generally restricted to higher elevation, mesic forest communities and *S. longirostris* restricted to lower elevation, xeric communities and early successional stages (Ford et al. 2006). Furthermore, the two are distinguishable on the basis of bivariate comparisons of body and skull features (Junge and Hoffman 1981, Laerm et al. 1996). The pygmy shrew occurs sympatrically with the masked

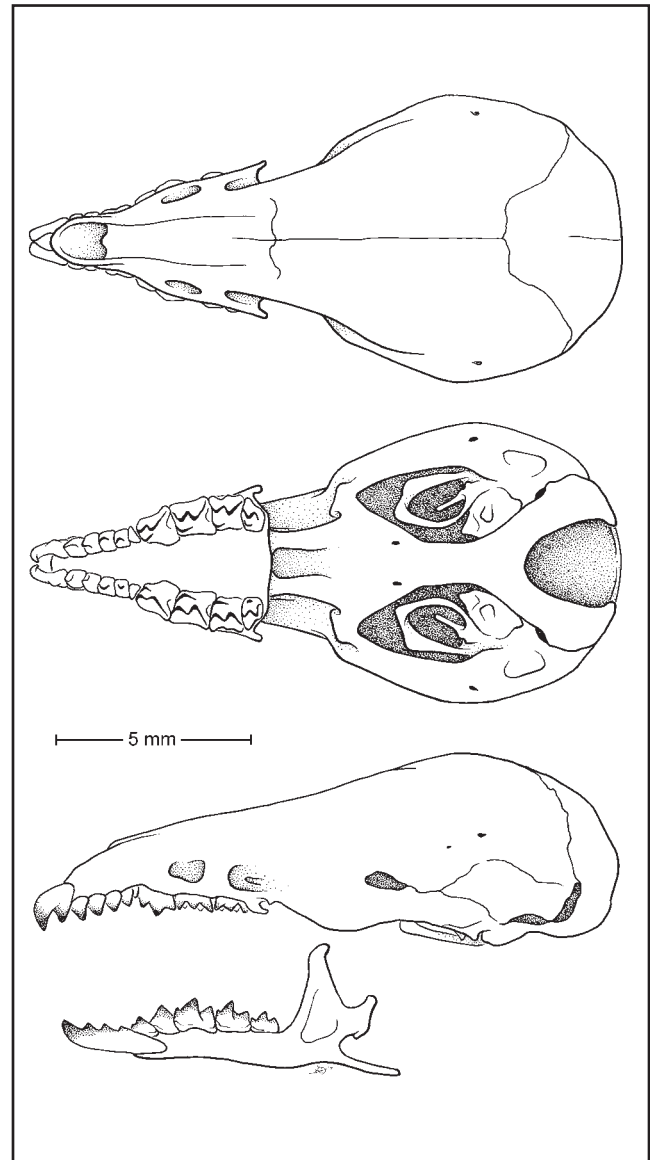


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sorex cinereus* from Giles County, Virginia (USNM 462614, female).

shrew; it is smaller and the skull is easily distinguished by minute third and fifth upper unicuspid.

CONSERVATION STATUS

The masked shrew has a global rank of Secure (NatureServe 2007). It is Secure in Virginia and Apparently Secure in North Carolina and Tennessee.

It is Vulnerable in Kentucky and Imperiled in Georgia. It is unranked in South Carolina.

DISTRIBUTION

The masked shrew has the largest range and exhibits the greatest geographic variation of any North American *Sorex*, ranging throughout the transcontinental boreal forests in Alaska and Canada into the Ohio Valley, the Appalachians, and Rocky Mountains. Figure 2 depicts the distribution of the masked shrew in the South. The species occurs throughout western and northern Virginia (Pagels and Tate 1976, Pagels and Handley 1989, Bulmer and King 1992, Pagels et al. 1994, Mitchell et al. 1997, Linzey 1998), extreme eastern Kentucky (Barbour 1951, Bryan 1991), eastern Tennessee (Conaway and Howell 1953, Smith et al. 1974, Harvey et al. 1991, 1992; Linzey 1995), western North Carolina (Conaway and Howell 1953, Johnston 1967, Gentry et al. 1968, Whitaker et al. 1975, Lee et al. 1982, Adams and Hammond 1991, Ford et al. 1999, Brannon 2000, Ford et al. 2000, 2001), extreme northwestern South Carolina (Laerm et al. 1995), and northeastern Georgia (Wharton 1968, Ford et al. 1994, 2001). In the Appalachians, it exhibits an altitudinal distribution along a latitudinal gradient. It does not occur below 400 m elevation in northern Virginia (Pagels and Handley 1989) or below 600 m in the southern Appalachians of Georgia and North Carolina (Ford et al. 2001, 2006). *Sorex c. fontinalis* is restricted to the Delmarva peninsula of Virginia regionally (Moncrief and Dueser 1998) and *S. c. lesueurii* is found only in northcentral Kentucky in Henderson and Union counties (French 1978, Bryan 1991).

ABUNDANCE STATUS

In the South, the masked shrew is the most common long-tailed shrew throughout much of the central and southern Appalachian Mountains. Kalko and Handley (1993) reported that it constituted 52% of all long-tailed shrews that were snap-trapped over a 15-year period and 77% of all small mammals recovered from pitfall traps in a one-year study at Mountain Lake Biological Station, Giles County, Virginia. In high elevation red spruce (*Picea rubens*) forest communities of Virginia, 89.4% of the soricids were masked shrews (Pagels et al. 1994). Similarly, Harvey et al. (1991, 1992) reported that 45% and 61.2%, respectively, of all soricids recovered from 2 pitfall surveys in the Cherokee National Forest in eastern Tennessee were *S. cinereus*. In western North Carolina, Ford et al. (1999, 2001, 2006) found that masked shrews constituted more than 75% of the long-tailed soricids in high elevation forests (>1,000 m). However, at the periphery of its range in northern Georgia (Ford et al. 1994, 2006) and South Carolina (Laerm et al. 1995,

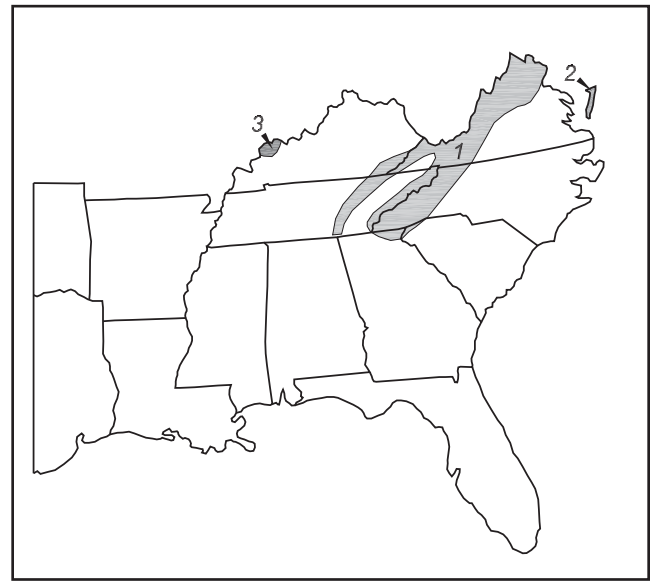


Figure 2. Distribution of *Sorex cinereus* in the South: (1) *S. c. cinereus*; (2) *S. c. fontinalis*; (3) *S. c. lesueurii*.

Ford et al. 2006), it is rare, occurring only at the highest elevations or in sheltered cove and riparian areas.

PRIMARY HABITATS

Throughout most of its range, the masked shrew is a habitat generalist (Kirkland 1991, Mitchell et al. 1997, Ford and Rodrigue 2001), although it is considered more of a high elevation, mesic cover type specialist in the southern Appalachians (Pagels et al. 1994, Ford et al. 2006). Moisture is the primary factor that influences local distribution and abundance (Pruitt 1959, Getz 1961, Spencer and Pettus 1966, Pagels et al. 1994). In the southern Appalachians, it occurs in hardwood and coniferous forests with shady, moist conditions. In Georgia and South Carolina, the presence of masked shrews indicates vegetative and faunal conditions of functional, high integrity northern communities (Ford et al. 2004). Structural features providing cover such as heavy leaf litter, fallen logs, rocks, and stumps are important, but its abundance is correlated with soil moisture capacity, organic matter, and understory vegetation rather than the actual structural components (Pagels et al. 1994, Brannon 2000, Ford and Rodrigue 2001, Ford et al. 2002). Conditions favorable to the masked shrew occur in red spruce-Fraser fir (*Abies fraseri*), northern hardwood, cove hardwood, mesic oak (*Quercus* spp.), and white pine-eastern hemlock (*Pinus strobus*-*Tsuga canadensis*) cover types with abundant rhododendron (*Rhododendron maximum*) understories. In favorable habitat at higher elevations, masked shrews are present in most seral stages (Ford et al. 2000, Ford and Rodrigue 2001). In the southern Appalachians above 1,000 m elevation, it also can occur in more xeric cover types

including pitch pine (*P. rigida*)-dry site oak, but typically only in areas with dense mountain laurel (*Kalmia latifolia*) or vaccinium (*Vaccinium* spp.) shrub layers, substantial coarse woody debris, or other structural features such as emergent rock with moist microhabitats (Ford et al. 1999, 2006). Masked shrews utilize shallow burrows or runways of other small mammals, or construct their own, beneath leafy cover and under other debris. Spherical nests a few centimeters in diameter, constructed of leaves and grasses, are located under logs, rocks, stumps, and other debris.

REPRODUCTION

Breeding occurs from April through October, and there usually are 2 litters/breeding season, one in the spring and another in the fall (French 1985). Gestation lasts 19–22 days, and 1–7 young are produced per litter. Breeding occurs after their first winter (French 1980, 1985). Few survive more than 1 year.

FOOD HABITS

The masked shrew appears to be a food generalist (Getz 1961, Pagels et al. 1994). It consumes chilopods, arachnids, larval coleoptera and lepidoptera, hymenoptera, gastropods, and possibly small vertebrates such as larval salamanders (Whitaker and Mumford 1972, French 1984, Whitaker and French 1984, Ryan 1986, Whitaker and Hamilton 1998). Because of the shrew's small size, prey items <10 mm in length dominate the diet (Churchfield 2002).

ASSOCIATED SPECIES

In the South, masked shrews are associated with insectivores such as the northern short-tailed shrew (*Blarina brevicauda*), smoky shrew (*Sorex fumeus*), and pygmy shrew. Masked shrews also share habitats with rodent species including the white-footed mouse (*Peromyscus leucopus*), deer mouse (*P. maniculatus*), southern red-backed vole (*Clethrionomys gapperi*), and woodland jumping mouse (*Napaeozapus insignis*).

VULNERABILITY AND THREATS

Outside of the Appalachians and north-central Kentucky, the masked shrew is largely absent in the South. Because the species is on the periphery of its range regionally and is linked to montane communities with northern affinities, potential threats might include global climate change or activities that further reduce the extent of northern hardwood or spruce-fir forests. Future local extirpation of eastern hemlock from hemlock adelgid (*Adelges tsugae*)

defoliation at mid-elevation riparian areas might be detrimental to masked shrews.

MANAGEMENT SUGGESTIONS

At higher elevations in the central and southern Appalachians, masked shrews are tolerant of forest management activities such as timber harvest and prescribed burning so long as cool, moist conditions are retained (Kirkland 1990, Ford et al. 2000, Ford and Rodrigue 2001). Management impacts at lower elevations or on the southernmost periphery of its range in the region are unknown, although disturbance and vegetation change could ecologically favor southeastern and pygmy shrews over masked shrews.

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Long-tailed Shrew

Sorex dispar (Batchelder, 1911)

Joshua Laerm and W. Mark Ford

CONTENT AND TAXONOMIC COMMENTS

Two subspecies of the long-tailed shrew (*Sorex dispar*) currently are recognized (Kirkland 1981), but controversy exists regarding their distributions. Schwartz (1956) described *S. d. blitchi*, restricting the species to Tennessee and North Carolina. Populations from Pennsylvania northward were referred to as *S. d. dispar*, whereas populations in Virginia and West Virginia were characterized as intergrades. Handley (1979, 1991) and Pagels and Tate (1976) considered populations from West Virginia and Virginia as far south as Clinch Mountain and Whitetop Mountain as *S. d. dispar*. However, Kirkland and Van Duesen (1979) and French and Kirkland (1983) revised the taxon and referred to populations south of Pennsylvania as *S. d. blitchi*. The long-tailed shrew also is known as the rock shrew, an alternate vernacular name. The literature on the long-tailed shrew is reviewed by Kirkland (1981).

DISTINGUISHING CHARACTERISTICS

The long-tailed shrew is a medium-sized, long-tailed soricid. The measurements of the long-tailed shrew are: total length, 103–136 mm; tail, 46–67 mm; hind foot, 12–16 mm; weight, 3–8 g. The dental formula is: I 3/1, C 1/1, P 3/1, M 3/3 = 32 (Figure 1). The small ears are concealed in pelage, which is slate gray dorsally and slightly paler ventrally. The tail usually is well furred (older individuals may lose significant tail hair), but not distinctly bicolored. Specimens of the long-tailed shrew are often confused with the smoky shrew (*S. fumeus*) because both species are of similar size and coloration. However, the smoky shrew has a wider body and a shorter, bicolored tail (Junge and Hoffman 1981).

CONSERVATION STATUS

The long-tailed shrew has a global rank of Apparently Secure (NatureServe 2007). It is Vulnerable in Virginia and Imperiled in North Carolina and Tennessee. Both Georgia and Kentucky classify it as Critically Imperiled.

DISTRIBUTION

Figure 2 depicts the distribution of the long-tailed shrew in the South. The shrew is endemic to the Appalachian Mountains and is distributed from New Brunswick south to Georgia (Schwartz 1956, Handley

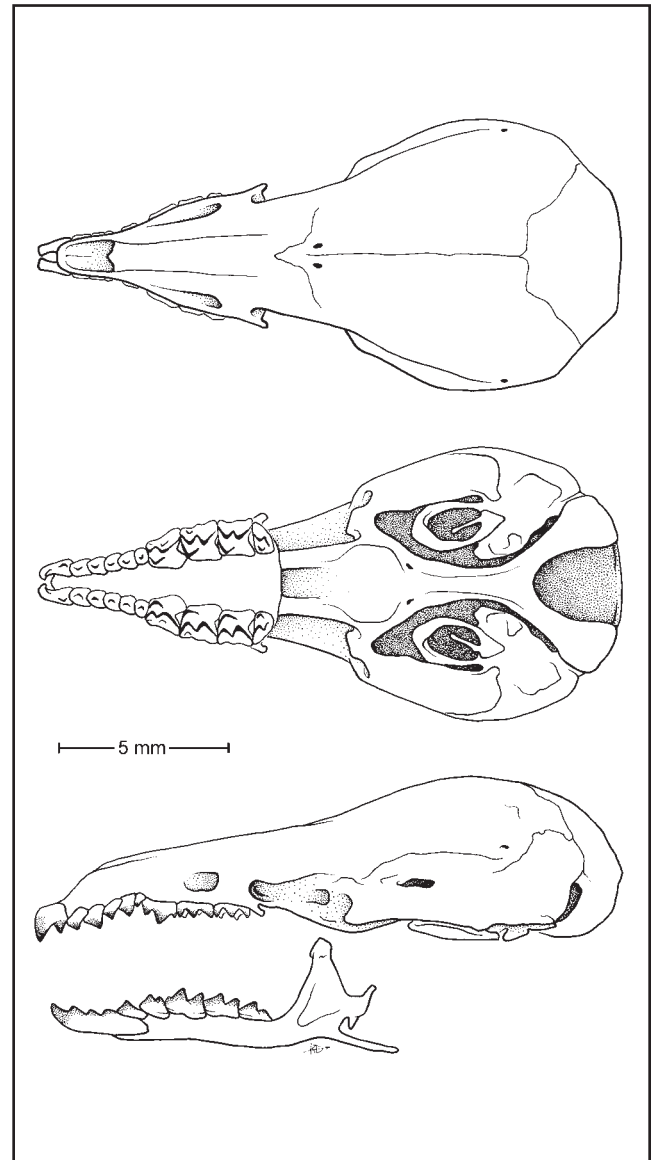


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sorex dispar* from Ulster County, New York (USNM 555462, male).

1971, Kirkland et al. 1979, Kirkland 1981, French and Kirkland 1983, Laerm et al. 1997). It occurs in the higher elevations of western Virginia (Holloway 1957, Pagels and Tate 1976, Pagels 1987, Mitchell et al. 1997, Linzey 1998), and eastern Tennessee (Conaway and Pfitzer 1952, Conaway and Howell 1953, Linzey and Linzey 1971, Smith et al. 1974, Harvey et al. 1992). Stucki

(1967) reported a disjunct population of long-tailed shrews in the gorge of Fall Creek Falls State Park, Van Buren County, Tennessee. Two additional records have been recorded from Savage Gulch, Grundy County, Tennessee (M. L. Kennedy, University of Memphis, personal communication), perhaps indicating that the species might occur in other rock-strewn, sheltered gorge areas in the Cumberland Mountains and Plateau of Kentucky and Tennessee. The long-tailed shrew is known from Black Mountain in Harlan and Letcher counties, Kentucky (Caldwell 1980, Bryan 1991) and one record in Rabun County in northeastern Georgia (Laerm et al. 1997). Its range is extremely limited in western North Carolina (Lee et al. 1982, Webster 1987), and it has not been reported in South Carolina (Laerm et al. 1999).

ABUNDANCE STATUS

Once considered very rare in the central and southern Appalachians, the long-tailed shrew is considered more widely distributed than previously supposed (Kirkland and Van Duesen 1979, Pagels 1987, Handley 1991, Kalko and Handley 1993, Laerm et al. 1999). No population estimates are available. Published records and trapping records do suggest that it is rare, particularly near its distributional limit in the southern Appalachians. It may be locally common in appropriate habitat (Ford and Rodrigue 2001). Kalko and Handley (1993) reported that the long-tailed shrew constituted about 10% of the *Sorex* recovered over a 15-year period in the northern Ridge and Valley section of Virginia.

PRIMARY HABITATS

Forest cover associations inhabited by the long-tailed shrew include red spruce–Fraser fir (*Picea rubens*–*Abies fraseri*), northern hardwood, cove hardwood, and white pine–eastern hemlock (*Pinus strobus*–*Tsuga canadensis*). Although it has been reported from younger seral stages (Kirkland 1977, Ford and Rodrigue 2001), it is found generally in mature forest stands characterized by cool, moist, moss-covered, humus-laden habitats associated with rockslides and talus slopes at high elevations or north-facing slopes. In the northern and central Appalachians, the long-tailed shrew can occur along small mountain streams and bogs (Kirkland et al. 1976, Kirkland and Van Duesen 1979, Kirkland 1981). Ford and Rodrigue (2001) speculate that moist and sheltered riparian zones might serve as dispersal or travel corridors between suitable talus areas. Most descriptions emphasize the significance of emergent rock and colluvial talus in the habitat (Richmond and Grimm 1950, Conaway and Pfitzer 1952, Holloway 1957, Kirkland and Van Duesen 1979, Kirkland 1981, Laerm et al. 1997, Ford et al.

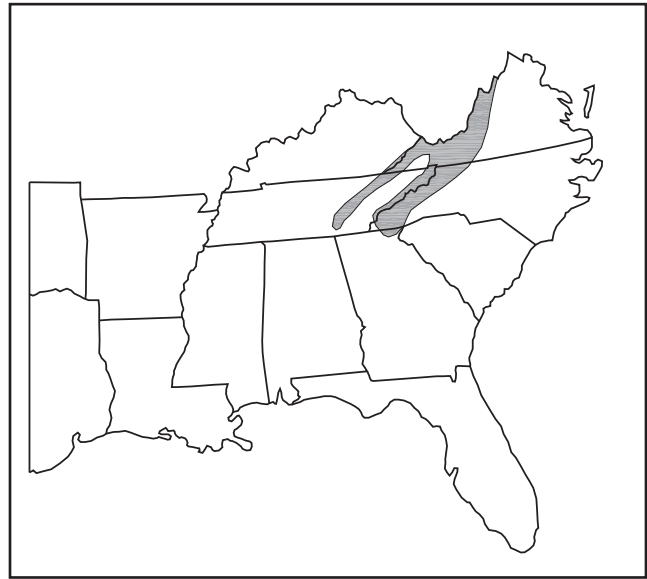


Figure 2. Distribution of *Sorex dispar* in the South.

2006), where it apparently uses cool deep crevices and underground burrows. Elevational distributions range from as low as approximately 500 m on the Cumberland Plateau of Tennessee and possibly Kentucky to 1,000 m and higher in the Allegheny Plateau and northern Ridge and Valley in Virginia and throughout the Blue Ridge in Georgia, Tennessee, North Carolina and Virginia.

REPRODUCTION

Little is known about reproduction in the long-tailed shrew. The breeding season extends from early spring to late summer. Pregnant and lactating females are reported from May through August. Two to 5 embryos are documented, but gestation is unknown. Reproductive information is reviewed by Richmond and Grimm (1950), Kirkland and Van Duesen (1979), and Kirkland (1981).

FOOD HABITS

The food habits of the long-tailed shrew also are poorly known. The species feeds on invertebrates including dipterans, coleopterans, orthopterans, arachnids, and chilopods (Richmond and Grimm 1950, Conaway and Pfitzer 1952, Kirkland 1981).

ASSOCIATED SPECIES

Within emergent rock habitat, the long-tailed shrew commonly is associated with other insectivores that occur in Appalachian mesophytic forests including the northern short-tailed shrew (*Blarina brevicauda*), masked shrew (*S. cinereus*), and smoky shrew, as well as rodents such as the deer mouse (*Peromyscus*

maniculatus), southern red-backed vole (*Clethrionomys gapperi*), and rock vole (*Microtus chrotorrhinus*). The American water shrew (*S. palustris*) and woodland jumping mouse (*Napaeozapus insignis*) are common associates along streamsides where long-tailed shrews occur in the central Appalachians.

VULNERABILITY AND THREATS

The long-tailed shrew is one of the least known mammals of the region; additional demographic and life history data are needed. The extent of appropriate forest cover types and microhabitat features throughout the Appalachians suggests that significant habitat exists in public ownership on inoperable and rugged sites, and there are no apparent short-term threats to the species viability. Nonetheless, impacts to long-tailed shrews from eastern hemlock reduction from hemlock woolly adelgid (*Adelges tsugae*) and global climate change effects on northern hardwood and montane boreal communities in the southern Appalachians merit future research and monitoring.

MANAGEMENT SUGGESTIONS

Although management effects on long-tailed shrews are unknown, activities that reduce micro-site moisture and shading in talus slopes or along montane riparian zones should be avoided. Kirkland (1977) found long-tailed shrews in young clearcuts in red spruce (<5 years) and Ford and Rodrigue (2001) found them in intermediate-aged (e.g., 70 yrs) red spruce-eastern hemlock stands indicating this species is tolerant of overstory disturbance in very mesic, high elevation sites. Regardless, identification and protection of favorable colluvial rock habitats would benefit this species.

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Smoky Shrew

Sorex fumeus (Kerr, 1792)

Joshua Laerm, W. Mark Ford, and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Two subspecies of smoky shrew (*Sorex fumeus*) are recognized (Hall 1981). Only one, *S. f. fumeus*, occurs in the South. Huggins and Kennedy (1989) examine patterns of morphological variation in the species. Owen (1984) reviews the literature on the smoky shrew.

DISTINGUISHING CHARACTERISTICS

The smoky shrew is a medium-sized, long-tailed soricid with the following measurements: total length, 104–130 mm; tail, 37–52 mm; hind foot, 12–15 mm; weight, 6–10 g. This species has small ears that are concealed in the pelage, minute eyes, and a long, pointed snout. The dorsal pelage is dark gray in summer and gray with brown overtones in winter, whereas the ventral pelage is slightly paler than the dorsum. The tail is distinctly bicolored, dark above and pale below. Specimens of the smoky shrew are most often confused with the long-tailed shrew (*S. dispar*) which has a longer, unicolor tail (Junge and Hoffman 1981). The dental formula of the smoky shrew is: I 3/1, C 1/1, P 3/1, M 3/3 = 32 (Figure 1). See keys for additional details.

CONSERVATION STATUS

The smoky shrew has a global rank of Secure (NatureServe 2007). It is Secure in Kentucky and Virginia, and Apparently Secure in North Carolina, South Carolina, and Tennessee. Georgia lists it as Vulnerable.

DISTRIBUTION

The range of the smoky shrew extends from south-central Ontario and the Maritime Provinces south throughout the Appalachian Highlands to Georgia. Figure 2 depicts the distribution of the shrew in the South. The species ranges from the foothills of the Blue Ridge across western Virginia (Pagels and Tate 1976, Handley 1992, Kalko and Handley 1993, Mitchell et al. 1997, Linzey 1998), eastern and middle Kentucky except portions of the Bluegrass (Barbour 1951, Fassler 1974, Bryan 1991, Kiser and Meade 1993), eastern Tennessee west to the eastern Highland Rim of the Interior Low Plateau (Howell and Conaway 1952, Smith et al. 1974, Kennedy and Harvey 1980,

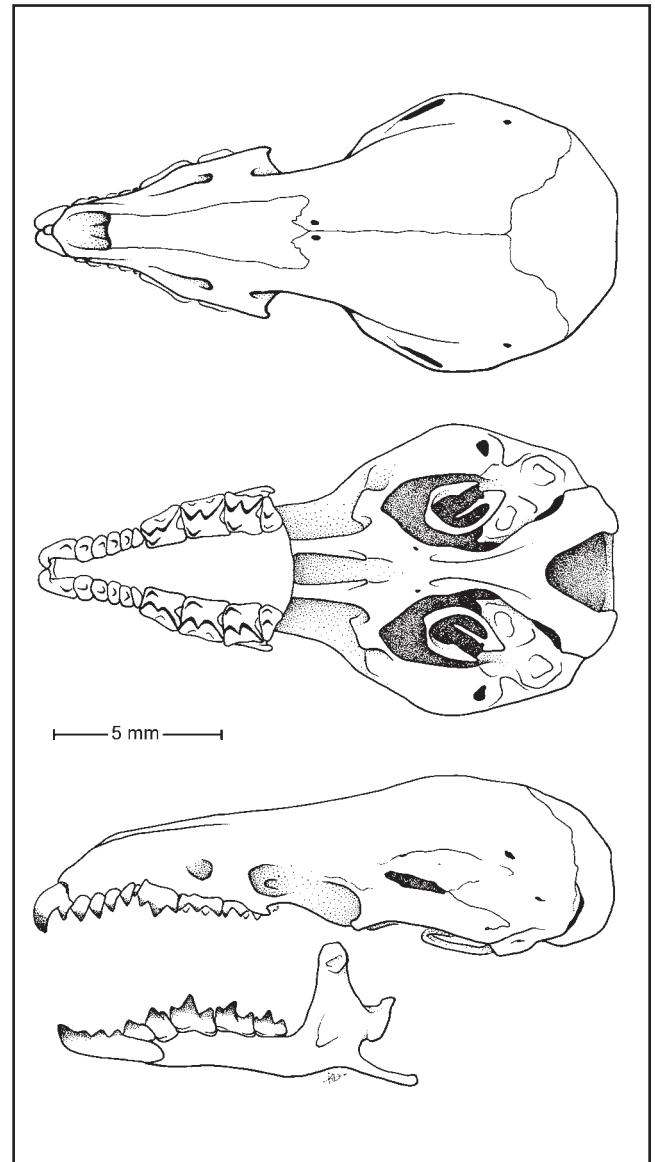


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sorex fumeus* from Hampshire County, West Virginia (USNM 489760, male).

Harvey et al. 1991, 1992; Linzey 1995) and in the Upper Piedmont and Blue Ridge of western North Carolina, northwestern South Carolina, and northern Georgia (Coleman 1948, Odom 1949, Schwartz 1955, Paul and Quay 1963, Johnston 1967, Whitaker et al. 1975, Lee et al. 1982, Mengak et al. 1987, Ford et al. 1994, Laerm et al. 1995a, 1999; Brannon 2000, Ford et al.

2000, 2006). A lone record from the Cumberland Plateau in northwestern Georgia (Laerm et al. 1995b) may indicate its possible presence nearby in Alabama.

ABUNDANCE STATUS

The smoky shrew is a common component of small mammal communities throughout the Appalachian Mountains and portions of the Interior Low Plateau, but it is absent in the Coastal Plain and most of the Piedmont in the southern United States. The smoky shrew is very abundant in suitable habitat (Pagels and Tate 1976, Caldwell and Bryan 1982, McComb and Rumsey 1982, Mengak et al. 1987, Harvey et al. 1991, 1992; Kalko and Handley 1993, Ford et al. 1994, 1997; Laerm et al. 1999, Ford et al. 2006). Data extrapolated from Ford et al. (1997) indicate that densities can reach over 75/ha in mesic forest habitat at mid-elevations in the Blue Ridge of Georgia. Although higher densities have been reported north of the region (Bole 1939), the smoky shrew often is the most abundant in southern Appalachian surveys (Laerm et al. 1999, Ford et al. 2006).

PRIMARY HABITATS

Owen (1984) reviews the literature on the habitat and ecological preferences of the smoky shrew. It is reported from red spruce-Fraser fir (*Picea rubens-Abies fraseri*), northern hardwood, cove hardwood, white pine-eastern hemlock (*Pinus strobus-Tsuga canadensis*), oak hickory (*Quercus-Carya* spp.), mixed pine-hardwood, and xeric cover types. Optimal habitats are high-elevation (>700 m), mesic forest communities with abundant debris such as colluvial talus, emergent rock and large coarse woody debris (Ford et al. 1997, 2006) along with moist, friable soils, herbaceous ground vegetation, and considerable leaf litter (Barbour 1951, Howell and Conaway 1952, Mengak et al. 1987, Ford et al. 1994, Laerm et al. 1995a, 1999). In North Carolina, Brannon (2000) found that the smoky shrew was linked to coarse woody debris in advanced decay states that enabled it to burrow after prey, reducing competition with the smaller masked shrew (*S. cinereus*). The smoky shrew also can occur in bogs and swamps at one end of the moisture gradient, and xeric forest communities on dry, south-facing slopes, exposed ridgelines, and grassy ecotones (McComb and Rumsey 1982, Laerm et al. 1999, Ford et al. 1999, 2006). It is reported from all seral stages, but may be particularly abundant in forest regeneration and mature stands where there is considerable forest floor structure (Kirkland 1977, McComb and Rumsey 1982, Ford et al. 1997, Ford and Rodrigue 2001).

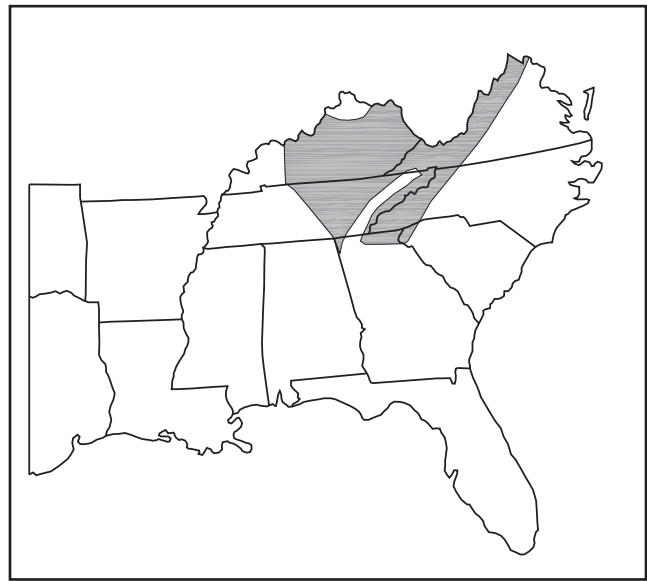


Figure 2. Distribution of *Sorex fumeus* in the South.

REPRODUCTION

Breeding probably occurs all year (McCay et al. 1998), but most reproductive activities are concentrated from March to October (Owen 1984). The gestation period is 19–22 days; 2–3 litters may be produced each year, with 2–8 young/litter. Sexual maturity usually is reached after overwintering. The maximum longevity is about 98 weeks.

FOOD HABITS

Whitaker et al. (1975), Linzey and Linzey 1973, and Owen (1984) report that smoky shrew diets include the larvae and adults of numerous insects and arachnids, chilopods, annelids, and gastropods. The smoky shrew may also feed on vertebrate carrion and small quantities of vegetation and fungi (Linzey 1995).

ASSOCIATED SPECIES

The relatively high elevation habitats in the South occupied by the smoky shrew frequently contain other insectivores such as the northern short-tailed shrew (*Blarina brevicauda*), masked shrew, pygmy shrew (*S. hoyi*), long-tailed shrew, and American water shrew (*S. palustris*). The smoky shrew also shares habitat with various rodents including the white-footed mouse (*Peromyscus leucopus*), deer mouse (*P. maniculatus*), golden mouse (*Ochrotomys nuttalli*), southern red-backed vole (*Clethrionomys gapperi*), and the woodland jumping mouse (*Napaeozapus insignis*).

VULNERABILITY AND THREATS

Nowhere do there appear to be significant threats to its survival where it occurs in the southern Appalachians and Interior Low Plateau.

MANAGEMENT SUGGESTIONS

The smoky shrew is tolerant of most management activities including even-aged and uneven aged silviculture (McComb and Rumsey 1982, Ford et al. 1997, 2000) and prescribed burning (Ford et al. 1999), as long as moist conditions with abundant ground structure remain (Ford and Rodrigue 2001, Ford et al. 2002).

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Pygmy Shrew

Sorex hoyi (Baird, 1858)

W. Mark Ford, Joshua Laerm, and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Because early taxonomists overlooked a small, disk-like upper unicuspid, the pygmy shrew (*Sorex hoyi*) was originally described as *Microsorex hoyi*. The supposed total dental complement was less than the 32 teeth characteristic of the genus *Sorex*. The error was subsequently recognized, but *Microsorex* was retained until Diersing (1980) relegated it to subgeneric status. Many common references and most field guides erroneously retain *Microsorex* (Hall 1981). Five subspecies of *S. hoyi* currently are recognized (Diersing 1980). Pygmy shrews in the southern United States, referred to *S. h. winnemana* by Diersing (1980), were believed to be isolated from populations of *S. h. thompsoni* to the north by 300 km. Records from western Maryland and Pennsylvania (Kirkland et al. 1987, Kirkland and Sheppard 1994, Laerm et al. 1994) indicate that there is a continuous distribution of *S. hoyi* from the southern to the central Appalachians. The literature on *S. (Microsorex) hoyi* is reviewed by Long (1974).

DISTINGUISHING CHARACTERISTICS

The pygmy shrew is the smallest mammal in the southern United States. It is a minute, long-tailed soricid with a long, pointed snout. The measurements are: total length, 70–96 mm; tail, 25–34 mm; hind foot, 7–10 mm; weight, 2–4 g. This species has tiny eyes and ears that are concealed in the pelage. The dorsal pelage is reddish gray-brown during the summer, becoming gray-brown in the winter; the ventral pelage is much lighter. The tail is distinctly bicolored, dark above and pale below. The pygmy shrew may be confused with the masked shrew (*S. cinereus*) and southeastern shrew (*S. longirostris*) with which it is sympatric. The minute third upper unicuspid distinguishes *S. hoyi* from all other shrews in the region (Junge and Hoffman 1981). The dental formula of the pygmy shrew is: $I\ 3/1, C\ 1/1, P\ 3/1, M\ 3/3 = 32$ (Figure 1). See keys for additional details.

CONSERVATION STATUS

The pygmy shrew has a global rank of Secure (NatureServe 2007). It is Apparently Secure in Kentucky and Virginia, but Vulnerable in North Carolina and South Carolina. The shrew is Critically Imperiled

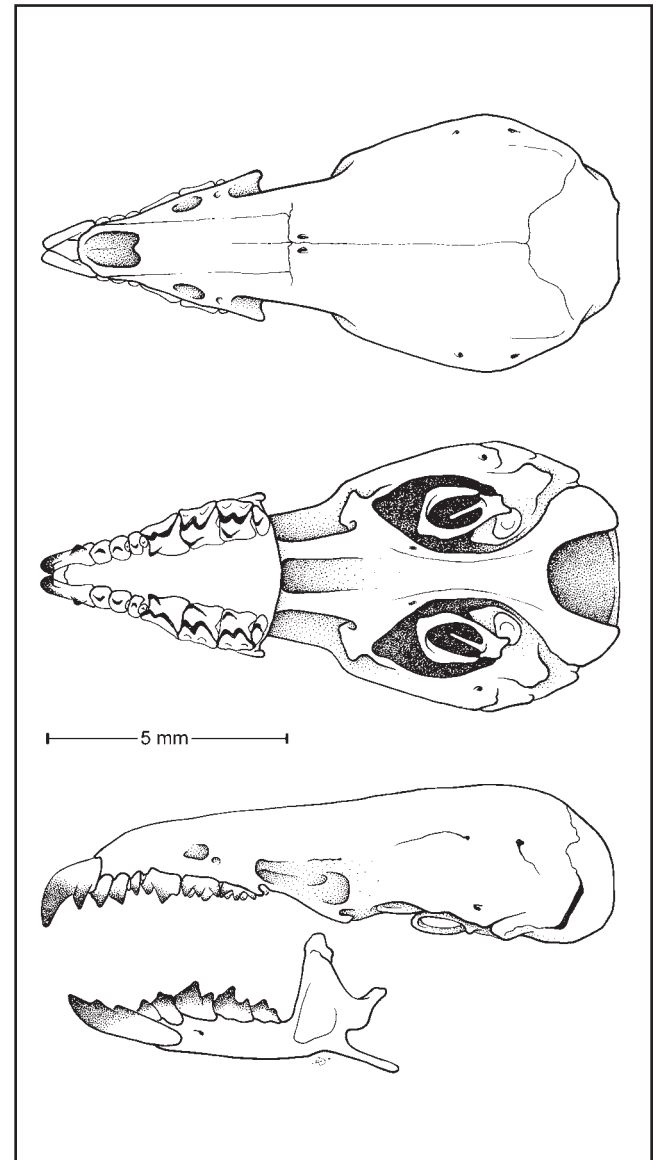


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sorex hoyi* from Ft. Resolution, Mackenzie, Northwest Territories (USNM 110889, female).

in Alabama. Georgia and Tennessee classify the pygmy shrew as Imperiled.

DISTRIBUTION

The pygmy shrew occurs from Alaska east throughout most of Canada and south into the United States

as far south as Colorado in the Rocky Mountains and the southern Appalachians to northwest Alabama and to the central portions of Virginia and North Carolina in the South (Long 1974, Diersing 1980). Figure 2 depicts the distribution of the pygmy shrew in the South. Once regarded as extremely rare, it is now known to be distributed across a wide range of habitats (Laerm et al. 1999). Considerable information on the distribution, abundance, and habitat associations of this species is now available for Virginia (Pagels 1987, Handley 1991, Pagels et al. 1992, Kalko and Handley 1993, Mitchell et al. 1993, 1997; Linzey 1998, Bellows et al. 2001), Kentucky (Caldwell 1980, Caldwell and Bryan 1982, Bryan 1991, Meade 1992, Feldhamer et al. 1993, Kiser and Meade 1993), Tennessee (Kennedy et al. 1979, Kennedy and Harvey 1980, Tims et al. 1989, Harvey et al. 1991, 1992; Feldhamer et al. 1993, Linzey 1995), North Carolina (Smith et al. 1960, Lee et al. 1982, Webster 1987, Padgett and Rose 1994, Laerm et al. 1995a, 1999; Ford et al. 2000), South Carolina (Mengak et al. 1987, Laerm et al. 1999, 2000), Georgia (Wharton 1968, Ford et al. 1994, Laerm et al. 1995b, Ford et al. 1997, Laerm et al. 1999, 2000), and Alabama (Laerm et al. 1996). With the exception of the southeastern shrew, the pygmy shrew is the most widespread of the long-tailed soricids in the region.

ABUNDANCE STATUS

The pygmy shrew often is ubiquitous, but rarely comprises a significant component of the small mammal community. No quantitative data on population densities are available from the region. Recovery rates per 100 pitfall trapnight efforts in the southern Appalachians range from 0.1–0.8 individuals (Mengak et al. 1987, Harvey et al. 1991, Kalko and Handley 1993, Ford et al. 2006). About 4% of small mammals collected in the central Piedmont of Virginia were pygmy shrews (Pagels et al. 1992). Similarly, Feldhamer et al. (1993) reported that 2.2% of the small mammals surveyed in the Land Between the Lakes area in western Kentucky and Tennessee were pygmy shrews.

PRIMARY HABITATS

The pygmy shrew is known from virtually the entire spectrum of forest communities, seral stages, moisture regimes, and elevations in the region from high elevation red spruce–Fraser fir (*Picea rubens*–*Abies fraseri*) in the mountains, to bottomland gum–bald cypress (*Nyssa* spp.–*Taxodium distichum*) in low lying coastal areas (Padgett and Rose 1994, Ford et al. 2006). It also is reported from oldfields and young clearcuts. Pygmy shrews appear to prefer areas with canopy closure in the Coastal Plain, whereas in mesic cove

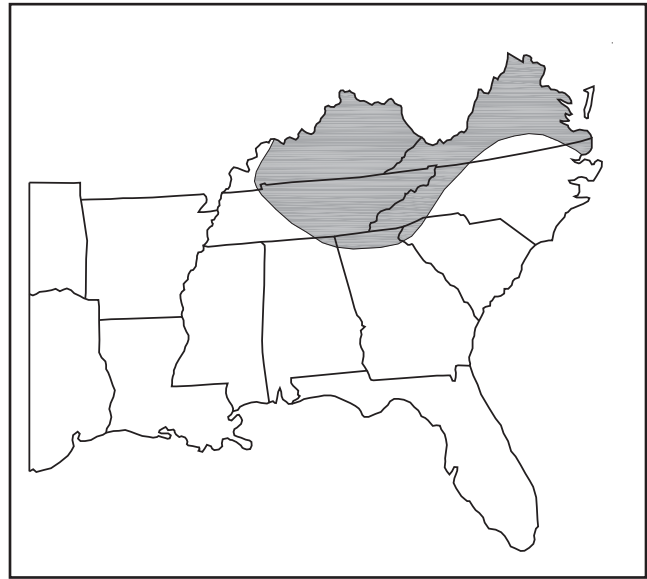


Figure 2. Distribution of *Sorex hoyi* in the South.

hardwoods in the southern Appalachians the opposite is true (Ford et al. 1997, Bellows et al. 2001). Similarly, substrate and understory associations also are varied, particularly its association with coarse woody debris (Bellows et al. 2001, Ford and Rodrigue 2001). Laerm et al. (1999, 2000) noted that it has the greatest niche breadth of all soricids in the southern Appalachians, although pygmy shrews may select or be competitively relegated to more xeric forest habitats where it is sympatric with masked shrews (Ford et al. 1994, 1997; Laerm et al. 1999).

REPRODUCTION

Long (1972, 1974, 1976) reviews the pertinent literature. Apparently, births of 3–7 young can occur throughout the year in the South, but reproduction is concentrated from November to March, with a peak in January (Feldhamer et al. 1993, McCay et al. 1998). In northern populations, births are reported from June through August with no evidence of a second breeding pulse. The gestation period is unknown. Adult size is reached in 8–12 weeks. Maximum longevity is approximately 70 weeks.

FOOD HABITS

Pygmy shrews are known to eat a wide variety of invertebrates including lepidopteran larvae, coleoptera, hymenoptera, and gastropods (Long 1974, Whitaker and French 1984, Ryan 1986, Whitaker and Cudmore 1987). Pygmy shrews concentrate primarily on invertebrate prey <10 mm in length (Churchfield 2002).

ASSOCIATED SPECIES

Depending on habitat, the pygmy shrew is associated with most insectivores and rodents that occur within its distribution. Ford et al. (2001) found pygmy shrews to be syntopic with the southeastern and masked shrews in the southern Appalachians at 53% and 68% of survey sites, respectively. Xeric forest sites where pygmy shrews and southeastern shrews occurred together often violate Fox and Kirkland's (1992) size-based assembly rules for shrews (Ford et al. 2006).

VULNERABILITY AND THREATS

Although the pygmy shrew is uncommon, there seem to be no obvious threats to its survival in the region.

MANAGEMENT SUGGESTIONS

The pygmy shrew appears tolerant of forest management activities such as regeneration harvest and prescribed burning (Ford et al. 1997, 1999, 2000; Bellows et al. 2001).

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Southeastern Shrew

Sorex longirostris (Bachman, 1837)

Joshua Laerm, W. Mark Ford, and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Three subspecies of the southeastern shrew (*Sorex longirostris*) are recognized: *S. l. longirostris*, *S. l. fisheri*, and *S. l. eionis*. All occur within the region. Literature on *S. longirostris* is reviewed by French (1980a,b).

DISTINGUISHING CHARACTERISTICS

The southeastern shrew is a small, long-tailed soricid with the following measurements: total length, 72–110 mm; tail, 24–37 mm; hind foot, 10–14 mm; weight, 3–6 g. The dental formula of the shrew is: I 3/1, C 1/1, P 3/1, M 3/3 = 32 (Figure 1). This species has small ears concealed in the pelage, minute eyes, and a long, pointed snout. The dorsal and ventral pelage is brown to reddish-brown, and the tail is indistinctly bicolored. The southeastern shrew may be confused with the masked shrew (*S. cinereus*) and pygmy shrew (*S. hoyi*). The pygmy shrew is smaller than the southeastern shrew and has minute third and fifth upper unicuspid (Junge and Hoffman 1981). The range of the masked shrew overlaps only in the southern Appalachians and north central Kentucky. Segregated along a habitat gradient, masked shrews are found in high elevation, mesic forest communities and southeastern shrews are restricted to low elevation, xeric forest communities and early successional stages (Ford et al. 2001, 2006). The two are distinguishable on the basis of bivariate comparisons of body and skull features (Junge and Hoffman 1981, Laerm et al. 1997).

CONSERVATION STATUS

The southeastern shrew has a global rank of Secure (NatureServe 2007). It is also considered Secure in Florida and Virginia. It is Apparently Secure in Alabama, Georgia, Kentucky, Mississippi, North Carolina, and Tennessee. Both Arkansas and Louisiana classify it as Imperiled. It is unranked in South Carolina. *Sorex l. fisheri* is listed as Threatened by the Virginia Department of Conservation and Recreation. The Florida Natural Areas Inventory considers *Sorex l. eionis* a Species of Special Concern. *Sorex l. longirostris* is monitored by the Arkansas and Mississippi Natural Heritage Programs, and is deemed In Need of Management by the Tennessee Department of Environment and Conservation.

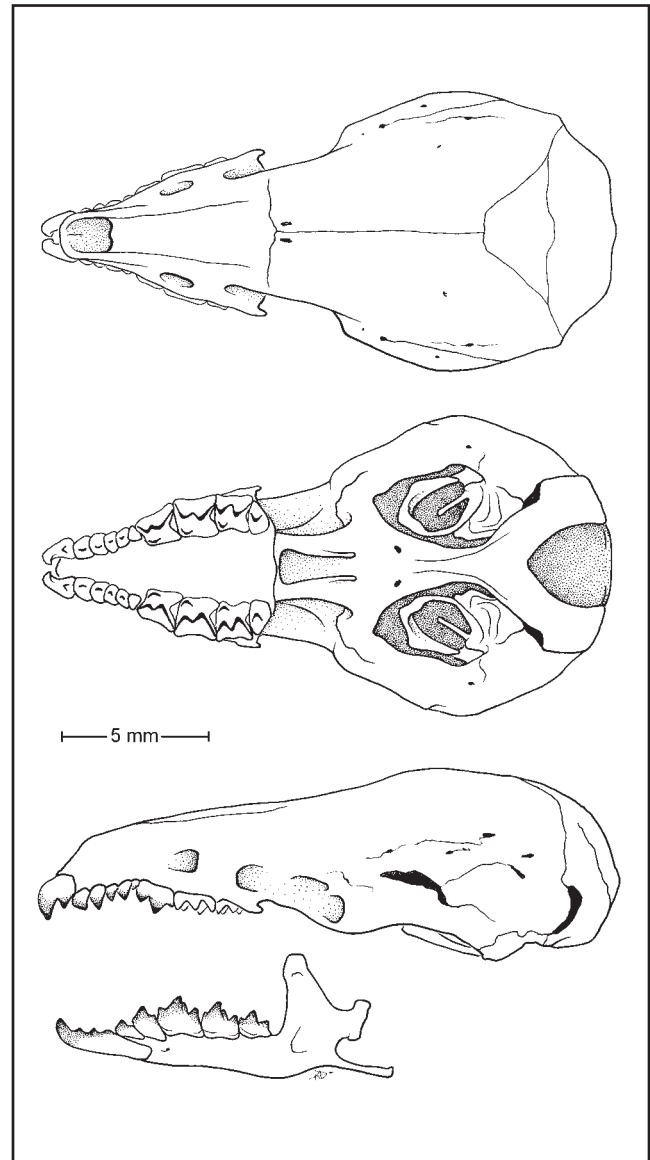


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sorex longirostris* from Fairfax County, Virginia (USNM 565900, gender unknown).

DISTRIBUTION

Sorex l. eionis is limited to the northern two-thirds of peninsular Florida (Moore 1944, Jones et al. 1991), but its overlap with *S. l. longirostris* is unknown (Figure 2). *Sorex l. fisheri* is restricted to the vicinity of the Great Dismal Swamp of Virginia and North Carolina

(Handley 1979, Pagels et al. 1982, Padgett et al. 1987, Rose et al. 1987, 1990; Webster 1987, Mitchell et al. 1993, Erdle and Pagels 1995). However, southeastern shrews 30 km beyond the Great Dismal Swamp are referable to as *S. l. longirostris*. Research is needed to define the range and degree of intergradation of *S. l. fisheri* with *S. l. longirostris* (Jones et al. 1991). The similarity of *S. l. longirostris* specimens from coastal South Carolina to specimens of *S. l. fisheri* also has been noted (Jones et al. 1991). *Sorex l. longirostris* is widely distributed, and its range includes most of Virginia (Pagels et al. 1982, Pagels and French 1987, Pagels and Handley 1989, Pagels et al. 1992, Linzey 1998, Bellows et al. 2001) and North Carolina (Engles 1941, Lee et al. 1982, Webster et al. 1984, Clark et al. 1985, Mitchell et al. 1995, Laerm et al. 1999, Ford et al. 2001). The southeastern shrew also occurs throughout South Carolina (Sanders 1978, Webster et al. 1985, Mengak et al. 1987, Cothran et al. 1991), all but the southeastern corner of Georgia (Golley 1962, Laerm et al. 1982, Ford et al. 1994, Laerm et al. 1999, Ford et al. 2001), the panhandle of Florida (Jones et al. 1991), all but coastal Mississippi (Cook 1942, Jones and Long 1961, Wolfe 1971, Kennedy et al. 1974, Wolfe and Esher 1981, Wolfe and Lohofener 1983, Jones and Carter 1989), throughout Tennessee (Goodpaster and Hoffmeister 1952, Tuttle 1964, Smith et al. 1974, Kennedy 1991, Harvey et al. 1991, 1992; Linzey 1995) and throughout Kentucky (Bryan 1991, Rose and Seegert 1982, Chadwick and Davis 1984, Meade 1992, Kiser and Meade 1993). It is found in most of Arkansas (Garland and Heidt 1989, Sealander and Heidt 1990), the Florida parishes of Louisiana (Lowery 1981, Constantine and Mitchell 1982, Jones et al. 1991), and extreme eastern Oklahoma (Taylor and Wilkinson 1988). Its possible occurrence in southeastern Arkansas and northeastern Louisiana is uncertain. The southeastern shrew does not occur above approximately 400 m elevation in northern Virginia grading higher to approximately 600 m in the extreme southern Appalachians (Pagels and Handley 1989, Ford et al. 2001, 2006).

ABUNDANCE STATUS

Historically, both *S. l. eionis* and *S. l. longirostris* were regarded as rare due to the inadequacy of conventional snap trap sampling. However, both subspecies now are considered common (George 1977, French 1980a,b; Wolfe and Esher 1981, Rose et al. 1987, Pagels and Handley 1989, Jones et al. 1991, Humphrey 1992, Feldhamer et al. 1993). French (1980a,b) reports densities at two Alabama study plots at 30–44 individuals/ha. *Sorex l. fisheri* may be locally abundant in portions of its restricted range (Rose et al. 1990).



Figure 2. Distribution of *Sorex longirostris* in the South: (1) *S. l. eionis*; (2) *S. l. fisheri*; (3) *S. l. longirostris*.

PRIMARY HABITATS

Jones et al. (1991) report that *S. l. eionis* occurs in bald cypress (*Taxodium distichum*) and bay swamps, hydric and xeric hammocks, slash pine (*Pinus elliotii*) flatwoods, longleaf pine (*P. palustris*) sandhills, sand pine (*P. clausa*) scrub, saw palmetto (*Serenoa repens*) thickets, and clearcuts. *Sorex l. fisheri* may be found in many habitat types, but it is most abundant in early to mid-successional disturbed woodlands with a dense understory, moderate leaf litter, and moist organic soils (Rose et al. 1990, Rose and Padgett 1991, Erdle and Pagels 1995). *Sorex l. longirostris* is known from a diversity of forested and non-forested habitats including oldfields, agricultural lands, dry upland hardwoods, pine forests, mixed pine-hardwoods, flatwoods, and the borders of moist or wet swamps, marshes, and rivers. In the Coastal Plain of Virginia, this subspecies prefers areas with heavy shrub cover (Bellows et al. 2001). It is associated primarily with heavy ground covers of grasses and sedges or moderate to heavy leaf litter where it nests under rotting logs or debris (Engles 1941, Cook 1942, Dusi 1959, Tuttle 1964, Negus and Dundee 1965, French 1980a, Rose 1980, Wolfe and Esher 1981, French 1984, Taylor and Wilkinson 1988, Jones et al. 1991, Laerm et al. 1997).

REPRODUCTION

French (1985) reported spring and fall peaks in reproduction with a reduction of breeding in summer. Pregnant females have been observed between March and October (Feldhamer et al. 1993). Litter size varies from 1–6, and 2 litters may be produced in a year

(French 1980a,b). Gestation length and lactation periods are unknown. Some individuals may reproduce in their first year, but most apparently overwinter before breeding (French 1980a, 1985). Maximum longevity is 70–90 weeks.

FOOD HABITS

The southeastern shrew eats arachnids, adult coleopterans, lepidopteran larvae, amphipods, isopods, chilopods, gastropods, and some vegetation (Whitaker and Mumford 1972, French 1980a,c; French 1984, Rose and Padgett 1991).

ASSOCIATED SPECIES

Depending on habitat, southeastern shrews associate with other insectivores such as the least shrew (*Cryptotis parva*), southern short-tailed shrew (*Blarina carolinensis*), and pygmy shrew. Ford et al. (2001) found that the southeastern shrew and the pygmy shrew were syntopic at 53% of sites surveyed in the foothills of the southern Appalachians and upper Piedmont. Rodent species that occupy the same habitats as the southeastern shrew include the white-footed mouse (*Peromyscus leucopus*), cotton mouse (*P. gossypinus*), oldfield mouse (*P. polionotus*), golden mouse (*Ochrotomys nuttalli*), eastern harvest mouse (*Reithrodontomys humulis*), hispid cotton rat (*Sigmodon hispidus*), marsh rice rat (*Oryzomys palustris*), and woodland vole (*Microtus pinetorum*).

VULNERABILITY AND THREATS

Sorex l. fisheri is restricted to areas in or around the Great Dismal Swamp, often at relatively low densities (Rose et al. 1990, Jones et al. 1991, Rose and Padgett 1991). Drainage of the Great Dismal Swamp and possible interbreeding by *S. l. longirostris* from surrounding areas are the primary threats to the subspecies (Rose et al. 1987, Rose and Padgett 1991). *Sorex l. eionis* is more abundant, less habitat specific, and more widespread than previously believed; it is not considered to be in danger (Jones et al. 1991, Humphrey 1992). *Sorex l. longirostris* is widely distributed, inhabits a variety of habitats, and is common to abundant. It is monitored by several states at the periphery of its range.

MANAGEMENT SUGGESTIONS

Maintenance of native vegetation and natural hydrologic regimes should be encouraged where *S. l. fisheri* occurs. The other subspecies are tolerant of most forest management activities, though *S. l. longirostris* can be displaced temporarily by least shrews in early successional habitat (Menzel et al. 2005).

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American Water Shrew

Sorex palustris (Richardson, 1858)

Joshua Laerm, W. Mark Ford, and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Ten subspecies of the American water shrew (*Sorex palustris*) are recognized (van Zyll de Jong 1983). Only *S. p. punctulatus* is found in the region (Hooper 1942). The literature on the species was reviewed by Beneski and Stinson (1987).

DISTINGUISHING CHARACTERISTICS

The American water shrew is a large, long-tailed soricid with a long, pointed snout. Its measurements are: total length, 130–156 mm; tail, 58–71 mm; hind foot, 18–20 mm; weight, 8–18 g. The dental formula of the American water shrew is: I 3/1, C 1/1, P 3/1, M 3/3 = 32 (Figure 1). The eyes and ears are small and concealed in the pelage. The pelage color is dark grayish-brown to black dorsally and light gray below, and can appear iridescent. The tail is distinctly bicolored, dark above and light below. Additionally, the hind feet of American water shrews are broad and fringed with stiff hairs. These characteristics make it unlikely that the water shrew would be confused with any other long-tailed shrew. Junge and Hoffman (1981) provide descriptive details. See keys for additional details.

CONSERVATION STATUS

The American water shrew has a global rank of Secure (NatureServe 2007). It is considered Imperiled in North Carolina and Tennessee, and Critically Imperiled in Georgia and Virginia. It is unranked in South Carolina. The Virginia Department of Conservation and Recreation lists *S. p. punctulatus* as Endangered. Kennedy and Harvey (1980), Linzey (1984, 1998), Webster (1987), Pagels and Handley (1991), and Laerm et al. (1995, 1999) provide substantive comments on the regional status of the species.

DISTRIBUTION

The American water shrew is associated closely with the Canadian and Hudsonian Life Zones. It is distributed throughout the boreal forest from Nova Scotia to Alaska and south through the Sierra Nevadas and Rockies in the West, and the Appalachians south to Tennessee, North Carolina, and Georgia in the east (Figure 2). It is restricted to the Blue Ridge and portions of the northern Ridge and Valley in a series of

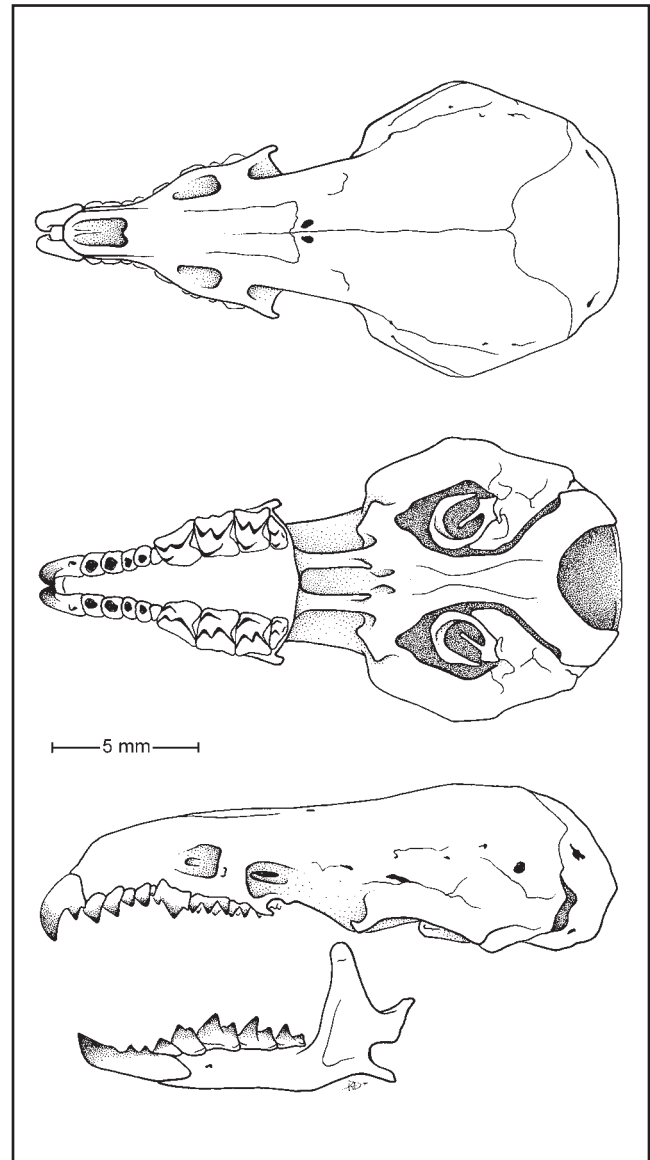


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sorex palustris* from Robinson Portage, Northwest Territory, Canada (USNM 107043, female).

apparently disjunct populations in Virginia (Pagels and Handley 1991, Linzey 1998, Pagels et al. 1998), Tennessee (Conaway and Pfitzer 1952, Kennedy and Harvey 1980, Linzey 1984, Harvey et al. 1991), North Carolina (Whitaker et al. 1975, Lee et al. 1982, Webster 1987, Laerm et al. 1999), and Georgia (Laerm et al.

1995). However, there is too little information available to adequately determine distribution extent or confirm the apparent disjunct nature of its populations.

ABUNDANCE STATUS

Collection records and existing literature indicate that the American water shrew may be extremely rare, although in Macon County, North Carolina, Laerm et al. (1999) recovered 10 individuals in 1,000 pitfall trapnights in three high elevation (> 1,300 m) first order streams. Because water shrews are very difficult to catch, it is possible that this species may be more widely distributed than present documentation suggests.

PRIMARY HABITAT

Beneski and Stinson (1987) reviewed the ecology and habitat associations of the American water shrew throughout its range. Hooper (1942), Conaway and Pfitzer (1952), Pagels and Tate (1976), Pagels and Handley (1991), Laerm et al. (1995, 1999), Pagels et al. (1998) and Ford et al. (2006) comment on habitat associations in the southern Appalachians. The American water shrew is found in association with high gradient, first and second order montane streams or seeps at high elevations where abundant cover from overhangs, rocks, roots, logs, and crevices exists (Pagels et al. 1998). In the Allegheny Mountains of West Virginia and Maryland, this species can occur in boggy habitats with little tree cover. In the southern Appalachians, the species is associated with riparian areas at medium to high elevation (900–1,800 m) in red spruce–Fraser fir (*Picea rubens*–*Abies fraseri*), northern hardwood, cove hardwood, and white pine–eastern hemlock (*Pinus strobus*–*Tsuga canadensis*) cover types, typically with dense rhododendron (*Rhododendron maximum*) understories. The American water shrew apparently has echolocation capabilities useful for orientation within the cluttered micro-environment where it exists (Buchler 1976).

REPRODUCTION

Conaway (1952) and Beneski and Stinson (1987) reviewed the information on reproduction of the American water shrew. Two or three litters with 4–7 young are produced annually. Gestation and lactation periods are not known, although they probably are similar to other shrews (approximately 21 days in most *Sorex*). Males and females usually are not active reproductively until they overwinter. The maximum life span is approximately 18 months.

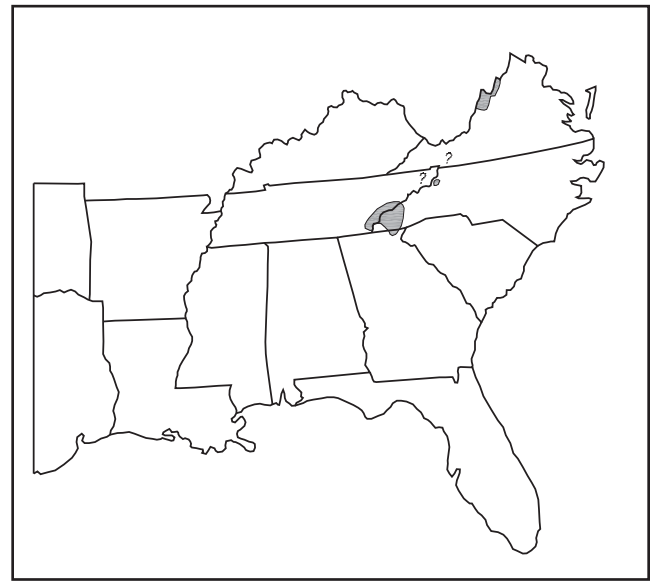


Figure 2. Distribution of *Sorex palustris* in the South.

FOOD HABITS

The food habits of the American water shrew were summarized by Beneski and Stinson (1987). This species primarily is insectivorous, feeding on both terrestrial and aquatic invertebrates such as larval plecoptera, ephemera, and trioptera (Linzey and Linzey 1973). It may also feed on fish and larval salamanders, but these items constitute a small component of the diet (Conaway 1952).

ASSOCIATED SPECIES

The American water shrew is associated with other high elevation forest insectivores in the southern Appalachians such as the northern short-tailed shrew (*Blarina brevicauda*), masked shrew (*S. cinereus*), smoky shrew (*S. fumeus*), and the hairy-tailed mole (*Parascalops breweri*). Rodents that occupy habitats associated with the water shrew include the deer mouse (*Peromyscus maniculatus*), southern red-backed vole (*Clethrionomys gapperi*), southern bog lemming (*Synaptomys cooperi*), and woodland jumping mouse (*Napaeozapus insignis*).

VULNERABILITY AND THREATS

Pagels and Handley (1991) suggest that the existing populations of the American water shrew in the southern and central Appalachians are Pleistocene relicts and populations have been small and scattered for most of the recent past. Water shrew distributions from Pennsylvania south may have been contracting for thousands of years in response to climate-driven

habitat changes, and this may be further accelerated by human-induced global climate change. Past land use practices in headwater streams used by this species probably have extirpated local populations, creating additional population fragmentation. Habitat degradation from logging, agriculture, and road construction negatively impact the riparian habitats favored by the American water shrew (Pagels et al. 1998).

MANAGEMENT SUGGESTIONS

Ford and Rodrigue (2001) suggest that streamside management zones with effective Best Management Practices are critical around riparian areas where American water shrews occur. Management impacts to water quality should be minimized or avoided. Additional surveys to adequately document American water shrew presence are needed in the southern Appalachians.

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American Water Shrew (*Sorex palustris*)

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Star-nosed Mole

Condylura cristata (Linnaeus, 1758)

Joshua Laerm, Brian R. Chapman, and W. Mark Ford

CONTENT AND TAXONOMIC COMMENTS

The star-nosed mole (*Condylura cristata*) includes two subspecies, *C. c. cristata* and *C. c. parva* (Yates 1978, Peterson and Yates 1980). The distribution of the subspecies in the region is confused. Paradiso (1959) defined the range of *C. c. cristata* as extending north from a line through central Virginia and southern West Virginia, and the range of *C. c. parva* was from south of that line. Biologists in the region (Handley and Gordon 1979, Webster 1987, Handley and Pagels 1991) followed this arrangement, but Peterson and Yates (1980) expanded the northern boundary of *C. c. parva* northward well into the Great Lakes and New England. Accordingly, all populations in the region are believed to be *C. c. parva*. The literature on the species is reviewed by Peterson and Yates (1980).

DISTINGUISHING CHARACTERISTICS

Condylura cristata is a medium-sized mole with a robust body. Its measurements are: total length, 153–238 mm; tail, 51–92 mm; hind foot, 23–32 mm; weight 35–70 g. The dental formula is: I 3/3, C 1/1, P 4/4, M 3/3 = 44 (Figure 1). The pinnae are absent, and the eyes are small and partially concealed in the pelage. The pelage is dense, soft, and silky, and is blackish-brown to black dorsally and only slightly paler ventrally. The tail is 1/3 to 1/2 of the body length and constricted at the base. The rostrum, the most distinctive feature of the species, has 22 fleshy appendages at the tip. It is unlikely that the star-nosed mole would be confused with any other mammals in the region. See keys for additional details.

CONSERVATION STATUS

The star-nosed mole has a global rank of Secure (NatureServe 2007). It is Apparently Secure in North Carolina and Virginia, but Vulnerable in South Carolina. Both Georgia and Tennessee classify the star-nosed mole as Imperiled. It is unranked in Florida.

DISTRIBUTION

The star-nosed mole is distributed throughout most of eastern Canada and the northeastern United States (Hall 1981). Figure 2 depicts the distribution of the star-nosed mole in the South. Lee (1987) provides a review of regional distribution records. The star-nosed

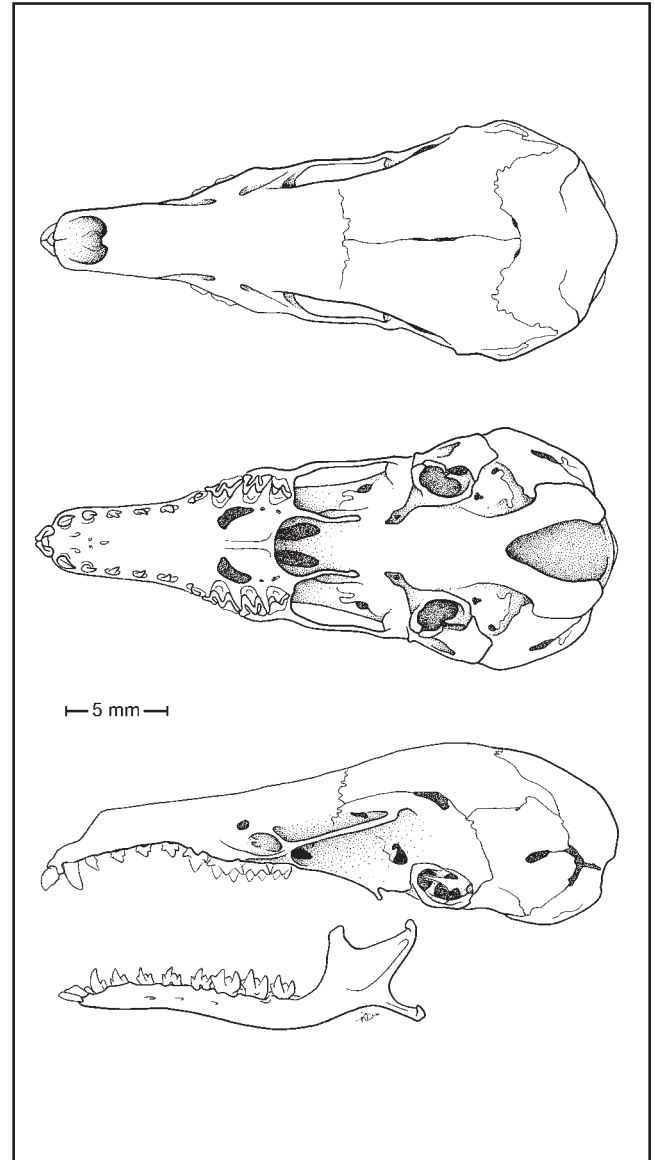


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Condylura cristata* from Fauquier County, Virginia (USNM 566403, gender unknown).

mole is known from scattered records in Virginia, but apparently is absent from the south-central portion of the state (Paradiso 1959, Handley 1979, Handley and Pagels 1991, Pagels et al. 1992, Linzey 1998, Bellows et al. 1999). From central Virginia south, populations appear to be restricted to either the Coastal Plain or the Blue Ridge and upper

Piedmont of North Carolina (Odom 1949, Johnston 1967, Lee et al. 1982, Clark et al. 1985, Webster 1987, Beane 1995), South Carolina (Grimm 1955, Cothran et al. 1991, Laerm et al. 1996, McCay et al. 1999), and Georgia (Harper 1927, Golley 1962, Laerm 1981, Laerm et al. 1982, Laerm 1992, Laerm et al. 1996). The star-nosed mole similarly is restricted to the Blue Ridge of Tennessee (Kellogg 1939, Smith et al. 1974, Kennedy and Harvey 1980, Harvey et al. 1992), and known from one extralimital record from northwestern Kentucky (R. Morton, Kentucky Department of Fish and Wildlife Resources, personal communication).

ABUNDANCE STATUS

The status of the star-nosed mole is uncertain. Most regional accounts indicate that it is rare to uncommon, though it may be locally abundant (Webster 1987). No population estimates are available for the species in the region. In the northern portion of its range, the star-nosed mole can reach 41/ha (van Zyll de Jong 1983).

PRIMARY HABITATS

The star-nosed mole is typically associated with moist, swampy habitats such as fens, bogs, seeps, and streambanks with heavy leaf litter in both forested and early successional communities. In the southern Appalachians, it is found in mesic forest cover types including northern hardwood, cove hardwood, and white pine-eastern hemlock (*Pinus strobus*-*Tsuga canadensis*) communities around streams and low wet sites. In the Coastal Plain, it generally occurs in saturated wetlands, pocosins, and forested floodplain cover types, although it also has been reported in an upland longleaf pine (*P. palustris*) habitat (McCay et al. 1999). The star-nosed mole constructs burrows and short surface runways, typically around coarse woody debris, tree roots, and rocks near water. These runways may open directly into aquatic habitats. Nests are constructed in the burrows above water level.

REPRODUCTION

Breeding in the star-nosed mole occurs from March to August with a peak in April or May (Peterson and Yates 1980). Gestation is about 45 days (Jackson 1961); a single litter is produced each year (Eadie and Hamilton 1956). The litter size ranges from 3–7 (Eadie and Hamilton 1956, Davis and Peek 1970). Individuals reproduce after their first winter.

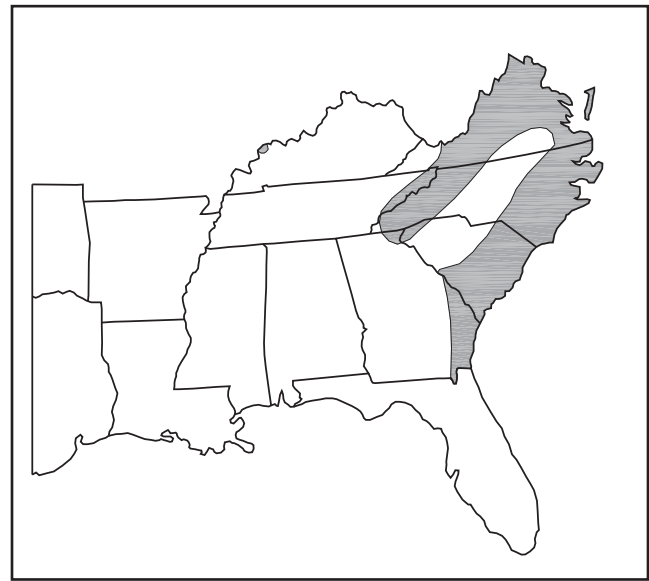


Figure 2. Distribution of *Condylura cristata* in the South.

FOOD HABITS

The nasal star is an electro-sensory device that is used to detect prey (Gould et al. 1993), best described as a “tactile” eye (Catania 1999), in leaf litter, underground burrows, and even water. Primary foods consist of terrestrial and aquatic annelids and insects, although crustaceans, mollusks, and fish also are eaten (Hamilton 1931, Rust 1966, Whitaker and Mumford 1972).

ASSOCIATED SPECIES

The star-nosed mole is associated with the hairy-tailed mole (*Parascalops breweri*), woodland jumping mouse (*Napaeozapus insignis*), and southern bog lemming (*Synaptomys cooperi*) in the Appalachian Mountains, and the eastern mole (*Scalopus aquaticus*) and rice rat (*Oryzomys palustris*) in the Coastal Plain.

VULNERABILITY AND THREATS

Little information is available regarding the abundance and life history traits of the star-nosed mole in the region. Because the species appears to be closely associated with moist habitats and friable soils, land management practices that impact aquatic ecosystems through draining, ditching, and conversion to agricultural or industrial uses probably are detrimental. As a result, the known geographic range appears fragmented and the species frequently is absent in otherwise appropriate habitat. Surveys to determine if this species occurs in the Cumberland Mountains and Plateau of Kentucky and Tennessee are warranted.

MANAGEMENT SUGGESTIONS

Activities that substantially alter natural hydrologic regimes should be avoided. The impact of locally raised water tables following timber harvest should be investigated.

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Hairy-tailed Mole

Parascalops breweri (Bachman, 1842)

Joshua Laerm, W. Mark Ford, and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The hairy-tailed mole (*Parascalops breweri*) is a monotypic species. Literature on the hairy-tailed mole was reviewed by Hallett (1978).

DISTINGUISHING CHARACTERISTICS

The hairy-tailed mole has a robust body. Its measurements are: total length, 139–174 mm; tail, 23–36 mm; hind foot, 17–21 mm; weight, 40–65 g. The dense, soft pelage is gray to black dorsally, but slightly paler ventrally. The eyes are concealed in the pelage and the ears lack pinnae. The short tail (less than 25% of the body length) is fleshy, constricted at the base, and densely furred with coarse hairs. The forefeet are not webbed. The hairy-tailed mole is unlikely to be confused with the eastern mole (*Scalopus aquaticus*), which has a naked tail, or with the star-nosed mole (*Condylura cristata*), that has numerous fleshy rostral appendages. The dental formula of hairy-tailed mole is: I 3/3, C 1/1, P 4/4, M 3/3 = 44 (Figure 1). See keys for additional details.

CONSERVATION STATUS

The hairy-tailed mole has a global rank of Secure (NatureServe 2007). It is Secure in Virginia, and Apparently Secure in Kentucky and North Carolina. Tennessee classifies it as Vulnerable. It is Critically Imperiled in Georgia. It is currently unranked in South Carolina, where it has been monitored as a Species of Special Concern.

DISTRIBUTION

The hairy-tailed mole is distributed from Ontario and Quebec (van Zyll de Jong 1983, Burns 1983) throughout the northeastern United States and south throughout the Appalachian Mountains. The distribution of the mole in the South is depicted in Figure 2. Its geographical range includes the Southern Appalachian Mountains of Virginia (Odom 1944, Handley 1971, Pagels and Tate 1976, Handley 1992, Linzey 1998), Kentucky (Welter and Sollberger 1939, Wallace and Houp 1968, Barbour and Davis 1974, Fassler 1974, Meade 1992), Tennessee (Smith et al. 1974, Copeland 1981, Allsbrooks et al. 1983, Linzey 1995), North Carolina (Gordon and Bailey 1963, Johnston 1967, Lee et al. 1982, Webster et al. 1984, Linzey 1995),

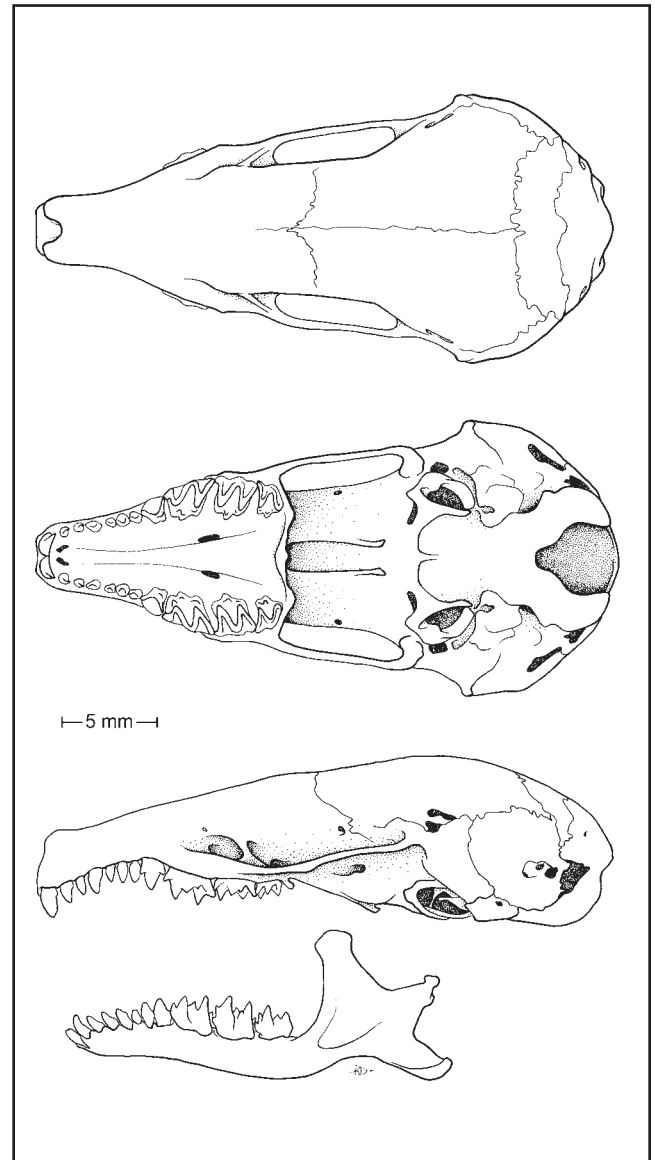


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Parascalops breweri* from Giles County, Virginia (USNM 364611, male).

South Carolina (Reese and Luckett 1979) and Georgia (Laerm 1992, Brown 1993).

ABUNDANCE STATUS

In the South, the hairy-tailed mole varies in its abundance where found. Average population density estimates reach 25–30/ha in New Hampshire (Eadie 1939)

and New York (Hamilton 1939). It is abundant at Mountain Lake Biological Station, Giles County, Virginia in the Ridge and Valley (C. O. Handley, Smithsonian Institution, personal communication). In the extreme southern Appalachians, it is rarely reported even in appropriate habitat. Consequently, density estimates are unavailable and its status is uncertain in the southernmost areas of its range.

PRIMARY HABITATS

Throughout its distribution, the hairy-tailed mole constructs tunnels for foraging and nesting (Hallett 1978). It occurs in deciduous and conifer forests, oldfields, pastures, and grassy roadsides in substrates composed of light, well-drained soils with substantial surface cover. This species avoids soils with high clay or moisture content (Eadie 1939). It occurs in red spruce–Fraser fir (*Picea rubens*–*Abies fraseri*), northern hardwoods, cove hardwoods, and white pine–eastern hemlock (*Pinus strobus*–*Tsuga canadensis*). Handley and Patton (1947) report that it is abundant above 900 m in northern hardwood forest habitats in Virginia. In the Great Smoky Mountains, the hairy-tailed mole is found between 450–1950 m. In western North Carolina and eastern Tennessee, it has been found in sheltered areas with dense rhododendron (*Rhododendron maximum*; Komarek and Komarek 1938, Johnston 1967). An individual was recovered at 800 m in a mesic hardwood community in northern Georgia (Laerm 1992) and a single specimen was collected at a similar elevation in a mixed pine-hardwood community with fine, loamy soil in the upstate of South Carolina (Reese and Luckett 1979).

REPRODUCTION

The breeding season is typically March through April, and it apparently does not extend into summer. The gestation period is 4–6 weeks. A single litter is produced annually and the normal litter size is 4–5 young (Eadie 1939), although litter sizes of up to 8 are reported (Richmond and Roslund 1949). Individuals become sexually mature in their second year. Longevity in the wild is approximately 4 years (Eadie 1939).

FOOD HABITS

The foods of the hairy-tailed mole include adult and larval insects, annelids, chilopods, gastropods, diplopods, and rootlets (Eadie 1939, Hallett 1978, Hamilton 1941).

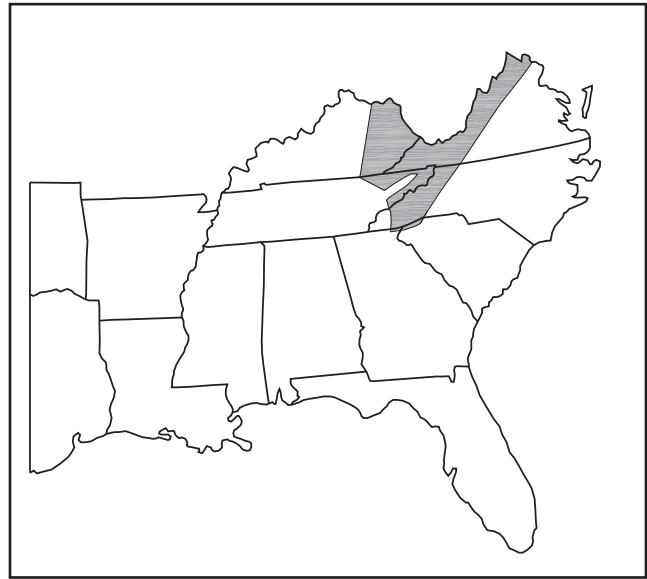


Figure 2. Distribution of *Parascalops breweri* in the South.

ASSOCIATED SPECIES

In the South, the hairy-tailed mole commonly occupies the same montane habitats as the masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), northern short-tailed shrew (*Blarina brevicauda*), deer mouse (*Peromyscus maniculatus*), southern red-backed vole (*Clethrionomys gapperi*), woodland vole (*Microtus pinetorum*), southern bog lemming (*Synaptomys cooperi*), and woodland jumping mouse (*Napaeozapus insignis*). It often is found in the same habitats as the star-nosed mole, but it apparently does not occur in association with the eastern mole (Hallett 1978). Shrews, particularly the northern short-tailed shrew, undoubtedly use tunnels and burrows constructed by the hairy-tailed mole.

VULNERABILITY AND THREATS

The hairy-tailed mole is widely distributed and common to abundant in appropriate habitats throughout most of its range. Its status in the southern Appalachians is less certain, but significant areas of appropriate habitat exist and there appear to be no known threats to the species.

MANAGEMENT SUGGESTIONS

Activities that permanently saturate the soil with water probably are detrimental. Surveys to ascertain the species full distribution south of the Great Smoky Mountains are needed.

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Eastern Mole

Scalopus aquaticus (Linnaeus, 1758)

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CONTENT AND TAXONOMIC COMMENTS

Sixteen subspecies of the eastern mole (*Scalopus aquaticus*) currently are recognized. Eleven subspecies occur in the region: *S. a. aereus*, *S. a. anastasae*, *S. a. aquaticus*, *S. a. australis*, *S. a. bassi*, *S. a. howelli*, *S. a. machrinoides*, *S. a. machrinus*, *S. a. nanus*, *S. a. parvus*, and *S. a. porteri* (Yates and Schmidly 1977, Yates and Schmidly 1978, Hall 1981). Yates (1978) examined the taxonomic relationships of populations of the eastern mole and questioned the validity of several regional subspecies. However, the conclusions of Yates (1978) were not followed by Yates and Schmidly (1978) or Hall (1981). The taxonomic relationships of populations west of the Mississippi River were revised by Yates and Schmidly (1977), but the eastern subspecies have not been revised since Jackson (1914, 1915) and subsequent descriptions of new subspecies have not been made (Howell 1939, Schwartz 1952a). Yates and Schmidly (1978) reviewed the literature on the eastern mole.

DISTINGUISHING CHARACTERISTICS

The eastern mole is a medium-sized mole with a robust body. Measurements are: total length, 129–208 mm; tail, 18–38 mm; hind foot, 15–22 mm; weight 65–115 g. Body size decreases from northern to southern latitudes, and males typically are larger than females. Like other moles, the eastern mole lacks ear pinnae and the eyes are concealed by a thin layer of skin. The dense, silky pelage varies in color; individuals are silver-gray, brown, or black dorsally and slightly paler below. Piebald specimens with white spots, particularly on the head, sometimes are seen. The tail is short (less than 1/6 of the body length) and either lacks hair or is sparsely haired. The forefeet of the eastern mole are greatly enlarged and webbed. *Scalopus aquaticus* is unlikely to be confused with the star-nosed mole (*Condylura cristata*) because of the latter's fleshy appendages around the nose, or the hairy-tailed mole (*Parascalops breweri*) which has a longer, hairy tail. The dental formula of the eastern mole is: $I\ 3/2, C\ 1/0, P\ 3/3, M\ 3/3 = 36$ (Figure 1). See keys for additional details.

CONSERVATION STATUS

The eastern mole has a global rank of Secure (NatureServe 2007). The species is also considered

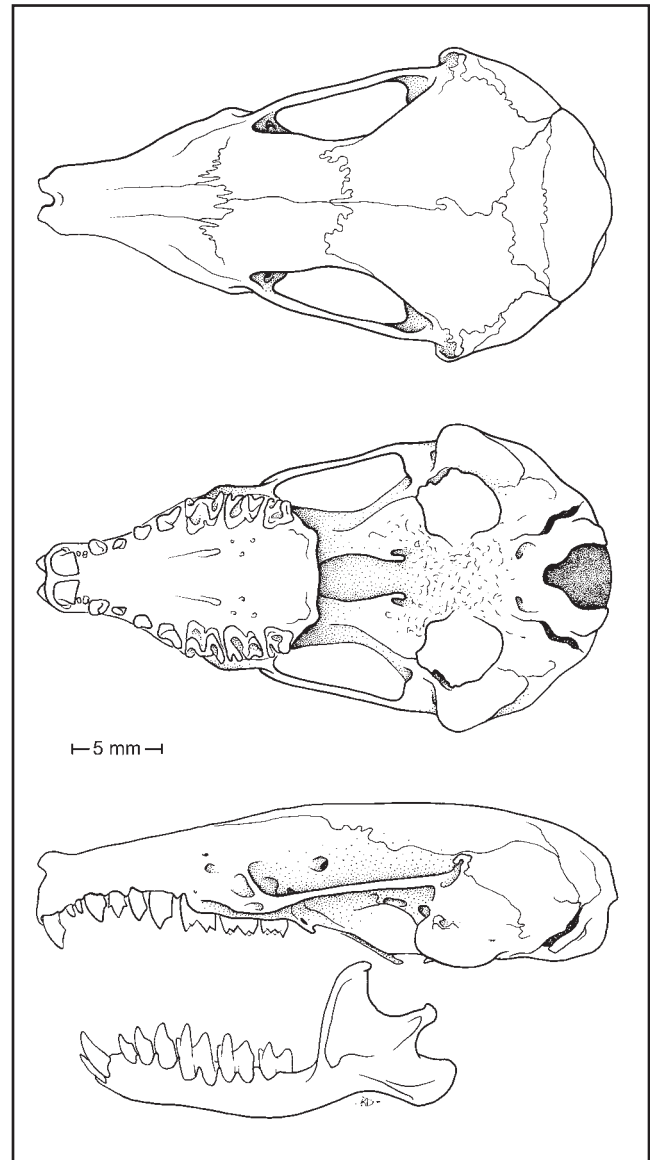


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Scalopus aquaticus* from Allegheny County, Maryland (USNM 506896, male).

Secure in those states where it occurs within the region except for Arkansas and Louisiana, where it is Apparently Secure. It is unranked in Florida and South Carolina.

DISTRIBUTION

The eastern mole has the most widespread geographic distribution of any North American mole. It ranges from the Great Lakes region and southern New England south to Florida and west through the Great Plains into Texas and northern Mexico. It occurs region-wide in areas with suitable soil types, though it is locally absent in portions of the southern Appalachians (Figure 2). It occurs throughout Virginia (Bailey 1946, Handley and Patton 1947, Jackson et al. 1976, Handley 1979, 1992; Webster et al. 1985, Pagels et al. 1992, Linzey 1998), North Carolina (Odom 1949, Johnston 1967, Lee et al. 1982, Clark et al. 1985, Webster et al. 1985, Webster 1988, Linzey 1995), South Carolina (Coleman 1941, Golley 1966, Sanders 1978, Schlacher and Pelton 1979, Webster et al. 1985, Cothran et al. 1991, Hartman and Krenz 1993, Hartman 1995, 1996; Hartman et al. 2000, 2001), and Georgia (Howell 1909, Harper 1927, Golley 1962, Neuhauser and Baker 1974, Wharton et al. 1981, Laerm et al. 1982, 1999; Ford et al. 1994) including many of the barrier islands of these states. Its range includes most of Florida except the Everglades (Hamilton 1941, Rand and Host 1942, Moore 1946, 1949; Schwartz 1952*a,b*; Pournelle and Barrington 1953, Ivey 1959, Layne 1974), Alabama (Howell 1921, Holliman 1963, Linzey 1970), Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989), and all but the coastal regions of Louisiana (Lowery 1974) and eastern Texas (Yates and Schmidly 1977, Schmidly 1983, Davis and Schmidly 1994). It is distributed throughout Tennessee (Calhoun 1941, Goodpaster and Hoffmeister 1952, Linzey and Linzey 1971, Smith et al. 1974, Kennedy 1991, Linzey 1995), Kentucky (Barbour and Davis 1974, Davis and Barbour 1979), Arkansas (Sealander and Heidt 1990), and eastern Oklahoma (Caire et al. 1989).

ABUNDANCE STATUS

Few population density estimates are available. Hartman and Krentz (1993) reported 1.3–3.0/ha in South Carolina. Many researchers indicate that it is locally abundant within the region (Golley 1962, Barbour and Davis 1974, Lowery 1974, Schmidly 1983, Webster et al. 1985, Caire et al. 1989, Sealander and Heidt 1990). Handley and Patton (1947) and Webster et al. (1985) suggest that it is uncommon at higher elevations of the Appalachians; however, there are records of the eastern mole at elevations up to 1375 m in Georgia (Howell 1909) and 1250 m in North Carolina (Johnston 1967). The species is common at lower elevations in the Appalachians (Linzey and Linzey 1971, Webster et al. 1985). Although Lowery (1974) and Schmidly (1983) indicate that *S. aquaticus* is absent from coastal areas of Louisiana and eastern

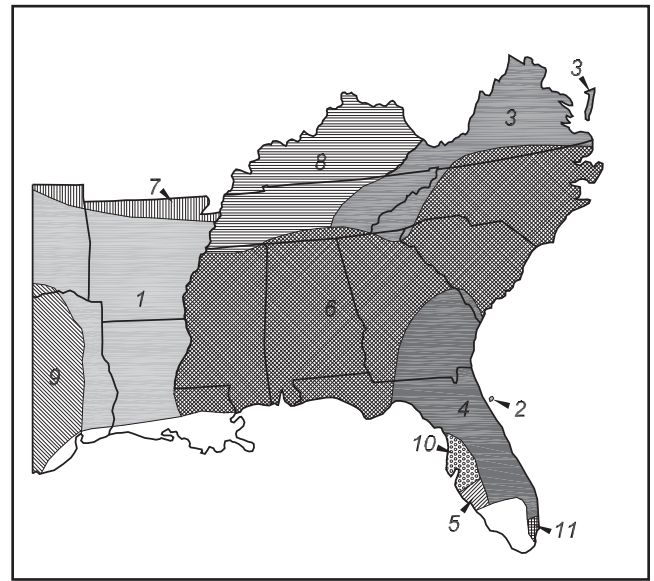


Figure 2. Distribution of *Scalopus aquaticus* in the South: (1) *S. a. aereus*; (2) *S. a. anastasiae*; (3) *S. a. aquaticus*; (4) *S. a. australis*; (5) *S. a. bassi*; (6) *S. a. howelli*; (7) *S. a. machrinoides*; (8) *S. a. machrinus*; (9) *S. a. nanus*; (10) *S. a. parvus*; (11) *S. a. porteri*.

Texas, it is common in coastal regions and barrier islands in Florida, Georgia, and the Carolinas (Bangs 1898, Golley 1962, Neuhauser and Baker 1974, Sanders 1978, Schlacher and Pelton 1979, Webster et al. 1985, Laerm et al. 1999, Hartman et al. 2000, 2001). Though absent from the Everglades, it is known from other major wetlands such as the Okefenokee Swamp (Harper 1927) and Great Dismal Swamp (Handley 1979).

PRIMARY HABITATS

The mole is fossorial, occurring in habitats with moist, loose, loamy, or sandy soil, but it is rare or absent in soils composed of heavily compacted clay or large amounts of stone or gravel (Jackson 1915, Arlton 1936, Davis 1942). Exceedingly dry or wet soils are avoided (Davis 1942, Yates and Schmidly 1978). Rivers and streams are not barriers to dispersal because the eastern mole is capable of swimming (Yates and Schmidly 1977). It occurs in many vegetation communities including residential gardens, lawns, grasslands, oldfields, and essentially all forest types in the region.

REPRODUCTION

Reproductive activities begin as early as January and February in Louisiana and Texas (Lowery 1974, Yates and Schmidly 1977), but are concentrated from late March to early May farther north (Conaway 1959).

Females breed in their second year (Conaway 1959), and a single litter of 2–5 young is produced. Mean survival is 2 years (Hartman 1995).

FOOD HABITS

The eastern mole feeds on annelids, coleopteran larvae, hymenopterans, and other invertebrates (Hisaw 1923, Calhoun 1941, Whitaker and Schmeltz 1974, Hartman et al. 2000), in addition to vertebrates (Christian 1950) and vegetable matter (Hisaw 1923, Hartman et al. 2000). The diet varies according to habitat type and prey availability. Comparison of food habits between the Coastal Plain of South Carolina and Indiana showed that earthworms comprised 8.3% of the diet of eastern moles in South Carolina versus 87.8% in Indiana (Hartman et al. 2000).

ASSOCIATED SPECIES

The eastern mole is widely distributed and occurs in such a variety of habitats that it is a component of most small mammal communities. The association of this species with *Blarina* spp. is especially important, as shrews commonly utilize mole tunnels as travel corridors.

VULNERABILITY AND THREATS

There appear to be no threats to the survival of the mole. *Scalopus aquaticus anastasae* and *S. a. bassi* are known only from type localities in Florida; these limited ranges present unique conservation problems.

MANAGEMENT SUGGESTIONS

The taxonomic distinctiveness and conservation status of the subspecies populations of the eastern mole should be addressed.

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Wagner's Bonneted Bat

Eumops glaucinus (Wagner, 1843)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Wagner's bonneted bat (*Eumops glaucinus*) originally was described as *Dysopes glaucinus* by Wagner (Koopman 1993). Allen (1932) described a new species, *Molossides floridanus*, on the basis of a fossil specimen from a Pleistocene deposit and this became the type specimen for the current subspecies when Koopman (1971) revised genus *Eumops*. Four subspecies are recognized; only *E. g. floridanus* occurs in the United States (Koopman 1971, Eger 1977, Hall 1981). This subspecies is often called the Florida mastiff bat (Owre 1978, Humphrey 1992).

DISTINGUISHING CHARACTERISTICS

Wagner's bonneted bat is a free-tailed bat with large ears. There are few published measurements for this species; the forearm length is known to average 57–66 mm (Barbour and Davis 1969). The glossy, thick pelage is usually black but individual hairs are sharply bicolor. The body hairs are white at the base, which sometimes makes the bat appear grayish. The ears of Wagner's bonneted bat are joined at the base where they meet above the face. The wings are long and narrow (wingspan approximately 470 mm) and the posterior half of the tail extends beyond the margin of the interfemoral membrane. The only other free-tailed bat in the region is the Brazilian free-tailed bat (*Tadarida brasiliensis*), a much smaller species. The forearm length of *T. brasiliensis* is less than 50 mm and its ears are not united at the base. The dental formula is I 1 1/2, C 1 1/1, P 2 2/2, M 3 3/3 = 30 (Figure 1).

CONSERVATION STATUS

Wagner's bonneted bat has a global rank of Secure (NatureServe 2007). It is Critically Imperiled in Florida.

DISTRIBUTION

Wagner's bonneted bat occurs in the West Indies, Central and South America, and the southern portion of the Florida peninsula (Barbour and Davis 1969, Hall 1981). Several records for the bat are from the Miami area (Barbour 1936, Eger 1977, Owre 1978). Most of the Florida specimen records date from 1955 to 1967 (Owre 1978). Belwood (1981) reported finding a small colony of *E. g. floridanus* near Punta Gorda, Charlotte County in 1979. This report, the first record

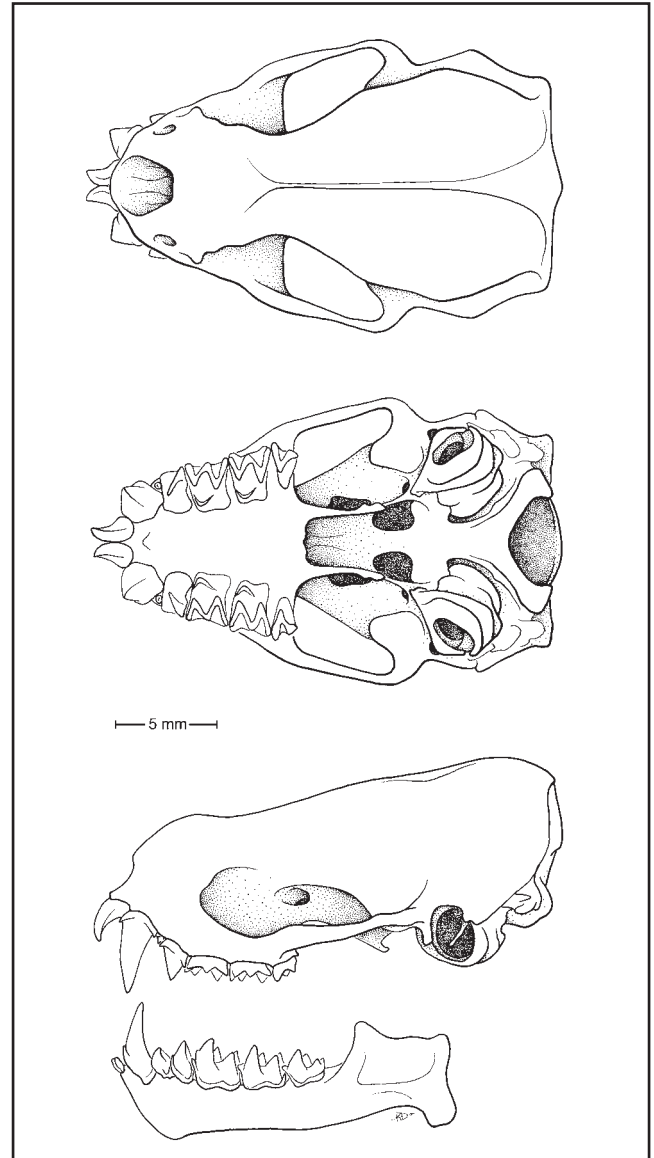


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Eumops glaucinus* from Jamaica (USNM 399618, gender unknown).

of the species in Florida since 1967, extended the known distributional range of the species 200 km westward and confirmed the existence of the species in the state (Figure 2). Robson et al. (1989) found an additional specimen of the Wagner's bonneted bat in Coral Gables in 1988.

ABUNDANCE STATUS

Jennings (1958) reported that 20 specimens of *E. g. floridanus* were known when he surveyed the bats of Florida. Based upon those numbers, Jennings speculated that the bat was probably common on the eastern coast of southern Florida. However, the Florida mastiff bat has been reported only twice since 1967. In both instances, pregnant females were reported (Belwood 1981, Robson et al. 1989) indicating that a reproductive population exists in southern Florida. The density of the current population of *E. g. floridanus* is not known.

PRIMARY HABITATS

In the Miami area, most *E. g. floridanus* are found roosting under shingles on buildings with Spanish tile roofs (Barbour and Davis 1969). Individuals also are found inside buildings (Robson et al. 1989) and in low shrubbery near buildings (Jennings 1958). Recently, a group of eight individuals was recovered from a cavity in longleaf pine (*Pinus palustris*) located in pine flatwoods habitat (Belwood 1981). The cavity, which was approximately 4.6 m high, was excavated originally by a red-cockaded woodpecker (*Picoides borealis*) and enlarged by a pileated woodpecker (*Dryocopus pileatus*). Woodpecker cavities (Silva-Taboada 1979) and buildings (Tyson 1964) also are used as roosting locations by the Cuban subspecies, *E. g. glaucinus*.

REPRODUCTION

Members of the Cuban subspecies, *E. g. glaucinus*, are polyestrous and give birth to a single young (Silva-Taboada 1979). Polyestry also may occur in the Florida subspecies. Young Wagner's bonneted bats have been reported in June (Humphrey 1992), August (Robson et al. 1989), and September (Belwood 1981). Furthermore, of the six female bats examined by Belwood (1981) in early September, five were post-lactating and one was pregnant. The sex ratio of the group found by Belwood (1981) suggests that the bats represented a male and a harem. Harem behavior has been described in other bats, including the molossid, *Tadarida midas* (Bradbury 1977). This type of behavior may be facilitated by the use of tree cavities, which can be defended against other males (Morrison 1979).

FOOD HABITS

The food habits of Wagner's bonneted bat have been poorly studied. Belwood (1981) recovered the fecal material from the bottom of a tree cavity roost used

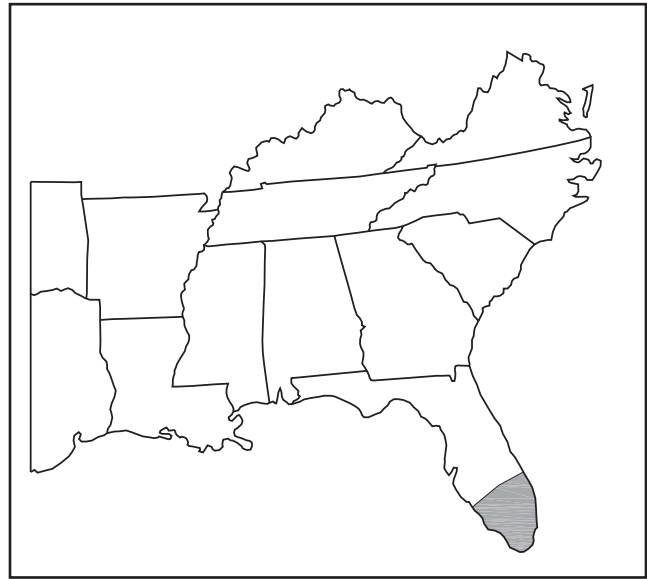


Figure 2. Distribution of *Eumops glaucinus* in the South.

by *E. g. floridanus* in southwestern Florida. The primary insects found were (by volume): Coleoptera (55%), Diptera (15%), and Hemiptera (10%). In Cuba, Coleoptera, Lepidoptera and Orthoptera were the main dietary items of *E. g. glaucinus* (Silva-Taboada 1979).

ASSOCIATED SPECIES

Eumops g. floridanus does not share communal roosts with other species. Because it uses buildings and tree cavities as roost sites, it may associate with species having similar roost preferences.

VULNERABILITY AND THREATS

The Wagner's bonneted bat is vulnerable to the destruction of roost sites in both urban and forested areas. In forested areas, efforts should be made to prevent the elimination of older trees with cavities. It may be influenced by pesticide application for mosquito control (Humphrey 1992).

MANAGEMENT SUGGESTIONS

Management activities focus on the provision of large, old trees and the maintenance of snags as roosting cavities (Bat Conservation International 2001). Additional research is needed on foraging and roosting requirements, and the impact of environmental contaminants.

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Brazilian Free-tailed Bat

Tadarida brasiliensis (Geoffrey Saint-Hilaire, 1824)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The Brazilian free-tailed bat (*Tadarida brasiliensis*) was described originally in 1824 by Geoffrey Saint-Hilaire as *Nyctinomus brasiliensis* (Wilkins 1989). Lyon (1914) suggested the use of *Tadarida* as a substitute for *Nyctinomus*; subsequent authors have followed that suggestion. The taxonomic history of *T. brasiliensis* is described by Owen et al. (1990). Nine subspecies were recognized by Schwartz (1955) and were verified by Freeman's (1981) morphometric comparison of 78 molossid species. One subspecies, (*T. b. cynocephala*), occurs within the South; however, the range of the western *T. b. mexicana* may overlap seasonally with *T. b. cynocephala* in eastern Oklahoma, eastern Texas, and Louisiana (Carter 1962, Spenrath and LaVal 1974, Hall 1981, Glass 1982). Because the eastern subspecies is a permanent resident throughout the South, the latter is considered in this account unless otherwise stated.

Carter (1962) provided considerable morphological, physiological, distributional, ecological, and behavioral evidence to suggest that the taxon represented by *T. b. cynocephala* should be elevated to the status of a separate species of *Tadarida*. Owen et al. (1990) evaluated this possibility, but followed the opinion of Wilkins (1989) who suggested that no taxonomic change occur until additional evidence is available. The Brazilian free-tailed bat also is known as the guano bat and the Mexican free-tailed bat, although these vernacular names are usually reserved for *T. b. mexicana* in the western United States. The south-eastern species is sometimes called Le Conte's free-tailed bat.

DISTINGUISHING CHARACTERISTICS

Tadarida brasiliensis is a medium-sized, dark brown bat that is easily recognized because the tail extends well beyond the uropatagial membrane. Measurements are: total length, 92–105 mm; tail, 30–42 mm; hind foot, 7–10 mm; ear, 12–14 mm; forearm, 30–37 mm; weight, 7–14 g. The dense fur of this species varies in color from a dark blackish-brown to a brownish-gray. Small patches of white fur may be found on some individuals. The base of the ears almost meets at the midline of the head, but they are not joined. The wings are long and narrow and almost half of the tail is free from the uropatagial

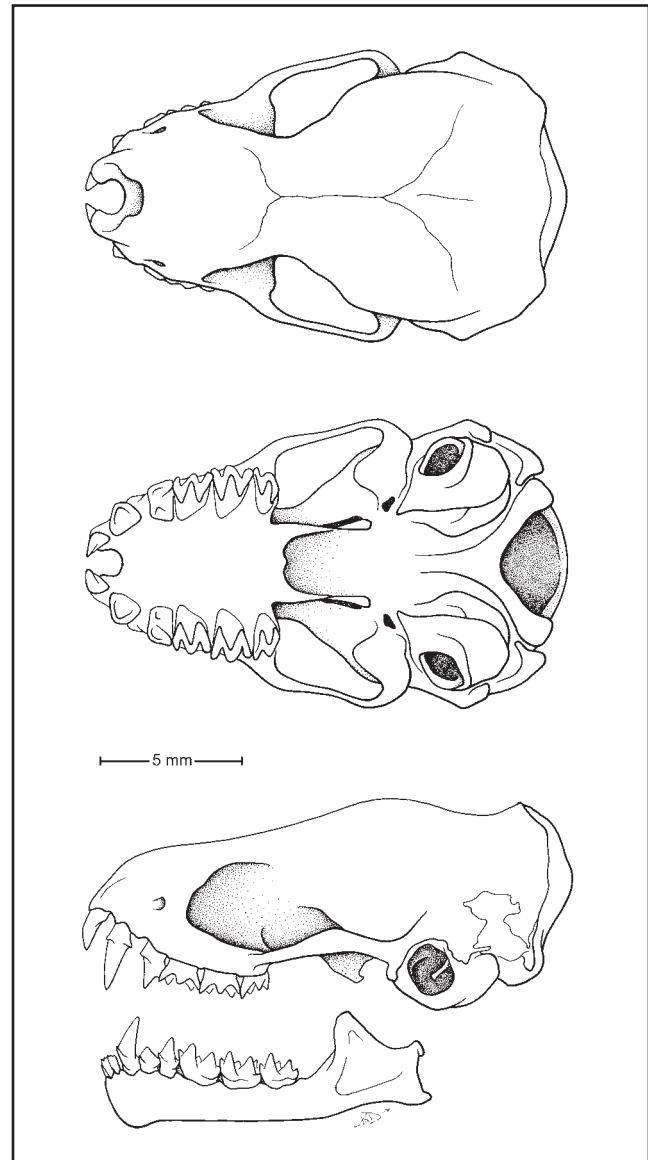


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Tadarida brasiliensis* from Alachua County, Florida (USNM 239257, male).

membrane. The other free-tailed bat in the region, Wagner's bonneted bat (*Eumops glaucinus*), is much larger. The forearm length of *E. glaucinus* is always greater than 50 mm. The dental formula is I 1/2, C 1/1, P 2/2, M 3/3 = 30 (Figure 1).

CONSERVATION STATUS

The Brazilian free-tailed bat has a global rank of Secure (NatureServe 2007). It is Secure in Mississippi and Texas, and Apparently Secure in Georgia, Louisiana and North Carolina. It is listed as Vulnerable in Alabama, Arkansas and Oklahoma. It is unranked in Florida and South Carolina.

DISTRIBUTION

The Brazilian free-tailed bat is distributed in the United States from southern Oregon to northern Utah and eastward to southern North Carolina (Hall 1981). The species also occurs throughout Mexico, Central America, and in large portions of western and central South America (Hall 1981, Koopman 1982, Wilkins 1989). Figure 2 depicts the distribution of the species in the South. *Tadarida brasiliensis cynocephala* reaches its western distributional limits in southeastern Oklahoma (Caire et al. 1989, Caire 1998) and eastern Texas (Spenrath and LaVal 1974, Schmidly 1983, 1991; Schmidly et al. 1977) where its range overlaps seasonally with migrating *T. b. mexicana* (Carter 1962, Spenrath and LaVal 1974, Glass 1982). The northern distributional limits of the southeastern subspecies extend from central Arkansas (Sealander and Price 1964, Saugey et al. 1983, 2001; Sealander and Heidt 1990) to southeastern North Carolina (Lee and Marsh 1978, Lee et al. 1982, Webster et al. 1985). Snyder (1993) reported the first record of *T. b. cynocephala* from Tennessee. *Tadarida brasiliensis cynocephala* is widely distributed in Louisiana (Tesh and Schneidau 1967, Lowery 1974), Mississippi (Tesh and Schneidau 1967, Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989), Alabama (Howell 1921, DiSalvo et al. 1969, Kiser 1996, 2000; Henry et al. 2000, Hilton and Best 2000), Georgia (Benson 1944, Golley 1962, Laerm et al. 1980, 1981; Menzel et al. 2000), and South Carolina (Golley 1966, Webster et al. 1985, Menzel et al. 2002). The bat is distributed throughout the mainland of Florida (Jennings 1958, Bain 1981, Humphrey 1992); it is known from the Florida Keys from a single undocumented occurrence (Lazell and Koopman 1985).

ABUNDANCE STATUS

There are few estimates of the density of *T. b. cynocephala* in the South. Information on the abundance of the subspecies is so poorly known in Oklahoma that the Nongame Section of the Oklahoma Department of Wildlife Conservation placed the bat on the state list of Species of Special Concern (Caire 1988). The Brazilian free-tailed bat appears to be expanding its range northward in Arkansas (Saugey et al. 1983, 1988) and North Carolina (Lee and Marsh

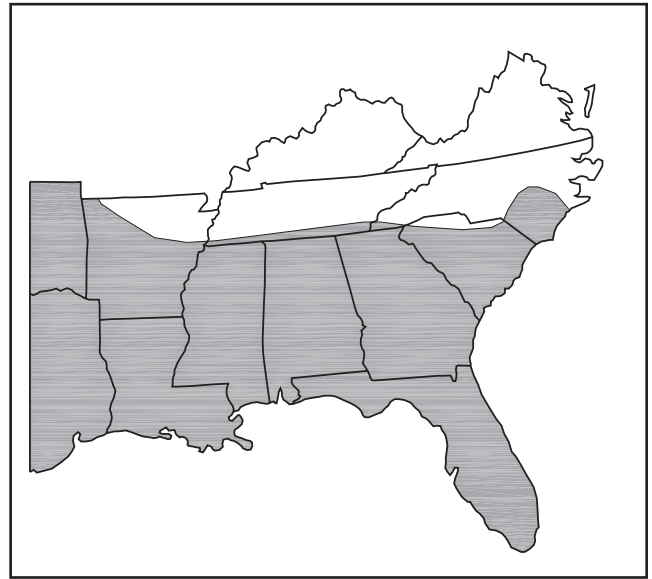


Figure 2. Distribution of *Tadarida brasiliensis* in the South.

1978). In Arkansas, nursery colonies located near the center of the state contain from a few hundred to a few thousand individuals (Saugey et al. 1988). Large colonies once were known in Alabama and Florida, but the population of *T. b. cynocephala* may have declined in these states. Jennings (1958) reported colonies throughout mainland Florida and suggested that many of these colonies contained 50,000 or more bats. However, Humphrey (1992) reported that *T. b. cynocephala* is no longer as common as it once was in Florida. Although some colonies in Florida may contain 20,000 Brazilian free-tailed bats, such colonies are rare.

PRIMARY HABITATS

Unlike the western subspecies that is associated with caves, *T. b. cynocephala* roosts almost exclusively in the hollows of trees or human structures. Tree cavities are the natural roost of the southeastern subspecies of the Brazilian free-tailed bat (Jennings 1958, Lowery 1974). Recent northward expansions of the range may be in response to the increased availability of human-made roost sites (Jennings 1958, Lee and Marsh 1978, Saugey et al. 1983, 1988). These roosts include buildings, bridges, and structures constructed of wood, concrete, brick, or steel. Laerm et al. (1981) observed the bat roosting in a stream tunnel underneath Sanford Stadium on the University of Georgia campus. In Florida, Jennings (1958) suggested that the bat may shift roosts in response to disturbance or local environmental conditions. Although the western subspecies regularly migrates south for the winter, the southeastern subspecies apparently is nonmigratory (Barbour and Davis 1969, Lowery 1974,

Wilkins 1989). Roost sites often are located near fresh water and in areas free from obstruction because the bats drop 3–8 m when departing from a roost (Jennings 1958). Foraging areas may be located far from the roost site. When foraging, it often flies in open areas and frequently flies above water (Norberg 1981, Menzel et al. 2002). Echolocation used while foraging ranges from 25–50 kHz (Simmons et al. 1979).

REPRODUCTION

Although males become sexually mature in their second year, female *T. b. cynocephala* reach sexual maturity at 9 months (Sherman 1937). In Florida populations, ovulation occurs within a 3 week period in March; mating takes place in a 5 week period centered on that time. In eastern Alabama, the testes undergo recrudescence and reach greatest length in February (Henry et al. 2000). After mating, the sexes segregate and the females occupy maternity colonies (Sherman 1937, Pagels and Jones 1974, Saugey et al. 2001). Normally each pregnant female carries a single embryo; however, half of the females in an Alabama sample carried twins or triplets (DiSalvo et al. 1969). The sex ratio at birth is usually 1:1. The young average 25.8 mm in crown-to-rump length and are born naked and blind (Sherman 1937). Parturition in Florida occurs during a 2–3 three-week period in June. New-born young attach themselves to a nipple and begin nursing immediately after birth, but the females leave the young at the roost when they forage at night. Despite some reports that females nurse random young when they return from feeding flights (Davis et al. 1962), using electrophoretic analysis McCracken (1984) demonstrated that females correctly located and nursed their own young 83% of the time. The young reach adult size in 3 weeks and begin flight at 35 days of age (Kunz and Robson 1995).

FOOD HABITS

The Brazilian free-tailed bat typically leaves the roost shortly after sunset to begin foraging, rests during the middle of the night, and engages in a second feeding bout in the early morning hours (Whitaker et al. 1996). Individuals from large colonies of the western subspecies make 65–80 km flights to their feeding grounds (Davis et al. 1962) and exploit insect prey at high altitudes (Williams et al. 1973, Griffin and Thompson 1982). The foraging flight distances and feeding altitudes of the southeastern subspecies are not known. The bat is insectivorous with 90% of the diet consisting of small moths that range from 5–9 mm (Storer 1926, Ross 1961, Barbour and Davis 1969). It also consumes chironomid midges, winged ants, and chalcids (Sherman 1939) but apparently eats few mosquitoes (Storer 1926). Whitaker et al.

(1996) demonstrated that *T. b. mexicanus* eats large numbers of beetles, especially carabids and lygaeids during the evening feeding bout and a higher percentage of moths when feeding in the early morning. The morning feeding bouts correspond temporally with radar observations of northward dispersals of large moths (Raulston et al. 1986, Wolf et al. 1990). Many of the moth species involved in these pre-dawn movements are significant agricultural pests (Wolf et al. 1986, 1990, 1994).

ASSOCIATED SPECIES

Tadarida b. cynocephala sometimes shares structures with other species that roost in buildings, but they do not roost communally with other species. In Florida, Jennings (1958) found these bats in association with the big brown bat (*Eptesicus fuscus*), evening bat (*Nycticeius humeralis*), and southeastern myotis (*Myotis austroriparius*).

VULNERABILITY AND THREATS

Populations of *T. brasiliensis* in the United States have declined drastically in the past four decades (McCracken 1986). Several factors have contributed to the decline, including pesticide exposure (Clark et al. 1975, Geluso et al. 1976, McCracken 1986), heavy metals (Reidinger 1972, Clark 1981), and roost disturbance (Lowery 1974, Humphrey 1992). Young bats are susceptible to poisoning during their first migratory flight because their mother's milk and post-weaning diet may contain high pesticide levels (Clark et al. 1975). In the South, the bat is vulnerable to roost disturbance.

MANAGEMENT SUGGESTIONS

The use of insecticides should be carefully monitored (Bat Conservation International 2001). The razing of old buildings that serve as roosting habitat should be avoided. When nuisance management is needed, bat-friendly exclusion methods are an effective long-term solution. The provision of artificial roosts may be beneficial. The bat migrates seasonally between the southern United States and Mexico, facing conservation challenges on both sides of the border (Pierson 1998).

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Rafinesque's Big-eared Bat

Corynorhinus rafinesquii (Lesson, 1827)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Rafinesque's big-eared bat (*Corynorhinus rafinesquii*) has a complex taxonomic history. The species originally was described as *Vespertilio megalotis* by Rafinesque (1818) based on specimens collected from the lower Ohio Valley. Lesson (1827 in Handley 1959) paraphrased the original description of the species and renamed it *Plecotus rafinesquii*. However, LeConte (1831 in Handley 1959) renamed the species to *P. macrotus*, which was used for several years. The bat also was described as *P. lecontei* by Cooper (1837) and *C. rafinesquii* by Allen (1916). In his revision of the genus, Handley (1959) noted that the name *V. megalotis* had been used for an African species prior to Rafinesque's description. Consequently, the original scientific name for Rafinesque's big-eared bat was rendered invalid. From the remaining available names, the name *P. rafinesquii* had priority (Handley 1959). Recently, Tumlison and Douglas (1992) evaluated the relationships of several genera of big-eared bats and concluded that the big-eared bats of the eastern United States should be assigned to *Corynorhinus*. For many years, various authors confused Rafinesque's and Townsend's big-eared bats (*C. townsendii*). Some of the literature using *C. (=Plecotus) rafinesquii* actually refers to *C. (=Plecotus) townsendii*, a species with a more western distribution. Although the geographic ranges of the two species overlap in portions of the Appalachian Highlands and Ozark Mountains, the two species have never been taken at the same locality. Two recognized subspecies of *C. rafinesquii* occur in the southern United States: *C. r. rafinesquii* inhabits an area west of the Appalachians and north of Alabama, Mississippi, and Louisiana; *C. r. macrotis* occurs in the Atlantic and Gulf Coast regions (Handley 1959, Hall 1981). Additional vernacular names that have been used for this species include eastern lump-nosed bat, eastern big-eared bat, eastern long-eared bat, and southeastern big-eared bat. A comprehensive review of the species is provided by Jones (1977).

DISTINGUISHING CHARACTERISTICS

Rafinesque's big-eared bat is a medium-sized, long-eared bat with two prominent fleshy lumps on the nose. Measurements are: total length, 95–105 mm; tail, 33–54 mm; hind foot, 8–13 mm; ear, 27–37 mm;

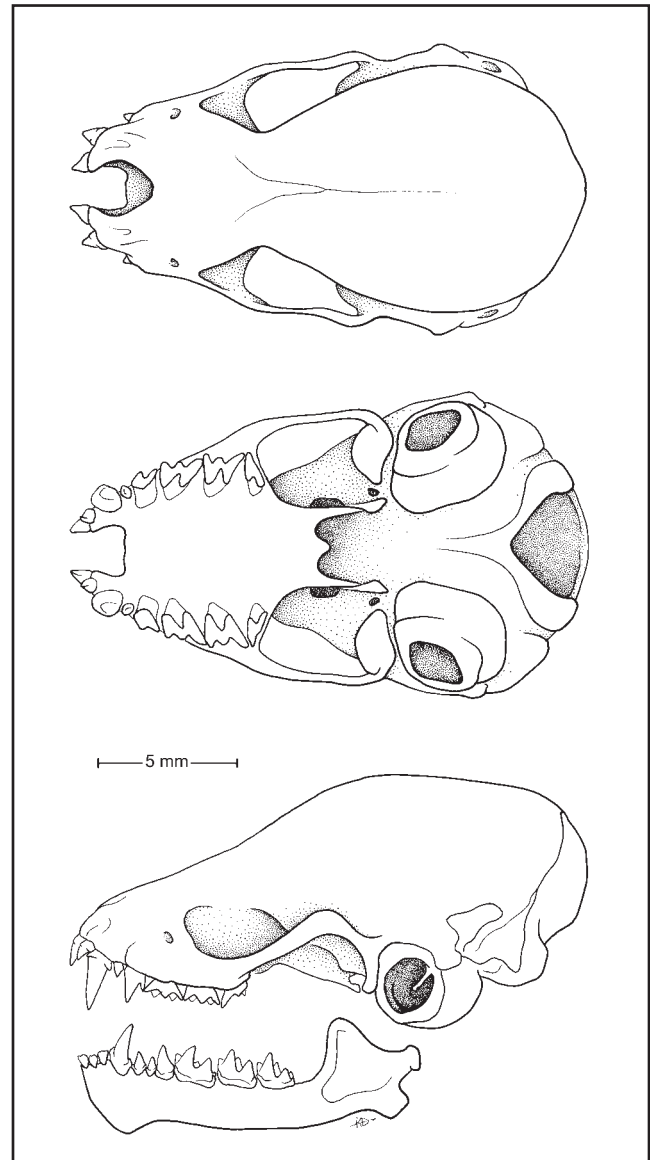


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Corynorhinus rafinesquii* from Marion Parrish, Louisiana (USNM 136100, gender unknown).

forearm, 38–44 mm; and weight, 6.0–9.5 g. The ears are joined at the base, taper to a narrow tip and are curled like a ram's horn when the bat is at rest or in a torpid state. Prominent lumps are present on each side of the muzzle and in front of the eyes. The pelage is grayish-brown or gray on the dorsum and white or

buff on the venter. In the South, only bats of the genus *Corynorhinus* and Wagner's bonneted bat (*Eumops glaucinus*) possess ears that exceed 20 mm in length. However, the latter species does not possess lumps on the muzzle and its dorsal coloration is black or dark grayish black. *Corynorhinus rafinesquii* is distinguished from *C. townsendii* by the ventral pelage color pattern (Handley 1959, Jones 1977). The abdominal hairs of Rafinesque's big-eared bat have grayish-black bases but the tips are white or whitish forming a sharp contrast. The bases of the hairs on the venter of *C. townsendii* are grayish-brown, the tips of the hairs are brown or buff, and there is no sharp contrast. The hairs on the feet of *C. rafinesquii* project well beyond the toes, whereas in *C. townsendii* they do not exceed the toes in length. The first incisor of *C. rafinesquii* is bicuspidate, but the first incisor of *C. townsendii* is unicuspid. Sealander and Heidt (1990) provide additional details and keys to species identification. The dental formula is I 2/3, C 1/1, P 2/3, M 3/3 = 36 (Figure 1).

CONSERVATION STATUS

Rafinesque's big-eared bat has a global rank of Vulnerable (NatureServe 2007). Georgia, Kentucky, Louisiana, Mississippi, North Carolina, Tennessee and Texas also classify it as Vulnerable. It is Imperiled in Alabama, Arkansas, Florida, South Carolina, and Virginia. Oklahoma lists it as Critically Imperiled.

DISTRIBUTION

Corynorhinus rafinesquii occurs throughout the South (Figure 2) from the Ohio River Valley of southern Illinois, Indiana, and Ohio westward to southeastern Missouri and eastern Oklahoma and Texas (Handley 1959, Hall 1981). The species has been taken from southeastern Oklahoma (Blair 1939, Caire et al. 1989) and from a small region of eastern Texas (Michael and Birch 1967, Davis 1974, Schulz et al. 1975, Schmidly et al. 1977, Schmidly 1983, 1991). The bat occurs in all physiographic provinces of Arkansas except the Ozark Mountains (Black 1936, Baker and Ward 1967, McDaniel and Gardner 1977, Gardner and McDaniel 1978, Heath and Heidt 1983, Pitts 1988, Saugey et al. 1989, 1993; Sealander and Heidt 1990, Tumblison et al. 1992, England and Saugey 1999). *Corynorhinus rafinesquii* occurs in Louisiana (Lowery 1943, 1974; Dalquest and Werner 1954, Jones and Suttikus 1975, Lance and Garrett 1997, Lance et al. 2001), Mississippi (Allen 1894, Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989, Best and Caesar 2000), Alabama (Allen 1894, Miller 1897, Howell 1921, Holliman 1983), Georgia (Miller 1897, LeConte 1855, Harper 1927, Golley 1962, Laerm et al. 1980, 1981; Menzel et al. 2000), Florida (Allen 1894,

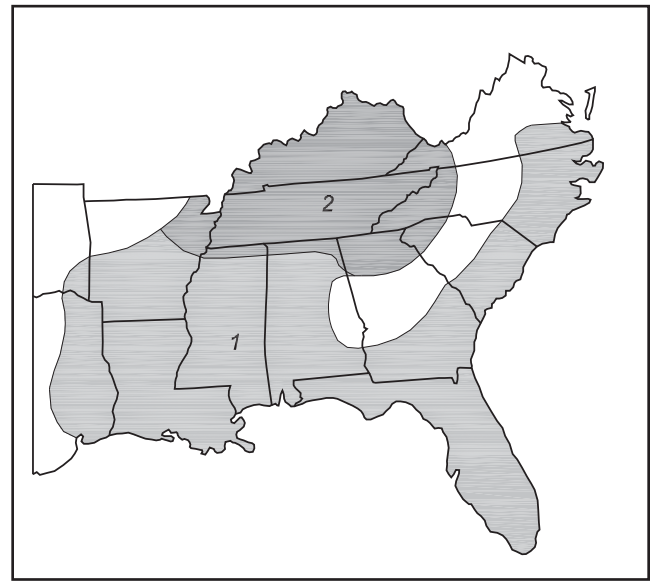


Figure 2. Distribution of *Corynorhinus rafinesquii* in the South: (1) *C. r. macrotis*; (2) *C. r. rafinesquii*.

Moore 1949a,b; Neill 1953, Pearson 1954, Jennings 1958, Brown 1974, 1978, 1993; Humphrey 1992, Brown and Brown 1993), Tennessee (Goodpaster and Hoffmeister 1952, Linzey and Linzey 1971, Graves and Harvey 1974, Smith et al. 1974, Kennedy and Harvey 1980, Harvey et al. 1991, Kennedy 1991), and Kentucky (Miller 1897, Allen 1916, Barbour 1957, Hall 1963, Barbour and Davis 1974, Harvey et al. 1991, Hurst and Lacki 1997, 1999; Lacki 2000, Lacki and Ladeur 2001). The species also occurs in eastern South Carolina (Penny 1950, Golley 1966) and North Carolina (Lee et al. 1982). South Carolina records are from the Coastal Plain and the Appalachian Mountains in the extreme northwest part of the state (Golley 1966, Webster et al. 1985, Menzel et al. 2001, 2002, 2003), and all North Carolina records are from the Coastal Plain and Appalachian Mountains (Brimley 1905, 1945; Komarek and Komarek 1938, Odum 1949, Barbour 1957, Handley 1959, Lee et al. 1982, Webster et al. 1985, Clark 1991). In Virginia, the bat is found in the extreme southwestern tip (Howell 1909, Bailey 1946) and in the southeast (Miller 1897, Webster et al. 1985, Handley 1991, Linzey 1998).

ABUNDANCE STATUS

Known roost sites of *C. rafinesquii* are scattered and the species is considered rare throughout its range. Although individuals occasionally roost alone, Rafinesque's big-eared bats normally roost in small clusters that range in size from 2–100 bats (Goodpaster and Hoffmeister 1952, Handley 1959, Baker and Ward 1967, Jones and Suttikus 1975, Hurst and Lacki 1999). Clusters in the northern parts of the range tend to be larger than those in the South (Jones 1977). Within a

given area, the bat changes roost locations frequently (Barbour and Davis 1969, Jones and Suttkus 1975). During winter, some *C. rafinesquii* enter a torpid state (McNab 1974, Pearson 1962) but the details and timing of this period of inactivity are unknown (Jones 1977). There is no evidence that this species migrates (Handley 1959).

PRIMARY HABITATS

Rafinesque's big-eared bat occurs in several forest types. When caves are available, they may be used as roost sites (Hurst and Lacki 1999, Best and Caesar 2000). When roosting in caves, the bat selects roosting sites located in cool zones near the cave entrance to offer twilight illumination. The bat roosts in different rooms of a cave in winter and summer (Hurst and Lacki 1999). Most roost sites, however, are located in partially illuminated, human-constructed shelters, and buildings (Jones 1977, Menzel et al. 2002). Recorded roost sites for this species include occupied buildings (Komarek and Komarek 1938, Odum 1949), a turpentine still (Harper 1927), well shafts and cisterns (Goodpaster and Hoffmeister 1952, Baker and Ward 1967, Schulz et al. 1975, England and Saugey 1999), concrete culverts (Moore 1949a), bridges (Lance et al. 2001), an artificial limestone grotto (Neill 1953), hollow trees (Harper 1927, Lowery 1943), and crevices behind loose bark (Handley 1959). Menzel et al. (2001) found that the mean home range size of male Rafinesque's big-eared bats was 93.1 ha.

REPRODUCTION

Details of the reproductive biology of *C. rafinesquii* are poorly known. Apparently, copulation takes place in fall and winter (Hoffmeister and Goodpaster 1963). Specimens with enlarged testes have been found in August (Hall 1963, Menzel et al. 2002) and September (Baker and Ward 1967, Menzel et al. 2002); individuals with sperm in the epididymis have been collected in January (Layne 1958, Handley 1959). Information on fertilization, implantation, and gestation period is lacking. Parturition occurs during late May-early June in the northern portion of the range (Baker and Ward 1967, Barbour and Davis 1969) and may occur somewhat earlier in southern areas (Jones and Suttkus 1975). Females give birth to a single young which is born blind and naked (Handley 1959). The young develop a dark pelage within a few days after birth and then undergo a molt to adult pelage after 3 months (Jones and Suttkus 1975). The age at sexual maturity is not known. One female bat reached a known age of 10 years and 1 month (Paradiso and Greenhall 1967).

FOOD HABITS

Rafinesque's big-eared bat emerges well after dark to forage and returns to its roost before twilight (Handley 1959, Barbour and Davis 1969). Male bats forage during two periods: within 4 hours after sunset and again 2 hours before sunrise (Menzel et al. 2001). Rafinesque's big-eared bat feeds on nocturnal flying insects (Clark 1991, Lacki and Ladeur 2001, Menzel et al. 2002). Hurst and Lacki (1997) found that *C. rafinesquii* in Kentucky took insects from 7 orders, particularly moths ranging 31–57 mm in size. The relatively large, broad wings with low wing loading (Jones and Suttkus 1971) suggests that the bat is a slow flier with excellent maneuverability (Norberg and Rayner 1987). The species produces low amplitude echolocation calls with frequencies from 60–90 kHz (Humphrey 1992). These collective characteristics indicate that *C. rafinesquii* may forage by gleaning insects from vegetation (Simmons et al. 1979, Belwood 1988). Lacki and Ladeur (2001) observed *C. rafinesquii* gleaning moths from the walls and ceilings of caves.

ASSOCIATED SPECIES

Rafinesque's big-eared bat frequently roosts in the same cave or structures as other species of bats, but usually roosts alone or in single-species clusters (Jones and Suttkus 1975). The eastern pipistrelle (*Pipistrellus subflavus*) is often found in the same roost. *C. rafinesquii* also shares roosts with the eastern small-footed myotis (*Myotis leibii*), little brown myotis (*M. lucifugus*), Indiana myotis (*M. sodalis*), southeastern myotis (*M. austroriparius*), gray myotis (*M. grisescens*), and big brown bat (*Eptesicus fuscus*).

VULNERABILITY AND THREATS

Rafinesque's big-eared bat is not considered abundant in any portion of its range. Alteration and destruction of roost sites may eliminate local populations. Its habit of roosting in abandoned buildings and in the twilight zone of caves renders it susceptible to human disturbance (Lacki 2000).

MANAGEMENT SUGGESTIONS

Historically, this species may have roosted in large, hollow trees in old-growth conditions such as those found in bottomland forests (M. K. Clark, North Carolina Museum of Natural Science, personal communication). Throughout the range of the bat, roosts have been lost in upland areas where timber harvests have occurred (D. Saugey, USDA Forest Service, personal communication). Protection of existing hollow tree roosts and the promotion of forest conditions

with large bores in areas adjacent to water are important.

The bat is routinely observed in abandoned cabins, shacks, barns, and outbuildings (Menzel et al. 2001). Identifying and retaining these structures can be beneficial when safety allows. The conflict between the bat and humans in anthropogenic structures is an important conservation issue; fears regarding rabies and misconceptions about health risks have resulted in unnecessary eradication (Pierson 1998). Education is a key component in securing protection of the bat. Artificial roosts provide alternatives in areas where large hollow trees and abandoned buildings are not available. Where the species occupies caves and rock shelters, protection of these sites is desirable (Bat Conservation International 2001). Additional research is needed on maternity, hibernation, and foraging requirements.

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Townsend's Big-eared Bat

Corynorhinus townsendii (Cooper, 1837)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Cooper (1837) described Townsend's big-eared bat (*Corynorhinus townsendii*). After a revision of the genus by Handley (1959), the species was known for many years as *Plecotus townsendii*. *Corynorhinus* was considered a subgenus of *Plecotus* (Hall 1981, Koopman 1993). Tumlison and Douglas (1992) restored *Corynorhinus* to generic status after evaluating the relationships of the big-eared bats. For several years following the revision by Handley (1959), some authors confused *C. townsendii* with Rafinesque's big-eared bat (*C. rafinesquii*). This confusion arose because *C. rafinesquii* was misapplied to the western forms of *C. townsendii* (Handley 1959). The two species are superficially similar which may have contributed to the confusion, particularly in the southeastern United States. There are five recognized subspecies, two of which, the Virginia big-eared bat (*C. t. virginianus*) and the Ozark big-eared bat (*C. t. ingens*), occur in the South (Handley 1959, Hall, 1981). The species is also known as the lump-nosed bat. The literature on *C. townsendii* is reviewed by Kunz and Martin (1982), Bagley (1984), U. S. Department of Interior (1995), and Pierson et al. (1999).

DISTINGUISHING CHARACTERISTICS

Corynorhinus townsendii is a large-eared, brownish bat with two obvious, fleshy lumps on its nose. Measurements are: total length, 95–115 mm; tail, 42–54 mm; hind foot, 10–13 mm; ear, 32–38 mm; forearm, 40–45 mm; weight, 7–12 g. The dorsal pelage of individual bats varies in color from light brown to dark brown; the venter is pale brown. The extremely large ears and lumps on the nose are the noticeable characteristics of the genus. *Corynorhinus townsendii* is characterized by brown hair on the ventral side and short toe hairs whereas *C. rafinesquii* has white hair and hairs extending past the toes. The first incisor of Townsend's big-eared bat has a single cusp rather than the bicuspidate *C. rafinesquii*. The dental formula is $I\ 2/3, C\ 1/1, P\ 2/3, M\ 3/3 = 36$ (Figure 1). Tumlison (1995) presents a description of cranial features that also can be used to distinguish the two species. The only other bat in the region with ears as large is Wagner's bonneted bat (*Eumops glaucinus*), which does not have lumps on its nose.

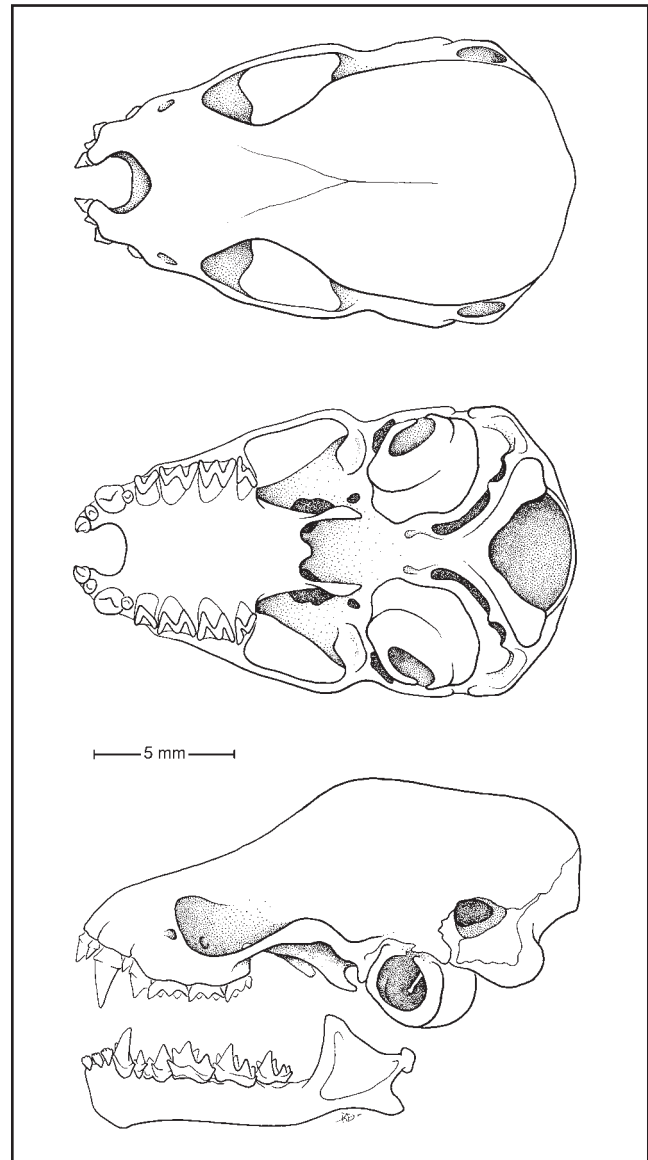


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Corynorhinus townsendii* from Lee County, Kentucky (USNM 360513, male).

CONSERVATION STATUS

Townsend's big-eared bat has a global rank of Apparently Secure (NatureServe 2007). It is Vulnerable in Oklahoma and Texas, and Critically Imperiled in Arkansas, Kentucky, and North Carolina. It is unranked in Tennessee. The U. S. Fish and Wildlife

Service (U. S. Department of the Interior 2007) lists the Virginia and Ozark big-eared bats as Endangered.

DISTRIBUTION

Townsend's big-eared bat is distributed throughout the western half of the United States from British Columbia to southern Mexico. Isolated populations occur in eastern Oklahoma, Arkansas, Missouri, Kentucky, West Virginia, and Virginia (Figure 2). *C. t. ingens* is known from a few caves in eastern Oklahoma (Caire et al. 1989, Clark 1991, Clark et al. 1993, 1996, 2002; Wethington et al. 1997), northwestern Arkansas (Sealander 1951, Sealander and Young 1955, McDaniel and Gardner 1977, Gardner and McDaniel 1978, Harvey 1975, 1978; Harvey et al. 1978, 1979; Harvey and Barkley 1990, Sealander and Heidt 1990, Wilhide et al. 1998a,b; Prather and Briggler 2002), and southwestern Missouri (Harvey 1975, Finnley 1978, 1979a). *Corynorhinus townsendii virginianus* populations are centered in eastern Kentucky (Rippy and Harvey 1965, Barbour and Davis 1974, Harvey et al. 1991, Lacki et al. 1993, 1994; Adam et al. 1994, Burford and Lacki 1998), southwestern Virginia (Bailey 1946, Tipton 1983, Dalton et al. 1986, Handley 1991, Linzey 1998), northwestern North Carolina (Clark and Lee 1987), and eastern West Virginia (Finnley 1979a,b).

ABUNDANCE STATUS

The population of the Virginia big-eared bat was estimated at 4,000 in 1978 (Finnley 1978). An additional small population was discovered in North Carolina caves (Clark and Lee 1987); there are fewer than five known caves containing nursery colonies of this subspecies. Currently, there are approximately 13,000–14,000 Virginia big-eared bats. The population of the Ozark big-eared bat in Arkansas was estimated as less than 200 individuals (Finnley 1978, Harvey et al. 1979). Clark et al. (1997a,b) reported 515 females leaving Oklahoma maternity caves in 1995 and estimated that the total population was approximately 1800.

PRIMARY HABITATS

Townsend's big-eared bat occasionally roosts in buildings during summer (Harvey et al. 1978); one individual was found roosting on a rhododendron (*Rhododendron maximum*) near the entrance to a cave (Clark and Lee 1987). Both subspecies inhabit caves during summer and winter. Several caves used by the bat are in limestone formations (Barbour and Davis 1969, Sealander and Heidt 1990, Lacki et al. 1994, Clark et al. 1997a,b; Prather and Briggler 2002), whereas some in North Carolina are in a schist formation (Clark and Lee 1987). The habitat surrounding cave entrances do not appear to affect cave selection

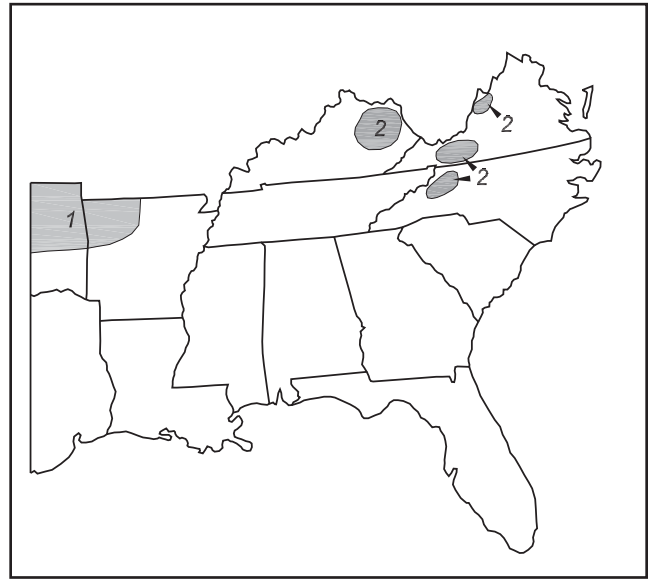


Figure 2. Distribution of *Corynorhinus townsendii* in the South: (1) *C. t. ingens*; (2) *C. t. virginianus*.

(Wethington et al. 1997). During summer, females form nursery colonies while males remain solitary (Barbour and Davis 1969). Males often occupy the same cave as maternity colonies (Wilhide et al. 1998b). Maternity colonies, which may be composed of one to several clusters, are located in warm parts of caves (Twente 1955, Humphrey and Kunz 1976, Clark et al. 1996), but in locations with cooler temperatures than those used by solitary bats (Clark et al. 1996). Roost sites are located on open ceilings easily reached by flying because the Townsend's big-eared bat does not crawl away from its landing spot (Barbour and Davis 1969). Most roosts are situated in dim light near the zone of total darkness; roost locations are shifted throughout the summer (Twente 1955, Graham 1966). Maternity colonies break up in August (Pearson et al. 1952). During winter, most *C. townsendii* roost singly, but clusters containing approximately 400 individuals are known (Twente 1955, Rippy and Harvey 1965, Harvey 1975, Humphrey and Kunz 1976, Clark and Lee 1987, Clark et al. 1997a,b). Clusters of hibernating Ozark big-eared bats were located in the coldest regions of caves in Oklahoma (Clark et al. 1996, 2002). The bat prefers relatively cold, well-ventilated locations and often are found near cave entrances when hibernating (Dalquest 1947, Twente 1955, Martin and Hawks 1972, Humphrey and Kunz 1976, Clark et al. 1996). Throughout winter, hibernating bats periodically awaken and change locations within a hibernaculum or move among nearby hibernacula (Rippy and Harvey 1965, Humphrey and Kunz 1976, Clark et al. 2002).

REPRODUCTION

The only information on reproduction in this species is derived from a study of a California population of *C. townsendii* by Pearson et al. (1952). Whether this applies to the eastern subspecies of the Townsend's big-eared bat is not known. Spermatogenesis in adult males begins during summer and reaches a maximum in September (Pearson et al. 1952). Females have a short proestrus in late summer followed by estrus and mating in fall. Copulation takes place from November–February, and occurs after a ritualized precopulatory behavior that consists of audible vocalizations and head nuzzling. A female stores sperm until spring when ovulation occurs. The gestation period is 56–100 days, and is influenced by ambient temperature and torpor period. A single young is born in May–early July. The young are capable of flight by 3 weeks of age and are fully weaned within 6 weeks. Young females mate during their first autumn but some young males do not reach sexual maturity until their second fall.

FOOD HABITS

Corynorhinus townsendii emerges late in the evening to begin foraging activity (Dalquest 1947, Pearson et al. 1952, Twente 1955, Cockrum and Cross 1964, Jones 1965, Ross 1967). There are also records of twilight emergence (Barbour and Davis 1969, Caire et al. 1986, Clark et al. 1993, 2002). Late evening foraging departures are often preceded by light sampling behavior (Pearson et al. 1952, Clark et al. 1993). The Townsend's big-eared bat feeds on flying insects including Coleoptera (beetles), Diptera (flies, midges, mosquitoes, gnats), Homoptera (leaf hoppers), Hymenoptera (flying ants), Lepidoptera (moths), Neuroptera (lacewings), Plecoptera (stoneflies), Trichoptera (caddisflies), and others (Ross 1967, Whitaker et al. 1977, Hamilton and Whitaker 1979, Dalton et al. 1986, Sample and Whitmore 1993). The most common prey are small moths 3–10 mm in size (Ross 1967, Sample and Whitmore 1993). Fecal pellets examined in Virginia were dominated by lepidopterans (Dalton et al. 1986). The 45 species of moths collected at feeding roosts in Kentucky represented six families (Burford and Lacki 1998). Like most vespertilionids, the bat forages aerially over many substrates (Barbour and Davis 1969, Adam et al. 1994, Wethington et al. 1996). It also gleans insects from leaves (Howell 1920) and feeds in close proximity to canyon walls (Caire et al. 1986). Townsend's big-eared bat drinks in flight from ponds and sometimes from pools in road ruts (Wilhide et al. 1998a). After feeding, the Townsend's big-eared bat retires to a night roost to digest its food (Pearson et al. 1952, Lacki et al. 1994). The roosts are located in caves (Twente 1955) or rock crevices (Caire et al. 1986, Lacki et al. 1994), and are

often shared with other bat species (Dalquest 1947, Pearson et al. 1952). Because *C. townsendii* usually does not return to a day roost until sunrise, Pearson et al. (1952) suggest that a second feeding bout may take place in the early morning hours.

ASSOCIATED SPECIES

In the South, the bat occupies the same caves as Rafinesque's big-eared bat, eastern pipistrelle (*Pipistrellus subflavus*), northern long-eared myotis (*Myotis septentrionalis*), and eastern small-footed myotis (*M. leibii*; Sealander and Young 1955, Rippey and Harvey 1965, Clark and Lee 1987). Allegheny woodrats (*Neotoma magister*) and southern red-backed voles (*Clethrionomys gapperi*) were found in North Carolina caves that contained Townsend's big-eared bats (Clark and Lee 1987).

VULNERABILITY AND THREATS

Isolated populations of *C. townsendii* are vulnerable to human disturbance (Graham 1966, Barbour and Davis 1969, Humphrey and Kunz 1976). Cave disturbance, vandalism, and loss of habitat have influenced the decline of the bat (Harvey 1975, Humphrey and Kunz 1976).

MANAGEMENT SUGGESTIONS

Populations of Townsend's big-eared bat are highly localized in a few caves and mines; maternity and hibernating colonies are extremely susceptible to human disturbance and vandalism. Excessive human intrusion may result in cave abandonment (Bat Conservation International 2001). Cave protection is critical for this species (Martin and Hanks 1972, White and Seginak 1987). Conservation actions center on the protection of roosting sites, foraging habitat, and prevention of cave disturbance (including recreational caving) by limiting public access during critical use periods (Bat Conservation International 2001). Populations have increased at several sites following reduction of human disturbance (C. Stihler, West Virginia Department of Natural Resources, personal communication). Recovery can be quite slow.

Bat gates (angle iron) designed by American Cave Conservation Association, that do not interfere with airflow or bat passage, work well for the big-eared bat. Gates at small cave entrances may increase predation; the establishment of the gate into the cave interior may be necessary (C. Stihler, West Virginia Department of Natural Resources, personal communication). Another management option is using 10-foot fencing to surround the cave entrance. Additional actions include the retention of hollow trees, prevention of

cave entrance and mine portal modification, and monitoring populations on an annual basis. In locations where the bat might roost in buildings, the maintenance of old sheds and farm buildings is beneficial. Retaining oldfields and other openings within densely forested locations may improve foraging opportunities. Additional research is needed on roost temperature requirements at maternity and hibernation sites.

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Big Brown Bat

Eptesicus fuscus (Palisot de Beavois, 1796)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The big brown bat (*Eptesicus fuscus*) originally was described as *Vespertilio fuscus*. When Miller (1897) revised the North American bats, *V. fuscus* was retained. The present taxonomic combination was first used by Mehely (1900 in Miller and Kellogg 1955). Eleven subspecies currently are recognized (Hall 1981). Two subspecies, *E. f. fuscus* and *E. f. osceola*, occur in the South. The literature on this species is reviewed by Kurta and Baker (1990).

DISTINGUISHING CHARACTERISTICS

Eptesicus fuscus is a large, dark brown bat. Measurements are: total length, 112–130 mm; tail, 38–50 mm; hind foot, 9–13 mm; ear, 16–20 mm; forearm, 42–50 mm; weight, 13–25 g. The dorsal fur is long and each hair is darker at the base than at the tip giving the bat a dark, cinnamon brown or slightly bronzy appearance. The venter is pale brown. The black, leathery ears are short and rounded; the tragi are broad and rounded. The wings and interfemoral membranes are blackish-brown and are almost devoid of fur. The calcar is keeled. Because of its size and color, this species is not easily confused with other bats of the region. The two subspecies are distinguished primarily on the basis of pelage coloration. *Eptesicus f. osceola*, which is restricted to Florida and southeastern Georgia, is darker than *E. f. fuscus* (Rhoads 1902). The dental formula is $I\ 2/3, C\ 1/1, P\ 1/2, M\ 3/3 = 32$ (Figure 1).

CONSERVATION STATUS

The big brown bat has a global rank of Secure (NatureServe 2007). It is also considered Secure in Alabama, Georgia, Kentucky, Mississippi, North Carolina, Tennessee, Texas and Virginia. It is Apparently Secure in Arkansas and Oklahoma. Florida classifies it as Vulnerable, and Louisiana classifies it as Critically Imperiled. It is unranked in South Carolina.

DISTRIBUTION

The big brown bat occurs from the southern parts of all Canadian provinces (van Zyll de Jong 1985) and throughout most of the continental United States, Mexico, Central America and the Greater Antilles, south to northwestern South America (Kurta and

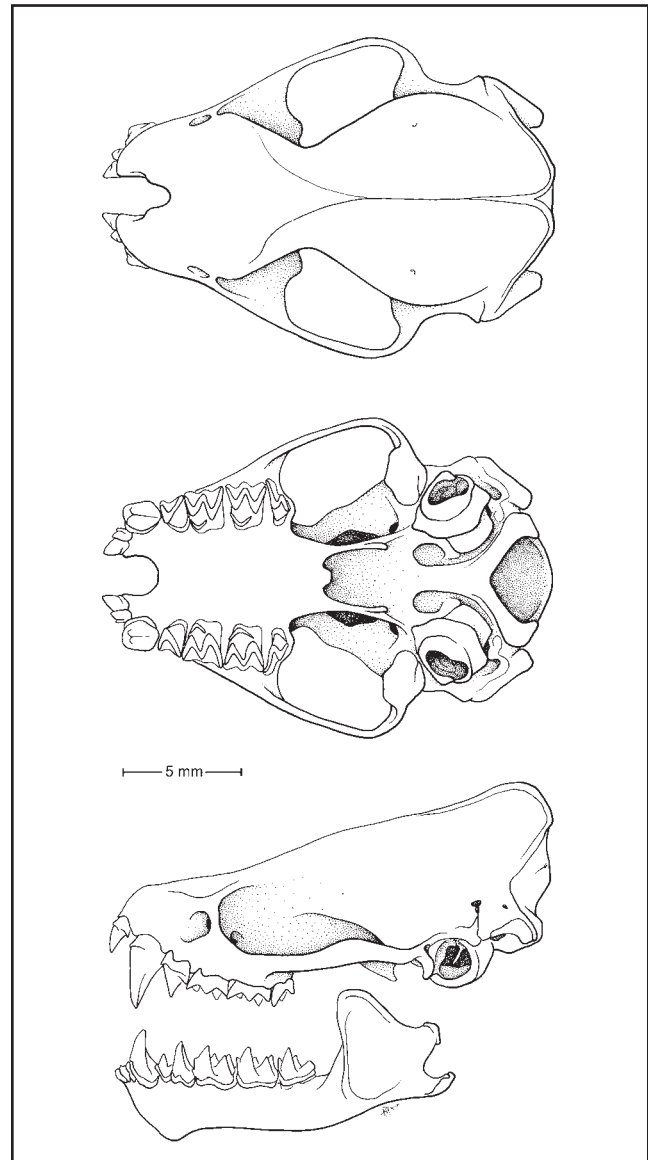


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Eptesicus fuscus* from Botetourt County, Virginia (USNM 279422, female).

Baker 1990). *Eptesicus f. fuscus* occurs throughout most of the eastern United States and all of the southern states except southeastern Georgia and peninsular Florida (Figure 2). In eastern Texas, the big brown bat apparently is restricted to the pine-oak (*Pinus-Quercus*) and longleaf pine (*P. palustris*) vegetation zones (Schmidly et al. 1977, Schmidly 1983, 1991; Manning

et al. 1989). The species does have a wide range in Louisiana but has not been taken near the coast (Lowery 1974, Lance and Garrett 1997). It occurs throughout Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989, Best and Caesar 2000) and Alabama (Howell 1921, Henry et al. 2000, Hilton and Best 2000). It is found in Georgia (Harper 1927, Golley 1962, Baker 1965, Laerm et al. 1980, 1981; Menzel et al. 1997, 2000, 2001; Carter et al. 1998, Johnson et al. 2002, Menzel et al. 2003) except where it intergrades with *E. f. osceola* in the southeastern corner of the state (Laerm et al. 1981). It is found throughout South Carolina (Golley 1966, Webster et al. 1985, Menzel et al. 2002) and probably occurs statewide in North Carolina (Lee et al. 1982, Webster et al. 1985) but there are few records from the Coastal Plain (Lee et al. 1982). It is found statewide in Virginia (Bailey 1946, Webster et al. 1985, Handley 1991, Linzey 1998), Kentucky (Humphrey and Cope 1976, Barbour and Davis 1974), Tennessee (Linzey and Linzey 1971, Harvey et al. 1991, Kennedy 1991), Arkansas (McDaniel and Gardner 1977, Gardner and McDaniel 1978, Saugey et al. 1978, 1988, 1989; Sealander and Heidt 1990, Wilhide et al. 1998) and Oklahoma (Caire et al. 1984, 1989; Caire 1998). *Eptesicus f. osceola* occurs in the southeastern corner of Georgia and most of Florida except the western portion of the Florida Panhandle (Jennings 1958, Hall 1981, Humphrey 1992).

ABUNDANCE STATUS

The big brown bat is regarded as common in many areas of the South, but no population density estimates are available for the region. Still, it is considered uncommon in Louisiana (Lowery 1974), South Carolina, North Carolina, and Virginia (Webster et al. 1985). Few specimen records are available from the southeastern portion of North Carolina where they are considered rare (Lee et al. 1982, Webster et al. 1985). According to Kurta et al. (1989), *E. fuscus* decreases in abundance from the southern deciduous forests to the northern coniferous forests.

PRIMARY HABITATS

After dispersing from hibernacula during March–April, adult female big brown bats form maternity colonies that are located in caves, mines, buildings, tree cavities, or rock crevices (Christian 1956, Barbour and Davis 1969, Kurta 1980, Whitaker and Gummer 1992, Whitaker 1995, Betts 1996, Menzel et al. 1997, Williams and Brittingham 1997, Best and Caesar 2000, Lausen and Barclay 2002). Ambient temperature may be important to the location of a maternity colony because female *E. fuscus* change locations within a site and may abandon a site entirely if the

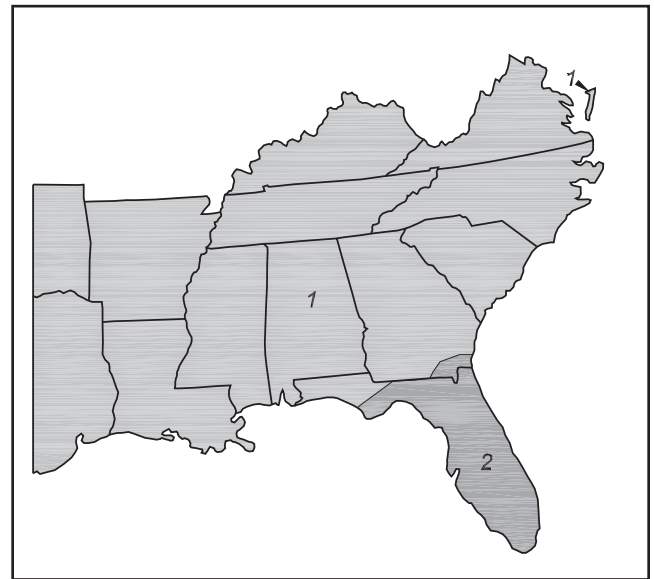


Figure 2. Distribution of *Eptesicus fuscus* in the South: (1) *E. f. fuscus*; (2) *E. f. osceola*.

temperature exceeds 35° C (Davis et al. 1968, Williams and Brittingham 1997). The number of individuals in a maternity colony varies from 5–700; however, many colonies contain 25–75 adults (Davis et al. 1968, Mills et al. 1975). Big brown bats in a maternity colony are highly philopatric, but may use bat boxes of the proper design and placement if excluded from a maternity roost (Brittingham and Williams 2000). *Eptesicus fuscus* apparently uses odor to locate colony sites and mates (Bloss et al. 2002). Approximately 72% of the adult females return to the same maternity colony site in successive years and 10–30% of the immature females return to their natal site (Davis 1967, Brenner 1968, Mills et al. 1975). Some males may occupy maternity colonies but most males roost solitarily or in all-male colonies (Davis et al. 1968, Barbour and Davis 1969). The summer colonies begin to break up in August, but the big brown bat usually does not appear at hibernacula until November (Barbour and Davis 1969). *Eptesicus fuscus* hibernates singly or in small groups (Beer 1955, Mumford 1958, Phillips 1966, Nagorsen 1980) within caves, mines, storm sewers, or buildings (Goehring 1954, Mills et al. 1975, Price et al. 1982, Vonhof 1995). The locations of hibernacula rarely are more than 80 km from their summer roosts (Mills et al. 1975). The bat usually selects hibernating perches with ambient temperatures below freezing, sometimes wedging into cracks and crevices or under rocks on a cave floor (Barbour and Davis 1969, Fenton 1972). Generally, hibernation sites are cooler, drier, and more exposed to air currents than those selected by the little brown myotis (*Myotis lucifugus*; Goehring 1972, Raesly and Gates 1987). The big brown bat often selects roosting sites near the cave entrance (Rysgaard 1942, Baker 1965). During periodic winter warm spells, the species

may awaken and become active outside the hibernaculum (Beer 1955, Mumford 1958).

REPRODUCTION

Production of sperm in the male big brown bat occurs from June–October (Christian 1956). Copulation takes place from September–March (Mumford 1958, Phillips 1966). Ovulation and fertilization is delayed until after the female arouses from hibernation (Wimsatt 1944). During ovulation, females may release 2–5 eggs; only 1 embryo reaches full term in each uterine horn (Birney and Baird 1985). The gestation period is approximately 60 days (Wimsatt 1944). The litter size varies from 1–2 and most births occur May–July (Barbour and Davis 1969, Henry et al. 2000). Although newborn *E. fuscus* are blind, naked, and immobile; their eyes and ears open within a few hours after birth (Gould 1971). The postnatal growth of the big brown bat is slow relative to other species of similar size (Burnett and Kunz 1982), but juveniles are capable of flight at 18–35 days (Kurta and Kunz 1987). Most males become sexually mature within 6 months after birth (Christian 1956), but many juvenile females do not reproduce during their first year (Schowalter and Gunson 1979).

FOOD HABITS

Foraging activity begins just after sundown and the peak of activity occurs within 2 hours. Foraging may occur at any hour of the night (Phillips 1966, Kunz 1973, Kurta 1980, Caire et al. 1984). The bat usually forages within 2 km of the roost site and they may use temporary night roosts to rest and digest food (Brigham and Fenton 1986, Vonhof 1995). The species forages over urban areas, open ground, forest canopies, wetlands, and calm water (Phillips 1966, Kurta 1982, Geggie and Fenton 1985, Furlonger et al. 1987, Mackey and Barclay 1989, Kurta et al. 1993, Menzel et al. 2001, 2005a, 2005b). It rarely forages under a forest canopy (Davis 1974, Menzel et al. 2002, 2005b). Grindal (1995, in Brigham et al. 1997) found that the big brown bat foraged significantly more often at forest clearcut edges than in adjacent intact forests or in the openings created by logging. Foraging activities, however, are not limited by the presence of structural clutter (Mackey and Barclay 1989, Brigham et al. 1997). The most common prey are Coleoptera (small beetles), Lepidoptera (small moths), and Trichoptera (adult caddisflies); a bat may discard the elytra and wings before ingesting the prey (Hamilton 1933, Ross 1967, Black 1972, Coutts et al. 1973, Freeman 1981, Mumford and Whitaker 1982, Brigham and Fenton 1991, Carter et al. 1998). Keeler and Studier (1992) found that

pregnant big brown bats ate large numbers of June beetles. The bat occasionally ingests vegetation and non-flying insects (Whitaker 1972). Because the silver-haired bat (*Lasionycteris noctivagans*) forages later in areas where *E. fuscus* is common, Reith (1980) suggests that the two species may compete for food. The big brown bat is aggressive towards other species of bats at feeding locations (Fenton 1980). The activities of a foraging bat believed to be *E. fuscus* caused a common nighthawk (*Chordeiles minor*) to forage in less desirable habitats when a localized food source was available (Shields and Bildstein 1979).

ASSOCIATED SPECIES

Eptesicus fuscus roosts singly or in single-species groups in caves, mines, and buildings where it is likely to share the general location with any other cave dwelling species. In western Oklahoma, Caire et al. (1984) found the bat in the same cave as the eastern pipistrelle (*Pipistrellus subflavus*), cave myotis (*M. velifer*), Townsend's big-eared bat (*Corynorhinus townsendii*), and Brazilian free-tailed bat (*Tadarida brasiliensis*).

VULNERABILITY AND THREATS

The big brown bat is distributed widely and relatively common in many portions of the southern region. Individuals may survive up to 19 years in the wild (Paradiso and Greenhall 1967), which contributes to population stability. There are no current threats to its existence.

MANAGEMENT SUGGESTIONS

Conservation actions focus on the protection of winter hibernacula (Bat Conservation International 2001). Loss of roosting snags may contribute to the use of barns, attics, house siding, and culverts (Menzel et al. 2003). The conflict between the species and humans in anthropogenic structures is an important conservation issue; fears regarding rabies and misconceptions about health risks have resulted in unnecessary eradication (Pierson 1998). When nuisance management is needed, proper exclusion methods are an effective long-term solution when combined with provision of artificial roosts. Selective harvests that maintain old, dominant trees may benefit the big brown bat, as well as the use of low intensity burns to maintain open habitat. Young clearcut stands and open bogs and beaver ponds are important foraging habitat (Owen et al. 2004). The bat may not use isolated trees; the maintenance of tree strips is beneficial (Vonhof 1995).

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Big Brown Bat (*Eptesicus fuscus*)

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Silver-haired Bat

Lasionycteris noctivagans (LeConte, 1831)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The silver-haired bat (*Lasionycteris noctivagans*) originally was described as *Vespertilio noctivagans*. The first use of the present scientific name was by Peters (1865, in Kunz 1982). *Lasionycteris* is a monotypic genus that may be closely related to *Myotis* (Tate 1942). Although the baculum of this species is distinct (Hamilton 1949) and the karyotype differs from most other vespertilionids (Baker and Patton 1967), Bickham (1979) suggested that the karyotype of *L. noctivagans* could have evolved from a myotine ancestor. There are no recognized subspecies. The literature is reviewed by Kunz (1982).

DISTINGUISHING CHARACTERISTICS

Lasionycteris noctivagans is a medium-sized, dark-colored bat. Measurements are: total length, 90–106 mm; hind foot, 7–10 mm; ear 14–17 mm; forearm, 38–43 mm; weight 8.5–12.5 g. The dorsal pelage is black to brownish-black with a noticeable frosting of silvery white. The venter is slightly paler, yet dark in appearance. The wing membranes are black and the proximal half of the tail membrane is heavily furred dorsally. The short, rounded ears appear to lack hair. The silver-haired bat is distinguished easily from all other regional species by its distinctive pelage coloration. The dental formula is $I\ 2/3, C\ 1/1, P\ 2/3, M\ 3/3 = 36$ (Figure 1).

CONSERVATION STATUS

The silver-haired bat has a global rank of Secure (NatureServe 2007). It is Secure in Georgia and Apparently Secure in Tennessee and Texas. Virginia ranks the bat as Still Under Review / Apparently Secure. North Carolina, Arkansas, and Oklahoma list it as Imperiled, with Louisiana listing it as Critically Imperiled. Kentucky lists the bat as Not Applicable (i.e., the species is not a suitable target for conservation activities). It is unranked in Florida, South Carolina, and Alabama.

DISTRIBUTION

During summer, the silver-haired bat is found in southeastern Alaska, southern Canada, and throughout most of the United States (Hall 1981, Kunz 1982). The distribution of the bat in the South is depicted in

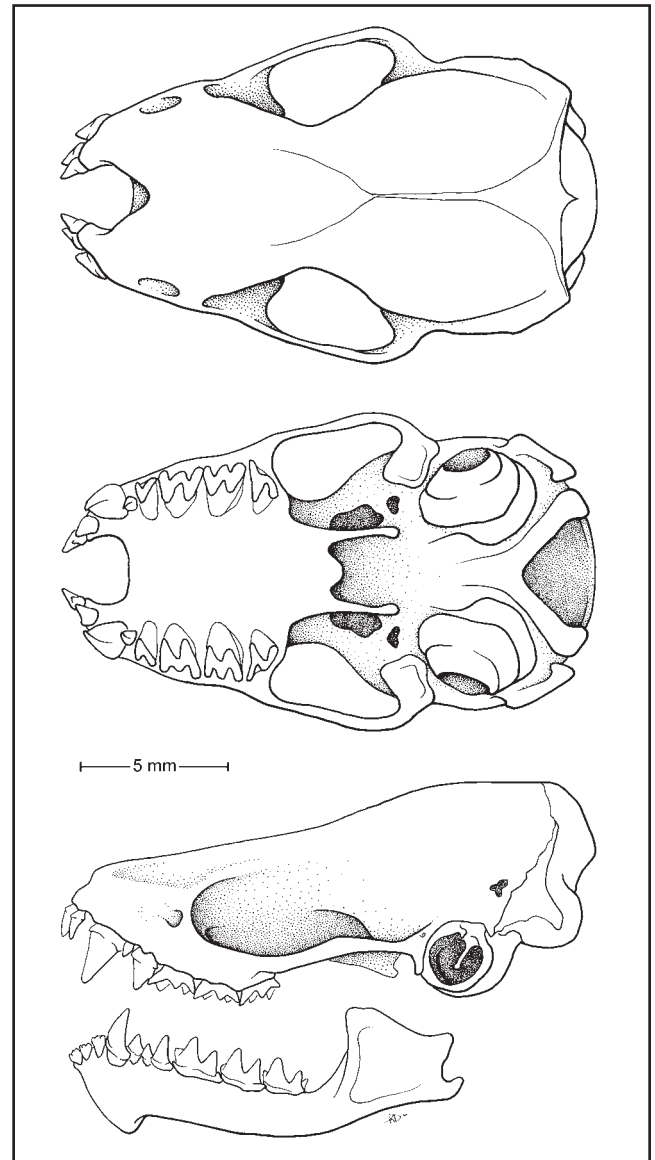


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Lasionycteris noctivagans* from Juneau County, Wisconsin (USNM 82724, female).

Figure 2. The species also is found in northeastern Mexico near the Texas border (Davis 1974, Hall 1981). Mid-summer records of *L. noctivagans* are rare in the South, and most of the available records are male specimens. This may indicate that silver-haired bats (particularly females) migrate north to spend the summer. The silver-haired bat is known from eastern

Texas (Schmidly et al. 1977, Schmidly 1983, 1991; Dowler et al. 1992), central and west-central Louisiana (Lowery 1974, Lance and Rogowski 1999), Alabama (Howell 1921, LaVal 1967), Georgia (Golley 1962, Baker 1965, Laerm et al. 1981), South Carolina (Golley 1966, Webster et al. 1985), North Carolina (Lee et al. 1982, Rabinowitz 1982, Webster et al. 1985), Virginia (Bailey 1946, Webster et al. 1985, Linzey 1998), Kentucky (Welter and Sollberger 1939, Barbour and Davis 1974), Tennessee (Linzey and Linzey 1971, Rabinowitz 1982, Kennedy 1991), Arkansas (Sealander 1960, Gardner and McDaniel 1978, Saugey et al. 1978, 1993; Izor 1979, Heath et al. 1983, 1986; Sealander and Heidt 1990, Harvey et al. 1991), and central and eastern Oklahoma (Caire et al. 1989, Sheffield and Chapman 1990). There is only one record for the species in Florida (Brown 1986, Humphrey 1992), one record for Mississippi (Carter et al. 1987, Jones and Carter 1989), and there are no summer records of the silver-haired bat in Virginia (Webster et al. 1985). During winter, the silver-haired bat may hibernate in suitable caves almost anywhere in the United States except the northern Midwest and Great Plains (Pearson 1962, Gosling 1977, Izor 1979). However, a large portion of the population apparently migrates to southern locations that are above a -6.7°C mean daily minimum temperature isotherm (Izor 1979). Most available records for wintering *L. noctivagans* in the South are from Oklahoma, Arkansas, Kentucky, Virginia, and North Carolina (Izor 1979).

ABUNDANCE STATUS

In certain western states and northern reaches of its range, *L. noctivagans* is regarded as a common bat (Barbour and Davis 1969, Kunz 1982). There are no accounts of its abundance or population dynamics in the southern United States.

PRIMARY HABITATS

The silver-haired bat generally is assumed to be a solitary, tree-roosting species. Despite this assumption, there are few records of this species roosting in trees (Kunz 1982) and small roosting groups are known (Novakowski 1956, Parsons et al. 1986, Clark 1993). The summer roosting preferences are poorly known. During the summer months, individuals may roost in foliage, tree cavities, woodpecker holes, bird nests, or beneath loose bark (Barbour and Davis 1969, Barclay 1984, Betts 1996, Mattson et al. 1996, Vonhof 1996). Menzel et al. (2000) found a silver-haired bat ground-roosting under deciduous leaf litter in a West Virginia forest. A newborn bat was found hanging from a dead white spruce (*Picea galuca*) twig that was approximately 6 m above the ground (Norris-Elye

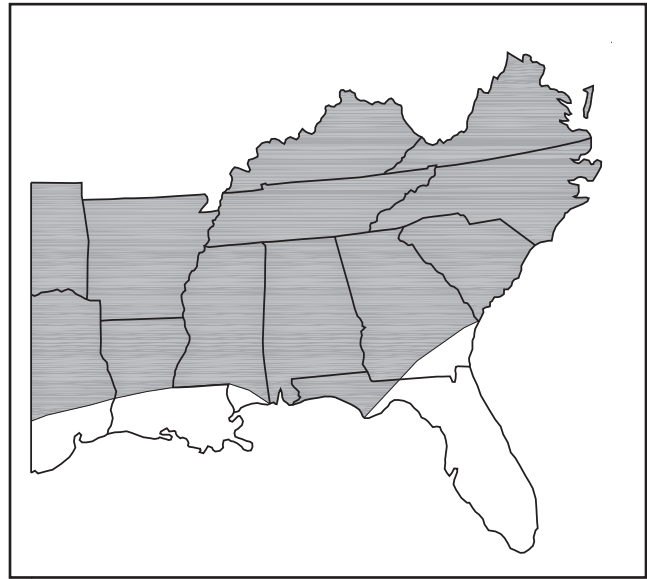


Figure 2. Distribution of *Lasionycteris noctivagans* in the South.

1951) indicating that the mother used the twig as a perch or roost. Although there have been several anecdotal reports of large maternity colonies of silver-haired bats (Barbour and Davis 1969), only two confirmed reports exist of small roosting groups (20 or fewer animals) that included young (Novakowski 1956, Parsons et al. 1986). Both of these roosts were located in tree cavities. The winter roosts of *L. noctivagans* are well known. During late fall, winter, and early spring, the bat roosts in caves (Beer 1956, Krutzsch 1966), mines (Layne 1958, Pearson 1962), rock crevices (Frum 1953), hollow trees (Jackson 1961), buildings, and other structures (Frum 1953, Bartsch 1956), and under loose bark (Cowan 1933, Tyler and Payne 1982). The silver-haired bat typically forages near forested habitats and adjacent to or over water-courses, ponds, or large water bodies (Davis and Hardin 1967, Kunz 1973, Barclay 1985, Brown 1986).

REPRODUCTION

The breeding behavior of the silver-haired bat remains to be described. Copulation occurs in fall and the females store the sperm until the following spring (Druecker 1972). Ovulation occurs late April–early May; following a 10-day period between ovulation and implantation, gestation lasts 50–60 days (Druecker 1972). Some females give birth to only 1 young but 2 is the usual litter size (Seton 1907, Turner and Jones 1968, Kunz 1971). Most births occur in June and July (Easterla and Watkins 1970, Kunz 1971) but pregnant bats are known from early August (Parsons et al. 1986). The period of lactation lasts approximately 36 days and the young are capable of flight soon after weaning (Kunz 1971). In central Iowa, Kunz (1971) found

flying young in late July. Young bats reach sexual maturity during their first summer (Druecker 1972).

FOOD HABITS

Lasionycteris noctivagans begins its foraging activity late in the evening (Seton 1907, Kunz 1973). However, the timing of its foraging activity may be influenced by the presence of other bat species. In areas where the eastern red bat (*Lasiurus borealis*) and hoary bat (*L. cinereus*) occur, the three species are active at different times (Seton 1907, Kunz 1973). The silver-haired bat may forage at later hours where the big brown bat (*Eptesicus fuscus*) is common (Reith, 1980) and earlier in areas where *E. fuscus* is rare (Whitaker et al. 1977). In Iowa, *L. noctivagans* exhibits two periods of feeding activity each night (Kunz 1973). The first period, which occurs 2–4 hours after sunset, is the major period of foraging. A second period of activity occurs 6–8 hours after sunset. Despite Black's (1974) assertion that the silver-haired bat is a "moth strategist," the species appears to be opportunistic in its feeding habits. Arthropods from many insect orders (e.g., Lepidoptera, Hemiptera, Coleoptera, Diptera, Trichoptera, and Isoptera) have been found in the stomachs or fecal remains of *L. noctivagans* (Gould 1955, Whitaker 1972, Jones et al. 1973, Black 1974, Whitaker et al. 1977).

ASSOCIATED SPECIES

The silver-haired bat occasionally forages in the same areas as the eastern pipistrelle (*Pipistrellus subflavus*), big brown bat, evening bat (*Nycticeius humeralis*), Townsend's big-eared bat (*Corynorhinus townsendii*), hoary bat, eastern red bat, little brown myotis (*Myotis lucifugus*), eastern small-footed myotis (*M. leibii*), and northern long-eared myotis (*M. septentrionalis*; Jones 1965, Turner and Jones 1968, Kunz 1973, Sheffield and Chapman 1990).

VULNERABILITY AND THREATS

Lasionycteris noctivagans is widely distributed in North America and appears to be abundant in the western and northern portions of its range. Although it may be relatively uncommon in the southern United States, there appear to be no threats to its existence.

MANAGEMENT SUGGESTIONS

Management actions center on the provision of high densities (21/ha) of large snags, maintenance of structural complexity in upland and riparian areas (Campbell et al. 1996), and the provision of roosting sites in mature forest (Bat Conservation International

2001). The bat roosts in hardwood cavities and under the peeling bark of small diameter shortleaf pines; management guidelines that retain trees with suitable peeling bark characteristics are beneficial (D. Saugey, USDA Forest Service, personal communication). The provision of forested corridors and diverse age structure is also helpful. Selective harvesting to maintain old, dominant trees may benefit the silver-haired bat, as well as low intensity prescribed burns to maintain open habitat. Because it roosts in hollow trees and caves, and feeds over or near water, cave protection and streamside management zones are beneficial to the species. Additional research is needed on winter roosting needs in the south.

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Eastern Red Bat

Lasiurus borealis (Müller, 1776)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The eastern red bat (*Lasiurus borealis*) originally was described as *Vespertilio borealis*. Although the species once was included in the genus *Nycteris*, Hall and Jones (1961) revised the lasiurine bats and assigned the species to its current genus following a 1914 ruling of the International Committee of Zoological Nomenclature in which the Law of Priority was suspended. Although this ruling favored the use of *Lasiurus* rather than *Nycteris*, some authors (Hall 1981) persisted in using the discarded taxon. Five subspecies are recognized by some authors (Hall 1981, Shump and Shump 1982), but one of the subspecies was elevated recently to specific status. Baker et al. (1988) suggested that *L. borealis* is monomorphic and the four other subspecies should be reassigned to *L. blossevillei*. Although Morales and Bickham (1995) confirmed the separation of the two species, the subspecific changes have not been uniformly accepted (Koopman 1993). Only one subspecies, *L. b. borealis*, occurs in the South. The natural history of the species is reviewed by Shump and Shump (1982).

DISTINGUISHING CHARACTERISTICS

Lasiurus borealis is a medium-sized, reddish-orange to chestnut-colored bat. Measurements are: total length, 108–125 mm; tail, 45–60 mm; hind foot, 8–10 mm; ear, 8–13 mm; forearm, 37–42 mm; weight 6–14 g. The hairs on the dorsum are white-tipped giving the pelage a frosted appearance; there are conspicuous yellowish-white patches at the shoulder and on the wing near the thumb. The venter usually is pale yellow or orange. White specimens occur but are rare (Rudd 2002). The ears are broad basally, short and rounded at the tip and the outer surfaces are partially furred. The tail membrane is heavily furred on the dorsal surface. The eastern red bat may be confused with the Seminole bat, *L. seminolus*, which it resembles in size and appearance. Pelage color is the most reliable characteristic to separate adult eastern red bats from adult Seminole bat specimens (Laerm et al. 1999). The dorsal pelage of the Seminole bat is a darker, mahogany brown to almost brownish-purple. The skull of the eastern red bat usually possesses a distinct lacrimal ridge whereas the skull of the Seminole bat often lacks a ridge. The dental formula is $I\ 1/3$, $C\ 1/1$, $P\ 2/2$, $M\ 3/3 = 32$ (Figure 1).

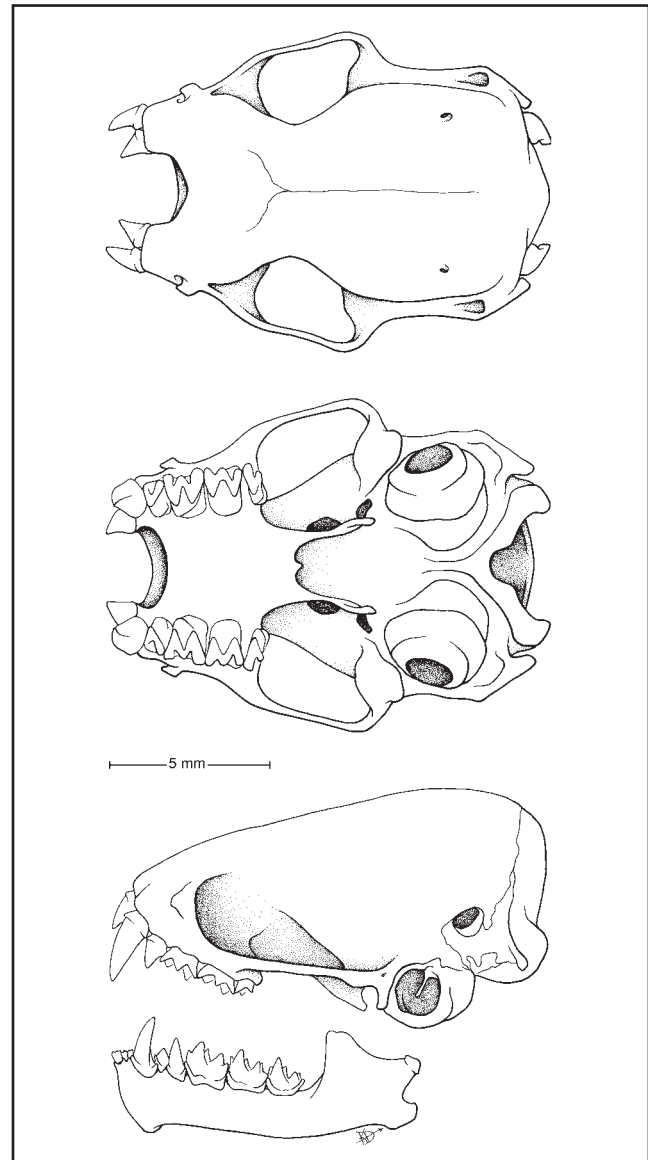


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Lasiurus borealis* from Giles County, Virginia (USNM 364633, male).

CONSERVATION STATUS

The eastern red bat has a global rank of Secure (NatureServe 2007). It is Secure in Alabama, Georgia, Kentucky, North Carolina, and Tennessee. It is Apparently Secure in Arkansas, Louisiana, Mississippi, Oklahoma, Texas and Virginia. It is unranked in Florida and South Carolina.

DISTRIBUTION

Lasiurus borealis is found from southern Canada southward throughout the central and eastern United States and northeastern Mexico (Hall 1981). The species has been recorded from all states in the South (Figure 2). The species is found throughout Louisiana but may not occur in coastal marshes west of the Mississippi River (Lowery 1974, Lance and Garrett 1997). The eastern red bat occurs statewide in Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989, Best and Caesar 2000), Alabama (Howell 1921, Pearson 1960), Georgia (Harper 1927, Constantine 1958, Pearson 1960, Golley 1962, Laerm et al. 1981, Menzel et al. 1998, 2000; Chapman et al. 2000, Johnson et al. 2002, Menzel et al. 2003), South Carolina (Golley 1966, Webster et al. 1985, Menzel et al. 2000, 2002), North Carolina (Lee et al. 1982, Webster et al. 1985), Virginia (Bailey 1946, Webster et al. 1985, Linzey 1998), Kentucky (Barbour and Davis 1974, Harvey et al. 1991, Hutchinson 1998, Hutchinson and Lacki 2000a,b; Tennessee (Linzey and Linzey 1971, Harvey et al. 1991, Kennedy 1991, Linzey 1995), Arkansas (Sealander and Heidt 1990, Saugey et al. 1998, Wilhide et al. 1998), Oklahoma (Caire et al. 1988, 1989), and eastern Texas (Schmidly et al. 1977, Schmidly 1983, 1991). It is absent from the southern half of the Florida peninsula (Jennings 1958, Pearson 1960, Zinn 1977, Hall 1981).

ABUNDANCE STATUS

The bat is regarded as common throughout the South. There are no population density estimates available in the region due to its solitary roosting habits. However, McClure (1942) estimated the density to be approximately 0.4 bats/ha in Iowa. Constantine (1966) found that there were more bats in the surrounding countryside than in the same Iowa town. Mumford (1973) estimated the population density of eastern red bats to be 1.0/ha in Indiana. Kunz (1973) observed that the species was uncommon in areas where the hoary bat (*L. cinereus*) was abundant.

The eastern red bat is migratory and moves southward in groups from Canada and the northern United States during September–October (Davis and Lidicker 1956, Banfield 1974). As a result, winter populations may increase in Kentucky but do not fluctuate noticeably in states to the south (Barbour and Davis 1974). Instead, some populations may make relatively small seasonal movements, which result in slightly denser populations to the south (Sealander and Heidt 1990). Northward migration occurs in March–April (Barbour and Davis 1969). Some bats hibernate during cold periods and become

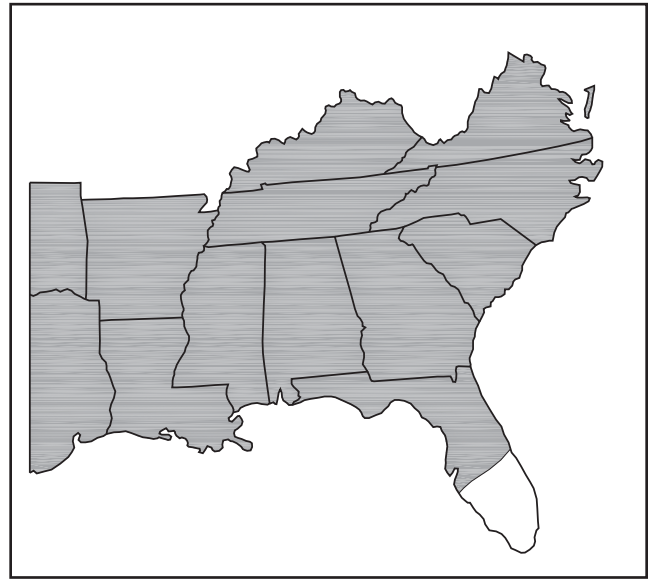


Figure 2. Distribution of *Lasiurus borealis* in the South.

active during temporary warm periods in winter (Gardner and McDaniel 1978, Koontz and Davis 1991). Davis and Lidicker (1956) observed the bat emerging from a woodlot in Illinois when the temperature was 19° C. Emergence was in late afternoon and most feeding activity ceased before sunset. Lewis (1940) found active eastern red bats in Virginia when the temperature rose to 13° C or above.

PRIMARY HABITATS

The eastern red bat is solitary and roosts primarily in trees or shrubs (Barbour and Davis 1969, Mumford 1973). Many species of trees are used as roosts (McClure 1942, Constantine 1966, Hart et al. 1993, Kurta et al. 1993, Hutchinson 1998, Menzel et al. 2000); hardwoods are used predominantly (Hutchinson 1998, Menzel et al. 1998, Hutchinson and Lacki 2001). Roost sites are usually 1–7 m above ground. Typical roost sites are high in the canopy near the outer edge of the foliage (Hutchinson 1998, Menzel et al. 1998). Roost sites share common characteristics: protection from view in all directions except from below; lack of obstruction below the roost; an absence of lower perches from which a potential predator could see the bat; dark cover beneath the roost; sufficient adjacent vegetation to reduce wind currents; and location on the southern or southwestern side of the tree (Constantine 1966, Menzel et al. 1998, Saugey et al. 1998, Mager and Nelson 2001). Eastern red bats in Kentucky seldom showed site fidelity to a single roost tree but used several roost trees within a small (<40 m²) core area (Hutchinson 1998). Trees with larger diameters are preferred for roost sites (Menzel et al. 1998, 2000) as are trees located >50 m

from any edge (Hutchinson 1998). Other roost locations include clumps of Spanish moss (*Tillandsia usneoides*; Constantine 1958), leaf litter (Mager and Nelson 2001), grass (Mager and Nelson 2001), the undersides of sunflower (*Helianthus* spp.) leaves (Downes 1964), and under the shingles of houses (Mager and Nelson 2001). Some *L. borealis* occasionally enter caves (Quay and Miller 1955, Myers 1960, Kurta 1980, McDaniel and Gardner 1978, Saugey et al. 1978, Best and Caesar 2000). The winter hibernacula of the eastern red bat are poorly known. Fassler (1975) found one torpid individual in an abandoned woodpecker hole on a cold day in March. For many years, foresters observed that eastern red bats commonly were “smoked” from their hibernacula during winter prescribed burning (Saugey et al. 1989). Recently, Saugey et al. (1998) reported that some individuals winter-roost beneath leaf litter on the forest floor.

REPRODUCTION

Copulation takes place during flight (Saugey et al. 1998) between August–September; fertilization does not occur until spring (Stuewer 1948, Layne 1958, Glass 1966, Hamilton and Whitaker 1979, Caire et al. 1988). After a gestation period of 80–90 days (Jackson 1961), the young are born in May–July (Kunz 1971, Mumford 1973, Saugey et al. 1988). The number of young/litter ranges from 1–5 (Hamilton and Stalling 1972, Mumford 1973) and the mean number of young/litter is 2.3 (McClure 1942, Constantine 1966, Birney and Rising 1968). The young are nursed 38 days (Kunz 1971) and are capable of independent flight 3–6 weeks after birth. Development of the young is inadequately studied; however, information is provided by Jackson (1961), Barbour and Davis (1969), Kunz (1971), Whitaker and Mumford (1972), and Hamilton and Whitaker (1979).

FOOD HABITS

The eastern red bat begins foraging activities 1–2 hours after sunset and some activity occurs throughout the night (Kunz 1973, Lacki and Bookhout 1983, Caire et al. 1988, Carter et al. 1999). Barbour and Davis (1969) observed that initial evening flights occur at high altitudes; after 15–30 minutes, the bats descend to feed at levels from treetop to ground. Davis (1974) and Jackson (1961) noted that eastern red bats use the same foraging flight pathway each evening. The echolocation calls of the eastern red bats may attract other bats to the feeding area (Caire et al. 1988). Eastern red bats in the Coastal Plain of South Carolina foraged in both cluttered and uncluttered habitats, however foraging activity is overwhelmingly concentrated over wetland, riparian, and bottomland habitats (Menzel et al. 2005a,b). Individual bats apparently

respond to the echolocation calls of conspecifics, identifying areas where prey is available (Hickey and Fenton 1990). This results in large, concentrated groups within a feeding area (LaVal and LaVal 1979); there is no consistent evidence for temporal resource partitioning among conspecifics (Hickey et al. 1996). Resource partitioning does occur when a related species such as the hoary bat is present (Hickey et al. 1996). Whitaker (1972) analyzed the stomach contents of 128 eastern red bats in Indiana and found that 28% of the contents were comprised of Lepidoptera. Prey includes beetles (Coleoptera), true bugs (Hemiptera), flies (Diptera), wasps (Hymenoptera), butterflies (Lepidoptera), and crickets (Orthoptera; Lewis 1940, Ross 1967, Connor 1971, Mumford 1973, Hamilton and Whitaker 1979, Acharya and Fenton 1992, Hickey et al. 1996). Other prey includes agricultural pests (Feldhamer et al. 1995). Foraging occurs over many substrates and elevations (Lacki and Bookhout 1983, Schmidly 1991). The eastern red bat sometimes feeds around insect light traps, street lamps, or floodlights (Wilson 1965, Hamilton and Whitaker 1979, Shump and Shump 1982, Hickey et al. 1996).

ASSOCIATED SPECIES

The eastern red bat roosts solitarily but it often forages in close proximity to other species including the evening bat (*Nycticeius humeralis*), eastern pipistrelle (*Pipistrellus subflavus*), big brown bat (*Eptesicus fuscus*), Seminole bat, and hoary bat (Barbour and Davis 1969, Kunz 1973, Sheffield and Chapman 1992, Menzel et al. 2005b).

VULNERABILITY AND THREATS

Although this species is widely distributed and highly migratory, little is known of its population status in the region. However, there appears to be no threats to the survival of the species at the present time.

MANAGEMENT SUGGESTIONS

Forest management strategies that promote long rotations, complex canopy structure, and snag formation are beneficial for roost provision (Menzel et al. 2000). Crampton and Barclay (1995) suggest retention of old growth patches, forest connectivity, and increased forest edge during timber harvest. Kern and Humphrey (1995) found that pine plantations had little habitat value for bats, possibly due to the lack of roosting substrate (broadleaf foliage, tree cavities) and less productive insect resource base. The red bat may hibernate in leaf litter (Moorman et al. 1999) and pine-hardwood litter (Saugey et al. 1998, 1989). Careful timing of prescribed fire activity minimizes mortality in areas where the bat hibernates on the ground.

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Hoary Bat

Lasiurus cinereus (Palisot de Beauvois, 1796)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The hoary bat (*Lasiurus cinereus*) was originally described as *Vespertilio cinereus*. Allen (1864) was the first to use the current scientific name. *Lasiurus* was approved for use by a 1914 ruling of the International Committee on Zoological Nomenclature in which the Law of Priority was suspended. Hall and Jones (1961) followed this ruling when they revised the genus *Lasiurus*, but some authors (Hall 1981) use another generic name, *Nycteris*. The accepted scientific name for the hoary bat is *L. cinereus* (Jones et al. 1992). There are three recognized subspecies (Shump and Shump 1982); one (*L. c. cinereus*) occurs in the South. The literature on this species is reviewed by Shump and Shump (1982).

DISTINGUISHING CHARACTERISTICS

The hoary bat is the largest bat in the region. Measurements are: total length, 133–150 mm; tail, 48–52 mm; hind foot, 11–14 mm; ear, 17–20 mm; forearm, 52–57 mm; weight, 20–35 g. The dorsal pelage of the hoary bat varies from yellowish-brown to mahogany; the tips of the hairs are silvery-white giving the bat a frosted appearance. A distinct collar of yellow or buffy fur is present from the throat to the ears and the venter is buffy-white. A buff-colored spot is present at the wrist and a yellow patch is present at the shoulder. The wing lining appears yellowish. The ears are short, rounded, and furred externally. The wings are long and pointed and the tail membrane is covered in fur dorsally. The dental formula is $I\ 1/3, C\ 1/1, P\ 2/2, M\ 3/3 = 32$ (Figure 1).

CONSERVATION STATUS

The hoary bat has a global rank of Secure (NatureServe 2007). It is Secure in Tennessee and Apparently Secure in Louisiana, Georgia and Texas. Arkansas, Mississippi and Oklahoma classify it as Vulnerable. North Carolina classifies it as Critically Imperiled. It is unranked in South Carolina and Alabama, and it is Under Review in Florida and Virginia. The bat is classified as Not Applicable by Kentucky (i. e., the species is not considered a suitable target for conservation activities).

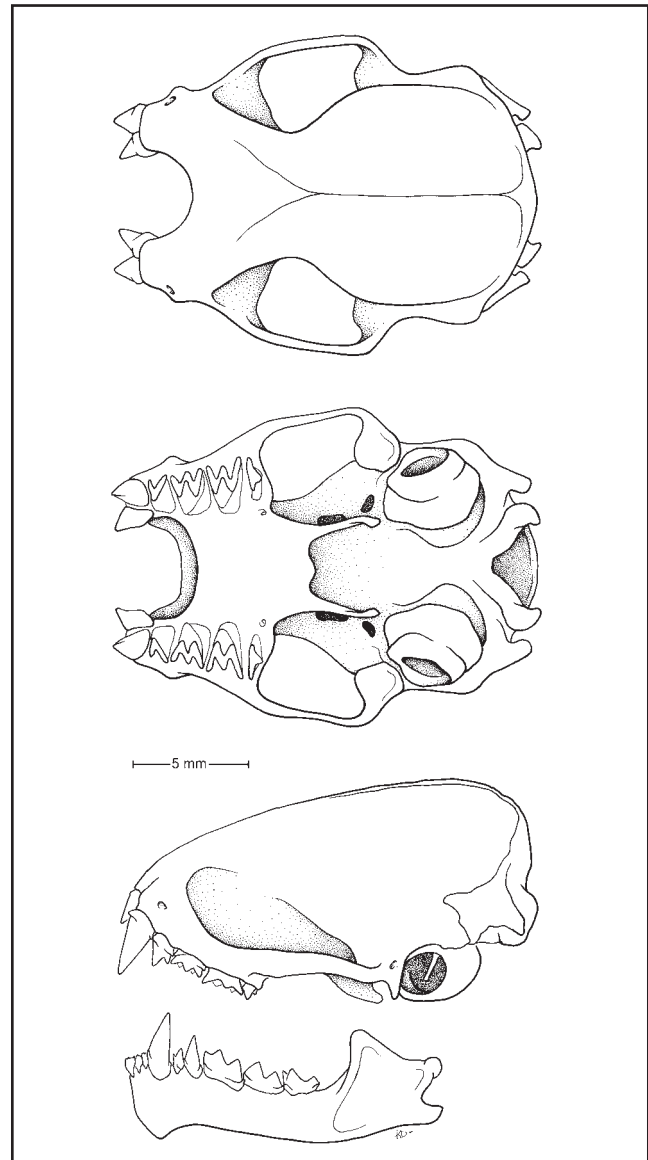


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Lasiurus cinereus* from Beachton, Florida (USNM 276572, female).

DISTRIBUTION

The hoary bat occurs in all 48 contiguous states of the United States as well as Hawaii and is widely distributed in Canada and Mexico (Shump and Shump 1982). The distribution pattern of the bat in the South

is confounded by the migratory behavior of the species. There is evidence that the bat migrates southward during the fall (Merriam 1887, Allen 1940, Findley and Jones 1964). Zinn and Baker (1979) report that the hoary bat migrates southward in groups through northern Florida in October and November, returning northward during February through May. Although Findley and Jones (1964) and Jones et al. (1977) suggest that the hoary bat winters in areas south of the United States, specific wintering locations and habitats are unknown and migratory routes are not documented. Furthermore, some hoary bats apparently remain in portions of their summer range and hibernate during the coldest winter months (Whitaker 1967, Barbour and Davis 1969, Whitaker and Mumford 1972, Lowery 1974). The species is recorded from all states in the region (Figure 2). Collection records from the summer months are rare in some parts of the South (Bailey 1946, Webster 1985), but acoustical surveys indicate their uncommon but widespread presence (Menzel et al. 2005b, Ford et al. 2006).

ABUNDANCE STATUS

Although the hoary bat may be more common in the Great Plains states than in the eastern portion of the country, surveys involving mist-netting have shown it to be more common in some southern states than previously suspected (Baker and Ward 1967, Caire et al. 1986, Sealander and Heidt 1990, Menzel et al. 2003a). Population density estimates are not available for any southern state, but it is regarded as locally common in most states where it is known to occur. Competitive exclusion may limit its abundance in areas where the eastern red bat (*L. borealis*) is common. The hoary bat is rare when the eastern red bat is abundant, and eastern red bats are rare in areas of hoary bat abundance (Kunz 1973). Findley and Jones (1964) and several other authors suggest that male and female bats may segregate sexually during the breeding season, but Caire et al. (1986) indicated that this pattern may not exist.

PRIMARY HABITATS

The hoary bat is a solitary, tree-roosting species (Shump and Shump 1982). Most roost sites are located in trees near the edge of clearings (Barclay 1984). The hoary bat roosts on leaf-concealed branches that are 3–5 m above ground and with open space underneath (Constantine 1966). Unusual roost locations include an eastern gray squirrel (*Sciurus carolinensis*) nest (Neill 1952), underneath a driftwood plank (Connor 1971), on the side of a building (Bowers et al. 1968), and in a woodpecker hole (Cowan and Guiguet 1965). The hoary bat occasionally occupies

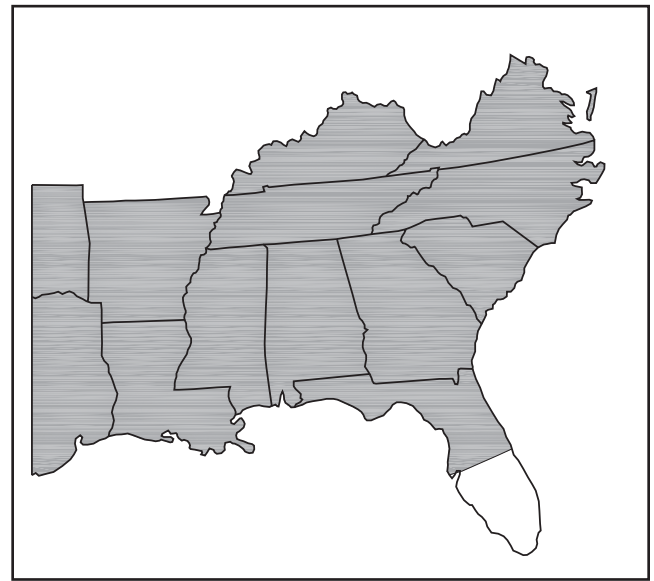


Figure 2. Distribution of *Lasiurus cinereus* in the South.

caves (Mumford 1953, Beer 1954, Grove 1974), but Myers (1960) suggests that most individuals that roost in a cave cannot find their way out and eventually die. Foraging begins in late evening. The peak activity period is 3–6 hours after sunset but it sometimes emerges before sunset on warm evenings (Kunz 1973). The hoary bat is a strong flier and may forage at considerable distances from roost locations (Black 1974). In urban locations, the hoary bat may feed around streetlights (Hickey 1992, Hickey et al. 1996). Because of its tendency to forage in open areas above vegetation, Black (1974) classified *L. cinereus* as an “open-air forager.” Recent research has confirmed that hoary bats forage primarily in open, uncluttered habitats such as fields, early-successional area water courses, and wetlands throughout the region (Ford et al. 2005, Menzel et al. 2005a, Menzel et al. 2005b). Foraging in forested habitats often occurs above the canopy (Menzel et al. 2005b) due to the species high wing loading and low echolocation characteristics (Menzel et al. 2003b). Caire et al. (1986) collected large numbers of this species by using elevated mist nets placed over and adjacent to streams.

REPRODUCTION

The breeding behavior of the hoary bat is poorly known. It may copulate before or during autumnal migration, or on the wintering grounds (Finley and Jones 1964, Bouchard et al. 2001). Implantation may be delayed until early spring, but Bouchard et al. (2001) report a pregnant bat in December. Most births are reported from mid-May through early July; litter size ranges from 1–4 with 2/litter the most common occurrence (Wood 1922, Provost and Kirkpatrick

1952, Jackson 1961, Gottschang 1966, Barbour and Davis 1969, Druker 1972, Whitaker and Mumford 1972). Although the post-natal growth rate of the hoary bat is slow (Koehler and Barclay 2000), volant young are present in early July (McClure 1942). Details of parturition and development of the young are provided by Munyer (1967) and Bogan (1972).

FOOD HABITS

The food habits of the hoary bat are poorly known. It apparently preys on only a few kinds of insects, which may indicate that it is a food specialist. It exhibits a preference for Lepidoptera (moths), which it captures in flight (Ross 1967, Black 1972, Hickey et al. 1996). Only the abdomen and thorax of a moth are ingested; the wings and head are discarded. The bat also feeds upon beetles, grasshoppers, termites, dragonflies, flies, and wasps (Dalquest 1953, Ross 1967, Zinn and Baker 1979, Rolseth et al. 1994). Whitaker (1967) found grass leaves and a snakeskin in the stomach of a hibernating hoary bat in Indiana. This may indicate that the hoary bat occasionally forages on the ground and may not be selective during the colder months. There are accounts (Bishop 1947, Orr 1950) of a hoary bat attacking an eastern pipistrelle (*Pipistrellus subflavus*), but in neither case was predation documented.

ASSOCIATED SPECIES

The hoary bat roosts solitarily but sometimes forages in a group of conspecifics. The hoary bat feeds near the big brown bat (*Eptesicus fuscus*), Indiana myotis (*Myotis sodalis*), silver-haired bat (*Lasionycteris noctivagans*), evening bat (*Nycticeius humeralis*), eastern red bat, and eastern pipistrelle (Mumford 1969).

VULNERABILITY AND THREATS

Although this species is widely distributed, little is known about its population status, habitat requirements, and migratory behavior. There appear to be no threats to the survival of the species in the South.

MANAGEMENT SUGGESTIONS

The maintenance of large trees and small openings within forested habitats benefits the hoary bat (Bat Conservation International 2001). Young clearcut stands and open bogs/beaver ponds are important foraging habitat (Francl et al. 2004, Owen et al. 2004, Ford et al. 2005). Additional research is needed on population trends, roosting habits, hibernation requirements, and habitat preferences.

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Hoary Bat (*Lasiurus cinereus*)

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Northern Yellow Bat

Lasiurus intermedius (H. Allen, 1862)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The northern yellow bat was described originally as *Lasiurus intermedius* based upon a specimen from Mexico. However, when Miller (1902) described a new species from Florida, he placed both *L. intermedius* and the new species (*Dasypterus floridanus*) into the genus *Dasypterus*. Tate (1942) recognized the hoary and red bats in the genus *Lasiurus*, and the yellow bats in the genus *Dasypterus*. Hall and Jones (1961) considered the two species as conspecific and relegated them to subspecific status in the genus *Lasiurus* following Handley (1960). The use of *Lasiurus* (rather than *Nycteris*) followed a 1914 ruling of the International Committee on Zoological Nomenclature (ICZN) in which the Law of Priority was suspended. Recently, the generic placement of the yellow bat again was a source of debate. Although Hill and Harrison (1987) reinstated the genus *Dasypterus* for the yellow bat, evidence indicates that all the yellow bats should be included in *Lasiurus* (Baker and Patton 1967, Bickham 1979, 1987; Baker et al. 1988, Jones et al. 1992, Morales and Bickham 1995). Three subspecies of *Lasiurus intermedius* currently are recognized: one (*L. i. floridanus*) occurs in the South. The literature on *L. intermedius* is reviewed by Webster et al. (1980).

DISTINGUISHING CHARACTERISTICS

Lasiurus intermedius is a relatively large bat with predominantly yellow pelage. Measurements are: total length, 121–132 mm; tail, 51–60 mm; hind foot, 8–11 mm; ear, 15–17 mm; forearm, 45–52 mm; weight, 15–19 g. The dorsal pelage is silky; color varies from yellow to yellowish-brown to yellowish-gray. The ventral pelage is light yellow; the wing membranes are brown. The northern yellow bat does not have light-colored patches at the shoulder or wrist. Unlike other species of *Lasiurus* in the region, the tail membrane is heavily furred dorsally only on the proximal half and the ears are pointed. The dental formula is $I\ 1/3, C\ 1/1, P\ 2/2, M\ 3/3 = 32$ (Figure 1).

CONSERVATION STATUS

The northern yellow bat has a global rank of Apparently Secure (NatureServe 2007). Louisiana and Texas classify it as Apparently Secure, but Georgia and Mississippi classify it as Imperiled. It is Critically

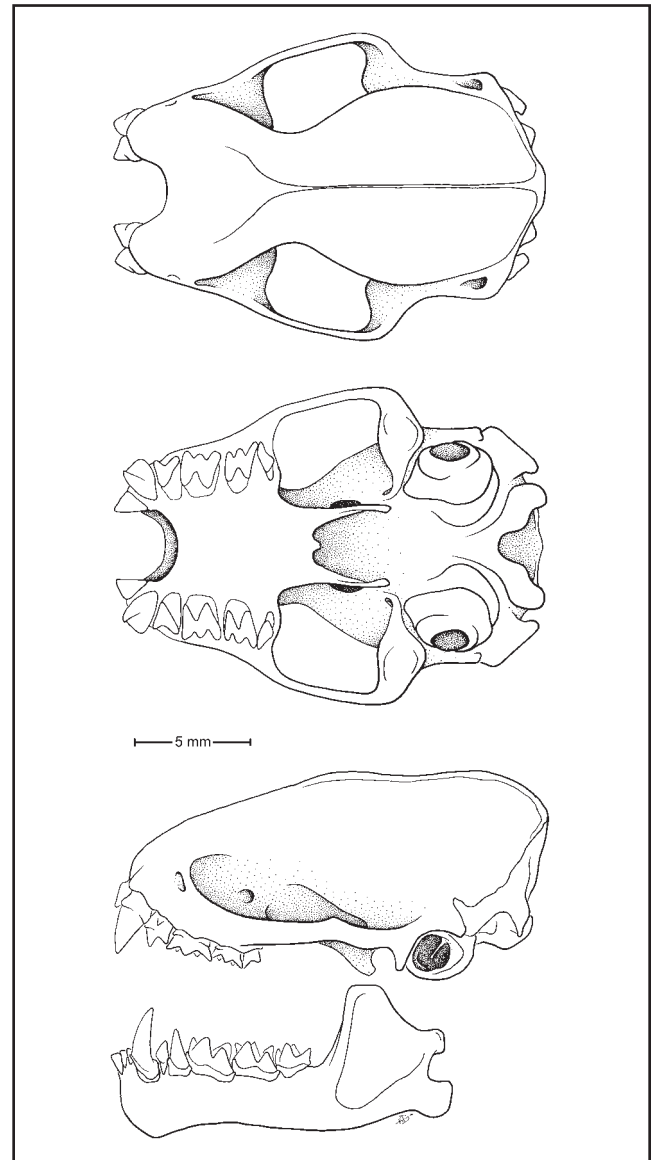


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Lasiurus intermedius* from Seven Oaks, Florida (USNM 230802, female).

Imperiled in Alabama. It is unranked in Florida and South Carolina and Under Review in North Carolina.

DISTRIBUTION

The northern yellow bat is known from the coastal United States from South Carolina to southern Texas (Figure 2). It occurs primarily in the lowland prairie

and marsh areas in Texas but has been collected in inland wooded habitats (Schmidly 1991). The northern yellow bat is found in the Coastal Plains of Louisiana (Lowery 1974), Mississippi (Wolfe 1971, Kennedy et al. 1974), Georgia (Golley 1962, Krishon et al. 1997, Carter et al. 1998, Chapman et al. 2000), and South Carolina (Golley 1966, Webster et al. 1985). It is the dominant bat species on dry upland sites in the central peninsula of Florida and is found throughout the state (Sherman 1939, 1944; Jennings 1958, Zinn 1977, Humphrey 1992). The species may be resident in the Coastal Plains of North Carolina (Lee et al. 1982, Linzey 1998) and southeastern Virginia (Handley 1991, Linzey 1998). Extralimital records of the northern yellow bat have come from as far north as Norfolk County, Virginia (Rageot 1955) and Union County, New Jersey (Koopman 1965).

ABUNDANCE STATUS

The population status of the northern yellow bat is virtually unknown. It is regarded as the most abundant bat in the central region of Florida (Humphrey 1992), but its status is not adequately assessed elsewhere in its range. Schmidly et al. (1977) regards the species as rare in eastern Texas based upon a paucity of specimen records from the area.

PRIMARY HABITATS

The northern yellow bat typically inhabits wooded areas near permanent water. In the southern United States, it occupies both coniferous and deciduous forests but its roost sites are poorly known. In the South, *L. intermedius* is regarded as a solitary rooster, but many individuals aggregate in a single tree (Jennings 1958). Barbour and Davis (1969) suggest that the range of the northern yellow bat coincided somewhat with the range of Spanish moss (*Tillandsia usneoides*). The species may use a variety of other roosts as demonstrated by the occurrence of bat roosting on dried corn (*Zea mays*) stalks (Baker and Dickerman 1956). It roosts in clumps of Spanish moss within live oak (*Quercus virginiana*) trees (Jennings 1958), among palm (*Sabal* spp.) fronds (Davis 1970, 1974; Spencer et al. 1988), and in pine-oak woodlands (Sherman 1944). Foraging habitat preferences of *L. intermedius* are poorly known, although it is regarded as a high-flying species. According to Barbour and Davis (1969) and Schmidly (1983), the northern yellow bat usually forages 5–7 m above the ground. Krishon et al. (1997) estimated a home range of 10.5 ha for one northern yellow bat on Sapelo Island, Georgia. It forages over open areas (e.g., croplands, pastures, airports, golf courses, marshes, lake margins) and above forest openings (Jennings 1958, Zinn 1977, Schmidly 1983, Krishon et al. 1997). Moore (1949)

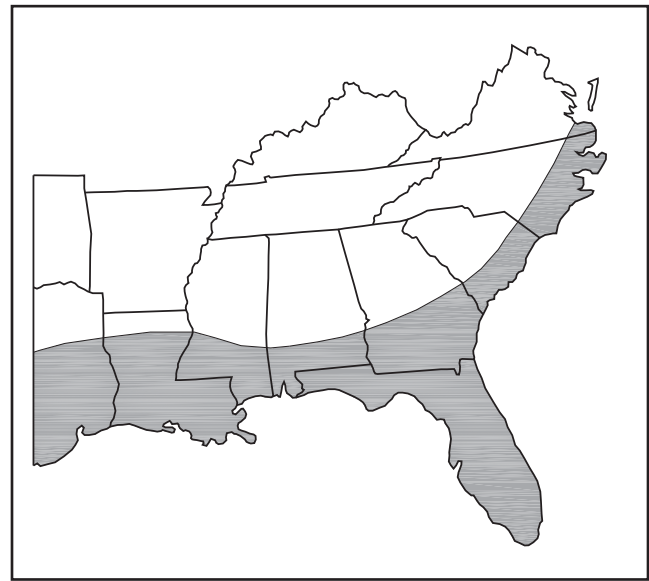


Figure 2. Distribution of *Lasiurus intermedius* in the South.

reported the bat foraging over piles of sawdust in Florida. Many specimens in museum collections were taken in mist nets set over water (Webster et al. 1980). Lowery (1974) reported *L. intermedius* foraging over the Mississippi River. The winter habits of the species are poorly known. The species has been captured throughout the year in southern Louisiana (Lowery 1974) and Florida (Jennings 1958); it may hibernate or migrate in other areas. Golley (1966) refers to January records of the species in South Carolina, which indicate that the species may overwinter there. Schmidly (1983) suggests that the northern yellow bat may migrate out of eastern Texas during winter.

REPRODUCTION

The breeding behavior of the northern yellow bat is unknown. Mating may occur in fall; fertilization is delayed until spring (Webster et al. 1980, Schmidly 1983). Females bear 2–3 young in May–June (Barbour and Davis 1969). Adult females and young are found in roosting and feeding aggregations during June–August (Barbour and Davis 1969, Davis 1974).

FOOD HABITS

The food habits of the northern yellow bat are known from few samples. Sherman (1939) reported that prey includes damselflies (Odonata), flies (Diptera), bark and other beetles (Coleoptera), and flying ants (Hymenoptera). Carter et al. (1998) examined fecal pellets from two bats captured on Sapelo Island, Georgia. The pellets were composed of Hymenoptera (69%) and Coleoptera (31%) fragments. Zinn (1977)

also found that the diet of the northern yellow bat included many hymenopterans and coleopterans.

ASSOCIATED SPECIES

The northern yellow bat is normally a solitary roosting species and does not associate with other bats. Because it commonly uses Spanish moss or trees as roosting habitats, it may occupy the same trees as the Seminole bat (*L. seminolus*), eastern red bat (*L. borealis*), hoary bat (*L. cinereus*), silver-haired bat (*Lasionycteris noctivagans*), and evening bat (*Nycticeius humeralis*). It uses the same roosts as the southern yellow bat (*L. ega*) in southern Texas (Spencer et al. 1988).

VULNERABILITY AND THREATS

Little is known about the population status and habitat requirements of the bat; its vulnerability is difficult to assess. The loss of sandhill and oak hammock habitats to residential development and the planting of citrus groves may seriously impact the species (Humphrey 1992). A former threat to the species, the harvest of Spanish moss, may have been reduced by the development of synthetic materials.

MANAGEMENT SUGGESTIONS

Protection of roosting areas in Spanish moss is beneficial (Bat Conservation International 2001). The practice of removing old palm fronds eliminates roosting habitat; removal should be avoided in spring when young of the year are present. Pesticide use should be investigated in areas of population decline. Additional research is needed on foraging habits.

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Seminole Bat

Lasiurus seminolus (Rhoads, 1895)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The Seminole bat (*Lasiurus seminolus*) originally was described as *Atalapha borealis seminola*, a subspecies of the eastern red bat (*L. borealis*). Although Koopman (1993) regards the Seminole bat as a subspecies of *L. borealis*, the most widely accepted interpretation recognizes *L. seminolus* as a separate species (Poole 1932, 1949; Barkalow 1948, Coleman 1950, Davis 1957, Lowery 1974, Bickham 1979, 1987; Hall 1981, Baker et al. 1988, Jones et al. 1992, Morales and Bickham 1995). At the generic level, the use of *Lasiurus* resulted from a ruling of the International Committee on Zoological Nomenclature (ICZN) in which the Law of Priority was suspended. Hall and Jones (1961) and subsequent authors followed the ICZN ruling, using *Lasiurus* for all bats previously described as *Atalapha* and *Nycteris*. The Seminole bat is a monotypic species. The literature is reviewed by Shump and Shump (1982) and Wilkins (1987).

DISTINGUISHING CHARACTERISTICS

The Seminole bat is a medium-sized bat with a strong resemblance to the eastern red bat. Measurements are: total length, 90–110 mm; tail, 35–50 mm; hind foot, 8–10 mm; ear, 10–13 mm; forearm, 37–43 mm; weight, 8.5–12.5 g. The Seminole bat can be distinguished from the eastern red bat by pelage color and a diagnostic skull feature (Laerm et al. 1999). The Seminole bat has a rich, mahogany brown pelage and a poorly defined lacrimal shelf (Wilkins 1987), whereas the eastern red bat is brick red to rusty red and possess a strongly developed lacrimal shelf (Shump and Shump 1982). Both species have distinct white spots at the shoulder and wrist, short, rounded ears, and a heavily furred tail membrane. Both species exhibit a frosted appearance dorsally. The dental formula is $I\ 1\frac{1}{3}, C\ 1\frac{1}{1}, P\ 2\frac{2}{2}, M\ 3\frac{3}{3} = 32$ (Figure 1).

CONSERVATION STATUS

The Seminole bat has a global rank of Secure (NatureServe 2007). It is Secure in Georgia and Apparently Secure in Louisiana and Alabama. North Carolina, and Texas classify it as Vulnerable. It is Critically Imperiled in Arkansas and Oklahoma. It is unranked in Florida, Mississippi, and South Carolina.

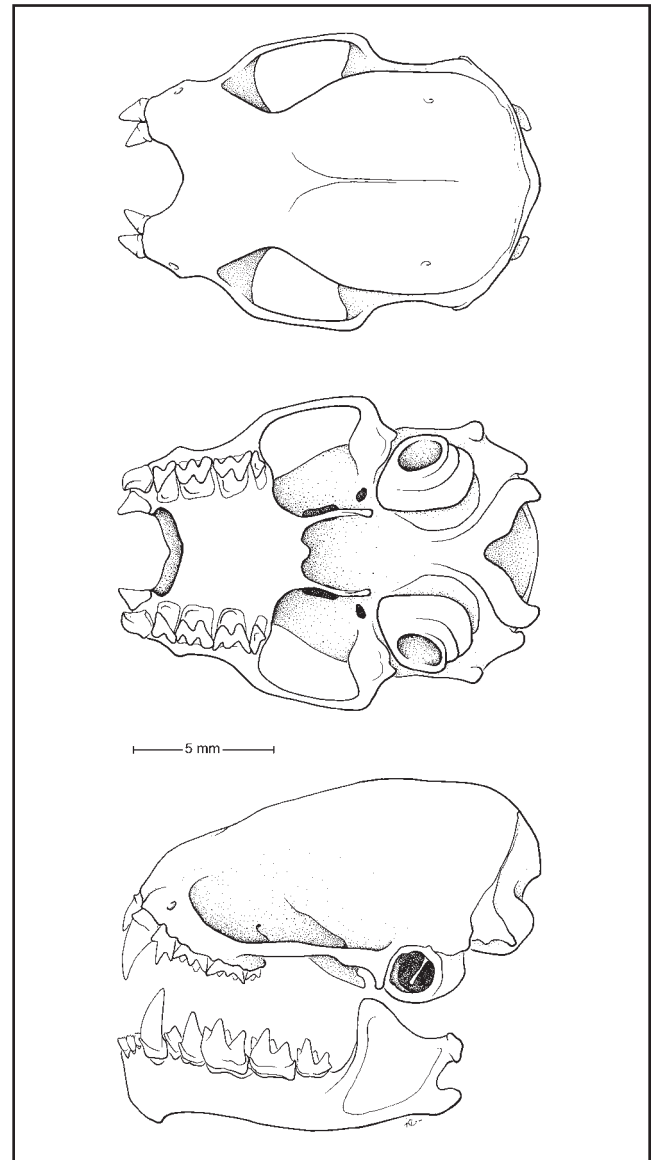


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Lasiurus seminolus* from Taylor County, Florida (USNM 234388, male).

DISTRIBUTION

Figure 2 shows that the range of the Seminole bat includes most portions of the South that border the Gulf of Mexico and the Atlantic Ocean from eastern Texas to North Carolina (Wilkins 1987). Barbour and Davis (1969) suggested that the range of this bat coincided largely with the range of Spanish moss (*Tillandsia*

usneoides), frequently used as a roost site. In Texas, the Seminole bat is found in the eastern forested areas (Davis 1974, Schmidly et al. 1977, Schmidly 1983, 1991). The species occurs throughout Louisiana (Lowery 1974, Lance and Garrett 1997, Lance and Rogowski 1999), Mississippi (Wolfe 1971, Kennedy et al. 1974), Alabama (Barkalow 1948, Barbour and Davis 1969), Georgia (Constantine 1958, Golley 1962, Krishon et al. 1997, Carter et al. 1998, Menzel et al. 1998, Chapman et al. 2000), Florida (Jennings 1958, Layne 1974), and South Carolina (Golley 1966, Neuhauser and DiSalvo 1972, Webster et al. 1985, Menzel et al. 1998, 2002, 2003) at elevations below 500 m. It is common in the lower Piedmont and Coastal Plain of North Carolina (Barkalow and Adams 1955, Barkalow and Funderburg 1960, Webster et al. 1985), and has been occasionally observed in the mountains (Webster 2000). The Seminole bat also occurs in southern Tennessee (Kennedy et al. 1984, Harvey et al. 1991), central Arkansas (Sealander and Hoiberg 1954, Heath et al. 1983, Saugey et al. 1989, Wilhide et al. 1998), and southeast Oklahoma (Glass 1958, Caire and Thies 1987, Caire et al. 1989). There is one record of the species from Virginia (Padgett 1987). Extralimital records exist from Pennsylvania (Poole 1932, 1949), New York (Layne 1955), western Texas (Lee 1987), and extreme southern Texas (Hall 1981). Individuals of this species apparently wander when young; this tendency may account for unusual occurrences outside the known range (Barbour and Davis 1969). Despite Barkalow's (1948) contention that the Seminole bat is migratory, the species is regarded as a year-round resident in Texas (Schmidly 1991), Louisiana (Lowery 1974), Georgia (Constantine 1958, Golley 1962), South Carolina (Coleman 1950), and Florida (Moore 1949, Jennings 1958). Constantine (1958) demonstrated that the species does not enter into deep hibernation, but may become torpid during cold periods. It may become active on warm winter evenings, but requires a higher temperature for activity than the eastern red bat.

ABUNDANCE STATUS

No population density estimates are available, but many authorities believe that the Seminole bat is abundant in suitable habitat (Lowery 1974, Barbour and Davis 1969, Webster et al. 1985, Schmidly 1991).

PRIMARY HABITATS

The Seminole bat occurs in forested habitats where it roosts in clumps of Spanish moss (Constantine 1958, Barbour and Davis 1969, Krishon et al. 1997); tree branches (Barkalow and Adams 1955, Menzel et al. 2000a,b); clumps of foliage (Sealander and Heidt 1990, Krishon et al. 1997, Menzel et al. 2000a,b); and

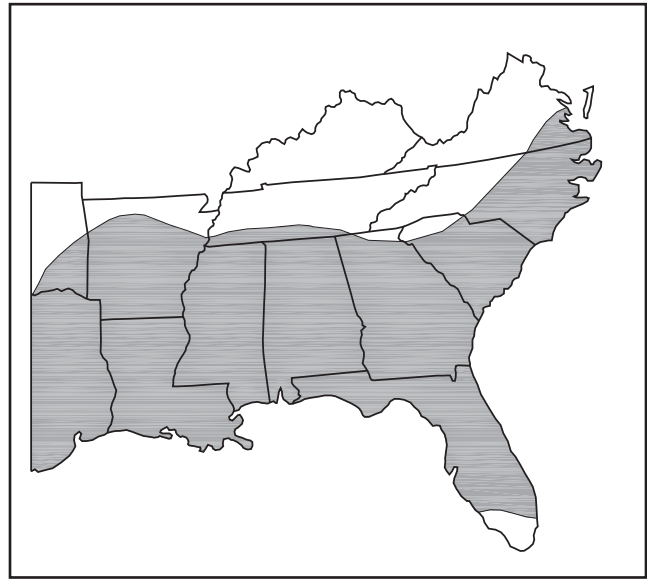


Figure 2. Distribution of *Lasiurus seminolus* in the South.

beneath loose bark (Sealander 1979). Menzel et al. (1998, 2000a) found that most roosts were located in large diameter pine (*Pinus* spp.) overstory. *Lasiurus seminolus* is a solitary bat and its roosts are occupied by individuals or by females with young. Constantine (1958) observed that the bat usually selects roosts from 1.1–4.5 m above the ground with a southwest exposure. One bat was captured near the entrance to an abandoned mine shaft in Arkansas, suggesting that the species opportunistically roosts in mines or caverns (Heath et al. 1983). In eastern Texas, *L. seminolus* occurs in pine-oak (*Pinus-Quercus*) and longleaf pine (*P. palustris*) habitat. It also is found in lesser numbers in oak-hickory (*Quercus-Carya*) forests (Schmidly et al. 1977). In Florida, the Seminole bat occupies a wide range of habitat including pine flatwoods, scrubby flatwoods, hammocks, lowland forests, and river swamps (Moore 1949, Jennings 1958, Ivey 1959, Zinn 1977). In the Okefenokee Swamp, Laerm et al. (1980) found *L. seminolus* foraging in uplands, prairies, shrub swamps, black gum (*Nyssa* spp.) forests, and bald cypress (*Taxodium distichum*) forests. Lee et al. (1982) collected Seminole bats from sand ridges where longleaf pine and turkey oak (*Q. laevis*) were dominant.

REPRODUCTION

Mating behavior has not been described for the Seminole bat. Mating may occur in early spring. Most young are born in early summer. Males taken in Georgia during February and April were found with descended testes (Constantine 1958). Pregnant females were collected during May in South Carolina (Coleman 1950) and Alabama (Barkalow 1948). Pregnant females

have been found in May and mid-June in Florida (Moore 1949, Jennings 1958). Lactating females have been found in June and early July (Moore 1949, Barkalow and Funderburg 1960). Females carry up to 4 embryos but usually give birth to 2 young (Moore 1949, Jennings 1958, Barbour and Davis 1969).

FOOD HABITS

The Seminole bat feeds at the treetop level over pine barrens, forest clearings, watercourses, coastal prairies, and hammocks (Harper 1927, Menzel et al. 2002, Menzel et al. 2005a,b). It occasionally gleans insects from foliage (Barbour and Davis 1969). Prey includes Hemiptera (true bugs), Diptera (flies), Coleoptera (beetles), and Hymenoptera (bees; Harper 1927, Sherman 1939, Zinn 1977, Carter et al. 1998). Sherman (1935) collected a Seminole bat that had a flightless cricket (*Gryllus assimilis*) in its mouth indicating that the bat may occasionally forage on the ground.

ASSOCIATED SPECIES

The Seminole bat normally roosts alone and does not associate with other species. Because it sometimes uses Spanish moss or trees as roosts, it may occupy the same roosting areas as the eastern red bat, northern yellow bat (*L. intermedius*), hoary bat (*L. cinereus*), silver-haired bat (*Lasionycteris noctivagans*), and evening bat (*Nycticeius humeralis*). It may use the same roost sites as *L. intermedius* and *L. borealis*, but on different days (Constantine 1958). It frequently forages in the same areas as the eastern red bat (Barkalow 1948).

VULNERABILITY AND THREATS

Lasiurus seminolus is widely distributed in the South and appears common throughout most of its range. It may be the most abundant summer bat in portions of Florida and southern Georgia. Although it may be uncommon in higher elevations and the periphery of its range, there appear to be few threats to its survival.

MANAGEMENT SUGGESTIONS

Forest management strategies that promote long rotations, complex canopy structure, and snag formation are beneficial for roost provision (Menzel et al. 2000a). Kern and Humphrey (1995) found that pine plantations had little habitat value for bats due to the lack of roosting substrate (broadleaf foliage, tree cavities) and less productive insect resource base. Preliminary research results suggest that forested corridors retained in intensively managed landscapes may be used extensively for roosting (S. Castleberry, University of Georgia, personal communication). Due to winter and spring roosting within Spanish moss

(Menzel et al. 2000b), the influence of professional moss gatherers requires monitoring (Bat Conservation International 2001). Additional research is needed on the foraging and roosting requirements of the Seminole bat.

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Southeastern Myotis

Myotis austroriparius (Rhoads, 1897)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The southeastern myotis (*Myotis austroriparius*) was described by Rhoads (1897) as a new subspecies of little brown myotis (*M. lucifugus*). When little brown myotis was placed in the genus *Vespertilio*, Rhoads (1897) named the new subspecies *Vespertilio lucifugus austroriparius*. In a subsequent revision of the genus *Myotis*, Miller and Allen (1928) used the current name combination. Several authors described subspecies of *M. austroriparius* based primarily on dorsal color variation; the species now is considered as monotypic (LaVal 1970). Three former subspecific taxa (*M. a. austroriparius*, *M. a. mumfordi*, and *M. a. gatesi*) are no longer recognized (Lowery 1974, Hall 1981, Koopman 1993). The southeastern myotis is known locally by several names including the Mississippi myotis, the southeastern little brown bat, and the southeastern brown bat (Humphrey 1992). The literature on this species is reviewed by Zinn (1977) and Jones and Manning (1989).

DISTINGUISHING CHARACTERISTICS

Myotis austroriparius is a small to medium-sized bat with rounded or blunt-tipped ears and narrow, pointed tragi. Measurements are: total length, 78–92 mm; tail, 34–42 mm; hind foot, 8–11 mm; ear, 12–15 mm; forearm, 36–41 mm; weight, 4–9 g. The calcar lacks a keel and the feet have long hairs that extend beyond the toes. LaVal (1970) indicated that the dorsal color of this bat is variable and may appear gray, brown or yellowish-brown. There usually is scant contrast in color between the tips and the base of the hairs. The dorsal pelage appears woolly. The venter is usually paler than the dorsum and the hairs may appear white-tipped. The wing and tail membranes are grayish black and are darker than the dorsum. *Myotis austroriparius* is most easily confused with the little brown myotis, which has smooth, glossy fur. The dental formula is I 2/3, C 1/1, P 3/3, M 3/3 = 38 (Figure 1).

CONSERVATION STATUS

The southeastern myotis has a global rank of Vulnerable (NatureServe 2007). The species is considered Apparently Secure in Louisiana, but is Vulnerable in Florida, Georgia, North Carolina, Tennessee, and Texas. It is Imperiled in Alabama and Arkansas. It is

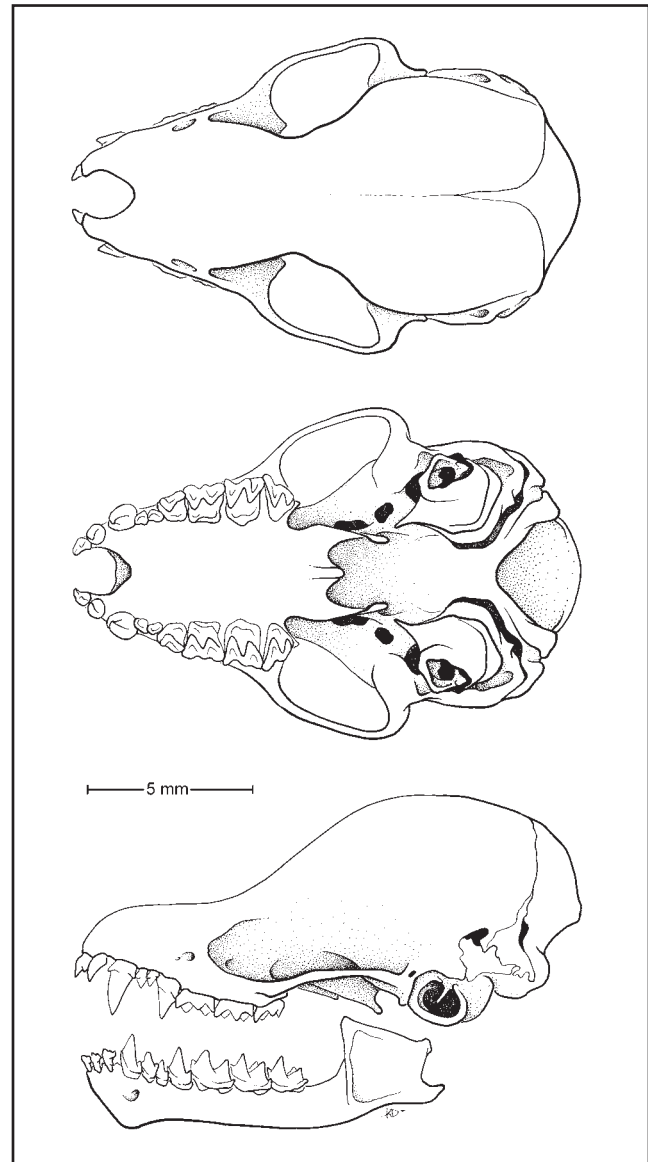


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Myotis austroriparius* from Citrus County, Florida (USNM 260855, female).

Critically Imperiled in Kentucky, Mississippi, Oklahoma, South Carolina, and Virginia.

DISTRIBUTION

The geographic range of *M. austroriparius* extends from eastern Texas and southeastern Oklahoma up the Mississippi Valley to Illinois and Indiana and

eastward to southern North Carolina (Figure 2). The western limits of the species coincide with the edge of the eastern deciduous forests in Texas (Packard 1966, Michael et al. 1970, Schmidly 1983, 1991; Schmidly et al. 1977) and the Little River drainage basin in Oklahoma (Caire et al. 1989). The species is found throughout Louisiana (Lowery 1974, Lance and Garrett 1997). The southeastern myotis occurs in Arkansas (Davis et al. 1955, Baker and Ward 1967, McDaniel and Gardner 1977, Heath et al. 1986, Sealander and Heidt 1990, Saugey et al. 1993) and is common throughout the western half of Tennessee (Graves and Harvey 1974, Harvey et al. 1991, Kennedy 1991) and Kentucky (Barbour and Davis 1974, Harvey et al. 1991). The species occurs throughout Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989, Best and Caesar 2000), and portions of Alabama (Howell 1921, Hall 1981, Hilton and Best 2000), Georgia (Golley 1962, Davis and Rippy 1968, Laerm et al. 1981), though rare north of the Fall Line (Menzel et al. 2000) in South Carolina (Webster et al. 1985, Menzel et al. 2002, Menzel et al. 2003a,b), and North Carolina (Davis and Rippy 1968). The species occurs in the northern half of the Florida peninsula and throughout the panhandle; it is most common in areas with karst topography (Rice 1957, Jennings 1958, Zinn 1977, Zinn and Humphrey 1981, Humphrey 1992, Ludlow and Gore 2000). Lee et al. (1982) reported a colony of several thousand near central North Carolina.

ABUNDANCE STATUS

The southeastern myotis once was regarded as one of the rarest bats in the southern United States (Miller and Allen 1928). However, as new roosting colonies of the species were discovered, the species came to be regarded as one of the most abundant bats in Florida (Rice 1957) and as relatively common in Louisiana (Lowery 1974). At one time, at least 18 maternity caves containing 400,000 adult females were known in Florida (Gore and Hovis 1992). There is considerable evidence that the population of this species has declined drastically in recent years. In a survey of roosting sites, Gore and Hovis (1992) found 200,000 adult females in 8 maternity caves, 3 of which were new locations. In many of the abandoned maternity roosts, there were signs of human disturbance.

PRIMARY HABITATS

In the northern portions of its range, caves are frequently used as roosting sites although other shelters are used (Barbour and Davis 1969). In the relatively caveless areas of the Gulf Coastal Plain, this bat roosts in hollow trees, buildings, and other protected

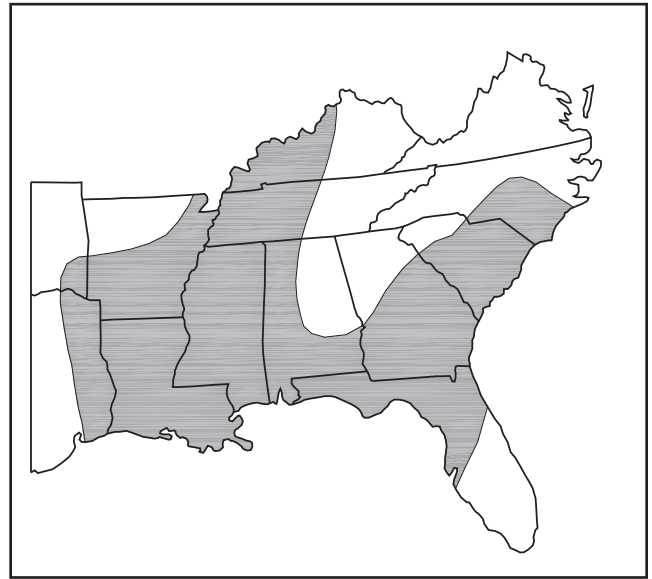


Figure 2. Distribution of *Myotis austroriparius* in the South.

locations (Lowery 1974, Foster et al. 1978, Sealander and Heidt 1990). Where the species roosts in Florida caves, clusters of *M. austroriparius* occur in locations above water (Rice 1957, Zinn 1977) which may afford protection from predators (Foster et al. 1978). However, water is not a prerequisite for a cave roost location because Mumford and Whitaker (1982) found the southeastern myotis roosting in dry caves in Indiana. These bats occupy different roosts in winter and summer (Bain 1981), but the pattern of movement between the sites is not known (Jones and Manning 1989). During winter in locations that are more northerly, the southeastern myotis may hibernate (Heath et al. 1986); the species does not enter torpor for extended periods in the southern portion of its range (Jennings 1958, Lowery 1974, Sealander and Heidt 1990). Zinn and Humphrey (1981) noted that the habitats used in foraging from a winter roost in northern Florida included a lake, marsh, riparian forest, shrubby oldfield, and small woodlot of slash pine (*Pinus elliottii*) and laurel oak (*Quercus laurifolia*). The foraging habitat used by bats from a nursery colony in the same area was a mesic forest dominated by sweet gum (*Liquidambar styraciflua*) and pignut hickory (*Carya glabra*). Both Menzel et al. (2005) and Ford et al. (2006) observed high levels of southeastern myotis activity around Carolina Bay wetlands as well as large bottomland river swamps in South Carolina.

REPRODUCTION

Mating behavior and details concerning reproduction in the southeastern myotis are poorly known (Jones and Manning 1989). Rice (1957) suggested that most reproductive activity occurred in spring based on

findings of males with enlarged epididymes during February–April in Florida. However, Mumford and Whitaker (1982) found that male *M. austroriparius* in Indiana also had scrotal testes (an indication of breeding activity) during August. After mating, the sexes segregate and the females join maternity colonies (Rice 1957, Zinn 1977, Bain 1981). In Florida, parturition in this species occurs from late April–May; most births take place in mid-May (Rice 1957). Parturition also may occur in mid-May in Oklahoma (Caire et al. 1989). Sherman (1937) described parturition in the southeastern myotis, the only species of *Myotis* in the United States that consistently bears twins (Barbour and Davis 1969). In the Florida population studied by Rice (1957), 90% of the females gave birth to twins; the remainder produced a single young. Foster et al. (1978) found one pregnant female carrying triplets. Young bats are carried by the female during the first day but are left behind thereafter (Sherman 1937). Young are capable of flight within 5–6 weeks after birth (Rice 1957).

FOOD HABITS

The food habits of *M. austroriparius* have not been described adequately. The southeastern myotis emerges late in the evening to begin foraging, flying low over open fields and watercourses (Rice 1957, Jennings 1958). Like most vespertilionids, *M. austroriparius* feeds on flying insects (Rice 1957). Zinn and Humphrey (1981) compared insect remains in southeastern myotis fecal pellets to insect availability and found that the bat selected large prey from several taxonomic groups. On cool spring evenings, only dipterans were active, and the bat selected mosquitoes and crane flies. During the warm spring and summer nights, flying insects were diverse and abundant; the bat selected from several taxonomic groups including Coleoptera, Lepidoptera, and culicid Diptera.

ASSOCIATED SPECIES

The southeastern myotis is primarily a cave-roosting species, and occasionally shares roost caves and mines with other bat species. During most months of the year, it is found in association with the little brown bat (*Nycticeius humeralis*; Rice 1957, Jones and Suttkus 1973, 1975), and Rafinesque's big-eared bat (*Corynorhinus rafinesquii*; Jones and Suttkus 1973, 1975). Sherman (1937) found *M. austroriparius* roosting with the Brazilian free-tailed bat (*Tadarida brasiliensis*) in a Florida building.

VULNERABILITY AND THREATS

Cave roosting populations of *M. austroriparius* are susceptible to disturbance from humans, flooding, sedimentation, closure of the cave entrance, and improper gating (Gore and Hovis 1992). Changes in land use and destruction of buildings that result in the loss of roost sites affect populations of the southeastern myotis in areas where the species roosts in tree cavities and human structures. Although insecticides have resulted in mortality (Clark 1981), exposure of *M. austroriparius* to high levels of cadmium, lead, chromium, and zinc did not affect survival rates in a Florida population (Clark et al. 1986).

MANAGEMENT SUGGESTIONS

Management centers on the protection of key roost sites from human disturbance and flooding. The maintenance of forested wetlands with large hollow trees also benefits the southeastern myotis (Bat Conservation International 2001, Menzel et al. 2003b). Management practices that change water quality and aquatic insect abundance may affect this species. Additional research is needed on critical habitat requirements, viability of small maternity colonies, and effects of disturbance on survival and reproductive success. Information is also needed on summer and winter roosting needs. A large maternity colony (>500) has been using a bridge over Lake Greeson in southwest Arkansas, yet members of this colony also use abandoned mines along the shore (D. Saugey, USDA Forest Service, personal communication).

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Gray Myotis

Myotis grisescens (Howell, 1909)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The gray myotis (*Myotis grisescens*), is a monotypic species (Hall 1981) that is often referred to as the gray bat. The literature is reviewed by Decher and Choate (1995).

DISTINGUISHING CHARACTERISTICS

The gray myotis is the largest species of *Myotis* in the South. Measurements are: total length, 90–107 mm; tail, 32–47 mm; hind foot, 9–13 mm; ear, 12–16 mm; forearm, 40–46 mm; weight, 7–14 g. The dorsal pelage is grayish-brown and the hair shafts are uniformly gray from base to tip. The ventral pelage is whitish or pale buff and the hairs are darker at the base. During summer, individuals may appear russet-colored or cinnamon-brown dorsally. The reddish appearance results from exposure to ammonia fumes in the summer colony (Tuttle 1979a). The calcar is not keeled. The sagittal and lambdoidal crests on the skull are distinctive (Hall 1981, Decher and Choate 1995). The gray myotis may be confused with the southeastern myotis (*M. austroriparius*), little brown myotis (*M. lucifugus*), northern long-eared myotis (*M. septentrionalis*), and Indiana myotis (*M. sodalis*). It can be distinguished by the uniformly colored dorsal fur (in the other species, the base and the tip of dorsal hairs are in contrasting shades) and by the wing membrane, which attaches at the ankle of the foot rather than at the base of the toes (see Barbour and Davis 1969:63). The dental formula is I 2/3, C 1/1, P 3/3, M 3/3 = 38 (Figure 1).

CONSERVATION STATUS

The gray myotis has a global rank of Vulnerable (NatureServe 2007). It is Imperiled in Alabama, Arkansas, Kentucky, Oklahoma, and Tennessee and Critically Imperiled in Florida, Georgia, South Carolina, and Virginia. The U. S. Fish and Wildlife Service (U. S. Department of Interior 2007) list the gray myotis as Endangered. A recovery plan for the species has been published (Brady et al. 1982).

DISTRIBUTION

Myotis grisescens is limited to a relatively small geographic area in the eastern United States (Figure 2). During summer, it occurs from the Florida Panhandle

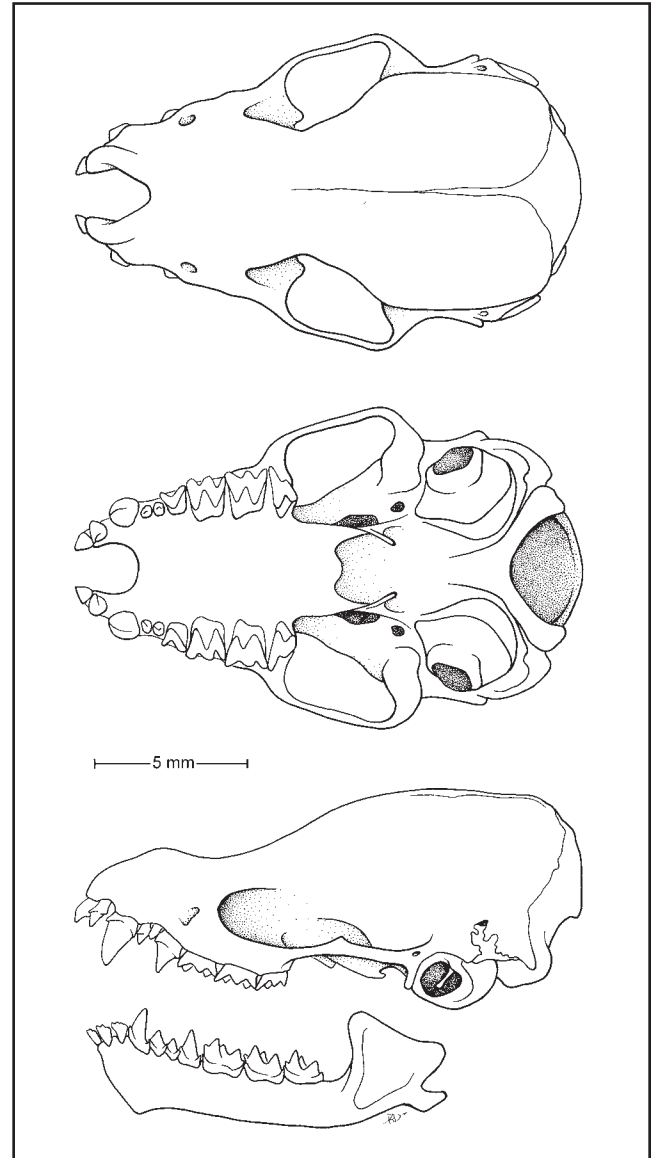


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Myotis grisescens* from Adair County, Kentucky (USNM 547689, female).

northward to Ohio, Indiana, and Illinois, and westward to southeastern Kansas and northeastern Oklahoma. In winter, its range is restricted; hibernacula are found in the limestone caves of Missouri, northern Arkansas, Tennessee, Alabama, and Kentucky. The gray myotis is known from the Appalachian Mountains in the southwestern tip of Virginia (Holsinger 1964, Handley 1991, 1992; Linzey 1998). The bat has been reported from western North Carolina

(Tuttle and Robertson 1969, Lee et al. 1982, Webster et al. 1985). The species has not been collected in South Carolina. Until recently, the gray myotis was known only from a few locations in Georgia (Golley 1962, Baker 1965, Laerm et al. 1981, Menzel et al. 2000). However, Martin and Sneed (1990) found a summer colony containing 4,000–9,000 gray myotis in a limestone cave in northwestern Georgia. Subsequent research has identified at least three bachelor colonies in this corner of Georgia (Johnson 2002). In the Florida Panhandle, the species occurs primarily in a small complex of caves and vicinity in Jackson County (Sherman 1934, Rice 1955, Jennings 1958, McNab 1974, Lee 1976, Lee and Tuttle 1979, Wenner 1984, Tuttle 1985, 1986; Clark et al. 1986, Gore 1992, Ludlow and Gore 2000). *Myotis grisescens* is known from several locations in Alabama, with the largest densities occurring in caves along the Tennessee River in the northern part of the state (Howell 1921, Miller and Allen 1928, Mohr 1932, 1933; McNab 1974, O'Shea et al. 1980, Clark et al. 1981, 1988; Stevenson and Tuttle 1981, Tuttle 1986, Best et al. 1997, Thomas and Best 2000). The gray myotis is reported from northeastern Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989). It is limited to a region of limestone hills in the northeastern corner of Oklahoma (Blair 1939, Glass and Ward 1959, Caire et al. 1989, Grigsby et al. 1993, Caire 1998) and is found in the northern counties of Arkansas (Sealander and Young 1955, Myers 1964, Krulin and Sealander 1972, Harvey 1976, 1984; McDaniel and Gardner 1977, Saugey 1978, Saugey et al. 1978, Sealander and Heidt 1990, Nelson et al. 1991, Timmerman and McDaniel 1992, Wilhide et al. 1998). The species is widely distributed in Tennessee (Howell 1909, Miller and Allen 1928, Mohr 1932, 1933; LaVal 1967, Tuttle and Robertson 1969, Harvey et al. 1991, Kennedy 1991) and Kentucky (Hall and Wilson 1966, Barbour and Davis 1974, Rabinowitz and Tuttle 1980, MacGregor and Westerman 1982, Harvey et al. 1991, Bat Conservation International 1993).

ABUNDANCE STATUS

The current population of the gray myotis is unknown. During the winter, 9 hibernacula were censused in Alabama, Arkansas, Kentucky, Missouri, and Tennessee and they contained an estimated 1,575,000 *M. grisescens* (Brady et al. 1982). Based on previous censuses of all known hibernacula, this may represent about 95% of the total gray myotis population. The majority of these bats overwinter at a few locations. James Cave in southwestern Kentucky is considered to be the third most important hibernaculum and over winters approximately 200,000 gray myotis (Bat Conservation International 1993). There are more

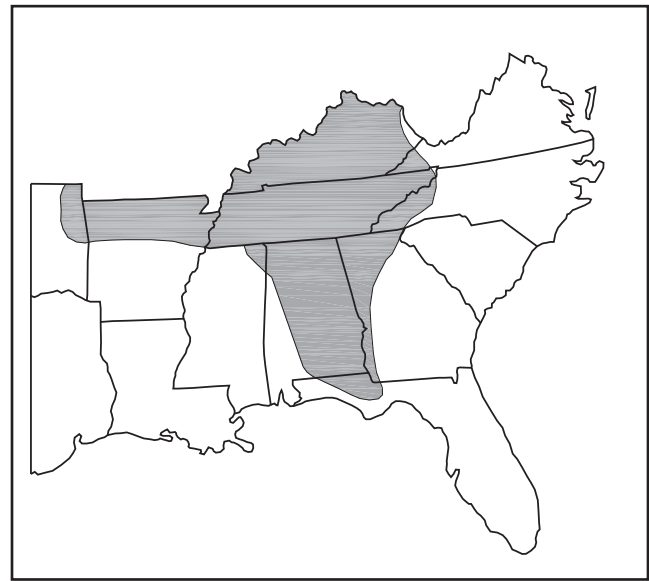


Figure 2. Distribution of *Myotis grisescens* in the South.

summer nursery colony and roost locations than there are winter hibernacula, but some of the summer colonies can be large. Blowing Wind Cave in Alabama serves as a summer roost for approximately 500,000 gray myotis (Tuttle 1986).

PRIMARY HABITATS

The gray myotis roosts in caves during winter and summer. Because the winter hibernacula differ environmentally from the caves used during summer, migratory movements occur between the winter and summer caves during spring and fall (Guthrie 1933a, LaVal and LaVal 1980). During winter, *M. grisescens* hibernates in humid caves that trap cold air (Tuttle 1975, 1976a). Tuttle (1985) found that <0.1% of the caves in the bat's range exhibit the appropriate temperature and humidity characteristics. Most of the hibernacula are located in areas characterized by limestone karst at latitudes < 39°N (Tuttle 1975). In summer, the gray myotis selects warm, humid caves that usually are located within 2 km of a river or lake (Tuttle 1976b) but maternity colonies sometimes have been found in storm sewers (Long 1961, Hays and Bingman 1964, Grigsby 1965, Timmerman and McDaniel 1992, Choate and Decher 1996). Maternity roosts are located in places such as ceiling pockets or high domes in caverns that trap heat. Night-roosting under bridges between foraging bouts has been observed (Johnson et al. 2002b). The mean temperatures in gray myotis maternity roosts in six caves in Alabama and Tennessee ranged from 13.9–26.3°C and the mean relative humidity in the same locations ranged from 86%–99% (Tuttle 1976a). Maximum migratory movements of 775 km between a winter hibernaculum in northern Tennessee and a summer

roost in Florida have been recorded (Tuttle 1976a). Each gray myotis colony appears to exhibit strong winter and summer philopatry to certain winter, summer, and transient caves which are used from year to year (Myers 1964, Tuttle 1976a, LaVal and LaVal 1980). Much circumstantial evidence indicates that the gray myotis may travel in aggregates between summer and winter caves (Barbour and Davis 1969, Elder and Gunier 1978).

REPRODUCTION

Saugey (1978) determined that the male and female reproductive cycles are asynchronous. The completion of spermatogenesis precedes ovulation by at least 7 months. In Florida, most copulation occurs in fall or early winter (McNab 1974); in Arkansas, copulation also takes place in spring (Saugey 1978). During copulation, a male mounts a female from behind and grasps the female's nape with his mouth (Lee 1976). Females mated in fall or winter store sperm; delayed fertilization takes place in March–April (Guthrie 1933b, Krulin and Sealander 1972, Tuttle 1975). A single embryo implants in the right uterine horn and fetal development takes place during April–May (Guthrie et al. 1951). Females return to maternity caves in May or June and give birth (Rice 1955, Guthrie 1933b). Young are born naked and blind; juveniles continue to nurse until they can fly, usually in late July or early August (Guthrie 1933b, Tuttle 1975, Saugey 1978). Females do not usually carry their young when they depart on foraging flights (Mohr 1933). Tuttle (1975) observed that in large maternity colonies, the young began to fly at an earlier age (24 days) than in smaller colonies (33 days). The difference in growth rates was attributed to the increase in ambient roost temperature caused by the larger population. Young weighing less than 7g at first flight have a high mortality rate (Tuttle 1976b) and young bats have higher mortality rates than adults (Tuttle and Stevenson 1977). One bat lived for 16.5 years between banding and recapture (Stevenson and Tuttle 1981).

FOOD HABITS

The gray myotis forages primarily over water (e. g., rivers, streams, or pools with forested edges) below treetop height (Tuttle 1976a, LaVal et al. 1977, Johnson 2002). Upon departure from its summer roost, the gray myotis tends to travel directly to a foraging area. Bats from a particular roost may travel up to 35 km to a foraging area, but usually confine their feeding activities to an area 1 km in length (LaVal et al. 1977). The average home range size of the gray bat in Alabama is 97 km² (Thomas and Best 2000). The echolocation sounds emitted during foraging range from 45–100 kHz (Shimozawa et al. 1974). The gray

bat probably feeds opportunistically on whatever prey species is most abundant (Lacki et al. 1995, Best et al. 1997). Its main prey includes insects from the orders of Ephemeroptera, Coleoptera, Diptera, Trichoptera, and Lepidoptera (Tuttle 1976b, Rabinowitz and Tuttle 1980, Lacki et al. 1995, Best et al. 1997). The gray bat culls the wings from its coleopteran prey (Brack and Mumford 1983). Tuttle (1979a) suggests that a colony of 250,000 gray myotis could consume approximately 1000 kg of insects in a single night.

ASSOCIATED SPECIES

Although other species may occupy the same caves, the gray myotis associates closely with few other bats. The gray myotis occasionally hibernates in clusters with the Indiana bat in Missouri (Myers 1964) and Arkansas (Sealander and Young 1955). In Florida, the eastern pipistrelle (*Pipistrellus subflavus*) associates with the gray myotis in fall roosts (Lee 1976) and the southeastern myotis clusters with the gray myotis in summer roosts (Tuttle 1976a).

VULNERABILITY AND THREATS

Declines in populations of the gray myotis have been observed throughout its range (Hall and Wilson 1966, Tuttle 1979a, Brady et al. 1982, MacGregor and Westerman 1982). The most commonly cited reasons for the declines are human disturbance and vandalism (Mohr 1972, Stevenson and Tuttle 1981), habitat destruction (Tuttle 1979b, Wenner 1984), and pollution (Clark et al. 1978, 1981, 1983a,b, 1986, 1988; Clawson 1991). Because large populations of the gray myotis congregate in few caves during winter, the species is particularly vulnerable to disturbance. Disruption during hibernation may result in the depletion of energy reserves and increased mortality. Many important hibernacula and nursery colonies have been gated or protected, and the bat populations in some of these have increased (Brady et al. 1982, Wenner 1984, Tuttle 1987, but see Ludlow and Gore 2000). The winter population of gray bats at 4 protected caves in Oklahoma was approximately 54,200 in 1991, which was a 33% increase from the past decade (Grigsby et al. 1993).

MANAGEMENT SUGGESTIONS

Conservation actions focus on the protection of maternity and hibernation caves from human disturbance and dam-induced flooding (Bat Conservation International 2001). Gated hibernation and maternity caves have led to stable and growing populations in many areas. Properly designed gates that do not restrict airflow or bat movement are beneficial (Tuttle 1979b). Human visitation to caves that potentially

contain gray myotis colonies should not occur during winter (November–March) or the maternity period (June–July). Gray bat echolocation calls are quite distinctive and can be used to finely delineate foraging habitat (Johnson et al. 2002a). The gray myotis is adversely affected by pesticides, siltation, and other factors that influence aquatic insect abundance.

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Eastern Small-footed Myotis

Myotis leibii (Audubon and Bachman, 1942)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

For many years, the taxon represented by the eastern small-footed myotis (*Myotis leibii*) was considered to be a subspecies of *M. subulatus* (Miller and Allen 1928). Glass and Baker (1968) rejected the name *M. subulatus* and replaced it with *M. leibii*. However, van Zyll de Jong (1984) suggested that the western forms of the species represented a separate species, *M. ciliolabrum*, a diagnosis confirmed by electrophoresis (Herd 1987). Consequently, *M. leibii* now represents a distinct, monotypic species. The natural history of the bat is reviewed by Best and Jennings (1997).

DISTINGUISHING CHARACTERISTICS

M. leibii is the smallest myotis in eastern North America. Measurements are: total length, 75–85 mm; tail, 32–39 mm; hind foot, 7–9 mm; ear, 13–15 mm; forearm, 30–34 mm; weight, 3.5–6 g. The dorsal pelage is yellowish brown; the tips of the hair are shiny giving the bat an overall sheen. The ventral pelage is pale and appears buff-colored. The eastern small-footed myotis has a distinctly black facial mask and the ears and membranes are dark brown. The calcar is strongly keeled and the tragus is pointed. The species can be confused with the little brown myotis (*M. lucifugus*) and eastern pipistrelle (*Pipistrellus subflavus*). The little brown myotis is larger, lacks a facial mask, and its calcar lacks a keel. The eastern pipistrelle is about the same size and coloration, but it has a blunt tragus and its forearms are distinctly paler than the rest of the wing membranes. The dental formula is I 2/3, C 1/1, P 3/3, M 3/3 = 38 (Figure 1).

CONSERVATION STATUS

The eastern small-footed myotis has a global rank of Vulnerable (NatureServe 2007). Georgia, Kentucky, North Carolina and Tennessee classify it as Imperiled, and it is Critically Imperiled in Alabama, Arkansas, Oklahoma, South Carolina, and Virginia.

DISTRIBUTION

The eastern small-footed myotis occurs in the Ozark Mountains of Arkansas, Missouri, and Oklahoma and in the Appalachian Mountains northward to south-eastern Canada and the New England states (Figure 2). In the South, the easternmost population is restricted

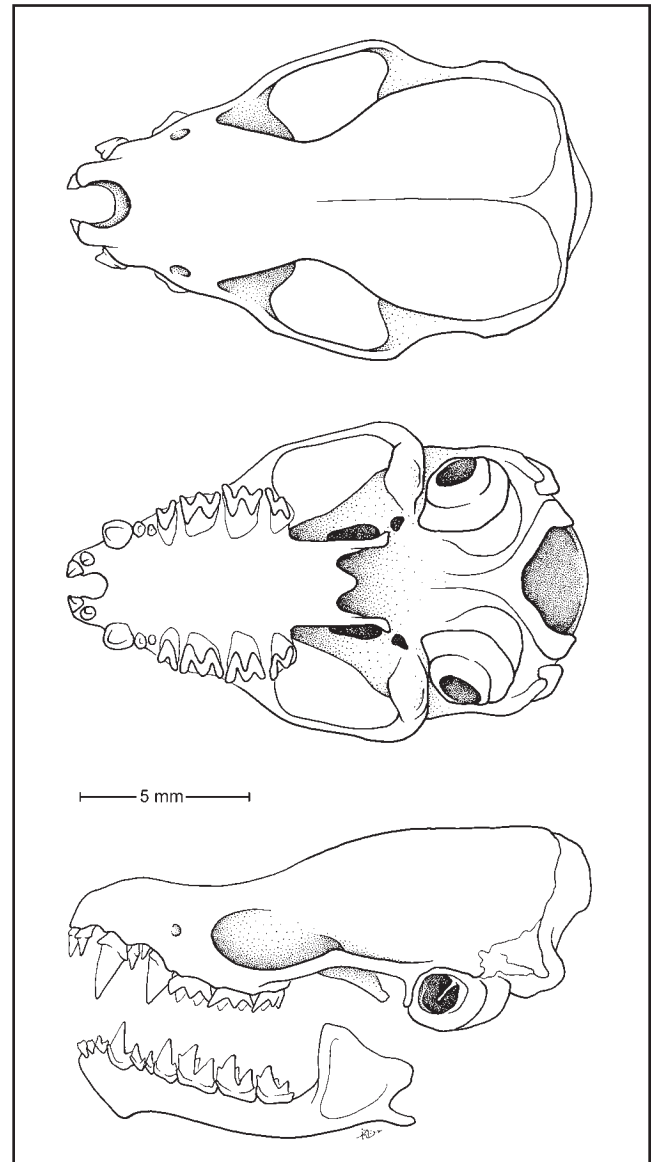


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Myotis leibii* from San Diego County, California (USNM 52803, gender unknown).

to caves and rocky outcrops associated with the Appalachian highlands of Virginia (Johnson 1950, Dalton 1987, Handley 1991, 1992; Linzey 1998), North Carolina (Adams 1950, Schwartz 1954, Lee et al. 1982), South Carolina (Menzel et al. 2003) northern Georgia (Baker 1967, Laerm et al. 1981, Menzel et al. 2000), Tennessee (Tuttle 1964, Neuhauser 1971, Webster et al.

1985, Harvey et al. 1991), and Kentucky (Bailey 1933, Barbour 1951, 1963; Davis et al. 1965, Barbour and Davis 1974, Fassler 1974, Harvey et al. 1991, Kiser and Meade 1993). The western population of this species in the South is known from locations in eastern Oklahoma (Glass and Ward 1959, Caire et al. 1989, Saugey et al. 1990) and northcentral Arkansas (Sealander 1967, McDaniel et al. 1982, Sealander and Heidt 1990, Saugey et al. 1993, Wilhide et al. 1998).

ABUNDANCE STATUS

Available collection records suggest that the eastern small-footed myotis is uncommon (Webster et al. 1985). However, the species may be abundant locally. Dalton (1987) observed the bat hibernating in 15% of the 106 caves that he examined in Virginia. Mist netting in cave mouths during the late summer, Handley (1979a, in Handley 1991) found that *M. leibii* comprised 15% of the total *Myotis* community in Virginia caves. Krutzsch (1966) and Dunn and Hall (1989) suggested that this species may be overlooked in cave surveys because it usually roosts alone at inconspicuous sites.

PRIMARY HABITATS

The habitat requirements of *M. leibii* are poorly known. During summer, the eastern small-footed myotis may occupy many types of roosts including buildings, caves, hillside rocks, and beneath the bark of trees (Webb and Jones 1952, Hitchcock 1955, Barbour and Davis 1969, Sealander and Heidt 1990, Handley 1991). During winter, the species hibernates in caves (Krutzsch 1966, Barbour and Davis 1969, 1974; Dalton 1987) and mines (Davis et al. 1955). It exhibits a preference for dry passages in colder caves and mines where temperatures drop below freezing and the humidity is low (Gunier and Elder 1973). It frequently roosts in locations subject to drafts near the mouth of the cave (Krutzsch 1966, Barbour and Davis 1969). Dunn and Hall (1989) found that 52% of the *M. leibii* in Pennsylvania surveys were hibernating in caves less than 150 m long. Although the bat occasionally hibernates in small groups (Krutzsch 1966, Dunn and Hall 1989), it usually roosts alone on cave ceilings (Krutzsch 1966); in small cracks, crevices, or depressions in the cavern walls or ceiling (Barbour and Davis 1969, Martin et al. 1966); beneath rock slabs (Tuttle 1964), and beneath rocks on the cave floor (Davis 1955, Krutzsch 1966, Martin et al. 1966). The eastern small-footed myotis is one of the last species to hibernate in winter and is one of the first to emerge in spring (Barbour and Davis 1974, Fenton 1972, Gates et al. 1984, Hitchcock et al. 1984).

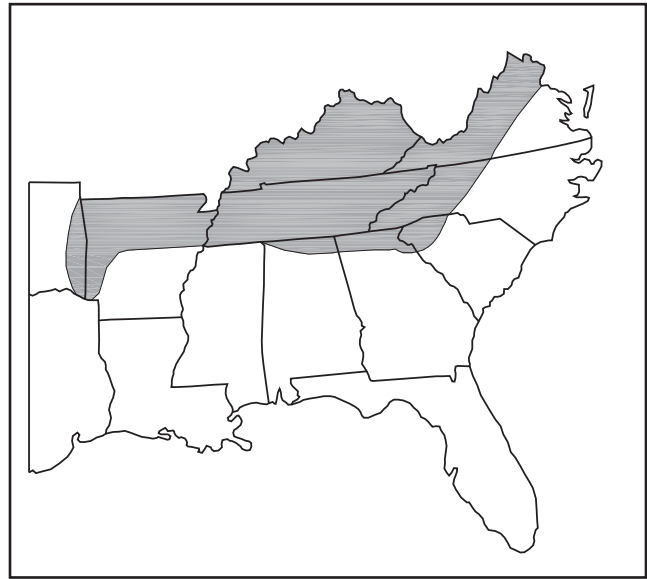


Figure 2. Distribution of *Myotis leibii* in the South.

REPRODUCTION

Most aspects of the reproductive ecology of *M. leibii* are not known. Barbour and Davis (1969) indicated that the species produces 1 young/yr. Because the weight of most vespertilionid neonates is 20–35% of the female's body weight (Kleiman and Davis 1979), Hitchcock et al. (1984) suggested that the addition of another fetus would be too great a burden for a female to carry while foraging. One nursery colony located inside a building contained 12 individuals (Hitchcock 1955). Based on banding and recapture data, Hitchcock et al. (1984) determined that *M. leibii* had a relatively low annual survival rate. Males and females had estimated mean annual survival rates of 0.75 years and 0.42 years, respectively, thereby explaining why *M. leibii* is uncommon.

FOOD HABITS

There are no published accounts of the food habits or foraging behavior of this species. Fenton and Bell (1981) analyzed the echolocation frequency of one *M. leibii* and found that it emitted ultrasonic pulses that modulated from 41 to 55 kHz in bursts lasting up to five milliseconds.

ASSOCIATED SPECIES

The eastern small-footed myotis usually roosts alone. However, it occupies caves that may contain the little brown myotis, southeastern myotis (*M. austroriparius*), northern long-eared myotis (*M. septentrionalis*), Indiana myotis (*M. sodalis*), big brown bat (*Eptesicus fuscus*), and eastern pipistrelle (Mohr 1936, Davis et al. 1955, Gates et al. 1984, Hitchcock et al. 1984, Dunn and Hall 1989).

VULNERABILITY AND THREATS

Populations of *M. leibii* in the southern United States are invariably small. Nevertheless, they are subject to the types of disturbance and destruction discussed by Thomas (1995). Because populations of *M. leibii* occupy small caves, the species may be vulnerable to habitat loss and degradation.

MANAGEMENT SUGGESTIONS

Management centers on the protection of hibernation sites in caves and mines (Bat Conservation International 2001). In locations where the bat roosts in rock outcrops, recreational activity around the outcrops and cliffs may disrupt the species. High intensity rock climbing could be detrimental, particularly where habitat is limited (C. Stihler, West Virginia Department of Natural Resources, personal communication). The provision of a water source on dry ridges is also beneficial. Additional research is needed on feeding patterns, roosting requirements, and nightly emergence patterns.

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Little Brown Myotis

Myotis lucifugus (LeConte, 1831)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The little brown myotis (*Myotis lucifugus*) originally was described as *Vespertilio lucifugus* by LeConte (1831, in Miller and Allen 1928). When he revised the Vespertilionidae, Allen (1893) placed the species in genus *Myotis* and was the first to use the present combination. Inadequate original descriptions and errors in identification have led to confusion in the distribution patterns of the southeastern myotis (*M. austroriparius*) and *M. lucifugus*. Originally classified as a subspecies of *M. lucifugus*, *M. austroriparius* was recognized as a distinct species by Miller and Allen (1928). Errors in identification, which have confused certain distributional records, persisted for many decades (Davis and Rippey 1968, Laerm et al. 1981). Six subspecies of *M. lucifugus* are recognized (Fenton and Barclay 1980, Hall 1981); one subspecies, *M. l. lucifugus*, occurs in the South. A comprehensive review of the species is provided by Barbour and Davis (1969) and Fenton and Barclay (1980).

DISTINGUISHING CHARACTERISTICS

Myotis lucifugus is a small to medium-sized, glossy-furred bat. Measurements are: total length, 75–105 mm; tail, 30–48 mm; hind foot, 8–11 mm; ear, 13–16 mm; forearm, 34–41 mm, weight, 5–12 g. The ears are narrow, pointed and when laid forward do not extend beyond the tip of the nose. The wing and tail membranes are dark brown to nearly black, and are sparsely haired. The dorsal pelage is tan, olive-brown, or dark brown and the ventral pelage is grayish tinged with buff. Hairs on the dorsum have dark bases and light-colored tips giving the bat a glossy or metallic sheen. The calcar lacks a keel and toe hairs extend beyond the tips of the claws. The little brown myotis is easily confused with the Indiana myotis (*M. sodalis*), which has a keeled calcar and short hairs on the feet that do not extend beyond the claws. The southeastern myotis has woolly dorsal fur. The gray myotis (*M. grisescens*) has uniformly-colored hairs on the dorsum and lacks a glossy sheen. The northern long-eared myotis (*M. septentrionalis*) and the eastern small-footed myotis (*M. leibii*) also lack a glossy sheen. The northern long-eared myotis has longer ears (17–19 mm) and a long, narrow tragus. The eastern small-footed myotis has a black facial mask and a tiny (8 mm) foot. The dental formula is I 2/3, C 1/1,

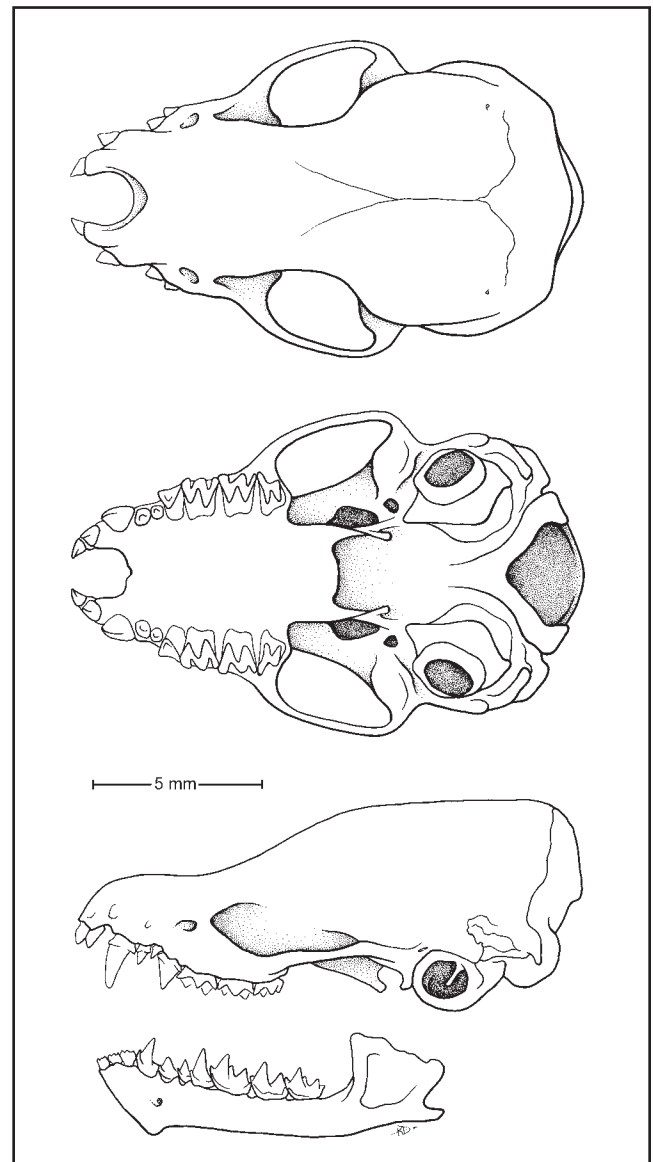


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Myotis lucifugus* from Rutland County, Vermont (USNM 181290, male).

P 3/3, M 3/3 = 38 (Figure 1). Species characteristics are described by Barbour and Davis (1969).

CONSERVATION STATUS

The little brown myotis has a global rank of Secure (NatureServe 2007). It is Secure in Kentucky, Tennessee, and Virginia. It is Apparently Secure in North

Carolina. It is Vulnerable in Alabama, Arkansas, Georgia, Mississippi, and South Carolina. Only Oklahoma classifies it as Critically Imperiled. It is unranked in Florida.

DISTRIBUTION

The range of *M. lucifugus* covers most of North America and extends southward through the central zone of Mexico, but the species is absent from the southern-central United States (Hall 1981). The distribution of the bat in the South is depicted in Figure 2. One specimen was reported from north central Texas (Baker 1964, Schmidly 1991). The species has not been recorded from Louisiana (Lowery 1974) and has been found only in eastern Oklahoma (Glass and Ward 1959, Caire et al. 1989). The little brown myotis occurs statewide in Arkansas during summer (Griffin and Hitchcock 1965, McDaniel and Gardner 1977, Gardner and McDaniel 1978, Saugey et al. 1978, 1988; Sealander and Heidt 1990). The species occurs in northern Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989), Alabama (Howell 1921, Holliman 1963, LaVal 1967) and Georgia (Morlan 1952, Golley 1962, Hall 1981, Laerm et al. 1981, Menzel et al. 2000). The sole Florida record of the species occurred in the northern panhandle (Brown 1985). It is found in the upper Piedmont of South Carolina (Allen 1893, Pickens 1928, Penny 1950, Golley 1966, Menzel et al. 2003) and the mountains and Piedmont of North Carolina (Odum 1949, Lee et al. 1982). The little brown myotis occurs throughout Tennessee (Linzey and Linzey 1971, Smith et al. 1974, Harvey et al. 1991, Kennedy 1991), Virginia (Bailey 1946, Handley and Patton 1947, Webster et al. 1985, Linzey 1998), and Kentucky (Barbour 1951, Davis et al. 1965, DeBlase et al. 1965, Barbour and Davis 1974, Humphrey and Cope 1976, Harvey et al. 1991).

ABUNDANCE STATUS

Although the little brown myotis is rare to locally common in scattered colonies in Arkansas, Mississippi, Alabama, Georgia, Florida, South Carolina, and North Carolina, it may be abundant in other portions of the region. It is difficult to estimate population size because the bats in a given area may switch roosts during a single night, over a period of several days, and seasonally (Barbour and Davis 1969, Humphrey and Cope 1976). Most summer colonies range in size from a few individuals to several hundred; colonies of 300–800 are common (Barbour and Davis 1969). Cockrum (1956) reported a colony of more than 30,000. From 50 nursery colonies in Indiana and Kentucky, Humphrey and Cope (1976) banded 71,706 little brown myotis. Hibernacula also may contain large concentrations of this species. Dunn and Hall (1989) found

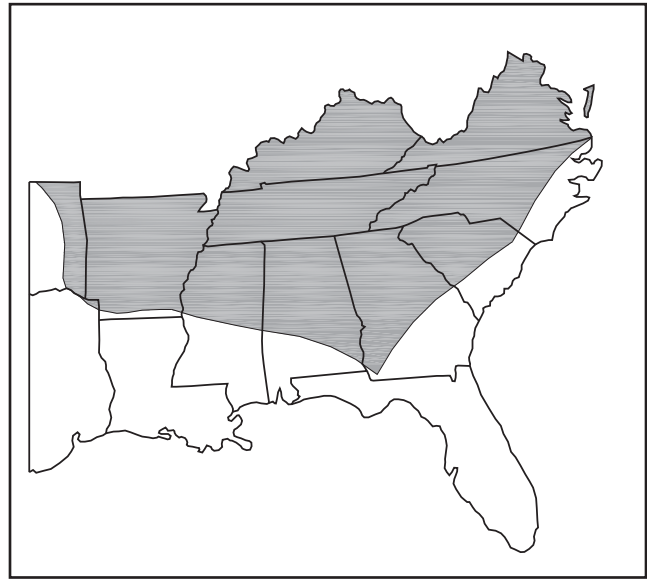


Figure 2. Distribution of *Myotis lucifugus* in the South.

over 20,000 bats in 71 colonies in Pennsylvania; 87% of these bats were found in 17 sites.

PRIMARY HABITATS

Although much of its foraging activity is associated with aquatic habitats, *M. lucifugus* feeds in many cover types and likely uses all available cover types in areas where the bat occurs (Barbour and Davis 1969, Fenton and Barclay 1980, Kurta 1982). The little brown myotis uses four types of roosts: day roosts, night roosts, nursery colonies, and hibernacula. Day roosts are used by active bats during the warmer months of the year and include dark or dimly lighted sites in buildings, tree cavities, log piles, caves, and under rocks (Barbour and Davis 1969, Kurta 1980, Burnett and August 1981, Kurta et al. 1993). Night roosts used temporarily by bats after their initial evening feeding are located near the day roosts and often in the same structure (Barclay 1982). Night roosts are in smaller, confined areas than day roosts, which suggest that the bats pack into these areas and use their body heat to speed the digestive processes (Buchler 1975). Night roosts result in the accumulation of feces away from the day roosts, which may render the day roost less susceptible to predators (Barbour and Davis 1969). Nursery colonies are used by females during parturition and rearing of the young (Davis and Hitchcock 1965). Hibernacula are located in caves or abandoned mines and are characterized by high humidity (>90%) and temperatures above freezing (Hitchcock 1949, 1965; Fenton 1970, Humphrey and Cope 1976, Nagorsen 1980). In the South, the species enters hibernacula in November and remains until mid-March. During winter, however, the bat occasionally arises from torpor and changes locations

within a hibernaculum (Guthrie 1933a, Folk 1940, Mumford 1958, Davis 1964a). When conditions are favorable, the little brown myotis may emerge from a hibernaculum and feed before returning to resume torpor (Whitaker and Rissler 1993).

REPRODUCTION

During fall and early winter, the little brown myotis congregates at hibernacula sites and engages in a behavior known as “swarming” (Davis 1964b, Schowalter 1980). When swarming, a group of bats fly in circular patterns near the outside entrance to a hibernaculum and inside the cave or mine. Mating relationships are initiated during this activity and copulation usually occurs within a month of the onset of swarming (Thomas et al. 1979). Both males and females are promiscuous (Thomas et al. 1979). Although Guthrie (1933b) and Wimsatt (1944) indicate that only torpid females retain sperm through hibernation, Thomas et al. (1979) suggest that most mated females store sperm through the winter months. Ovulation and fertilization occurs when the females complete hibernation in the spring (Buchanan 1987). The gestation period varies from 50–60 days (Wimsatt 1945). Before giving birth, females congregate at nursery colonies that are located in abandoned buildings or hollow trees that provide a relatively high ambient temperature (Davis and Hitchcock 1965, Humphrey and Cope 1976, Schowalter et al. 1979, Whitaker and Lawhead 1992). In Kentucky, parturition occurs from mid-May to late June (Humphrey and Cope 1976). A single young is born blind and covered with a fine, silky pelage. The eyes and ears open within a few hours (Gould 1971, Adams 1992); the young can thermoregulate within 10 days (Studier and O’Farrell 1972), and independent flight begins at 3 weeks (Anthony and Kunz 1977, Adams 1997). Female bats reach sexual maturity in their first year but males do not mate until their second year (Gustafson and Shemesh 1976, Thomas et al. 1979). The little brown myotis commonly lives 10 years (Paradiso and Greenhall 1967). One individual lived for 24 years (Griffin and Hitchcock 1965). Bowles (1983) captured a 23 year-old banded pregnant female that subsequently gave birth in a holding cage.

FOOD HABITS

The food habits of *M. lucifugus* have been studied extensively in both laboratory (Griffin et al. 1960) and field situations (Poole 1932, Griffin 1958, Belwood and Fenton 1976, Buchler 1975, 1976; Anthony and Kunz 1977, Fenton and Bell 1979, Kunz and Whitaker 1983, Griffin and Gates 1985, Whitaker and Lawhead 1992). The little brown myotis feeds on nocturnal flying

insects and preys heavily on insects that are associated with aquatic habitats (Belwood and Fenton 1976, Anthony and Kunz 1977). It prefers insects that are 3–10 mm in length (Anthony and Kunz 1977) and those that appear in hatching swarms (Fenton and Bell 1979). Lactating females select slightly larger prey than males or non-lactating females. Although its foraging pattern may vary, the little brown myotis begins its foraging in the early evening along the margins of lakes, rivers, or streams where it zigzags around vegetation 2–5 m tall (Anthony and Kunz 1977, Kurta 1982). Later in the night, it forages in groups within 2 m of the water surface (Fenton and Bell 1979, Kurta 1982, Barclay 1991). When foraging over streams, it exhibits a preference for calm-water pools rather than riffles (von Frenckell and Barclay 1987, Mackey and Barclay 1989). In the central Appalachians, little brown myotis were most associated with riparian zones along streams > 3rd order (Ford et al. 2005). Individual bats often return to the same feeding area (Hough 1957) but there is no evidence that this species defends feeding territories. When feeding, *M. lucifugus* echolocates with high intensity, frequency-modulated calls that sweep downward from 40–80 kHz, with most of the energy at 45 kHz (Griffin 1958, Barclay et al. 1979, Fenton and Bell 1979, Simmons et al. 1979, Pearl and Fenton 1996).

ASSOCIATED SPECIES

Although rarely found in mixed-species clusters, the little brown myotis shares roosts and hibernacula with other species that use caves or human structures. Common associates include the Indiana myotis, eastern small-footed myotis, southeastern myotis, northern long-eared myotis, gray myotis, eastern pipistrelle (*Pipistrellus subflavus*), big brown bat (*Eptesicus fuscus*), and Rafinesque’s big-eared bat (*Corynorhinus rafinesquii*).

VULNERABILITY AND THREATS

The little brown myotis is one of the most abundant and widespread bat species in North America and there are few threats to its existence. Caves and mines are fragile habitats and many populations of bats have been adversely affected through human disturbance (Humphrey 1964, 1978; Humphrey and Cope 1976, LaVal and LaVal 1980, Thomas 1995) or cave flooding (DeBlase et al. 1965). Bat populations also may be reduced locally through pesticide application (Geluso et al. 1976, Clark et al. 1978, Clark 1988), destruction of riparian feeding habitats, or alteration of buildings that serve as roost sites (Humphrey and Cope 1976).

MANAGEMENT SUGGESTIONS

Conservation actions focus on the protection of key hibernation roosts; the dominant threats to survival are disturbance of winter hibernacula and closure of abandoned mines. Recreational caving in known maternity and hibernacula caves should be prohibited to prevent disturbance. Cave gates that do not restrict airflow or bat movement are beneficial, as are forest management practices that provide roosting opportunities in large snags near water. Activities to safely exclude bats from dwellings may be appropriate (Bat Conservation International 2001.)

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Northern Long-eared Myotis

Myotis septentrionalis (Trouessaret, 1897)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The northern long-eared myotis (*Myotis septentrionalis*) is a monotypic species (van Zyll de Jong 1979, 1985; Jones et al. 1992) that once was considered a subspecies of Keen's myotis (*Myotis keenii*; Hall 1981). Fitch and Shump (1979) reviewed the literature on *M. keenii* including the former subspecies *M. keenii septentrionalis*. The literature on *M. septentrionalis* is reviewed by Caceres and Barclay (2000). Alternate vernacular names for this species include northern long-eared myotis, long-eared bat, and northern bat.

DISTINGUISHING CHARACTERISTICS

Myotis septentrionalis is a small to medium-sized bat with long ears and a long, pointed tragus. Measurements are: total length, 78–96 mm; tail, 32–35 mm; hind foot, 8–10 mm; ear, 16–18 mm; forearm, 32–37 mm; weight, 5–10 g. The dorsal pelage is dull brown and the venter may vary from pale grayish-brown to pale brown. The calcar is slightly keeled and may appear to lack a keel. The ears and wing membranes are slightly darker than the dorsal pelage. The bat is similar in appearance to the little brown myotis (*M. lucifugus*) from which it can be distinguished by longer ears and the number of elastic bands in the tail membrane. When laid forward, the ears of the northern long-eared myotis extend beyond the tip of the nose whereas in the little brown myotis they do not. There are 11 or fewer elastic bands in the tail membrane of *M. septentrionalis*. The dental formula is $I\ 2/3, C\ 1/1, P\ 3/3, M\ 3/3 = 38$ (Figure 1).

CONSERVATION STATUS

The northern long-eared myotis has a global rank of Apparently Secure (NatureServe 2007). Kentucky, Tennessee and South Carolina also classify it as Apparently Secure. It is Vulnerable in Georgia, Mississippi, North Carolina, and Virginia and Imperiled in Alabama, Arkansas, and Oklahoma. It is Possibly Extirpated in Florida. The bat is unranked in Louisiana.

DISTRIBUTION

The northern long-eared myotis ranges from Manitoba across southern Canada to Newfoundland, south to the Florida Panhandle and west to Wyoming

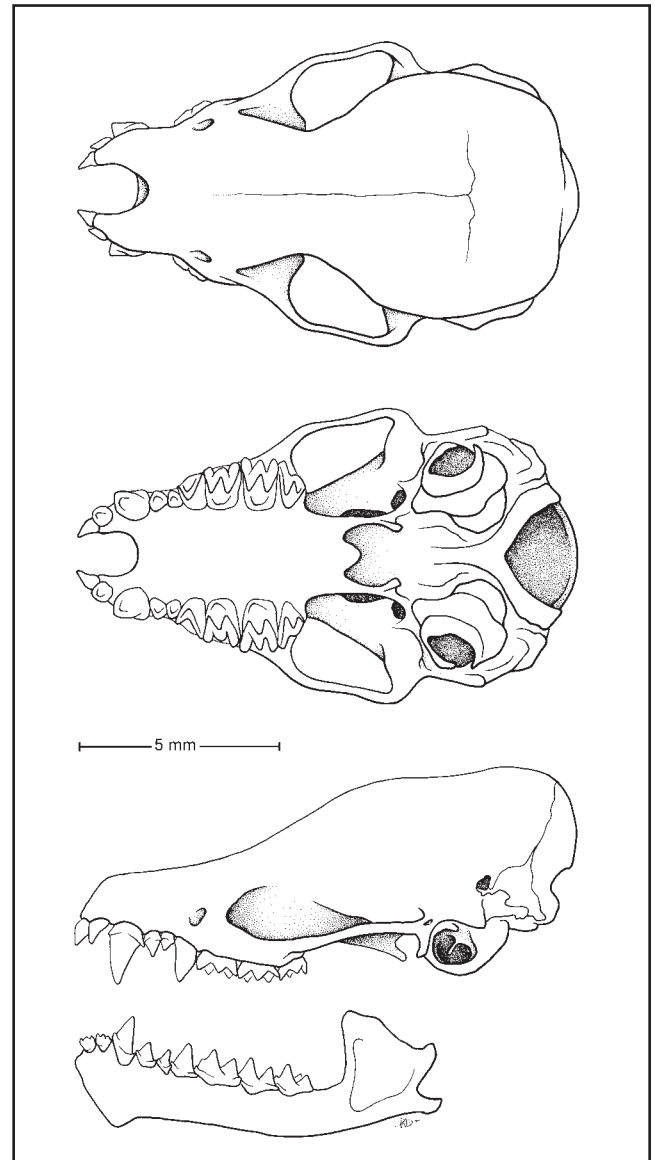


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Myotis septentrionalis* from Pulaski County, Kentucky (USNM 187424, male).

(Barbour and Davis 1969). It is more common in the northern portion of its range than in the southern United States (Figure 2). The species occurs throughout Virginia (Miller and Allen 1928, Bailey 1946, Dalton 1987, Handley 1979, 1991, 1992; Padgett and Rose 1991, Linzey 1998). The northern long-eared myotis occurs in North Carolina (Miller and Allen 1928, Schwartz 1954, Johnston 1967, Lee et al. 1982, Webster

et al. 1985) and South Carolina (Miller and Allen 1928, Hamilton 1943, Penny 1950, Golley 1966, Menzel et al. 2003) where it is essentially confined to the Blue Ridge mountains. There are two early (1890–1930) records of the species from the Piedmont of North Carolina (Lee et al. 1982). In Georgia, the species is found in the northern and western portions of the state (Hamilton 1943, Golley 1962, Laerm et al. 1981, Menzel et al. 1997, Menzel et al. 2000, Johnson et al. 2002). The northern myotis reaches its southern limit in the Florida Panhandle where it is known from a single specimen (Rice 1955, Jennings and Layne 1957, Jennings 1958). *Myotis septentrionalis* is known from eastern Alabama (Chermock and White 1953) and Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989). The species has not been recorded in Louisiana or Texas, but has been collected in five counties in eastern Oklahoma (Glass and Ward 1959, Caire et al. 1989, Saugey et al. 1990). In Arkansas, the species appears to be restricted to the Interior Highlands (Dellinger and Black 1940, Davis et al. 1955, Harvey and McDaniel 1983, Heath et al. 1986, Saugey et al. 1990, 1993; Sealander and Heidt 1990, Wilhide et al. 1998). The northern long-eared myotis occurs throughout Tennessee, and is more common in the eastern half of the state (Goodpasture and Hoffmeister 1952, Linzey and Linzey 1971, Smith et al. 1974, Harvey et al. 1991, Kennedy 1991). The species is distributed widely in Kentucky (Barbour and Davis 1974, Harvey et al. 1991, Kiser and Meade 1993, Lacki and Schwierjohann 2001).

ABUNDANCE STATUS

The northern long-eared myotis is relatively uncommon in most of the South (Barbour and Davis 1969, Sealander and Heidt 1990). Although this species usually roosts singly, colonies of hibernating bats vary from 3–350 (Hitchcock 1949, Heath et al. 1986). However, Harvey and McDaniel (1983) found that mist netting at cave entrances produced more specimens of northern long-eared myotis than indicated by observation of hibernating bats in caves. Because it roosts singly and often wedges into crevices or small interstices in cave formations, it can be overlooked in surveys (Dunn and Hall 1989, Whitaker and Rissler 1992a). During summer, the bats disperse into smaller groups that may be segregated by sex (Fitch and Shump 1979).

PRIMARY HABITATS

During daylight hours in summer, *Myotis septentrionalis* roosts in a variety of situations. Summer shelters include old buildings (Doutt et al. 1966), shutters (Mumford 1969), roof shingles (Brandon 1961), tarpaper (Barbour and Davis 1969), tree cavities, and

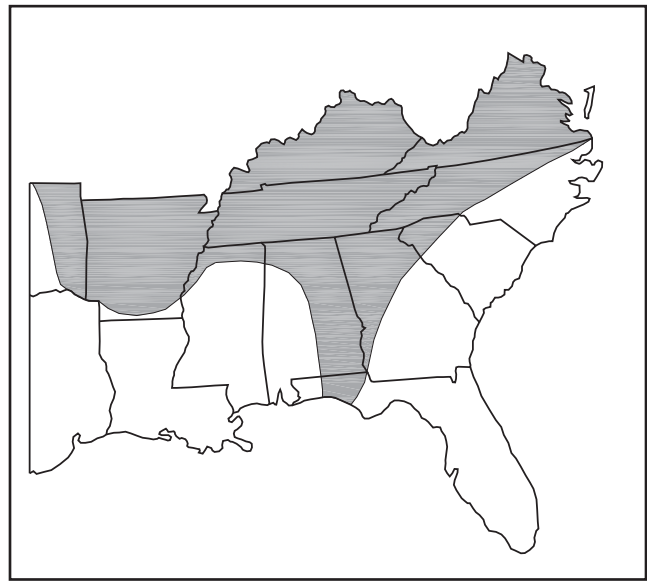


Figure 2. Distribution of *Myotis septentrionalis* in the South.

under exfoliating tree bark (Mumford and Cope 1964, Foster and Kurta 1999, Lacki and Schwierjohann 2001, Menzel et al. 2002, Owen et al. 2002). Most summer day roosts are in trees. At night, the bat uses temporary resting roosts in caves (Poole 1932, Jones 1964, Barbour and Davis 1969). During winter, the bat selects hibernating roosts in caves and mines where the air is cool and there is little air movement (Fitch and Shump 1979). Three northern long-eared myotis were found hibernating in a sewer where the air temperature was 1.5°C and the relative humidity was 69% (Goehring 1954). Swarming at the entrance to hibernacula begins in late August–early September (Davis 1964, Schowalter 1980, Dunn and Hall 1989). In the northern part of its range, *M. septentrionalis* may begin hibernation as early as August. It may remain in the hibernaculum for as long as nine months (Stones and Fritz 1969). Little is known of its hibernation period in the southern United States; in Indiana, the bat enters hibernation in September and arouses completely by late March (Whitaker and Rissler 1992a,b). The northern long-eared myotis apparently arouses from hibernation during warm spells and may change locations within the cave or fly outside before returning (Whitaker and Rissler 1992a,b; 1993).

REPRODUCTION

Little is known of the reproductive and developmental patterns of *M. septentrionalis*. Similar to *M. lucifugus*, the northern long-eared myotis probably breeds in fall and the females store sperm until spring (Barbour and Davis 1969). In spring, females segregate themselves from the males and form maternity colonies that may contain 3–30 individuals (Mumford and

Cope 1964). Maternity colonies have been found in buildings (Brandon 1961, Barbour and Davis 1969) or under tree bark (Mumford and Cope 1964). Pregnant females were found from late May–late June in Iowa (Kunz 1971). Easterla (1968) reported a parturition date of early June in Missouri. Lactating females have been captured from June–August in the mid-western United States (Brandon 1961, Kunz 1971, Turner 1974). Volant young were found in late July in Iowa (Kunz 1971). One banded individual lived for 18.5 years (Hall et al. 1957).

FOOD HABITS

The northern long-eared myotis begins foraging at dusk, roosts during the middle part of the night, and forages again before dawn (Barbour and Davis 1969, Kunz 1973). Foraging flights take place primarily in forests on hillsides and ridges where the bats feed between the forest canopy and shrub layer, often gleaning insects directly from foliage (LaVal et al. 1977, Owen et al. 2003, Ford et al. 2005). The species also forages above the surface of ponds (Cowan and Guiguet 1965, Brack and Whitaker 2001) and on the ground (Kirkland 1997). During aerial foraging, the northern long-eared myotis emits ultrasonic echolocation pulses that are 1–2 milliseconds in duration that modulate downward from 80–40 kHz (Fenton and Bell 1981, Fenton et al. 1983). The northern long-eared myotis feeds on flying insects including Lepidoptera, Coleoptera, Diptera, Trichoptera, and Plecoptera, with a preference for moths (Whitaker 1972, Belwood 1979, LaVal and LaVal 1980, Griffith and Gates 1985, Brack and Whitaker 2001).

ASSOCIATED SPECIES

The northern long-eared myotis often shares hibernacula with other species: eastern pipistrelle (*Pipistrellus subflavus*), big brown bat (*Eptesicus fuscus*), eastern small-footed myotis (*M. leibii*), Indiana myotis (*M. sodalis*), and little brown myotis (Swanson and Evans 1936, Griffin 1940, Hitchcock 1949, Stones and Fritz 1969, Gates et al. 1984, Griffith and Gates 1985, Schowalter 1980, Dunn and Hall 1989, Saugey et al. 1990, Whitaker and Rissler 1992a,b; Kurta et al. 1993).

VULNERABILITY AND THREATS

Populations of *M. septentrionalis* in the South are small and widely dispersed. Because of the philopatry exhibited by this species for winter roosts (Caire et al. 1979), the northern long-eared myotis may be locally vulnerable to extirpation if a hibernaculum is modified or destroyed. The northern myotis is sensitive to nontactile human stimuli in hibernacula (Thomas 1995). It may arouse and fly after human visits; this

causes premature depletion of fat reserves that may decrease survival.

MANAGEMENT SUGGESTIONS

Forest management that maintains existing snags and large trees benefits the northern long-eared myotis (Bat Conservation International 2001). Maternal and male roosts can be quite variable including live trees and snags in all size classes (Menzel et al. 2002, Ford et al. 2006). Lacki and Schwierjohann (2001) suggest that forest management practices that sustain diversity in tree species, size classes, and snag conditions provide the best habitat for northern bats in mixed mesophytic forests. The bat often forages in closed upland forest and high order stream or seep areas with small canopy openings (Owen et al. 2003, Ford et al. 2005); the maintenance of closed forest conditions is a prudent management measure. Additional research is needed on life history, hibernation roost requirements, and population trends.

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Indiana Myotis

Myotis sodalis (Miller and Allen, 1928)

W. Mark Ford and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The Indiana (or social) myotis (*Myotis sodalis*) is a monotypic species (Hall 1981). Before the Indiana myotis was described, several authors identified specimens as the little brown myotis (*M. lucifugus*). Cases of misidentification were described by Miller and Allen (1928) and Mumford and Cope (1964). The literature on the life history, ecology, and management of the Indiana myotis is reviewed by Thomson (1982), Menzel et al. (2001), and Kurta and Kennedy (2002).

DISTINGUISHING CHARACTERISTICS

The Indiana myotis is a small-to-medium-sized bat. Measurements are: total length, 70.8–90.6 mm; tail, 27–43.8 mm; hind foot, 7.2–8.6 mm; forearm, 36–40.4 mm; weight, 5–11 g. The color of the dorsal pelage varies, but is often dull brownish-gray to dark pinkish-gray. Individual dorsal hairs are tricolor. The ventral pelage is lighter and appears pinkish-white. The bat is most easily confused with the little brown myotis and southeastern myotis (*M. austroriparius*) from which it can be distinguished by its short, almost inconspicuous toe hairs and a small but noticeable keel on the calcar (Menzel et al. 2002). The dental formula is: I 2/3, C 1/1, P 3/3, M 3/3 = 38 (Figure 1).

CONSERVATION STATUS

The Indiana myotis has a global rank of Imperiled (NatureServe 2007). It is also Imperiled in Alabama, and Arkansas, and is Critically Imperiled in Kentucky, Oklahoma, South Carolina, Tennessee and Virginia. In North Carolina, the status of the bat is Under Review. The U. S. Fish and Wildlife Service (U. S. Department of Interior 2007) lists the Indiana myotis as Endangered. A recovery plan for the species has been published (U. S. Fish and Wildlife Service 1983) and a revised draft is available (U. S. Fish and Wildlife Service 2007).

DISTRIBUTION

The Indiana myotis occurs widely across much of the eastern half of the United States from Iowa and Oklahoma east to Vermont and south to Alabama (Figure 2). In part, its range is linked to the distribution of limestone caverns used as winter hibernacula

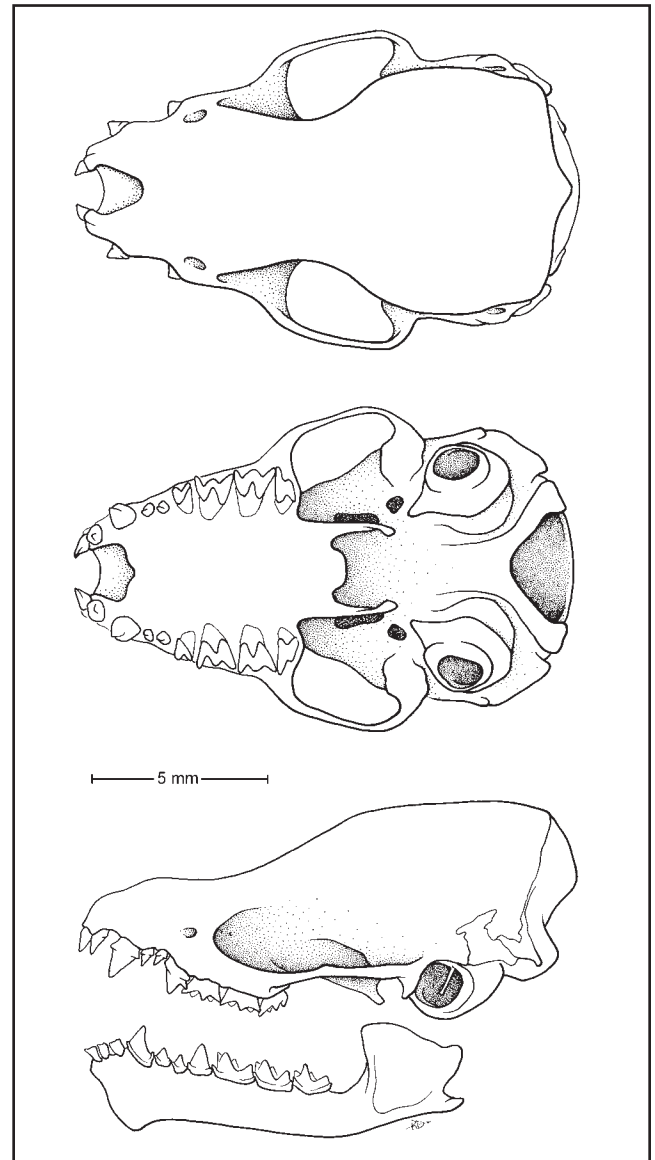


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Myotis sodalis* from Bath County, Virginia (USNM 268520, male).

(Hall 1962). During the summer, the range extends beyond the karst areas into the mid-Mississippi and upper-Ohio River Valleys north to Iowa, southern Michigan and southern Wisconsin (Mumford and Cope 1964, Hall 1962, Barbour and Davis 1969, Gardner and Cook 2002). Over half of known Indiana myotis utilize 9 Priority I hibernacula caves or mines

(i. e., those containing >30,000 wintering individuals) in Indiana, Kentucky, and Missouri (Humphrey 1978, Richter et al. 1978, U. S. Fish and Wildlife Service 2007, Clawson 2002). The remaining bats occupy approximately 330 Priority II and III hibernacula throughout winter range in the lower Midwest and upper Midsouth, as well as along the Appalachian Mountains from Alabama north to Vermont (Humphrey 1978, Dunn and Hall 1989, Menzel et al. 2001, Clawson 2002, U. S. Fish and Wildlife Service 2007).

In the South, scattered caves and mines serving as Indiana myotis hibernacula occur in western Virginia, northern Arkansas, northern Alabama, and throughout Kentucky and Tennessee east of the northward bend of the Tennessee River (U. S. Fish and Wildlife Service 2007, Harvey 2002). Kentucky and Tennessee contain over 100 hibernacula (Harvey et al. 1991). Small numbers of Indiana myotis hibernate in eastern Oklahoma (Saughey et al. 1990) and scattered records of hibernating individuals have been reported for western North Carolina, northwestern South Carolina, northwestern Georgia, northeastern Mississippi, and the Florida Panhandle near the Alabama and Georgia borders (Penny 1950, Jennings and Layne 1957, Golley 1962, Golley 1966, Kennedy et al. 1974, Jones and Carter 1989).

During spring post-hibernation emergence and summer maternity season, male Indiana myotis often remain locally near hibernacula sites (Ford et al. 2002, Harvey 2002, Whitaker and Brack 2002). Most maternity activity and summer distribution of females occurs outside of the South, north of the Ohio River and west of the Appalachians (Hall 1962, Humphrey 1978, Gardner and Cook 2002, Harvey and Britzke 2002), although maternity activity in Kentucky along the Ohio and Mississippi rivers has been known for some time (Harvey et al. 1991). Recent surveys have documented maternity activity and female distribution in the vicinity of the Great Smoky Mountains in North Carolina and Tennessee (Harvey 2002, Harvey and Britzke 2002). Lactating females and juveniles indicative of maternity activity have been reported in the Cumberland Mountains and Plateau of eastern Kentucky (Garner and Cook 2002, B. Palmer-Ball, Kentucky State Nature Preserve Commission, personal communication) and southwestern Virginia (Virginia Department of Game and Inland Fisheries 2003), and along the Allegheny Mountains and Plateau in north-central and southwestern of West Virginia (Owen et al. 2001, Beverly and Gumbert 2004). Relative to maternity activity in the Midwest, observations in the Appalachians seem to be ephemeral with little year-to-year fidelity (Carter 2006).

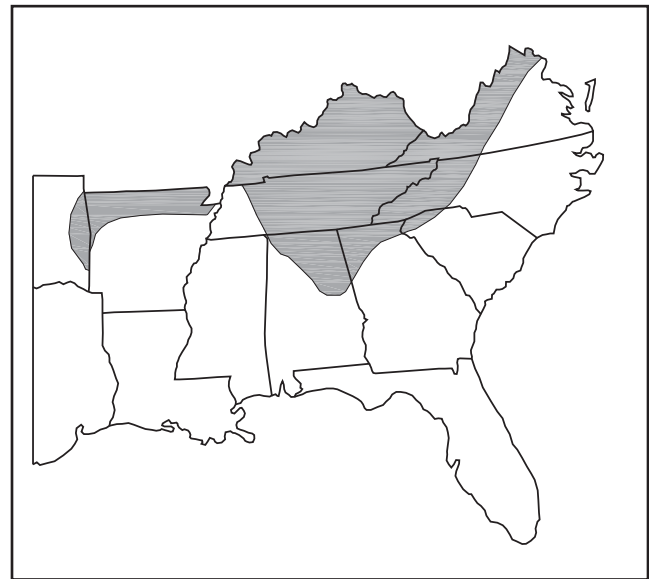


Figure 2. Distribution of *Myotis sodalis* in the South.

ABUNDANCE STATUS

In the 1960s, populations of Indiana myotis were estimated at 750,000 individuals; by the middle 1990s, this estimate declined to approximately 380,000 (Clawson 2002). Approximate overwinter hibernacula numbers by state include Alabama (250), Arkansas (2,500), Kentucky (60,000), Tennessee (10,000), and Virginia (1,000). Georgia, Mississippi, North Carolina, and South Carolina have overwintered small numbers from year to year less than 100 bats (Clawson 2002, U. S. Fish and Wildlife Service 2007). In the summer months, a foraging population density in Kentucky was measured at 11–17/ha (Humphrey et al. 1977). Although endangered, the Indiana myotis may be locally common near winter hibernacula during the fall “swarm” period.

PRIMARY HABITATS

The summer habitat requirements of the Indiana myotis are poorly known. Females and their young typically roost in maternity colonies located under loose, exfoliating bark of living and dead trees during June–July in bottomland and upland forest, and in residual trees in pastures, harvested stands, and suburban areas. Roosts usually receive full sunlight regardless of the sex and the geographic locale. Foraging often occurs along forested riparian areas (Cope et al. 1974, Humphrey et al. 1977, Kurta et al. 1993a,b; Callahan et al. 1997, Stihler 1998, Foster and Kurta 1999, Menzel et al. 2001, Belwood 2002, Whitaker and Brack 2002, Ford et al. 2005, Menzel et al. 2005). Approximately 25 tree species have been identified as maternity roosts (Menzel et al. 2001). Females

exhibit strong inter-annual fidelity for maternity sites (Kurta and Murray 2002). Although males often remain near hibernacula during summer (Henshaw and Folk 1966, Menzel et al. 2001, Harvey 2002, Whitaker and Brack 2002), many of the summer roosting habits of male bats are unknown. In the Allegheny Mountains of West Virginia, a male Indiana myotis roosted under exfoliating bark of large diameter (61 cm) shagbark hickory (*Carya ovata*) and sugar maple (*Acer saccharum*) in a close-canopied mature forest and a clearcut with residual trees, respectively (Ford et al. 2002). In southern Indiana, Brack (1983) found that 4 adult males roosted in moderately large diameter snags and trees (38 cm) in stands with 50% canopy closure. These included pine (*Pinus* spp.), shagbark hickory, white oak (*Quercus alba*), northern red oak (*Q. rubra*), and American elm (*Ulmus americana*).

During winter, Indiana myotis select caves and mines with noticeable airflow and temperatures that remain cool and stable (Henshaw 1965, Humphrey 1978, Menzel et al. 2001). When bats arrive at hibernacula between October–November, they begin the hibernation period in a relatively warm area deep within the cavern (Hall 1962, Myers 1964, Hassell 1967, Hardin and Hassell 1970, Kurta et al. 1996). The Indiana myotis may move nearer the cave entrance when temperatures cool. The Indiana myotis may arouse every 8–10 days; intra-hibernacula movements to favorable microclimates occur (Hardin 1967, Menzel et al. 2001). Many hibernation sites have mid-winter temperatures within 4–8°C (Hall 1962, Myers 1964, Humphrey 1978). Extremes of -1.6°–17°C have been reported (Barbour and Davis 1969, Humphrey 1978); however, those caves often report population declines (Tuttle and Kennedy 2002). Raesly and Gates (1986) found that Indiana myotis tend to occupy open cave ceilings where the ambient air and cave wall temperature were low, relative humidity was high, and airflow was great as compared to sites selected by other bat species. Guthrie (1933a) referred to Indiana myotis as the “cluster bat” because individuals pack together in tight groups or clusters on the ceilings of caves. These clusters have relatively high densities of bats (3000/m²; Hall 1962, Barbour and Davis 1969). This clustering behavior may buffer individuals from fluctuations in temperature (Clawson et al. 1980).

REPRODUCTION

Similar to other life history aspects, the reproductive ecology of Indiana myotis also is poorly known. Mating likely occurs during the fall swarm (Hall 1962), a behavior pattern in which many bats fly in and out of cave entrances from dusk to dawn (Poole 1932, Davis

and Hitchcock 1965, Fenton 1969, Cope and Humphrey 1977). Mating may also occur in winter or early spring (Guthrie 1933a, Hall 1962, Barbour and Davis 1969, LaVal and LaVal 1980). Females store sperm through winter; ovulation, fertilization, and implantation occur after the females leave the hibernacula in spring (Guthrie 1933b, Easterla and Watkins 1969, Thomson 1982). During gestation and lactation, females form small nursery colonies that typically do not contain males (Humphrey et al. 1977). Maternity roosts commonly are found in forested bottomland or riparian areas (Gardner et al. 1991, Callahan et al. 1997, Whitaker and Brack 2002); they also can occur in residual trees and snags in pastures and upland hardwoods (Kurta et al. 1993a,b; Whitaker and Hamilton 1998), and one colony has been reported in an abandoned church (Butchkoski and Hassinger 2002). Few maternity colonies are located in tree cavities; maternity roosts are found under exfoliating bark in large live trees and snags that receive maximum solar exposure (Cope et al. 1974, Humphrey et al. 1977, Kurta et al. 1993a,b; Rommé et al. 1995, Callahan et al. 1997, Menzel et al. 2001, Beverly and Gumbert 2004). Excellent summaries on maternity roost characteristics are presented by Gumbert et al. (2002), Kurta et al. (2002), Miller et al. (2002) and Whitaker and Brack (2002). The gestation period is unknown but parturition occurs in June or July. A single young is born (Easterla and Watkins 1969, Humphrey et al. 1977). Juveniles are weaned in 25–37 days (Humphrey et al. 1977). Volant young have been captured as early as mid-July (LaVal and LaVal 1980). The age at sexual maturity is unknown. Reported longevity is noteworthy, with individuals over 10 years recorded (Paradiso and Greenhall 1967, Humphrey and Cope 1977); one bat was recaptured 20 years after banding (LaVal and LaVal 1980).

FOOD HABITS

The Indiana myotis feeds in forested riparian areas (Humphrey et al. 1977, LaVal and LaVal 1980, Kessler et al. 1981, Brack 1983, Menzel et al. 2005, Ford et al. 2005), upland forests, and woodlots (Easterla and Watkins 1969, LaVal et al. 1977, Brack 1983, Hobson and Holland 1995). Open pasture and cropland may be avoided (Menzel et al. 2001, Menzel et al. 2005). Foraging occurs 2 m above the forest floor to canopy height (30 m) in closed forests and selectively logged stands (Brack 1983, Callahan 1993). There is strong evidence suggesting Indiana myotis return to foraging areas annually (Menzel et al. 2001, Gumbert et al. 2002). During the summer months in Indiana, Whitaker (1972) reported stomach contents contained Hymenoptera (50%), Coleoptera (24%), and Homoptera (19%). Belwood (1979) and Murray and Kurta (2002) analyzed fecal pellets from a maternity

roost and found that females and their young had consumed insects representing 8 and 12 orders, respectively. Belwood (1979) found that lepidopterans comprised 57% of total prey taken over the entire season, increasing to 70% during lactation. Brack and LaVal (1985) found that a diversity of insects was consumed early in the season; however, lepidopterans were selected for disproportionately. Diptera and Coleoptera are other important components of the diet (Kurta and Whitaker 1998, Murray and Kurta 2002). When foraging or orienting, the Indiana myotis emits ultrasonic pulses that sweep downward from 96 to 40 kHz in relatively short (0.5–1.5 millisecond) bursts, an adaptation for foraging in closed habitats (Thomson 1982, O'Farrell 1999, Owen et al. 2004, Ford et al. 2005).

ASSOCIATED SPECIES

Indiana myotis usually cave roost in large, dense, single-species clusters. However, roosting can occur in association with the gray myotis (*M. grisescens*; Sealander and Young 1955), little brown myotis (Griffin 1940, Cope et al. 1974), northern long-eared myotis (*M. septentrionalis*; van Zyll de Jong 1979), southeastern myotis (LaVal 1967), big brown bat (*Eptesicus fuscus*; Fenton 1966), and eastern pipistrelle (*Pipistrellus subflavus*; Kunz and Schlitter 1968).

VULNERABILITY AND THREATS

Myotis sodalis is extremely vulnerable to natural and human-caused perturbations due to concentrated populations in few winter hibernacula. Cave flooding (Hall 1962, DeBlase et al. 1965) and extremely low temperatures (Humphrey 1978) have resulted in mortality. However, the major threat is human disturbance (Humphrey 1992). Human visitation to hibernacula that cause arousal can result in weight loss than impacts survival (Johnson et al. 1998). Population declines also have been related to changes in cave temperature and airflow regimes caused by cave entrance alterations or improper gating (Humphrey 1978, Menzel et al. 2001, Currie 2002, Tuttle and Kennedy 2002), and destruction of summer foraging and roosting habitat by deforestation and stream channelization (Humphrey et al. 1977, U. S. Fish and Wildlife Service 1983, Carter 2007).

MANAGEMENT SUGGESTIONS

Human entry into hibernacula should be prohibited from September–May (Brady 1985). Improperly designed gates or unnatural obstructions that alter cave humidity and temperature require modification. In some instances, cold-air dams or ventilation shafts can restore favorable conditions (Tuttle and Kennedy

2002). Johnson et al. (2002) provides a summary on hibernacula management. Conservation actions also include the protection of roost trees and forested habitats where maternity activity occurs (Bat Conservation International 2001). Land-clearing activities near hibernacula and important maternity areas can be restricted to the hibernation period to avoid direct harm to Indiana myotis. Trees with exfoliating bark such as shagbark hickory should be retained. Forest management techniques, including the use of prescribed fire, that promote multiple-aged stands with large residual trees and snags should be investigated (Gumbert et al. 2002, Krusac and Mighton 2002, Keyser and Ford 2006).

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Evening Bat

Nycticeius humeralis (Rafinesque, 1818)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The evening bat (*Nycticeius humeralis*) originally was described as *Vespertilio humeralis* and as *Nycticeia crepuscularis*. The bat was placed in its current genus by Miller (1897). Three subspecies are recognized; two occur in the South (Hall 1981). The subspecies occurring throughout most of the southern United States is *N. h. humeralis*, while *N. h. subtropicalis* occurs in the extreme southern portion of the Florida peninsula (Schwartz 1951). Individuals of this subspecies are brighter in color and smaller in total length and tail length than *N. h. humeralis*. The literature is reviewed by Watkins (1972a).

DISTINGUISHING CHARACTERISTICS

The evening bat is a medium-sized, brown bat. Measurements are: total length, 80–96 mm; tail, 30–40 mm; hind foot, 7–9 mm; ear, 12–14 mm; forearm, 30–37 mm; weight 7–14 g. The dorsal pelage is dark brown to blackish-brown; the ventral pelage is slightly lighter in color. The ears are short and rounded, and the wing membranes are dark brown-black and are devoid of fur. The evening bat may be confused with several species of *Myotis*. Two characteristics are used to avoid misidentification: the evening bat has only two upper incisors, but *Myotis* species possess four and the tragus of the evening bat is short and curved with a rounded tip. Tragi of most *Myotis* are long with a straight edge and pointed tip. The dental formula is $I\ 1/3, C\ 1/1, P\ 1/2, M\ 3/3 = 30$ (Figure 1).

CONSERVATION STATUS

The evening bat has a global rank of Secure (NatureServe 2007). It is Secure in Alabama, Georgia, Louisiana, North Carolina, Tennessee and Texas. It is Apparently Secure in Oklahoma and Virginia. Arkansas and Kentucky classify the evening bat as Vulnerable. It is unranked in Florida, Mississippi, and South Carolina.

DISTRIBUTION

The evening bat occurs throughout the South at elevations from sea level to 300 m or more in temperate deciduous forest habitats (Figure 2). It is rare or absent from much of the southern Appalachians (Barbour and Davis 1974, Webster et al. 1985), but is found in all states of the region (Handley and Patton 1947, Golley 1962, 1966; Wolfe 1971, Lowery 1974,

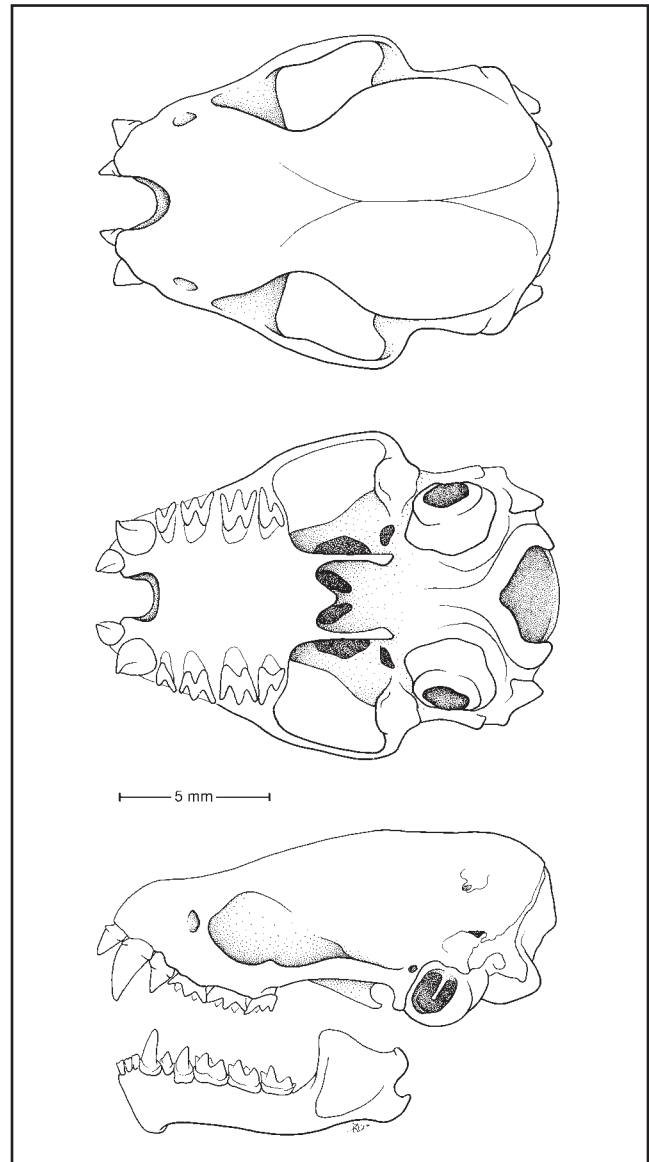


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Nycticeius humeralis* from Wilkinson County, Mississippi (USNM 348354, female).

Laerm et al. 1981, Lee et al. 1982, Caire et al. 1989, Jones and Carter 1989, Sealander and Heidt 1990, Harvey et al. 1991, Kennedy 1991, Schmidly 1991, Krishon et al. 1997, Lance and Garrett 1997, Taylor and Lehman 1997, Carter et al. 1998, Linzey 1998, Willhide et al. 1998, Menzel et al. 2000, 2001, 2002, 2003a, 2003b; Hutchinson 2001).

ABUNDANCE STATUS

The evening bat is a common bat in the southern portions of its range (Lowery 1974, Saugey et al. 1988). Maternity colonies often consist of hundreds of individuals, but there is no information on the density of the evening bat in any area. The absence of this species in the northern parts of its range during winter (Watkins 1972a), documented recoveries of banded bats (Humphrey and Cope 1968), and autumnal fat deposition (Baker et al. 1968) strongly suggest that the evening bat migrates south before winter. Winter roosting sites in southern states have not been described.

PRIMARY HABITATS

The evening bat probably evolved as a tree-roosting species. Literature records of *N. humeralis* roosting in trees are relatively scarce, but most tree roosts are discovered after trees are cut down. Tree roosts are located in cavities (Schwartz 1952, Barbour and Davis 1969, Menzel et al. 2000, 2001), under loose bark (Bailey 1933, Jennings 1958, Chapman and Chapman 1990, Menzel et al. 1999, 2000, 2001), in palm fronds (*Sabal* spp.; Taylor and Lehman 1997), and in Spanish moss (*Tillandsia usneoides*; Jennings 1958). Most observations of the evening bat are from communal roosts in buildings (Baker and Ward 1967, Easterla and Watkins 1970a, Watkins 1972a, Watkins and Shump 1981, Chapman and Chapman 1990). There are observations of the species roosting in a cave (Easterla 1965), and the species has been captured in the vicinity of cave openings on several occasions, sometimes using cave habitat as roost sites (Bailey 1933, Davis 1964, Glass and Humphrey 1971). There are two peaks of foraging activity: one during the early evening hours and a second just before dawn (Watkins 1972b). Foraging primarily occurs along riparian areas, wetlands and bottomlands (Schmidly 1991, Kirshon et al. 1997, Menzel et al. 2002). Although activity along riparian areas in South Carolina was significantly greater than upland areas (Menzel et al. 2005), pine (*Pinus* spp.) savannas are readily utilized (Ford et al. 2006). Considerable evening bat foraging activity occurs above forest canopies (Menzel et al. 2005). Several authors mention an association between the evening bat and bald cypress (*Taxodium distichum*; Harper 1927, Jennings 1958, Layne 1974, Zinn 1977, Laerm et al. 1980).

REPRODUCTION

Mating occurs in the fall, but copulatory behavior has not been observed in this species (Watkins 1972a). Birth of young occurs between May–July (Lindsay 1956, Easterla and Watkins 1970b). During this period, the females join nursery colonies that rarely contain males.

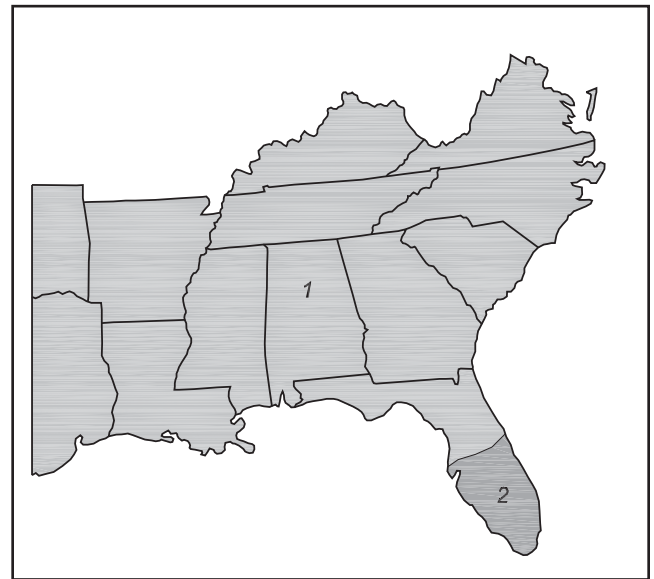


Figure 2. Distribution of *Nycticeius humeralis* in the South: (1) *N. h. humeralis*; (2) *N. h. subtropicalis*.

Litter size varies from 1–3; many females give birth to 2 young. Jones (1967) gives details concerning the growth and development of juvenile bats. Young are capable of flight within 20 days after birth and reach adult size within 30 days. The pelage color of juvenile evening bats is much darker (dark brown-black) than that of adults (yellowish brown). Presumably, the molt to adult pelage occurs during the first year.

FOOD HABITS

The evening bat preys on species of Hymenoptera, Hemiptera, Coleoptera, Diptera, and Lepidoptera (Ross 1967, Mumford and Whitaker 1982, Feldhamer et al. 1995, Carter et al. 1998). The evening bat is not a selective consumer (Carter et al. 1998).

ASSOCIATED SPECIES

The evening bat commonly roosts in buildings with the Brazilian free-tailed bat (*Tadarida brasiliensis*), but evening bats usually form discrete roosting groups (Barbour and Davis 1969). Other species that roost in association with the evening bat includes the big brown bat (*Eptesicus fuscus*) and the southeastern myotis (*M. austroriparius*).

VULNERABILITY AND THREATS

Nycticeius humeralis is widely distributed and relatively common in the southern United States. Threats to its survival are not apparent; however, local extirpations may occur if roosting sites or nursery locations are eliminated. Streamside management zones benefit this species.

MANAGEMENT SUGGESTIONS

Conditions that promote mature forests with snags and large trees with broken tops are beneficial (Menzel et al. 2001). Trees and snags with exfoliating bark characteristics should be retained when possible. Preliminary research in mature longleaf pine (*P. palustris*) and loblolly pine (*P. taeda*) plantations in southwest Georgia indicate considerable plasticity in roost structure (Miles et al. 2006). Beaver activity on the landscape provides foraging and roosting habitat by creating ponds that provide water and insect prey (Menzel et al. 2001). Loss of roost sites occurs when barns and old buildings are replaced by structures that bats cannot use. Provision of artificial roosts may aid this species (Bat Conservation International 2001). Additional research is needed on migration dynamics and winter habitat requirements.

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Eastern Pipistrelle

Pipistrellus subflavus (F. Cuvier, 1832)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The eastern pipistrelle (*Pipistrellus subflavus*) originally was described as *Vespertilio subflavus* (Hall 1981, Fujita and Kunz 1984). It was included in its present genus by Miller (1897). Four subspecies of *P. subflavus* currently are recognized (Davis 1959a, Hall 1981); two occur in the South. Recent genetic analysis has called into question the validity of the eastern pipistrelle's membership in the genus *Pipistrellus* (Hoofer et al. 2006). *P. s. floridanus* occurs in peninsular Florida and in the southeastern corner of Georgia. *P. s. subflavus* is widely distributed in the South except in areas occupied by *P. s. floridanus*. The literature is reviewed by Fujita and Kunz (1984).

DISTINGUISHING CHARACTERISTICS

The eastern pipistrelle is a small, light-colored bat with dark wing membranes. Measurements are: total length, 75–90 mm; tail, 33–41 mm; hind foot, 8–10 mm; ear, 12–14 mm; forearm, 31–45 mm; weight, 4–8 g. The dorsal pelage varies in coloration from pale yellowish-brown to pale grayish-brown. The venter is lighter than the dorsum and the body coloration contrasts sharply with the dark blackish wing membranes. The forearms, which are reddish or pink, are lighter in color than the rest of the wings; this feature is used to distinguish the species either from a distance or in the hand. The calcar is not keeled. The anterior third of the tail membrane is covered with fur. The eastern pipistrelle might be confused with the eastern small-footed myotis (*Myotis leibii*) because of the similarity in size and body coloration. However, the eastern pipistrelle lacks a dark facial mask, its ears are about the same color as the dorsal pelage, and it has a blunt (rather than a pointed) tragus. The two subspecies of *P. subflavus* are distinguished primarily on the basis of pelage coloration. *Pipistrellus s. subflavus* is characterized by reddish tones in the pelage whereas *P. s. floridanus* lacks reddish coloration. The size of *P. s. floridanus* does not differ from specimens of *P. s. subflavus* from the Atlantic seaboard, but they are slightly larger than *P. s. subflavus* from Kentucky and the Ohio River Valley (Davis 1959a). The dental formula is $I\ 2/3, C\ 1/1, P\ 2/3, M\ 3/3 = 36$ (Figure 1).

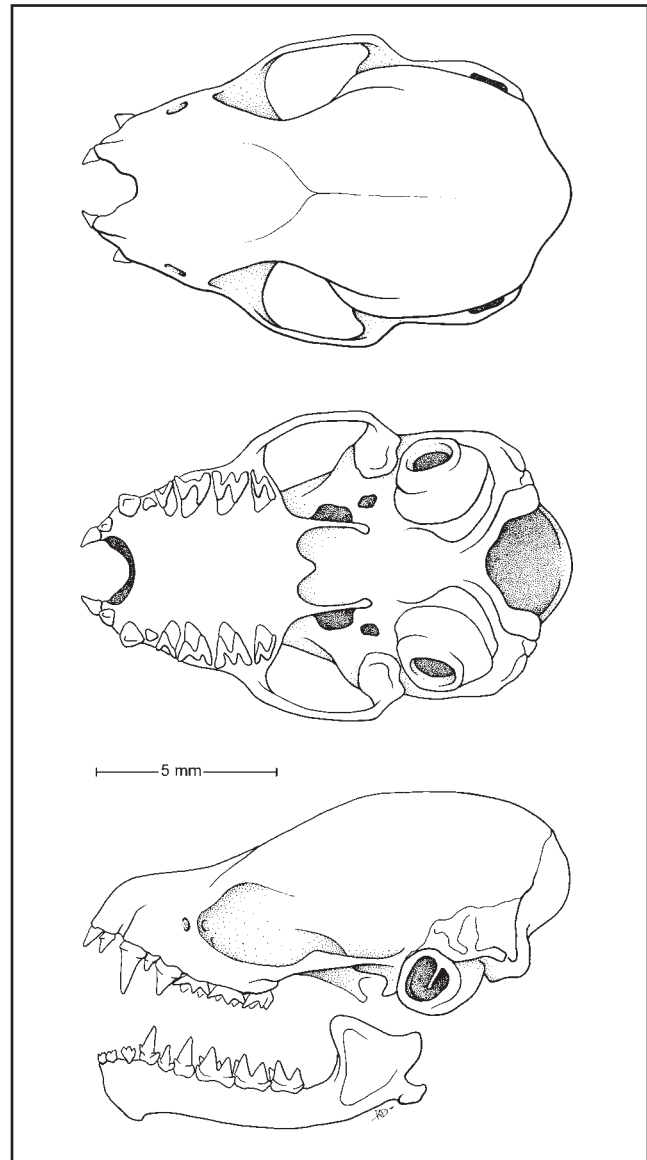


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Pipistrellus subflavus* from Potomac River, Virginia (USNM 156966, female).

CONSERVATION STATUS

The eastern pipistrelle has a global rank of Secure (NatureServe 2007). It is Secure in Alabama, Georgia, Mississippi, North Carolina, Tennessee, Texas and Virginia. It is Apparently Secure in Arkansas, Kentucky, Louisiana, and Oklahoma. It is unranked in Florida and South Carolina.

DISTRIBUTION

The eastern pipistrelle occurs throughout most of the eastern half of North America and along the Gulf of Mexico coast to the Yucatan Peninsula in Mexico (Hall 1981). The distribution of the eastern pipistrelle is depicted in Figure 2. *P. s. subflavus* occurs throughout Texas (Davis 1974, Schmidly 1983, 1991; Schmidly et al. 1977, Walker et al. 1996, Sandel et al. 2001), Louisiana (Jones and Pagels 1968, Jones and Suttikus 1973, Lowery 1974, Lance and Garrett 1997), Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989, Best and Caesar 2000), Alabama (Howell 1921, Hilton and Best 2000), South Carolina (Golley 1966, Webster et al. 1985, Menzel et al. 2002a, Menzel et al. 2003a,b), North Carolina (Lee et al. 1982, Webster et al. 1985), Virginia (Lewis 1940, Bailey 1946, Webster et al. 1985, Handley 1991, Linzey 1998), Kentucky (Hall 1963, DeBlase et al. 1965, Barbour and Davis 1969, 1974; Harvey et al. 1991), Tennessee (Linzey and Linzey 1971, Rabinowitz 1981, Harvey et al. 1991, Kennedy 1991), Arkansas (Sealander and Young 1955, McDaniel and Gardner 1977, Gardner and McDaniel 1978, Saugey et al. 1978, 1988, 1989; Heath et al. 1986, Sealander and Heidt 1990, Wilhide et al. 1998), and Oklahoma (Caire et al. 1984, 1989). *Pipistrellus s. subflavus* also is found in Georgia (Davis 1959a, Golley 1962, Laerm et al. 1981, Menzel et al. 1996, 1997, 2000, 2002b; Krishon et al. 1997, Carter et al. 1998, 1999; Johnson 2002) except in the southeastern part of the state. *Pipistrellus s. floridanus* occurs throughout the northern half of the Florida peninsula (Blair 1935, Jennings and Layne 1957, Jennings 1958) and occurs in southeastern Georgia (Davis 1959a). The southernmost record of *P. s. floridanus* is from Okeechobee County, Florida, and the northernmost record is from Long County, Georgia (Davis 1959a, Hall 1981).

ABUNDANCE STATUS

No population estimates are available, but the eastern pipistrelle is abundant throughout the southern United States. *Pipistrellus subflavus* may be the most abundant bat in Arkansas (Sealander and Heidt 1990).

PRIMARY HABITATS

During winter, the eastern pipistrelle hibernates in caves (Myers 1960, Hall 1963, Davis 1964, 1966), tunnels (Mohr 1942), mines (Heath et al. 1986, Sealander and Heidt 1990, Menzel et al. 1997), storm sewers (Goehring 1954), highway culverts (Walker et al. 1996, Sandel et al. 2001), or buildings (Jones and Suttikus 1973). Most eastern pipistrelles hibernate singly (Guthrie 1933a, Hitchcock 1949, McNab 1974). The bat often selects the deep parts of caves or other

microhabitats with high humidity (Caire et al. 1989), constant temperature (Hitchcock 1949, Rabinowitz 1981), and minimal air flow (Rabinowitz 1981). Males and females may occupy the same hibernacula (Griffin 1940), but disproportionate sex ratios in hibernating populations are reported (Mohr 1942, Hitchcock 1949, Sealander and Young 1955, Davis 1959b). The differences in sex ratios may indicate that males and females select different hibernating locations to some degree (Jones and Pagels 1968) or that males have a higher survival rate than females (Davis 1959b). The number of evening pipistrelles that use a winter hibernaculum is correlated with the amount of forest and agricultural areas around each site (Sandel et al. 2001). The abundance of *P. subflavus* at cave hibernacula in Missouri in late July–August and again in late April–May suggests that the species is among the first bats to enter hibernacula in fall and the last to leave in spring (LaVal and LaVal 1980). Upon departure from the hibernacula in spring, males and females disperse and use separate roosts during the maternity period (Hamilton and Whitaker 1979). Females establish maternity roosts in barns (Poole 1938, Lane 1946, Hoying 1983), other structures (Allen 1921, Jones and Pagels 1968, Jones and Suttikus 1973, Whitaker 1998), hollow trees (Sealander and Heidt 1990, Menzel et al. 1996), caves, or rock crevices (Allen 1921, Humphrey et al. 1976, Caire et al. 1989). Summer roosting groups may use more than one roost location (Whitaker 1998). The habitat preferences of males are less well known, but they are presumed to be solitary (Lane 1946) and, similar to females in late summer, may roost in trees (Findley 1954, Krishon et al. 1997, Carter et al. 1999). In arid regions, both males and females may select summer roosts in caves with high humidity (Caire et al. 1984). Most foraging activity takes place over forested waterways and along forest edges (Blair 1935, Davis and Mumford 1962, Paradiso 1969, LaVal et al. 1977, Schmidly et al. 1977, Caire et al. 1984, Krishon et al. 1997, Carter et al. 1999, Ford et al. 2005).

REPRODUCTION

Although breeding behavior has not been described for the eastern pipistrelle, copulation occurs in fall (Poole 1938, LaVal and LaVal 1980). Spermatozoa are stored in the uterus of females during winter and fertilization occurs at the time of ovulation in spring (Guthrie 1933b). From 1–7 eggs are shed; 4 fertilized eggs implant in the uterus (Wimsatt 1945). Two of the developing embryos usually are resorbed. The average litter of 2 young (Allen 1921, Brimley 1923, Wimsatt 1945, Lane 1946, Cope and Humphrey 1972) is born after a gestation period of 44 days (Wimsatt 1945). Births occur from early June–late July (LaVal and LaVal 1980). The development of the young is described by Lane (1946) and Hoying (1983). The

young begin flying at 3 weeks of age and are capable of adult-like foraging flights within a week of the first flight (Hoying 1983). In the southern United States, the eastern pipistrelle reaches sexual maturity at the age of 3–11 months (Davis 1963).

FOOD HABITS

Pipistrellus subflavus is among the earliest bats to begin foraging in the evening (Hamilton and Whitaker 1979, Caire et al. 1984, Winchell and Kunz 1996). They are distinguished by their slow, erratic flight while foraging (Lewis 1940, Patterson and Hardin 1969, Hoying 1983). The eastern pipistrelle feeds on small insects that range from 4–10 mm in length (Ross 1967). The known diet includes species of Coleoptera, Homoptera, Diptera, Hymenoptera, Hemiptera, and Lepidoptera (Sherman 1939, Ross 1967, Whitaker 1972, Carter et al. 1998, Menzel et al. 2002b).

ASSOCIATED SPECIES

Although the eastern pipistrelle roosts alone during hibernation, it often occupies the same hibernacula as the little brown myotis (*Myotis lucifugus*), Indiana myotis (*M. sodalis*), southeastern myotis (*M. austroriparius*), gray myotis (*M. grisescens*), northern myotis (*M. septentrionalis*), and big brown bat (*Eptesicus fuscus*; Guthrie 1933a, Swanson and Evans 1936, Folk 1940, Griffin 1940, Hitchcock 1949, Goehring 1954, Jennings and Layne 1957, Myers 1960, Hall 1963, Davis 1964, Fenton 1970, Mumford and Whitaker 1975, Menzel et al. 1997). During summer in western Oklahoma, the eastern pipistrelle shares caves with the cave myotis (*M. velifer*), Townsend's big-eared bat (*Corynorhinus townsendii*), Brazilian free-tailed bat (*Tadarida brasiliensis*), and big brown bat (Caire et al. 1984).

VULNERABILITY AND THREATS

Pipistrellus subflavus is widely distributed in the South and is relatively common throughout its range. There are no current threats to its existence. Because the species exhibits a tendency to utilize the same summer roosts each year (Griffin 1934), habitat disturbance may affect large numbers of the species in an area. Loss of *P. subflavus* has occurred due to cave flooding in Kentucky (DeBlase et al. 1965).

MANAGEMENT SUGGESTIONS

Management centers on the protection of winter hibernacula. Bat-compatible gates can prevent human disturbance at hibernation sites (Bat Conservation International 2001). Appropriate actions to exclude the eastern pipistrelle from dwellings may

be required (D. Saugey, U. S. Forest Service, personal communication). Additional research is needed on summer roosting and foraging habitat requirements of the species.

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Nine-banded Armadillo

Dasypus novemcinctus (Linnaeus, 1758)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Seven subspecies of nine-banded armadillo (*Dasypus novemcinctus*) are recognized (Wetzel and Mondolfi 1979, Hall 1981). Only one, *D. n. mexicanus*, occurs in the United States (Hall 1981). Wetzel and Mondolfi (1979) and Wetzel (1985) review the taxonomy of the armadillo. The literature is summarized by Galbreath (1982), McBee and Baker (1982), and Montgomery (1985).

DISTINGUISHING CHARACTERISTICS

The nine-banded armadillo is the most unique mammal in the South. The dorsum is covered with an armored carapace consisting of bony plates that are fused to form a scapular shield, nine flexible bands at mid-body, a pelvic shield, and a bony head plate. Twelve bony rings enclose the tail. The remainder of the scantily haired body is covered by a leathery epidermis. Measurements are: total length, 600–800 mm; tail, 245–370 mm; hind foot, 75–107 mm; ear, 30–40 mm; weight, 3–8 kg. The back and sides are brownish-gray and the underside is light gray to yellowish. The toes have long, white claws. The skull is elongate and is characterized by a narrow, extended rostrum. Incisors and canines are absent. The peg-like cheek teeth vary in number from seven to nine on each side of the upper and lower jaws. The dental formula is: I 0/0, C 0/0, P 4–6/4–6, M 3/3 = 28–36 (Figure 1).

CONSERVATION STATUS

The nine-banded armadillo has a global rank of Secure (NatureServe 2007). It is Secure in Alabama, Louisiana, Mississippi, and Texas and Apparently Secure in Arkansas, Georgia and Oklahoma. It is Vulnerable in Tennessee. It is unranked in Florida and South Carolina. North Carolina has assigned it a conservation rank of Not Applicable (i.e., the species is not considered a suitable target for conservation).

DISTRIBUTION

Known only from southern Texas before the turn of the 20th century (Cope 1880, Bailey 1905), the armadillo rapidly expanded its range throughout the southern United States (Talmage and Buchanan 1954, Humphrey 1974, Galbreath 1982, McBee and Baker 1982). From its original range, the species spread

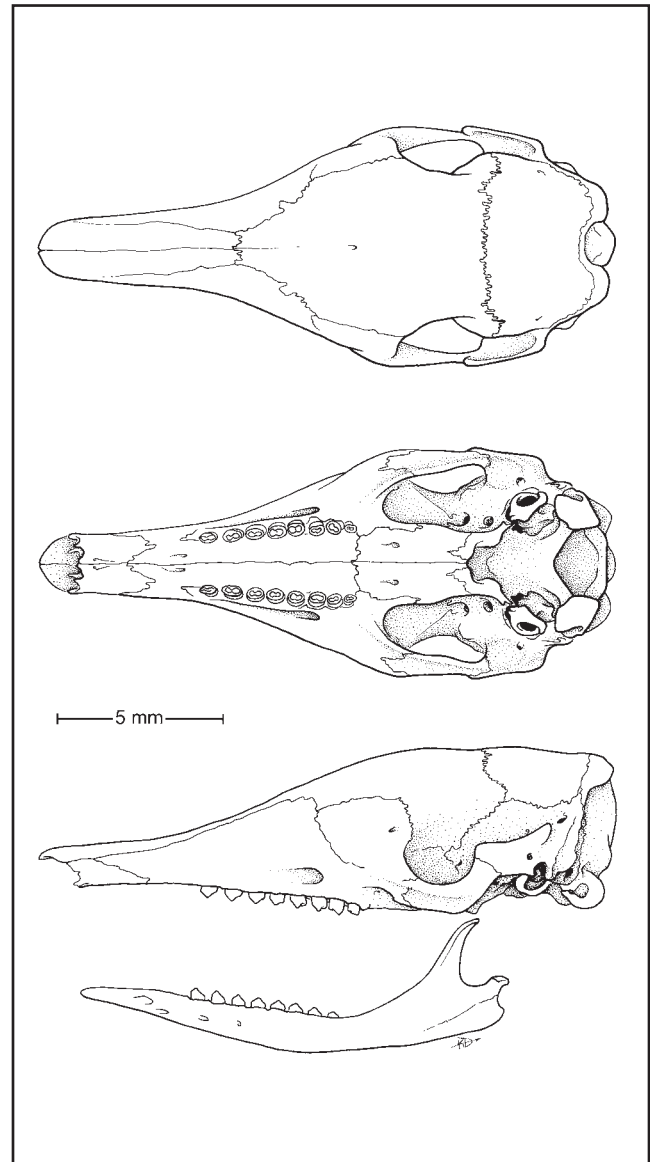


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Dasypus novemcinctus* from Wakulla County, Florida (USNM 527296, female).

northward throughout central and eastern Texas (Hollander et al. 1987, Jones and Jones 1992, Jones et al. 1993, Davis and Schmidly 1994) and Oklahoma (Gardner 1948, Caire et al. 1989). The species dispersed eastward throughout Louisiana (Lowery 1974), Arkansas (Sealand and Heidt 1990, Sikes et al. 1991), western Tennessee (Henning 1980,

Kennedy 1991), southern and western Mississippi (Kennedy et al. 1974, Jones and Carter 1989), and southern Alabama (Humphrey 1974, Breece and Dusi 1985). The nine-banded armadillo was introduced into Florida in the early 1900s (Bailey 1924, Talmage and Buchanan 1954) and with exception of the Everglades, expanded its range throughout the state (Humphrey 1974, McDonough and Loughry 1997, Inbar and Mayer 1999). The species moved north from Florida into the Coastal Plain and coastal islands of Georgia (Golley 1962, Humphrey 1974, Laerm et al. 1982, Bond et al. 2000, Osborn et al. 2000) and continued northward to the Fall Line of South Carolina (Mayer 1989, Platt and Snyder 1995) and Georgia. Today, western and eastern populations are continuous across western Florida and Alabama (Figure 2). The rapid expansion is attributed to progressively warmer climatic conditions, habitat modification, and removal of large carnivores (Humphrey 1974, McBee and Baker 1982). Its western and northward expansion may be limited by aridity and cold (McBee and Baker 1982, Taulman and Robbins 1996).

ABUNDANCE STATUS

The nine-banded armadillo is common to abundant throughout the main portion of its range, but becomes increasingly less so toward the northern limits. Population density estimates range broadly from 0.05–3.0/ha (Kalmbach 1943, Taylor 1946, Galbreath 1982). In Louisiana, mean home ranges of 20 ha have been recorded, whereas in Florida, ranges of 1.1–13.8 ha have been measured (Whitaker and Hamilton 1998).

PRIMARY HABITATS

Dasypus novemcinctus occurs in several habitats including pine (*Pinus* spp.) savanna, southern scrub oak (*Quercus* spp.), sand pine (*P. clausa*), longleaf pine (*P. palustris*), mesic hardwoods, slash pine (*P. elliottii*), oak-gum-cypress (*Nyssa-Taxodium*), and maritime live oak (*Q. virginiana*). It occurs in various grassland communities, agricultural areas, and orchards. The armadillo tends to concentrate near streams, rivers, or other water sources. Much of its dispersal is attributed to its use of riverine and bottomland corridors (Humphrey 1974, McBee and Baker 1982). During the winter months in Florida, dense woodlands were centers of activity (Inbar and Mayer 1999). Nine-banded armadillos construct burrows and nest in underground dens (Bond et al. 2000).

REPRODUCTION

Several aspects of armadillo reproduction are unique among mammals of the region. Although ovulation and breeding activities are concentrated in summer

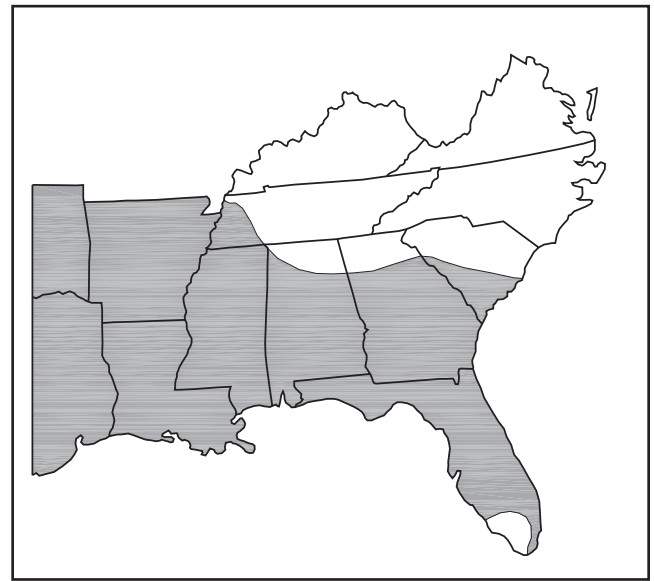


Figure 2. Distribution of *Dasypus novemcinctus* in the South.

(June–August), implantation of the fertilized egg is delayed until late November or December. After implantation in the uterine wall, a single blastocyst typically splits to form 4 embryos (i.e., monozygotic polyembryony). After several months of gestation, a litter of 4 identical young are born between March–April. The young develop rapidly, are capable of walking within hours, and follow their mother within weeks. The young remain with the mother for several months. Individuals do not reproduce until their second year (Buchanan 1957, Enders 1966, Galbreath 1982, McBee and Baker 1982).

FOOD HABITS

Dasypus novemcinctus is an opportunistic insectivore that feeds primarily on invertebrates such as beetles, ants, caterpillars, centipedes, millipedes, snails, and slugs that live in soil, litter, and decomposing wood. The diet varies by season and availability; fruits may be locally important during summer. Vertebrates, particularly reptiles, may comprise up to 25% of the diet in winter (Whitaker and Hamilton 1998). The armadillo also consumes bird eggs, carrion, and small quantities of vegetable matter (Kalmbach 1943, McBee and Baker 1982, Breece and Dusi 1985, Wirtz et al. 1985, Sikes et al. 1991, Tyler et al. 1996, Osborn et al. 2000). The species relies heavily on a keen sense of smell and powerful digging claws while searching for food.

ASSOCIATED SPECIES

Nine-banded armadillos occur in association with numerous small mammals of the Coastal Plain

region. It sometimes shares its burrow systems with the Virginia opossum (*Didelphis virginiana*), eastern cottontail (*Sylvilagus floridanus*), marsh rabbit (*S. palustris*), swamp rabbit (*S. aquaticus*), hispid cotton rat (*Sigmodon hispidus*), or striped skunk (*Mephitis mephitis*). It is preyed upon by the feral dog (*Canis familiaris*) and coyote (*C. latrans*).

VULNERABILITY AND THREATS

The species is widespread and abundant in the region; there appear to be no threats to its survival. Mortality on the highway probably accounts for more deaths than does predation (Whitaker and Hamilton 1998). Prolonged drought may result in increased adult mortality; most juvenile mortality is due to predation (McDonough and Loughry 1997). The armadillo cannot survive prolonged freezing weather.

MANAGEMENT SUGGESTIONS

The nine-banded armadillo is an important control agent of destructive agricultural arthropod pests. It is also sometimes used for human food and is important in leprosy research. The armadillo does not require active management other than population control for agricultural or suburban damage. The species prefers to have its burrow located in dense cover; removal of brush or other such cover discourages establishment. Fencing or barriers may exclude the armadillo under certain conditions. Additional research is needed on the early stages of development and variation among geographic areas.

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Snowshoe Hare

Lepus americanus (Erxleben, 1777)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Fifteen subspecies of snowshoe hare (*Lepus americanus*) are recognized (Hall 1981, Bittner and Rongstad 1982). Only one subspecies, *L. a. virginianus*, occurs in the South. Populations in Virginia and adjacent areas of West Virginia were augmented by introductions of *L. a. struthopus*. These introductions generally were unsuccessful and there is no evidence that the native genotype was impacted (Brooks 1955, Handley 1979, 1991; Fies 1991). Literature on the species is reviewed by Bittner and Rongstad (1982); its status in the South is discussed by Handley (1979) and Fies (1991).

DISTINGUISHING CHARACTERISTICS

Snowshoe hares are large lagomorphs. Measurements are: total length, 360–520 mm; tail, 25–55 mm; hind foot, 112–150 mm; ear, 62–70 mm; weight, 1.0–2.3 kg. The pelage of the snowshoe hare varies by season. During summer, snowshoe hares are rusty brown above with a dark brown to black snout. The nostrils are edged in white, the chin and belly are white to grayish, the tail is white above and gray below, and the ears are tipped in black. The winter pelage is pure white except for black-tipped ears and a brownish wash on the feet. The interparietal bone is indistinct in all *Lepus* species, and is fused to the parietals. The dental formula is: I 2/1, C 0/0, P 3/2, M 3/3 = 28 (Figure 1). Snowshoe hares may be readily distinguished from sympatric eastern cottontails (*Sylvilagus floridanus*) and Appalachian cottontails (*S. obscurus*) on the basis of size. The cottontails are smaller, have larger and whiter tails, and the dorsal pelage of both is neither as white in winter or as rusty in summer.

CONSERVATION STATUS

The snowshoe hare has a global rank of Secure (NatureServe 2007). It is Critically Imperiled in Virginia and Presumed Extirpated in North Carolina. Snowshoe hares are a regulated game species with a hunting season in West Virginia.

DISTRIBUTION

Lepus americanus is distributed throughout the transcontinental coniferous forests of the Canadian Arctic from Alaska to Newfoundland south into the Rocky Mountains, extreme northern portions of the Great

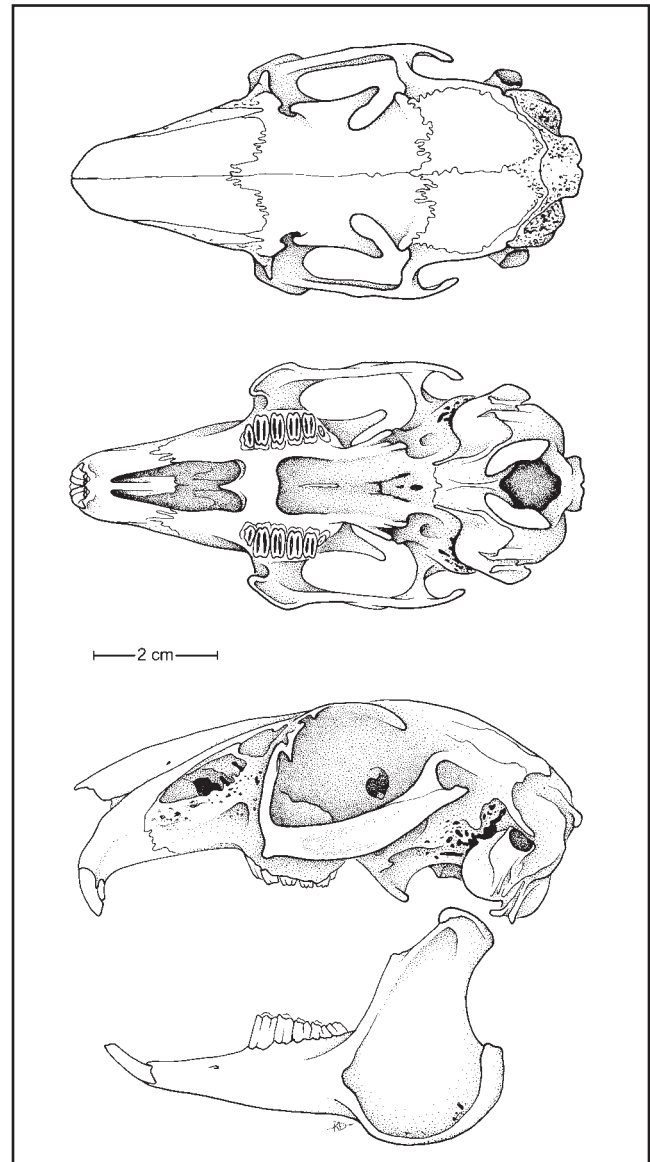


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Lepus americanus* from Sagadahoc County, Maine (USNM 507455, male).

Lakes, and New England south to Pennsylvania (Genoways 1985). The distribution of the snowshoe hare in the South is depicted in Figure 2. It is the only hare native to the eastern United States. It may have once ranged south through the higher portions of the Blue Ridge to Tennessee and North Carolina (Hall 1981, Bittner and Rongstad 1982, Linzey 1995), but

the extent of its current southern Appalachian distribution is uncertain. Based upon available evidence, it is not present in Maryland (Mansueti 1953, Paradiso 1969, Lee 1984). Populations in the region are restricted to eastern West Virginia (McKeever 1952, Brooks 1955, Rieffenberger 1966, Fies 1991) and three localities in northwestern Highland County, Virginia (Fies 1991, Linzey 1998). Anecdotal information obtained from local residents during Kellogg's (1939) review of Tennessee mammals formed the basis for Hall and Kelson's (1959) marginal records and later reports (Blair et al. 1968, Bittner and Rongstad 1982) indicating that the species ranged as far south as the Great Smoky Mountains of Tennessee and North Carolina. Linzey and Linzey (1971) commented that the species was not present in the Great Smoky Mountains National Park and they doubted that it occurred there before the park was established.

ABUNDANCE STATUS

Handley (1979, 1991) and Fies (1991) suggest that prior to the overharvest of red spruce (*Picea rubens*) forests in Virginia in the early 1900s, the snowshoe hare probably was more widespread in western Virginia and adjacent West Virginia. In 1978, estimates of snowshoe hare populations in Virginia numbered from a few dozen to hundreds of hares (Handley 1979). In 1991, there were reports that there were less than a few dozen hares in a small, forested tract of approximately 25 km² in Highland County (M. Fies, Virginia Department of Game and Inland Fisheries, personal communication). However, the snowshoe hare population in Highland County is part of the metapopulation in the Allegheny Mountains of West Virginia where the species occupies an area that may include 200,000 to 400,000 ha.

PRIMARY HABITATS

The habitat of the snowshoe hare in the South is restricted to areas that are now or have been dominated by red spruce (Brooks 1955, Rieffenberger 1966, Fies 1991). Although most relict stands of red spruce have been removed from Virginia, there are pockets of red spruce regeneration; the snowshoe hare survives in these and second growth forests of northern hardwood cover types where thick stands of rhododendron (*Rhododendron* spp.) and mountain laurel (*Kalmia latifolia*) dominate the understory (Brooks 1955, Fies 1991). Adequate understory may be the most critical aspect of snowshoe hare habitat (Rieffenberger 1966, Brocke 1975, Conroy et al. 1979, Buehler and Keith 1982, Fies 1991). In Virginia and adjacent West Virginia, spruce thickets, extensive tangles of rhododendron, mountain laurel, blueberry (*Vaccinium* spp.), and yellow birch (*Betula alleghaniensis*)

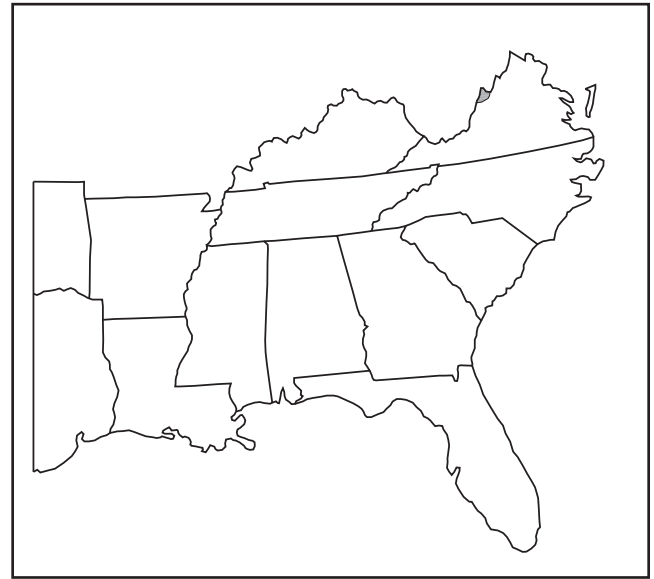


Figure 2. Distribution of *Lepus americanus* in the South.

provide preferred cover (Handley 1979, Fies 1991). Brocke (1975) and Wolfe et al. (1982) suggest the most suitable hare habitat contains horizontal vegetation densities of 40–100% (with 0–60% visibility). In areas where woody cover is sparse, patchy habitat and continuity of cover are important for snowshoe hare movements (Brocke 1975, Conroy et al. 1979).

REPRODUCTION

The reproductive biology of the snowshoe hare has been studied extensively (Bittner and Rongstad 1982). The hare breeds from February to mid-August with the onset of breeding correlated with weather and light (Severaid 1942, Meslow and Keith 1971, Cary and Keith 1979). The species is polyestrous and postpartum breeding permits up to 4 litters/season. Gestation ranges from 34–40 days (Severaid 1942, Meslow and Keith 1971). Litter size varies by latitude, with southern populations having smaller litter sizes than those to the north. Litter size in the South is unknown, but probably ranges between 1–5 (Keith et al. 1966). Young are weaned at 4 weeks and typically breed the spring following their birth (Keith and Meslow 1967).

FOOD HABITS

The diet of snowshoe hares varies by season (Brooks 1955, Rieffenberger 1966, Handley 1979, Fies 1991). In summer, snowshoe hares graze on a variety of grasses, clover, herbs, and tender parts of woody plants. In winter, *Vaccinium* spp. is a predominant food source along with the buds, bark, twigs, shoots of eastern hemlock (*Tsuga canadensis*), red spruce,

rhododendron, maple (*Acer* spp.), birch, and serviceberry (*Amelanchier* spp.).

ASSOCIATED SPECIES

Because of its limited distribution and restricted habitat in the South, the snowshoe hare likely occurs only with boreomontane mammals. These include the masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), northern short-tailed shrew (*Blarina brevicauda*), Appalachian cottontail, northern flying squirrel (*Glaucomys sabrinus*), red squirrel (*Tamiasciurus hudsonicus*), deer mouse (*Peromyscus maniculatus*), southern bog lemming (*Synaptomys cooperi*), southern red-backed vole (*Clethrionomys gapperi*), and woodland jumping mouse (*Napaeozapus insignis*).

VULNERABILITY AND THREATS

Loss of habitat due to even-aged succession of forests is the primary threat to remaining populations of snowshoe hares in Virginia (Fies 1991). In addition, the population of snowshoe hares in Virginia is separated from those to the north by at least 150 km. The absence of timber harvesting has resulted in the loss of second growth forests, canopy closure, and sparse understory vegetation (Krebs et al. 1995). Browsing by white-tailed deer (*Odocoileus virginianus*) may decrease food and cover for snowshoe hares (Bookout 1965). Duration of snow cover may limit the distributional range of the species.

MANAGEMENT SUGGESTIONS

The snowshoe hare requires suitable cover conditions that can be provided by young, regenerating forests (particularly red spruce). Active management requires periodically thinned forest stands to encourage the regenerative growth that provides suitable habitat conditions. Habitat conditions can be improved by thinning stands with dense overstories where mountain laurel survives in the understory and creating small openings in dense cover for summer foraging (Fies 1991).

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Black-tailed Jackrabbit

Lepus californicus (Gray, 1837)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Seventeen subspecies of black-tailed jackrabbit (*Lepus californicus*) are recognized (Hall 1981, Dunn et al. 1982). Two subspecies (*L. c. melanotis* and *L. c. merriami*) occur in the South. The literature on the black-tailed jackrabbit is reviewed by Dunn et al. (1982) and Best (1996).

DISTINGUISHING CHARACTERISTICS

Lepus californicus is the largest lagomorph in the region. Measurements are: total length, 465–630 mm; tail, 50–112 mm; hind foot, 112–145 mm; ear, 99–131 mm; weight, 1.5–3.5 kg. The dorsal pelage is gray or grayish brown and washed with black. The sides are grayish and the abdomen is white. The tail is black dorsally and white ventrally. The ears are tipped in black. The black-tailed jackrabbit occurs sympatrically with the eastern cottontail (*Sylvilagus floridanus*) and the swamp rabbit (*S. aquaticus*), from which it is readily distinguishable on the basis of its larger ears, hind feet, overall size, and black tail. The dental formula is: I 2/1, C 0/0, P 3/2, M 3/3 = 28 (Figure 1).

CONSERVATION STATUS

The black-tailed jackrabbit has a global rank of Secure (NatureServe 2007). It is Secure in Texas and Oklahoma, but Vulnerable in Arkansas. A conservation status rank of Not Applicable has been assigned by Florida and Virginia; the species is not a suitable target for conservation activities in those states.

DISTRIBUTION

The black-tailed jackrabbit is the most common jackrabbit in the western United States. It ranges from the Pacific Northwest east to the grasslands of South Dakota and south through western Missouri and Arkansas into Texas and Mexico (Hall 1981). The distribution of the jackrabbit in the South is depicted in Figure 2. Populations occur in the Ozark Highlands, Arkansas Valley, and Boston Mountains of northwestern Arkansas (Sealander and Heidt 1990). The species is common throughout western Oklahoma, but it is known from only a few records in the eastern and none from southeastern portion of the state (Caire et al. 1989). Schmidly (1983), Cleveland et al. (1984), and Davis and Schmidly (1994) report that the

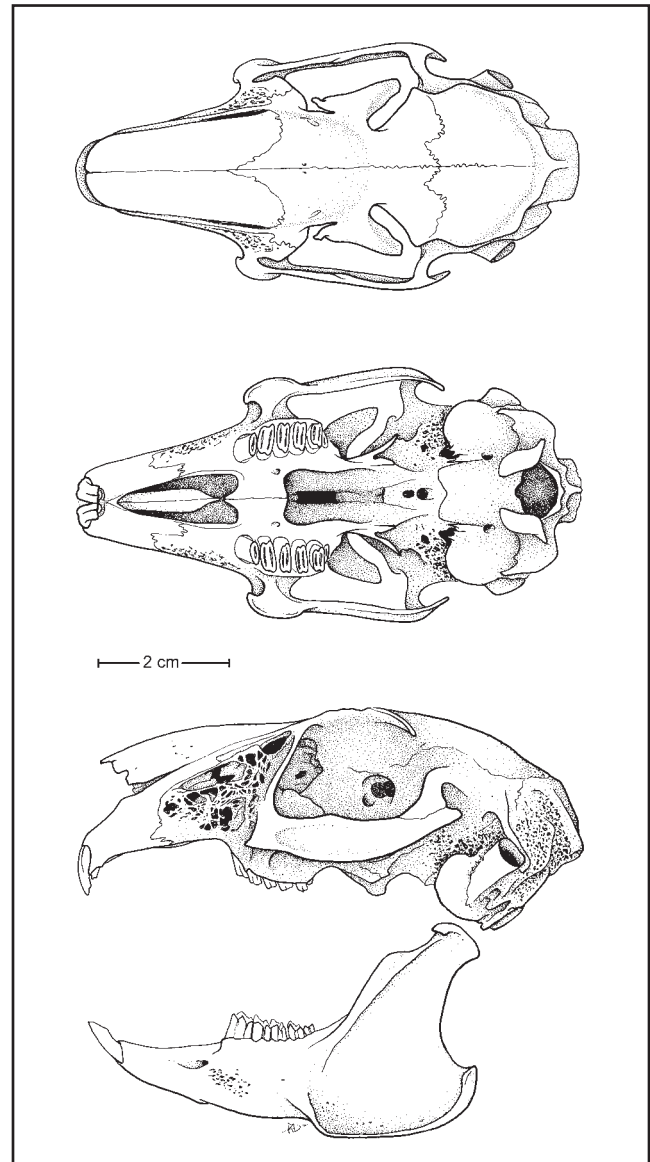


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Lepus californicus* from Crowley County, Colorado (USNM 564055, female).

species is characteristic of western Texas and coastal prairies. However, only disjunct populations of the species occur in eastern Texas, where some populations may be the result of translocations (McCarley 1959). Unsuccessful introductions have been made into southern Florida (Layne 1965, 1974), where the

jackrabbits were restricted to Dade and Broward counties, as well as Mercer, Pendleton, and Hancock counties, Kentucky (Myers 1952 in Clapp et al. 1976). Populations were introduced to the Eastern Shore of Maryland (Chapman and Sandt 1977) and Virginia where a population is now restricted to Cobb Island (Clapp et al. 1976, Dueser et al. 1979, Fies 1991, Linzey 1998).

ABUNDANCE STATUS

Populations of black-tailed jackrabbits in the region are at the eastern margin of their range. No density estimates are available. Sealander and Heidt (1990) indicate that *L. californicus* is reported from only a few counties in Arkansas (Black 1936); it is locally common only in the extreme northwestern part of the state. Schmidly (1983) reports that black-tailed jackrabbits are common in blackland and coastal prairies, but uncommon elsewhere. Similarly, it is rare in eastern Oklahoma (Caire et al. 1989).

PRIMARY HABITATS

The species occupies a diversity of habitats as reflected in its broad distribution throughout the West and Great Plains grasslands (Dunn et al. 1982). The black-tailed jackrabbit is common in arid short-grass rangelands, desert shrub habitats, sagebrush-creosote communities, and agricultural lands. Timberland and tall grass habitats typically are avoided. It occurs in the prairie peninsulas of western Arkansas and Oklahoma where Sealander and Heidt (1990) and Caire et al. (1989) suggest that following widespread clearing of forested land for agriculture, the species expanded its range into open grasslands, pasture, and cultivated areas. In Texas, it is associated with prairie communities of the blackland and coastal prairies, but rare in the oak-hickory (*Quercus-Carya*) and pine-oak (*Pinus-Quercus*) cover types. Schmidly (1983) noted that the occurrence of black-tailed jackrabbits in eastern Texas is a recent phenomenon associated with timber cutting and land-clearing of the past 40–50 years. By day, black-tailed jackrabbits rest in a shallow depression hollowed out in clumps of grass at the base of a shrub or in open grassland.

REPRODUCTION

The reproductive biology is reviewed by Dunn et al. (1982). No information is available on populations in the region. Elsewhere, the length of the breeding season varies depending upon latitude and environmental factors, but extends from February through August (Orr 1940, Haskell and Reynolds 1947, Bronson and Tiemeier 1958, Lechleitner 1959, Gross et al. 1974). The species is polyestrous and characterized by postpartum

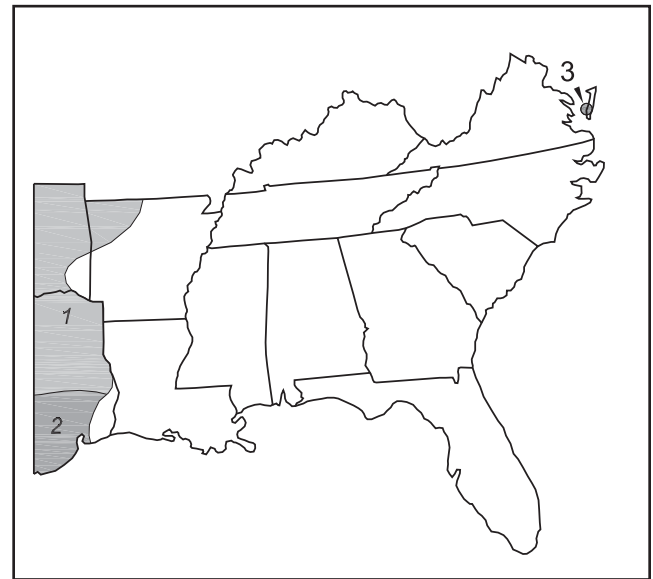


Figure 2. Distribution of *Lepus californicus* in the South: (1) *L. c. melanotis*; (2) *L. c. merriami*; (3) Introduced Population (Subspecies unknown).

breeding with 2–7 litters/season depending upon latitude and environmental factors (Feldhamer 1979). Gestation ranges from 41–47 days (Haskell and Reynolds 1947). Litter size varies by latitude and southern populations have smaller litter sizes than those to the north. Litter size in the South is unknown, but it is reported to be 2.8 in southwest Kansas (Dunn et al. 1982). Most breeding begins in the second year (Haskell and Reynolds 1947, Bronson and Tiemeier 1958, but see Gross et al. 1974).

FOOD HABITS

The diet varies depending upon locality and availability (Dunn et al. 1982). The black-tailed jackrabbit selects succulent vegetation when it is available. In summer, it feeds on grasses, sedges, forbs, and herbs, but the species focuses on shrubs in winter (Lechleitner 1958, Hansen and Flinders 1969, Flinders and Hansen 1972, Dunn et al. 1982).

ASSOCIATED SPECIES

The black-tailed jackrabbit is expected to occur with other open grassland species such as the least shrew (*Cryptotis parva*), southern short-tailed shrew (*Blarina carolinensis*), Elliot's short-tailed shrew (*B. hylophaga*), eastern cottontail, hispid pocket mouse (*Chaetodipus hispidus*), harvest mouse (*Reithrodontomys* spp.), deer mouse (*Peromyscus maniculatus*), and hispid cotton rat (*Sigmodon hispidus*). The coyote (*Canis latrans*) and white-tailed deer (*Odocoileus virginianus*) also commonly occupy the same habitats as the black-tailed jackrabbit.

VULNERABILITY AND THREATS

Populations of *L. californicus* are scattered and isolated at the extreme eastern margin of their range. The occurrence of the species in the region is largely a consequence of the clearing of forests for agricultural purposes. Pastureland and agricultural openings have provided avenues for dispersal eastward from the prairies. Most populations are secure because open grassland habitats are now extensive throughout the region. However, some isolated populations of the black-tailed jackrabbit may be vulnerable if succession results in the loss of open grassland habitats.

MANAGEMENT SUGGESTIONS

Prescribed fire in coniferous forest stands benefits the jackrabbit by reducing overstories, creating edge habitat, and enhancing grass and forb production. In areas of crop depredation, management centers on population control: trapping, hunting, and removing habitat cover.

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Swamp Rabbit

Sylvilagus aquaticus (Bachman, 1837)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Two subspecies of the swamp rabbit (*Sylvilagus aquaticus*) are recognized by Hall (1981) and Chapman and Feldhamer (1981). Both subspecies, *S. a. aquaticus* and *S. a. littoralis*, occur in the South. Although we include it here, the validity of *S. a. littoralis*, restricted to the lower Coastal Plain of Mississippi, Louisiana, and Texas (Nelson 1909), was questioned by Lowery (1974) and the taxon was rejected by Schmidly (1983). Lowe (1958) and Jenkins and Provost (1964) suggested that *S. aquaticus* and the marsh rabbit (*S. palustris*) interbreed in portions of Georgia and South Carolina. In some parts of the region, swamp rabbits also are called “cane cutters.” Chapman and Feldhamer (1981) and Chapman et al. (1982) review the literature on the species.

DISTINGUISHING CHARACTERISTICS

The swamp rabbit is the largest of the *Sylvilagus* species. Measurements are: total length, 450–550 mm; tail, 50–74 mm; hind foot, 90–113 mm; ear, 60–80 mm; weight, 1.6–2.6 kg. The dorsal parts of the head and body of swamp rabbits are rusty brown to black. The nape is a dark cinnamon and the undersurface of the throat, abdomen, and tail is white. Swamp rabbits have a black spot between their ears. The basilar length of the skull is usually greater than 63 mm, larger than other *Sylvilagus*. The dental formula is: I 2/1, C 0/0, P 3/2, M 3/3 = 28 (Figure 1). Swamp rabbits may be confused with marsh rabbits, but the latter are usually distinguishable because of their smaller size, shorter ears, small slender feet, and grayish tail. Eastern cottontails (*S. floridanus*) also often occupy the same habitats as swamp rabbits. Eastern cottontails are smaller than swamp rabbits and lack the black spot between the ears (Bond et al. 2000).

CONSERVATION STATUS

The swamp rabbit has a global rank of Secure (NatureServe 2007). It is also Secure in Alabama, Georgia, Louisiana, Mississippi, and Texas. It is Apparently Secure in Tennessee, Vulnerable in Arkansas and Kentucky, and Imperiled in Oklahoma and South Carolina.

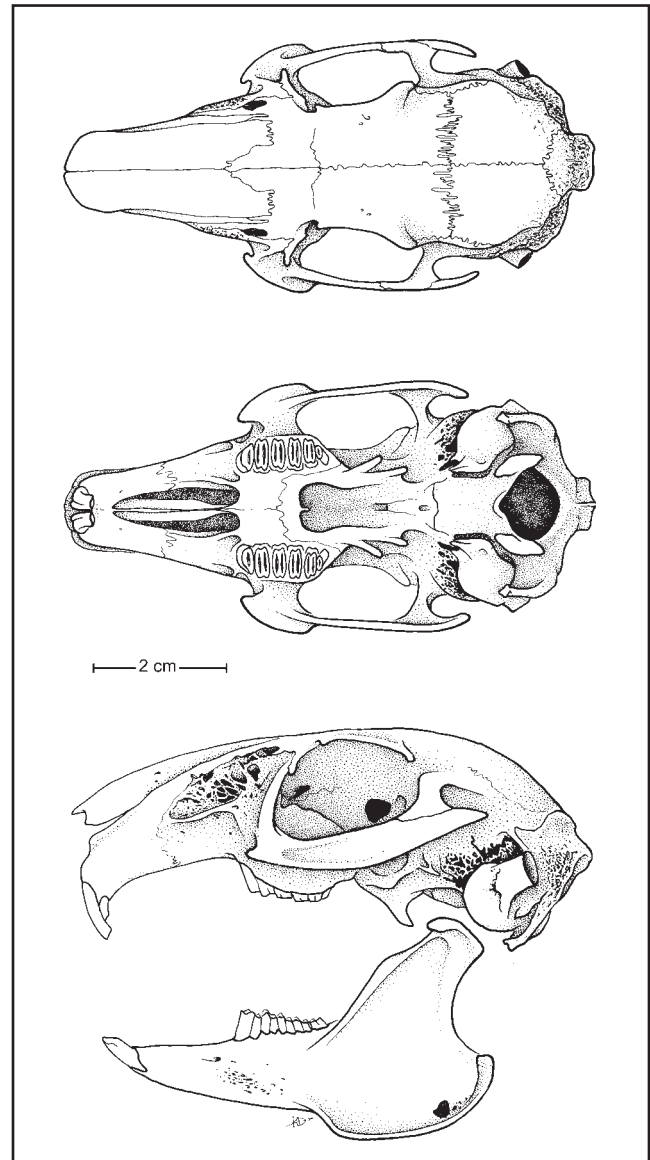


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sylvilagus aquaticus* from Stoddard County, Missouri (USNM 349485, female).

DISTRIBUTION

The range of *S. aquaticus* is contiguous with that of the marsh rabbit to the south. It is known from a single record in the Blue Ridge Province of North Carolina

(Lee et al. 1982, Betsill 1987). It occurs in the Blue Ridge and upper Piedmont of South Carolina (Sherman 1939, Jenkins and Provost 1964, Golley 1966, Cothran et al. 1991, Platt and Bunch 2000), the Piedmont and Ridge and Valley of Georgia (Lowe 1958, Golley 1962, Wharton et al. 1981, Laerm et al. 1982) and west throughout all but extreme southeastern Alabama (Holliman 1963, Hill 1967, Linzey 1970, McCollum and Holler 1994), into Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989, Palmer et al. 1991), Louisiana (Lowery 1974, Heuer and Perry 1976), Arkansas (Sealander and Heidt 1990, Zollner et al. 1996), eastern Texas (Schmidly 1983, Jones and Jones 1992, Davis and Schmidly 1994), and eastern Oklahoma (Caire et al. 1989). It ranges up the Mississippi River Valley into western Tennessee (Goodpaster and Hoffmeister 1952, Kennedy and Harvey 1980, Kennedy 1991) and Kentucky (Barbour and Davis 1974). The distribution of the swamp rabbit in the South is depicted in Figure 2.

ABUNDANCE STATUS

Despite its wide distribution, little specific information on abundance is available. The species is common in suitable habitat from Georgia west to eastern Texas. Published density estimates include approximately 0.1/ha in bottomlands of Georgia (Lowe 1958), 0.4/ha in southern Indiana (Harrison and Hickie 1931, Terrel 1972), 0.3/ha in Texas (Davis 1974), and 1.2/ha in western Kentucky (Barbour and Davis 1974). The species is rare to uncommon in South Carolina (Platt and Bunch 2000). Populations of *S. aquaticus* are declining in Tennessee (Kennedy and Harvey 1980), Oklahoma (Caire et al. 1989), and Arkansas (Sealander and Heidt 1990). These population declines are related to the conversion of the forested habitats that this species prefers to agricultural or human development (Smith et al. 1993).

PRIMARY HABITATS

The swamp rabbit is associated with southern swamp forest communities, predominantly oak-gum-cypress (*Quercus-Nyssa-Taxodium*) and elm-ash-cottonwood (*Ulmus-Fraxinus-Populus*) forest types. It usually is restricted to small sloughs, low ridges, and grassy marshes associated with bottomlands and floodplains of large and small rivers, streams, and swamps (Svihla 1929, Lowe 1958, Hunt 1959, Hill 1967, Terrel 1972, Fredrickson 1980, Schmidly 1983, Allen 1985, Caire et al. 1989, Sealander and Heidt 1990, McCollum and Holler 1994, Zollner 2000a,b). In such habitats, it occurs in thickets of greenbrier (*Smilax* spp.) and honeysuckle (*Lonicera* spp.), cane (*Arundinaria* spp.), and dense shrub thickets both in mature and early forest stands (Chapman and Feldhamer 1981,

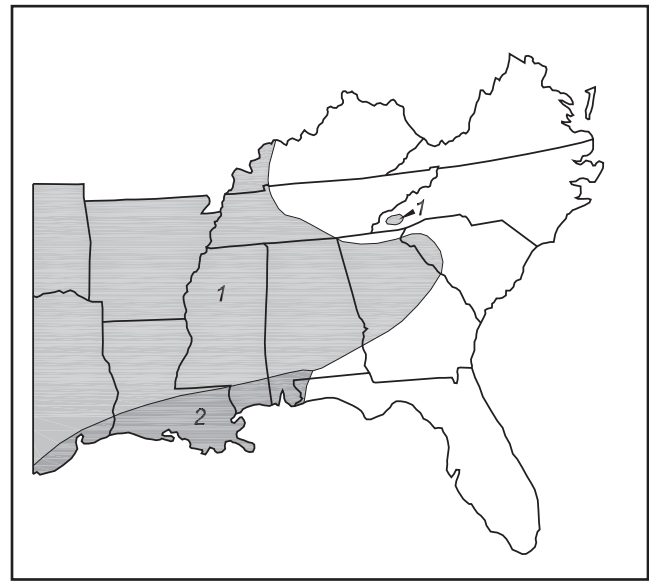


Figure 2. Distribution of *Sylvilagus aquaticus* in the South: (1) *S. a. aquaticus*; (2) *S. a. littoralis*.

Chapman et al. 1982). Forms and runways occur in brushy vegetation. Logs are used as latrines (Zollner et al. 1996). It also uses ground or tree holes as form sites (Hunt 1959). The eastern cottontail in dry upland and grassy areas (Toll et al. 1960, Terrel 1972) generally replaces it.

REPRODUCTION

The species is a synchronous breeder (Sorensen et al. 1968). The breeding season is longest in the South and progressively shorter to the north (Hunt 1959). Breeding begins as early as February. First litters are produced in March and the last in September; there is a depression in breeding activity between September and December (Svihla 1929, Lowe 1958, Toll et al. 1960, Hill 1967, Sorensen et al. 1968). However, the swamp rabbit may breed year-round in Texas (Hunt 1959). Gestation varies from 35–40 days (Hunt 1959, Holler et al. 1963, Sorensen et al. 1968). Mean litter size ranges from 2.8–3.7 (Lowe 1958, Hunt 1959, Toll et al. 1960, Palmer et al. 1991). Individuals breed at 23–30 weeks (Sorensen et al. 1968, but see Hunt 1959). One to five litters are produced per year (Holler et al. 1963, Sorensen et al. 1968).

FOOD HABITS

The swamp rabbit feeds on various plants including emergent aquatic vegetation, grasses, sedges, cane, tree seedlings, bark, and twigs (Svihla 1929, Toll et al. 1960, Terrel 1972). The species will often forage on cultivated crops, such as corn (*Zea mays*) and sorghum (*Sorghum* spp.), where the fields adjoin areas in which the rabbit lives. Swamp rabbits also engage

in coprophagy (Hamilton 1955, Layne 1958, Toll et al. 1960).

ASSOCIATED SPECIES

The swamp rabbit occurs in association with other swamp forest and marshland species including the southeastern shrew (*Sorex longirostris*), southern short-tailed shrew (*Blarina carolinensis*), marsh rice rat (*Oryzomys palustris*), cotton mouse (*Peromyscus gossypinus*), and hispid cotton rat (*Sigmodon hispidus*). The distributional range of the swamp rabbit is contiguous with that of the marsh rabbit.

VULNERABILITY AND THREATS

Hardwood swamp and river bottom habitats have decreased in the region as a result of wetland drainage, stream channelization, and clearing of forests. Habitat reduction for farming and other uses appears to be the major problem for this species. The rabbit may now be rare or absent where it once was common.

MANAGEMENT SUGGESTIONS

The restoration of forested wetlands in much of the range of the swamp rabbit is critical to the survival of the species. The protection of bottomlands, wooded swamps and marshes, estuaries, and tributaries of rivers and streams is beneficial. Maintenance of high quality escape cover (e.g., thickets of cane, privet [*Ligustrum* spp.], blackberry, and greenbrier) and abundant food sources are essential for viable populations of the swamp rabbit. This is one of the least studied species of *Sylvilagus*, and additional research is warranted in the South.

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Eastern Cottontail

Sylvilagus floridanus (Allen, 1890)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Twenty-three subspecies of the eastern cottontail (*Sylvilagus floridanus*) are recognized in North America (Chapman et al. 1980, Hall 1981). Seven of the subspecies, *S. f. alacer*, *S. f. ammophilus*, *S. f. floridanus*, *S. f. hitchensi*, *S. f. mallurus*, *S. f. mearnsi*, and *S. f. paulsoni*, are known in the South. As a result of numerous translocations, subspecies distinctions may be meaningless (Chapman and Morgan 1973). The literature is reviewed by Chapman et al. (1980) and Chapman et al. (1982).

DISTINGUISHING CHARACTERISTICS

Sylvilagus floridanus is a medium-sized cottontail. Measurements are: total length, 375–463 mm; tail, 25–65 mm; hind foot, 85–105 mm; ear, 49–68 mm; weight, 0.8–1.5 kg. The color of the dorsum is ochraceous buff overlain by a black wash that gives the appearance of finely streaked penciling. The sides are less heavily washed with black and are slightly paler than the dorsum. The venter is whitish to light gray. The ears are long and pointed, and lack black edging along the anterior outer borders. A white spot is usually present on the forehead, but there is no black spot between the ears. A rusty patch is present on the middle of the nape. The postorbital processes are thick and are in broad contact with the skull at their tips. The sutures between the frontal and nasal bones are smooth in outline. The dental formula is: I 2/1, C 0/0, P 3/2, M 3/3 = 28 (Figure 1). The pelage and cranial features of the eastern cottontail and the Appalachian cottontail (*S. obscurus*) are similar.

CONSERVATION STATUS

The eastern cottontail has a global rank of Secure (NatureServe 2007). The species is also considered Secure in those states where it occurs within the region, except for Arkansas where it is Apparently Secure. It is unranked in Florida and South Carolina.

Sylvilagus f. hitchensi, restricted to Smith and Fisherman's Islands, Northampton County, Virginia, is known only from historical records; no specimens have been documented in the past 80 years (Fies 1991).

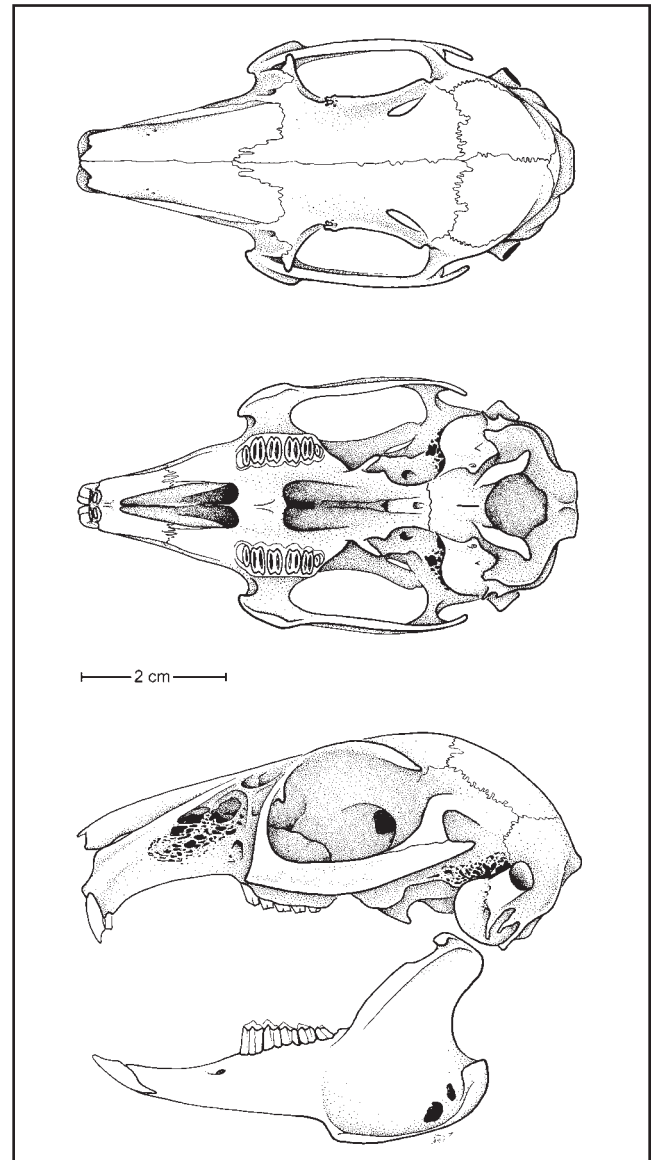


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sylvilagus floridanus* from Monroe County, Arkansas (USNM 551500, male).

DISTRIBUTION

The eastern cottontail has the largest distribution of any North American rabbit. Its geographical range extends from southern Canada throughout the United States from New England to the Upper Great Plains and south into Central and South America (Hall 1951, 1981; Chapman et al. 1980, Diersing and

Wilson 1980). The eastern cottontail has been widely translocated (Chapman et al. 1982). Its range overlaps other *Sylvilagus* species in the region. The eastern cottontail occurs throughout Virginia (Mearns 1911, Bailey 1946, Handley and Patton 1947, Handley 1979, 1991, 1992; Jackson et al. 1976, Jacobson et al. 1978, Dueser et al. 1979, Webster et al. 1985, Pagels et al. 1992, Linzey 1998), North Carolina (Engels 1942, Johnston 1967, Lee et al. 1982, Clark et al. 1985, Webster et al. 1985, Webster 1988, Lunk 1993), South Carolina (Jenkins and Provost 1964, Wood and Odom 1965, Golley 1966, Sanders 1978, Schacher and Pelton 1979, Webster et al. 1985, Cothran et al. 1991), and Georgia (Harper 1927, Golley 1962, Pelton 1969, Pelton and Jenkins 1971, Pelton and Provost 1972, Laerm et al. 1982). Populations occur throughout Florida (Howell 1939, Rand and Host 1942, Moore 1946, Schwartz 1952, 1956; Ivey 1959, Layne 1974), Alabama (Holliman 1963, Linzey 1970, Hill 1966, 1972; King et al. 1991), Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989, Bond et al. 2000, 2001), Tennessee (Calhoun 1941, Goodpaster and Hoffmeister 1952, Howell and Conaway 1952, Linzey and Linzey 1971, Kennedy 1991), Kentucky (Bruna 1952, Barbour and Davis 1974, Fassler 1974, Giuliano 1990, Giuliano et al. 1993, 1994; McGehee-Marsh et al. 1993, Sole 1995, Giuliano and Elliott 1997), Louisiana (Lowery 1974, Hamilton et al. 1987, Mullin and Williams 1987), Arkansas (Sealander and Heidt 1990), eastern Texas (Taylor and Lay 1944, Schmidly 1983, Jones and Jones 1992, Davis and Schmidly 1994), and eastern Oklahoma (Caire et al. 1989). The distribution of the cottontail in the South is depicted in Figure 2.

ABUNDANCE STATUS

The eastern cottontail is abundant in suitable habitat throughout its range. Estimates of local abundance range from 0.7–20/ha (Jenkins and Provost 1964, Trent and Rongstad 1974, Bittner and Chapman 1981, Chapman et al. 1982). The species undergoes both long-term and short-term population fluctuations on both local and regional scales (Bailey 1968, Sadler 1981, Chapman et al. 1982, Giuliano et al. 1993).

PRIMARY HABITATS

The eastern cottontail occurs in a diversity of habitats and vegetational communities. Habitat use varies by season. Throughout much of the southern United States, it is associated with upland cover types along wooded margins adjacent to open habitats such as oldfields. It is particularly common in agricultural areas where weedy grasses, briars, and low-growing woody perennials are abundant (Swihart and Yahner 1982a,b; Giuliano and Elliott 1997, Mankin and Warner 1999). Preferred habitats include cover for

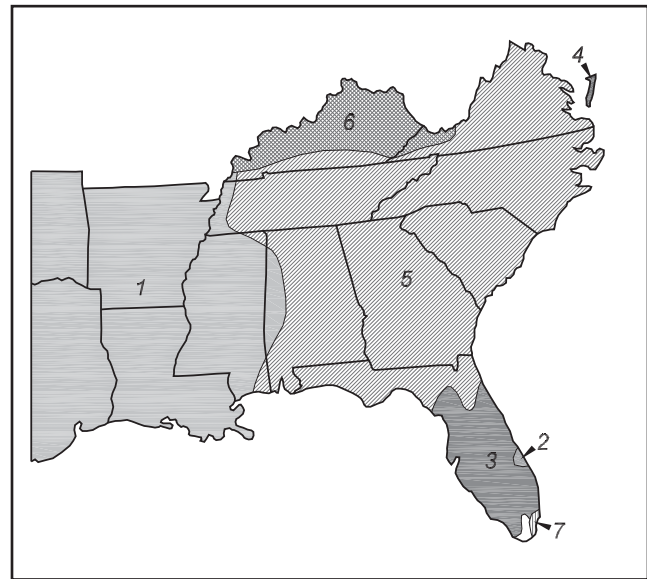


Figure 2. Distribution of *Sylvilagus floridanus* in the South: (1) *S. f. alacer*; (2) *S. f. ammophilus*; (3) *S. f. floridanus*; (4) *S. f. hitchensi*; (5) *S. f. mallurus*; (6) *S. f. mearnsi*; (7) *S. f. paulsoni*.

hiding, nesting, and resting. The eastern cottontail is not usually associated with marshes and swampy bottomlands that are inhabited by the marsh rabbit (*S. palustris*) and swamp rabbit (*S. aquaticus*).

REPRODUCTION

The onset of breeding occurs early in lower latitudes and elevations (Barkalow 1962, Conaway et al. 1974, Chapman et al. 1977). Breeding onset may vary between populations and within the same population from year to year (Conaway and Wight 1962, Hill 1966, 1972; Chapman et al. 1977). Reproductive activity apparently begins in response to temperature or availability of succulent vegetation. Eastern cottontails are synchronous breeders that may breed throughout the year in southern Texas (Bothma and Teer 1977), February to mid-October in Louisiana (Lowery 1974) and Georgia (Pelton and Provost 1972), February–March to August–September in Arkansas (Sealander and Heidt 1990), and March–September in Kentucky (Barbour and Davis 1974, Giuliano et al. 1993). From 5–8 litters may be produced per breeding season (Chapman et al. 1982). Gestation is 25–35 days (Chapman et al. 1982). During gestation, the female scoops out a hollow in the ground, lining the hollow first with grass and fur (Beule 1940, Lord 1963). The young are deposited in the nest and concealed by a layer of dead grasses. The number of young/litter usually varies from 3–5 depending upon latitude, time of year, age, and soil fertility (Llewellyn and Handley 1947, Pelton and Jenkins 1971, Hill 1972, Bothma and Teer 1977, Chapman

et al. 1982), but litters of up to 12 young are reported (Chapman et al. 1982). The potential life span is 10 years (Lord 1963) but average longevity in the wild is 15 months (Bruna 1952). Bond et al. (2001) reported a high mortality rate for eastern cottontails during the first year.

FOOD HABITS

Feeding occurs in two distinct periods: 3 to 4 hours after sunrise and within one hour of sunset (Dalke and Sime 1941). Eastern cottontails feed on a variety of grasses and herbaceous plants in the summer. During the fall, there is a transition from herbaceous to woody vegetation and in winter, the diet consists of woody shrubs and tree species (Chapman et al. 1982, Giuliano et al. 1994). The eastern cottontail exhibits coprophagy (Hamilton 1955). They produce two types of pellets: The hard, brown pellet has few nutrients and is not eaten whereas the green pellet type that is produced in the caecum and contains vitamin B is reingested (Bailey 1969).

ASSOCIATED SPECIES

The eastern cottontail is associated with upland early successional and mixed woodland habitat species including the short-tailed shrews (*Blarina* spp.), least shrew (*Cryptotis parva*), southeastern shrew (*Sorex longirostris*), white-footed mouse (*Peromyscus leucopus*), cotton mouse (*P. gossypinus*), oldfield mouse (*P. polionotus*), Texas mouse (*P. attwateri*), hispid cotton rat (*Sigmodon hispidus*), and meadow jumping mouse (*Zapus hudsonius*). Wide-ranging predators such as the coyote (*Canis latrans*), red fox (*Vulpes vulpes*), gray fox (*Urocyon cinereoargenteus*), and bobcat (*Lynx rufus*) occupy the same habitats.

VULNERABILITY AND THREATS

The eastern cottontail is an abundant game species where there is suitable habitat. It appears secure throughout the South, although local populations in mature forest habitats in the Appalachians and in managed pine plantations in the Coastal Plain may be low. There also are concerns that some populations have declined with "clean farming" practices (Edwards et al. 1981, Mankin and Warner 1999).

MANAGEMENT SUGGESTIONS

Management centers on the provision of cover for nesting, escape from predators, and shelter from the weather. Early seral stands and young pine plantations provide excellent cover. Productive habitat can be provided by maintaining a variety of different aged stands interspersed with open fields, areas of

thick cover, and succulent green forage. Strip disking, bush hogging, and prescribed burning on a 4–5 year rotation can enhance habitat productivity and provide woody winter cover. However, the presence of altricial young dictates that active management occurs outside of the nesting season. In winter, the planting of oats (*Avena sativa*) and winter wheat (*Triticum aestivum*), and clover next to ground cover is beneficial.

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Appalachian Cottontail

Sylvilagus obscurus (Chapman, Cramer, Dippenaar, and Robinson, 1992)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

In a review of the systematics and biogeography of the New England cottontail (*Sylvilagus transitionalis*), Chapman et al. (1992) found two morphologically and karyologically distinct forms previously referred to as *S. transitionalis*. The New England cottontail is restricted to New England from the Hudson River east. The new monotypic species, the Appalachian cottontail (*S. obscurus*), is distributed from the Hudson River west and south throughout the Appalachian Highlands to Alabama. Some authors prefer to use Allegheny cottontail as the common name for Appalachian cottontail. Literature for the Appalachian cottontail is included in reviews of the New England species by Chapman (1975) and Chapman et al. (1982).

DISTINGUISHING CHARACTERISTICS

The Appalachian cottontail is a medium-sized rabbit. Measurements are: total length, 386–430 mm; tail, 22–65 mm; hind foot, 87–97 mm; ear, 54–63 mm; weight, 0.7–1.0 kg. Dorsal parts of the head and body of the rabbit are pinkish to ochraceous buff overlain by a black wash resulting in a finely streaked effect. The sides are a slightly paler buff that is less heavily washed with black but often grayish. The venter is whitish to light gray. A black spot is usually present between the ears, but the Appalachian cottontail never has a white spot on forehead. The ears are short and rounded, and usually have distinct black edging along anterior outer borders. The nape of the neck is rust-colored. The postorbital processes of the cottontail are thin and taper to a slender point that slightly or not at all touches the skull at their tips. The sutures between the frontal and nasal bones are jagged and irregular in outline. The dental formula is: I 2/1, C 0/0, P 3/2, M 3/3 = 28 (Figure 1). It is often difficult to distinguish between this species and the eastern cottontail (*S. floridanus*). The pelage of the eastern cottontail is similar, but the dorsum of the eastern species has less of a pinkish cast and fewer black streaks. The ears of the eastern species are longer and more pointed, and the borders are pale buff to white rather than black. Most eastern cottontails have a white spot or at least a few white hairs on forehead and lack a black spot between the ears. The postorbital process of the eastern cottontail is broad and flat, and often has broad contact with skull. In addition, the naso-frontal sutures are smooth in outline.

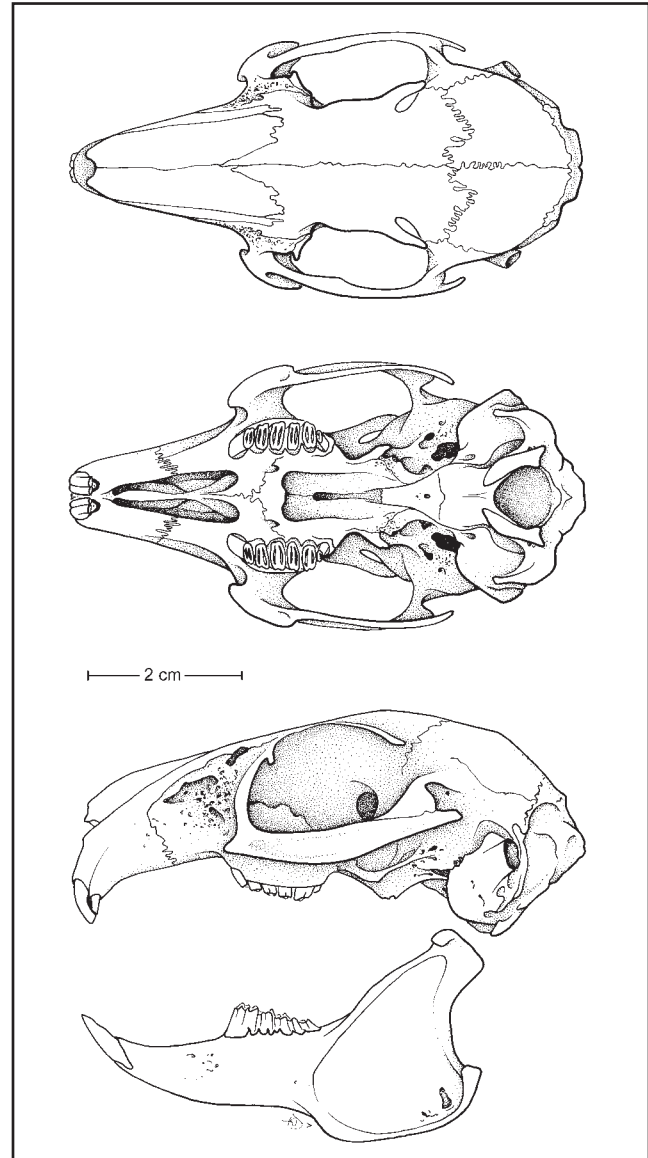


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sylvilagus obscurus* from Carter County, Tennessee (USNM 332330, male).

CONSERVATION STATUS

The Appalachian cottontail has a global rank of Apparently Secure (NatureServe 2007). The species is considered Apparently Secure in Virginia and Vulnerable in North Carolina, South Carolina, and Tennessee. It is Critically Imperiled in Alabama and Georgia. Mount (1986), Betsill (1987), Handley (1991), and Laerm (1993) review the status of the species in the region.

DISTRIBUTION

Chapman et al. (1992) suggest that the Appalachian cottontail is a cold-adapted, or boreal northern forest specialist restricted in the southern Appalachian Highlands to a series of isolated, relictual refugia. These refugia are associated with balds and higher elevations extending down the Appalachian Highlands of Virginia (Llewellyn and Handley 1945, Handley and Patton 1947, Blymer 1976, Handley and Gordon 1979, Fies and Coggin 1985, Handley 1991, Linzey 1998), Kentucky (Barbour 1951, Barbour and Davis 1974), Tennessee (Kellogg 1939, Conaway and Howell 1953, Linzey and Linzey 1971, Kennedy and Harvey 1980, Linzey 1995), North Carolina (Odom 1949, Johnston 1967, Betsill 1987, Linzey 1995), South Carolina (Russell et al. 1999), Georgia (Golley 1962, Laerm 1981, 1993) and Alabama (Howell 1921, Holliman 1963, Mount 1986). The species may be distributed more widely than previously believed in the Blue Ridge Province of Virginia (Fies and Coggin 1985, Handley 1991, Linzey 1998), upper Piedmont and Blue Ridge of South Carolina (M. Strayer, Personal Communication), and the Cumberland Plateau of Kentucky (Sole 1999) and Tennessee (M. Kennedy, Personal Communication). Distributional records are listed in Chapman et al. (1992). The distribution of the Appalachian cottontail is depicted in Figure 2.

ABUNDANCE STATUS

Based on collection records, Chapman et al. (1992) considered the Appalachian cottontail as rare over most of its range. Although little quantitative information is available, the species may be more abundant than present documentation suggests. Fies and Coggin (1985) note that of 210 cottontails collected in 19 counties in western Virginia, 70 were Appalachian cottontails. In 24 counties of Kentucky, 39 of 363 rabbits obtained from hunters were identified as Appalachian cottontails (J. Sole, The Nature Conservancy personal communication). In extreme northwestern South Carolina, the species is considered common in suitable habitat (M. Bunch, South Carolina Wildlife Resources Commission, personal communication).

PRIMARY HABITATS

The habitat of the Appalachian cottontail is described by Llewellyn and Handley (1945), Barbour (1951), Eabry (1968), Chapman and Paradiso (1972), Chapman and Morgan (1973), Blymer (1976), Chapman et al. (1977), Handley and Gordon (1979), Chapman and Stauffer (1981), Chapman et al. (1992), Laerm (1993) and Russell et al. (1999). The species is associated with dense cover, especially scrubby vegetation and ericaceous shrubs such as mountain laurel (*Kalmia*

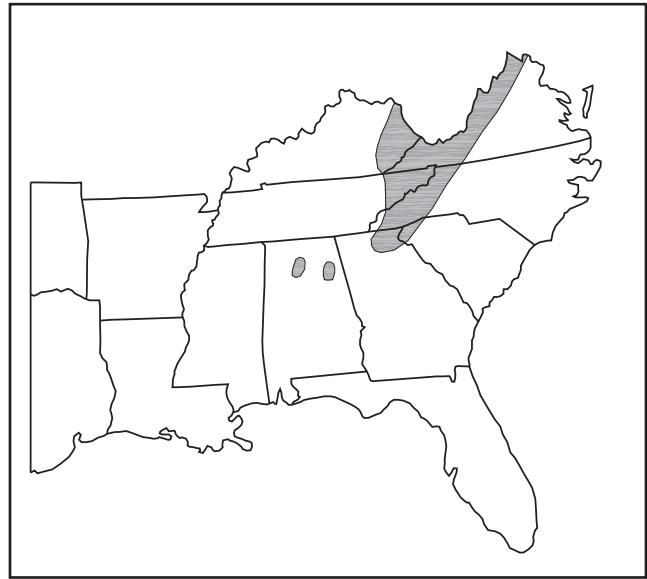


Figure 2. Distribution of *Sylvilagus obscurus* in the South.

latifolia), rhododendron (*Rhododendron* spp.), or blueberry (*Vaccinium* spp.), that are typical in forested habitats of the Appalachian Mountains. Populations occur as low as 250 m on the Cumberland Plateau of Kentucky (Sole 1999). The Appalachian cottontail often is associated with overgrown farmsteads and heath-conifer habitat. In Virginia, the Appalachian cottontail is reported from 6–7 year old clearcuts at high elevations (Handley 1991, Linzey 1998). In the central and southern Appalachians, it is most abundant in 5–10 year old clearcuts and older scrubby vegetation that is associated with high elevation stream bottoms. Throughout the central and southern Appalachians, the cottontail is found in association with red spruce-fir (*Picea rubens-Abies* spp.), northern hardwood, cove hardwood, white pine-hemlock (*Pinus strobus-Tsuga* spp.), oak-hickory (*Quercus-Carya*), and mixed pine-hardwood cover types (Chapman et al. 1992).

REPRODUCTION

The reproductive biology of the Appalachian cottontail was studied extensively in western Maryland (Chapman et al. 1977). Appalachian cottontails are synchronous breeders. The duration of the breeding season is from early March to early September with a peak of activity between March and July. Mean litter size is 3.5. Individuals are sexually active in the year following their birth. Chapman et al. (1977) reported that the species has a longer breeding season and smaller litters than the eastern cottontail. Juveniles also engage in significantly more breeding.

FOOD HABITS

The diet of Appalachian cottontail is probably similar to that of New England and eastern species (Dalke and Sime 1941). However, Nottage (1972) suggests that the eastern cottontail is better adapted to a wider variety of foods than either the Appalachian or New England rabbits. During the summer, the diet includes grasses and clover, twigs, buds, seeds, and fruits (Dalke 1937). Autumn and winter foods include shrubby and herbaceous plants and conifer needles (Spencer and Chapman 1986).

ASSOCIATED SPECIES

The Appalachian cottontail occupies the same habitats as other montane, early successional species including the masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), deer mouse (*Peromyscus maniculatus*), meadow vole (*Microtus pennsylvanicus*), and southern red-backed vole (*Clethrionomys gapperi*). In many areas, the eastern cottontail occurs sympatrically with the Appalachian cottontail (Chapman and Morgan 1973). Potential predators that occur within the range of the cottontail include the bobcat (*Lynx rufus*), gray fox (*Urocyon cinereoargenteus*), red fox (*Vulpes vulpes*), coyote (*Canis latrans*), fisher (*Martes pennanti*), weasel (*Mustela* spp.), great horned owl (*Bubo virginianus*), barred owl (*Strix varia*), and red-tailed hawk (*Buteo jamaicensis*).

VULNERABILITY AND THREATS

There are two major threats to the Appalachian-New England cottontail complex throughout their range. First, the decline in early seral conditions in eastern forests (Trani et al. 2001) has negatively influenced both the New England (Litvaitis and Villafuerte 1996) and Appalachian species (M. Fies, Virginia Game and Inland Fisheries, personal communication). Second, the complete clearing of land for agricultural use has facilitated competitive exclusion by the eastern cottontail. This species has made extensive incursions into the range of the Appalachian cottontail. However, Fies and Coggin (1985) hypothesize that in Virginia, the Appalachian cottontail may be able to withstand the encroachment of eastern cottontails within the confines of isolated high elevation balds and forests.

MANAGEMENT SUGGESTIONS

Management activity that provides thick escape cover is the key to enhancing populations of the Appalachian cottontail (M. Fies, Virginia Game and Inland Fisheries, personal communication). This includes prescribed burning, thinning, or clearcutting

that create habitat edge, reduce canopy closure, and enhance dense understory cover (e.g., blueberry, mountain laurel, and rhododendron thickets). The maintenance of large blocks of forest with a thick understory provide the cottontail with areas of refuge, which may provide this species with a competitive edge. Litvaitis and Villafuerte (1996) suggest the creation of a network of patches of early successional habitat in remote mountain areas to provide optimum habitat for the cottontail. To facilitate exchange of individuals between populations, the patches should be placed in close proximity (<500m) to one another.

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Marsh Rabbit

Sylvilagus palustris (Bachman, 1837)

Brian R. Chapman and Margaret K. Trani

CONTENT AND TAXONOMIC COMMENTS

Three subspecies of marsh rabbit (*Sylvilagus palustris*) are recognized: *S. p. palustris*, *S. p. paludicola*, and *S. p. hefneri* (Lazell 1984). Literature on the marsh rabbit is reviewed by Chapman and Willner (1981) and Chapman et al. (1982).

DISTINGUISHING CHARACTERISTICS

The marsh rabbit is a moderate-sized cottontail. Measurements are: total length, 400–450 mm; tail, 30–40 mm; hind foot, 85–100 mm; ear, 45–55 mm; weight, 1.0–2.2 kg. The dorsal parts of head and body may vary from chestnut brown to rusty red, but all specimens have a dark cinnamon nape. The middle of abdomen is white and the remainder of belly is pale brown to buff. The feet are slender and reddish to buffy. The ventral surface of the tail, unlike other *Sylvilagus*, is dingy gray. The basilar length of the skull of *S. palustris* is usually less than 63 mm. The anterior portion of the supraorbital process is typically absent and the posterior portion of the supraorbital process is fused to the skull. The dental formula is: I 2/1, C 0/0, P 3/2, M 3/3 = 28 (Figure 1). Marsh rabbits may be confused with swamp rabbits (*S. aquaticus*) and eastern cottontails (*S. floridanus*). Marsh rabbits usually are distinguishable from both species due to a larger body size and a gray tail. Marsh rabbits also possess a black spot between the ears, which is absent in the eastern cottontail.

CONSERVATION STATUS

The marsh rabbit has a global rank of Secure (NatureServe 2007). The species is considered Secure in Georgia and North Carolina, and Vulnerable in Alabama and Virginia. It is unranked in Florida and South Carolina. The U. S. Fish and Wildlife Service (U. S. Department of the Interior 1993, 2007) lists the Lower Keys marsh rabbit (*S. p. hefneri*) as Endangered.

DISTRIBUTION

The marsh rabbit is restricted to the Coastal Plain of the southeastern Atlantic and Gulf Coasts (Figure 2). Its northern limit is in extreme southeastern Virginia (Payne 1975, Hall 1981, Webster et al. 1985, Fishel and McCravy 1988, Padgett 1989, Fies 1991, Linzey 1998), but Handley (1991) noted no recent sightings

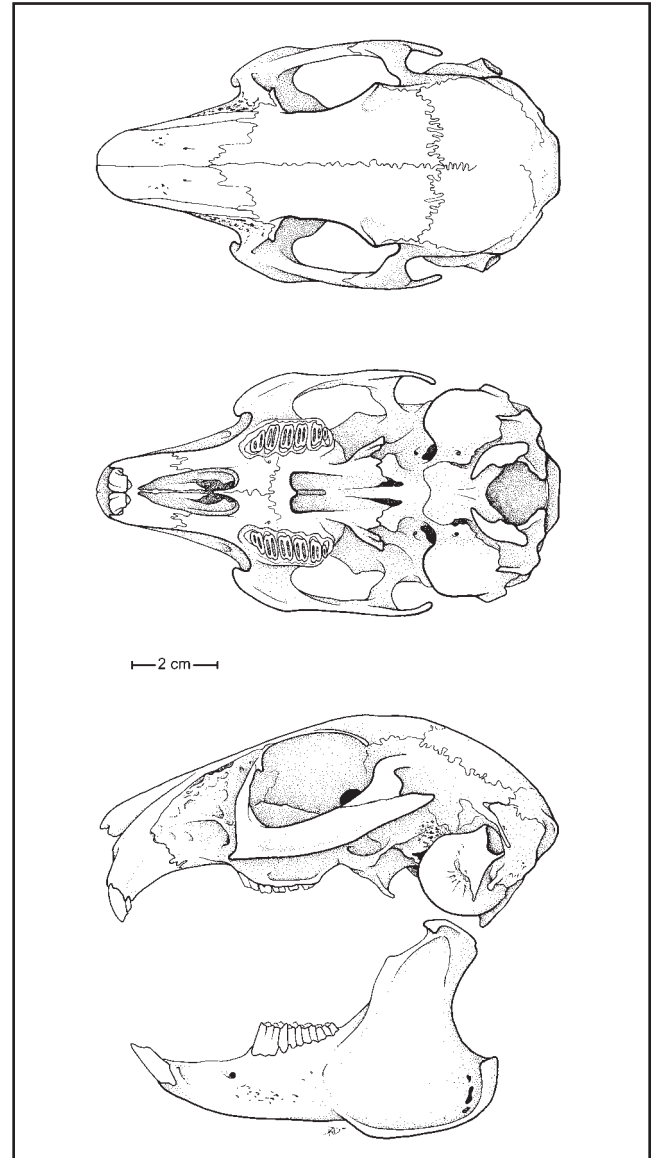


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sylvilagus palustris* from Palm Beach County, Florida (USNM 349493, male).

from the Eastern Shore. It is distributed throughout the Coastal Plain and barrier islands of North Carolina (Lee et al. 1982, Webster 1988, Markham and Webster 1993), South Carolina (Golley 1966, Sanders 1978, Schacher and Pelton 1979, Cothran et al. 1991), and Georgia (Harper 1927, Tompkins 1935, 1955; Lowe 1958, Golley 1962, Caldwell 1966, Neuhauser and

Baker 1974, Wharton et al. 1981, Laerm et al. 1982). It occurs throughout Florida (Blair 1936, Hamilton 1941, Rand and Host 1942, Moore 1946, Pournelle 1950, Schwartz 1952, Layne 1974, Holler and Conaway 1979, Lazell 1984, 1989; Wolfe 1992, Fors and Humphrey 1996, 1997) and west to Mobile Bay, Alabama (Holliman 1963, Linzey 1970, Lazell 1984, French 1986). *Sylvilagus p. hefneri* is restricted to the Lower Keys of Florida from Big Pine Key to Key West (Lazell 1984, 1989; Howe 1988, Wolfe 1992, Fors and Humphrey 1996, 1997; Cox and Kautz 2000). *Sylvilagus p. paludicola* occurs throughout the peninsular and southern Florida Panhandle (Lazell 1984, Wolfe 1992).

ABUNDANCE STATUS

Little is known of the population density in the South, but the marsh rabbit is thought to be quite abundant in appropriate habitat (Whitaker and Hamilton 1998). It is considered rare in southeastern Virginia (Fies 1991) and Alabama (French 1986). The current population of the endangered Lower Keys marsh rabbit is 150–400 individuals (Cox and Kautz 2000), which is considered to be in decline (U. S. Department of Interior 1998).

PRIMARY HABITATS

The marsh rabbit inhabits freshwater and brackish marsh habitat (Tomkins 1935, Blair 1936, Chapman and Willner 1981, Chapman et al. 1982). The species is often found in raised hummocks in marshes of southern magnolia (*Magnolia grandiflora*), water tupelo (*Nyssa aquatica*), sweetgum (*Liquidambar styraciflua*), and blackberry (*Rubus* spp.) shrubs. It occurs in freshwater swamps (Harper 1927), floodplains, tributaries, and estuaries of rivers and streams from coastal areas to the upper limits of the Coastal Plain (Chapman et al. 1982). In coastal areas and islands, the marsh rabbit may inhabit sand dunes, salt and freshwater pond margins, road edges, and open fields adjacent to marshes and dunes (Schacher and Pelton 1979, Fies 1991). The species also is associated with sugarcane (*Saccharum officinarum*) plantations in southern Florida (Holler and Conaway 1979), where it is considered a pest (Whitaker and Hamilton 1998). The endangered Lower Keys subspecies occupies transitional salt marsh habitat dominated by thick grasses and shrubs (Cox and Kautz 2000). Fors et al. (1996) estimate that 320 ha of habitat exist in the Lower Keys. Two locations have substantial areas of potential habitat: Key Deer National Wildlife Refuge and Key West Naval Air Station. The latter is estimated to contain approximately half the remaining population and one third of the remaining habitat.

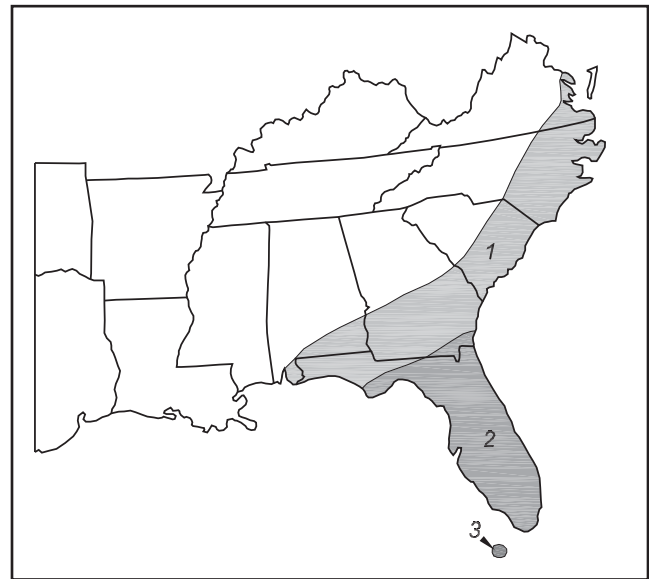


Figure 2. Distribution of *Sylvilagus palustris* in the South: (1) *S. p. palustris*; (2) *S. p. paludicola*; (3) *S. p. hefneri*.

REPRODUCTION

Marsh rabbits breed year-round (Holler and Conaway 1979) and produce approximately 6 litters/year (Chapman et al. 1982). The gestation period is estimated between 30–37 days (Holler and Conaway 1979). The female constructs a nest that is about 20 cm deep and lined with soft grass and fur (Tomkins 1935). The litter size may vary from 3–5 (Harper 1927). The Lower Keys marsh rabbit is sexually mature at 9 months of age. Similar to other subspecies of marsh rabbit, the species is polygamous and also breeds year around (U. S. Department of Interior 1998). There is no apparent seasonal breeding pattern, however. The highest proportion of females with litters occurs in March and September.

FOOD HABITS

Marsh rabbits feed on a variety of seeds, fruits, leaves, and twigs of numerous trees, shrubs, woody vines, and cultivated plants (Chapman and Willner 1981, Wolfe 1992, Markham and Webster 1993). In captivity, they prefer herbaceous plants such as centella (*Centella* spp.), marsh pennywort (*Hydrocotyle* spp.), cattail (*Typha* spp.), rush (*Juncus* spp.), water hyacinth (*Eichhornia crassipes*), and the bulbs of several plants (Blair 1936). The endangered marsh rabbit consumes bushy seaside oxeye (*Borrchia frutescens*), an important food that is common in the saltmarsh areas. Other foods include dropseed grass (*Sporobolus virginicus*), glasswort (*Salicornia virginica*), and cordgrass (*Spartina spartinae*; U. S. Department of Interior 1998).

ASSOCIATED SPECIES

The geographical range of the marsh rabbit overlaps that of the swamp rabbit. The marsh rabbit also associates with other wetland forest and marshland mammals such as the southern short-tailed shrew (*Blarina carolinensis*), marsh rice rat (*Oryzomys palustris*), and cotton mouse (*Peromyscus gossypinus*). The Lower Keys marsh rabbit occupies habitat that overlaps with the endangered Key deer (*Odocoileus virginianus clavium*) and the silver rice rat (*Oryzomys argentatus*). Predators of the marsh rabbit include the great horned owl (*Bubo virginianus*), barred owl (*Strix varia*), northern harrier (*Circus cyaneus*), red-tailed hawk (*Buteo borealis*), bobcat (*Lynx rufus*), red fox (*Vulpes vulpes*), gray fox (*Urocyon cinereoargenteus*), weasel (*Mustela* spp.), mink (*M. vison*), and American alligator (*Alligator mississippiensis*). The cottonmouth (*Agkistrodon piscivorus*) and eastern diamondback rattlesnake (*Crotalus adamanteus*) are known to prey on the young (Whitaker and Hamilton 1998).

VULNERABILITY AND THREATS

The species is considered rare at the limit of its range in Alabama (French 1986) and Virginia (Handley 1991). Whitaker and Hamilton (1998) consider habitat fragmentation to be the major environmental problem for this species, since it results in the loss of available suitable habitat and may adversely affect dispersal and genetic diversity. The species prefers relatively undisturbed marshes, which are rapidly disappearing throughout the South as human populations expand (Trani 2002). The loss of suitable habitat stems from land conversion and degradation through dredge and fill operations.

The endangered subspecies in the Lower Keys is vulnerable to habitat destruction associated with residential and commercial construction activities, contaminants, vehicular traffic, feral cat (*Felis catus*) predation, and red imported fire ants (*Solenopsis invicta*; U. S. Department of Interior 1998). Land development in transitional wetlands is regulated by federal, state, and local governments; permits that result in habitat losses continue to be issued (Cox and Kautz 2000). One-third of the habitat of the species is held in private ownership.

MANAGEMENT SUGGESTIONS

Maintenance of high quality escape cover and abundant food sources are essential for viable populations of the marsh rabbit. Forest management that results in dense secondary growth near a water source is beneficial, whereas activities that reduce cover near water are detrimental. In those states where known

populations are classified as Vulnerable (Alabama and Virginia), habitat should be protected and maintained. Four major recovery objectives have been identified for the Lower Keys marsh rabbit. These include the acquisition of suitable habitat with an upland buffer, control of predation by feral and domestic cats, monitoring of existing rabbit populations, and reintroduction of rabbits into unoccupied suitable habitat (U. S. Department of Interior 1998).

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American Beaver

Castor canadensis (Kuhl, 1820)

Michael T. Mengak and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Twenty-four subspecies are recognized, four of which occur in the South: *C. c. carolinensis*, *C. c. canadensis*, *C. c. missouriensis*, and *C. c. texensis* (Jenkins and Busher 1979, Hall 1981, Baker and Hill 2003). The North American beaver is genetically, morphologically, and behaviorally distinct from the Eurasian beaver (*C. fiber*; Lavrov and Orlov 1973, Sieber et al. 1999). Hall (1981) identified *C. c. carolinensis* as the most widespread subspecies in the region. However, Whitaker and Hamilton (1998) found no evidence of primary isolation mechanisms and concluded that there was no need to recognize subspecies. Further, extirpation of regional populations due to historic over harvest and reintroduction may have altered gene pools such that subspecies distinctions are meaningless. An extensive array of literature is available (Yeager and Hay 1955, Hodgdon and Larson 1980), much of which is reviewed by Jenkins and Busher (1979), Novak (1987), and Baker and Hill (2003).

DISTINGUISHING CHARACTERISTICS

The beaver is the largest rodent in North America, characterized by a heavy, compact body, large dorso-ventrally flattened heavily scaled tail, and short legs with webbed hind feet. Measurements are: total length, 1,000–1,200 mm; tail, 230–325 mm; hind foot, 150–205 mm; ear, 23–31 mm; weight, 11–31 kg. Males are slightly larger than females. Dorsal pelage consists of soft, dense underfur and long, coarse guard hairs. Color ranges from chocolate brown to blond. The cranium is large, the infraorbital canals are slit-like, the frontals are without prominent postorbital processes, and there is a distinct depression in the basioccipital. The front surface of the incisors is orange. The dental formula is I 1/1, C 0/0, P 1/1, M 3/3 = 20 (Figure 1). See keys for details.

CONSERVATION STATUS

The American beaver has a global rank of Secure (NatureServe 2007). The species is also considered Secure in those states where it occurs within the region except for Arkansas and Oklahoma, where it is Apparently Secure. It is unranked in Florida and South Carolina. The American beaver is a protected furbearer with regulated harvests in most states of the South.

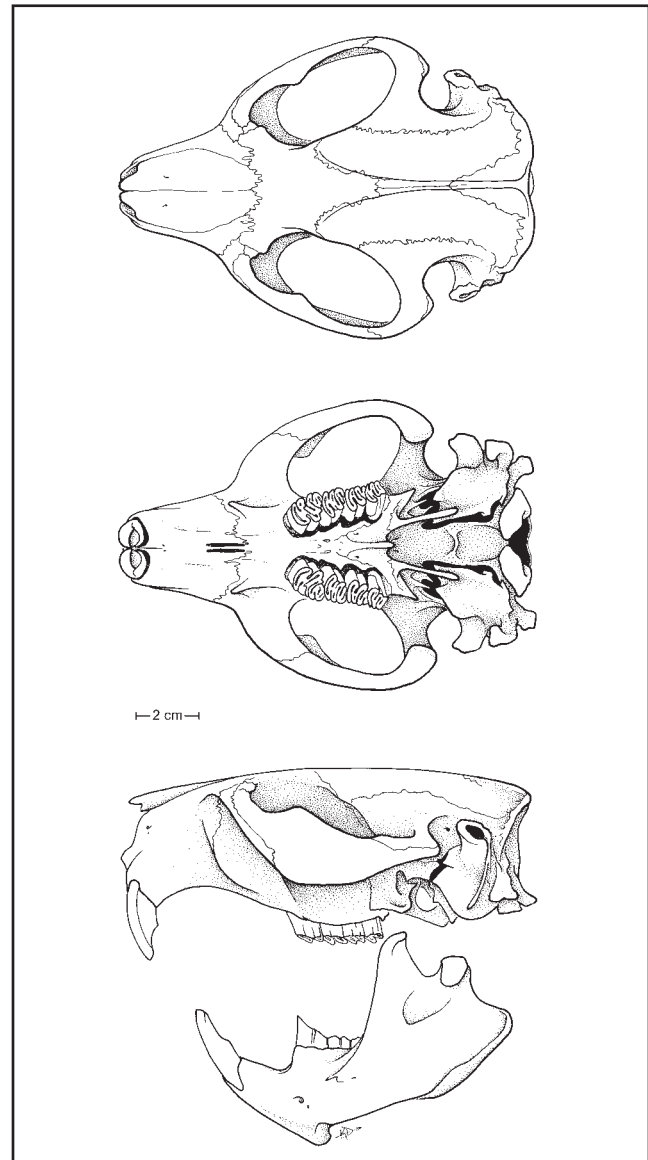


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Castor canadensis* from Cherry County, Nebraska (USNM 250191, female).

DISTRIBUTION

The beaver ranges throughout North America from Alaska east to Newfoundland and south into northern Mexico. It is absent from areas above the tree line, southwestern deserts, portions of the Midwest, and southern Florida (Larson and Gunnison 1983).

The species occurs throughout Virginia (Bailey 1946, Handley and Patton 1947, Handley 1979, 1992; Echternach and Rose 1987, Linzey 1998) and North Carolina (Lee et al. 1982, Clark et al. 1985, Woodward et al. 1985; Figure 2). Populations have been expanding from sources in the Savannah and Pee Dee rivers in South Carolina (Golley 1966, Woodward et al. 1976, Shipes et al. 1979, Cothran et al. 1991, SCDNR 2005) and Georgia (Golley 1962, Wharton et al. 1981, Laerm et al. 1982). Populations in Florida occur as far south as Ocala and are present on the Osceola and Apalachicola National Forest (Gore and Baker 1989, Robson 1989, J. Gore, Florida Fish and Wildlife Conservation Commission, personal communication). The species occurs throughout Alabama (Holliman 1963, Beshears 1967, Pullen 1975, Johnson and Aldred 1984), Mississippi (Wolfe 1971, Kennedy et al. 1974, Arner and Dubose 1978, 1982; Brown 1981, Wigley et al. 1983, Johnson and Aldred 1984, Roberts and Arner 1984, Jones and Carter 1989), and all but coastal Louisiana (Chabreck 1958, Smith 1964, Lowery 1974, Johnson and Aldred 1984, Elsey and Kinler 1996). Beaver occur throughout Tennessee (Goodpaster and Hoffmeister 1952, Schultz 1957, Linzey and Linzey 1971, Smith et al. 1974, Kennedy 1991, Houston et al. 1995), Kentucky (Barbour and Davis 1974, Fassler 1974), and west throughout Arkansas (Heidt et al. 1985, Sealander and Heidt 1990) into eastern Texas (Schmidly 1983, Davis and Schmidly 1994) and Oklahoma (Glass 1960, Reynolds 1977, Caire et al. 1989).

ABUNDANCE STATUS

Beaver populations throughout much of the South were decimated by over harvest before the turn of the 20th century. Largely as a result of introductions, propagation programs, and range expansions from these sources, the beaver has once again become a common mammal in the South. Novak (1987) indicates that density is dependent upon a number of factors including habitat (associated vegetative community, water quality, and food), mortality (trapping pressure, predation, and disease), and behavior (territorial and intra-group relations). Population densities are usually expressed in family groups per unit area or per length of watercourse (Hill 1982). Mean colony size varies between 5 and 8 individuals, and the density of groups normally ranges 0.4–1.0/km² up to as high as 3.9/km² (Busher et al. 1983, Novak 1987, Baker and Hill 2003).

In Tennessee, Houston (1998) removed 169 resident beaver in a 7-month period and an additional 153 immigrants over the next 40 months from a 1,619 ha area. Food cache size is not a reliable estimator of colony size (Osmundson and Buskirk 1993) and it is unlikely that southern beaver rely on stored food for

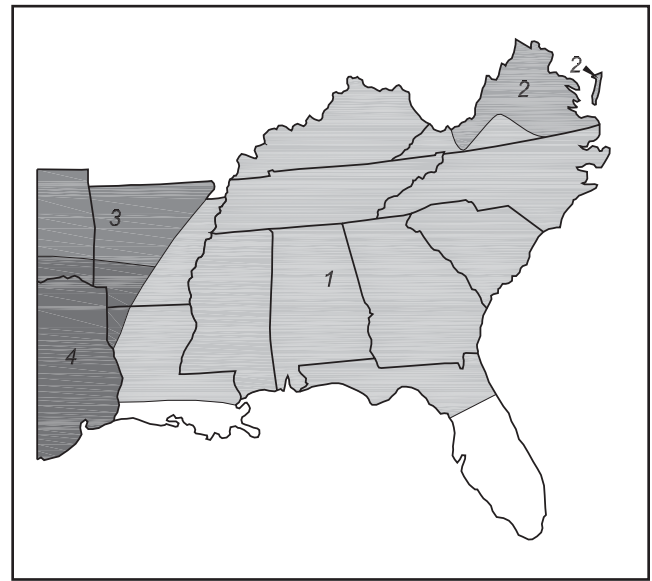


Figure 2. Distribution of *Castor canadensis* in the South: (1) *C. c. carolinensis*; (2) *C. c. canadensis*; (3) *C. c. missouriensis*; (4) *C. c. texensis*.

the winter, unlike their northern counterparts where ice is a significant habitat factor.

PRIMARY HABITATS

The American beaver is associated with water. Although they can survive in areas with poor food supply, they cannot survive in areas where water supply fluctuates dramatically seasonally. Fast moving, rocky streams or rivers, those subject to periodic flooding, lakes with rocky shorelines, or lakes with significant wave action are avoided. Optimal beaver habitat includes natural and artificial ponds and small lakes with muddy bottoms and slow moving meandering streams (Novak 1987). Habitat models predicting suitable beaver habitat incorporate features such as water supply, gradient, valley (i.e., drainage) width, and food supply (Allen 1982, Novak 1987). Beaver construct elaborate dome-shaped mud and wood “lodges” in open waters, frequently impounded by dams. Lodges typically have two or more underwater entrances. Internal chambers are constructed a few inches above water level. However, in areas along larger streams and lakes, beaver may construct dens and nests in banks.

REPRODUCTION

Beaver are monogamous and produce a single litter/year. In the southern portion of its range, beaver breed between October and March (Moore and Martin 1949, Wilkinson 1962, Woodward 1977, Wigley et al. 1983, Lizotte 1994). Gestation is 105–107 days (Hill 1982). Mean litter size in the South ranges from 2.1–2.7

(Wilkinson 1962, Woodward 1977, Wigley et al. 1983). Weaning occurs between 6–8 weeks. Females may breed as early as 21–24 months; males are reproductively active by 21–24 months. Longevity averages 6–10 years up to a maximum of 20 years. See Baker and Hill (2003) and Novak (1987) for extensive reviews.

FOOD HABITS

The beaver is a generalist herbivore (Novak 1987), feeding on a variety of woody and herbaceous species. The number of plant species fed upon varies inversely with latitude. Roberts and Arner (1984) report 42 species of trees, 36 genera of herbaceous plants, 4 woody vines, and many species of grass utilized as food. Regional food habits are discussed by Phares (1950), Thigpen (1950), Chabreck (1958), Wilkinson (1962), Woodward (1977), Shipes et al. (1979), Roberts (1981), Arner and Bullock (1982), Roberts and Arner (1984), Echternach and Rose (1987), and Tumilson and Karnes (1987).

ASSOCIATED SPECIES

Beaver activity can alter forest landscapes and habitat for a variety of species. In the South Carolina Piedmont, amphibian richness, diversity and evenness were higher in unimpounded streams than in beaver ponds (Metts et al. 2001). However, for reptiles, those same three ecological parameters were significantly higher in beaver ponds than in free-flowing streams (Metts et al. 2001). Edwards and Otis (1999) identified 77 species of birds associated with beaver ponds in the upper Piedmont of South Carolina. Bird use varied seasonally; however, beaver ponds provided important wetland habitat throughout the year. Waterfowl also use beaver ponds seasonally. Wood ducks (*Aix sponsa*) commonly use beaver ponds for brood rearing and nesting especially in snags or artificial boxes within the pond.

Numerous mammals benefit from the activities of beaver such as the marsh rice rat (*Oryzomys palustris*), common muskrat (*Ondatra zibethicus*), northern river otter (*Lontra canadensis*), small mammals (Edwards 1983), mink (*Mustela vison*), white-tailed deer (*Odocoileus virginianus*), and swamp rabbit (*Sylvilagus aquaticus*). Beaver impoundments create habitat for muskrats and are also used by mink and otter for foraging. Small mammals, deer, and rabbits benefit from the early successional habitats created when beaver flood timber stands.

VULNERABILITY AND THREATS

Beaver perhaps have had a greater impact on the environments where they occur than any other North American animal. Impacts range from detrimental (damming culverts, flooding roads and timber, feeding damage to trees) to beneficial (creation of wetlands which provide habitat for numerous other species, slowing erosion, creation of early successional habitat). It has had a considerable economic impact, both negatively (e.g., as a pest and destructive agent) as well as positively (fur value). As a result of range expansion and furbearer regulation, it is unlikely that the beaver will become vulnerable to the threats that imperiled it in the past.

MANAGEMENT SUGGESTIONS

As a result of past conservation and restoration efforts, the American beaver is well established in the South. Management currently centers on the removal of nuisance individuals (Woodward 1983). Damage occurs to crops and agricultural fields, road culverts, timber, and residential landscapes. Hill (1976) reviewed control methods for the South. In the nearly 30 years since, numerous other studies have addressed the management of beaver damage. Non-lethal methods of control include installation of water control structures (Wood and Woodward 1992) and removal of dams.

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Baird's Pocket Gopher

Geomys breviceps (Baird, 1855)

Steven B. Castleberry and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

The taxonomy of the genus *Geomys* has undergone considerable revision in the past 25 years. Although formerly considered a subspecies of the plains pocket gopher (*Geomys bursarius*), *G. breviceps* now is recognized as a distinct species based on chromosomal, morphological, and protein allozyme differences (Honeycutt and Schmidly 1979, Bohlin and Zimmerman 1982, Heaney and Timm 1983, Dowler 1989). There are two recognized subspecies, *G. b. breviceps* and *G. b. sagittalis* (Honeycutt and Schmidly 1979, Bohlin and Zimmermann 1982), both of which occur in the South. The literature on Baird's pocket gopher is reviewed by Chase et al. (1982) and Sulentic et al. (1991).

DISTINGUISHING CHARACTERISTICS

Baird's pocket gopher has a cylindrical body that is thickest at the back of the head and tapers to the tail. The large forelimbs have well developed claws used in digging. The eyes and ears are reduced, and the tail is short and nearly hairless. The short, fine pelage is light brown to almost black dorsally, and slightly paler beneath. External, fur-lined cheek pouches are used to transport food. Measurements are: total length, 190–250 mm; tail, 54–75 mm; hind foot, 20–30 mm; weight, 200–400 g. Males are approximately 10% larger than females. The dental formula is I 1/1, C 0/0, P 1/1, M 3/3 = 20 (Figure 1). See keys for details.

Although difficult to distinguish externally, *G. breviceps* is generally smaller than Attwater's pocket gopher (*Geomys attwateri*) and the plains pocket gopher (Honeycutt and Schmidly 1979), with which it has areas of contact on the western periphery of the distribution. *Geomys b. breviceps* can be distinguished from *Geomys b. sagittalis* by its slightly larger size and melanistic pelage found in most individuals (Honeycutt and Schmidly 1979).

CONSERVATION STATUS

Baird's pocket gopher has a global rank of Secure (NatureServe 2007). However, the isolated population that constitutes the subspecies *Geomys b. breviceps* in Prairie Mer Rouge, Morehouse Parish, Louisiana is considered Vulnerable/Apparently Secure. *Geomys b. sagittalis* is classified as Apparently Secure in Texas and has no ranking in Arkansas and Oklahoma.

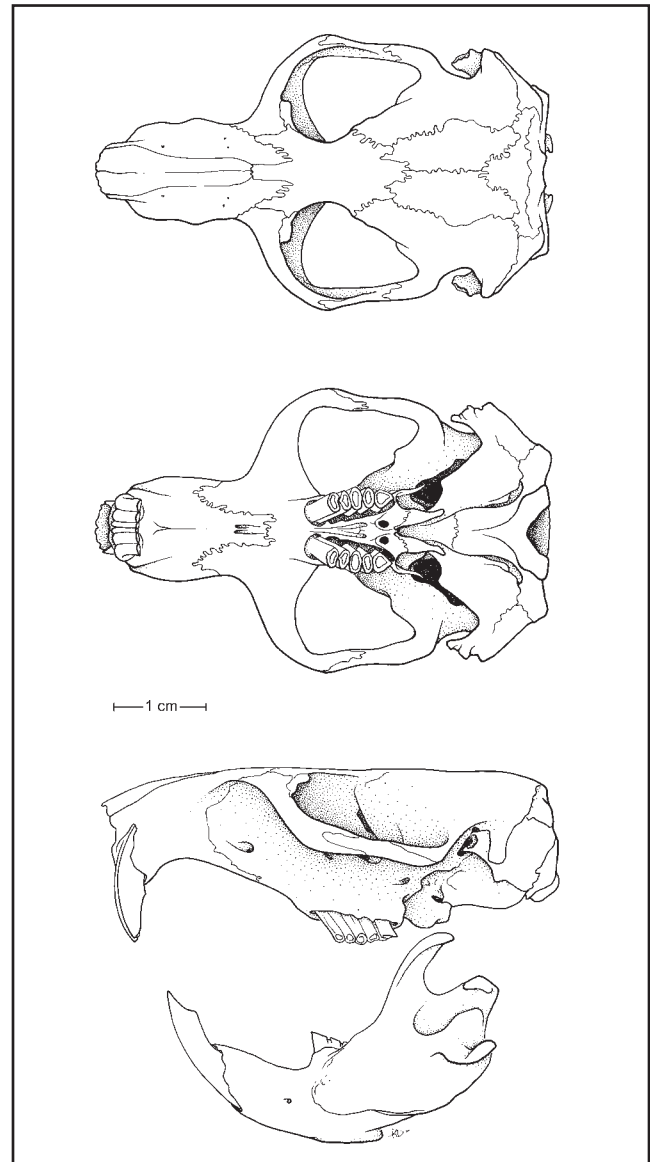


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Geomys breviceps* from Oldham County, Texas (USNM 97169, female).

DISTRIBUTION

Geomys b. sagittalis ranges throughout eastern Texas and eastern Oklahoma, eastward into western Louisiana and southwestern Arkansas (Lowery 1974, Honeycutt and Schmidly 1979, Caire et al. 1989, Sealander and Heidt 1990; Figure 2). *Geomys b. breviceps* is represented by an isolated population restricted to

Morehead Parrish, Louisiana (Honeycutt and Schmidly 1979). The two subspecies are geographically separated by the Ouachita River Basin, an area of unsuitable pocket gopher habitat that experiences annual flooding (Demastes 1994).

ABUNDANCE STATUS

Baird's pocket gopher is abundant in appropriate habitat throughout its range. Because pocket gophers are fossorial, soil characteristics are probably the most important factor influencing distribution and abundance (Davis et al. 1938, Honeycutt and Schmidly 1979). However, population density may also depend on moisture, drainage, land use, and vegetative cover (Chase et al. 1982). Available density estimates from Texas range from 0.55/ha (Davis et al. 1938) to 16.8/ha (Schmidly 1983).

PRIMARY HABITATS

Pocket gophers spend virtually their entire lives in underground burrows except occasionally to feed, disperse, or search for mates. Baird's pocket gopher typically occurs in fine sandy soils, sandy and silty loams, fine loams, silty clays, and clay loams with a topsoil depth >10 cm (Davis et al. 1938, Honeycutt and Schmidly 1979, Tucker and Schmidly 1981, Sulentic et al. 1991). They are less common in soils with high moisture content, poorly drained soils, or those that become hard and compact when dry (Davis et al. 1938). Generally, pocket gopher habitat corresponds to soils having low clay content, low soil moisture, and high sand content (Honeycutt and Schmidly 1979). However, Baird's pocket gopher has a wider tolerance of these soil characteristics than other pocket gopher species. The species is most common in tall grass vegetation, agricultural land, pastures, roadsides, and residential areas, and less so in open grassy areas within oak-hickory (*Quercus-Carya*) and mixed pine (*Pinus* spp.)-hardwood cover types (Buechner 1942, Chase et al. 1982, Sulentic et al. 1991). Although soil characteristics largely determine the distribution, pocket gophers do not always occur in areas that would otherwise appear to be appropriate habitat (Lowery 1974).

REPRODUCTION

Reproduction in Baird's pocket gopher is poorly understood (Pitts et al. 1992). Although most breeding is thought to occur from February to August, with a peak in June and July (Wood 1949), individuals have been documented breeding from November to January (Pitts et al. 1992). The average number of litters/year is estimated to be 1.7 and 2 litters may be produced in rapid succession (Wood 1949). Although

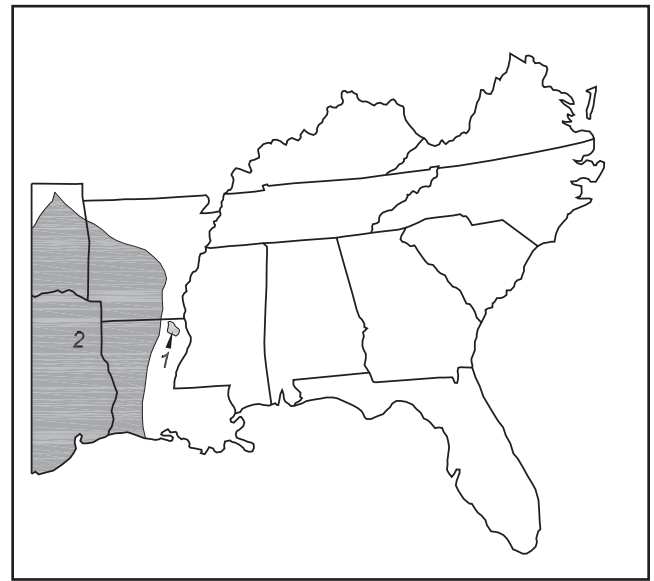


Figure 2. Distribution of *Geomys breviceps* in the South: (1) *G. b. breviceps*; (2) *G. b. sagittalis*.

the gestation period of Baird's pocket gopher is unknown, it is thought to be similar to other pocket gophers, with estimates ranging from 4 weeks (Wood 1949) to 7 weeks (Pitts et al. 1992). The young are raised in nests within large soil mounds up to 1.8 m in diameter and projecting 30–60 cm above the ground surface (English 1932). The number of young/litter ranges from 1–4 and average litter size has been estimated at 2.4 (English 1932) and 2.6 (Wood 1949). Sex ratios tend to be skewed towards females (English 1932, Wood 1949). Young probably are weaned and leave the parental nest 35–40 days after birth (Wood 1949). Sexual maturity is attained at about 90 days (Wood 1949) and longevity is approximately 5 years (Chase et al. 1982).

FOOD HABITS

The diet of Baird's pocket gopher is a function of available food in proximity to their burrow system, but typically includes a variety of roots, bulbs, underground stems, succulent stems of grasses and legumes, leaves, fruits, and grains (English 1932). Some specific foods documented from caches in Texas include Bermuda grass (*Cynodon dactylon*), nut grass (*Cyperus esculentus*), crow-poison (*Nothoscordum bivalve*), bur clover (*Medicago denticulata*), Johnson grass (*Sorghum halepense*), mullein (*Verbascum thapsus*), plantain (*Plantago occidentalis*), bull nettle roots (*Cnidioscolus texana*), pale dock (*Rumex altissimus*), vetch (*Vicia leavenworthii*), catbrier (*Smilax* spp.) tubers, blackjack oak (*Quercus marilandica*) acorns, sensitive pea (*Chamaecrista robusta*), small ragweed (*Ambrosia artemisiifolia*) and black-eyed Susan (*Rudbeckia hirta*; English 1932). Feeding occurs in lateral tunnels that

radiate out from the nest mound. Food items are transported in the cheek pouches and stored in nest mounds or in short tunnels near the nest. The caecum of pocket gophers contains bacteria that aid in digesting cellulose (Boley and Kennerly 1969).

ASSOCIATED SPECIES

Baird's pocket gopher occurs in areas inhabited by other grassland and early successional species including the southern short-tailed shrew (*Blarina carolinensis*), Elliot's short-tailed shrew (*B. hylophaga*), least shrew (*Cryptotis parva*), eastern mole (*Scalopus aquaticus*), white-footed mouse (*Peromyscus leucopus*), cotton mouse (*P. gossypinus*), Texas mouse (*P. attwateri*), and hispid cotton rat (*Sigmodon hispidus*). The range of Baird's pocket gopher contacts the ranges of the plains pocket gopher and Attwater's pocket gopher in Texas and limited hybridization occurs (Tucker and Schmidly 1981, Cothran and Zimmerman 1985). Although there are few predators because of the fossorial nature of the species, some mortality is attributed to kingsnakes (*Lampropeltis getula*), great-horned owls (*Bubo virginianus*), red-tailed hawks (*Buteo jamaicensis*), long-tailed weasels (*Mustela frenata*), and striped skunks (*Mephitis mephitis*; English 1932). Baird's pocket gopher is host to several ectoparasites, including chewing lice (*Geomydoecus* spp.) that have been studied as a model of coevolutionary relationships between hosts and parasites (Timm and Price 1980, Demastes and Hafner 1993).

VULNERABILITY AND THREATS

Baird's pocket gopher is common in suitable habitats throughout the range and there are no obvious threats to survival of the species. The isolated peripheral population in Morehouse Parrish, Louisiana (*G. b. breviceps*) has a restricted range and is vulnerable to extirpation, although no direct threats have been identified.

MANAGEMENT SUGGESTIONS

Most management considerations for Baird's pocket gopher focus on minimizing damage to agricultural crops, orchards, and golf courses. Trapping and toxicants are the most effective control methods. However, because burrowing activities can be beneficial in increasing soil fertility and aeration, rate of soil formation, and water infiltration, judicious use of control practices are recommended.

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Southeastern Pocket Gopher

Geomys pinetis (Rafinesque, 1817)

Steven B. Castleberry and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

The southeastern pocket gopher (*Geomys pinetis*) was originally described as *Mus tuza* (Pemberton and Williams 1978), but the original description was vague and the name was subsequently discarded (Harper 1952). The formerly recognized species *Geomys colonus*, *G. cumberlandius*, and *G. fontanelus* now are regarded as synonyms of *G. pinetis* based on morphological, mitochondrial DNA, protein electrophoretic, and karyotypic evidence (Avice et al. 1979, Williams and Genoways 1980, Laerm 1981a, Laerm et al. 1982). Although Pemberton and Williams (1978) list five subspecies (*G. p. austrinus*, *G. p. floridanus*, *G. p. goffi*, *G. p. mobilensis*, and *G. p. pinetis*), only two subspecies, *G. p. fontanelus* and *G. p. pinetis*, were recognized in a subsequent taxonomic revision based on karyotypic analysis (Williams and Genoways 1980). However, Humphrey (1981, 1992) suggests retention of *G. p. goffi* pending a review of diagnostic features that were not examined by Williams and Genoways (1980). Avice et al. (1979) suggests that there may be two alternate forms of *G. p. pinetis* separated by the Chattahoochee-Apalachicola river system. The literature is reviewed by Pemberton and Williams (1978).

DISTINGUISHING CHARACTERISTICS

The southeastern pocket gopher has a thick, cylindrical body, large forelimbs, and long, well-developed claws. Like other pocket gophers, *G. pinetis* has small eyes and ears, and a short naked tail. Measurements are: total length, 250–335 mm; tail, 76–96 mm; hind foot, 30–37 mm; weight, 220–420 g. Males are approximately 10% larger than females. The pelage is short and light brown to dark grayish-brown above, and lighter grayish-brown below. External fur-lined cheek pouches are present. The dental formula is I 1/1, C 0/0, P 1/1, M 3/3 = 20 (Figure 1). See keys for details.

CONSERVATION STATUS

The southeastern pocket gopher has a global rank of Secure (NatureServe 2007). It is considered Secure in Florida, Apparently Secure in Georgia, and Vulnerable in Alabama. The subspecies *Geomys p. fontanelus*, restricted to the type locality near Savannah, Chat-ham County, Georgia, and *Geomys p. goffi*, restricted to the type locality in Eau Gallie, Brevard County,

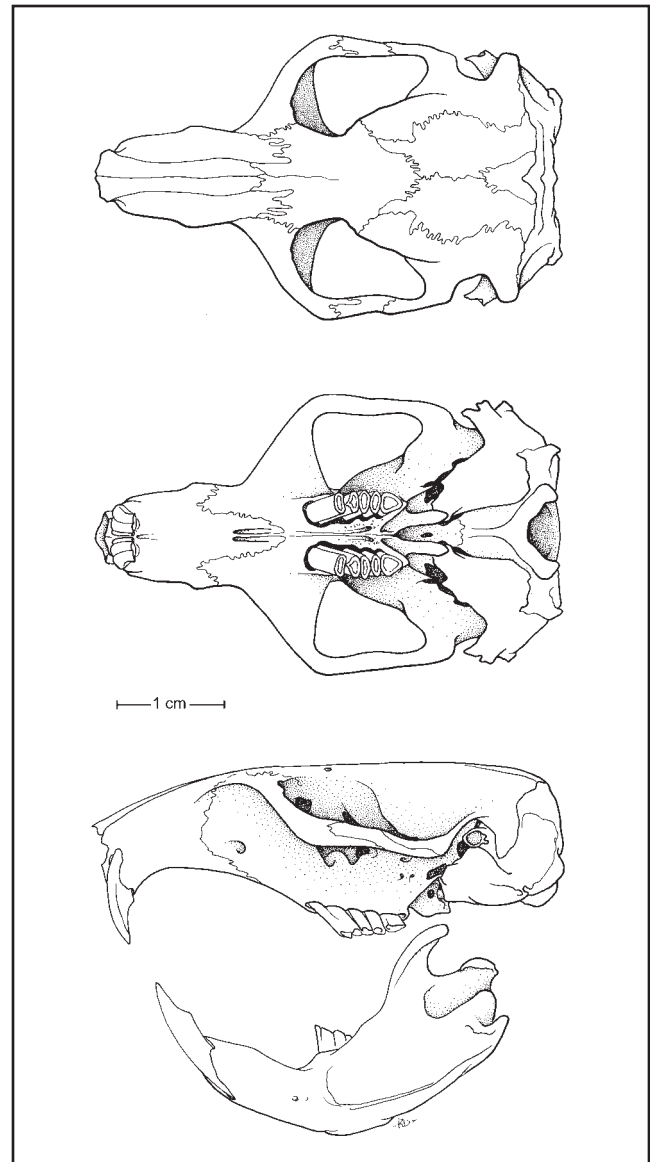


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Geomys pinetis* from Escambia County, Alabama (USNM 64949, male).

Florida, are considered extinct (Laerm 1981b, Humphrey 1981).

DISTRIBUTION

The southeastern pocket gopher is restricted to northern and central Florida (Bangs 1898, Sherman 1937,

Humphrey 1992), southern Georgia (Harper 1952, Golley 1962, Laerm 1981b), and southeastern Alabama (Linzey 1970; Figure 2). The population of southeastern pocket gophers restricted to Cumberland Island, Camden County, Georgia (formerly referred to as *G. cumberlandius*) was extant as late as 1956 (Laerm 1981a), but is now believed extinct.

ABUNDANCE STATUS

Throughout their range, southeastern pocket gopher populations occur in suitable habitat patches in a matrix of unsuitable habitat (McNab 1966, Wilkins 1987). Presence depends primarily on vegetative associations and soil characteristics (McNab 1966, Wilkins 1987). Density estimates of 0.1–0.15/ha have been obtained by counting the number of separate burrow systems (Winchester and DeLotelle 1978).

PRIMARY HABITATS

Primary southeastern pocket gopher habitat is the Coastal Plain sandhill ecosystem, which is characterized by longleaf pine (*Pinus palustris*) and turkey oak (*Quercus laevis*; Golley 1962, Wilkins 1987). It also occurs in xeric hammocks that are characterized by the presence of live oak (*Q. virginiana*) and other hardwoods, and slightly more organic soils (Wilkins 1987). Grassy roadsides, right-of-ways, and pastures also provide suitable habitat (Humphrey 1981). McNab (1966) suggests that pocket gopher habitat in Florida corresponds to soils with high rates of gas exchange and low water-holding capacity, both of which influence respiratory ability of fossorial species. Frequent fire is important in maintaining the understory vegetative associations of the sandhill ecosystem that provides primary habitat for the species (Gates and Tanner 1988), and fire encourages many herbaceous species found in the diet (Golley 1962). Pocket gophers are tolerant of vegetation changes due to variable burning regimes (Gates and Tanner 1988).

REPRODUCTION

The southeastern pocket gopher breeds year round, with peaks of breeding activity in late winter and early summer in southern Florida (Brown 1971) and spring and late summer in northern Florida (Wing 1960). Two litters are usually produced per year, corresponding to the peaks in breeding activity (Wing 1960, Brown 1971, Ewel 1971). The gestation period is unknown. The number of young per litter ranges from 1–3 and average litter size has been estimated at 1.5 (Wing 1960) and 1.7 (Brown 1971), the lowest reported for any pocket gopher species. One to 2 young are most common, with 3 being rare (Brown

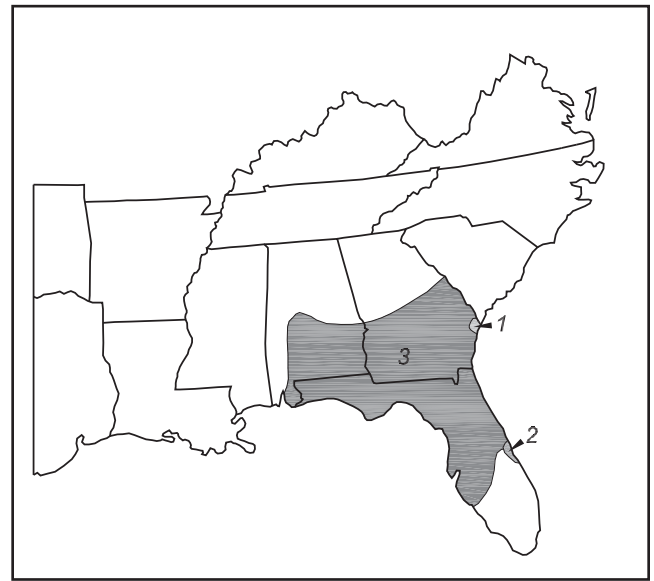


Figure 2. Distribution of *Geomys pinetis* in the South: (1) *G. p. fontanelus*; (2) *G. p. goffi*; (3) *G. p. pinetis*.

1971). The young are born nearly hairless with pinkish wrinkled skin (Barrington 1942). A furrow is present at birth on the sides of the mouth where the cheek patches will later form. The young are weaned and disperse in approximately 4 weeks, and sexual maturity is thought to occur in 4–6 months (Wing 1960, Brown 1971). Sex ratios tend to be skewed towards females (Wing 1960). However, Brown (1971) noted that sex ratios were skewed towards males in the juvenile age class, approximately equal in subadults, and skewed toward females in adults, indicating lower survival in males.

FOOD HABITS

Food habits of the southeastern pocket gopher are largely unknown (Gates and Tanner 1988). Foods generally include a variety of roots, bulbs, and leaves (Golley 1962). Items documented from food caches include bahia grass (*Paspalum notatum*), dog fennel (*Eupatorium* spp.), lambsquarters (*Chenopodium* spp.), and false moneywort (*Alysicarpus vaginalis*; Ross 1976). Captive animals have eaten nut grass (*Cyperus esculentus*), Bermuda grass (*Cynodon dactylon*), and wiregrass (*Aristida* spp.; Barrington 1940). Food items are transported in the cheek pouches to storage chambers (Golley 1962).

ASSOCIATED SPECIES

Pocket gophers occur in areas inhabited by Coastal Plain sandhills community species. Mammalian associates include the southeastern shrew (*Sorex longirostris*), least shrew (*Cryptotis parva*), southern short-tailed shrew (*Blarina carolinensis*), eastern mole (*Scalopus*

aquaticus), eastern harvest mouse (*Reithrodontomys humulis*), hispid cotton rat (*Sigmodon hispidus*), cotton mouse (*Peromyscus gossypinus*), and oldfield mouse (*P. polionotus*). Reptilian associates include gopher tortoise (*Gopherus polyphemus*), eastern diamondback rattlesnake (*Crotalus adamanteus*), and eastern indigo snake (*Drymarchon corais couperi*). Potential predators include owls (Brown 1971), long-tailed weasels (*Mustela frenata*; Sherman 1929), and spotted skunks (*Spilogale putorius*; Howell 1920).

VULNERABILITY AND THREATS

The southeastern pocket gopher is widely distributed and may be common in appropriate habitat. However, populations generally have declined because much of the sandhills community type throughout the region has been converted to other uses (Winchester and DeLotelle 1978). Comparisons of present distribution records with historical occurrences suggest that previously known populations are no longer extant and that many populations are decreasing (Laerm 1981b). The extinction of *G. p. goffi* was likely a result of conversion of suitable habitat to urban and suburban land uses and habitat changes resulting from fire exclusion (Humphrey 1981). Pocket gophers are viewed as an agricultural, silvicultural, and residential pest and are sometimes exterminated.

MANAGEMENT SUGGESTIONS

Frequent fire is required to maintain the sandhills habitat in a condition suitable for pocket gopher habitation (Humphrey 1981, Gates and Tanner 1988). Conversion of sandhill habitat to intensively managed forests may reduce the suitability as pocket gopher habitat (Winchester and DeLotelle 1978).

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Hispid Pocket Mouse

Chaetodipus hispidus (Baird, 1858)

Steven B. Castleberry and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Formerly included in the genus *Perognathus* (Hall 1981), the hispid pocket mouse and other spiny-rumped pocket mice are now referred to the genus *Chaetodipus* (Hafner and Hafner 1983). Four subspecies are recognized, two of which occur in the South: *C. h. hispidus* and *C. h. spilotus* (Hall 1981, Paulson 1988). The literature is reviewed by Paulson (1988).

DISTINGUISHING CHARACTERISTICS

The hispid pocket mouse is the only representative of the Family Heteromyidae occurring in the region. The genus *Chaetodipus* is characterized by elongate hind limbs, external fur-lined cheek pouches, and elongate spiny bristles on rump. The hispid pocket mouse has a bristly pelage that is olive to ochraceous and interspersed with black hairs above. The sides are buffy and the venter is white. Measurements are: total length, 184–237 mm; tail, 82–114 mm; hind foot, 23–29 mm; ear, 8–12 mm; weight, 30–47 g. In comparison to other *Chaetodipus* species, the tail is shorter and not crested. The dental formula is I 1/1, C 0/0, P 1/1, M 3/3 = 20 (Figure 1). See keys for details.

CONSERVATION STATUS

The hispid pocket mouse has a global rank of Secure (NatureServe 2007). It is considered Secure in Oklahoma and Texas, and it is considered Imperiled in Louisiana.

DISTRIBUTION

The regional distribution of the hispid pocket mouse is limited to eastern Texas and Oklahoma and western Louisiana (Hall 1981, Paulson 1988; Figure 2). *Chaetodipus h. spilotus* occurs throughout eastern Oklahoma, except the extreme northeastern and southeastern corners (Caire et al. 1989), and the Red River Valley of northeastern Texas (Glass 1947). *Chaetodipus h. hispidus* occurs south of the Red River Valley in eastern Texas, east into adjacent areas of west-central Louisiana (Glass 1947, Lowery 1974).

ABUNDANCE STATUS

The hispid pocket mouse is widespread and common in the western and northern portions of the

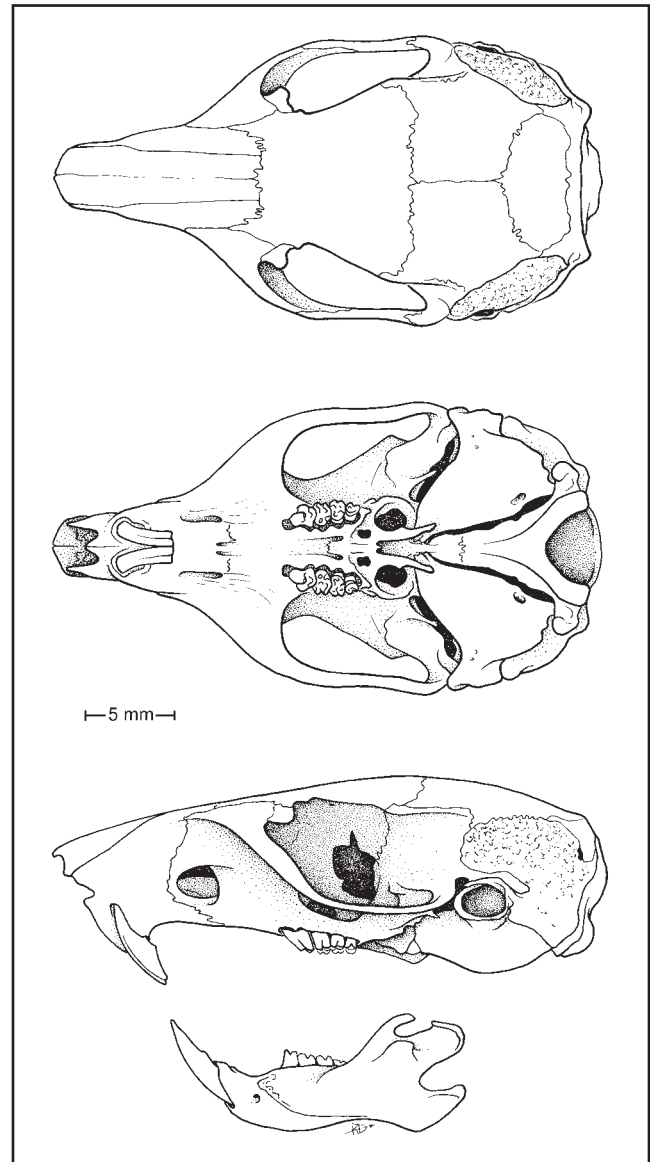


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Chaetodipus hispidus* from Wichita County, Texas (USNM 348437, male).

distribution (Jones et al. 1983). Regionally, the species is apparently abundant in eastern Oklahoma (Blair 1938) and Texas (Schmidly 1983), but relatively rare near the eastern limit of the range in Louisiana (Lowery 1974). No regional population estimates are available.

PRIMARY HABITATS

The hispid pocket mouse occurs in a wide variety of grassland vegetation and soil types throughout the range. In the South, it appears to be most commonly associated with grassland habitats containing broom-sedge (*Andropogon* spp). In eastern Texas, it is reported to be most common in areas containing well-drained soils with vegetation dominated by broomsedge and weeds, with scattered stands of woody vegetation (Schmidly 1983). It also is reported from sandy hillsides dominated by shortleaf pine (*Pinus echinata*) and black locust (*Robinia pseudoacacia*). In eastern Oklahoma, it occurs in a wide variety of grassland habitats, including the grama-beardgrass, plum thicket, and sumac-grama associations (Blair 1938). In western Louisiana it has been collected in grassy fields with dense stands of broomsedge and in grassy areas within longleaf pine (*P. palustris*) forests (Lowery 1974).

Hispid pocket mice construct burrows that are apparently used for nesting and food storage (Blair 1937, Thompson and Barrett 1967). Burrow entrances typically are associated with areas of dense vegetation (Blair 1937, Hill 1942). Blair (1937) noted that burrows in northeastern Oklahoma were typically found underneath limestone slabs. Nests are constructed 0.3–0.6 m below the surface and are comprised of grass strips placed in a small chamber at the end of the burrow (Thompson and Barrett 1967). A separate chamber for food storage may also be present (Blair 1937).

REPRODUCTION

Little information is available regarding reproduction in the hispid pocket mouse. The breeding season is variable depending on location (Schmidly 1983). Generally, 1–2 litters are produced annually from June to August in the northern parts of the range, whereas breeding occurs year round in the southern extent. In western Texas, the hispid pocket mouse may breed year-round when conditions are favorable, but breeding activity is greater from early spring to autumn (Choate and Jones 1989). On the northern Great Plains, the reproductive season lasts from spring through late summer (Jones et al. 1983). The number of young per litter is reported to range from 2–9 with an average of 6 (Schmidly 1983). Choate and Jones (1989) documented a range of 4–10 embryos with a mean of 6.3. Females apparently can reproduce during the breeding season of birth (Choate and Jones 1989).

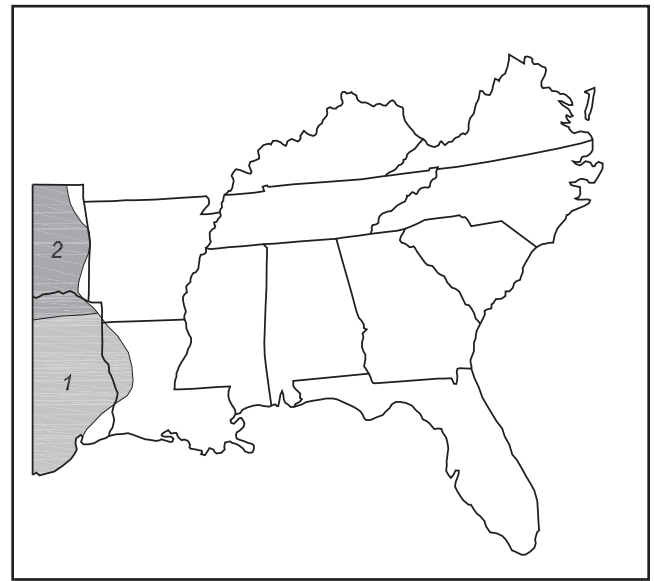


Figure 2. Distribution of *Chaetodipus hispidus* in the South: (1) *C. h. hispidus*; (2) *C. h. spilotus*.

FOOD HABITS

The regional diet is relatively unknown, but probably consists primarily of seeds and leaves of available vegetation. Availability of plants in proximity to the burrow is an important factor in food selection (Blair 1937). Based on observations of seeds stored in burrows, Blair (1937) concluded that blanket flower (*Gaillardia pulchella*) and prickly pear (*Opuntia* spp.) were important foods in northeastern Oklahoma. He also noted insects as a regular part of the diet. Sericea lespedeza (*Lespedeza sericea*) seeds were found in the cheek pouches of a specimen captured near Fort Jesup, Louisiana (Lowery 1974). Hispid pocket mice are known to cache large quantities of seeds in their underground burrows (Blair 1937, Schmidly 1983).

ASSOCIATED SPECIES

Common mammalian associates of the hispid pocket mouse may include Elliott's short-tailed shrew (*Blarina hylophaga*), least shrew (*Cryptotis parva*), harvest mice (*Reithrodontomys* spp.), white-footed mouse (*Peromyscus leucopus*), deer mouse (*P. maniculatus*), eastern woodrat (*Neotoma floridana*), southern plains woodrat (*N. micropus*), hispid cotton rat (*Sigmodon hispidus*), northern pigmy mouse (*Baiomys taylori*), and thirteen-lined ground squirrel (*Spermophilus tridecemlineatus*; Caire et al. 1989, Roberts et al. 1997). Known predators of hispid pocket mice are western diamondback rattlesnakes (*Crotalus atrox*; Beavers 1976), great-horned owls (*Bubo virginianus*; Tyler and Jensen 1981), and common barn owls (*Tyto alba*; Pesaturo et al.

1989). They probably also are preyed upon by other snakes, owls, and mammals (Paulson 1988).

VULNERABILITY AND THREATS

Although the hispid pocket mouse is apparently rare in Louisiana, no specific threats to the survival of those populations have been identified.

MANAGEMENT SUGGESTIONS

Although somewhat tolerant of agricultural practices (Fleharty and Navo 1983, Kaufman et al. 2000), conversion of native prairie to wheat (*Triticum aestivum*) production may reduce abundance of the species (Kaufman and Kaufman 1990). Hispid pocket mice can cause considerable damage to agricultural operations by digging up seeds of planted crops (Davis and Schmidly 1994).

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Northern Pygmy Mouse

Baiomys taylori (Thomas, 1887)

Joshua Laerm, W. Mark Ford, and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

There are eight described subspecies of the northern pygmy mouse (*Baiomys taylori*). Two occur in the South: *B. taylori subater* and *B. t. taylori* (Packard 1960, Hall 1981). The literature was summarized by Eshelman and Cameron (1987).

DISTINGUISHING CHARACTERISTICS

The northern pygmy mouse is among the smallest of North American rodents. Its measurements are: total length, 87–123 mm; tail, 34–53 mm; hind foot, 12–15 mm; ear, 9–12 mm; weight, 6–10 g. The dental formula is: I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). The pelage of the northern pygmy mouse is highly variable, ranging from grizzled reddish-brown to gray dorsally, and white, creamy buff, or gray ventrally. The tail is short, sparsely haired, and can be either uniformly gray or bicolored. This species is not confused easily with other sympatric sigmodontine rodents, such as *Peromyscus* and *Reithrodontomys*, all of which have longer body, tail, and hind foot lengths. See keys for details.

CONSERVATION STATUS

The northern pygmy mouse has a global rank of Apparently Secure. Texas also considers it Apparently Secure. (NatureServe 2007).

DISTRIBUTION

The northern pygmy mouse has been expanding its range both northward and eastward since the turn of the 20th century (Hunsaker et al. 1959, Schmidly 1983, Eshelman and Cameron 1987, Davis and Schmidly 1994). Figure 2 depicts the distribution of the northern pygmy mouse in the South. This species ranges throughout central and northern Mexico north into Arizona and New Mexico, with another northern extension into central and eastern Texas along the coast close to Louisiana (Blair 1941, Blair 1950, Hunsaker et al. 1959, Dalquest 1968, Schmidly 1983). Significant northern and western range expansions have been reported in Texas (Austin and Kitchens 1986, Hollander et al. 1987, Pitts and Smolen 1989, Jones and Manning 1989, Roberts et al. 1997, Pitts et al. 2001). Northern pygmy mice also have been reported recently from south-central and

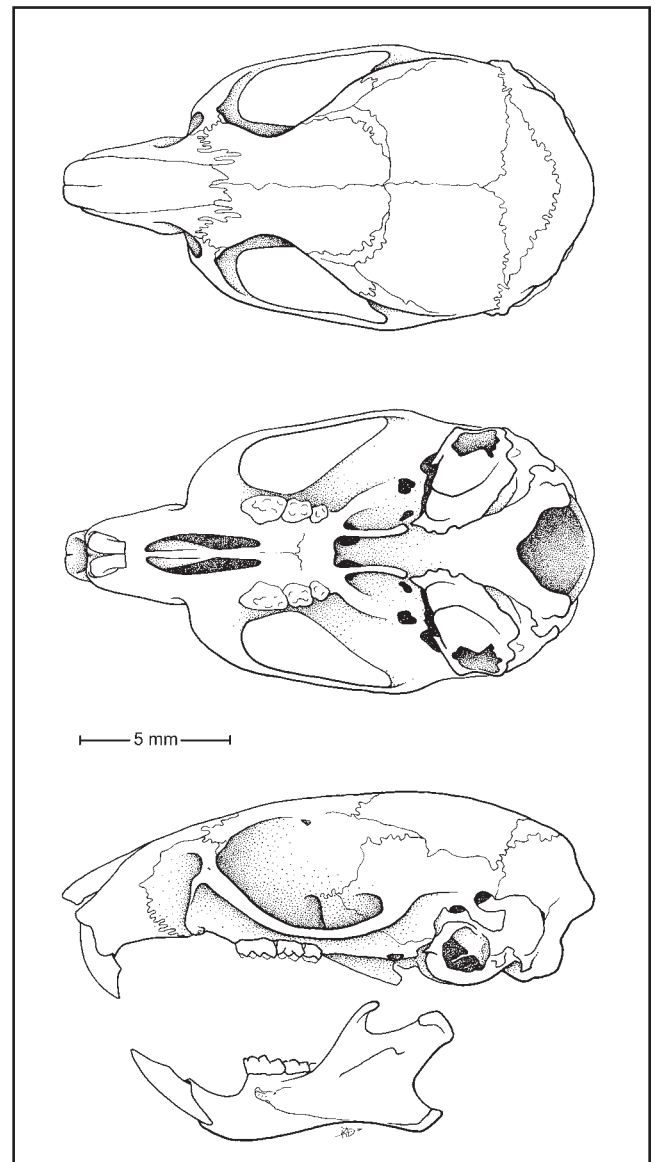


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Baiomys taylori* from Nayarit, Mexico (USNM 509957, female).

western Oklahoma (Stangl and Dalquest 1986, Cleveland 1986, Caire et al. 1989, Tumlinson et al. 1993). Northern populations probably are limited by cold weather and winter-kill (Caire 1991).

ABUNDANCE STATUS

The species is relatively common throughout most of eastern Texas, with density estimates ranging from 2 to 84 animals/ha depending upon habitat type, ground cover density, and time of year (Raun and Wilks 1964, Schmidly 1983, Grant et al. 1985). Densities are lowest in summer and highest in early fall and winter, apparently because populations can be depressed by high populations of hispid cotton rats (*Sigmodon hispidus*; Stickel and Stickel 1949, Raun and Wilks 1964, Schmidly 1983, Grant et al. 1985).

PRIMARY HABITATS

Although the northern pygmy mouse occurs in a wide variety of habitats throughout its range (Eshelman and Cameron 1987), in Texas this species occurs in dense grassy areas such as oldfields, prairies, pastures, and road banks and ditches. It also has been reported in orchards, post oak (*Quercus stellata*) savannas, pine-oak (*Pinus-Quercus*) forests, and oak-hickory (*Carya* spp.) forests (Bailey 1905, Blair 1941, Taylor and Davis 1947, Blair 1952, Hunsaker et al. 1959, Schmidly 1983, Tumlinson et al. 1993, Hanchey and Wilkins 1998, Pitts et al. 2001).

REPRODUCTION

The northern pygmy mouse is capable of breeding year-round, with an average of 4 weeks between litters. However, there are peaks in the early spring and late fall (Blair 1941, Packard 1960, Asdell 1964, Raun and Wilks 1964). Gestation is 20–23 days and litter size ranges from 1 to 5. Young are weaned at 17–24 days and average sexual maturity is 60–90 days (Blair 1941, Packard 1960, Quadagno et al. 1970, Hudson 1974). Morrison et al. (1977) reported a median life span of 23 weeks (up to a maximum of 170 weeks) in captivity.

FOOD HABITS

Common foods of the northern pygmy mouse include prickly pear cactus (*Opuntia* spp.) stems and fruits, grass shoots and seeds, mesquite (*Prosopis glandulosa*) seeds, and granjero (*Celtis* spp.) berries. It also will feed on terrestrial gastropods and, at least in captivity, will consume small snakes and insects (Raun and Wilks 1964, Pitts 1978).

ASSOCIATED SPECIES

The northern pygmy mouse is sympatric with other early successional habitat and grassland/prairie species such as the least shrew (*Cryptotis parva*), fulvous harvest mouse (*Reithrodontomys fulvescens*), plains

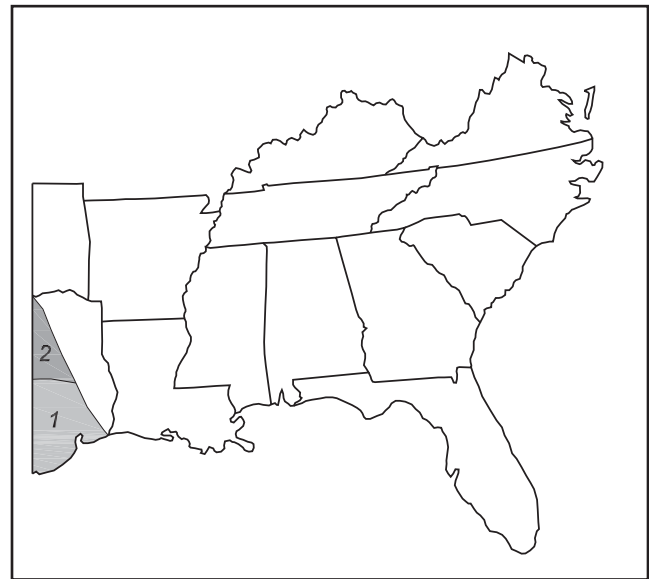


Figure 2. Distribution of *Baiomys taylori* in the South: (1) *B. t. subater*; (2) *B. t. taylori*.

harvest mouse (*R. montanus*), rice rat (*Oryzomys palustris*), white-footed mouse (*Peromyscus leucopus*), and hispid cotton rat.

VULNERABILITY AND THREATS

The northern pygmy mouse is at the eastern and northern limit of its range in this region. However, it is common, occurs in a variety of habitats, and shows evidence of range expansion. Killion et al. (1995) suggest that the northern pygmy mouse avoids locating its burrows in areas with high concentrations of the red imported fire ant (*Solenopsis invicta*). Although undetermined, red imported fire ants may adversely affect the reproductive success of the northern pygmy mouse.

MANAGEMENT SUGGESTIONS

Periodic habitat disturbance such as fire or mowing probably maintains northern pygmy mouse populations from a landscape perspective, particularly in wooded and shrubby conditions of eastern and coastal Texas. Nonetheless, activities that reduce or maintain a low ground cover density could favor other species that compete with the northern pygmy mouse, such as the hispid cotton rat.

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Southern Red-backed Vole

Clethrionomys gapperi (Vigors, 1830)

John F. Pagels and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Twenty-nine subspecies of the southern red-backed vole, *Clethrionomys gapperi*, are recognized (Hall 1981, Merritt 1981). Three of the subspecies occur in the South: *C. g. carolinensis*, *C. g. gapperi*, and *C. g. maurus*. In portions of southwestern Virginia and nearby states, the geographical distributions of the three subspecies are poorly defined. Additional collecting and genetic studies, (e.g. see Reese et al. 2001) are needed to clarify geographical boundaries. Merritt (1981) reviewed the literature.

DISTINGUISHING CHARACTERISTICS

The southern red-backed vole is a medium-sized vole with small eyes and relatively prominent ears. Measurements of are: total length, 120–170 mm; tail, 30–50 mm; hind foot, 16–21 mm; ear, 12–16 mm; weight, 16–42 g. Coloration of the dorsal pelage is variable, but as the name suggests it is characterized by a broad rusty or reddish band that extends from forehead to rump in much of its range. The sides of the head and body are grayish washed with buff and the venter is pale gray to silverish. *Clethrionomys g. gapperi*, which occurs in most of western Virginia, is generally lighter than the darker *C. g. carolinensis* and *C. g. maurus* and has the typical reddish dorsum. In all forms, summer pelage tends to be darker than winter pelage and pelage of young tends to be darker than pelage of adults. The tail is bicolored, dark brown to black above and white to pale gray below. The southern red-backed vole is readily distinguished from other voles by its reddish back, grayish sides, and paler venter. The skull lacks the grooved incisors characteristic of *Synaptomys*, and unlike voles of the genus *Microtus*, the posterior border of the palate is a thin-edged straight shelf. The dental formula is $I\ 1/1, C\ 0/0, P\ 0/0, M\ 3/3 = 16$ (Figure 1). See keys for details.

CONSERVATION STATUS

Clethrionomys gapperi has a global rank of Secure (NatureServe 2007). It is considered Apparently Secure in North Carolina and Tennessee, and Secure in Virginia. In Georgia, the conservation status of *C. gapperi* is uncertain; however, it is considered to be within the range of Vulnerable/Apparently Secure. The global rank of *C. g. maurus* is uncertain but is

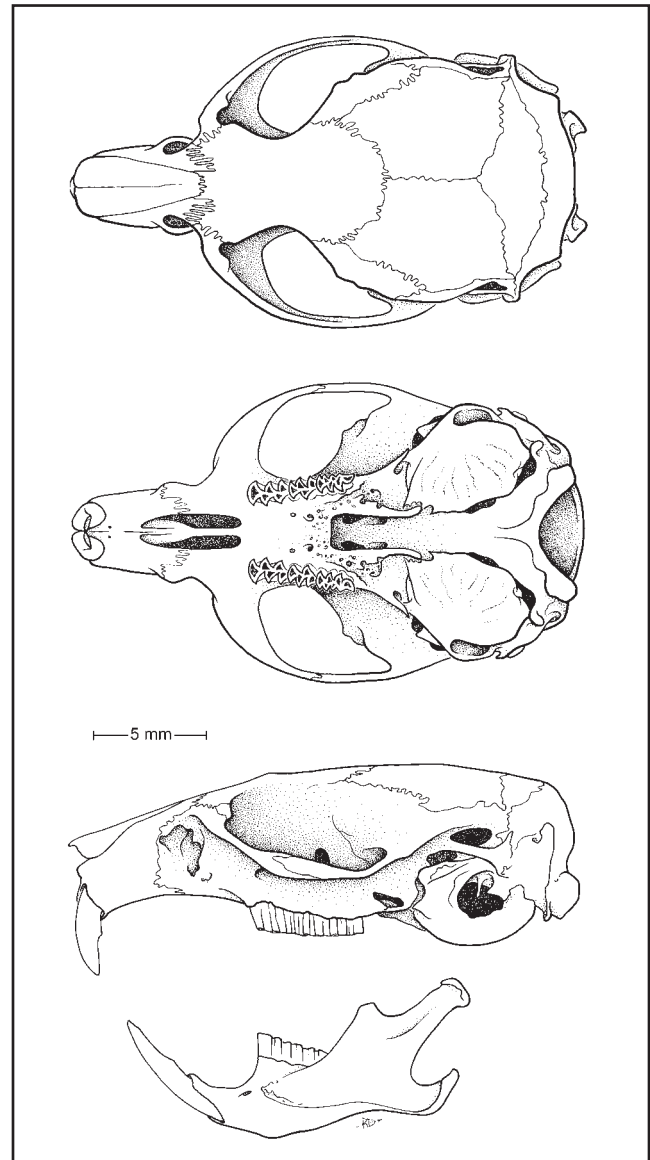


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Clethrionomys gapperi* from Roan Mountain, Pisgah National Forest, North Carolina (USNM 54463, male).

considered to be within the range of Vulnerable/Apparently Secure; it is classified as Vulnerable in Kentucky. *Clethrionomys g. carolinensis* has a global rank of Apparently Secure and is considered to be within the range of Imperiled/Vulnerable in South Carolina.

DISTRIBUTION

The southern red-backed vole occurs throughout the transcontinental coniferous forests from the Canadian Arctic south into the northern United States with extensions into the Rocky and Appalachian Mountains (Figure 2). In the region, it is restricted to higher elevations of the Appalachian Mountains of Virginia (Kellogg 1939, Handley and Patton 1947, Handley 1971, Kalko and Handley 1993, Reese et al. 2001), eastern Kentucky (Kellogg 1939, Barbour and Davis 1974), eastern Tennessee (Howell and Conaway 1952, Conaway and Howell 1953, Linzey and Linzey 1968, Smith et al. 1974, Harvey et al. 1991, 1992; Todd 1992), western North Carolina (Odum 1949, Johnston 1967, Gentry et al. 1968, Lee et al. 1982), extreme northwestern South Carolina (Pivorun et al. 1984, Laerm et al. 1995), and northeastern Georgia (Wharton and White 1967, Laerm et al. 1982, Ford et al. 1994).

ABUNDANCE STATUS

The southern red-backed vole is a common species in high elevation mesic hardwood and coniferous forests of the Appalachian Mountains. No density estimates are available for the region, but densities elsewhere have been reported to range from 24–65/ha in New York, up to 37/ha in Connecticut and Vermont, and 1–7/ha in New Hampshire (Miller and Getz 1972, 1977; Merritt 1981). Most reports for the region note the species is common to abundant in appropriate high elevation mesic habitats (Conaway and Howell 1953, Johnston 1967, Linzey and Linzey 1968, Orrock et al. 2000, McShea et al. 2003), but uncommon (Howell and Conaway 1952, Ford et al. 1994) to rare (Pivorun et al. 1984, Laerm et al. 1995) in xeric lower elevation cover types or at the periphery of its range. Steele and Powell (1999) found that the density of *C. gapperi* was greater on northern mountaintops than on the more southern mountaintops on major mountain ranges in North Carolina.

PRIMARY HABITATS

In the southern extent of its range the southern red-backed vole is largely restricted to red spruce-fir (*Picea rubens-Abies*), maple-beech-birch (*Acer-Fagus-Betula*), cove hardwood, mesic oak-hickory (*Quercus-Carya*), and white pine-hemlock (*Pinus strobus-Tsuga canadensis*) cover types where there is an abundance of fallen logs, moss-covered rocks, talus, and coarse woody debris (Kellogg 1939, Conaway and Howell 1953, Johnston 1967, Wharton and White 1967, Linzey and Linzey 1968, Smith et al. 1974, Wharton 1978, Pivorun et al. 1984, Cranford and Maly 1986, Barry et al. 1990, Laerm et al. 1995,

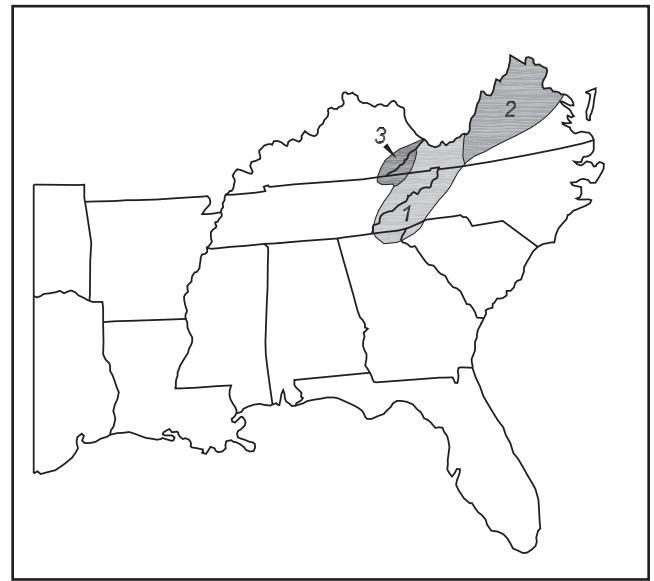


Figure 2. Distribution of *Clethrionomys gapperi* in the South: (1) *C. g. carolinensis*; (2) *C. g. gapperi*; (3) *C. g. maurus*.

Steele and Powell 1999, Orrock et al. 2000, McShea et al. 2003). In forested habitat in Virginia, the presence and abundance of *C. gapperi* are greater in mesic situations and with presence of the aforementioned forest floor features (Orrock et al. 2000, McShea et al. 2003). The species has been captured at the bases of shrubs on grassy balds (Komarek and Komarek 1938). Ford et al. (1999) found no differences in numbers of red-backed voles in post-fire burn and control sites following a prescription fire. Like many species of small mammals, abundance of the southern red-backed vole usually increases temporarily in recent clearcuts as a result of herbaceous plant growth (Kirkland 1990). Similarly, Menzel et al. (1999) found relatively high numbers of *C. gapperi* at field and forest edges at wildlife openings, presumably the result of the increased plant diversity and structural heterogeneity in edges. Cranford and Maly (1986) collected *Clethrionomys* in a small wildlife clearing and suggested they may have been transient individuals from the surrounding forest. While it is known from elevations as low as 533 m in the Great Smoky Mountains, it is most common above 914 m (Linzey and Linzey 1968). The red-backed vole typically utilizes the burrows and runways of other species. Nests are usually constructed underground or located under coarse woody debris.

REPRODUCTION

Little specific information on breeding is available for the red-backed vole in the southern portion of its range. Linzey and Linzey (1968) reported reproductive activity between April and late August in the

Great Smoky Mountains. Merritt (1981) summarized data on reproduction. In most areas breeding occurs from late winter to late fall. The species is polyestrous and exhibits a post-partum estrus. Gestation is reported at 17–19 days and litter size ranges from 2–8. Weaning occurs at 12–17 days and sexually maturity at 2–4 months. Maximum longevity is about 20 months but most live less than one year.

FOOD HABITS

Foods of the red-backed vole include the vegetative portions of plants, fruits, seeds, roots, berries, fungi, mosses, lichens, ferns, and some insects (Linzey and Linzey 1973, Schloyer 1977, Perrin 1979, Merritt 1981). Merritt's (1981) summary noted the species is an opportunistic feeder whose diet shifts in response to availability. Orrock and Pagels (2002) found *C. gapperi* is a fungal generalist and suggest analysis of its diet may be useful for assessing the availability of fungi for other, more specialized mycophagists.

ASSOCIATED SPECIES

In the region, the red-backed vole is typically most abundant in mesic habitats that include the northern short-tailed shrew (*Blarina brevicauda*), masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), deer mouse (*Peromyscus maniculatus*), woodland jumping mouse (*Napaeozapus insignis*), and rock vole (*Microtus chrotorrhinus*; Pagels and Tate 1976, Pagels 1990, Steele and Powell 1999, McShea et al. 2003). It has been captured with other species, including the nearly ubiquitous white-footed mouse (*P. leucopus*), golden mouse (*Ochrotomys nuttali*), and southern bog lemming (*Synaptomys cooperi*).

VULNERABILITY AND THREATS

While it is limited in its southern distribution, and restricted to high elevation mesic forests, the southern red-backed vole is common to abundant in appropriate habitats. Nowhere does there appear to be threats to its survival. However, in southerly portions of its range it is more isolated at high elevations than farther to the north. Long-term events that could lead to warming and drying in the southern Appalachians would lead to still greater distributional isolation on high elevation islands, or loss of its presence in the region.

MANAGEMENT SUGGESTIONS

Although the red-backed vole appears to be secure in the region providing that appropriate mesic habitat with sufficient cover is available, activities that warm or dessicate such habitats should be discouraged.

Maintenance of suitable habitat for the red-backed vole will benefit a suite of boreal northern organisms, including many forms that are uncommon or rare.

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Rock Vole

Microtus chrotorrhinus (Miller, 1894)

John F. Pagels and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Three subspecies of the rock vole, *Microtus chrotorrhinus*, are recognized (Hall 1981, Kirkland and Jannett 1982). *Microtus c. carolinensis*, occurs in the South. Kirkland and Jannett (1982) summarize literature on *M. chrotorrhinus*, and chapters in Tamarin (1985) provide information on New World members of the genus *Microtus*, including *M. chrotorrhinus*.

DISTINGUISHING CHARACTERISTICS

The rock vole is similar in size to the more widespread meadow vole (*Microtus pennsylvanicus*). Rock voles leave the nest when relatively young and, like many species, measurements of young and adults may vary considerably. Total length may range from 140–185 mm; tail, 42–64 mm; hind foot, 18–24 mm; ear, 12–18 mm; and weight, 30–48 g. The pelage tends to be coarser than the meadow vole. The upper surface is grayish to glossy brown with black-tipped hairs, and the venter is pale gray to grayish white. The face usually exhibits a yellow to dull orange wash, most prominent on the snout and fading toward the ears. The tail is somewhat bicolored, dark above and lighter below. The dental formula is $I\ 1/1$, $C\ 0/0$, $P\ 0/0$, $M\ 3/3 = 16$ (Figure 1). The skull of *M. chrotorrhinus* is distinguishable from other species of *Microtus* only by examination of teeth. See keys for details.

CONSERVATION STATUS

Microtus chrotorrhinus has a global rank of Apparently Secure; however, the subspecies *M. c. carolinensis* has a global rank of Vulnerable (NatureServe 2007). In Virginia, *M. c. carolinensis* is classified as Critically Imperiled and is listed as Endangered by the Virginia Department of Game and Inland Fisheries. It is considered Vulnerable in North Carolina. In South Carolina, the conservation status of *M. chrotorrhinus* is uncertain. It is unranked in Tennessee. Adams (1987) and Handley and Pagels (1991) comment on the conservation status of the species in the South.

DISTRIBUTION

The geographic range of the rock vole extends from Labrador and the Maritime Provinces west to southwestern Ontario and northeastern Minnesota (Timm

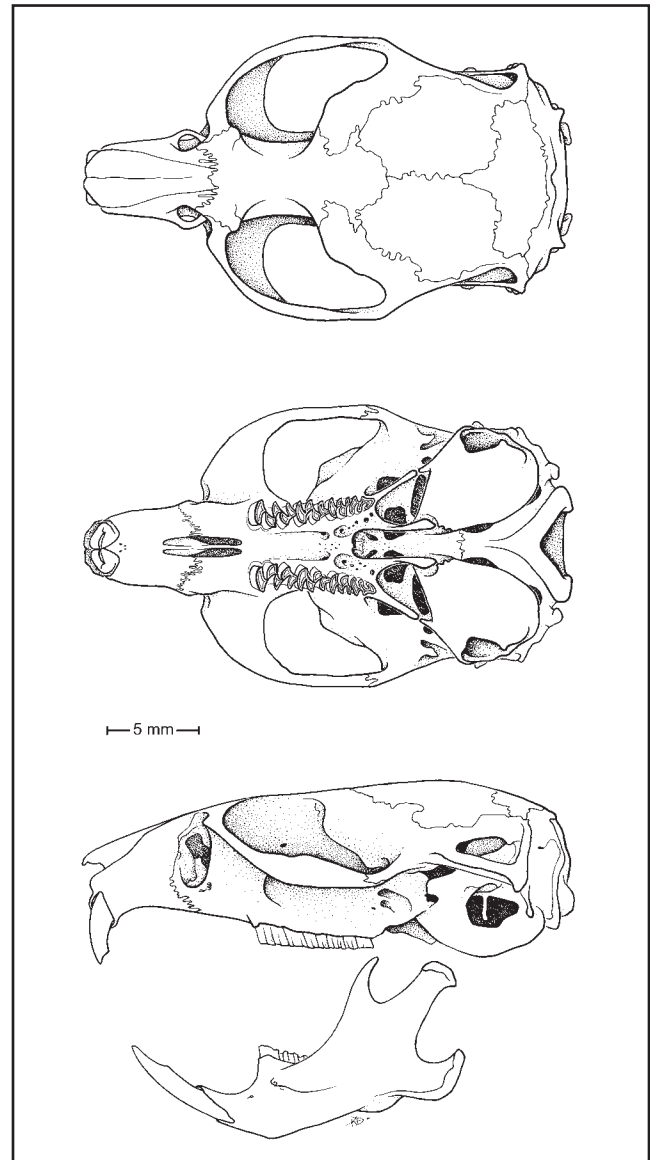


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Microtus chrotorrhinus* from Great Smoky Mountains National Park, North Carolina (USNM 250443, female).

et al. 1977, Jannett 1990), south throughout the Adirondack and Appalachian mountains into northeastern Pennsylvania (Kirkland 1985, Kirkland and Hart 1999). There is a large hiatus between populations in northeastern Pennsylvania and the nearest localities to the south in western Maryland (D. Feller,

Maryland Department of Natural Resources, personal communication), and West Virginia (Kirkland 1977a, Handley and Pagels 1991). Approximately 35 km separates West Virginia populations from four known Virginia localities in Bath and Highland counties (Pagels 1990, Orrock et al. 1999). The Virginia populations are disjunct from sites in the Great Smoky Mountains of eastern Tennessee (Linzey and Linzey 1968, Steele and Powell 1999) and the few scattered localities in extreme western North Carolina (Lee et al. 1982, Adams 1987, Steele and Powell 1999). In the late Pleistocene, the rock vole had a greater distribution in the Appalachian Mountains than now (Guilday 1962, Guilday et al. 1977). The species exists in the mid-to-southern Appalachian Mountains as a Pleistocene relict (Figure 2).

ABUNDANCE STATUS

The status of the rock vole is uncertain because of its fragmented distribution. Only five specimens were taken from the four Virginia localities (Pagels 1990, Orrock et al. 1999). In the Great Smoky Mountains, the rock vole is apparently common and has been reported from several localities and a range of elevations (Linzey and Linzey 1968, Steele and Powell 1999). In western North Carolina, it is rare and localized with most of its localities known from single specimen records (Lee et al. 1982, Adams 1987, Steele and Powell 1999). In addition to typical forested habitat, in eastern West Virginia it occurs locally in clearcuts of northern coniferous and mixed deciduous forests where emergent rock is abundant (Kirkland 1977a,b; Healy and Brooks 1988).

PRIMARY HABITATS

The rock vole is often associated with boreal forests of the Canadian Life Zone; however, it also occurs in deciduous Transition Zone forests (Martin 1971, Kirkland and Knipe 1979, Kirkland and Jannett 1982). It is associated with cool, moist, log-strewn, moss covered rocks, boulders, and talus where it inhabits subsurface runs in red spruce-fir (*Picea rubens-Abies*), maple-beech-birch (*Acer-Fagus-Betula*), cove hardwood, mesic oak-hickory (*Quercus-Carya*), and white pine-hemlock (*Pinus strobus-Tsuga canadensis*) cover types (McKeever 1952, Linzey and Linzey 1968, Martin 1971, Kirkland 1977a,b; Timm et al. 1977, Kirkland and Knipe 1979, Kirkland 1985, Adams 1987, Healy and Brooks 1988, Pagels 1990, Orrock et al. 1999). Water in the form of streams, springs, or seeps is often a conspicuous element of the habitat (Martin 1971, Kirkland and Knipe 1979, Kirkland and Jannett 1982, Handley and Pagels 1991). In northern deciduous and boreal coniferous forests of West Virginia, Kirkland (1977a,b) found

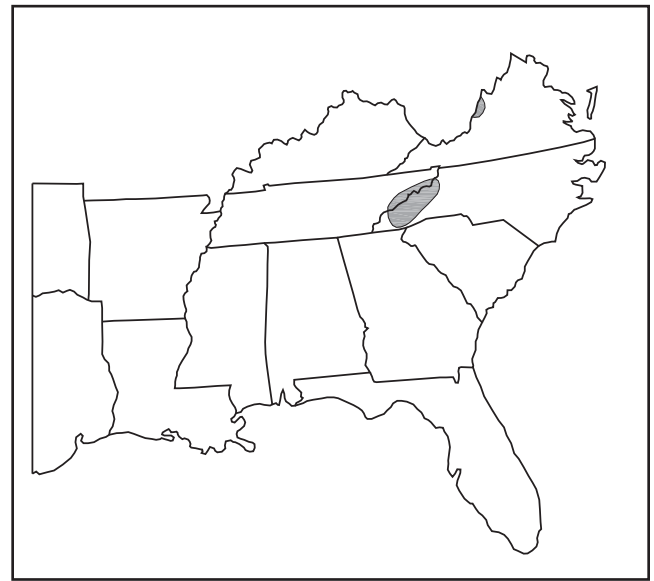


Figure 2. Distribution of *Microtus chrotorrhinus* in the South.

that numbers of rock voles increased dramatically in red spruce clearcuts and decreased slightly in deciduous forest clearcuts compared to older, forests of each type. Kirkland's data indicate the rock vole exploits recently disturbed sites and, similar to many species of small mammals (Kirkland 1990), temporarily benefits from the lush herbaceous growth that follows clearcutting. All four Virginia sites where *M. chrotorrhinus* has been found are mixed mesophytic habitats dominated by yellow birch (*Betula alleghaniensis*) with a forest floor of moss-covered talus or rocks (Pagels 1990, Orrock et al. 1999). In the Great Smoky Mountains and western North Carolina, it has been found at elevations ranging from 790 to 1675 m associated with maple-beech-birch stands and red spruce-fir habitat and sometimes in open balds or grassy areas (Komarek and Komarek 1938, Linzey and Linzey 1968, Adams 1987, Steele and Powell 1999).

REPRODUCTION

Breeding is reported from March to October (Linzey and Linzey 1968, Martin 1971, Kirkland 1977a). Gestation is 19–21 days (Kirkland and Jannett 1982). Litter size ranges from 1–7 with means ranging from 2.9–3.7 (Martin 1971, Timm et al. 1977). Females exhibit a postpartum estrus (Kirkland and Jannett 1982).

FOOD HABITS

Observations on food habits of the rock vole are primarily from other parts of the species range. Individuals from the northeastern United States and Canada were reported to feed on vegetation including significant amounts of bunchberry (*Cornus canadensis*),

seeds, berries of a variety of plants, including blueberries (*Vaccinium* spp.), ferns, grass stems and leaves, roots, and fungi (Whitaker and Martin 1977). A subadult rock vole in captivity consumed numerous insects (Timm et al. 1977).

ASSOCIATED SPECIES

In southern portions of its range, the rock vole is frequently found in association with other small mammal species with boreal affinities, the most notable being the red-backed vole (*Clethrionomys gapperi*). Other common associates include the masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), deer mouse (*Peromyscus maniculatus*), and woodland jumping mouse (*Napaeozapus insignis*). Kirkland and Jannett (1982) noted that *C. gapperi* is a consistent associate of the rock vole in other areas, and Orrock and Pagels (2003) suggest that in Virginia a high abundance of *C. gapperi* is a useful indicator of suitable *M. chrotorrhinus* habitat.

VULNERABILITY AND THREATS

The rock vole is able to exploit, if not benefit from, disturbed forest communities, but presumably source populations must be very near or contiguous, and as a result of forest succession the benefit of disturbance is temporary (Kirkland 1990). The species may be more widely distributed in the southern Appalachians than currently recognized. However, after many years of small mammal surveys, in general, and many recent surveys directed towards boreal species in particular, it appears that in the central to southern Appalachian Mountains the rock vole occurs in isolated, relict populations. As such, any factors (i.e. global warming) whether anthropomorphic or natural, and other more localized activities that could warm or dessicate islands of suitable habitat where it now occurs, could negate future occurrence at such islands.

MANAGEMENT SUGGESTIONS

Because of the island-like occurrence of rock vole populations, management considerations where the vole occurs should be directed towards those practices that maintain cool, moist areas characterized by moss-covered talus and boulders. Further, similar sites should not be disturbed to allow for potential repopulation of such sites by the rock vole, and to provide habitat that is required by other small boreal species in the southern Appalachians (McShea et al. 2003).

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Prairie Vole

Microtus ochrogaster (Wagner, 1852)

Joshua Laerm and W. Mark Ford

CONTENT AND TAXONOMIC COMMENTS

There are seven subspecies of prairie voles (*Microtus ochrogaster*) currently recognized, three of which are regionally extant: *M. o. haydenii*, *M. o. ochrogaster*, and *M. o. ohionensis* (Stalling 1990). Disjunct populations referred to *M. o. ludovicianus* are believed extinct. The literature was reviewed by Johnson and Johnson (1982), Tamarin (1985) and Stalling (1990).

DISTINGUISHING CHARACTERISTICS

The prairie vole is a robust, medium-sized vole with short tail. Its measurements are: total length, 125–172 mm; tail, 24–45 mm; hind foot, 17–22 mm; ear, 11–15 mm; weight, 37–73 g. The dental formula is: I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). The molar dentition of the skull is unique, as the third upper molar has two closed triangles, whereas the first lower molar has three closed and two open triangles. The pelage is long and coarse, with grizzled brown or grayish brown dorsal coloration, paler sides, and gray or gray washed with pale cinnamon coloration ventrally. The tail is slightly bicolor. See keys for details.

CONSERVATION STATUS

The prairie vole has a global rank of Secure (NatureServe 2007). The species is also considered Secure in Oklahoma and Kentucky. It is listed as Apparently Secure in Arkansas, but is Vulnerable in Tennessee, Imperiled in Alabama, and Critically Imperiled in Texas. It is Presumed Extirpated in Louisiana.

DISTRIBUTION

The prairie vole ranges from Alberta, Canada south throughout the Great Plains and upper Midwest south to New Mexico and east to extreme western West Virginia (Figure 2). In the South, the species is at the southern and eastern limits of its range. Prairie voles occur throughout all but the mountainous portions of southeastern Kentucky (Barbour and Davis 1974, McPeck et al. 1983, Davis and Kalisz 1992, Kalisz and Davis 1992, Kiser and Meade 1993), most of central and northwestern Tennessee (Goodpaster and Hoffmeister 1952, Dimmick 1969, Severinghaus and Beasley 1973, Kennedy 1991), and south into

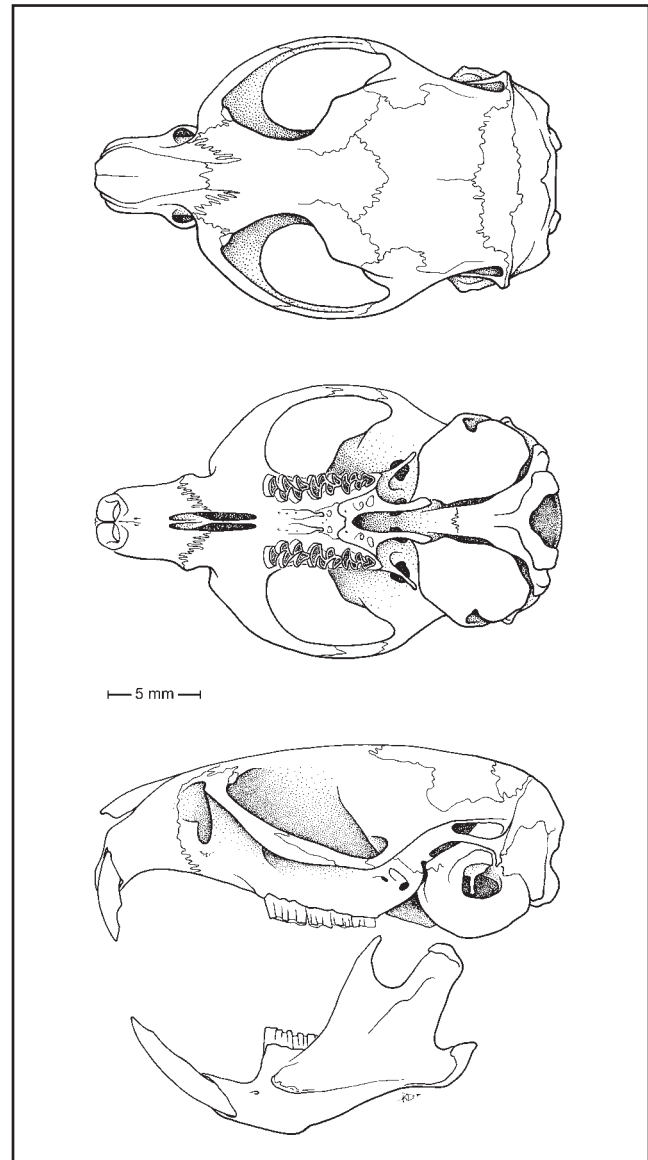


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Microtus ochrogaster* from Alexander County, Illinois (USNM 159613, female).

north-central Alabama (Whitaker and Zimmermann 1968, Allen and Dusi 1980). It ranges west through northern Arkansas (Sealander and Heidt 1990) into northeastern Oklahoma (Choate and Williams 1978, Caire et al. 1989). A disjunct race, *M. o. ludovicianus*, from Calcasieu Parrish, Louisiana and Hardin County,

Texas (Bailey 1900, Bailey 1905) is considered extinct (Lowery 1974, Schmidly 1983).

ABUNDANCE STATUS

The prairie vole is a common to abundant species in grassland habitats within its distribution. Populations fluctuate dramatically on an annual basis with highest densities reported from April to June and October and November (Gaines and Rose 1976, Taitt and Krebs 1985, Getz et al. 1987). Additionally, it exhibits large population fluctuations, which peak between 2–4 years (Hamilton 1937, French et al. 1976, Getz et al. 2001, Stalling 1990). These multi-annual population fluctuations may be related to food availability with high cycle densities as great as 1000/ha (Crawford 1971, Taitt and Krebs 1985).

PRIMARY HABITATS

The prairie vole inhabits a diversity of grassland habitats including tall grass prairie communities, abandoned fields and pastures, cultivated fields, fencerows, railroad rights-of-way, roadside corridors, and lawns—wherever grass is sufficiently thick for nesting cover and runway construction (Getz 1985, Stalling 1990). Where sympatric with meadow voles, (*M. pennsylvanicus*), prairie voles often are restricted to the more sparsely vegetated sites (Getz 1985). However, in areas where they coexist with cotton rats, (*Sigmodon hispidus*), the opposite pattern of selection is observed, as prairie voles choose dense grass cover and micro-topographic relief (Stokes 1995). Power-line and transportation corridors are believed to be an important means of dispersal that have provided an opportunity for range expansion of the species range, especially at the periphery (Getz et al. 1978, Sealander and Heidt 1990). It rarely, if ever, is found in woodland habitats.

REPRODUCTION

Breeding occurs year-round, though activity is highest in summer and lowest in winter (Keller 1985). Gestation is 20–23 days and mean litter size is approximately 3.5 (Nadeau 1985). Young are weaned within 3 weeks and individuals are reproductively active at 6–7 weeks (Richmond and Conaway 1969). Longevity in the wild is approximately 2–3 years (Fisher 1945, Martin 1956).

FOOD HABITS

The diet is composed of seeds, stems, and roots of a wide variety of plants (Zimmerman 1965, Fish 1974, Fleharty and Olsen 1969a,b; Batzli and Cole 1979, Batzli 1985). Arthropods make up a significant part

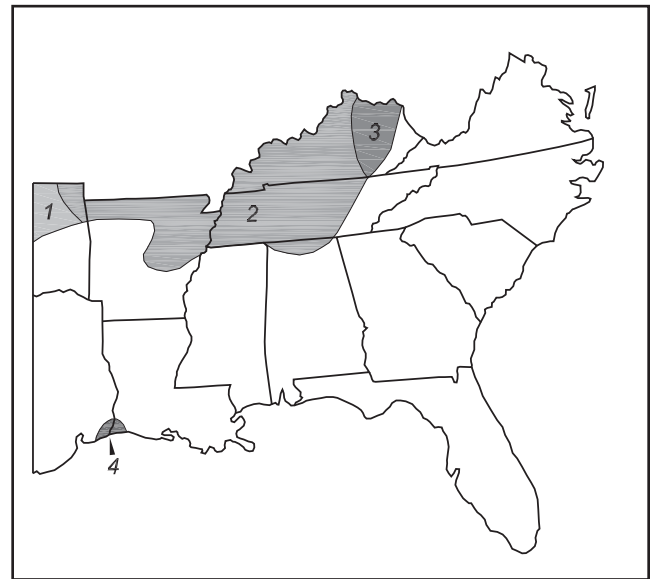


Figure 2. Distribution of *Microtus ochrogaster* in the South: (1) *M. o. haydenii*; (2) *M. o. ochrogaster*; (3) *M. o. ohionensis*; (4) *M. o. ludovicianus*.

of the diet when available (Fish 1974, Stalling 1990). In areas where prairie voles and meadow voles coexist, prairie voles consume significantly more monocots in early autumn than where meadow voles are absent, perhaps due to interspecific competition (Haken and Batzli 1996).

ASSOCIATED SPECIES

In the South, the prairie vole is associated with Elliot's short-tailed shrew (*Blarina hylophaga*), least shrew (*Cryptotis parva*), eastern harvest mouse (*Reithrodontomys humulis*), western harvest mouse (*R. megalotis*), white-footed mouse (*Peromyscus leucopus*), deer mouse (*P. maniculatus*), hispid cotton rat, and meadow vole. The prairie vole exhibits strong competitive interactions with hispid cotton rats, meadow voles, and possibly deer mice (Stalling 1990, Stokes 1995, Haken and Batzli 1996).

VULNERABILITY AND THREATS

On a regional basis, there appears to be no threats to prairie vole survival.

MANAGEMENT SUGGESTIONS

Maintenance of relict tall grass prairie and oldfield grassland habitats in the region would benefit this species.

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Meadow Vole

Microtus pennsylvanicus (Ord, 1815)

W. Mark Ford, Jane L. Rodrigue, and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

There are currently 27 subspecies of the meadow vole (*Microtus pennsylvanicus*) recognized (Hall 1981, Reich 1981, Hoffman and Koepl 1985, Woods et al. 1982); three occur in the South: *M. p. dukecampbelli*, *M. p. nigrans*, and *M. p. pennsylvanicus*. The literature was reviewed by Reich (1981) and Johnson and Johnson (1982).

DISTINGUISHING CHARACTERISTICS

The meadow vole is a large vole with a moderately long tail. The measurements are: total length 140–198 mm; tail 33–56 mm; hind foot 16–25 mm; ear 11–18 mm; weight 25–65 g. The pelage is yellowish chestnut to dull brown dorsally and silver-gray ventrally with a slightly bicolor tail. The dental formula is: I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). The skull is distinctive with the third upper molar having three closed triangles, the first lower molar five (sometimes six) closed triangles, and the third lower molar three transverse loops and no closed triangles. See keys for details.

CONSERVATION STATUS

The Florida salt marsh vole (*M. p. dukecampbelli*) is listed as Endangered by the U. S. Fish and Wildlife Service (U. S. Department of the Interior 1997, 2007). The meadow vole has a global rank of Secure (NatureServe 2007). The species is also considered Secure in those states where it occurs within the region, except for Georgia where it is listed as Vulnerable. It is unranked in Florida and South Carolina; however, South Carolina monitors *M. p. pennsylvanicus* as a Species of Concern.

DISTRIBUTION

The meadow vole is the most widely-distributed North American *Microtus*, ranging from the Alaskan and trans-Canadian Arctic south across much of the northern United States (Figure 2). Regionally, the meadow vole occurs throughout Virginia from the Delmarva Peninsula and coastal islands to the Appalachians (Jackson et al. 1976, Dueser et al. 1979, Handley 1979, Carter and Merritt 1981, Cranford and Maly 1986, Cranford and Maly 1990, Rose et al. 1990, Pagels et al. 1992). It also is widely distributed in

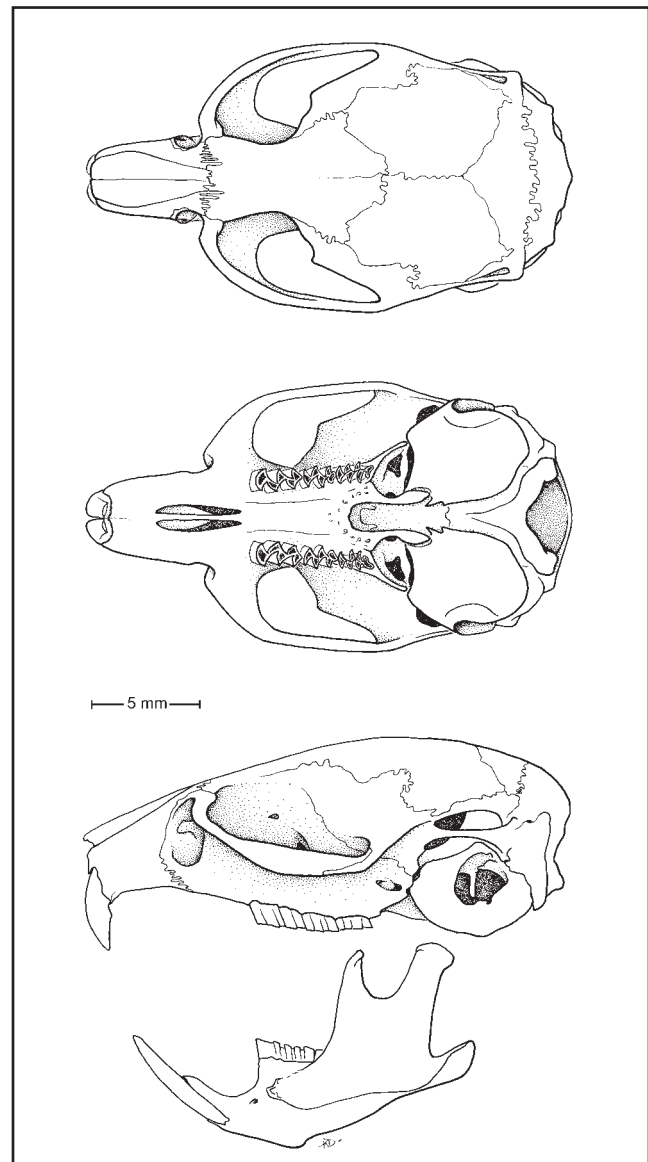


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Microtus pennsylvanicus* from Grafton County, New Hampshire (USNM 294792, female).

North Carolina, except the Coastal Plain south of the Pamlico River (Linzey and Linzey 1967, Lee et al. 1982, Clark et al. 1985, Webster et al. 1985, Adams et al. 1987). Most of Virginia and North Carolina are occupied by *M. p. pennsylvanicus*; however, specimens from southeastern Virginia and northeastern North

Carolina in and around the Great Dismal Swamp are referable to *M. p. nigrans*. In South Carolina, the meadow vole has been reported from the western Piedmont and Blue Ridge, the Upper Coastal Plain at the Savannah River Site, and from coastal areas of Charleston County including several barrier islands (Cottam and Nelson 1937, Golley 1966, Sanders 1978, Chamberlain 1979, Webster et al. 1985, Feldhamer et al. 1987, Hart 2000). Georgia populations are restricted to the upper Piedmont and scattered localities in the Blue Ridge in the northeastern part of the state (Odum 1948, Golley 1962, Laerm et al. 1982). Tennessee records occur mainly in the northeast portion of the state in the Blue Ridge and Ridge and Valley (Smith et al. 1974), although the species probably is present throughout much of eastern Tennessee along the Virginia and Kentucky borders. In Kentucky, it is present in the Bluegrass Region (Barbour and Davis 1974), and recent records indicate presence throughout the Cumberland Plateau and Mountains to the south and east (S. Thomas, National Park Service, personal communication), perhaps attributed in part to the abundance of reclaimed surface mines in the eastern coalfields. *Microtus p. dukecampbelli* was described by Woods et al. (1982) as a Pleistocene relict, isolated from other *M. pennsylvanicus* populations and restricted to high saltmarsh habitat in a single locality in Levy County, Florida (Woods et al. 1982, Smith 1990, Woods 1992).

ABUNDANCE STATUS

Regional patterns of the species distribution and abundance are complex. Meadow voles are very common in the Bluegrass of central Kentucky (Barbour and Davis 1974) and throughout much of Virginia, but often rare in the Piedmont and Upper Coastal Plain in the Carolinas and Georgia (Golley 1966, Lee et al. 1982, Webster et al. 1985). Paradoxically, it can be locally common along the Atlantic coast (Feldhamer et al. 1987). Throughout forested regions of Appalachia, it is mostly restricted to grassy roadsides, montane meadows, and wildlife openings (Menzel et al. 1999). Recent surveys for *M. p. dukecampbelli* in Florida indicate the current population is very low – only a single specimen taken in a reported 1,025 trap night survey (Woods 1992). Densities in northern United States and southern Canada range from 10–410/ha (Whitaker and Hamilton 1998). Similar to other microtines, the meadow vole is susceptible to competition with other small mammals and dramatic cyclic population fluctuations (Rose and Birney 1985, Taitt and Krebs 1985, Getz et al. 1987, Krupa and Haskins 1996, Getz et al. 2001).

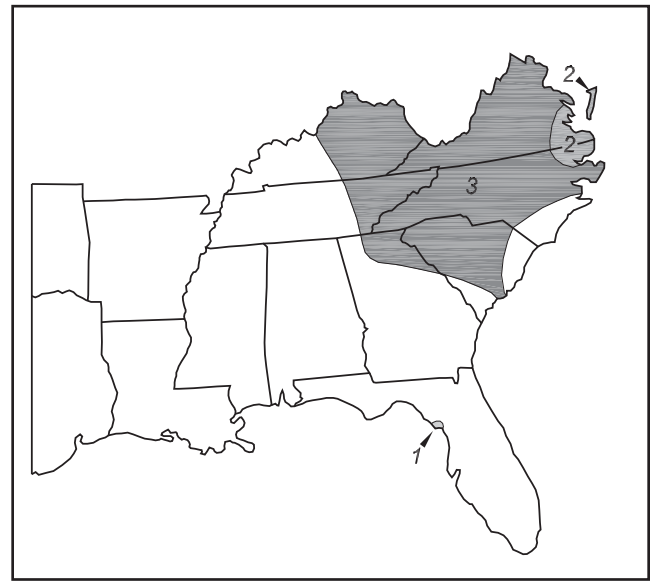


Figure 2. Distribution of *Microtus pennsylvanicus* in the South: (1) *M. p. dukecampbelli*; (2) *M. p. nigrans*; (3) *M. p. pennsylvanicus*.

PRIMARY HABITATS

The meadow vole inhabits a diversity of open habitats with low, dense vegetative cover (Getz 1985, Getz et al. 2001) including coastal and salt marshes, grassy meadows, pastures, fence rows, reclaimed surface mines, early successional seres, montane meadows, and bogs with thick grasses and sedges (Odum 1949, Harris 1953, Woods et al. 1982, Linzey and Cranford 1984, Linzey 1984, Rose 1986, Kirkland 1988, Cranford and Maly 1990, Pagels et al. 1992, Woods 1992, Kalko and Handley 1993, Franc 2003). The species typically is absent from later successional stages of forests (Pagels et al. 1992, Kalko and Handley 1993), but has been reported from isolated shrubby dominated forest clearings with a grassy understory (Getz 1985).

REPRODUCTION

The breeding season is year-round with highest activity in summer and lowest in winter (Keller 1985, Rose 1986). Gestation is 20–21 days; mean litter size ranges from 4.0–6.0 (Reich 1981, Nadeau 1985). Young are weaned 11–14 days (Nadeau 1985). Meadow voles are prolific breeders capable of multiple litters annually (Whitaker and Hamilton 1998). Longevity estimates in the field range from 2–16 months (Beer and MacLeod 1961, Hamilton 1941).

FOOD HABITS

The meadow vole feeds on a variety of plant stems, leaves, flowers, seeds, and roots, as well as fungi, insects, and occasionally carrion (Zimmerman 1965, Fish 1974, Batzli 1985). The species caches food for use in the dormant season (Whitaker and Hamilton 1998).

ASSOCIATED SPECIES

Common faunal associates of the meadow vole include the least shrew (*Cryptotis parva*), marsh rice rat (*Oryzomys palustris*), white-footed mouse (*Peromyscus leucopus*), eastern harvest mouse (*Reithrodontomys humulis*), hispid cotton rat (*Sigmodon hispidus*), prairie vole (*Microtus ochrogaster*), southern bog lemming (*Synaptomys cooperi*), and meadow jumping mouse (*Zapus hudsonius*). It apparently avoids microhabitats inhabited by the northern short-tailed shrew (*Blarina brevicauda*; Eadie 1952, Funk 1972). Competitive interactions may play a significant role in the limited distribution and patchiness of regional meadow vole populations (Rose and Birney 1985).

VULNERABILITY AND THREATS

Throughout most of its range, the meadow vole is a common component of grassland small mammal communities with few threats to its long-term viability. Conversely, isolated populations such as those in coastal South Carolina (which may represent an undescribed subspecies) and those in Florida may be highly vulnerable. Given the dramatic population fluctuations that characterize microtines in general, the possibility for competitive exclusion by other rodent species, the inundation of coastal areas by storms, and habitat destruction due to development, these populations are beset with a myriad of serious threats (Woods 1992, U. S. Fish and Wildlife Service 1997).

MANAGEMENT SUGGESTIONS

Preventing woody succession through the maintenance of grassy habitats such as wildlife openings, relict prairies, powerline, railroad and highway rights-of-way, and pastures by mowing, grazing, or herbicide control are beneficial to the meadow vole.

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Woodland Vole

Microtus pinetorum (LeConte, 1830)

W. Mark Ford, Jane L. Rodrigue, and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Currently, seven subspecies of the woodland vole (*Microtus pinetorum*) are recognized (Smolen 1981), six of which occur in the South: *M. p. auricularis*, *M. p. carbonarius*, *M. p. nemoralis*, *M. p. parvulus*, *M. p. pinetorum*, and *M. p. scalopsoides*. Van der Meulen (1978) and Repenning (1983) regarded *M. p. nemoralis* and *M. p. parvulus* as distinct species, although Whitaker and Hamilton (1998) suggest that all subspecies occurring in the South are referable to *M. p. pinetorum*. *Pitymys* was a recent generic synonym (Van der Meulan 1978, Zakrzewski 1985). Smolen (1981) reviewed the literature on this species. The woodland vole also is commonly referred to as the pine vole (Whitaker and Hamilton 1998).

DISTINGUISHING CHARACTERISTICS

The woodland vole is a small vole with a short tail and well developed forelimbs and claws that reflect semi-fossorial habits. The measurements are: total length 97–145 mm; tail 17–25 mm; hind foot 13–20 mm; ear 8–13 mm; weight 18–35 g. The dental formula is: I 1 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). The pelage is smooth and silky, reddish to chestnut brown dorsally, and buffy to silvery gray ventrally. The tail is short (less than 20% body length) and slightly bicolor. The woodland vole skull is distinctive, with the third upper molar having two closed triangles, and the first lower molar three closed triangles and two anterior loops. See keys for details.

CONSERVATION STATUS

Microtus pinetorum has a global rank of Secure (NatureServe 2007). It is also considered Secure in Alabama, Georgia, Kentucky, Mississippi, North Carolina, Oklahoma, Tennessee, and Virginia. Louisiana and Arkansas list it as Apparently Secure, and Texas lists it as Vulnerable. It is unranked in South Carolina and Florida.

DISTRIBUTION

The woodland vole is distributed across most of eastern United States and southern Canada. It is present throughout the South (Figure 2) including all of Virginia (Handley and Patton 1947, Handley 1979, Rose et al. 1990, Handley 1992, Pagels et al. 1992), Kentucky

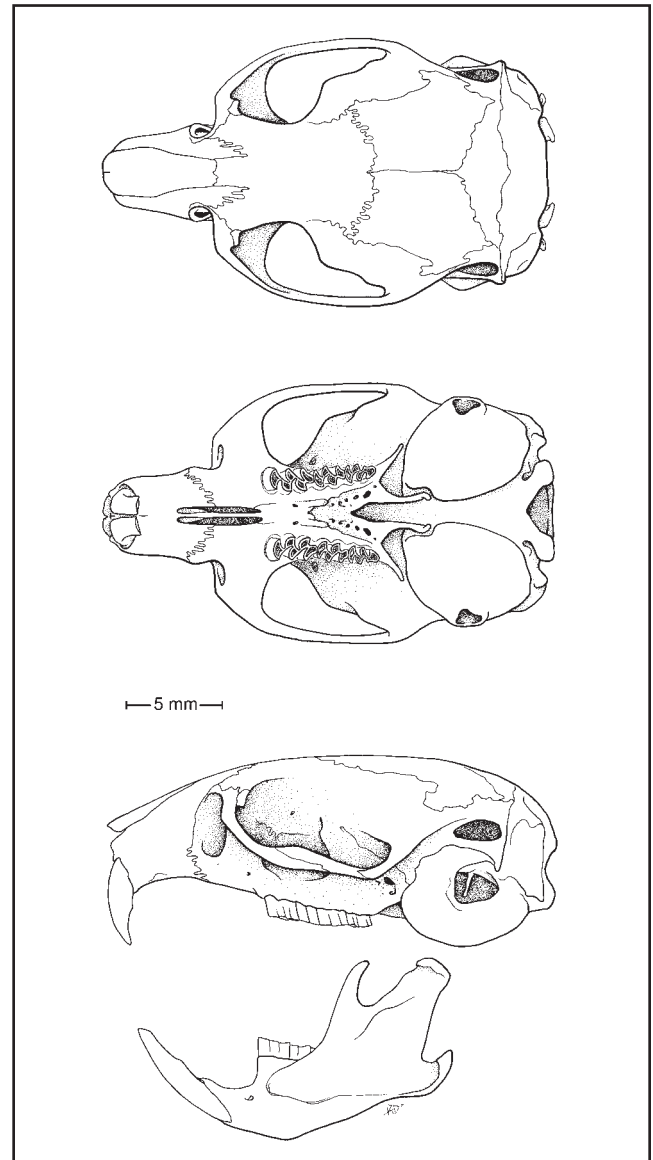


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Microtus pinetorum* from Autauga County, Alabama (USNM 222589, male).

(Barbour and Davis 1974, McPeck et al. 1983, McGehee-Marsh et al. 1992, Kiser and Meade 1993), Tennessee (Smith et al. 1974, Kennedy 1991, Linzey 1995), North Carolina (Lee et al. 1982, Clark et al. 1985), South Carolina (Golley 1966, Cothran et al. 1991), Arkansas (Sealander and Heidt 1990), and eastern Oklahoma (Caire et al. 1989, Haner et al. 1999). In Georgia, the

species occurs statewide except for the extreme southeastern coast (Golley 1962, Laerm et al. 1982, Ford et al. 1994). Apart from the coastal regions along the Gulf of Mexico, the woodland vole also occurs throughout Alabama (Holliman 1963, Wolfe and Rogers 1969, Linzey 1970), Mississippi (Kennedy et al. 1974, Jones and Carter 1989), Louisiana (Lowery 1974, Williams et al. 1980, Mullin and Williams 1987) and eastern Texas (Davis and Schmidly 1994). In Florida, the species is restricted to the northern portions of the panhandle and north-central peninsular area as far south as the Ocala area (Neill and Boyles 1955, Arata 1965, Whitaker and Hamilton 1998).

ABUNDANCE STATUS

The species is common to abundant throughout the region. Density estimates vary by season and habitat, and have been reported as high as 14/ha (Fitzgerald and Madison 1983). The species undergoes cyclic population fluctuations, usually with peak numbers in spring and fall and fewer in summer, primarily due to reproductive output (Paul 1970). Typically, woodland voles are underreported in most small mammal surveys in the region, in part due to their semi-fossorial nature and difficulty of capture (Bellows et al. 2001).

PRIMARY HABITATS

The woodland vole occurs in a diversity of woodland and grassland habitats throughout the South, but deciduous forest sites with mesic, well-drained soils and a dense ground cover of litter or vegetation are preferred (Smolen 1981, Miller and Getz 1969, Getz 1985, Rhoades and Richmond 1985, Linzey 1995, Haner et al. 1999). The species spends much of its time below the litter and humus layer where it burrows and nests. There are records of woodland voles at elevations >1,300 m in the Southern Appalachian Mountains. However, generally the species is found from 600 m in the mountains to near sea level on the Atlantic Coastal Plain (Linzey 1995, Ford et al. 2000, Bellows et al. 2001). It also is commonly associated with upland grassy areas, fencerows, railroad rights-of-way, cropland, and orchards. Woodland voles frequently can cause economic damage by root-girdling fruit trees and consuming crops (Eadie 1954, Anthony and Fisher 1977, Smolen 1981, Whitaker and Hamilton 1998).

REPRODUCTION

Regional populations probably breed throughout the year; however, some studies suggest most activity is concentrated from March through October. Farther north, reproductive activity may decline or cease by

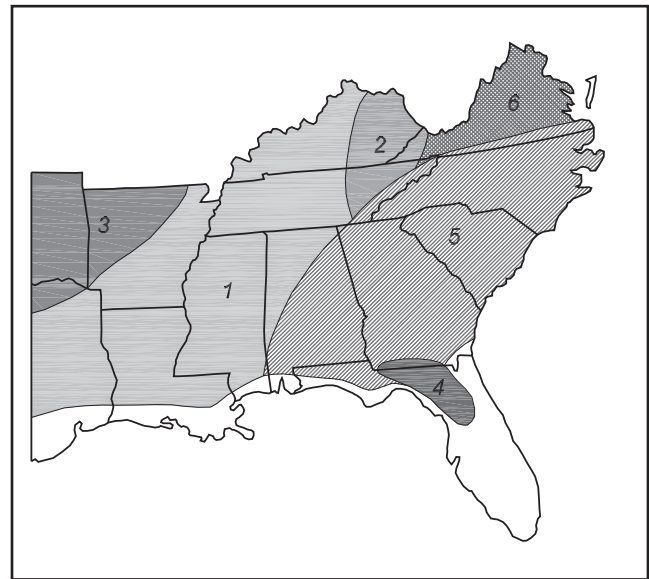


Figure 2. Distribution of *Microtus pinetorum* in the South: (1) *M. p. auricularis*; (2) *M. p. carbonarius*; (3) *M. p. nemoralis*; (4) *M. p. parvulus*; (5) *M. p. pinetorum*; (6) *M. p. scalopsoides*.

early winter (Benton 1955). Upwards of 4 litters of 1–6 neonates are produced annually. Gestation is 20–24 days and young are weaned at 17–21 days. Individuals are sexually mature by 6–8 weeks (Horsfall 1963, Paul 1970, Valentine and Kirkpatrick 1970, Goertz 1971, Cengel et al. 1978, Schadler and Butterstein 1979).

FOOD HABITS

The diet of the woodland vole varies seasonally. Forb and grass roots predominate in spring and summer, whereas seeds and fruit are consumed in fall, and bark and roots are utilized in winter (Benton 1955, Cengel et al. 1978). Habitat also effects diet as woodland voles in orchards and grassland habitats feed primarily on green vegetation and those in woodlands eat greater seed amounts. *Endogone* fungus and insects also have been recorded as food items (Linzey 1995, Whitaker and Hamilton 1998).

ASSOCIATED SPECIES

Regionally, common faunal associates include the northern short-tailed shrew (*Blarina brevicauda*), southern short-tailed shrew (*B. carolinensis*), least shrew (*Cryptotis parva*), pygmy shrew (*Sorex hoyi*), southeastern shrew (*S. longirostris*), cotton mouse (*Peromyscus gossypinus*), white-footed mouse (*P. leucopus*), eastern harvest mouse (*Reithrodontomys humulis*), hispid cotton rat (*Sigmodon hispidus*), meadow jumping mouse (*Zapus hudsonius*), meadow vole (*M. pennsylvanicus*), and prairie vole (*M. ochrogaster*).

VULNERABILITY AND THREATS

Nowhere within its distribution does the woodland vole appear to be vulnerable or threatened. Locally, it often is regarded as a serious pest species for agriculture, horticulture, and silviculture that requires control (Eadie 1954, Anthony and Fisher 1977, Smolen 1981).

MANAGEMENT SUGGESTIONS

In areas where woodland voles are causing economic damage, population control or cultural management may be necessary. Grass and weed control around young fruit trees can prevent damage by woodland voles during the growing season. Additionally, stem enclosures constructed of hardware cloth extending below the ground line can protect young trees during the winter months. Toxic baits have been used in areas where high vole populations are causing extensive damage (Fisher and Hygnstrom 2003).

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House Mouse

Mus musculus (Linnaeus, 1758)

Joshua Laerm and Wm. David Webster

CONTEXT AND TAXONOMIC COMMENTS

The house mouse is one of three commensal murids introduced into North America during early European colonization. Subspecies designations (Schwarz and Schwarz 1943) have little practical value because of the historic mixing of populations throughout North America. We follow Jones et al. (1997) in the use of *M. musculus*, following Bonhomme (1986), who relegated *M. m. domesticus* to subspecies (see Musser and Carleton 1993 for additional references relating to taxonomy). There is a considerable amount of literature concerning this species, most of it relating to its relationship to humans, its impact as a pest species, and its control (see Brooks 1973, Marsh and Howard 1977, Pratt et al. 1977, Jackson 1982, 1990; Meehan 1984, Lund 1994, Timm 1994).

DISTINGUISHING CHARACTERISTICS

The house mouse is a small mouse with a slender, scantily haired, scaled tail equal to or greater than the head and body length. External measurements are: total length, 140–190 mm; tail, 75–100 mm; hind foot, 16–21 mm; ear, 11–18 mm; weight, 18–23 g. Its pelage is typically grayish brown to pale brown above, grading to buffy gray below, although white, blonde, dark brown, black, and piebald individuals also occur. The dental formula is I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). The upper molars have cusps arranged in three longitudinal rows, superorbital ridges are absent, and the incisors are notched in lateral view. See keys for details.

CONSERVATION STATUS

The house mouse has a global rank of Secure (NatureServe 2007). A conservation status rank of Not Applicable has been assigned for each state in the region where it occurs; the species is not a suitable target for conservation activities.

DISTRIBUTION

This ubiquitous commensal occurs throughout the South, both in association with humans and related structures, and as feral populations (Handley and Patton 1947, Howell and Conaway 1952, Schwartz 1952, Conaway and Howell 1953, Golley 1962, 1966; Holliman 1963, Linzey and Linzey 1971, Wolfe 1971,

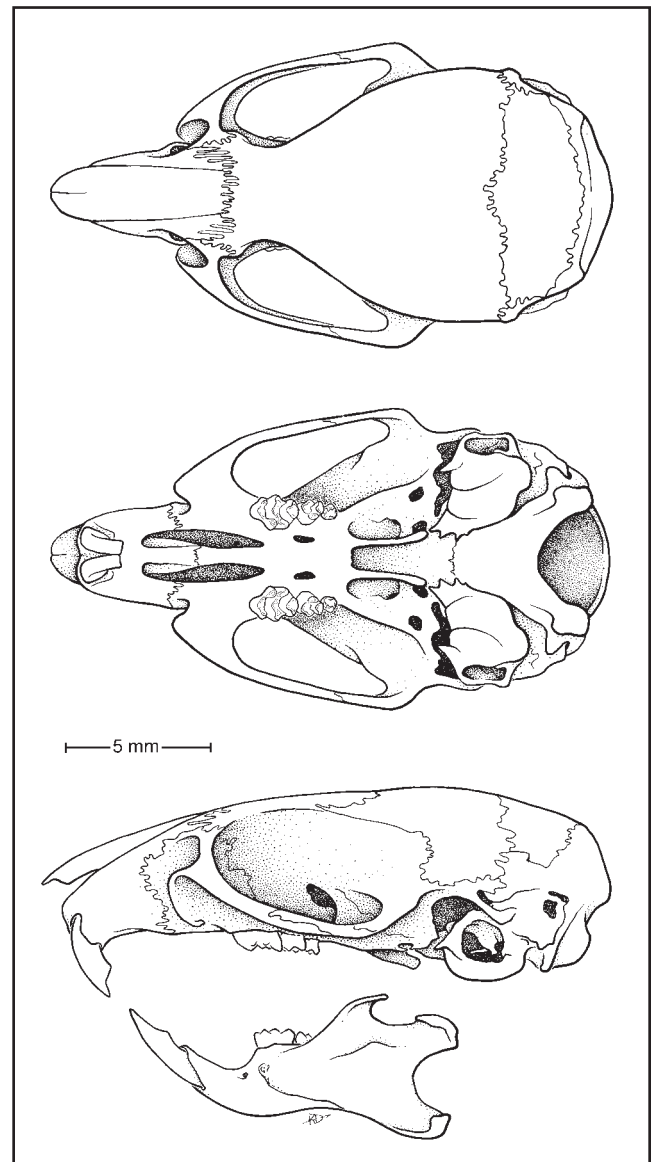


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Mus musculus* from Djakarta, Indonesia (USNM 522001, female).

Barbour and Davis 1974, Kennedy et al. 1974, Layne 1974, Lowery 1974, Neuhauser and Baker 1974, Smith et al. 1974, Sanders 1978, Andre 1981, Laerm et al. 1982, Lee et al. 1982, Schmidly 1983, Webster et al. 1985, Webster 1988, Caire et al. 1989, Jones and Carter 1989, Sealander and Heidt 1990, Kennedy 1991, McGehee-Marsh et al. 1992, Davis and Schmidly 1994, Linzey 1998; Figure 2).

ABUNDANCE STATUS

Feral house mouse populations vary considerably in size depending upon the availability of food and appropriate habitat. Density estimates range from 8–10/ha (Brown 1953, Howell 1954, Caldwell 1964, Lowery 1974, Cranford and Maly 1990, Cothran et al. 1991) to 750/ha (Pearson 1963, Lidicker 1966). House mice have the capacity to reach phenomenally high densities as evidenced in 1926 and 1927 in Kern County, California, when populations reached 175,000/ha (Hall 1927).

PRIMARY HABITATS

Mus musculus is a common rodent most typically associated with humans and their structures, particularly in agricultural areas. Feral populations are common in grain fields, early stages of oldfield succession, fencerows, abandoned fields, and dunes and associated grasslands on barrier islands (Gentry 1966). It occasionally inhabits young pine plantations and woodlands (Mitchell et al. 1995). Nests are made of shredded materials and located in trash piles, under logs, and in the burrows and nests of other animals.

REPRODUCTION

Breeding in feral populations tends to be bimodal, with highest activity in the spring and fall months. Commensal populations in favorable habitats will breed throughout the year. Gestation is 18–21 days and litter size ranges from 1–8. Up to 12 litters may be produced each year. Young are weaned by 3 weeks and reach sexually maturity as early as 5 weeks. Reproductive data are in Laurie (1946), Smith (1954), Pearson (1963), Brooks (1973), Marsh and Howard (1977), Pratt et al. (1977), Jackson (1982, 1990), Meechan (1984), Hayassen et al. (1993), and Lund (1994).

FOOD HABITS

The diet is highly variable. House mice will feed on essentially any human food or that of their livestock and companion animals. In addition, *Mus* feeds on a variety of seeds, plant material, insects, and other animal matter.

ASSOCIATED SPECIES

The house mouse is commonly associated with other small mammals that occupy early successional habitats, including a variety of shrews. These include the least shrew (*Cryptotis parva*), southeastern shrew (*Sorex longirostris*), several *Blarina* species, as well as the marsh rice rat (*Oryzomys palustris*), hispid cotton rat (*Sigmodon hispidus*), meadow jumping mouse

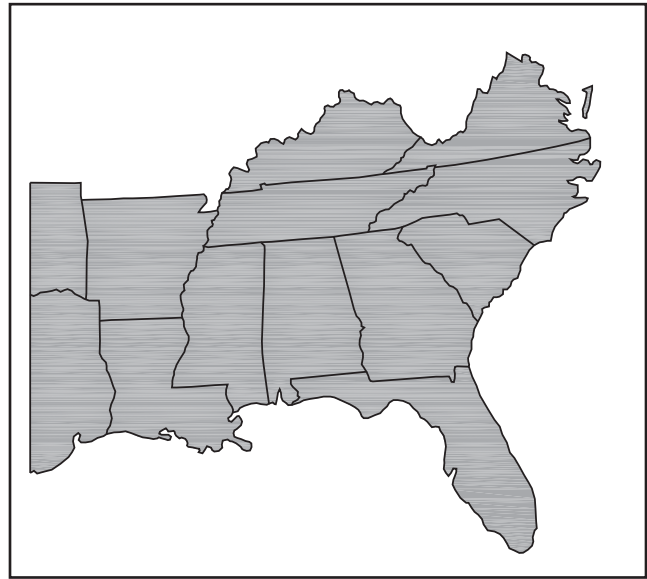


Figure 2. Distribution of *Mus musculus* in the South.

(*Zapus hudsonius*), *Reithrodontomys* spp., *Peromyscus* spp., and *Microtus* spp.

VULNERABILITY AND THREATS

This species is widespread and abundant in both commensal situations and natural habitats. It is one of the most significant pest species, causing wide ranging negative health and agricultural impacts.

MANAGEMENT SUGGESTIONS

As an exotic, this species requires scant management, except as a potential reservoir for diseases that negatively affects humans.

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Round-tailed Muskrat

Neofiber alleni (True, 1884)

Steven B. Castleberry and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Schwartz (1953) recognized five subspecies of the round-tailed muskrat (*Neofiber alleni*): *N. a. alleni*, *N. a. apalachicola*, *N. a. exoristus*, *N. a. nigrescens*, and *N. a. struix*. Although Burt (1954) questioned the validity of the subspecies designations stating they were arbitrarily based on small differences in coloration, body proportions, and size, the subspecies designations of Schwartz (1953) generally are recognized (Birkenholz 1972, Perry 1982, Lefebvre 1992). The literature was reviewed by Birkenholz (1972), Perry (1982), and Boutin and Birkenholz (1987).

DISTINGUISHING CHARACTERISTICS

The dense underfur is gray at the base, grading into brown at the tips on the dorsum and buffy at the tips on the venter. The guard hairs are dark brown and glossy. Stiff fringe hairs are present on the margins of the hind feet. The round-tailed muskrat resembles the common muskrat (*Ondatra zibethicus*) except it is smaller and has a round, sparsely-haired tail. Measurements are: total length, 285–381 mm; tail, 99–168 mm; hind foot, 40–50 mm; ear, 15–22 mm; weight, 187–357 g. Males are slightly heavier than females. The skull is similar to the common muskrat, but smaller (length less than 50 mm) and with non-rooted (ever growing) cheek teeth. The dental formula is $I\ 1/1, C\ 0/0, P\ 0/0, M\ 3/3 = 16$. See keys for details.

CONSERVATION STATUS

The round-tailed muskrat has a global rank of Vulnerable (NatureServe 2007). Florida and Georgia also consider it Vulnerable.

DISTRIBUTION

The distribution of the round-tailed muskrat is restricted to Florida and southeastern Georgia. In Florida, it ranges from the Everglades north throughout the state, as far west as the Choctawhatchee River in the western panhandle (Paul 1967, Birkenholz 1972, Wassmer and Wolfe 1983). Throughout the distribution in Florida it occurs in patchy, localized populations in favorable habitat. Populations are not documented from the northwestern peninsula or the extreme northeastern counties (Lefebvre 1992). In Georgia, the primary distribution is in the

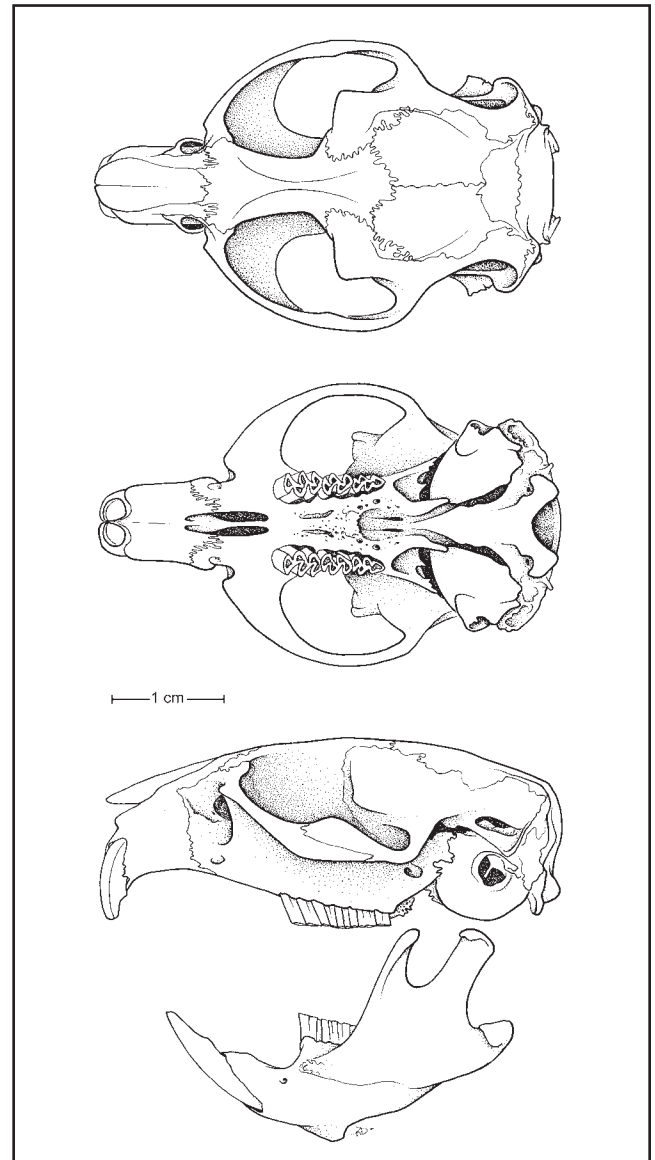


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Neofiber alleni* from Franklin County, Florida (USNM 263703, male).

Okefenokee Swamp (Golley 1962, Laerm 1981), but it is also found west of the Okefenokee from smaller, isolated wetland systems (Bergstrom et al. 2000). Skull remains from barn owl (*Tyto alba*) pellets are documented from Camden county, east of the Okefenokee (Schantz and Jenkins 1950), but populations there have not been documented.

ABUNDANCE STATUS

The round-tailed muskrat can be extremely abundant in quality habitat. Density estimates in north-central Florida ranged from 250–300 individuals/ha (Birkenholz 1963). In the northern extreme of the distribution in the Okefenokee Swamp in Georgia, density estimates were up to 150/ha in some areas (Laerm 1981). Birkenholz (1963) reported that the highest densities occur from December through February. Local populations can fluctuate greatly over short periods of time depending on water levels and habitat suitability (Birkenholz 1963, Lefebvre 1992). Displacement by flooding and drought has a greater impact on abundance than predation (Birkenholz 1963).

PRIMARY HABITATS

The round-tailed muskrat typically inhabits shallow freshwater marshes with dense emergent vegetation (Birkenholz 1963, Lefebvre 1992), but also is reported from brackish marsh (Ehrhart 1984, Lefebvre 1992). The highest densities occur in marshes dominated by maidencane (*Panicum hemitomon*), pickerelweed (*Pontederia cordata*), and rice cut-grass (*Leersia hexandra*; Birkenholz 1963, Wassmer and Wolfe 1983). Other plant species associated with round-tailed muskrat habitat include blue-tongue arrow-head (*Sagittaria lancifolia*), pipewort (*Eriocaulon* spp.), Virginia chainfern (*Woodwardia virginica*), dog fennel (*Eupatorium* spp.), redroot (*Lachnanthes caroliniana*), and yellow-eyed grass (*Xyris ambigua*; Birkenholz 1963, Wassmer and Wolfe 1983, Smith and Franz 1994, Bergstrom et al. 2000). Floating mats of *Sphagnum* covered by herbaceous and woody plants are important habitat components in small, isolated wetlands in southern Georgia (Bergstrom et al. 2000). Birkenholz (1963) found that preferred plant communities in a north-central Florida marsh were best developed at water depths of 15–46 cm. The stable water levels and deep muck soils associated with commercial sugarcane (*Saccharum officinarum*) fields provide suitable habitat (Lefebvre 1992). Round-tailed muskrats may burrow into banks or under marsh bottom substrate when water levels are below the soil surface (Birkenholz 1963, Lefebvre 1992, Bergstrom et al. 2000).

Spherical or dome-shaped houses, 17–60 cm diameter, are constructed of tightly-woven plant material, typically in water <1 m deep (Golley 1962, Birkenholz 1963, Smith and Franz 1994). Houses may be constructed on decaying vegetation at water level or attached to emergent plants just above water level. Females raise the floor of the house higher above the water prior to parturition (Birkenholz 1963). There

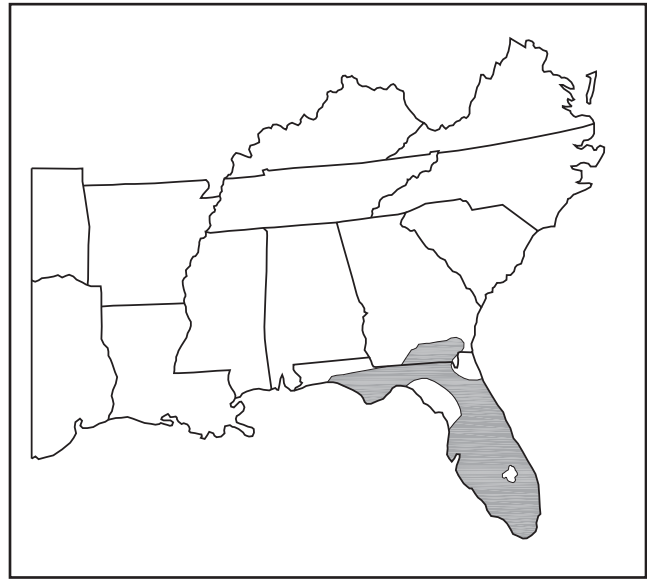


Figure 2. Distribution of *Neofiber alleni* in the South.

typically are two exit holes (plunge holes) in the house floor. Round-tailed muskrats also construct feeding platforms of vegetation just above water level in the vicinity of their houses (Birkenholz 1963, 1972). Feeding platforms may be covered when emergent vegetation is sparse.

REPRODUCTION

Breeding occurs year-round with most young born between October and January (Birkenholz 1963). Although there are no seasonal breeding cycles, periods of increased reproduction are related to increased water levels and cover of emergent vegetation. Gestation is 26–29 days and litter size is 1–4 with reported means ranging from 1.8 in South Florida (Lefebvre 1982) to 2.3 in north-central Florida (Birkenholz 1963). Based on placental scar counts, Birkenholz (1963) suggested that females may produce 4–5 litters/year under appropriate environmental conditions. Young are weaned by 21 days and both sexes become sexually mature at 90–100 days (Birkenholz 1963).

FOOD HABITS

The round-tailed muskrat is herbivorous, feeding primarily on the roots, stems, and seeds of aquatic and semiaquatic vegetation. In north-central Florida, Birkenholz (1963) reported maidencane as the primary food consumed, being found in stomach contents year round. Other important food plants include pickerelweed, rice cut-grass, blue-tongue arrow-head, and watershield (*Brasenia schreberi*). In sugarcane fields, they feed on the sprouts, stalks, and roots of sugarcane, as well as sedges (*Cyperus* spp.) and

grasses (*Panicum* spp.; Lefebvre 1982). Birkenholz (1963) found no evidence of animal matter in an examination of the stomach contents of 330 round-tailed muskrats. Most feeding activity occurs just after dark and within 10 m of the currently occupied house (Birkenholz 1963).

ASSOCIATED SPECIES

Marsh rice rats (*Oryzomys palustris*), hispid cotton rats (*Sigmodon hispidus*), and cotton mice (*Peromyscus gossypinus*) frequently use abandoned round-tailed muskrat houses and tunnels (Birkenholz 1963, Bergstrom et al. 2000). Remains of marsh rice rat feeding activities are commonly found on round-tailed muskrat feeding platforms (Birkenholz 1963). A variety of amphibians and reptiles use round-tailed muskrat houses for protection, feeding, and basking (Smith and Franz 1994). Bergstrom et al. (2000) suggest that the greater sandhill crane (*Grus canadensis tabida*) is important in maintaining round-tailed muskrat habitat by promoting and maintaining early successional marsh. Important predators include American alligator (*Alligator mississippiensis*; Kinsella 1982, Bergstrom et al. 2000), barn owl (*Tyto alba*; Schantz and Jenkins 1950, David 1996), northern harrier (*Circus cyaneus*), and cottonmouth (*Agkistrodon piscivorus*; Birkenholz 1963). Other reported predators include great blue heron (*Ardea herodias*; Ehrhart 1984), red-shouldered hawk (*Buteo lineatus*; Bergstrom et al. 2000), red-tailed hawk (*Buteo jamaicensis*), bobcat (*Lynx rufus*; Birkenholz 1963), and feral cat (*Felis catus*; Harper 1927). Most predation occurs on individuals displaced by flooding (Birkenholz 1963).

VULNERABILITY AND THREATS

Although round-tailed muskrat populations can be drastically reduced by natural processes such as drought and flooding (Birkenholz 1963), human activities represent the greatest threat to the species. The major threats to populations are destruction of wetland habitat by draining and filling and alteration of natural hydrologic regimes (Lefebvre 1992, Bergstrom et al. 2000). Fragmentation of suitable habitat isolates populations, preventing dispersal and subsequent recolonization of extirpated wetlands. Winter prescribed burning of wetlands may reduce populations by reducing emergent cover, increasing susceptibility to predation (Lefebvre 1992). Suppression of natural summer fires which prevent encroachment of woody plants and perpetuate the "floating-mat" habitat may decrease habitat quality (Bergstrom et al. 2000). Alteration of natural fire and hydrologic regimes allows colonization of wetlands by red imported fire ants (*Solenopsis invicta*) that may

negatively affect round-tailed muskrat populations (Bergstrom et al. 2000).

MANAGEMENT SUGGESTIONS

Because of its specific habitat requirements, existing suitable wetland habitat should be protected and natural fire and hydrologic processes maintained (Lefebvre 1992). Habitat conditions in degraded wetlands can be improved by restoring natural fire and hydrologic regimes (Bergstrom et al. 2000). Recent reintroductions of Florida sandhill cranes (*G. c. pratensis*) into southern Georgia wetland systems may enhance habitat conditions for the species (Bergstrom et al. 2000).

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Eastern Woodrat

Neotoma floridana (Ord, 1818)

Michael T. Mengak and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

The taxonomic relationship of eastern woodrats west of the Mississippi River has been examined by Birney (1973) and those to the east by Hayes and Harrison (1992) and Hayes and Richmond (1993). Eight subspecies of *N. floridana* are currently recognized, six of which occur regionally. The Allegheny woodrat, *N. magister* is regarded as a distinct species (Hayes and Harrison 1992, Hayes and Richmond 1993, Edwards and Bradley 2001). However, patterns of genetic variation in *N. floridana* do not support current subspecies taxonomy and their present distribution. In particular, Hayes and Harrison (1992:342) note that *N. f. haematoreia* "...as currently recognized is not at all distinct from *N. f. floridana*, and probably does not represent a valid taxonomic entity". However, see Schwartz and Odum (1957). Ray (2000) examined the phylogenetics of woodrats in the Southern Appalachian Mountains of North Carolina.

Lazell (1987) suggested that *N. f. smalli* was sufficiently distinct morphologically to warrant species recognition, a notion for which Humphrey (1988, 1992) and Hayes and Harrison (1992) found little justification. The latter regard *N. f. smalli* as distinct at the subspecies level. Additional studies are required to define the taxonomic relationships of remaining populations. *Neotoma f. osagensis* is regarded a synonym of *N. f. attwateri* (Birney 1973, Wiley 1980), despite Hall (1981), Caire et al. (1989) and Sealander and Heidt (1990). *N. f. illinoensis* and *N. f. rubida* are also reported in the South. The literature is reviewed by Wiley (1980).

DISTINGUISHING CHARACTERISTICS

The eastern woodrat is a large rodent with long vibrissae, large ears and eyes, and a moderately haired tail. Total length is 310–490 mm; tail, 140–207 mm; hind foot, 28–44 mm; ear, 24–30 mm; weight, 220–360 g. Males are slightly larger than females. Pelage is soft, upper parts are brown to brownish gray tinged with black-tipped hairs, sides light brown grading into white undersides; tail long, sparsely haired and sharply bicolored, dark above, white below; feet whitish. Semiprismatic molars characterize *Neotoma*, with cusps elongated into transverse lophs forming one medial and two lateral entrant angles resembling the letter "E." *Neotoma floridana* is

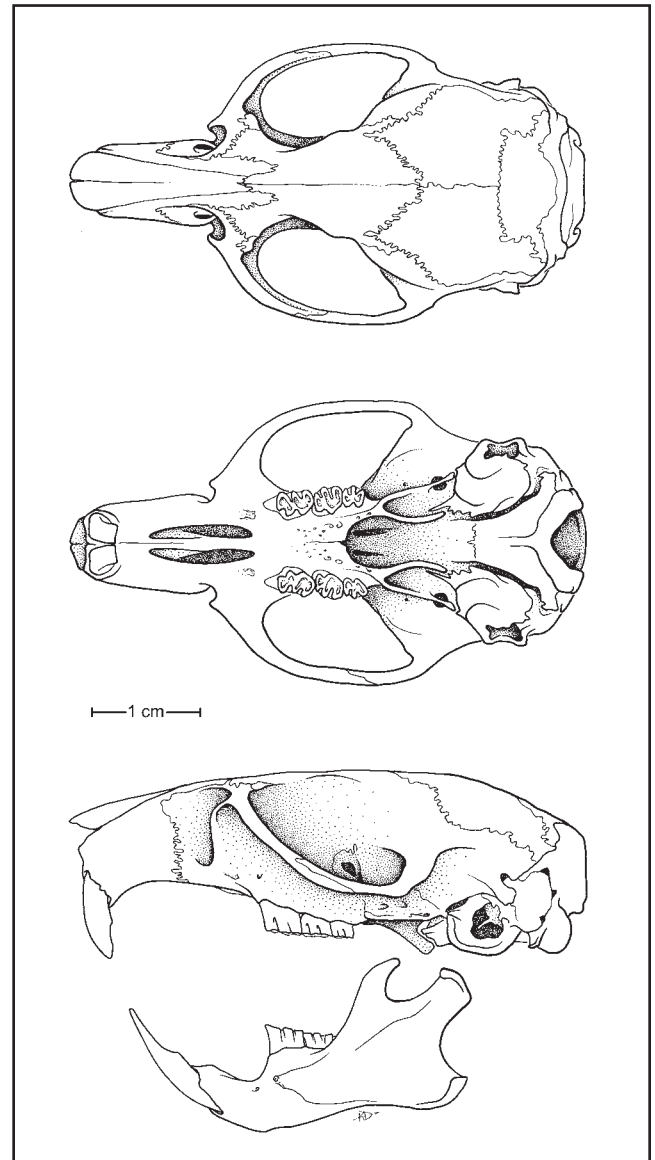


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Neotoma floridana* from Madison Parrish, Louisiana (USNM 132256, male).

distinguished from the Allegheny woodrat by the lack of a maxillo-vomerine notch in the former and usually having a bifurcated anterior palatal spine. The dental formula is $I\ 1/1, C\ 0/0, P\ 0/0, M\ 3/3 = 16$ (Figure 1). See keys for details.

CONSERVATION STATUS

The eastern woodrat has a global rank of Secure (NatureServe 2007). It is also considered Secure in Florida, Georgia, Oklahoma, and Texas. It is Apparently Secure in Alabama, Arkansas, and Louisiana. It is considered Vulnerable in North Carolina and South Carolina, and unranked in Mississippi and Tennessee. *Neotoma f. smalli* is listed as Endangered by the U. S. Fish and Wildlife Service (1993, 2007). The U. S. Fish and Wildlife Service (1991) considered *Neotoma f. haematoreia* a Category 2 taxon prior to elimination of sub-category designators. *Neotoma f. floridana* is listed as Threatened by the North Carolina Natural Heritage Program. *Neotoma f. illinoensis* is monitored by the Tennessee Department of Environment and Conservation as a Species of Special Concern.

DISTRIBUTION

Distribution records are reported by Schwartz and Odum (1957), Birney (1973), and Hayes and Richmond (1993). The eastern woodrat occurs in two disjunct regions of North Carolina, the extreme southeastern and southwestern portions (Howell 1933, Lee et al. 1982, Adams 1987, Webster et al. 1987). In South Carolina it is restricted to the Coastal Plain and three Blue Ridge counties (Chamberlain 1928, Coleman 1949, Golley et al. 1965, Golley 1966, Schacher and Pelton 1979, Cothran et al. 1991) and the Coastal Plain, Blue Ridge, Ridge and Valley, and Cumberland Plateau of Georgia (Howell 1933, Golley 1962, Neuhauser and Baker 1974, Laerm 1981). Contrary to maps of Wiley (1980) and Hall (1981), woodrats are not known from the Piedmont of Georgia (Laerm 1981). Populations extend into south-central Florida (Layne 1974, Greer 1978, Humphrey 1988, 1992) but are disjunct from those referred to *N. f. smalli*, which is restricted to Key Largo (Hersch 1981, Barbour and Humphrey 1982, Lazell 1989, Humphrey 1992). The Tennessee River represents the southern limit of the range of *N. magister* (Hayes and Richmond 1993) and populations to the east and south of the river in eastern Tennessee, including the Great Smoky Mountains (Holliman 1963, Linzey and Linzey 1968, 1971), south and west of the river in Alabama (Zamernardi 1956, Linzey 1970, Hayes and Richmond 1993) and western Tennessee (Goodpaster and Hoffmeister 1952, Beasley and Severinghaus 1973, Kennedy and Harvey 1980, Kennedy 1991) are referred to as *N. floridana* (Hayes and Richmond 1993). We have modified the subspecies distribution map from Hall (1981) but retain his subspecies designations. However, the validity of *N. f. haematoria* as a subspecies has been questioned by Hayes and Harrison (1992). To the west, the species occurs throughout Mississippi (Kennedy et al. 1974,

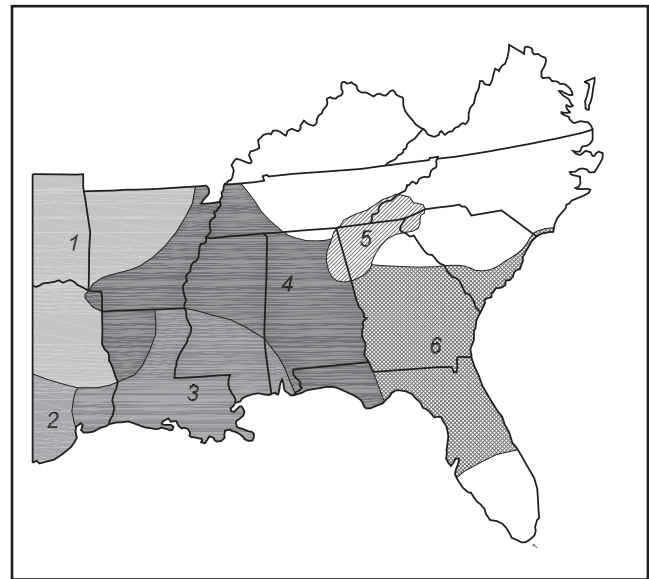


Figure 2. Distribution of *Neotoma floridana* in the South: (1) *N. f. osagensis*; (2) *N. f. attwateri*; (3) *N. f. rubida*; (4) *N. f. illinoensis*; (5) *N. f. haematoreia*; (6) *N. f. floridana*.

Jones and Carter 1989) but there is disagreement about the line of separation between subspecies (Wiley 1980, Jones and Carter 1989). Two subspecies are recognized in Louisiana (Neal 1967, Lowrey 1974), Arkansas (Sealander and Heidt 1990, Tumlinson et al. 1992) and Texas (Birney 1973, Schmidly 1983). In Oklahoma, the subspecies is *N. f. attwateri* (Goertz 1970, Birney 1973, Caire et al. 1989).

ABUNDANCE STATUS

The status of the species varies considerably throughout its range. It is rare to uncommon in the Carolinas (Lee et al. 1982, Webster et al. 1985, Adams 1987, Webster et al. 1987) and Georgia (Golley 1962, Laerm 1981). It is regarded as poorly known and apparently rare in western Tennessee (Beasley and Severinghaus 1973, Kennedy 1991). It is considered relatively common in Louisiana (Lowrey 1974), Arkansas (Sealander and Heidt 1990), Texas (Schmidly 1983) and Oklahoma (Caire et al. 1989). Woodrat populations are cyclic (Fitch and Rainey 1956, Rainey 1956). Density estimates are reported at 0.2–0.8/ha in Mississippi River hardwood bottoms (Neal 1967), 0–5/ha in Texas woodlands (Schmidly 1983), 4.0/ha in eastern Kansas (Fitch and Rainey 1956) and 2.2–12.2/ha on Key Largo in Florida (Hersch 1981, Humphrey 1988, 1992).

PRIMARY HABITATS

In the South, woodrat habitat is quite variable. In the Appalachian region of the Carolinas, they occur in

rock crevices, overhangs, abandoned buildings, and dense brush (Howell 1933, Coleman 1949, Golley 1962, 1966; Adams 1987). In the Coastal Plain they are associated with hardwood bottoms and swamps (Howell 1921, Harper 1927, Chamberlain 1928, Pearson 1952, Neal 1967, Webster et al. 1987), hammocks (Pearson 1952), marshes (Svihla and Svihla 1933), and mature, dry tropical forest (Hersch 1981, Humphrey 1992). They are widely distributed in upland hardwood forests (Caire et al. 1989, Sealander and Heidt 1990, Cothran et al. 1991). Woodrats in the central United States construct stick houses that shelter a nest (Rainey 1956, Birney 1973). However, this behavior is variable in southeastern populations (Hayes and Richmond 1993) and the propensity to do so may vary with microhabitat differences (Humphrey 1988). Oak (*Quercus* spp.) and other hardwoods provide mast but woodrats are also found in early successional habitat in certain parts of the range. Shrubs and hedgerows provide cover as do caves and rock outcrops.

REPRODUCTION

The breeding season varies geographically. In most areas, breeding occurs from early spring through late fall (Rainey 1956, Wiley 1980), although populations in northern Florida and coastal Georgia are reported to breed year-round (Pearson 1952, Golley 1962). Gestation is 32–39 days (Pearson 1952). Two litters are produced per year with 1–4 young/litter (Rainey 1956, Goertz 1970). Based on seven laboratory litters, mean litter size was 3.0 (Pearson 1952). Young are weaned in 4 weeks. Early born individuals may breed their first year but most do so in their second year (Fitch and Rainey 1956, Rainey 1956). Maximum longevity in the wild is 3–4 years (Fitch and Rainey 1956).

FOOD HABITS

Woodrats are primarily herbivorous with a generalist diet that varies with respect to region and availability (Wiley 1980). The species consumes acorns, leaves, bark, seeds, and small amounts of animal matter including bats and northern bobwhite quail (*Colinus virginianus*; Strecker 1929, Goodpaster and Hoffmeister 1952, Rainey 1956, Finley 1958, Neal 1967, Wiley 1980, Williams et al. 2000).

ASSOCIATED SPECIES

The eastern woodrat commonly occurs in habitats occupied by northern short-tailed shrew (*Blarina brevicauda*), southern short-tailed shrew (*B. carolinensis*), Elliott's short-tailed shrew (*B. hylophaga*), smoky shrew (*S. fumeus*), southeastern shrew (*S. longirostris*), least shrew (*Cryptotis parva*) and rodents such as the

marsh rice rat (*Oryzomys palustris*), white-footed mouse (*Peromyscus leucopus*), deer mouse (*P. maniculatus*), and cotton mouse (*P. gossypinus*). Predators include the bobcat (*Lynx rufus*), feral cat (*Felis catus*), snakes, and owls.

VULNERABILITY AND THREATS

The status of the eastern woodrat is uncertain. In the central United States, the species is widespread and apparently abundant. However, in the South local populations such as those in southeastern North Carolina (Adams 1987, Webster et al. 1987) and Key Largo, Florida (Humphrey 1988, 1992) are severely threatened. Other regional populations (such as those referred to as *N. f. haematoreia* or those in western Tennessee) are too poorly known to assess status (Kennedy and Harvey 1980, Laerm 1981).

MANAGEMENT SUGGESTIONS

Eastern woodrats favor pine (*Pinus* spp.) forest with an open canopy and grass understory maintained by periodic fire, brush piles in pine regeneration areas, and natural rock outcrops and crevices. Other habitats include hedgerows and dense understory of bottomlands. Judicious use of prescribed fire may enhance some habitats; the retention of mast-producing trees is also beneficial. Further study of the ecology of the species is warranted.

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Allegheny Woodrat

Neotoma magister (Baird, 1857)

Steven B. Castleberry and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

The Allegheny woodrat was first described as *Neotoma magister* by Baird (1857), but was later relegated to a subspecies of the eastern woodrat (*N. floridana*) by Schwartz and Odum (1957). Recent studies of mitochondrial DNA variation (Hayes and Harrison 1992, Planz et al. 1996, Edwards and Bradley 2001), cranial morphology (Hayes and Richmond 1993), and chromosome structure (Ray 2000) indicate that the Allegheny woodrat is a distinct species. The literature was reviewed by Castleberry et al. (2006).

DISTINGUISHING CHARACTERISTICS

Generally, the dorsal pelage is gray to brownish-gray, typically with more brown coloration present in adults. The entire ventral surface and the feet are white. The tail is long, moderately haired, and distinctly bicolored. The Allegheny woodrat has longer vibrissae than other woodrats, an adaptation for maneuvering in dark, tight crevices within rocky habitats (Ray 2000). Measurements reported by Ray (2000) are: total length, 311–451 mm; tail, 147–210 mm; hind foot, 35–46 mm; ear, 23–34 mm; weight, 203–444 g. Externally, the Allegheny woodrat and the eastern woodrat are difficult to separate. Although the Allegheny woodrat is generally larger and has longer vibrissae, identification in the field is not reliable because of overlap in size measurements (Ray 2000). Reliable identification can only be made based on genetic and chromosome differences (Ray 2000) and skull morphology, namely presence of a maxillovomerine notch (Hayes and Richmond 1993). The dental formula is I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). See keys for details.

CONSERVATION STATUS

The Allegheny woodrat has a global rank of Vulnerable (NatureServe 2007). It is considered Apparently Secure in Kentucky, Vulnerable in Alabama, Tennessee, and Virginia, and Imperiled in North Carolina.

DISTRIBUTION

The Allegheny woodrat generally ranges throughout the Appalachian and Interior Highland regions of the eastern United States (Figure 2). Regionally it is distributed throughout western Virginia (Mengak 2002a)

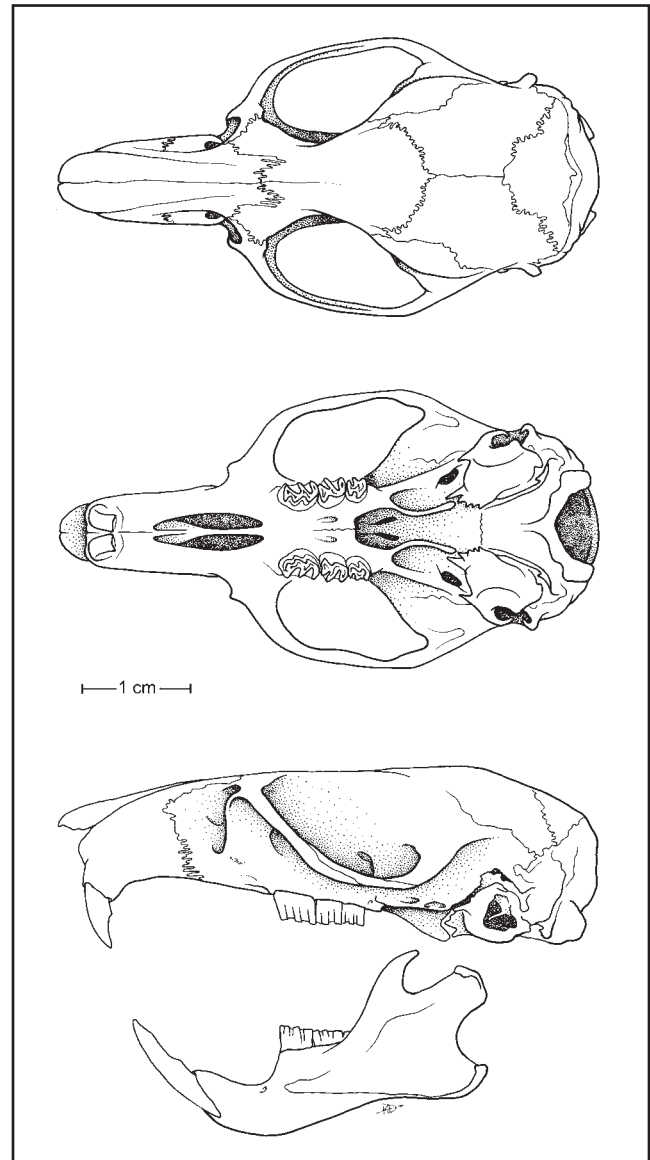


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Neotoma magister* from Colbert County, Alabama (USNM 207198, female).

southward down the Blue Ridge Mountains into North Carolina where it generally occurs at elevations above 640 m to its southern extent in Burke, McDowell, Buncombe, and Madison (northeast of the French Broad River) counties (Ray 2000). Woodrats that occur in southwestern North Carolina and adjacent regions of southeastern Tennessee east of the

Tennessee River, including Great Smoky Mountains National Park, are the Appalachian woodrat (*N. floridana haematoria*; Linzey and Linzey 1968, Ray 2000). Woodrat populations in northeastern Tennessee (Smith et al. 1974) and in central Tennessee (Kennedy and Harvey 1980) bordered on the east and west by the Tennessee River are presumed to be the Allegheny woodrat, although little work has been done in the area. In Alabama, it is known only from areas north of the Tennessee River, with the exception of 3 specimens collected south of the river near Muscle Shoals, Colbert County that were identified as Allegheny woodrats based on presence of the maxillo-vomerine notch (Hayes and Richmond 1993). The species is thought to occur throughout Kentucky east of the Tennessee River (Barbour and Davis 1974).

ABUNDANCE STATUS

Although populations in the northern and western peripheries of the distribution have experienced dramatic declines in recent years (Ford et al. 2006, LoGuidice 2006), the Allegheny woodrat is thought to be common in appropriate habitats in the South. Ray (2000) found the species to be relatively abundant in western North Carolina. In Tennessee, Kennedy and Harvey (1980) indicate that the Allegheny woodrat is commonly associated with cave entrances in the limestone regions of central Tennessee. In Kentucky, they are reportedly abundant in sandstone cliffs throughout the Cumberland Plateau, but are less abundant in the Highland Rim and Bluegrass regions of the state (Barbour and Davis 1974, Fassler 1974, Bommarito 1999). Population estimates for 2 sites in western Virginia studied intensively over an 11-year period ranged from 0–24.1 individuals with long-term averages of 10.6–6.3 (Mengak 2002a).

PRIMARY HABITATS

The Allegheny woodrat occurs in wide variety of vegetative associations including northern hardwood, red spruce-eastern hemlock (*Picea rubens-Tsuga canadensis*), mixed mesophytic, and mixed-oak pine (*Quercus-Pinus*) forest types (Fassler 1974, Castleberry et al. 2002a, Mengak et al. 2002). However, the presence of caves, cliff faces, boulder fields, or talus slopes typically is a prerequisite for occurrence regardless of forest type (Newcombe 1930, Poole 1940). Meyers (1997) found that rock outcrops occupied by woodrats in north-central West Virginia occurred on steeper slopes, were wider, had less leaf litter accumulation, and had fewer understory trees than unoccupied rock outcrops. In eastern Kentucky, occupied rock outcrops occurred on steeper slopes that had higher tree densities (Bommarito 1999). Allegheny woodrats select foraging areas that contain a diversity of

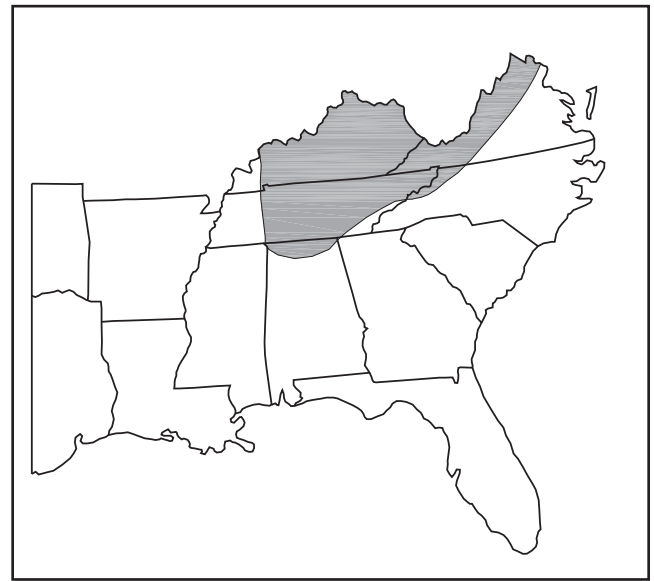


Figure 2. Distribution of *Neotoma magister* in the South.

understory vegetation (Castleberry et al. 2002b). Evidence of Allegheny woodrat presence at rock outcrops includes middens and latrines (Newcombe 1930, Poole 1940). Middens are conspicuous piles of sticks, green vegetation, and other debris located underneath overhanging rock ledges and other sheltered areas. Latrines tend to be located in the open on clean, flat rock surfaces, often with an overhanging ledge.

REPRODUCTION

Poole (1940) suggested that Allegheny woodrats in Pennsylvania have 2–3 litters/year between mid-March and October. In Virginia, they apparently are capable of breeding year-round, but with most young born between May and October (Mengak 2002b). Woodrats in Harlan County, Kentucky, were in reproductive condition in July and August (Barbour 1951). The gestation period is estimated to be between 30–36 days (Poole 1940). Typical litter sizes range from 1–4 with reported averages of 2.0 (Poole 1940) and 2.3 (Mengak 2002b). The young are reared in nests constructed of shredded wood fibers, bark, roots, and grasses (Newcombe 1930, Poole 1940) which typically are located inaccessibly deep in crevices or on ledges in caves. Age at sexual maturity is unknown, but is probably 3–4 months (Poole 1940). Individuals are known to give birth at 10 months. Allegheny woodrats are known to live up to about 48 months in captivity (Poole 1940) and up to 58 months in the wild (Mengak et al. 2002).

FOOD HABITS

Based on items found around nests and middens and from examination of stomach contents of collected specimens in Pennsylvania, Allegheny woodrats eat various parts of hemlock, black birch (*Betula lenta*), American chestnut (*Castanea dentata*), scrub oak (*Quercus ilicifolia*), flowering dogwood (*Cornus florida*), apple (*Malus pumila*), black cherry (*Prunus serotina*), mountain ash (*Sorbus americana*), and rhododendron (*Rhododendron maximum*; Poole 1940). Castleberry et al. (2002a) documented food habits of Allegheny woodrats from the Allegheny Plateau and Ridge and Valley in West Virginia and the adjacent Ridge and Valley in Virginia using microhistological analysis of fecal pellets. The most common food items consumed in an oak-pine (*Quercus-Pinus*) dominated forest in the Ridge and Valley were blackberry leaves (*Rubus* spp.), fungi, greenbrier (*Smilax* spp.) leaves, acorns, and oak leaves. In mixed oak-northern hardwood and red spruce-hemlock forests in the Allegheny Plateau the most common foods were fungi, acorns, holly (*Ilex* spp.), blueberry (*Vaccinium* spp.), fern (*Dryopteris* spp.), and lichen. They noted that acorn consumption reflected annual and seasonal availability, and that acorns were an important component of the diet in both regions. Although no regional food habits information is available, the diet is probably similar in the southern Appalachians.

ASSOCIATED SPECIES

The Allegheny woodrat commonly occurs in habitats occupied by the northern short-tailed shrew (*Blarina brevicauda*), masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), rock shrew (*S. dispar*), white-footed mouse (*Peromyscus leucopus*), deer mouse (*P. maniculatus*), rock vole (*Microtus chrotorrhinus*), and red-backed vole (*Clethrionomys gapperi*; Ford and Rodrigue 2001, Castleberry et al. 2003). Potential predators of Allegheny woodrats include owls, foxes, skunks, bobcats, weasels, and snakes (Poole 1940, Balcom and Yahner 1996). Northern raccoons (*Procyon lotor*) host the ascarid nematode *Baylisascaris procyonis*, which causes fatal neurological disease in aberrant hosts and has been implicated in Allegheny woodrat population declines in the northern parts of the range (LoGiudice 2001).

VULNERABILITY AND THREATS

Although the Allegheny woodrat is apparently abundant regionally, declines in western and northern areas have prompted conservation concerns throughout the range. The causes of its extirpation from Connecticut and New York, and declines in New Jersey, Pennsylvania, Maryland, Indiana, and Ohio, are still

not fully understood, but may be related to *Baylisascaris procyonis* parasitism, changes in forest composition and structure, forest fragmentation, and loss of hard mast resulting from declines in forest health (Balcom and Yahner 1996, Castleberry et al. 2001, Ford et al. 2006, LoGiudice 2006). Although Ray (2000) found the species to be relatively abundant in western North Carolina, he cautioned that because those populations are on the periphery of the range they are vulnerable to extirpation. Mengak (2002a) stated that although woodrats have an extensive distribution in Virginia, they occur in isolated locations with few individuals/site, and populations can decline to zero in a short period of time.

MANAGEMENT SUGGESTIONS

Despite their relatively narrow habitat requirements, Allegheny woodrats can tolerate a wide variety of forest overstory conditions (Castleberry et al. 2002b). Forest management practices have minimal direct impact on Allegheny woodrats if harvests are planned to retain the area immediately surrounding rock outcrops and on at least one adjacent side of rock outcrops in intact forest (Castleberry et al. 2001). If planned properly, clearcutting and deferment harvesting in the proximity of outcrops can benefit woodrats by providing abundant soft mast and succulent vegetation consumed by woodrats in spring and summer (Castleberry et al. 2002a). In general, management for the species should provide a mix of habitat conditions with abundant understory vegetation and consistent hard and soft mast production.

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Golden Mouse

Ochrotomys nuttalli (Harlan, 1832)

Steven B. Castleberry and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

The genus *Ochrotomys* was long considered a subgenus of the genus *Peromyscus* (Osgood 1909, Miller and Kellogg 1955, Hall 1981). However, based on baculum morphology (Hooper 1958, Hooper and Musser 1964), absence of an entepicondylar foramen on the humerus (Manville 1961), and chromosome structure (Patton and Hsu 1967), *Ochrotomys* is now recognized as a distinct genus containing a single species. Packard's (1969) taxonomic revision of the golden mouse recognized five subspecies: *O. n. aureolus*, *O. n. flammeus*, *O. n. floridanus*, *O. n. lisae*, and *O. n. nuttalli*, all of which occur regionally. The literature is reviewed by Linzey and Packard (1977).

DISTINGUISHING CHARACTERISTICS

The name *Ochrotomys* is derived from the Greek words *ochros*, meaning "pale" and *mys*, meaning "mouse" (Stangl et al. 1993), referring to the pale yellow to golden dorsal coloration. The feet and underparts generally are creamy white, but often tinged with pale yellow. The semi-prehensile tail is slightly bicolored. The pelage is soft and dense. Measurements are: total length, 127–180 mm; tail, 50–97 mm; hind foot, 12–29 mm; ear, 11–26 mm; weight, 12–31 g. The dental formula is I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). Although similar to *Peromyscus*, the golden mouse is easily distinguished by its golden coloration. See keys for details.

CONSERVATION STATUS

The golden mouse has a global rank of Secure (NatureServe 2007). It is considered Secure in Alabama, Georgia, North Carolina, and Tennessee; Apparently Secure in Arkansas, Kentucky, Louisiana, Texas, and Virginia; and Critically Imperiled in Oklahoma. It is unranked in Florida, Mississippi, and South Carolina.

DISTRIBUTION

The golden mouse ranges from southern Virginia (Handley 1992), south throughout North Carolina (Lee et al. 1982), South Carolina (Golley 1966), and Georgia (Golley 1962) to central Florida (Frank and Layne 1992; Figure 2). It ranges west throughout Alabama (Eads and Brown 1953) and Mississippi (Jones

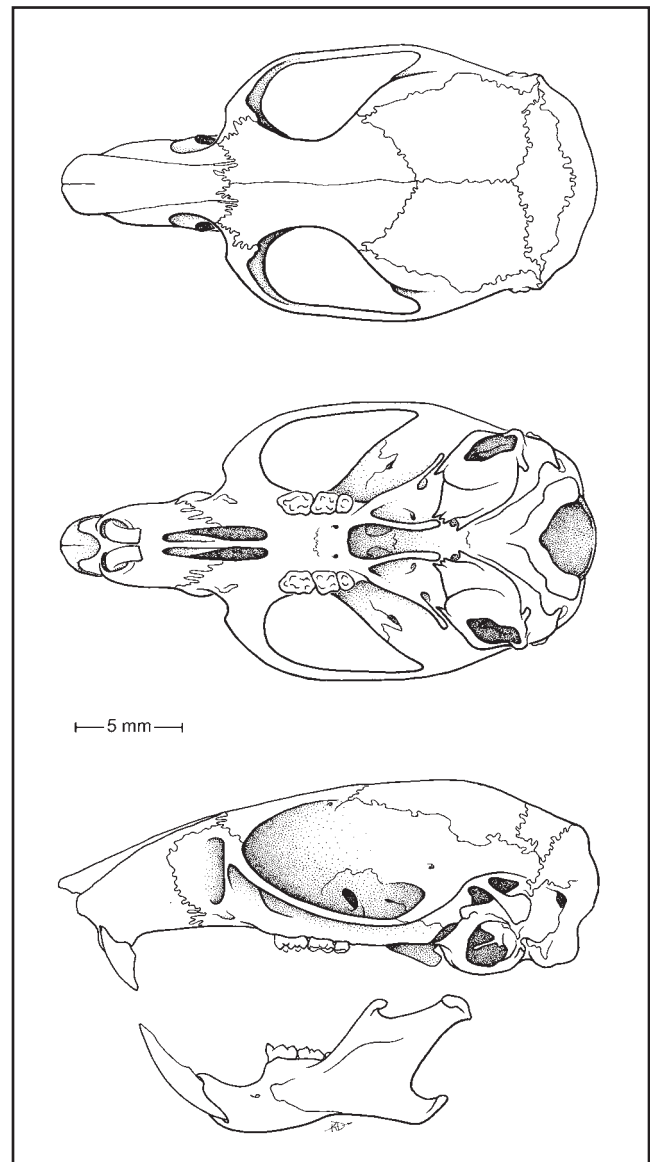


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Ochrotomys nuttalli* from Cumberland Gap National Historic Park, Kentucky (USNM 559123, male).

and Carter 1989), all but extreme southern Louisiana (Lowery 1974), and into eastern Texas (Davis and Schmidly 1994) and Oklahoma (Caire et al. 1989). To the north, the golden mouse occurs throughout Tennessee (Howell and Conaway 1952, Kennedy 1991) and into Kentucky (Barbour and Davis 1974), but is absent from the extreme northern portions of the state.

ABUNDANCE STATUS

The golden mouse is common in appropriate habitat throughout most of the region. Density estimates are 1.2/ha in oldfield habitat (Howell 1954) and 0.6–8.9/ha in mixed pine-hardwoods (Linzey 1968) in eastern Tennessee. Other estimates range from 0.7–5.4/ha in a forested hardwood floodplain in Texas (McCarley 1958), 5.4–7.1/ha in South Carolina lowland mesic-hardwood forest (Kaufman et al. 1971), and 0.5–6.9/ha in cutover loblolly-shortleaf (*Pinus taeda*-*P. echinata*) pine in Louisiana (Shadowen 1963). Blus (1966a) reported maximum density estimates from 1.7–74/ha in Illinois. Density of golden mice is directly correlated with the amount of underbrush and vines (Lowery 1974).

PRIMARY HABITATS

Linzey and Packard (1977) reviewed the considerable literature on habitat associations of the species. Although widely adaptable, golden mice generally occur in forested areas with dense understory vegetation. Common habitats include moist thickets, canebrakes, swampy woodlands, rhododendron (*Rhododendron* spp.) thickets, edges of broomsedge (*Andropogon* spp.) fields, thick timber bordering cypress (*Taxodium distichum*) swamps, rocky oak hill-sides, pine thickets, and pine-oak (*Quercus*) woodlands. High densities also have been documented from upland and bottomland deciduous forests with a dense understory of Chinese privet (*Ligustrum sinense*) and greenbrier (*Smilax* spp.; Pruett et al. 2002). Because the golden mouse is semi-arboreal, abundant undergrowth, especially dense tangles of vines, appear to be the controlling factor in habitat selection (Goodpaster and Hoffmeister 1954, McCarley 1958, Linzey 1968, Lowery 1974, Davis and Smith 1978, Mengak and Guynn 2003).

REPRODUCTION

The breeding season of the golden mouse is variable, depending on location. In Louisiana, breeding occurs year round but is concentrated in late autumn and early winter (Lowery 1974). In Texas, breeding begins in September and continues through winter and spring with little activity in summer (McCarley 1958). In Florida, breeding occurs at least 8 months of the year with reported peaks in early summer and autumn (Layne 1960). In eastern Tennessee, breeding apparently peaks in early spring and late summer and ceases in winter (Linzey and Linzey 1967). Gestation is variable, lasting from 25–30 days (Linzey and Linzey 1967). Litter size ranges from 1–4 with most reported averages near 2.6 (Layne 1960, Linzey and Linzey 1967). Litter sizes tend to be larger in the

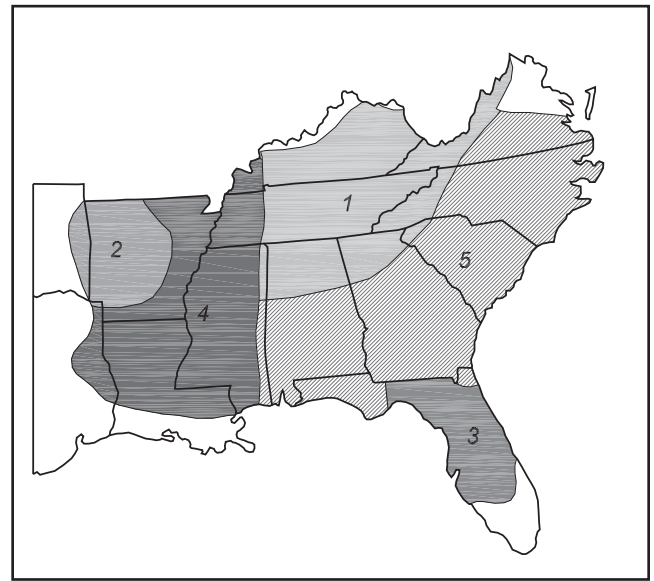


Figure 2. Distribution of *Ochrotomys nuttalli* in the South: (1) *O. n. aureolus*; (2) *O. n. flammeus*; (3) *O. n. floridanus*; (4) *O. n. lisae*; (5) *O. n. nuttalli*.

northern parts of the distribution (Blus 1966b). Young are weaned at about 17–18 days and individuals probably reach sexual maturity between 8–10 weeks (Linzey and Linzey 1967). Golden mice can produce several litters per year (Linzey and Packard 1977). The average life span ranges from about 6.5 months (McCarley 1958) to 2.5 years in the wild (Pearson 1953), although individuals are known to live 3 years or longer. Linzey (1968) documented one individual in Tennessee living 8 years and 5 months.

Golden mice construct arboreal nests used for shelter and rearing young (Goodpaster and Hoffmeister 1954, Linzey 1968). However, nests sometimes are located on the ground inside stumps or fallen logs (Goodpaster and Hoffmeister 1954) or under leaf litter (Frank and Layne 1992). Nests are globular masses with a single opening on one end constructed of available materials including leaves, twigs, shredded bark, pine needles, Spanish moss (*Tillandsia usenoides*), palmetto fibers, and grass (Linzey 1968, Frank and Layne 1992). Nests typically are 1.5–4.5 m above the ground and constructed in tangles of vines and understory shrubs (Goodpaster and Hoffmeister 1954, Blus 1966a, Linzey 1968, Wagner et al. 2000).

FOOD HABITS

Golden mice construct feeding platforms resembling nests that serve as feeding sites (Goodpaster and Hoffmeister 1954, Blus 1966a, Linzey 1968). Important food items documented from feeding platforms or stomach contents include the seeds of sumac (*Rhus* spp.), wild cherry (*Prunus* spp.), dogwood (*Cornus* spp.), blackberry (*Rubus* spp.), greenbrier, and poison ivy

(*Rhus radicans*; Goodpaster and Hoffmeister 1954, Blus 1966a, Linzey 1968). Acorns and smartweed (*Polygonum* spp.) seeds also have been documented from feeding platforms (Layne 1958, Blus 1966a). Linzey (1968) found invertebrate remains in the stomach contents of 47% to 57% of individuals examined from Tennessee. Golden mice have well-developed cheek pouches in which they transport food (Goodpaster and Hoffmeister 1954).

ASSOCIATED SPECIES

Golden mice are associated with a variety of rodents and soricids throughout the range. Some common small mammal associates include the northern short-tailed shrew (*Blarina brevicauda*), southern short-tailed shrew (*B. carolinensis*), smoky shrew (*Sorex fumeus*), white-footed mouse (*Peromyscus leucopus*), deer mouse (*P. maniculatus*), cotton mouse (*P. gossypinus*), Florida mouse (*Peromyscus floridanus*), woodland vole (*Microtus pinetorum*), and southern red-backed vole (*Clethrionomys gapperi*; Linzey 1968, Packer and Layne 1991, Frank and Layne 1992). Several studies have investigated competitive interactions, habitat segregation, and resource partitioning between golden mice and white-footed mice (Dueser and Hallett 1980, Knuth and Barrett 1984, Seagle 1985, Feldhamer and Maycroft 1992). Nests of the northern cardinal (*Cardinalis cardinalis*) and yellow-breasted chat (*Icteria virens*) are commonly used as foundations for golden mouse nests (Goodpaster and Hoffmeister 1954). Potential predators of golden mice are the common gray fox (*Urocyon cinereoargenteus*), long-tailed weasel (*Mustela frenata*), eastern spotted skunk (*Spilogale putorius*; Frank and Layne 1992), copperhead (*Agkistrodon contortrix*), black rat snake (*Elaphe obsoleta obsoleta*; Linzey 1968), hawks, and owls (Lowery 1974, Wagner et al. 2000).

VULNERABILITY AND THREATS

The species is widespread and regionally abundant. There appear to be no threats to its survival.

MANAGEMENT SUGGESTIONS

Forest management practices, such as timber harvesting and prescribed burning, that promote dense understory vegetation probably benefit the golden mouse (McCarley 1958, Blus 1966a, Wagner et al. 2000).

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Common Muskrat

Ondatra zibethicus (Linnaeus, 1766)

Steven B. Castleberry and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Sixteen subspecies of the common muskrat are recognized, four of which occur in the South: *O. z. zibethicus*, *O. z. cinnamominus*, *O. z. macrodon*, and *O. z. rivalicius* (Hall and Cockrum 1953, Perry 1982). The literature was reviewed by Willner et al. (1980), Perry (1982), and Boutin and Birkenholz (1987).

DISTINGUISHING CHARACTERISTICS

The pelage consists of dense, soft underfur overlain by long, glossy guard hairs. Color of the dorsal pelage is variable, ranging from tan to black. Ventral coloration typically is lighter. The tail and feet are dark brown to black. The eyes are small and the ears are short and rounded. Stiff fringe hairs are present on the margins of the partially webbed hind feet. The laterally compressed tail is sparsely haired and scaly. The common muskrat resembles the round-tailed muskrat except for the laterally compressed tail and a larger body size. Measurements are: total length, 406–640 mm; tail, 177–295 mm; hind foot, 63–92 mm; ear, 15–25 mm; weight, 900–1600 g. Males are slightly larger than females. The dental formula is $I\ 1/1, C\ 0/0, P\ 0/0, M\ 3/3 = 16$. See keys for details.

CONSERVATION STATUS

The common muskrat has a global rank of Secure (NatureServe 2007). The species is also considered Secure in those states where it occurs within the region except for Arkansas, Louisiana, and Oklahoma, where it is ranked as Apparently Secure. It is unranked in South Carolina. The common muskrat is regulated by all Southern state wildlife agencies as a furbearer.

DISTRIBUTION

The common muskrat is one of the most widely distributed mammals in North America, ranging from northern Alaska to the Gulf of Mexico (Willner et al. 1980). Numerous populations also exist throughout South America, Europe, Russia, and Japan as a result of introductions from fur farming operations. Although found throughout most of the South, the species is absent from some areas despite apparently suitable habitat (Dozier 1953, Wilson 1968). Within the region, the species occurs throughout Virginia, North Carolina (Lee et al. 1982, Webster et al. 1985),

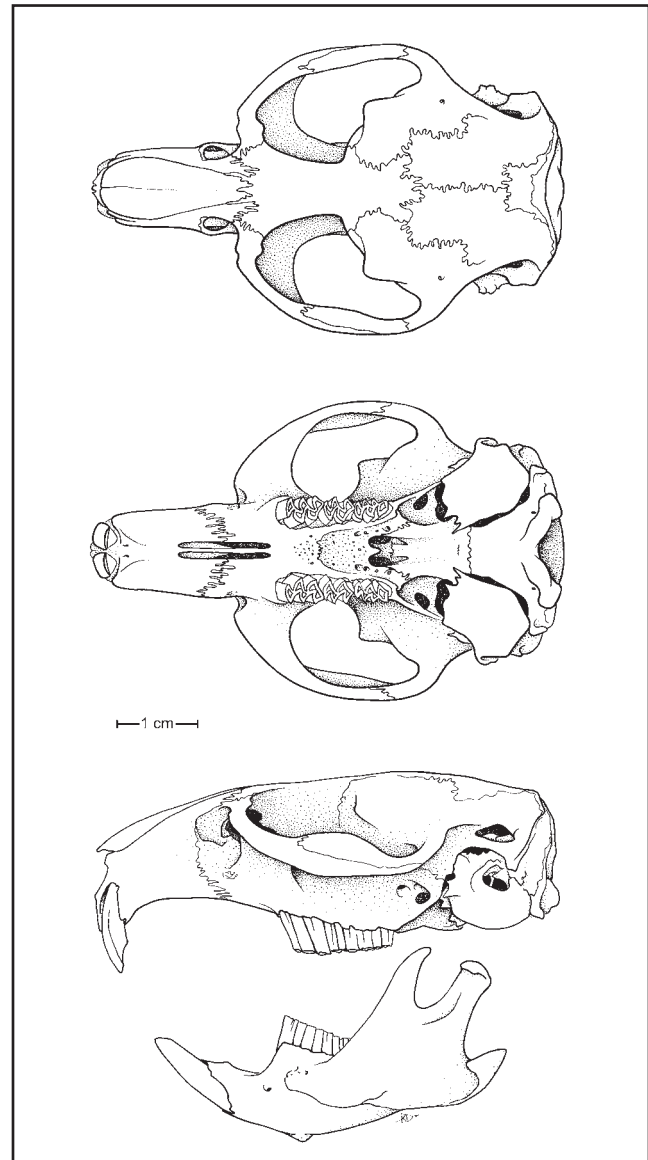


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Ondatra zibethicus* from Crawford County, Pennsylvania (USNM 265695, female).

and most of Alabama (Beshears 1951, Linzey 1970), but largely is absent from the Coastal Plain of South Carolina and Georgia (Golley 1962, 1966; Laerm et al. 1982). It apparently is absent from Florida. The range extends west into Mississippi, Louisiana, and eastern Texas (Schmidly 1983), although it is absent in parts of southern Mississippi (Kennedy et al. 1974) and

central Louisiana (Palmisano 1972, Lowery 1974). The common muskrat occurs throughout Tennessee (Schacher and Pelton 1975, Kennedy 1991), Kentucky (Barbour and Davis 1974), Arkansas (Sealand and Heidt 1990), and eastern Oklahoma (Caire et al. 1989).

ABUNDANCE STATUS

Common muskrat abundance varies considerably depending upon geographic region, habitat type and condition, water levels, harvest pressure, and disease (Lynch et al. 1947, Errington 1963, Lowery 1974, Boutin and Birkenholz 1987, Clark 1987, 1994). Certain populations throughout the distribution show 4–14 year cycles in abundance related to food availability, social interactions, disease, or predation (Lynch et al. 1947, Errington 1963, Erb et al. 2000, 2001). Population cycles in Louisiana coastal marshes are possibly related to overuse of habitat, referred to as “eat-outs,” and subsequent habitat recovery (Lynch et al. 1947, Palmisano 1972, Lowery 1974). Density estimates range widely across the distribution. The highest densities in the South are reported from coastal brackish marsh habitat (O’Neil 1949, Beshears and Haugen 1953, Palmisano 1972, Lowery 1974) where densities greater than 100/ha have been documented (Lynch et al. 1947).

PRIMARY HABITATS

Because of the wide distribution and diversity of habitats occupied, habitat relationships of the common muskrat are difficult to generalize. Essentially any aquatic habitat with relatively constant water levels and some form of aquatic or shoreline vegetation is potential habitat (Errington 1963). The most common habitats include fresh and brackish marshes, lakes, ponds, sloughs, and rivers, although man-made habitats such as ditches and canals also are inhabited. They generally are absent from large, open water bodies with significant wave action. Habitat selection at the local scale varies with population density (Messier et al. 1990, Clark 1994).

Common muskrats use either burrows (dens) excavated in shoreline banks or houses constructed of available vegetation as dwellings, depending on the local environment (Willner et al. 1980). Bank dens are variable, depending upon soil type and slope of the bank, but consist of a complex system of tunnels usually with several underwater entrances (Beshears 1951, Beshears and Haugen 1953, Errington 1963). Houses are conically-shaped and constructed of vegetation from the local area (Errington 1963, Perry 1982). Although houses are built above water level, there typically are several underwater entrance tunnels or “leads.” Common muskrats also construct

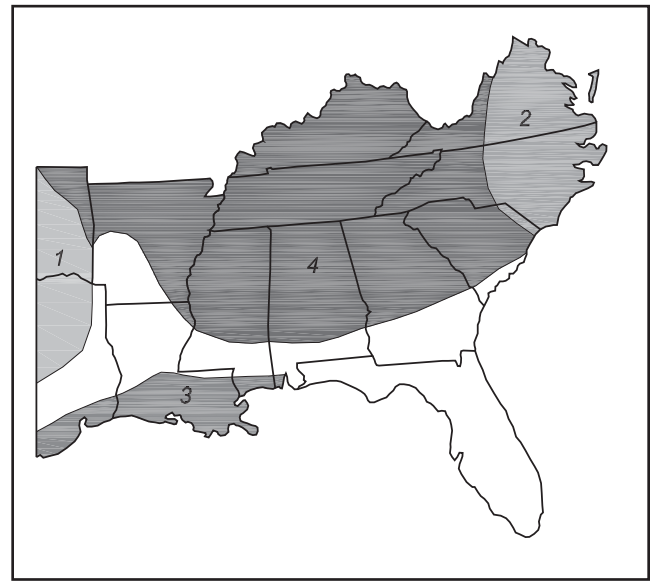


Figure 2. Distribution of *Ondatra zibethicus* in the South: (1) *O. z. cinnamominus*; (2) *O. z. macrodon*; (3) *O. z. rivalicius*; (4) *O. z. zibethicus*.

smaller feeding huts and platforms consisting of pads of vegetation slightly above water level.

REPRODUCTION

Although reproduction in northern common muskrat populations is restricted to spring and summer (Willner et al. 1980), populations in most of the South breed year-round (Svihla and Svihla 1931, Beshears 1951, Wilson 1954). However, in river and stream habitats in the southern Appalachian region of eastern Tennessee, timing of breeding activities is similar to northern populations (Schacher and Pelton 1975). Typical gestation is 29–30 days, but has been reported as short as 19 days (Errington 1937). Schacher and Pelton (1975) summarized litter sizes from across the distribution and noted that populations in the South typically have fewer young per litter, but more litters/year than northern populations. Mean litter size in the South ranges from 3.7 in coastal Louisiana (Svihla and Svihla 1931) to 5.4 in eastern Tennessee (Schacher and Pelton 1975). Weaning occurs by the fourth week, at which time the young are independent (Errington 1939). Time to sexual maturity is variable, but is generally reached within a year (Willner et al. 1980). Although common muskrats are known to live up to 4 years in the wild (Schwartz and Schwartz 1959), typical longevity is likely less than 2 years.

FOOD HABITS

The common muskrat is primarily herbivorous, feeding on the roots, bulbs, tubers, seeds, and stems of available aquatic and semi-aquatic vegetation (Svihla

and Svihla 1931, Lay and O'Neil 1942, Willner et al. 1975, Perry 1982). Specific foods vary considerably depending upon geographic location, habitat type, and plant species present. See Willner et al. (1975) and Perry (1982) for summarized lists of specific food plants from across the distribution. In coastal marshes of the South, the most important food plants are bulrushes (*Scirpus olneyi*, *S. americanus*, and *S. robustus*) and cattails (*Typha latifolia* and *T. angustifolia*; Svihla and Svihla 1931, Lynch et al. 1947, O'Neil 1949, Wilson 1968). Plants consumed in lower quantities include rushes (*Juncus effusus* and *J. roemerianus*), panic grasses (*Panicum hemitomon* and *P. virgatum*), and cordgrass (*Spartina* spp.). Animal matter, including crayfish, fish, and mollusks, is sometimes consumed, usually during times of food shortage or when one type of animal food is highly abundant (Schwartz and Schwartz 1959, Errington 1963, Willner et al. 1980).

ASSOCIATED SPECIES

A wide variety of vertebrates occupy the same habitats as common muskrats across the distribution. Many of these species use muskrat houses for resting, nesting, or basking (Svihla and Svihla 1931, Perry 1982). Although the nutria (*Myocastor coypus*), introduced into the United States from South America, has similar feeding habits (Willner et al. 1979), the presence of population-level effects resulting from competition between the species is equivocal (Willner et al. 1980, Perry 1982). Beavers (*Castor canadensis*) occupy many of the same habitats, but there is little competition because of the differences in feeding habits (Perry 1982). The mink (*Mustela vison*) generally is considered the most important predator of common muskrat, but mink predation varies among different environmental conditions (Errington 1943). Other important predators include raccoon (*Procyon lotor*), red fox (*Vulpes vulpes*), coyote (*Canis latrans*), northern harrier (*Circus cyaneus*), American alligator (*Alligator mississippiensis*), and cottonmouth (*Agkistrodon piscivorus*; Svihla and Svihla 1931, Errington and Scott 1945, Lay 1945, O'Neil 1949, Wilson 1953, Errington 1963).

VULNERABILITY AND THREATS

Because of the wide distribution and high reproductive potential, there are no known threats to survival of the species. Local common muskrat populations can be influenced by extreme environmental conditions, such as flooding and drought (Svihla and Svihla 1931, Errington 1963, Wilson 1968). Predation by mink may have a strong influence on local populations in northern areas of the distribution where prey diversity is low (Errington 1963, Erb et al. 2001).

Tyzzer's disease, a fatal bacterial disease known primarily from muskrats and rabbits in the wild, has decimated populations in the Midwest and Canada, but is not a significant source of mortality in the South (Davidson and Nettles 1997).

MANAGEMENT SUGGESTIONS

Throughout most of the 20th century, the common muskrat was one of the most highly valued furbearers in North America (Novak et al. 1987). Although management of wetlands specifically for muskrats was commonly practiced in the last century (Errington 1961), declines in fur demand and increased awareness of the role of wetlands in ecosystem function have led to more holistic approaches to wetland management. Most current management for the common muskrat is related to reducing damage from burrowing activities.

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Common Muskrat (*Ondatra zibethicus*)

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Marsh Rice Rat

Oryzomys palustris (Harlan, 1837)

Steven B. Castleberry and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

The taxonomy of the marsh rice rat has been the subject of considerable debate. Until recently, all rice rats in the eastern United States were considered a single species, *Oryzomys palustris*, with six recognized subspecies: *O. p. coloratus*, *O. p. natator*, *O. p. palustris*, *O. p. planirostris*, *O. p. sanibeli*, and *O. p. texensis* (Hall 1981, Wolfe 1982). In 1978, Spitzer and Lazell (1978) described a population of rice rats on the Lower Keys of Florida as a distinct species, *O. argentatus*. However, in a comprehensive review of the *Oryzomys* complex, Humphrey and Setzer (1989) found no species-level differences between *O. argentatus* and *O. palustris*, and recognized a single species with only two subspecies: *O. p. palustris* (= *O. p. palustris* and *O. p. texensis*) and *O. p. natator* (= *O. p. coloratus*, *O. p. natator*, *O. p. planirostris*, *O. p. sanibeli*, and *O. argentatus*). Goodyear (1991) later argued that retention of the species-level status of *O. argentatus* was justified. Although we follow Jones et al. (1997) and Musser and Carleton (1993) in accepting the synonymy of Humphrey and Setzer (1989), we recognize the need for additional diagnostic work. Wolfe (1982) reviewed the literature on the marsh rice rat.

DISTINGUISHING CHARACTERISTICS

The pelage coloration of the marsh rice rat is highly variable, but the dorsum generally ranges from gray to grayish-brown, mixed with black hairs. Some populations are reddish-brown or silver. The sides are lighter with less black and the venter and feet are grayish white. The long, sparsely haired tail is not distinctly bicolored. The underfur is dense, soft, and water repellent. Measurements are: total length, 226–332 mm; tail, 108–152 mm; hind foot, 28–40 mm; ear, 12–16 mm; weight, 45–80 g. The marsh rice rat is externally similar to the Norway rat (*Rattus norvegicus*) and black rat (*R. rattus*), but the pelage of *Oryzomys* is lighter below producing a conspicuous contrast with the dorsum. The dental formula is I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). See keys for details.

CONSERVATION STATUS

The marsh rice rat has a global rank of Secure (NatureServe 2007). It is considered Secure in Alabama, Florida, Georgia, Louisiana, Mississippi,

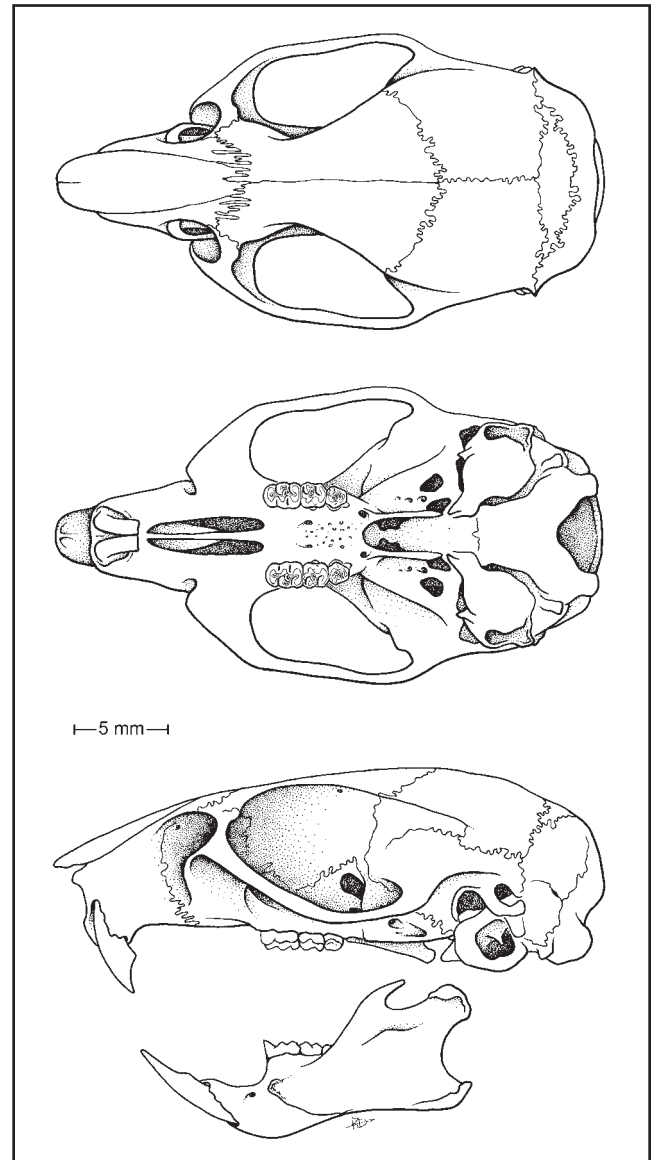


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Oryzomys palustris* from Lancaster County, Virginia (USNM 311824, female).

North Carolina, Tennessee, and Virginia, Apparently Secure in Arkansas, Kentucky, and Texas, and Imperiled in Oklahoma. It is unranked in South Carolina.

DISTRIBUTION

The marsh rice rat has a wide distribution throughout the southern United States (Figure 2). It is restricted to the lower Piedmont and Coastal Plain, including coastal areas of Virginia (Dueser et al. 1979, Pagels et al. 1992, Linzey 1998) and North Carolina (Lee et al. 1982, Webster et al. 1985). It occurs throughout South Carolina (Coleman 1948, Golley 1966), Georgia (Golley 1962, Neuhauser and Baker 1974), Florida (Pournelle and Barrington 1953, Layne 1974), Alabama (Linzey 1970), Mississippi (Jones and Carter 1989), Louisiana (Lowery 1974), and into eastern Texas (Schmidly 1983, Cleveland et al. 1984). To the north it ranges through all but extreme northeastern Tennessee (Howell and Conaway 1952, Goodpaster and Hoffmeister 1952), into southern Kentucky (Barbour and Davis 1974), west throughout all but north-central Arkansas (Sealander and Heidt 1990), and into southeastern Oklahoma (Caire et al. 1989).

ABUNDANCE STATUS

The marsh rice rat is one of the most abundant mammals of coastal marshes and freshwater wetlands. Density estimates range from 4.1/ha in a Texas coastal prairie (Joule and Jameson 1972) to as high as 48/ha in the Florida Everglades, where density was highest between August and October (Smith and Vrieze 1979). Monthly capture rates of 0.5–17.8/ha were reported by Negus et al. (1961) from a sedge-shrub coastal dune community in Louisiana. In a Mississippi tidal marsh, Wolfe (1985) reported population fluctuations from 2–25/ha over a 3-year period, which he attributed partly to juvenile recruitment in summer and fall. The marsh rice rat is uncommon in uplands away from water (Lowery 1974, Sealander and Heidt 1990).

PRIMARY HABITATS

The marsh rice rat is semi-aquatic and almost always associated with dense vegetation in wetland habitats (Harper 1927, Hamilton 1946). It is most common in coastal marshes (Harris 1953, Martin et al. 1991) and freshwater wetlands (Harper 1927, Smith and Vrieze 1979). Because of its ubiquitous distribution on the mainland and ability to disperse across water (Loxterman et al. 1998), it also is common on coastal barrier islands (Negus et al. 1961, Dueser et al. 1979). It is present but not particularly abundant in Carolina bays, pocosins, and other Coastal Plain wetlands unless early successional marsh vegetation is present (Clark et al. 1985). In upland areas it occurs along densely vegetated margins of ditches, canals, bottom-land forests, streams, and lakes (Goodpaster and

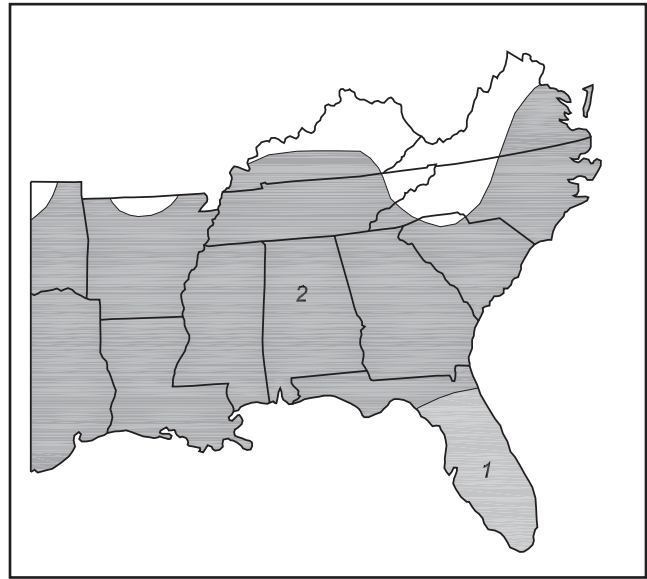


Figure 2. Distribution of *Oryzomys palustris* in the South: (1) *O. p. natator*; (2) *O. p. palustris*.

Hoffmeister 1952, Thurmond and Miller 1994, Wilkins and Broussard 2000). It is reported from wet meadows and marshy areas of the Appalachian Highlands (Coleman 1948, Linzey and Linzey 1971). The marsh rice rat is rarely found in dry upland grasslands or woodlands away from water (Wolfe 1982).

REPRODUCTION

The breeding season is variable depending on population density and environmental conditions (Negus et al. 1961). Breeding apparently occurs year-round in most populations (Conaway 1954, Negus et al. 1961), but may not occur in winter in some parts of the range (Svihla 1931, Worth 1950). Wolfe (1985) reported year-round breeding in Mississippi, but with the highest proportion of pregnant females in late spring and late summer. Gestation probably ranges between 21–28 days (Wolfe 1982). Litter sizes range from 3–7 with reported averages ranging from 3.0–4.8 (Svihla 1931, Hamilton 1946, Negus et al. 1961). The maximum number of litters per year is estimated at 5–6, but is probably less in most years (Negus et al. 1961). The young are raised in nests made of woven-grasses, sedges, and cattails (*Typha* spp.) built on the ground above water level (Hamilton 1946, Goodpaster and Hoffmeister 1952, Negus et al. 1961) or attached to marsh vegetation above water level (Sharp 1967). The young are weaned at 11–13 days (Hamilton 1946) and both sexes may reach sexual maturity as early as 40–60 days (Svihla 1931, Conaway 1954, Negus et al. 1961). The average life span in the wild is about 7 months (Negus et al. 1961), with a maximum reported longevity of 24 months (Wolfe 1985).

FOOD HABITS

The marsh rice rat has been characterized as predominantly carnivorous (Sharp 1967), but it also consumes significant amounts of seeds and plant material (Svihla 1931, Negus et al. 1961). Although the relative amount of plant and animal food varies seasonally, overall each makes up a relatively equal part of the diet (Negus et al. 1961).

Animal foods include insects, crabs, snails, fish, vertebrate carrion, and the eggs and young of turtles and birds (Hamilton 1946, Goodpaster and Hoffmeister 1952, Kale 1965, Sharp 1967, Lowery 1974, Goodyear 1992). Reported plant foods include cordgrass (*Spartina* spp.), glasswort (*Salicornia* spp.), gamma grass (*Tripsacum dactyloides*), wild rye (*Elymus virginicus*), peppervine (*Ampelopsis arborea*) fruits, and acorns (*Quercus* spp.; Hamilton 1946, Negus et al. 1961, Kincaid and Cameron 1982, Savage et al. 1996). In tidal marshes, marsh rice rats construct feeding platforms from the stems of marsh vegetation where they feed during high tides (Hamilton 1946).

ASSOCIATED SPECIES

Because of the wide distribution and diversity of habitats, the marsh rice rat is associated with a variety of species. Common small mammal habitat associates that have been studied as potential competitors include hispid cotton rats (*Sigmodon hispidus*), meadow voles (*Microtus pennsylvanicus*), and harvest mice (*Reithrodontomys* spp.; Harris 1953, Joule and Jameson 1972). Other associated small mammals include cotton mice (*Peromyscus gossypinus*), black rats, southern short-tailed shrews (*Blarina carolinensis*), and least shrews (*Cryptotis parva*; Hamilton 1946, Smith and Vrieze 1979, Wolfe 1985, Goodyear 1992). Known mammalian predators include northern raccoons (*Procyon lotor*), mink (*Mustela vison*), long-tailed weasels (*M. frenata*), striped skunks (*Mephitis mephitis*), red fox (*Vulpes vulpes*), and gray fox (*Urocyon cinereoargenteus*; Harris 1953, Hamilton and Whitaker 1979). Marsh rice rats comprise a large proportion of barn owl (*Tyto alba*) diets throughout the range (Wolfe 1982). Other avian predators include the northern harrier (*Circus cyaneus*; Harris 1953) and barred owl (*Strix varia*; Hamilton and Whitaker 1979). Snakes associated with aquatic habitats, such as cottonmouths (*Agkistrodon piscivorus*), probably also are predators (Svihla 1931).

VULNERABILITY AND THREATS

The species is so widespread and abundant that regionally there appear to be no threats to the species survival. However, significant loss or deterioration

of wetland habitats may make local populations vulnerable to extirpation.

MANAGEMENT SUGGESTIONS

Streamside management zones (SMZs) may provide habitat for marsh rice rats in intensively managed forests. In the Upper Coastal Plain of Georgia, Thurmond and Miller (1994) captured marsh rice rats in hardwood SMZs up to 50 m wide, but recorded no captures in young pine plantations 50 m from the SMZ edge. Clarke et al. (1985) suggested that fire and anthropogenic disturbance in pocosins and Carolina bays created and maintained a sedge/grass/rush community suitable as marsh rice rat habitat.

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Texas Mouse

Peromyscus attwateri (Allen, 1895)

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CONTENT AND TAXONOMIC COMMENTS

Peromyscus attwateri was previously regarded a subspecies of *P. boylii* by Bailey (1906), but more recent chromosomal (Lee et al. 1972) and morphological (Schmidly 1973) studies revealed *P. attwateri* to be a distinct species. Although populations of the Texas mouse vary morphologically and genetically, the degree of morphological variation does not justify recognition of subspecies (Schmidly 1973, Sugg et al. 1990a). The literature on the Texas mouse is reviewed by Schmidly (1974).

DISTINGUISHING CHARACTERISTICS

The Texas mouse is considered medium-sized for the genus *Peromyscus*. Measurements are: total length, 160–220 mm; tail, 68–116 mm; hind foot, 20–27 mm; ear, 15–22 mm; weight, 19–41 g. The dorsal coloration is grayish-brown to brown, with the mid-dorsal pelage darker brown mixed with black hairs. The sides are described as “pinkish cinnamon.” The venter and feet are white, but the ankles are dark or dusky. The tail is distinctly bicolored and well haired, with a prominent tuft at the tip. The long, tufted tail is used as a prop and for balance when climbing vertical rock surfaces (Long 1961). It is distinguished from sympatric *Peromyscus* within the region by the terminal tail tuft. The dental formula is I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). See keys for details.

CONSERVATION STATUS

The golden mouse has a global rank of Secure (NatureServe 2007). It is considered Secure in Texas and Apparently Secure in Oklahoma and Arkansas.

DISTRIBUTION

The distribution of the Texas mouse is discontinuous, reflecting the distribution of rocky habitats to which it is largely restricted (Schmidly 1974). Although the range extends from south-central Texas northward into southern Kansas and southwestern Missouri (Schmidly 1973), the species is absent from eastern Texas (Figure 2). In the South, the Texas mouse occurs in northwestern Arkansas (Sealander and Heidt 1990) and all of eastern Oklahoma (Caire et al. 1989).

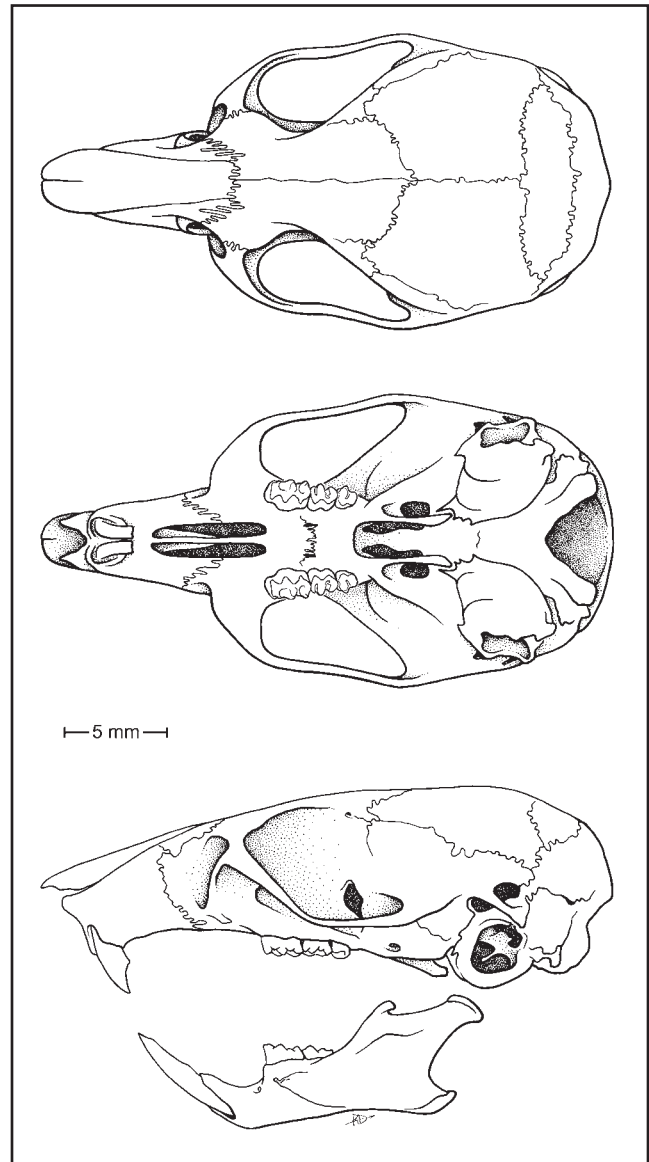


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Peromyscus attwateri* from the Wichita Mountains, Comanche County, Oklahoma (USNM 132777, female).

ABUNDANCE STATUS

The Texas mouse appears to be common in appropriate habitat throughout its range (Blair 1938, Glass and Halloran 1961, Taylor 1964). Although no regional density estimates are available, densities have been

estimated at 3.0–3.5/ha in west Texas (Garner 1967) and at 0.7–5.4/ha in Missouri (Brown 1964a).

PRIMARY HABITATS

The Texas mouse is restricted to rocky outcrops, particularly limestone or sandstone rocky prominences, such as escarpments or cliff faces where they inhabit crevices, cracks, or debris (Blair 1938, Brown 1964a, Montgomery 1984). In the Ozarks of Arkansas and southern Missouri, it is strongly associated with rocky outcrops in the eastern red cedar (*Juniperus virginiana*) glade-shrubby woodland-grasslands characteristic of shallow soils overlying dolomite (Brown 1964b). In the Ouachita Mountains of Arkansas, the Texas mouse is closely associated with rock outcrops and talus slopes in the pine-oak-hickory (*Pinus-Quercus-Carya*) woodlands, and with south-facing rocky ravines and exposed shale with associated eastern red cedar woodlands (Sealander and Heidt 1990). In northeastern Oklahoma, it occurs in narrow, steep-walled limestone ravines with vegetation characterized by oak, sugar maple (*Acer saccharum*), American elm (*Ulmus americana*), and black hickory (*Carya buckleyi*; Blair 1938).

REPRODUCTION

Little is known regarding reproduction in the Texas mouse. Breeding apparently occurs only in spring and fall in Missouri and Arkansas populations (Cockrum 1952, Brown 1964a). Garner (1967) documented breeding throughout winter in west Texas, but found no evidence of breeding in late spring and summer. In Kansas, Long (1961) noted breeding activity in every month except January. Length of gestation is uncertain. Litter size is reported to be between 3–6 (Cockrum 1952, Long 1961). Males and females typically attain sexual maturity between 12–19 weeks (Brown 1963). Mean life span has been reported at 6.8 months up to a maximum of 18 months in the wild (Brown 1964a).

FOOD HABITS

In southwest Oklahoma, Texas mice consumed primarily acorns, Johnson grass (*Sorghum halepense*), and centipedes (*Chilopoda* spp.) from March to June (wet season), while the most common foods from July to October (dry season) were prickly pear cactus (*Opuntia* spp.), darkling beetles (Tenebrionidae), and seed bugs (Lygaeidae; Stancampiano and Caire 1995). Brown (1964b) examined stomach contents of Texas mice in Missouri and found that seeds and insects comprised over 84% of the diet in all seasons. Camel crickets (*Ceuthophilus* spp.) and ground beetle larvae (Carabidae) were the most common insects

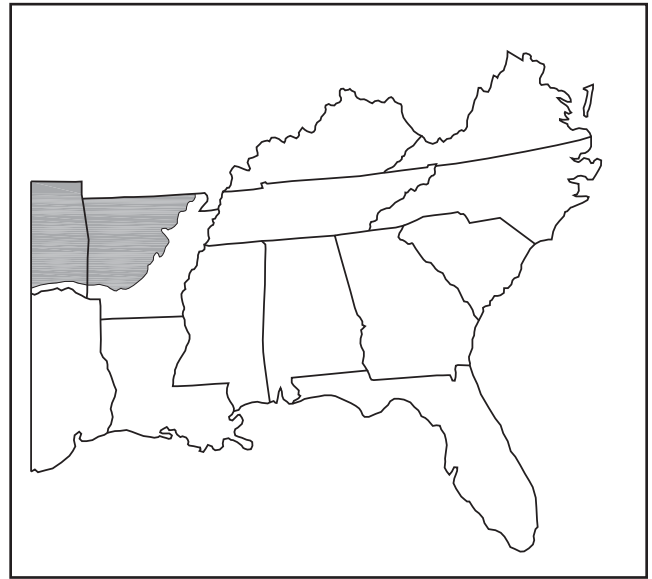


Figure 2. Distribution of *Peromyscus attwateri* in the South.

consumed. Other reported foods included berry integuments and green plant fragments. Long (1961) reported acorns as the most common food item in Kansas, with seeds and insects uncommon or absent. No regional information is available on food habits of the Texas mouse.

ASSOCIATED SPECIES

The Texas mouse may occur in association with the cotton mouse (*Peromyscus gossypinus*), white-footed mouse (*P. leucopus*), deer mouse (*P. maniculatus*), hispid cotton rat (*Sigmodon hispidus*), eastern woodrat (*Neotoma floridana*), eastern gray squirrel (*Sciurus carolinensis*), fox squirrel (*S. niger*), striped skunk (*Mephitis mephitis*), eastern mole (*Scalopus aquaticus*), eastern cottontail (*Sylvilagus floridanus*), and bats that occupy caves and crevices, such as the eastern pipistrelle (*Pipistrellus subflavus*; Blair 1938, Brown 1964b). Known predators of the Texas mouse are the broad-winged hawk (*Buteo platypterus*) and the coachwhip (*Masticophis flagellum*; Brown 1964a). Other snakes, owls, hawks, skunks, and foxes probably also are predators.

VULNERABILITY AND THREATS

Although the Texas mouse is somewhat restricted in distribution and habitat associations, there appear to be no threats to its survival.

MANAGEMENT SUGGESTIONS

Aside from protection of habitats associated with rock formations within the species range, no management guidelines are suggested.

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Cotton Mouse

Peromyscus gossypinus (LeConte, 1853)

Timothy S. McCay and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Traditionally, seven subspecies of the cotton mouse have been recognized. However, Boone et al. (1993) argued that populations on Anastasia Island and Cumberland Island (formerly *P. g. anastasae*) did not warrant subspecific recognition, and this suggestion has been widely adopted. Boone (1995) also found evidence that the Key Largo cotton mouse (*P. g. allapaticola*) is not sufficiently distinct to warrant its subspecific taxonomic distinction. However, that subspecies is still accepted by many authors. Several islands in Florida and Georgia (e.g., Amelia Island) are inhabited by cotton mice that are relatively distinct (Boone et al. 1999), though these populations are not currently recognized as taxonomically distinct. Overall, the pattern of genetic variability in this species does not closely match the current distribution of recognized subspecies (Robbins et al. 1985, Boone et al. 1999).

DISTINGUISHING CHARACTERISTICS

Of the five *Peromyscus* species in the South, the cotton mouse is the largest. Measurements are: total length, 142–206 mm; tail length, 55–97 mm; hind-foot length, 16–26 mm; ear length, 10–21 mm, and weight 17–46 g (Wolfe and Linzey 1977). Upper pelage of adults is golden brown to cinnamon, typically with a darker middle dorsal area. Venter is cream to white. Subadults, which may be encountered outside the nest, are gray above and white to cream below. The tail is bicolored, though indistinctly, dark above and light below. The pelage tends to be lighter in spring than in autumn or winter. The cotton mouse may be distinguished from similar sympatric species (*Peromyscus* and *Podomys* spp.), except the white-footed mouse (*P. leucopus*), by its possession of 6 plantar tubercles, large size, and tendency to have an indistinctly bicolored tail (Wolfe and Linzey 1977). Separating the cotton mouse from the white-footed mouse is difficult using observational characteristics (Barko and Feldhamer 2002), although the cotton mouse tends to have a longer hind foot (≥ 22 mm) and a greater mass (≥ 26 g) than the white-footed mouse (Hoffmeister 1989). Electrophoresis may be used to positively identify this species from blood or tissue samples (Price and Kennedy 1980). The skull is similar to other regional *Peromyscus* species, although it is generally larger than those of other congeners.

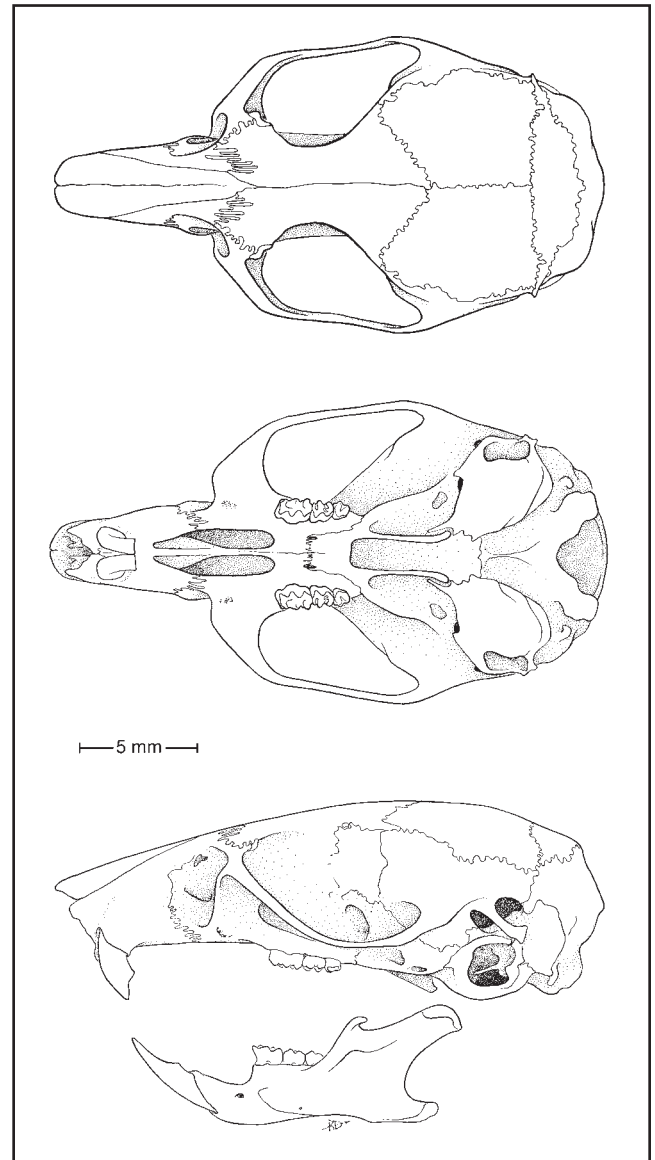


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Peromyscus gossypinus* from Marion County, Florida (USNM 510846, female).

Algebraic functions for discriminating among *Peromyscus* species on the basis of skull and external characters have been developed for application in Alabama (Linzey et al. 1976), Arkansas (McDaniel et al. 1983), Texas (Engstrom et al. 1982), and throughout much of the South (Laerm and Boone 1994). The dental formula is $I\ 1/1, C\ 0/0, P\ 0/0, M\ 3/3 = 16$ (Figure 1). See keys for details.

CONSERVATION STATUS

The Key Largo cotton mouse (*P. g. allapaticola*), which is found only on Key Largo, Monroe County, Florida, is listed as Endangered by the U. S. Fish and Wildlife Service (United States Department of the Interior 2007) and the Florida Fish and Wildlife Conservation Commission. The Chadwick Beach cotton mouse (*P. g. restrictus*), described from Sarasota County, Florida, has not been detected since 1938 and is likely extinct.

The cotton mouse has a global rank of Secure (NatureServe 2007). It is Secure in Alabama, Florida, Georgia, Louisiana, Mississippi, North Carolina, Tennessee and Texas. It is Apparently Secure in Arkansas. Virginia and Oklahoma classify it as Vulnerable and Kentucky lists it as Imperiled. It is unranked in South Carolina. States where the species is of conservation concern are on the periphery of the geographic range.

DISTRIBUTION

The cotton mouse ranges from southeastern Virginia (Ulmer 1963, Boone and Laerm 1993) along the Coastal Plain in North Carolina (Lee et al. 1982) and South Carolina (Golley 1966, Sanders 1978; Figure 2). In Georgia the cotton mouse is found in the Coastal Plain, Piedmont, Ridge and Valley, and Cumberland Plateau physiographic provinces (Laerm and Boone 1995), and it is found throughout Florida. The cotton mouse ranges north into Tennessee (Linzey and Linzey 1971, Kennedy 1991) and extreme western Kentucky (Barbour and Davis 1974). It ranges throughout Alabama (Linzey et al. 1976), Mississippi (Kennedy et al. 1974, Jones and Carter 1989), and Louisiana (Lowery 1974). The cotton mouse is found in all but extreme northern Arkansas (Sealander and Height 1990), eastern Texas (Davis and Schmidly 1994), and southeastern Oklahoma (Caire et al. 1989).

ABUNDANCE STATUS

The cotton mouse is both ubiquitous within its geographic range and locally dense; density is dependent on habitat, season, and climatic conditions. Density estimates for cotton mice include 1.5–9.7/ha in loblolly pine (*Pinus taeda*) plantations on the Coastal Plain of South Carolina (Smith et al. 1984), 3.8/ha in bottomland hardwood forest in the same region (Smith et al. 1971), up to 3.0/ha in a pine-hardwood forest in Louisiana (Shadowen 1963), and 0.7–6.7/ha in eastern Texas (McCarley 1954a). Calhoun (1941) reported 97/ha in a bottomland forest in Tennessee. Populations are generally highest in winter or late spring and lowest in summer (McCarley 1954a, Shadowen 1963). Smith et al. (1974) observed that the abundance of cotton mice fluctuated positively with summer precipitation and negatively with winter

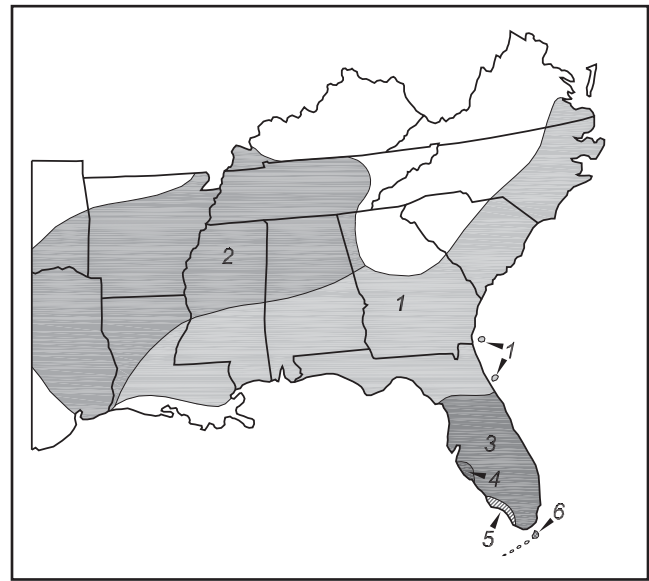


Figure 2. Distribution of *Peromyscus gossypinus* in the South: (1) *P. g. gossypinus*; (2) *P. g. megacephalus*; (3) *P. g. palmarius*; (4) *P. g. restrictus*; (5) *P. g. telmaphilus*; (6) *P. g. allapaticola*.

temperatures in the year prior. Humphrey (1988) estimated the density of the Key Largo cotton mouse at 15.5/ha near housing subdivisions and 26.9/ha away from subdivisions.

PRIMARY HABITATS

The cotton mouse is most common in bottomland hardwood forests, swamps, and hammocks; it also occurs in pine forests, mixed pine-hardwood forests, and early successional habitats (Wolfe and Linzey 1977). It is found in maritime forests along the coast and has been noted on rocky ledges in upland habitats. The cotton mouse has been found from lower elevations (< 600 m) in the Great Smoky Mountain foothills in eastern Tennessee (Linzey and Linzey 1971). In a fragmented landscape of early successional habitat within a matrix of pine plantations, cotton mice readily moved through the forested matrix (Mabry and Barrett 2002) and exhibited a relatively uniform abundance across the landscape (Danielson and Hubbard 2000, Mabry et al. 2003). Downed coarse woody debris is used extensively by cotton mice in managed pine forests (McCay 2000) and increased abundance of dead wood following a tornado was associated with increased abundance, survival, and reproduction (Loeb 1999). Cotton mice use decomposing stumps and root boles as daytime nest sites (Ivey 1949, Frank and Layne 1992, McCay 2000). Fallen logs are used as runways during nighttime movements (McCay 2000). Abundance of dead wood is an important determinant of capture success in young loblolly pine stands (Mengak and Guynn 2003).

REPRODUCTION

The breeding season is long and may extend throughout the year in the southern portions of its geographic range (Pournelle 1952). The cotton mouse exhibits lowest reproduction in summer (McCarley 1954a, O'Farrell et al. 1977), which may be due to the effects of high temperature on reproductive physiology (Pournelle 1952). Gestation is approximately 23 days; litters range from 1–7 young with a mean of 3 (Pournelle 1952, O'Farrell et al. 1977). A postpartum estrus occurs and females may have several litters per year. Young are weaned by the third or fourth week of life and individuals reach sexual maturity between 7–12 weeks (Pournelle 1952).

FOOD HABITS

The cotton mouse is omnivorous, eating a variety of plant and animal foods (Calhoun 1941, Rand and Host 1942). Common foods include beetles, caterpillars, and spiders (Calhoun 1941). Plant foods include hard mast and berries of wax myrtle (*Myrica cerifera*) (Calhoun 1941, Rand and Host 1942). Supplemental food resulted in an increase of cotton mice in loblolly pine forests (Smith et al. 1984) and sand-pine (*P. clausa*) scrub (Young and Stout 1986), suggesting that populations of cotton mice are limited by food in these habitats.

ASSOCIATED SPECIES

Due to its ubiquity, the cotton mouse occurs with a variety of rodents and insectivores. Notably, there is significant overlap in the ranges of the cotton mouse and the white-footed mouse. Where they are sympatric, the cotton mouse tends to be restricted to moist environments (e.g., river floodplains), while the white-footed mouse is restricted to upland habitats (McCarley 1963). Natural hybridization between these two species has been suggested on the basis of morphology (Howell 1921, McCarley 1954b) and genetic data (Barko and Feldhamer 2002). However, Price and Kennedy (1980) found no evidence of gene flow between these species in an area of sympatry. The Key Largo cotton mouse had a competitive effect on populations of Key Largo woodrats (*Neotoma floridana smalli*); however, the reverse was not the case (Sasso and Gaines 2002). Similarly, McCarley (1963) suggested that the cotton mouse was able to exclude the white-footed mouse from otherwise suitable habitat. The importance of the cotton mouse as a prey item in the South is not known; predators include owls (Adams et al. 1986) and snakes (Harper 1927). The spirochete that causes Lyme disease (*Borrelia burgdorferi*) has been isolated from tissues of the cotton mouse (Oliver et al. 1993, Clark et al. 2002).

VULNERABILITY AND THREATS

Because this species has broad habitat tolerance, it is secure throughout the majority of its range. Certain insular or coastal populations have been driven extinct; others may be at risk of extinction due to the loss of forested habitat by human development (Repenning and Humphrey 1986, Humphrey et al. 1988, Humphrey 1992).

MANAGEMENT SUGGESTIONS

Conservation of bottomland hardwood forests with abundant coarse woody debris would benefit this species.

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White-footed Mouse

Peromyscus leucopus (Rafinesque, 1818)

Joshua Laerm and Steven B. Castleberry

CONTENT AND TAXONOMIC COMMENTS

Seventeen subspecies of the white-footed mouse are recognized (Lackey et al. 1985), three of which occur in the South: *P. l. easti*, *P. l. leucopus*, and *P. l. noveboracensis*. Baker et al. (1983) reported two distinct chromosomal races, one in the north-central and northeastern United States and another in the southwestern portion of the range, with populations from much of the southeast as hybrids. Although the differences observed between the two chromosomal races is greater than those distinguishing some closely related species of *Peromyscus*, the hybrids reproduce successfully with no apparent reduction in fitness (Stangl 1986, Carleton 1989). The literature is reviewed by Lackey et al. (1985).

DISTINGUISHING CHARACTERISTICS

The dorsal pelage is brownish to grayish-brown, usually with a darker mid-dorsal area. Although the ventral pelage appears white, the base of the hairs are dark bluish gray. As the name implies, the feet are white. The tail is indistinctly bicolored and slightly less than half the total length. The white-footed mouse is considered medium-sized for the genus, but there is considerable size variation throughout the range. Measurements are: total length, 130–205 mm; tail, 45–100 mm; hind foot, 17–25 mm; ear, 15–18 mm; weight, 15–30 g. In the southern Appalachians, the white-footed mouse is most easily confused with the cloudland subspecies of the deer mouse (*P. maniculatus nubiterrae*), but the tail of the deer mouse usually is more than half the total body length. In the western part of the region, the tail of the prairie deer mouse (*P. m. bairdii*) is much less than half the body length and the hind foot is seldom more than 18 mm, whereas the hind foot of the white-footed mouse is usually 19 to 22 mm. The hind foot of the cotton mouse (*P. gossypinus*) is typically 22 mm or greater. The oldfield mouse (*P. polionotus*) is smaller and its tail is distinctly bicolored and considerably shorter than half the total length. Considerable variation and overlap of characters within and among species makes identification of *Peromyscus* in the field difficult. Discriminant models developed by Laerm and Boone (1994) and Bruseo et al. (1999) provide reasonably good discrimination (82–92% correct classification), but Bruseo et al. (1999) recommend using electrophoresis of

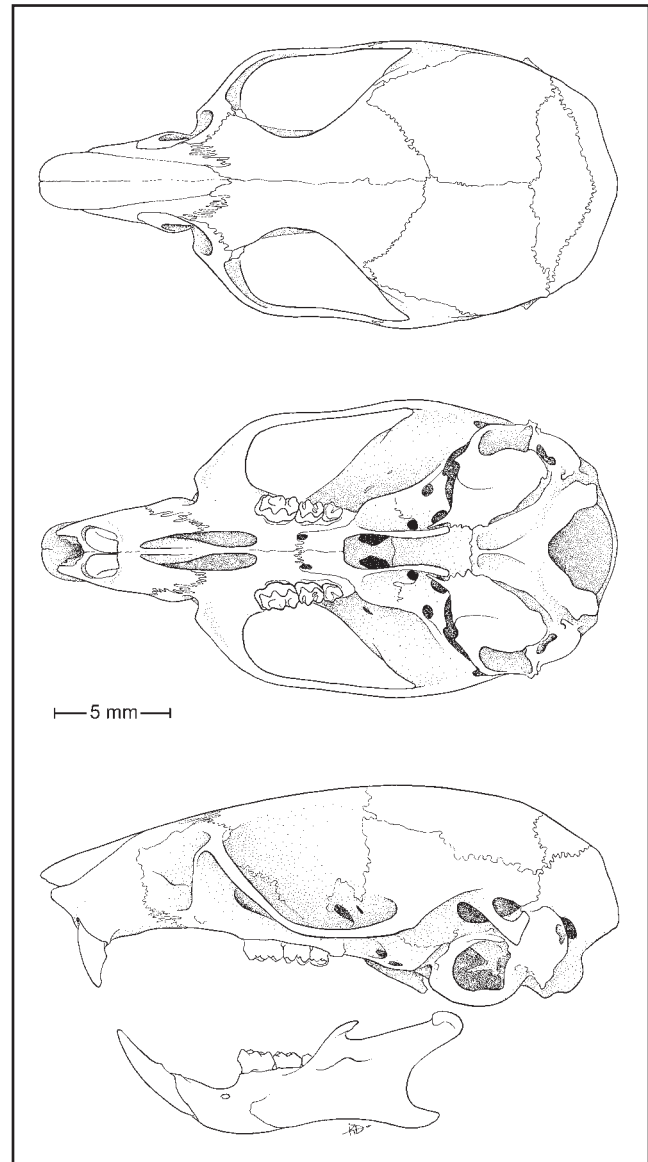


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Peromyscus leucopus* from Bath County, Virginia (USNM 489896, male).

salivary amylase in cases where unambiguous identification is needed. The dental formula is I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). See keys for details.

CONSERVATION STATUS

The white-footed mouse has a global ranking of Secure and state rankings of Secure in every state in the region except Alabama and Arkansas where it is ranked as Apparently Secure (NatureServe 2007). It is unranked in South Carolina.

DISTRIBUTION

The white-footed mouse occurs widely throughout most of the eastern United States (Figure 2). It occurs throughout every state in the region except Florida, the coastal areas of southeastern North Carolina (Webster et al. 1985), areas south of the Fall Line in South Carolina (Golley 1966), Georgia (Golley 1962, Laerm et al. 1982), and Alabama (Linzey et al. 1976), and extreme southeastern Mississippi (Jones and Carter 1989). *P. l. leucopus* is the most widespread subspecies, occurring in portions of every state in the distribution. *P. l. noveboracensis*, which inhabits most of the northeastern United States, occurs throughout western Virginia and eastern Kentucky, and extreme northwestern North Carolina and northeastern Tennessee. It also occurs in northern Arkansas and Oklahoma. *P. l. easti* has a limited distribution in extreme southeastern Virginia and northeastern North Carolina (Paradiso 1960).

ABUNDANCE STATUS

The white-footed mouse is one of the most ubiquitous and abundant small mammal throughout the South. It is most abundant on the edges of upland brushy fields and moist woodlands. Populations fluctuate seasonally and annually in response to demographic and environmental conditions such as food, weather, and density (Wolff 1985a). In particular, populations are strongly associated with temporal and spatial variations in oak (*Quercus* spp.) mast availability (McCracken et al. 1999). Densities typically range from 3–20/ha, although densities may exceed 50/ha in years of high mast production (Batzli 1977, Wolff 1985a, 1986; Vessey 1987, Krohne et al. 1988, Smith and Sloan 1988, Baccus and Wolff 1989, Kaufman and Kaufman 1989).

PRIMARY HABITATS

The white-footed mouse inhabits a diversity of habitats ranging from brushy stands of oldfield succession to mature upland pine (*Pinus*) and hardwood forests. It is an expected component of virtually all forest cover types in its range, but is most abundant in areas with dense coarse woody debris and a well-developed shrub layer (Dueser and Shugart 1978, 1979; Kaufman et al. 1983, Drickamer 1990,

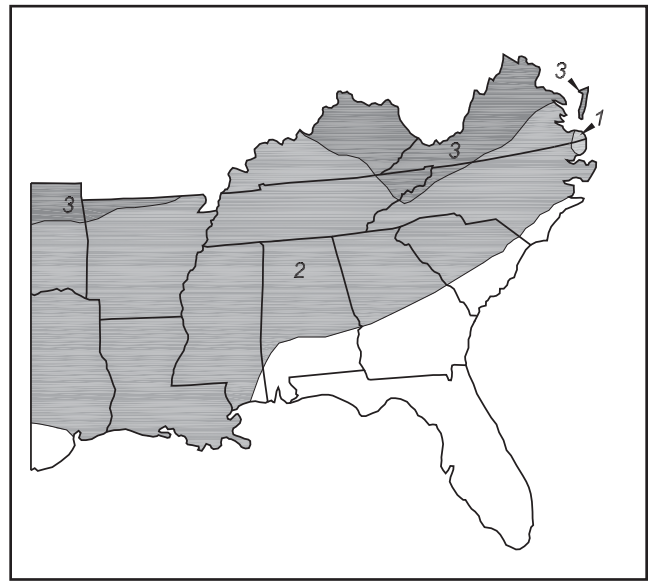


Figure 2. Distribution of *Peromyscus leucopus* in the South: (1) *P. l. easti*; (2) *P. l. leucopus*; (3) *P. l. noveboracensis*.

Menzel et al. 1999, Rossell and Rossell 1999, Greenberg 2002). Because it is considered semi-arboreal, a dense cover of vines and other understory vegetation is an important habitat component (Batzli 1977, Barry et al. 1984, 1990; Harney and Dueser 1987, Graves et al. 1988). It may select nest sites above the ground in tree cavities or on the ground in rock piles, logs, stumps, or burrows (Lackey et al. 1985).

REPRODUCTION

In higher latitudes the breeding season is bimodal, with peaks in spring and fall (Burt 1940). Southern populations tend to breed year-round, although a mid-season breeding hiatus occurs in many areas (Brown 1964, Lackey 1978, Wolff 1985a,b; Terman and Terman 1999). Gestation is 23–24 days, but may be extended in lactating females (Svihla 1932, Lackey 1978). Litter size ranges from 1–8 with reported means in the region ranging from 3.4–4.2 (Svihla 1932, Brown 1964, Wolff 1985b, Hawkins et al. 1986, Suttikus and Jones 1991). Young are weaned at 22–37 days (Svihla 1932) and become sexually mature at about 35–45 days depending on latitude (Layne 1968). Although white-footed mice may live longer than 2 years in the wild, few survive longer than 1 year (Schug et al. 1991).

FOOD HABITS

The variation in food habits of the white-footed mice reflects the diversity of habitats occupied. They are considered opportunistic omnivores, consuming relatively equal amounts of plant and animal foods.

Several studies have provided extensive lists of specific food items consumed by white-footed mice (Hamilton 1941, Whitaker 1966, Drickamer 1970, Linzey and Linzey 1973, Batzli 1977, Wolff et al. 1985). Generally, seeds and invertebrates are the most important year-round food items, although fruits and green vegetation may be consumed in significant amounts seasonally. They also will opportunistically feed on flowers, fungi, and occasionally other vertebrates.

ASSOCIATED SPECIES

Because of a wide distribution and habitat tolerance, the white-footed mouse is associated with a diversity of small mammals. Common associates include insectivores such as the masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), pygmy shrew (*S. hoyi*), southeastern shrew (*S. longirostris*), northern short-tailed shrew (*Blarina brevicauda*), southern short-tailed shrew (*B. carolinensis*), Elliott's short-tailed shrew (*B. hylophaga*), least shrew (*Cryptotis parva*), and rodents including the Texas mouse (*Peromyscus attwateri*), deer mouse, cotton mouse, golden mouse (*Ochrotomys nuttalli*), harvest mice (*Reithrodontomys* spp.), hispid cotton rat (*Sigmodon hispidus*), microtine voles (*Microtus* spp.), southern red-backed vole (*Clethrionomys gapperi*), woodland jumping mouse (*Napaeozapus insignis*), and meadow jumping mouse (*Zapus hudsonius*). White-footed mice and cotton mice hybridize in captivity (Dice 1937) and natural hybrids have been reported from Virginia (Dice 1940), Alabama (Howell 1921), Georgia (Golley 1962), and Texas (McCarley 1954).

VULNERABILITY AND THREATS

The species is so widespread and abundant that regionally there appear to be no threats to its survival.

MANAGEMENT SUGGESTIONS

Little management is required to promote this highly adaptable species. Forest management practices that promote oaks can positively influence abundance, body mass, and reproduction (McCraken et al. 1999). Group selection harvesting benefits white-footed mice by increasing understory cover and soft mast production while maintaining forest overstory cover (Buckner and Shure 1985, McCracken et al. 1999).

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White-footed Mouse (*Peromyscus leucopus*)

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Deer Mouse

Peromyscus maniculatus (Wagner, 1845)

Joshua Laerm and Steven B. Castleberry

CONTENT AND TAXONOMIC COMMENTS

Because of numerous geographic races, varying degrees of population differentiation, and several zones of intergradation among races, the deer mouse has served as a model species to study intraspecific variation and speciation processes for many years (Carleton 1989). However, these factors have created considerable difficulty in taxonomy. Although Hall (1981) recognized 69 subspecies, alternative classifications have been proposed (Hogan et al. 1993, Whitaker and Hamilton 1998). Despite the taxonomic complexity, most sources agree that three subspecies occur in the South: *Peromyscus maniculatus bairdii*, *P. m. nubiterrae*, and *P. m. ozarkiarum* (Hall 1981, Hogan et al. 1993, Whitaker and Hamilton 1998). No current review of the literature is available, but King (1968) and Kirkland and Layne (1989) provide useful overviews of many aspects of the biology.

DISTINGUISHING CHARACTERISTICS

The dorsal pelage typically is gray, but can vary from gray to brown. The underparts and feet are white. The tail is distinctly bicolored, but tail length varies among the subspecies. The tail of *P. m. nubiterrae* usually is equal to or slightly more than half the total length, whereas in *P. m. bairdii* and *P. m. ozarkiarum* the tail is less than half the total length. Generally, *P. m. nubiterrae* is larger than the other subspecies. Measurements for *P. m. nubiterrae* are: total length, 157–201 mm; tail, 80–106 mm; hind foot, 19–23 mm; ear, 17–20 mm; weight, 13–29 g. Measurements for *P. m. bairdii* and *P. m. ozarkiarum* are: total length, 116–154 mm; tail, 40–66 mm; hind foot, 15–20 mm; ear, 12–20 mm; weight, 16–26 g. The deer mouse is most easily confused with the white-footed mouse (*Peromyscus leucopus*). See the white-footed mouse species account for distinguishing characteristics. The dental formula is $I\ 1/1, C\ 0/0, P\ 0/0, M\ 3/3 = 16$ (Figure 1). See keys for details.

CONSERVATION STATUS

The deer mouse has a global ranking of Secure (NatureServe 2007) and state rankings of Secure in each state in the region except Arkansas and Kentucky, where it is ranked as Apparently Secure. It is unranked in South Carolina.

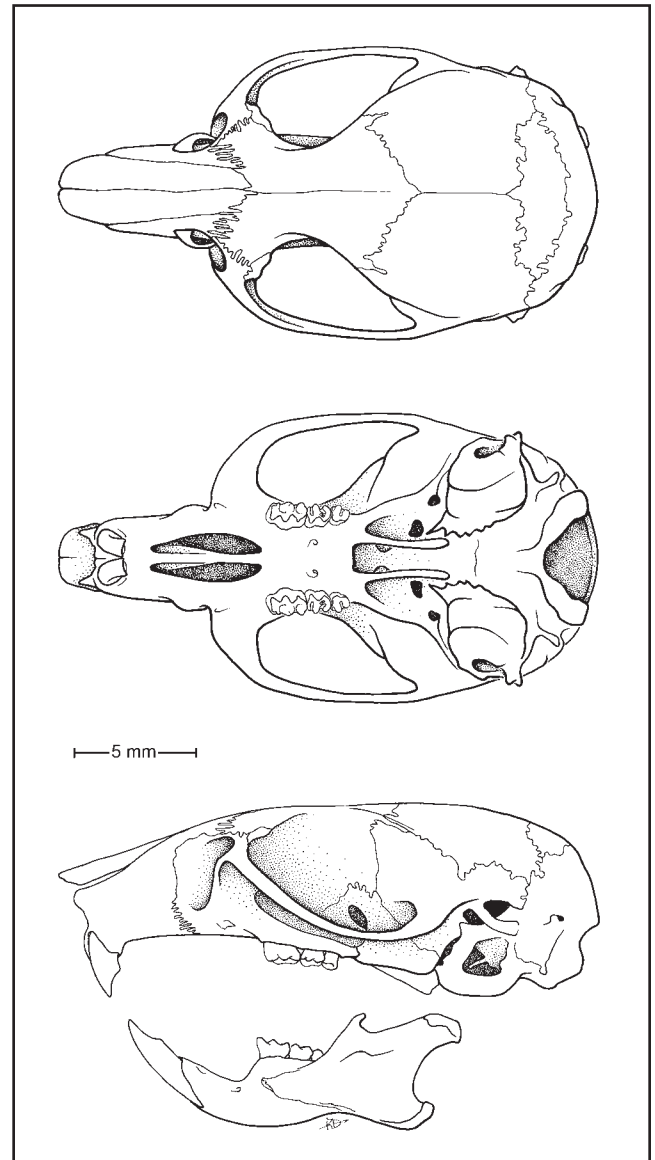


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Peromyscus maniculatus* from Howard County, Maryland (USNM 364983, female).

DISTRIBUTION

Although the deer mouse is one of the most widespread mammals in North America, generally ranging from the Canadian Arctic south into Mexico, the distribution is somewhat restricted in the region (Figure 2). The woodland or cloudland deer mouse

(*P. m. nubiterrae*) is restricted to the higher elevations of the Appalachian Mountains of Virginia (Linzey 1998), extreme eastern Kentucky (Barbour and Davis 1974), eastern Tennessee (Howell and Conaway 1952, Linzey and Linzey 1968, Smith et al. 1974), western North Carolina (Lee et al. 1982), western South Carolina (Golley 1966, Laerm et al. 1995), and north-eastern Georgia (Golley 1962, Laerm et al. 1982, Ford et al. 1994). The prairie deer mouse (*P. m. bairdii*) has recently expanded its range from the upper Midwest east across the Appalachians into eastern Pennsylvania, Maryland, and northern Virginia (Peacock and Peacock 1962, Pitts and Kirkland 1987). Elsewhere the prairie deer mouse ranges only west of the Appalachian Mountains in Kentucky (Barbour and Davis 1974), all but southeastern Tennessee (Beasley and Severinghaus 1973, Kennedy 1991), into eastern Arkansas (Sealander and Heidt 1990), and extreme northern Oklahoma (Caire et al. 1989). The Ozark deer mouse (*P. m. ozarkiarum*) ranges throughout northwestern Arkansas (Black 1935, Sealander and Heidt 1990) and northeastern Oklahoma (Caire et al. 1989), and probably intergrades with the prairie deer mouse in both states.

ABUNDANCE STATUS

The deer mouse is uncommon to abundant in various locations across the region. Reported densities range between 0.7–50/ha, but considerable variation in density occurs seasonally and among habitat types (Terman 1986, Wolff 1985a,b; Baccus and Wolff 1989). Although widely distributed at higher elevations in the southern Appalachians (Barbour 1951, Conaway and Howell 1953, Ford et al. 1994) and in western Tennessee (Goodpaster and Hoffmeister 1952, Kennedy 1991), it is not particularly abundant anywhere. Populations in some areas exhibit 4–6 year cyclical population fluctuations (Wolff 1985a).

PRIMARY HABITATS

Deer mice occupy a wide variety of habitats that vary considerably among subspecies. The woodland subspecies (*P. m. nubiterrae*) associated with the Appalachian Mountains, is largely restricted to higher elevation, mesic, deciduous, and mixed coniferous-deciduous woodlands (Barbour 1951, Conaway and Howell 1953, Lee et al. 1982, Ford et al. 1994). It is semi-arboreal, frequently climbing trees and shrubby vegetation for traveling, foraging, and nesting (Barry et al. 1984, 1990; Harney and Dueser 1987, Wolff and Durr 1986, Graves et al. 1988). Deer mice are frequently found in large trees (>30 cm dbh; Graves et al. 1988). Nests are constructed of grass and leaves and usually are located in large hollow trees, but sometimes in stumps, or under rocks or

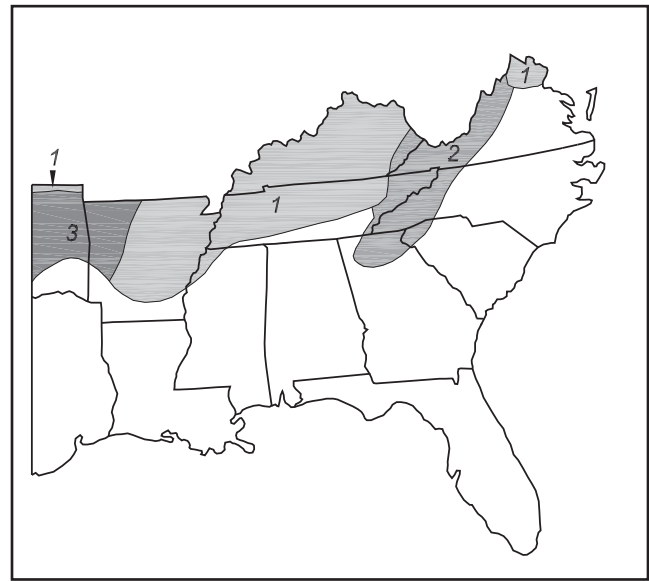


Figure 2. Distribution of *Peromyscus maniculatus* in the South: (1) *P. m. bairdii*; (2) *P. m. nubiterrae*; (3) *P. m. ozarkiarum*.

logs (Baker 1968, Wolff and Hurlbutt 1982, Wolff and Durr 1986, Graves et al. 1988). The grassland subspecies (*P. m. bairdii* and *P. m. ozarkiarum*) are associated with upland grassland habitats including fallow fields, fencerows, highway right-of-ways, pastures, and cultivated croplands (Black 1935, Goodpaster and Hoffmeister 1952, Harris 1952, Brown 1964, Kennedy 1991). These forms typically avoid woodlands, even though the ground cover may be grassy. They nest in underground burrows, beneath rocks, or in clumps of grass or weeds (Barbour and Davis 1974, Sealander and Heidt 1990).

REPRODUCTION

Breeding occurs year-round but tends to be concentrated in early spring and late fall with a mid summer low (Linzey 1970, Barbour and Davis 1974, Webster et al. 1985, Sealander and Heidt 1990). Litter size ranges between 1–9 with reported means ranging from 3.0–5.5 (Linzey 1970, Kirkland and Linzey 1973, McLaren and Kirkland 1979). Gestation lasts 22–35 days and there is a postpartum estrus (Svihla 1935, Myers and Master 1983, Millar and Millar 1989). Although deer mice in captivity can produce up to 12 litters/year (Kirkland and Linzey 1973), 2–4 litters/year are probably more typical in the wild, depending upon weather and food availability. Young are weaned in 22–37 days and sexually mature by 6–8 weeks (Svihla 1932). They seldom live more than 1 year in the wild (Sealander and Heidt 1990).

FOOD HABITS

Deer mice are omnivorous, consuming significant amounts of both plant and animal foods. Seeds, fruit, and insects are the most important food items year-round, although insects typically make up a greater portion of the diet in summer (Hamilton 1941, Whitaker 1966, Linzey and Linzey 1973, Wolf et al. 1985). Green vegetation and fungi also can be seasonally important components of the diet (Hamilton 1941, Wolf et al. 1985). Agricultural crops, including wheat (*Triticum aestivum*) seeds and soybeans (*Glycine* spp.), may be consumed in significant amounts when available (Whitaker 1966). For lists of specific food items see Hamilton (1941), Whitaker (1966), Linzey and Linzey (1973), and Wolf et al. (1985). Deer mice may hoard food items in tree cavities, burrows, and other sheltered areas (Tannenbaum and Pivorun 1987) or scatter hoard individual seeds in the vicinity of the nest (Vander Wall et al. 2001).

ASSOCIATED SPECIES

The deer mouse is commonly associated with insectivores such as the northern short-tailed shrew (*Blarina brevicauda*), southern short-tailed shrew (*B. carolinensis*), masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), water shrew (*S. palustris*), rock shrew (*S. dispar*), hairy-tailed mole (*Parascalops breweri*), and rodents such as harvest mice (*Reithrodontomys* spp.), hispid cotton rat (*Sigmodon hispidus*), white-footed mouse (*Peromyscus leucopus*), Texas mouse (*P. attwateri*), southern red-backed vole (*Clethrionomys gapperi*), southern bog lemming (*Synaptomys cooperi*), meadow jumping mouse (*Zapus hudsonius*), and woodland jumping mouse (*Napaeozapus insignis*). A variety of vertebrates prey on the deer mouse, including weasels (*Mustela* spp.), skunks, coyotes (*Canis latrans*), foxes, hawks, owls, and snakes (Sealander and Heidt 1990).

VULNERABILITY AND THREATS

The species is widespread and regionally abundant; there are no apparent threats to its survival.

MANAGEMENT GUIDELINES

Woodland deer mice are associated with closed-canopy forests and may be averse to regenerating forest stands (Ford et al. 1994).

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Oldfield or Beach Mouse

Peromyscus polionotus (Wagner, 1843)

Steven B. Castleberry and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Currently, 16 subspecies of the oldfield mouse are recognized based primarily on variation in cranial measurements and pelage coloration (Osgood 1909, Howell 1939, Schwartz 1954, Bowen 1968). However, in view of the complexity of patterns in morphological variation and the sensitivity regarding conservation concerns, the species is in need of taxonomic revision. Reviews of the biology and status of individual subspecies were conducted by Humphrey and Barbour (1981), Holliman (1983, 1986*a,b*), Holler et al. (1989), Holler and Rave (1991), Holler (1992*a,b*), Humphrey (1992*a,b*), James (1992), and Stout (1992).

DISTINGUISHING CHARACTERISTICS

The oldfield mouse shows a wide variation in pelage coloration throughout its range, with a tendency to match its background. The dorsal surface varies from pale to almost white in coastal and insular populations living on sandy soils, to brownish-gray in inland populations occurring in oldfield habitats. Populations occurring north of the fall line in South Carolina and Georgia have the darkest coloration. The underparts and feet are white. The tail is distinctly bicolored and relatively short, but length varies among populations (Schwartz 1954). Measurements are: total length, 119–142 mm; tail, 38–57 mm; hind foot, 15–19 mm; ear, 11–16 mm; weight, 8–19 g. Compared to the cotton mouse (*P. gossypinus*) and white-footed mouse (*P. leucopus*), with which they are often associated, oldfield mice generally are smaller and with a shorter, distinctly bicolored tail. The dental formula is I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). See keys for details.

CONSERVATION STATUS

As a species, the oldfield mouse has a global ranking of Secure (NatureServe 2007), and state rankings of Secure in Alabama, Florida, and Georgia. It is Apparently Secure in Tennessee, Imperiled in Mississippi, and Critically Imperiled in North Carolina. It is unranked in South Carolina. However, several subspecies are federally and state listed as Threatened or Endangered. The Choctawhatchee beach mouse (*P. p. allopheys*), Alabama beach mouse (*P. p. ammobates*), Anastasia Island beach mouse (*P. p. phasma*), Perdido Key beach mouse (*P. p. trissyllepsis*), and St. Andrew

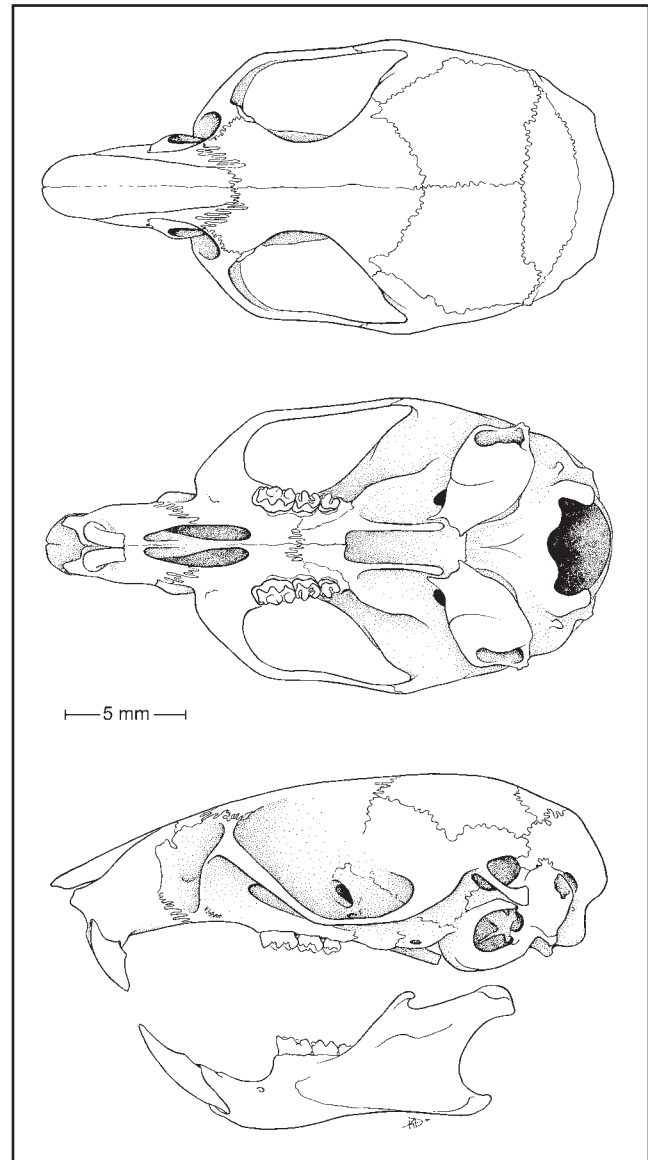


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Peromyscus polionotus* from Inlet Beach, Florida (USNM 308900, male).

beach mouse (*P. p. peninsularis*) are listed as Endangered by the United States Fish and Wildlife Service and by the wildlife conservation agencies of the states in which they occur (U. S. Department of Interior 2007). The southeastern beach mouse (*P. p. niveiventris*) is listed as threatened by the United States Fish and Wildlife Service and the Florida Fish

and Wildlife Conservation Commission. The pallid beach mouse (*P. p. decoloratus*), once considered for federal listing, is apparently now extinct (Humphrey and Barbour 1981, Humphrey 1992a).

DISTRIBUTION

The oldfield mouse occurs in a small area of the Piedmont of southwestern North Carolina (Lee et al. 1982) and throughout the Piedmont and Coastal Plain of South Carolina (Coleman 1939, 1948; Schwartz 1954) and Georgia (Golley 1962; Figure 2). It is not present in the Blue Ridge of Georgia as previously reported (Laerm and Boone 1995), but has been reported from many of the coastal islands (Laerm et al. 1999). The species is widely distributed in the sand ridges and barrier islands of the peninsula and panhandle of northern Florida, and along eastern coastal regions of southern Florida (Howell 1939, Humphrey 1992a,b; Holler 1992a,b; James 1992, Stout 1992). It ranges throughout much of Alabama, including the barrier islands (Bowen 1968, Wolfe and Rogers 1969, Holliman 1986a,b), and into extreme northeastern Mississippi (Jones and Carter 1989).

ABUNDANCE STATUS

The oldfield mouse is common in appropriate habitat in most inland areas of the distribution, although population densities vary by season and year. In oldfield habitats the Upper Coastal Plain of South Carolina, densities estimates fluctuated seasonally from 2.4–21.0/ha (Davenport 1964), and from 4.9–50.1/ha (Caldwell 1964). Generally, population densities tend to be highest in late fall, winter, and early spring, and lowest in summer. Total populations numbers for each of the Choctawhatchee beach mouse, Anastasia Island beach mouse, Perdido Key beach mouse, and St. Andrew beach mouse subspecies have been estimated at fewer than 500 individuals (Holler and Rave 1991, Holler 1992a,b; Humphrey 1992b, James 1992). Total population estimates of the Perdido Key beach mouse on Gulf Islands National Seashore after a reintroduction effort steadily increased from 20–97 individuals from 1986 to 1988 (Holler et al. 1989). The southeastern beach mouse appears to be relatively stable in the northern areas of the range, but rare or extirpated from the southern areas (Stout 1992, Mota et al. 2001). Extine and Stout (1987) reported the mean number of southeastern beach mice known alive on a 1.44 ha study site on Merritt Island, Florida to range from 19.8–93.7 over a 13-month period. Although the Alabama beach mouse has been extirpated from much of its former range, the remaining populations appear stable (Holler and Rave 1991).

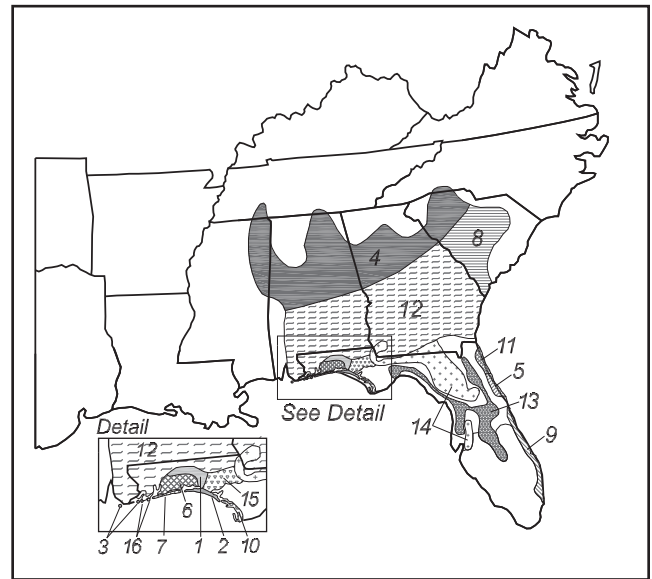


Figure 2. Distribution of *Peromyscus polionotus* in the South: (1) *P. p. albifrons*; (2) *P. p. allophrys*; (3) *P. p. ammobates*; (4) *P. p. colemani*; (5) *P. p. decoloratus*; (6) *P. p. griseobracatus*; (7) *P. p. leucocephalus*; (8) *P. p. lucubrans*; (9) *P. p. niveiventris*; (10) *P. p. peninsularis*; (11) *P. p. phasma*; (12) *P. p. polionotus*; (13) *P. p. rhoadsi*; (14) *P. p. subgriseus*; (15) *P. p. sumneri*; (16) *P. p. trissyllepsis*.

PRIMARY HABITATS

Mainland populations of the oldfield mouse are most common in the herbaceous stage of oldfield succession. They occur in variety of early successional habitats including fallow fields, young pine plantations, roadsides, and edges of cultivated fields, with vegetation characterized by broomsedge (*Andropogon* spp.), asters (*Aster* spp.), lespedeza (*Lespedeza* spp.), and grasses (Schwartz 1954, Davenport 1964, Golley et al. 1965, Thurmond and Miller 1994, Yates et al. 1997). Typical oldfield mouse habitat occurs on well-drained, sandy soils, but they also occur on clay and loamy soils (Coleman 1948). Optimal habitat for insular populations (referred to as beach mice) is generally considered primary and secondary dunes with vegetation characterized by sea oats (*Uniola paniculata*), beach grass (*Panicum amarum*), and bluestem (*Andropogon maritimus*; Holliman 1983, Holler et al. 1989, Holler and Rave 1991). They also occur in the scrub/transition zone habitat characterized by oaks (*Quercus* spp.), pines (*Pinus* spp.), saw palmetto (*Serenoa repens*), wax myrtle (*Myrica cerifera*), sea grape (*Coccoloba uvifera*), and rosemary (*Ceratiola ericoides*; Extine and Stout 1987, Holler and Rave 1991). Scrub/transition habitat provides important refugia for beach mice during hurricanes and other significant storm events (Swilling et al. 1998). Both mainland

and beach populations are primarily nocturnal and utilize simple burrows consisting of an entrance on a slope, a nest chamber, and an escape tunnel rising from the nest chamber (Hayne 1936, Blair 1951, Wolf and Esher 1978).

REPRODUCTION

Breeding can occur year-round in most populations (Davenport 1964, Rave and Holler 1992), but with seasonal peaks depending on location. Inland populations are reported to have peaks in fall and spring with lower reproduction in summer and winter (Golley 1962, Caldwell and Gentry 1965). However, Davenport (1964) reported the highest pregnancy rates from February to July, with an additional peak in September and October in South Carolina. Peaks in island populations have been reported to occur in fall and winter (Blair 1951, Rave and Holler 1992) and fall and spring (Holler 1992a). Gestation is 23–24 days (Layne 1968). Litter size ranges from 1–8 with reported means between 3.1–4.1 (Caldwell and Gentry 1965, Kaufman and Kaufman 1987). Both sexes may be reproductively active at 6–7 weeks (Blair 1951). Maximum reported longevity in the wild is 14 months (Caldwell 1964).

FOOD HABITS

Gentry and Smith (1968) list over 50 species of plant and 10 animal food items found at oldfield mouse burrow entrances and in nest cavities in South Carolina, Georgia, and Florida. Plant material consisted of seeds, seed shells, acorns, and nuts of a variety of trees, shrubs, legumes, forbs, and grasses. The majority of animal food items were insects. Foods from stomach contents reported by Caldwell (1964) from South Carolina included 74% seeds, 26% animal matter, and trace amounts of green plant material. Seeds consisted of lespedeza, buttonweed (*Diodia* spp.), milkpea (*Galactia* spp.), and cowpea (*Vigna* spp.); animal foods consisted of beetle and beetle larvae, flies and fly larvae, caterpillars, spiders, crickets, grasshoppers, and hymenopterans. Alabama beach mouse diets varied seasonally but generally consisted of bluestem, evening primrose (*Oenothera humifusa*), dune spurge (*Chamaesyce ammannioides*), jointweed (*Polygonella gracilis*), sea oats (*Uniola paniculata*), toad flax (*Linaria floridana*), beach pea (*Galactia* spp.), and pennywort (*Hydrocotyle bonariensis*; Moyers and Holler 1997).

ASSOCIATED SPECIES

A variety of invertebrates and vertebrate burrow associates have been reported (Gentry and Smith 1968). Common vertebrate burrow associates include

the gopher frog (*Rana capito*), six-lined racerunner (*Cnemidophorus sexlineatus*), coachwhip (*Masticophis flagellum*), corn snake (*Elaphe guttata*), and least shrew (*Cryptotis parva*). Other small mammal species that occupy the same habitats include the house mouse (*Mus musculus*), eastern harvest mouse (*Reithrodontomys humulis*), hispid cotton rat (*Sigmodon hispidus*), cotton mouse, and white-footed mouse (Caldwell 1964, Briece and Smith 1973, Yates et al. 1997). Potential predators include domestic cats (*Felis catus*), red fox (*Vulpes vulpes*), coachwhip, striped skunk (*Mephitis mephitis*), long-tailed weasel (*Mustela frenata*), and northern raccoon (*Procyon lotor*; Bowen 1968, Humphrey and Barbour 1981, Holler et al. 1989, Holler and Rave 1991).

VULNERABILITY AND THREATS

In general, mainland populations of the oldfield mouse are widespread and abundant, with limited threats to their survival. Populations of the numerous subspecies of the beach mouse, however, are among the most highly threatened and endangered mammals in the region. Remaining populations of beach mice typically are small and isolated, and thus vulnerable to extinction from habitat loss due to coastal development, direct mortality during tropical storms, predation by domestic cats, competition with house mice, and loss of genetic diversity (Bowen 1968, Holliman 1983, Briece and Smith 1973, Humphrey and Barbour 1981, Holler et al. 1989, 1992a; Humphrey 1992b, Rave and Holler 1992, Swilling et al. 1998).

MANAGEMENT GUIDELINES

The United States Fish and Wildlife Service recommends a three-phase program for recovery of beach mouse populations that includes maintenance and restoration of suitable habitat, reestablishment of extirpated populations, and public education of beach mouse conservation efforts (Holler 1992a,b). Reestablishment of extirpated populations appears to be a viable management strategy to increase genetic diversity and reduce the risk of extinctions where suitable habitat is available (Holler et al. 1989). Protection of remaining suitable habitat is critical for the long-term persistence of all populations (Holler and Rave 1991). In addition to primary dune areas, scrub transition habitat provides important refugia for beach mice during hurricanes and should be considered in conservation plans (Swilling et al. 1998).

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Florida Mouse

Podomys floridanus (Chapman, 1889)

Steven B. Castleberry and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

The Florida mouse (*Podomys floridanus*) was described by Chapman (1889) as *Hesperomys floridanus*. It was subsequently referred to the genus *Peromyscus* by Bangs (1898). Osgood (1909) placed *Peromyscus floridanus* in the subgenus *Podomys*, which Carleton (1980) elevated to full generic rank. No subspecies are recognized. The literature on the Florida mouse is reviewed by Jones and Layne (1993).

DISTINGUISHING CHARACTERISTICS

The Florida mouse closely resembles members of the genus *Peromyscus*. The pelage of adults is brownish to brownish-gray on the dorsum and orange-buff on the cheeks, sides, and shoulders. The feet and under parts are white, but often with a tawny patch on the breast. The pelage is soft and silky. The tail is indistinctly bicolor and about 80% of the body length. Measurements are: total length, 166–220 mm; tail, 70–101 mm; hind foot, 21–28 mm; ear, 16–23 mm; weight, 20–47 g. The Florida mouse is distinguished from sympatric *Peromyscus* (*P. gossypinus* and *P. polionotus*) by its larger size and 5 plantar tubercles on the feet, in contrast with 6 plantar tubercles in *Peromyscus*. *Podomys* is reported to have a skunk-like odor (Layne 1990). The dental formula is I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). See keys for details.

CONSERVATION STATUS

The Florida mouse has a global rank of Vulnerable (NatureServe 2007). The Florida Natural Heritage Inventory Program also classifies it as Vulnerable. The conservation status is reviewed by Layne (1992).

DISTRIBUTION

The species has one of the smallest distributions of any North America mammal, being restricted to the northern two-thirds of peninsular Florida and an isolated region of Franklin County in the eastern panhandle (Layne 1992, Jones and Layne 1993; Figure 2). Its distribution is characterized as patchy, reflecting the distribution of vegetative associations to which it is generally restricted (Layne 1992).

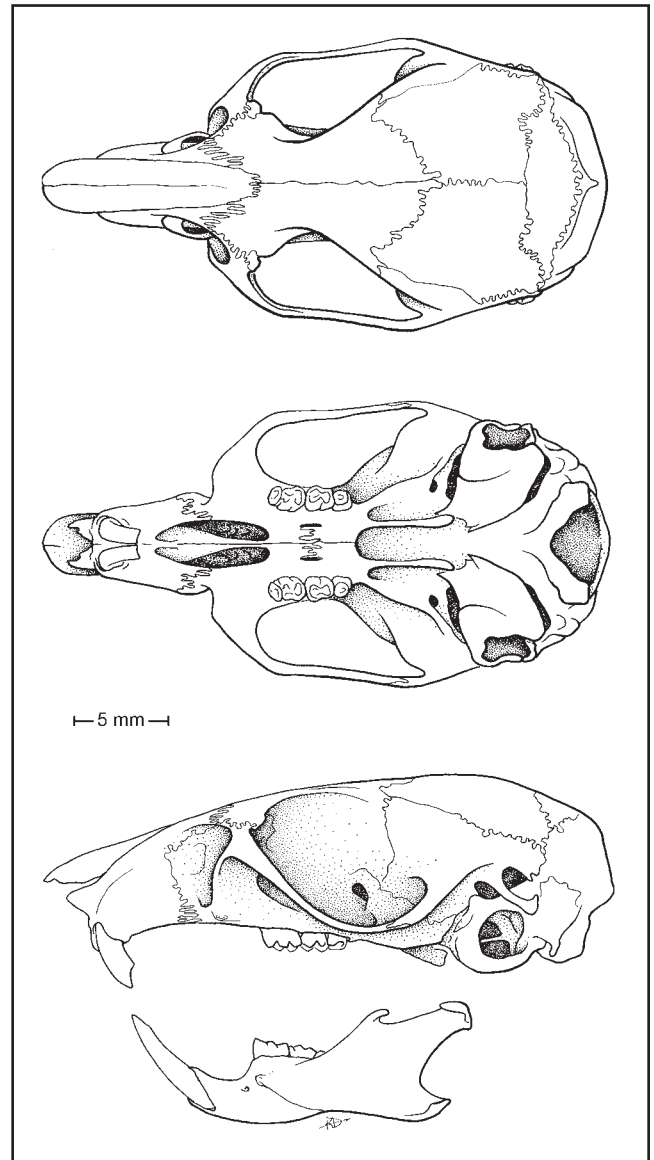


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Podomys floridanus* from Hernando County, Florida (USNM 248772, male).

ABUNDANCE STATUS

The Florida mouse is relatively common in preferred habitat. Population density estimates range from 1.6–28.0/ha (Layne 1992). Higher densities typically occur in sand pine (*Pinus clausa*) scrub habitat than in pineland habitat dominated by longleaf pine (*P. palustris*) and turkey oak (*Quercus laevis*; Layne 1990).

PRIMARY HABITATS

The Florida mouse occurs in xeric upland habitats with fire maintained vegetational communities and well-drained soils (Layne 1992). The primary habitats are sand pine scrub, scrubby flatwoods, and sandhills (longleaf and slash pine (*Pinus elliotii*)-turkey oak). Other habitats include coastal scrub, upland hammocks, and dry pine flatwoods. Layne (1992) suggests that scrub was the historic habitat for the species, and that sandhill habitat became more generally occupied after conversion to a drier, more open condition by human disturbance. Larger Florida mouse densities in scrub and scrubby flatwood habitats, compared to sandhill habitats, are related to higher and more consistent annual acorn production (Layne 1990).

REPRODUCTION

The Florida mouse may breed year-round (Dice 1954, Rood 1966, Drickamer and Vestal 1973), but most breeding occurs in late fall and winter (Layne 1966). Length of gestation is unknown, but is probably similar to most *Peromyscus* at about 23–24 days (Layne 1992). Average number of litters/year is 2.4 (Rood 1966). Litter size ranges from 1–5 with means reported to range from 1.7 (Rood 1966) to 3.4 (Layne 1966). Young are weaned at 3–4 weeks (Layne 1966). Reports of the average life span vary from 2–4 months (Layne 1990), but individuals may live more than a year in the wild (Jones 1990).

FOOD HABITS

The Florida mouse is an opportunistic feeder (Jones 1993), but acorns probably are the primary food source when available (Humphrey et al. 1985). Jones (1993) found turkey oak acorns to be the least preferred of 6 species offered to captive individuals. Other food items consumed by captive individuals included pine seeds, blueberries (*Vaccinium myrsinites*), deer berries (*V. stamineum*), gall berries (*Ilex glabra*), blackberries (*Rubus argutus*), gopher apples (*Licania michauxii*), pawpaw fruits (*Asimina* spp.), queen's delight (*Stillingia sylvatica*) flowers, and seed pods of *Crotalaria rotundifolia* and *Galactia elliotii*. Invertebrates probably comprise a small part of the diet. Florida mice forage primarily on the ground and appear to show no preference for foraging in open versus densely vegetated areas (Packer and Layne 1991).

ASSOCIATED SPECIES

The Florida mouse is a burrow-dwelling rodent, but they typically do not dig their own burrows. They are most commonly associated with the burrows of

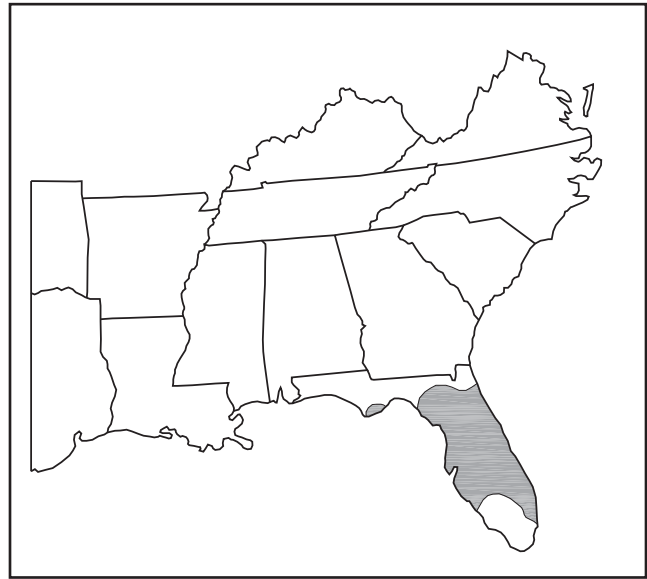


Figure 2. Distribution of *Podomys floridanus* in the South.

the gopher tortoise (*Gopherus polyphemus*; Jones and Franz 1990), but will also use the holes and burrows of the southeastern pocket gopher (*Geomys pinetis*), nine-banded armadillo (*Dasypus novemcinctus*), oldfield mouse (*Peromyscus polionotus*), and hispid cotton rat (*Sigmodon hispidus*; Layne and Jackson 1994). The Florida mouse also occurs in sympatry with the golden mouse (*Ochrotomys nuttalli*) and cotton mouse (*P. gossypinus*; Packer and Layne 1991), although the cotton mouse is usually associated with wetter habitats than the Florida mouse (Jones 1990). Potential predators include bobcat (*Lynx rufus*; Maehr and Brady 1986), northern raccoon (*Procyon lotor*), foxes, raptors, and snakes (Layne 1992).

VULNERABILITY AND THREATS

The Florida mouse has narrow habitat requirements and is susceptible to habitat loss. The well-drained xeric upland scrub and sandhills region of peninsular Florida favored by the Florida mouse are desirable for commercial, residential, and agricultural development. Extensive regions of Florida's scrub and sandhills communities have been converted to other uses, and suppression of fire has resulted in successional changes that have reduced or eliminated *Podomys* in remaining habitats (Layne 1992).

MANAGEMENT SUGGESTIONS

Although prescribed fire appears to have few short-term effects on Florida mouse populations (Jones 1992), long-term fire exclusion may result in vegetation changes that make habitats less suitable (Layne 1990). Because of their close association

(Jones and Franz 1990), management for gopher tortoise within suitable habitat may provide commensal benefits to the Florida mouse.

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Norway Rat

Rattus norvegicus (Berkenhout, 1769)

Joshua Laerm and Wm. David Webster

CONTEXT AND CONTENT

The Norway rat, *Rattus norvegicus*, was introduced into North America by European colonists and it is likely that these early introductions were assignable to *R. n. norvegicus*. However, introductions persist, resulting in a continued mixing of genetic stocks; therefore, it is moot to recognize subspecies at this time. Much of the literature on the Norway rat deals with its relationship with humans, impact as a pest species, and control (Calhoun 1963, Brooks 1973, Howard and Marsh 1976, Pratt et al. 1977, Jackson 1982, 1990; Meechan 1984, Lund 1994, Timm 1994). In addition, there is an extensive amount of literature about the role of the Norway rat as a vector for diseases that affect humans.

DISTINGUISHING CHARACTERISTICS

Rattus norvegicus is a relatively robust rat with a sparsely haired, scaled tail that is shorter than the head and body length. External measurements are: total length, 320–480 mm; tail, 150–220 mm; hind foot, 34–46 mm; ear, 15–25 mm; weight, 200–500 g. The pelage is short and coarse, grayish brown to brown above and buffy or pale gray below; the tail is indistinctly bicolored. The dental formula is $I\ 1/1$, $C\ 0/0$, $P\ 0/0$, $M\ 3/3 = 16$ (Figure 1). The upper molars have cusps arranged in three longitudinal rows. The superorbital and temporal ridges are distinct. The Norway rat may be confused only with the black rat, *R. rattus*, but the former is distinguished by its shorter tail, parallel temporal ridges, and parietal bones that are shorter than the distance between the temporal ridges. See keys for details.

CONSERVATION STATUS

The Norway rat has a global rank of Secure (NatureServe 2007). A conservation status rank of Not Applicable has been assigned for each state in the South where it occurs; the species is not a suitable target for conservation activities.

DISTRIBUTION

The Norway rat occurs throughout the South in association with humans and their structures, essentially anywhere it can find food and shelter (Figure 2). Representative records are from Virginia (Handley and

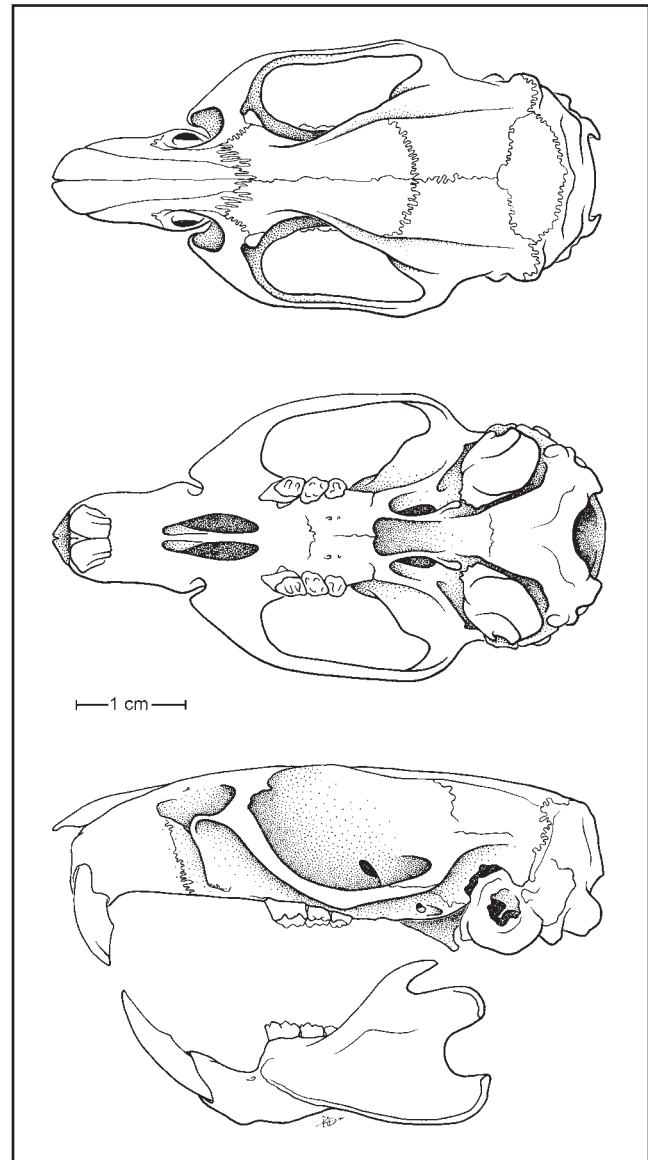


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Rattus norvegicus* from Newton County, Missouri (USNM 279722, female).

Patton 1947, Dueser and Porter 1986, Linzey 1998), North Carolina (Lee et al. 1982, Webster 1988), South Carolina (Golley 1966, Sanders 1978, Chamberlain 1979, Schacher and Pelton 1979, Feldhamer et al. 1987, Cothran et al. 1991), Georgia (Golley 1962, Neuhauser and Baker 1974, Laerm et al. 1982), and Florida (Schwartz 1952, Layne 1974), including most of the

coastal barrier islands of these states. It ranges west throughout Alabama (Holliman 1963, Wolfe and Rogers 1969, Linzey 1970), Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989), Louisiana (Goertz and Long 1973, Lowery 1974), north throughout Tennessee (Howell and Conaway 1952, Conaway and Howell 1953, Linzey and Linzey 1971, Smith et al. 1974, Kennedy 1991) and Kentucky (Barbour and Davis 1974), and west throughout Arkansas (Sealander and Heidt 1990) into Texas (Schmidly 1983, Davis and Schmidly 1994) and Oklahoma (Caire et al. 1989).

ABUNDANCE STATUS

Regional density estimates of commensal, semi-feral, or feral populations are not available. Most faunal reports indicate that Norway rats are relatively common to abundant in and around human structures but rare in natural habitats.

PRIMARY HABITATS

Rattus norvegicus is almost always associated with human habitations, and it is common in urban, suburban, and agricultural situations where it thrives on garbage and stored food sources. In summer it may invade cultivated fields and early stages of oldfield succession. Norway rats are good swimmers; feral and semi-feral populations are known from coastal islands, salt marshes, ditches, sewers, and among flotsam at the water's edge. It rarely inhabits woodlands. The species is colonial, living in groups of 10–12 individuals. Nests are made of shredded materials and located under floors, in walls of buildings, and under trash piles. It is a good burrower and often nests underground.

REPRODUCTION

In favorable habitats, Norway rats will breed throughout the year, producing 6–8 litters/year; however, most breeding activity is concentrated in the spring and fall. Reproductive output is dependent on food supply. Gestation is 21–23 days and litter size ranges from 6–22. Young are weaned by 3 weeks and they reach sexual maturity in 2–3 months. Reproductive information is in Davis (1953), Brooks (1973), Howard and Marsh (1976), Pratt and Brown (1976), Pratt et al. (1977), Jackson (1982, 1990), Meechan (1984), Hayassen et al. (1993), and Lund (1994).

FOOD HABITS

Norway rats are omnivorous and have food preferences that are similar to humans, feeding on grains,

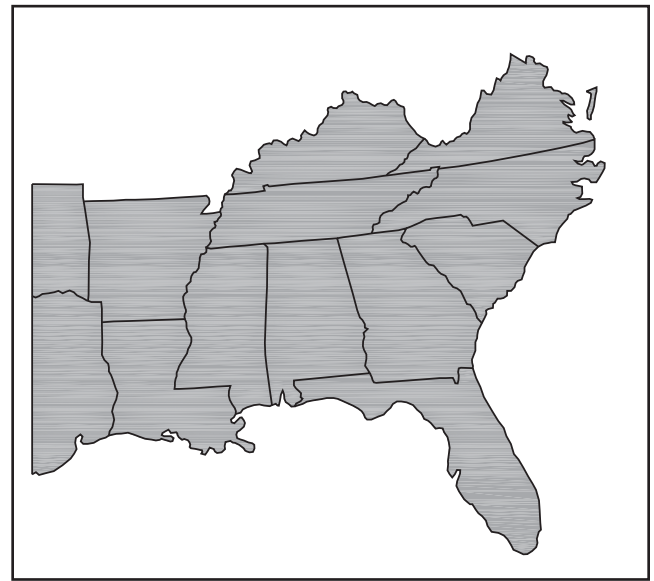


Figure 2. Distribution of *Rattus norvegicus* in the South.

grain products, fruits, and meats. Foods with high fat and protein content apparently are preferred. Eggs, chicks, birds, mice, and small fish are taken when available (Pisano and Storer 1948, Jackson 1965, Nieder et al. 1982).

ASSOCIATED SPECIES

In the South, Norway rats have been found in association with the hispid cotton rat (*Sigmodon hispidus*), eastern woodrat (*Neotoma floridana*), white-footed mouse (*Peromyscus leucopus*), black rat, and house mouse (*Mus musculus*).

VULNERABILITY AND THREATS

The Norway rat is widespread and abundant in commensal situations. Along with the black rat and house mouse, it is one of the most significant pest species in the region, having wide ranging negative public health and economic impacts.

MANAGEMENT SUGGESTIONS

This species warrants no management considerations given its introduced status, the negative economic effect that it has on humans, and the detrimental effect it has on native wildlife.

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Black Rat

Rattus rattus (Linnaeus, 1758)

Joshua Laerm and Wm. David Webster

CONTENT AND TAXONOMIC COMMENTS

Rattus rattus is one of three commensal rodents first brought into North America when the continent was colonized by Europeans. Three subspecies (*R. r. alexandrinus*, *R. r. frugivorus*, and *R. r. rattus*) have been introduced at various times in the past and extensive interbreeding has made it impossible to reliably assign individual specimens to a particular subspecies or to define their respective ranges in North America. Most literature deals with the black rat's relationship to humans, its impact as a pest species, and its control (Brooks 1973, Howard and Marsh 1976, Pratt et al. 1977, Jackson 1982, 1990; Meehan 1984, Lund 1994, Marsh 1994).

DISTINGUISHING CHARACTERISTICS

The black rat has a slender body, pointed snout, and a sparsely haired, scaly tail that is noticeably longer than the head and body length. External measurements are: total length, 320–450 mm; tail, 160–250 mm; hind foot, 27–42 mm; ear, 17–27 mm; weight, 120–350 g. The pelage is short and smooth, brownish to grayish to black above, grading to white to slate gray below, and the tail is not bicolored. The dental formula is $I\ 1/1$, $C\ 0/0$, $P\ 0/0$, $M\ 3/3 = 16$ (Figure 1). The upper molars have cusps arranged in three longitudinal rows and the superorbital and temporal ridges are well developed. The black rat is distinguished externally from the Norway rat (*R. norvegicus*) by its smaller size, slighter build, and longer tail. Cranially, the black rat has temporal ridges that are strongly bowed outward, rather than parallel, such that the length of the parietal bone along the temporal ridge is less than the distance between the temporal ridges. See keys for details.

CONSERVATION STATUS

The black rat has a global rank of Secure (NatureServe 2007). With the exception of Florida and Kentucky, a conservation status rank of Not Applicable has been assigned for each state in the region where it occurs; the species is not a suitable target for conservation activities. Ironically, it was listed as Endangered in Virginia from 1978 (Handley and Gordon 1979) until 1991, based on the assumption that it became rare in the Commonwealth after the Norway rat was introduced into North America. It was deleted from

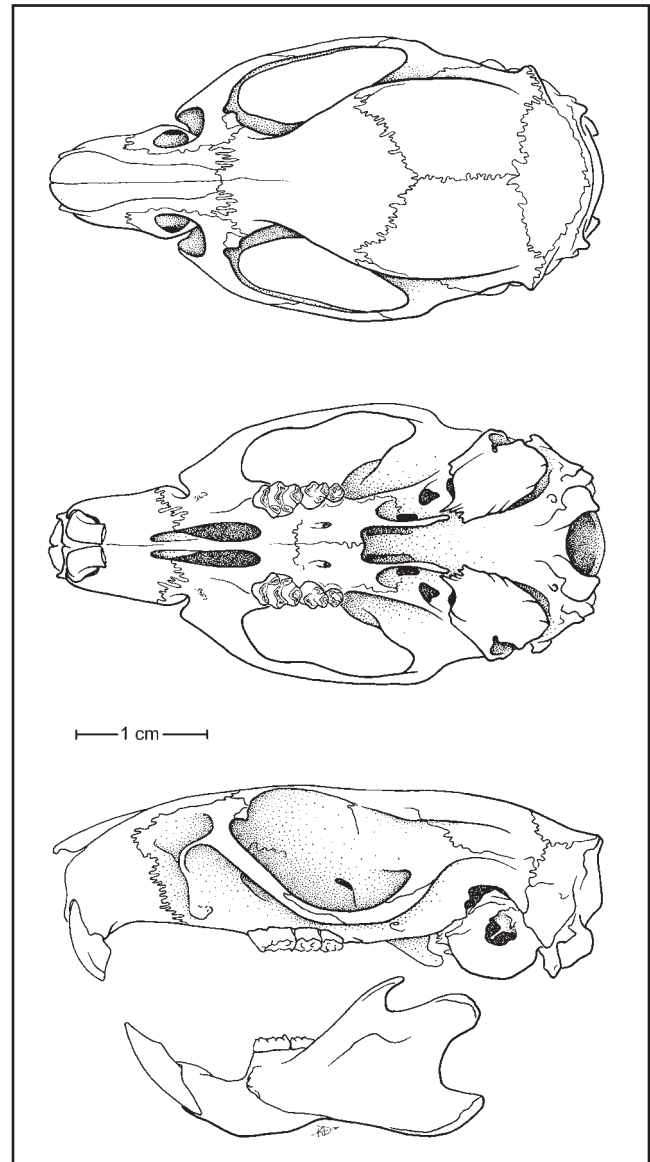


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Rattus rattus* from Leyte Island, Philippines (USNM 458853, male).

Virginia's list of endangered species because "it is not a native, it is a pest, and therefore it should not be sheltered by designating it to be endangered" (Handley 1991).

DISTRIBUTION

The black rat, when compared to the other introduced commensal murids (Norway rat and house mouse, *Mus musculus*), has a more restricted warm temperate geographic distribution because of its intolerance to cold temperatures. It occurs predominantly in coastal areas, particularly ports and adjacent inland shipping and commercial hubs in the southeastern Atlantic and Gulf Coast states (Figure 2). It less commonly occurs as a commensal further inland and only infrequently occurs in natural habitats. Representative records are provided from Virginia (Handley and Patton 1947, Linzey 1998), North Carolina (Lee et al. 1982, Webster 1988), South Carolina (Golley 1966, Sanders 1978, Chamberlain 1979, Cothran et al. 1991), Georgia (Golley 1962, Laerm et al. 1982), and Florida (Rand and Host 1942, Moore 1946, 1949; Worth 1950, Schwartz 1952, Pournelle and Barrington 1953, Layne 1974, Sasso and Gaines 2002), including most of the coastal barrier islands of these states. It ranges west throughout Alabama (Holliman 1963, Linzey 1970), Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989), Louisiana (Goertz and Long 1973, Lowery 1974), north into Tennessee (Linzey and Linzey 1971, Smith et al. 1974, Kennedy 1991) and west throughout Arkansas (Sealander and Heidt 1990) into Texas (Schmidly 1983, Davis and Schmidly 1994) and Oklahoma (Caire et al. 1989).

ABUNDANCE STATUS

Regional density estimates are not available, but most faunal reports indicate that the black rat is relatively common in and around human structures but rare in natural habitats.

PRIMARY HABITS

Although the black rat is considered a commensal rodent, it generally is more common in areas influenced by humans than in areas where it has direct contact with humans. However, black rats will live in the upper stories of buildings and other human habitations, particularly during the cold winter months. The black rat is more arboreal than the Norway rat, often living in trees and vine covered fences; heavily landscaped residential and industrialized areas provide good habitat. It also inhabits riparian zones where it inhabits thick vegetation, grain fields, early stages of oldfield succession, fencerows, and abandoned fields. In areas where it comes into contact with the Norway rat, the latter is dominant, often forcing the former into the attics of buildings.

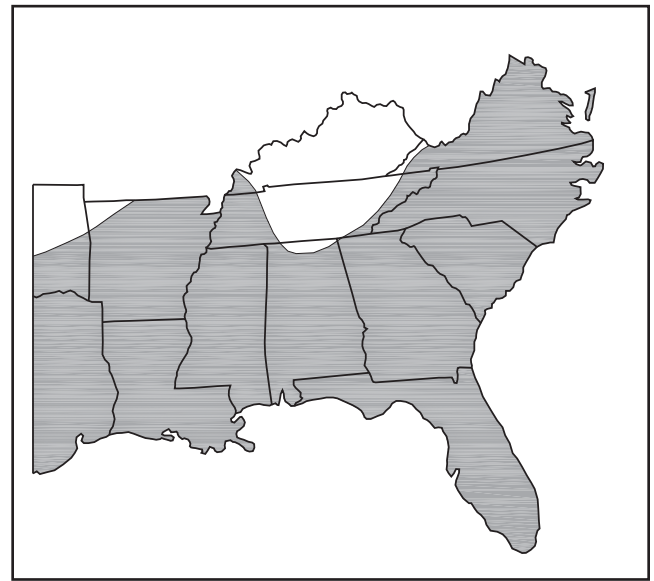


Figure 2. Distribution of *Rattus rattus* in the South.

REPRODUCTION

In favorable habitats, the black rat will breed throughout the year; however, most breeding activity is concentrated in the spring and autumn. Reproductive output is dependent on food supply and climatic conditions. Gestation is 21–23 days, litter size typically ranges from 4–8, and each female produces 4–6 litters/year. Young are weaned in 4–5 weeks and sexually mature in 2–3 months. Extensive reproductive information and additional references are in Davis (1953), Brooks (1973), Howard and Marsh (1976), Pratt and Brown (1976), Pratt et al. (1977), Jackson (1982, 1990), Meechan (1984), Hayassen et al. (1993), Lund (1994), and Marsh (1994).

FOOD HABITS

Although it is basically omnivorous, the black rat feeds extensively on fruits, nuts, grains, and vegetables (Goodyear 1992). Independent of humans, its diet is similar to that of tree squirrels (Jackson 1965).

ASSOCIATED SPECIES

The black rat is found commonly in association with the marsh rice rat (*Oryzomys palustris*), white-footed mouse (*Peromyscus leucopus*), cotton mouse (*P. gossypinus*), eastern woodrat (*Neotoma floridana*), Norway rat, and house mouse.

VULNERABILITY AND THREATS

This relatively widespread rodent is one of the most significant pest species in the region, having wide

ranging negative public health and economic impacts (Ecke 1955).

MANAGEMENT SUGGESTIONS

This species requires no management, as it is an introduced species that has detrimental effects on humans (Morlan et al. 1952).

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Fulvous Harvest Mouse

Reithrodontomys fulvescens (Allen, 1894)

Joshua Laerm and Michael T. Mengak

CONTENT AND TAXONOMIC COMMENTS

Seventeen subspecies are recognized by Hall (1981) and Spencer and Cameron (1982). A single subspecies occurs in the South, *R. f. aurantius*. Spencer and Cameron (1982) review the literature along with a key to North American *Reithrodontomys*.

DISTINGUISHING CHARACTERISTICS

The fulvous harvest mouse is a medium-sized mouse with a long tail (10–50% longer than body). Standard measurements are: total length, 134–189 mm; tail, 72–116 mm; hind foot, 16–22 mm; ear, 11–17 mm; weight, 10–12 g. The upper pelage is reddish brown and finely grizzled or streaked with black; the sides are cinnamon to yellow; venter is pale buff to whitish; tail is not sharply bicolor. It has grooved upper incisors and the folds of the third upper molar extend more than halfway across the crown of tooth; the worn surface of third lower molar is S-shaped. The dental formula of the fulvous harvest mouse is $I\ 1/1, C\ 0/0, P\ 0/0, M\ 3/3 = 16$ (Figure 1). See keys for details.

CONSERVATION STATUS

The fulvous harvest mouse has a global rank of Secure (NatureServe 2007). It is considered Secure in Texas and Apparently Secure in Arkansas, Louisiana, and Oklahoma. It is unranked in Mississippi.

DISTRIBUTION

The species ranges from southern Nebraska and Missouri south through Mexico into Central America (Hooper 1952, Hall 1981, Spencer and Cameron 1982). It is distributed throughout Arkansas (McDaniel et al. 1978, Sealander and Heidt 1990, Tumblison et al. 1992), much of central and eastern Oklahoma (Blair 1939, Goertz 1962, Caire et al. 1989), central and eastern Texas (Baker 1942, Taylor and Davis 1947, Kincaid and Cameron 1982, Schmidly 1983, Cleveland et al. 1984, Spencer and Cameron 1988, Choate et al. 1994), Louisiana except the southeastern tip (Lowery 1974, Abernathy et al. 1985, Martin et al. 1991), and most of southwestern and south central Mississippi (Kennedy et al. 1974, Jones and Carter 1989). The distribution map presented in Figure 2 includes more of Mississippi than that shown by Hall (1981) or Spencer

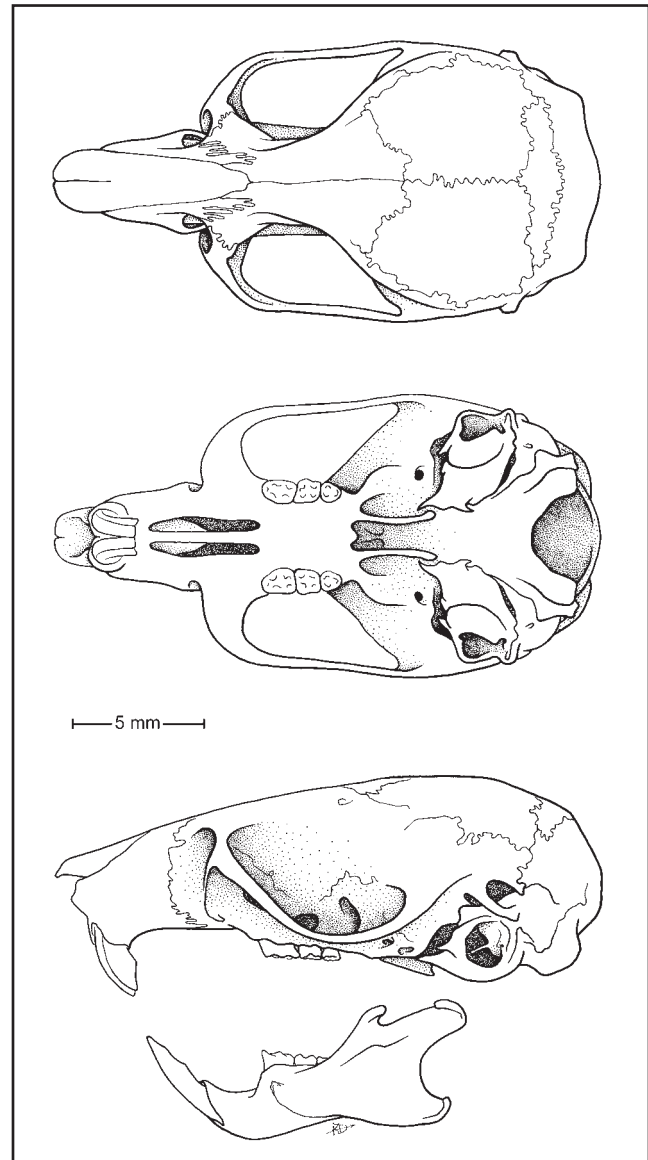


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Reithrodontomys fulvescens* from Kerr County, Texas (USNM 87841, male).

and Cameron (1982), but follows the recent work of Kennedy et al. (1974) and Jones and Carter (1989).

ABUNDANCE STATUS

In Texas, Schmidly (1983) reports the species as widespread and abundant. It exhibits a bimodal

annual density pattern with peaks in late summer and winter and troughs in spring. Density estimates (Packard 1968, Cameron 1977, Waggoner 1978, Wilkins and Schmidly 1980, Grant et al. 1985) range from 4–28/ha in winter, 0.7–13.3/ha in spring, 1–11/ha in summer, and 3–8.7 in fall. Schmidly (1983) indicates these fluctuations are correlated with abundance of preferred nesting vegetation. In Louisiana, it is ubiquitous and abundant, with densities reported as high as 45.3/ha (Shadowen 1956, Lowery 1974). Densities ranging from 1–50/ha were reported in the south-central states (Choate et al. 1994). In Arkansas, the fulvous harvest mouse is widely distributed but seldom attains high population levels (Sealand and Heidt 1990). It is widely distributed in Oklahoma but reported to be uncommon (Goertz 1962, Caire et al. 1989). Density estimates in Texas range from 5–27/ha (Grant et al. 1985).

PRIMARY HABITATS

The fulvous harvest mouse inhabits grassland and shrub communities. Hooper (1952) described the habitat to include rock outcrops, cactus or brush. In Texas it occurs in broomsedge-pine wood ecotones (Taylor and Davis 1947), bluestem and grassy fields (Hooper 1952), marshy areas along brushy and weedy fencerows (Baker 1942), briar thickets along cultivated fields (Bailey 1905), coastal prairies (Spencer and Cameron 1988), and unmanaged highway rights-of-way (Wilkins and Schmidly 1980). In north-central Texas, *R. fulvescens* achieved its highest abundance in grassland habitats (Hanchey and Wilkins 1998). Windberg (1998) caught this species most often in areas with medium to high shrub density (>1500–4000 stems/ha). Blair (1939) and Caire et al. (1989) note its association with brushy grasslands, prairie and forest-grassland ecotones, and lowland vine and brush thickets in Oklahoma. In Louisiana (Lowery 1974, Abernathy et al. 1985, Martin et al. 1991), it is associated with freshwater and coastal marsh edges, uncultivated fields, briar thickets on woodland borders, and dense tangles of low vegetation along fencerows. Similarly in Arkansas (Sealand and Heidt 1990), it is found in oldfields with dense stands of mixed grass and shrubs, tall grasses such as broomsedge (*Andropogon* spp.), Johnson grass (*Tridens flavus*) and purple top (*Sorghum halepense*), and along fencerows with tangles of grasses, vines and shrubs.

The species typically nests several inches above ground in a round, baseball-sized mass of shredded grasses and forbs. It is often caught in traps placed 1 m above ground. Fulvous harvest mice seem to prefer areas of heavy grass cover when its range overlaps with the plains harvest mouse (*R. montanus*), which

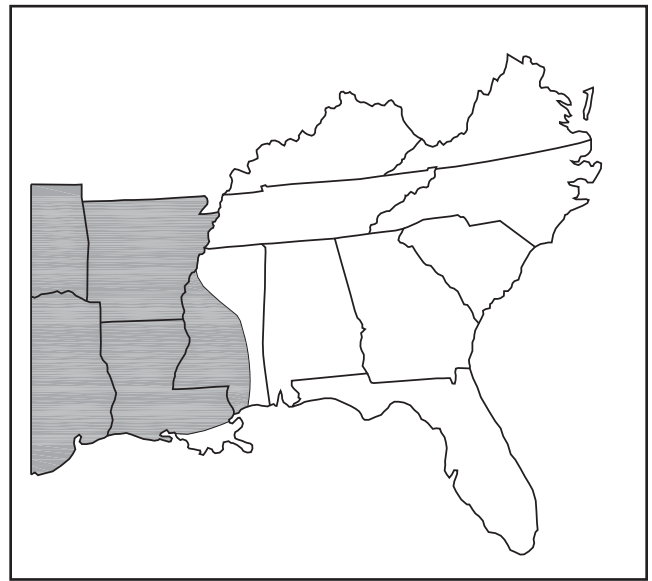


Figure 2. Distribution of *Reithrodontomys fulvescens* in the South.

prefers areas of sparse cover (Goertz 1962). Social behavior in the fulvous harvest mouse is poorly studied. It is seldom active during daytime and the home range has been reported at around 0.1 ha (Lowery 1974).

In north-central Texas, relative abundance of fulvous harvest mice was positively correlated with amount of dead vegetative material at 5, 10, and 25 cm heights; with Bermuda grass at 25 and 50 cm height; and with Johnson grass at 25 cm height. Relative abundance was negatively correlated with woody cover at 50 cm and > 1 m in height. There was also a significant correlation between fulvous harvest mice and stem count of sumac (*Rhus* spp.) and biomass of greenbrier (*Smilax* spp.). The fulvous harvest mouse was one of the most predominant rodents in grassland communities and its relative abundance was negatively correlated with woody material (Hanchey and Wilkins 1998).

REPRODUCTION

The breeding season extends from February to October throughout its range; it is markedly bimodal, with a peak in late spring-early summer and another in late summer-early autumn (Packard 1968, Cameron 1977). Gestation is 21–23 days. Litter size ranges from 1–7 (Svihla 1930, Cockrum 1952, Goertz 1962, Cameron 1977) with an average of 3.5 (Cameron 1977). Young are weaned by 13–16 days and become sexually mature at 2–3 months. Spring young may breed in fall. Longevity averages 2–3 months, up to a maximum of 14–15 months for males and 11–12 months for females (Spencer and Cameron 1982).

FOOD HABITS

Few studies have examined the diet of the fulvous harvest mouse, but seasonally it relies heavily on insects. In Arkansas, invertebrates dominate the diet in spring and summer while seeds predominate in fall and winter (Gaertner 1968). In Texas, however, invertebrates dominate the diet year-round due to their availability (Kincaid 1975). In coastal prairies, the diet consisted of 70% invertebrates and 28% monocot plants (Kincaid and Cameron 1982). However, Lowery (1974) reported that the diet consisted almost entirely of weed seeds but with occasional vegetation.

ASSOCIATED SPECIES

In the South, the fulvous harvest mouse may occur in association with least shrew (*Cryptotis parva*), southern short-tailed shrew (*Blarina carolinensis*), Elliot's short-tailed shrew (*B. hylophaga*), marsh rice rat (*Oryzomys palustris*), western harvest mouse (*R. megalotis*), plains harvest mouse, eastern harvest mouse (*R. humulis*), white-footed mouse (*Peromyscus leucopus*), hispid cotton rat (*Sigmodon hispidus*), and northern pygmy mouse (*Baiomys taylori*). The fulvous harvest mouse and the rice rat segregate on the basis of habitat, whereas the cotton rat and fulvous harvest mouse segregate temporally, spatially, and by food habits. Windberg (1998) found a significant positive correlation between cotton rats and the relative abundance of the species in southwest Texas.

Information on predators is sparse. Fulvous harvest mouse skulls have been identified from pellets of the common barn owl (*Tyto alba*; Goertz and Jimenez 1999) and barred owl (*Strix varia*; Spencer and Cameron 1982), and the stomach of a red-tailed hawk (*Buteo jamaicensis*; Spencer and Cameron 1982).

VULNERABILITY AND THREATS

The fulvous harvest mouse is widespread and occurs in a diversity of grassland and early successional communities. There seem to be no threats to its survival on a local or regional basis.

MANAGEMENT SUGGESTIONS

The fulvous harvest mouse relies on abundant grass cover and early seral habitat with limited amounts of brush and shrubs for climbing and nest sites. Oldfields, fencerows, and ditch banks are known habitat. Young clearcuts may provide habitat but this seral stage is uncommon in much of the species range.

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Eastern Harvest Mouse

Reithrodontomys humulis (Audubon and Bachman, 1841)

Michael T. Mengak and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Three subspecies are recognized, all of which occur in the South (Howell 1940, Hall 1981). Hooper (1943) examined geographic variation and reviewed taxonomy. Discrepancies exist among authors regarding subspecies distribution. In Oklahoma, the subspecies is reported as *R. h. merriami* by Hall (1981) and Stallings (1997), but as *R. h. humulis* by Caire et al. (1989). In Arkansas, *R. h. humulis* inhabits the eastern part of the state and *R. h. merriami* occupies the western part of the state (Sealander and Heidt 1990). The third subspecies, *R. h. virginianus*, occurs in Virginia (Wilson and Ruff 1999). The literature is reviewed by Kaye (1959), Dunaway (1968), Chandler (1984), and Stallings (1997).

DISTINGUISHING CHARACTERISTICS

The eastern harvest mouse is a small-sized mouse with a short tail (less than head and body length). Measurements are: total length, 99–152 mm; tail, 45–73 mm; hind foot, 15–18 mm; ear, 8–9 mm; weight, 5–11 g. Its upper pelage is grayish brown, finely grizzled or streaked with black; the sides are paler, the venter is buffy gray, and the tail is not sharply bicolored. The dental formula is $I\ 1/1, C\ 0/0, P\ 0/0, M\ 3/3=16$ (Figure 1). The upper incisors are grooved and the first primary fold of third upper molar is shorter than the second fold, extending less than halfway across crown of tooth. The worn surface of the left third lower molar is C-shaped. Labial ridges are present on first and second lower molars. See keys for details.

CONSERVATION STATUS

The eastern harvest mouse has a global rank of Secure (NatureServe 2007). It is also Secure in Alabama, Tennessee and Virginia, and listed as Apparently Secure in Georgia, Kentucky, North Carolina, and Texas. Louisiana classified it as Vulnerable, and Arkansas and Oklahoma classify it as Critically Imperiled. It is unranked in Florida, Mississippi and South Carolina.

DISTRIBUTION

The eastern harvest mouse is restricted to the southeastern United States (Figure 2). It ranges throughout

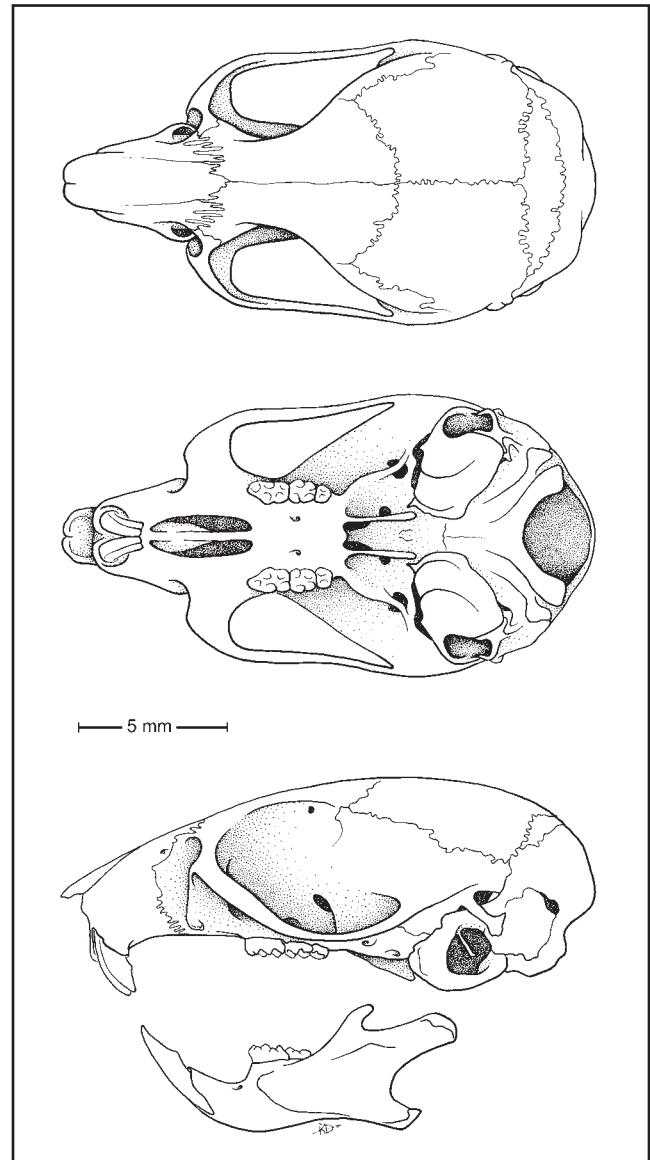


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Reithrodontomys humulis* from South Hampton County, Virginia (USNM 284985, female).

Virginia, except the Eastern Shore (Handley and Patton 1947, Jackson et al. 1976, Handley 1979, 1992; Chandler 1984, Rose 1986, Cawthorn and Rose 1989, Pagels et al. 1992, Mitchell et al. 1993); eastern North Carolina (Kaye 1959, 1961a; Lee et al. 1982, Clark et al. 1985, Adams et al. 1987, Webster 1988, Rose et al.

1990, Clark et al. 1993, Stalling 1997); South Carolina (Golley et al. 1965, Golley 1966, Briese and Smith 1974, Sanders 1978, Schacher and Pelton 1979, Andre 1981, Mengak et al. 1989, Cothran et al. 1991); and Georgia (Golley 1962, Neuhauser and Baker 1974, Atkeson and Johnson 1979, Laerm et al. 1980, 1982; Ford et al. 1994). It occurs north of Lake Okeechobee in Florida (Rand and Host 1942, Hooper 1943, Moore 1946, Layne 1974, Pournelle 1950) and west throughout Alabama (Holliman 1963, Wolf and Rogers 1969, Linzey 1970) and Mississippi (Wolfe 1971, Wolfe and Lohofener 1987, Jones and Carter 1989). Populations in Louisiana are apparently absent in coastal regions and the northcentral and northeastern portions of the state (Lowery 1974, Mullin and Williams 1987). It ranges into eastern Texas (Waggoner 1978, Schmidly 1983, Pitts and King 1988, Wilkins 1991, Davis and Schmidly 1994). The mouse occurs in eastern and western Tennessee (Goodpaster and Hoffmeister 1952, Howell 1954, Dunaway 1968, Linzey and Linzey 1971, Beasley and Severinghaus 1973, Smith et al. 1974, Harvey et al. 1991, Kennedy 1991, Stalling 1997) and eastern and western Kentucky (Barbour and Davis 1974, McGehee-Marsh et al. 1992, Kinser and Meade 1993) but is absent from central Tennessee and central Kentucky (Stalling 1997). Populations are restricted to northeastern and southwestern Arkansas (McDaniel et al. 1978, Tumblison et al. 1988, Sealander and Heidt 1990) and four counties in eastern Oklahoma (Jones and Anderson 1959, Smith 1964, Caire et al. 1989, Sheffield and Chapman 1992).

ABUNDANCE STATUS

Abundance estimates vary considerably throughout the species range depending upon region, habitat, and time of year. In the western portion of its range, it is regarded as uncommon to rare and with scattered populations (Schmidly 1983, Caire et al. 1989, Sealander and Heidt 1990, Kennedy 1991); however, some reports suggest it to be locally common (Tumblison et al. 1988). The species has not been reported in a number of local fauna reports; in cases where it is reported, the mouse is regarded as rare to common. In the Appalachians, it is rare above 600m elevation and absent from higher elevations (Linzey and Linzey 1971, Lee et al. 1982, Ford et al. 1994). Few density estimates are available; 8.75/ha have been reported at two sites in southwestern Virginia (Chandler 1984, Cawthorn and Rose 1989) where a high of 44.4/ha was recorded during fall and winter. Other studies indicate relative abundance as 0.4 captures/100 trap nights in the Upper Coastal Plain of Virginia (Bellows et al. 2001) and 0.41–0.57 captures/100 trap nights in South Carolina Piedmont (Mengak 1987, Menzel et al. 2005).

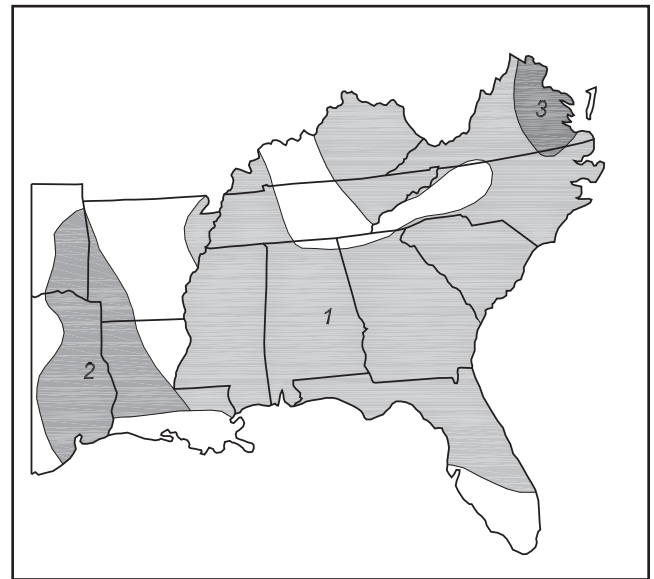


Figure 2. Distribution of *Reithrodontomys humulis* in the South: (1) *R. h. humulis*; (2) *R. h. merriami*; (3) *R. h. virginianus*.

PRIMARY HABITATS

The eastern harvest mouse is associated with early successional communities, pine regeneration areas, and grasslands. It is considered an oldfield specialist (Bellows et al. 2001). Preferred habitats include herbaceous stages of oldfield succession with dense stands of tall grasses, particularly broomsedge (*Andropogon* spp.), fescue (*Festuca* spp.), Johnson grass (*Sorghum halepense*), goldenrod (*Solidago* spp.), briar (*Rubus* spp.) patches, and honeysuckle (*Lonicera* spp.) thickets. The mouse also occurs in cultivated grain fields, weed-filled ditches, brackish meadows, and bottomlands. It will nest above ground in grass clumps, usually in thick cover or on the ground hidden near a grass clump (Stalling 1997). It rarely occurs in forested areas unless considerable open herbaceous areas are available.

Eastern harvest mice are difficult to collect. They have been captured in Victor mouse traps (Balkcom et al. 1996), Sherman live traps (Wilkins 1991), and pitfall traps (Mengak and Guynn 1987). Significantly more individuals were caught in pitfall traps than either Victor mousetraps or Museum Special traps in South Carolina (Mengak and Guynn 1987). Harvest mice were captured most often at trap stations where cotton rats (*Sigmodon hispidus*) occurred. Habitat models have examined the variables of tree height, distance to nearest tree, distance to nearest log, and percent vine cover; tree height was the most important criteria (Mengak and Guynn 2003). Preferred mouse habitat consists of areas with tree and grass cover;

coarse woody debris and dense vegetative cover are less suitable. Harvest mice occur in pine regeneration stands <8 years and in plantations <14 years (Mengak et al. 1989). Home range size averages 952.4 m² (Kaye 1961a, Stalling 1997).

REPRODUCTION

The breeding season is probably year-round with bimodal peaks in March–April and September–October (Layne 1959, Dunaway 1968, Chandler 1984, Cawthorn and Rose 1989). Gestation ranges from 21 (Kaye 1961b) to 24 days (Layne 1959). Litter size ranges from 1–8 with an average of 3.4 (Dunaway 1968). Young are weaned by 2–4 weeks and sexually mature at 10–12 weeks (Layne 1959, Kaye 1959, 1961b; Dunaway 1968). Age at first pregnancy for two females was 11 and 20 weeks (Stalling 1997); intervals between litters for one female varied from 24–49 days. Spring young may breed in fall. Mean life span is 3–4 months (Dunaway 1968, Cawthorn and Rose 1989). Schmidly (1983) suggests that low reproductive potential, coupled with high nest mortality, may account for low densities observed in many areas.

FOOD HABITS

Food habits for the eastern harvest mouse are poorly known. It is considered a granivore/omnivore (Kirkland 1977, Mengak et al. 1989), presumably feeding on seeds of grasses, herbaceous vegetation, and insects. Captive individuals are known to feed upon a diversity of seeds, fruits, succulents, rolled oats, and invertebrates (Layne 1959, Kaye 1959, 1961b).

ASSOCIATED SPECIES

In the South, the eastern harvest mouse may occur in association with least shrew (*Cryptotis parva*), south-eastern shrew (*Sorex longirostris*), northern short-tailed shrew (*Blarina brevicauda*), southern short-tailed shrew (*B. carolinensis*), Elliot's short-tailed shrew (*B. hylophaga*), marsh rice rat (*Oryzomys palustris*), western harvest mouse (*R. megalotis*), plains harvest mouse (*R. montanus*), fulvous harvest mouse (*R. fulvescens*), white-footed mouse (*Peromyscus leucopus*), oldfield mouse (*P. polionotus*), northern pygmy mouse (*Baiomys taylori*), hispid cotton rat, meadow vole (*Microtus pennsylvanicus*), and prairie vole (*M. ochrogaster*). Predators include the common barn owl (*Tyto alba*) and numerous meso-mammals such as bobcat (*Lynx rufus*), red fox (*Vulpes vulpes*) and coyote (*Canis latrans*; Adams et al. 1986, Stalling 1997).

VULNERABILITY AND THREATS

The eastern harvest mouse is widespread and occurs in a diversity of grassland and early successional communities. There seem to be no threats to its survival on a local or regional basis.

MANAGEMENT SUGGESTIONS

The maintenance of grass cover in oldfields benefits habitat of the mouse. Young pine (*Pinus* spp.) plantations with grass and vine cover provide suitable habitat as do old home sites, roadside ditches, and utility rights-of-way.

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Western Harvest Mouse

Reithrodontomys megalotis (Baird, 1855)

Joshua Laerm and Michael T. Mengak

CONTENT AND TAXONOMIC COMMENTS

Hall (1981) lists 17 subspecies, but Webster and Jones (1982) following Hoffmeister and Warnock (1955) recognized 16, which included a subspecies subsequently elevated to species level by Hood et al. (1984). Thus, of the 15 subspecies presently recognized, one occurs in the South, *R. m. dychei*. Webster and Jones (1982) reviewed the literature.

DISTINGUISHING CHARACTERISTICS

The western harvest mouse is a medium-sized mouse with a long tail (i.e., tail length is approximately equal to body length). Body measurements are: total length, 118–170 mm; tail, 55–96 mm; hind foot, 14–20 mm, ear, 10–16 mm; weight, 8–16 g. Its upper surface is buffy to brownish gray, finely grizzled or streaked with black. The sides are cinnamon and the venter is pale gray to whitish. The tail is somewhat bicolored, dark above and light below. As with all North American harvest mice, the upper incisors are grooved. The dental formula is $I\ 1/1, C\ 0/0, P\ 0/0, M\ 3/3 = 16$ (Figure 1). The folds of the upper molars extend less than halfway across crown of the tooth; the worn surface of the third lower molar is C-shaped; there is no labial ridge on lower molars; and the breadth of the braincase is greater than 9.5 mm. Additional measures used to separate sympatric *R. megalotis* and *R. montanus* are provided by Hoofer et al. (1999). See keys for details.

CONSERVATION STATUS

The western harvest mouse has a global rank of Secure (NatureServe 2007). It is classified as Vulnerable in Arkansas.

DISTRIBUTION

Figure 2 depicts the distribution of the western harvest mouse in the South. Regional populations are restricted to northeastern Arkansas, where it is known from the Mississippi Alluvial Plain (Sealander 1954, McDaniel et al. 1978, Sealander and Heidt 1990).

ABUNDANCE STATUS

The species is common in appropriate habitat (Sealander and Heidt 1990); there are no density

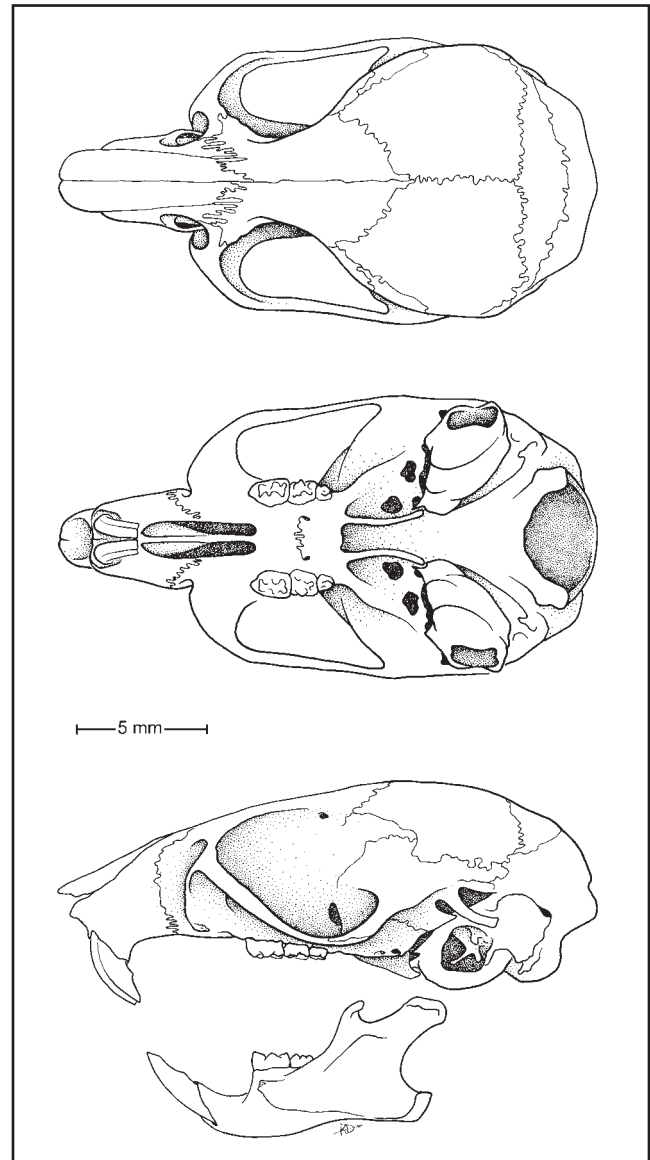


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Reithrodontomys megalotis* from Adair County, Missouri (USNM 548601, female).

estimates available for the South. Species densities of 11.9/ha were reported in Washington (Gray 1943), 4.0/ha in New Mexico, and up to 60/ha following seasonal rains (Whitford 1976). A home range of 3525 m² was measured in California (Meserve 1977). Mice seldom move >50 m within a month's time; however, male and female harvest mice have been recorded

moving distances of 385 m and 413 m, respectively (Skupski 1995).

PRIMARY HABITATS

In Arkansas, harvest mice inhabit oldfields, pastures, meadows, brushy areas bordering cultivated fields, and riparian zones. Areas that contain thick patches of grass bearing large seeds including brome (*Bromus* spp.), fescue (*Festuca* spp.), foxtail (*Alopecurus* spp.), switch grass (*Panicum virgatum*), grama (*Bouteloua* spp.), and bluestem (*Andropogon* spp.) are used (Ford 1977, Sealander and Heidt 1990). Harvest mice are rarely found in forested areas.

REPRODUCTION

In the South, breeding probably occurs year-round and may be concentrated in spring and fall (Hooper 1952, Verts 1960, Jackson 1961). Bancroft (1967) reported 14 litters produced in a single year under captive conditions but seven is typical. After a gestation period of 22–24 days, 1–9 young/litter are produced (Long 1962), with an average of 4.0 (Fisler 1971, Jones 1964). Weaning occurs in 24 days and females are sexually mature by 4 months (Richens et al. 1974, Sealander and Heidt 1990). The proportion of individuals in reproductive condition is highly variable; the largest percentage of reproductively active females occurs in August with a second peak in April (Skupski 1995). Few harvest mice live over a year (Fisler 1971); those individuals that reach 8 months of age generally survive 18–20 months (Skupski 1995).

FOOD HABITS

The species is generally granivorous, feeding on seeds of various grasses and weeds as well as other vegetation; lepidopteran larvae and other insects make up a significant to predominant component of the diet (Whitaker and Mumford 1972). In Idaho, western harvest mice are largely insectivorous from April to August, when arthropods comprise 67–81% of the diet (Johnson 1961). Lepidopteran larvae were detected in 54% and 87% of the mice examined during April and June, respectively (Johnson 1961).

ASSOCIATED SPECIES

The western harvest mouse may occur in association with southeastern shrew (*Sorex longirostris*), least shrew (*Cryptotis parva*), northern short-tailed shrew (*Blarina brevicauda*), Elliot's short-tailed shrew (*B. hylophaga*), deer mouse (*Peromyscus maniculatus*), white-footed mouse (*P. leucopus*), hispid cotton rat (*Sigmodon hispidus*), prairie vole (*Microtus ochrogaster*),

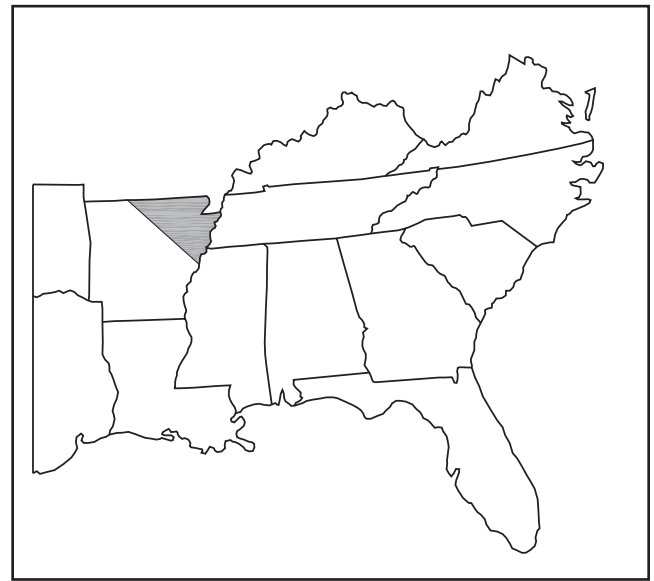


Figure 2. Distribution of *Reithrodontomys megalotis* in the South.

and house mouse (*Mus musculus*; Ford 1977, Mumford and Whitaker 1982, Hoffmeister 1989).

VULNERABILITY AND THREATS

The species occurs at the margin of its range in eastern Arkansas, but it is common in appropriate habitat. There appear to be no threats to the species survival.

MANAGEMENT SUGGESTIONS

The western harvest mouse prefers early successional habitats with thick grass cover, especially large-seeded grasses such as switch grass, foxtail, bluestem, and brome. It occurs in weedy fencerows bordering these fields. Transportation rights-of-way may act as dispersal corridors. Prescribed burning to remove brush and the conversion of fields to native warm season grasses may improve habitat conditions.

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Plains Harvest Mouse

Reithrodontomys montanus (Baird, 1855)

Joshua Laerm and Michael T. Mengak

CONTENT AND TAXONOMIC COMMENTS

Three subspecies are recognized (Bailey 1905, Hooper 1952, Smith 1964, Hall 1981, Wilkins 1986); one of which, *R. m. griseus*, occurs in the South. Wilkins (1986) reviewed the literature.

DISTINGUISHING CHARACTERISTICS

The Plains harvest mouse is a medium-sized mouse with a short tail (i.e., shorter than head and body length). Its measurements are: total length, 95–143 mm; tail, 41–63 mm; hind foot, 14–20 mm; ear, 12–16 mm; weight, 5–15 g. This species may be shorter in the South, with a total length usually < 130 mm and tail shorter than the head and body (Caire et al. 1989). The upper surface is characterized by a dark mid-dorsal stripe grading into gray to buffy grizzled sides; venter pale gray to whitish; tail slender and marked bicolored, dark above light below. Upper parts are less tawny than the western harvest mouse (*R. megalotis*). Most specimens lack the distinct dark mid-dorsal stripe and narrow, dark tail stripe commonly found in the western species (Caire et al. 1989). The dental formula is: I 1/1, C 0/0, P 0/0, M 3/3 = 16 (Figure 1). The upper incisors are grooved and the folds of the third upper molar extend less than halfway across the crown of each tooth. The worn surface of the third lower molar is C-shaped; there are no labial ridges on the lower molars, and the breadth of braincase is less than 9.6 mm. See keys for details.

CONSERVATION STATUS

The Plains harvest mouse has a global rank of Secure (NatureServe 2007). It is Secure in Oklahoma, and is Critically Imperiled in Arkansas.

DISTRIBUTION

The species ranges from southwestern North Dakota and eastern Wyoming south throughout the central and southern Great Plains into Texas, New Mexico, and northern Mexico (Hall 1981, Wilkins 1986). It has a marginal distribution in the region (Figure 2). Populations occur throughout Oklahoma (Caire et al. 1989), including the eastern oak-hickory-pine (*Quercus-Carya-Pinus*) communities in the Ouachita Highlands and the oak-hickory communities in the

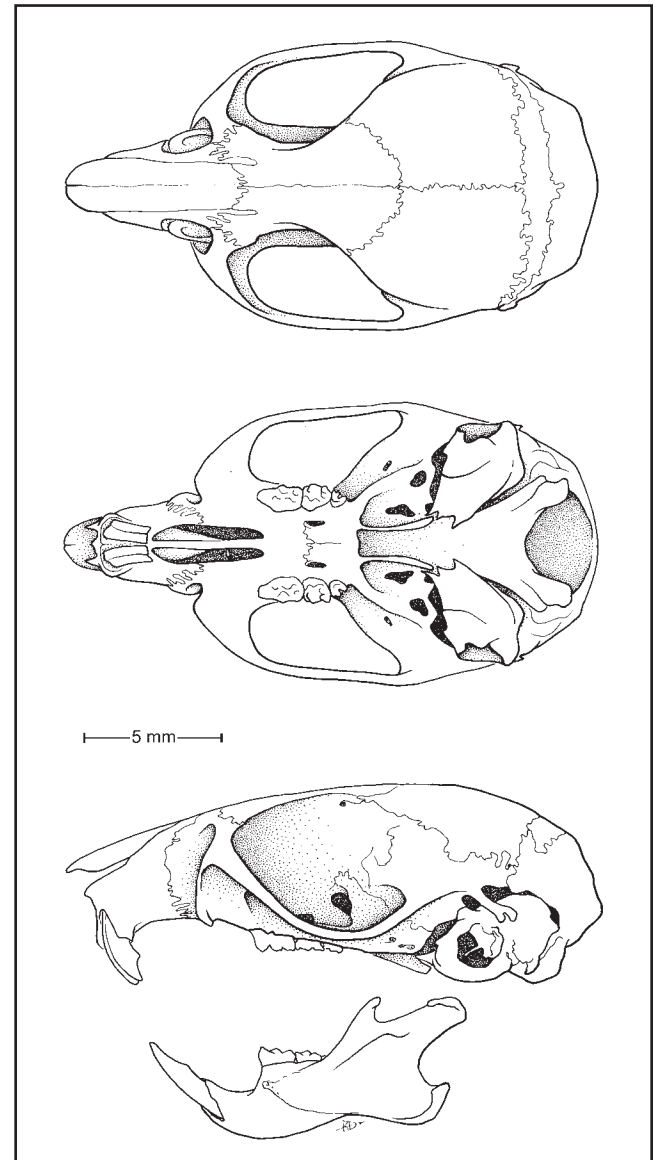


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Reithrodontomys montanus* from Harper County, Oklahoma (USNM 272575, male).

Ozark Plateau. It extends into the Ozark Plateau into northwestern Arkansas (James et al. 1979, Sealander and Heidt 1990) and into extreme Missouri (Long 1961). In eastern Texas, the plains harvest mouse occurs in the blackland prairies and post oak (*Q. stellata*) woodlands; although it has expanded its range into

eastern Texas along vegetated highway rights-of-way (Schmidly 1983, Wilkins 1990), the mouse has not been reported in southern mixed forest communities or coastal prairies (Schmidly 1983, Davis and Schmidly 1994). Schmidly (1983) reported the plains harvest mouse in eight counties of east Texas; Davis and Schmidly (1994) reported it from 11 counties east of the Balcones Fault Zone, which represents a dramatic change in climate, vegetation, soils and fauna from western counties (Schmidly 1983).

ABUNDANCE STATUS

The harvest mouse is at the margin of its range in the region; it is rare to uncommon (Blair 1939, Goertz 1963, Long 1961, Caire et al. 1989, Sealander and Heidt 1990). It is common in prairie-grassland communities to the west and north (Wilkins 1986), where density estimates range from 1.5–6.8/ha (Brown 1946, Waggoner 1975). Density estimates are not available for regional populations. Goertz (1963) collected only 97 individuals in > 72,200 trap nights over a 2-year period in Oklahoma. Home ranges of 0.17 ha (males) and 0.2 ha (females) were reported in Oklahoma (Caire et al. 1989), while home ranges in Texas varied from 0.23–0.84 ha (Schmidly 1983).

PRIMARY HABITATS

The mouse occurs in dry, upland, grassy areas including hay fields, pastures, highway rights-of-way, cultivated sorghum and wheat (*Triticum aestivum*) fields, sand sagebrush (*Artemisia filifolia*), grazed riparian woodlands, and abandoned grassy fields (Blair 1939, Hill and Hibbard 1943, Hall 1955, Goertz 1963, Jones 1964, Maxwell and Brown 1968, Kaufman and Fleharty 1974, Fleharty and Navo 1983, Wilkins 1986). Plains harvest mice are common in shortgrass associations.

REPRODUCTION

In the South, breeding probably occurs year-round in southern portions of the range (Goertz 1963, Davis 1974, Smith 1964, Schmidly 1983); in the northern parts of its range, breeding may be restricted to warmer months (Brown 1946, Goertz 1963, Jones 1964). Gestation is approximately 21 days (Schmidly 1983). Litter size ranges from 1–9 with an average of 4 young (Hall 1955, Goertz 1963, Smith 1964). Weaning occurs within 2 weeks and individuals are sexually mature by 2 months. Maximum longevity is approximately 14 months (Goertz 1963, Waggoner 1975).

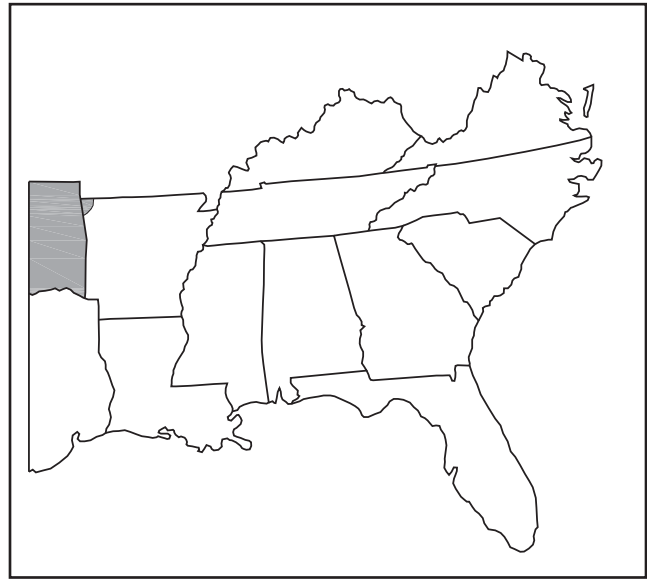


Figure 2. Distribution of *Reithrodontomys montanus* in the South.

FOOD HABITS

The diet consists of insects and the seeds, fruits, and flower heads of various grasses (Brown 1946, Wilkins 1986). Brown (1946) reported that harvest mouse diets in Kansas consisted of grasshoppers and buffalo grass seeds (*Buchloe dactyloides*) and switch grass (*Panicum virgatum*).

ASSOCIATED SPECIES

Regionally, the plains harvest mouse may occur in association with western harvest mouse, fulvous harvest mouse (*R. fulvescens*), eastern harvest mouse (*R. humulis*), deer mouse (*Peromyscus maniculatus*), white-footed mouse (*P. leucopus*), northern pygmy mouse (*Baiomys taylori*), hispid cotton rat (*Sigmodon hispidus*), eastern woodrat (*Neotoma floridana*), and house mouse (*Mus musculus*; Wilkins 1986). Faunal studies indicate the plains harvest mouse is the least abundant element of these small mammal assemblages (Wilkins 1986).

VULNERABILITY AND THREATS

The prairie harvest mouse is widespread and occurs in a diversity of shortgrass prairie communities. It is rare in grassy communities in the forested regions of eastern Oklahoma and northwestern Arkansas and absent from woodlands in eastern Texas. It is rare in the South due to the lack of appropriate habitat. However, other upland grass communities support populations of the species.

MANAGEMENT SUGGESTIONS

Little is known of the life history of this mouse. The maintenance of rights-of-way may promote the growth of grasses. The species has been collected in strip mine areas planted to crimson clover (*Trifolium incarnatum*) and coastal bermuda grass (*Cynodon dactylon*).

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Hispid Cotton Rat

Sigmodon hispidus (Say and Ord, 1825)

Michael T. Mengak and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Twenty-five subspecies are currently recognized (Cameron and Spencer 1981, Hall 1981), nine of which occur in the South: *S. h. exsputus*, *S. h. floridanus*, *S. h. hispidus*, *S. h. insulicola*, *S. h. komareki*, *S. h. littoralis*, *S. h. spadicipygus*, *S. h. texianus*, and *S. h. virginianus*. Several of these have very restricted distributions. Cleveland (1979) and Cameron and Spencer (1981) reviewed the literature.

DISTINGUISHING CHARACTERISTICS

The hispid cotton rat is medium-sized rodent with the following dimensions: total length, 225–350 mm; tail, 80–160 mm; hind foot, 28–40 mm; ear, 16–24 mm; weight, 100–225 g. Its pelage is diagnostic: upper parts are grizzled (hispid salt and pepper), yellowish to brownish gray; the venter is pale gray, sometimes slightly washed in buff; the sparsely haired tail is shorter than the head and body. The dental formula is $I\ 1\ 1/1, C\ 0\ 0/0, P\ 0\ 0/0, M\ 3\ 3/3 = 16$ (Figure 1). The upper molars are flat crowned in lateral view, with cusps forming a sigmoid or S-shape. See keys for details.

CONSERVATION STATUS

The hispid cotton rat has a global rank of Secure (NatureServe 2007). The species is considered Secure in the majority of states in the South. It is considered Apparently Secure in Arkansas and Vulnerable in Kentucky. It is unranked in South Carolina.

DISTRIBUTION

The species occurs widely throughout the southern United States to Central America. Figure 2 depicts the distribution of the hispid cotton rat in the South. It occurs in southern Virginia (Gardner 1946, Handley and Patton 1947, Pagels and Adleman 1971, Jackson et al. 1976, Davis and Barbour 1979, Rose 1986, Rose and Mitchell 1990, Rose et al. 1990, Pagels et al. 1992), including the Great Dismal Swamp region (contrary to Hall (1981) and Cameron and Spencer (1981)); North Carolina (Lee et al. 1982, Clark et al. 1985, Adams et al. 1987, Clark et al. 1993), South Carolina (Golley et al. 1965, Golley 1966, Sanders 1978, Schacher and Pelton 1979, Andre 1981, Cothran et al. 1991, Lidicker et al. 1992), Georgia (Harper

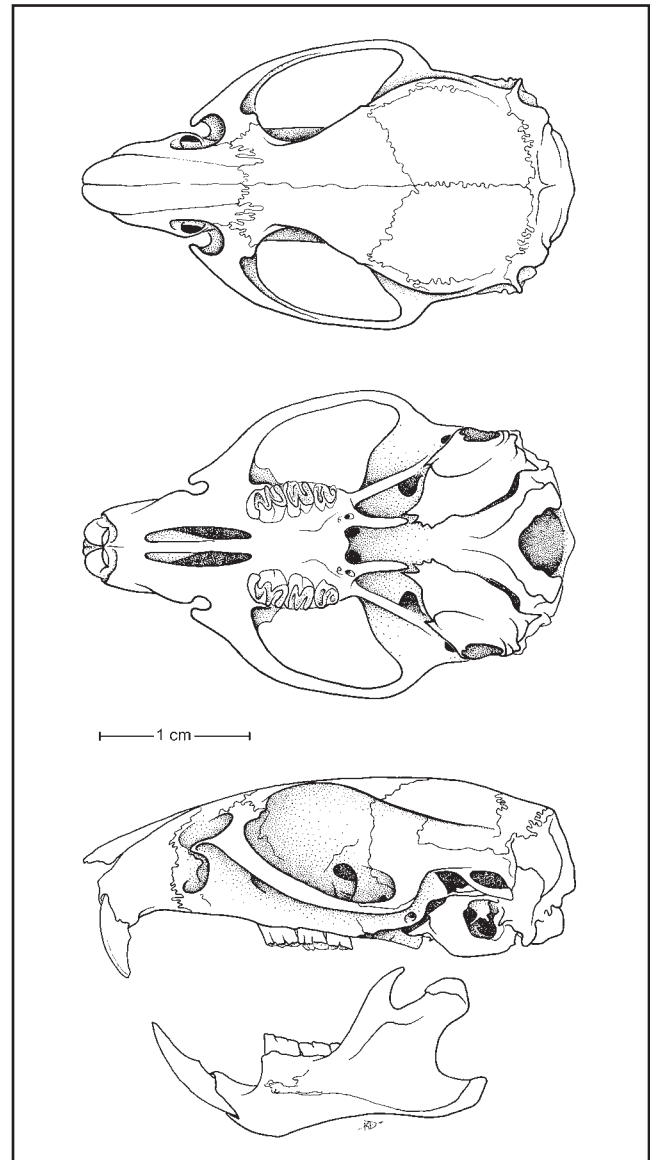


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sigmodon hispidus* from Wakulla County, Florida (USNM 527370, male).

1927, Odum 1955, Golley 1962, Neuhauser and Baker 1974, Atkeson and Johnson 1979, Laerm et al. 1982, Langley and Shure 1980, 1988; Thurmond and Miller 1994), and Florida (Howell 1943, Moore 1946, 1949; Pournelle 1950, Schwartz 1952, Layne 1974a,b; Bigler and Jenkins 1975, Lazell 1978, 1989, Layne 1978, Smith and Vrieze 1979, Stout and Demmer 1982, Sowders and Woodall 1985, Chicardi et al. 1990,

Humphrey 1992, Lips 1991, Packer and Layne 1991), including most of the coastal barrier islands. It ranges west throughout Alabama (Holliman 1963, Wolfe and Rogers 1969), Mississippi (Wolfe 1971, Kennedy et al. 1974, Wolfe and Lohofener 1987, Jones and Carter 1989), Louisiana (Goertz and Long 1973, Lowery 1974, Mullin and Williams 1987), and Arkansas (Sealander and Heidt 1990) into Texas (Schmidly 1983, Grant et al. 1985, Spencer and Cameron 1988, Davis and Schmidly 1994, Cameron 1995, Eshelman and Cameron 1996) and Oklahoma (Caire et al. 1989). It occurs in Tennessee (Howell and Conaway 1952, Howell 1954, Goodpaster and Hoffmeister 1952, Linzey and Linzey 1971, Beasley and Severinghaus 1973, Smith et al. 1974, Harvey et al. 1991, Kennedy 1991) and southeastern and western Kentucky (Robinson and Quick 1965, Barbour and Davis 1974, Davis and Barbour 1979). The species has an expanding range into the Great Plains region (Genoways and Schlitter 1967, Caire et al. 1989), and apparently in eastern Virginia and Kentucky (Davis and Barbour 1974, Pagels and Adleman 1971). It is rare to absent in the higher elevations of the Southern Appalachian Mountains above 900 m (Webster et al. 1985).

ABUNDANCE STATUS

Sigmodon hispidus is one of the most common mammals in the region and is numerous in appropriate habitat. Cotton rats were ranked as first or second in abundance in 6 of 9 studies conducted from 1949–1974 in Florida flatwoods (Layne 1974a). In the southern portion of its range, bimodal annual population fluctuations are often dramatic; to the north, fluctuations may not occur (Cameron and Spencer 1981). Population peaks typically occur in spring and autumn. Depending upon habitat and seasonality, density estimates range from 0.2–20/ha in South Carolina (Golley et al. 1965), 69/ha in Georgia (Odum 1955), 25/ha in Florida (Layne 1974a), 7–10/ha in Louisiana (Lowery 1974), and 3–51 in eastern Texas (Cameron 1977, Schmidly 1983).

PRIMARY HABITATS

The hispid cotton rat occurs primarily in well-drained, grass-dominated habitats where there is a heavy mat of dead vegetation providing cover. Grass height and density are important. It occurs in perennial grasses and forbs of oldfields transportation corridors, brushy fencerows, thickets, pine regeneration areas, and other early successional habitats (Mengak 1999, Mengak and Guynn 2003). Loeb (1999) reported that cotton rats were abundant in tornado-damaged pine forests. Measures of grass cover, foliage height, dry weight of vines, grasses, herbaceous and woody

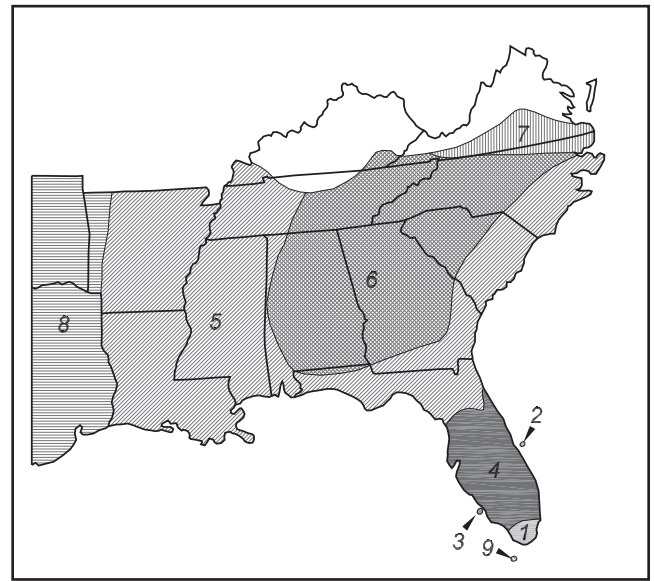


Figure 2. Distribution of *Sigmodon hispidus* in the South: (1) *S. h. spadicipyus*; (2) *S. h. littoralis*; (3) *S. h. insulicola*; (4) *S. h. floridanus*; (5) *S. h. hispidus*; (6) *S. h. komareki*; (7) *S. h. virginianus*; (8) *S. h. texianus*; (9) *S. h. exsputus*.

plants, and pine density were significantly greater at cotton rat capture stations than at non-capture stations; tree height was significantly lower (Mengak and Guynn 2003). This suggests that preferred cotton rat habitat consists of dense vegetation as found in 3–10 year old clearcuts.

Broomsedge (*Andropogon virginicus*), Johnson grass (*Sorghum halepense*), and Bermuda grass (*Cynodon* spp.) are preferred in grass-dominated habitats. In the mid-western portion of the range, fence rows (Clark et al. 1998) and oldfields are favorite habitat due to vegetation heterogeneity and the prevalence of forbs and woody plants (Sietman et al. 1994). In coastal areas, cotton rats are reported from dunes, open woodlands, ponds and marsh edges, and cord grass communities. In the extensive oak-hickory (*Quercus-Carya* spp.) and oak-pine (*Pinus* spp.) areas, they occur in open-canopy, grassy areas. Cotton rats are typically absent from closed canopy, mature forest. In upland sites of the upper Coastal Plain of South Carolina, *S. hispidus* was absent from clearcuts < 6 ha, but common to abundant in those > 10 ha (Yates et al. 1997) whereas in bottomland habitats, open areas (40 m radius) were occupied (Menzel et al. 2005).

REPRODUCTION

Breeding can occur throughout the year; it is markedly bimodal during the spring and fall (Cameron 1977, O'Farrell et al. 1977, Rose 1986). Gestation is

approximately 27 days and litter size ranges from 1–15 (Svihla 1929, Haines 1961, Goertz 1965, Kilgore 1970) with reported means ranging from 3.4–9.0 depending on locality and season (Keys 1958, Goertz 1965, Kilgore 1970, O'Farrell et al. 1977, McClenaghan and Gaines 1978). Individuals become sexually mature at 8–12 weeks (Meyer and Meyer 1944, Chipman 1965).

FOOD HABITS

Cotton rats are primarily herbivorous, feeding on a variety of succulent herbs, sedges, and seed heads of grasses (Cameron and Spencer 1981, Whitaker and Hamilton 1998). Seasonally, a significant portion of the diet includes insects (Fleharty and Olson 1969a,b; Cameron and Spencer 1981). Cotton rats may be important predators on bobwhite quail eggs (*Colinus virginianus*; Stoddard 1931, Trippensee 1948, Hawthorne 1983, Simpson 1976, Whitaker and Hamilton 1998); however, these reports offered no direct evidence of predation. In another study, bobwhite quail eggs were presented to captive cotton rats; there was no evidence to implicate cotton rats as important predators of nests (Ettel et al. 1998). Cotton rats did disturb the nests and may be indirectly responsible for quail mortality. The same study did show that cotton rat predation on songbird nests may be significant.

ASSOCIATED SPECIES

Cotton rats are commonly associated with the marsh rice rat (*Oryzomys palustris*), northern pygmy mouse (*Baiomys taylori*), meadow vole (*Microtus pennsylvanicus*), prairie vole (*M. ochrogaster*), and harvest mice (*Reithrodontomys* spp.). The least shrew (*Cryptotis parva*), southeastern shrew (*Sorex longirostris*), southern short-tailed shrew (*Blarina carolinensis*), white-footed mouse (*Peromyscus leucopus*), cotton mouse (*P. gossypinus*), and golden mouse (*Ochrotomys nuttalli*) also are sympatric with *S. hispidus* (Mengak and Gynnn 2003, Menzel et al. 2005). Cotton rats may behaviorally exclude *Microtus*, especially *M. ochrogaster* (Cameron and Spencer 1981). Cotton rats outcompete with northern pygmy mice for favorable habitat in south Texas (Raun and Wilks 1964).

Cotton rats are important prey for avian predators including the barred owl (*Strix varia*; Whitaker and Hamilton 1998) and barn owl (*Tyto alba*; Goetze and Jimenez 1999). Other predators include the common gray fox (*Urocyon cinereoargenteus*), northern raccoon (*Procyon lotor*; Whitaker and Hamilton 1998), canebrake rattlesnake (*Crotalus horridus atricaudatus*; Platt et al. 2001), and copperhead (*Agkistrodon contortrix*; Whitaker and Hamilton 1998). Barrett et al. (2001) documented predation by the common kingsnake

(*Lampropeltis getula*), timber rattlesnake (*C. h. horridus*), rat snake (*Elaphe obsoleta*), and great horned owl (*Bubo virginianus*). Cotton rats comprised > 31% of bobcat (*Lynx rufus*) diets in Florida (Maehr and Brady 1986) and 46.8% of the annual bobcat diet in eastern North Carolina (Murray et al. 1992). Cotton rats ranked first or second in frequency of occurrence in bobcat stomachs in the Coastal Plain and Piedmont of North Carolina, respectively (King et al. 1983); they ranked first in frequency in winter and spring, second to rabbits (*Sylvilagus* spp.) in summer, and third to rabbits and nutria (*Myocastor coypus*) in autumn in eastern North Carolina (Murray et al. 1992).

VULNERABILITY AND THREATS

The hispid cotton rat is widespread and abundant in appropriate habitat throughout the South. Nowhere does it appear to be vulnerable. Humphrey (1992) suggests that in the Lower Keys and other insular areas, cotton rats are more common and less vulnerable than previously thought.

MANAGEMENT SUGGESTIONS

Rights-of-way maintained in dense grass, vine and herbaceous cover would benefit the hispid cotton rat. Other practices that enhance vegetation cover such as thinning young pine plantations, retaining fencerows and windrows, and establishing brush piles and edges would be beneficial. Medium to large clearcuts with adequate grass and herbaceous vegetation provide excellent habitat.

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Hispid Cotton Rat (*Sigmodon hispidus*)

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Southern Bog Lemming

Synaptomys cooperi (Baird, 1858)

W. Mark Ford and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

There are eight subspecies of the southern bog lemming (*Synaptomys cooperi*) recognized, four of which occur in the South: *S. c. gossii*, *S. c. helaletes*, *S. c. kentucki*, and *S. c. stonei* (Wetzel 1955, Barbour 1956, Hall 1981, Linzey 1983, Long 1987). However, Whitaker and Hamilton (1998) indicate that *S. c. gossii*, *S. c. kentucki*, and *S. c. stonei* could be referable to *S. c. cooperi*. The literature was reviewed by Linzey (1983).

DISTINGUISHING CHARACTERISTICS

The southern bog lemming is a robust, short-tailed vole with a broad head, small ears, and small eyes. Its measurements are: total length, 119–154 mm; tail, 13–25 mm; hind foot, 16–24 mm; ear, 8–14 mm; weight, 20–50 g. The dental formula for this species is: I 1 1/1, C 0 0/0, P 0 0/0, M 3 3/3 = 16 (Figure 1). The pelage is bright chestnut to dark grizzled brown dorsally, grading into silver grayish white ventrally, with gray to brown feet and tail. The southern bog lemming readily is distinguished from other voles by its short tail (usually less than hind foot length), presence of a shallow longitudinal groove along upper incisors, and deep reentrant angles on molars. See keys for details.

CONSERVATION STATUS

The southern bog lemming has a global rank of Secure (NatureServe 2007). It is listed as Secure in Virginia and Apparently Secure in Kentucky and Tennessee. It is listed as Vulnerable in North Carolina, Imperiled in Arkansas, and Critically Imperiled in Georgia. It is unranked in South Carolina.

DISTRIBUTION

The southern bog lemming is distributed throughout southeastern Canada and the north-central and northeastern portions of the United States. Although specimen records have been reported from the Delmarva Peninsula of Maryland (Paradiso 1969), the species is not known from the Virginia portion of the peninsula. There are disjunct populations of *S. c. helaletes* in the Great Dismal Swamp region of Virginia to Carteret and Jones counties, North Carolina, approximately 170 km south (Handley 1979, Clark et al. 1985, Rose 1981, Webster et al. 1984, Rose et al.

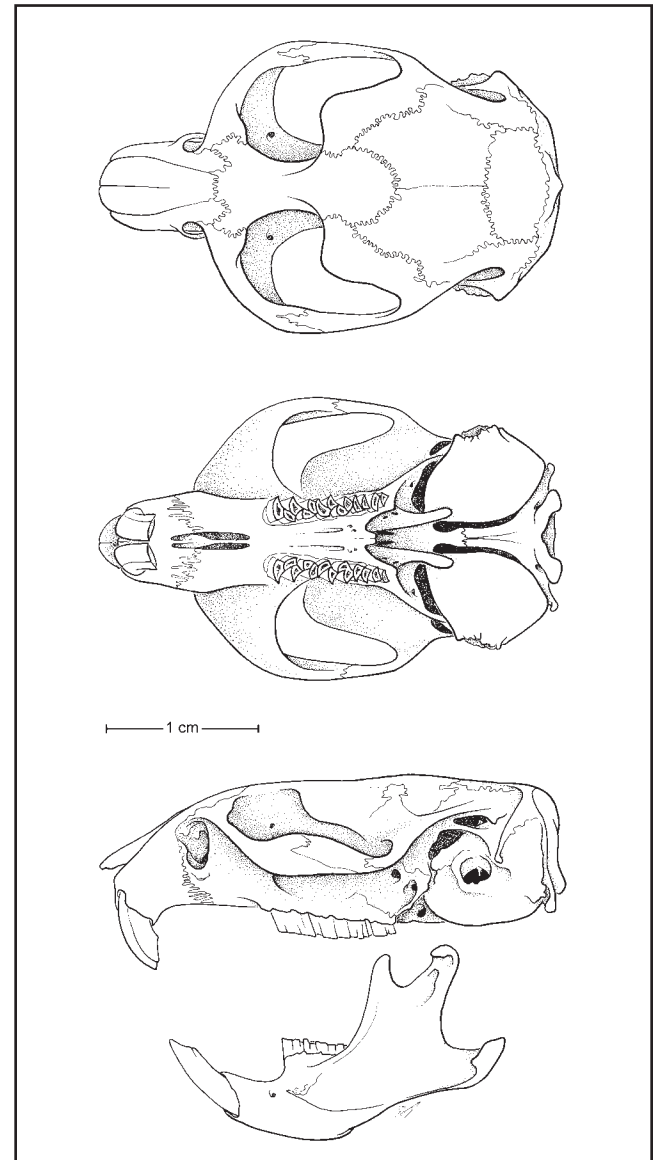


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Synaptomys cooperi* from The Great Dismal Swamp National Wildlife Refuge, Southampton County, Virginia (USNM 95879, male).

1990, Webster et al. 1992, Clark et al. 1993). *Synaptomys c. stonei* occurs at middle to high elevations throughout the Appalachian region of western Virginia (Stewart 1943, Smyth 1946, Linzey and Cranford 1984, Linzey 1984, Handley 1992), eastern Kentucky (Barbour and Davis 1974, Kiser and Meade 1993),

eastern Tennessee (Smith et al. 1974, Linzey 1995), western North Carolina (Odum 1948, Johnston 1967, Lee et al. 1982), and extreme northern Georgia (Brown 1993). Populations in the interior Bluegrass and eastern outer Bluegrass of Kentucky were thought to be *S. c. kentucki* by Barbour (1956), however Robinson (1981) was unable to discriminate between *S. c. kentucki* and *S. c. stonei* there.

Synaptomys c. gossii is found in northeastern Arkansas in the Mississippi Alluvial Valley (Sealander and Heidt 1990; Figure 2).

ABUNDANCE STATUS

The regional abundance of the southern bog lemming is poorly known. The species is difficult to catch and is underreported in small mammal survey efforts even where present (Linzey 1995, Whitaker and Hamilton 1998, Franci 2003). The species may be relatively common in the central Appalachians of Virginia where densities above 20/ha were reported (Linzey and Cranford 1984, Linzey 1984, Kalko and Handley 1993). Generally, in the southern Appalachians, the species is considered rare and patchily distributed (Johnston 1967, Lee et al. 1982, Kiser and Meade 1993, Linzey 1995). In the Kentucky Bluegrass, it is regarded as fairly common (Barbour and Davis 1974); Krupa and Haskins (1996) believed it was once the dominant small mammal in woodland openings and clearings. The southern bog lemming is sporadically distributed in small colonies in Arkansas (Sealander and Heidt 1990). Populations of *S. c. helaletes* may be locally common in the Great Dismal Swamp in Virginia (Rose et al. 1990), whereas Clark et al. (1993) thought the species to be comparatively rare in eastern North Carolina.

PRIMARY HABITATS

In the South, the southern bog lemming has been reported from a wide variety of relatively open habitats such as glades, grassy woodland openings, oldfields, moist meadows, marshes, and shrub bogs (Welter and Sollberger 1939, Conaway and Howell 1953, Barbour and Davis 1974, Smith et al. 1974, Linzey and Cranford 1984, Sealander and Heidt 1990, Krupa and Haskins 1996, Franci 2003). The species also has been reported on high elevation grassy balds, along roadside rights-of-way, and rhododendron (*Rhododendron maximum*) streamside communities in the southern Appalachians (Linzey 1995). Populations in the Coastal Plain of North Carolina and Virginia have been reported from pocosin swamps and young loblolly pine (*Pinus taeda*) plantations and new regeneration areas with thick herbaceous cover (Clark et al. 1985, Rose et al. 1990, Webster et al. 1992, Clark et al. 1993). Southern bog

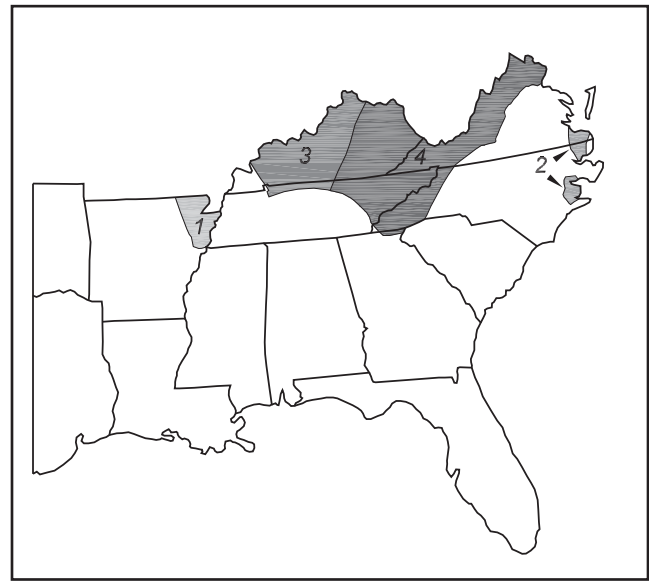


Figure 2. Distribution of *Synaptomys cooperi* in the South: (1) *S. c. gossii*; (2) *S. c. helaletes*; (3) *S. c. kentucki*; (4) *S. c. stonei*.

lemmings construct surface runways and underground burrows. Nests are usually constructed underground; in saturated soils, these may be located under coarse woody debris above ground. Fresh grass cuttings and bright green feces in the runways often indicate the presence of the southern bog lemming (Sealander and Heidt 1990). Home range is typically 0.3 ha or less (Whitaker and Hamilton 1998).

REPRODUCTION

Breeding occurs year-round, but is highest in warmer months and usually is depressed between November and February (Connor 1959, Robinson 1981, Linzey 1983). Gestation is 23–26 days; mean litter size is approximately 4 (Linzey 1983). Young are weaned in 2–3 weeks (Connor 1959). Longevity is 8–12 months (Connor 1959), although individuals living over 2 years have been reported (Whitaker and Hamilton 1998).

FOOD HABITS

Preferred foods of the southern bog lemming are the leaves, shoots, and roots of grasses and sedges, as well as fruits such as blackberries (*Rubus* spp.) or blueberries (*Vaccinium* spp.) and hypogaeal fungi (Hamilton 1941, Connor 1959, Knopf 1978, Linzey 1983, Linzey 1995, Whitaker and Hamilton 1998).

ASSOCIATED SPECIES

The masked shrew (*Sorex cinereus*), northern short-tailed shrew (*Blarina brevicauda*), eastern harvest mouse (*Reithrodontomys humulis*), white-footed mouse

(*Peromyscus leucopus*), deer mouse (*P. maniculatus*), southern red-backed vole (*Clethrionomys gapperi*), prairie vole (*Microtus ochrogaster*), and meadow jumping mouse (*Zapus hudsonius*) are common faunal associates of the southern bog lemming. In the South, the spotty distribution and ephemeral nature of regional populations are suggested to relate, in part, to competitive exclusion by meadow voles (*M. pennsylvanicus*) from areas with dense ground cover. Forest clearing and replacement of native grasses by introduced species apparently favors meadow voles to the exclusion of southern bog lemmings (Linzey 1983, Linzey 1984, Krupa and Haskins 1996). On the other hand, artificial clearings in larger forested areas not readily accessible to meadow voles may favor southern bog lemmings (Linzey 1983, Linzey 1984, Francl 2003).

VULNERABILITY AND THREATS

The southern bog lemming has a wide distribution, but is at the southern limit of its range regionally. In many areas, the species may be in competition with meadow voles. Local populations may be subject to extirpation due to successional change within isolated grassland, wet meadow or early succession habitats.

MANAGEMENT SUGGESTIONS

Maintenance of woodland openings, bogs and moist meadows, and montane grassy balds by periodic mowing, haying, or grazing seems appropriate. In prairie systems, southern bog lemmings showed negative short-term response to prescribed burning, though long-term impacts or benefits were not known (Clark and Kaufman 1990). Connections to habitats that allow expansion or establishment of meadow voles should be avoided.

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Nutria

Myocastor coypus (Molina, 1782)

Joshua Laerm and Wm. David Webster

CONTENT AND TAXONOMIC COMMENTS

A native to South America, the nutria was introduced first into the United States in 1899 and again in the 1930s. Some authorities suggest that these introduced populations are referable to *M. c. bonariensis*; however, individuals of three subspecies from numerous South American localities were brought into the United States at various times and cross-breeding was common to enhance the quality of their pelts, so subspecies designations are irrelevant at this time (Evans 1970). The literature is reviewed by Willner (1982), Kinler et al. (1987), and Woods et al. (1992).

DISTINGUISHING CHARACTERISTICS

This large semiaquatic rodent superficially resembles a beaver (*Castor canadensis*) or muskrat (*Ondatra zibethicus*) except for its long, round, nearly naked tail. It has relatively small eyes and short ears but long whiskers. External measurements are: total length, 850–1050 mm; tail, 300–450 mm; hind foot, 100–150 mm; ear, 20–30 mm; weight, 7–11 kg. Nutria have short, soft, dense underfur and long, coarse guard hairs. The upper parts are yellowish brown to dark brown, the underparts are gray, and the chin and the tip of the muzzle are frequently white to gray. The cranium is distinct; the large infraorbital openings are elongate vertically, the nasals are truncate posteriorly and end anterior to the posterior extent of the premaxillae, and the greatly elongate paraoccipital processes project ventrolaterally. The dental formula is $I\ 1\frac{1}{1}, C\ 0\ 0\ 0, P\ 1\frac{1}{1}, M\ 3\frac{3}{3} = 20$ (Figure 1). See keys for details.

CONSERVATION STATUS

The nutria has a global rank of Secure (NatureServe 2007). A conservation status rank of Not Applicable has been assigned for each state in the South where it occurs; the species is not a suitable target for conservation activities.

DISTRIBUTION

Nutria were introduced in the South, primarily in Louisiana, for use as weed control agents and fur farming, an effort that generally failed due to low pelt prices, low reproductive success, and poor competition with beaver pelts (Evans 1970, Linzey 1998,

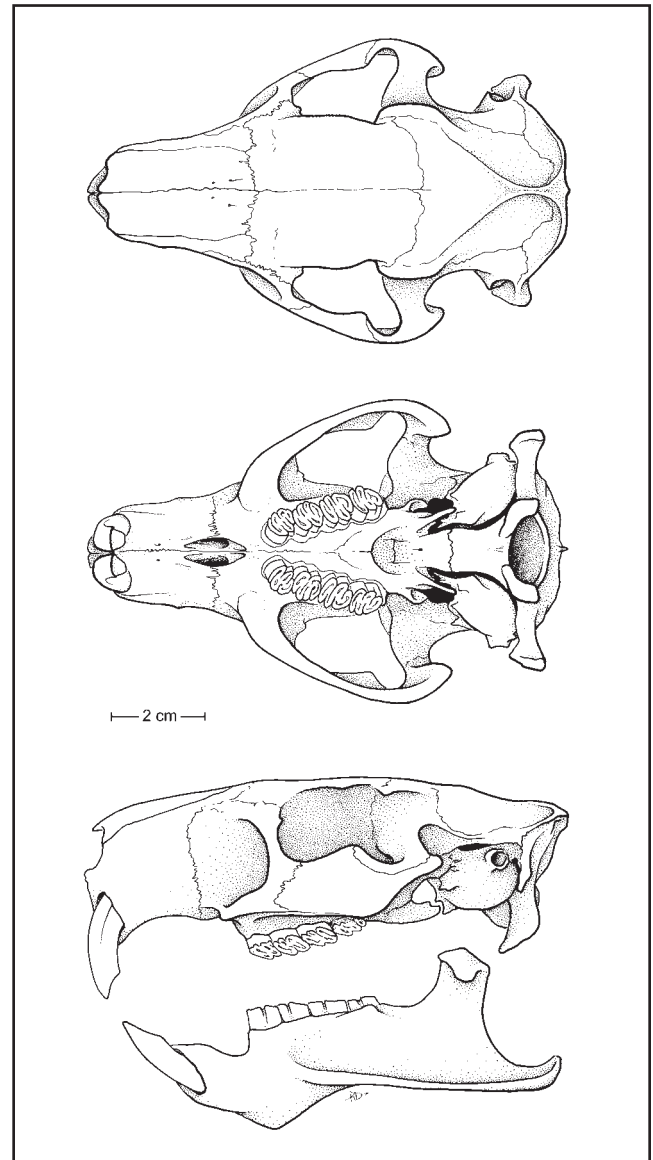


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Myocastor coypus* from St. Charles Parrish, Louisiana (USNM 559317, male).

Carter and Leonard 2002). However, numerous escapes and additional introductions have resulted in the establishment of viable populations throughout as many as 15 states (Evans 1970, Willner 1982, Kinler et al. 1987), mostly in the Gulf Coast region (Figure 2). Nutria occur throughout eastern Texas (Davis 1958, Schmidly 1983, Davis and Schmidly

1994) west to the Trans-Pecos (Hollander et al. 1992), north into southeastern Oklahoma (Dolgos and Earls 1973, Caire et al. 1989), and essentially all of Louisiana (Evans 1970, Lowery 1974, Fuller et al. 1984). Populations are present throughout southern and eastern Arkansas and the Arkansas River valley (Sealander and Heidt 1990), southwestern Tennessee (Kennedy and Kennedy 1998), western and southern portions of Mississippi (Kennedy et al. 1974, Jones and Carter 1989), and extreme southern Alabama and Georgia (Neuhauser and Baker 1974, Laerm et al. 1982). Localized populations occur throughout the panhandle and northern half of peninsular Florida (Griffo 1957, Brown 1975). Populations along the mid-Atlantic Coast currently are distributed in localized coastal areas from the southern portions of Pamlico Sound, North Carolina, northward along both sides of the Chesapeake Bay in Virginia and Maryland (Paradiso 1966, Lee et al. 1982, Webster et al. 1985, Webster 1988, Linzey 1998).

ABUNDANCE STATUS

Nutria are highly gregarious and they may be quite abundant in appropriate habitat. Density estimates vary depending upon such factors as the floral characteristics of the habitat, abundance of aquatic food plants, water levels, and weather (Brown 1975, Willner et al. 1979, Kinler et al. 1987). Density estimates from Florida range from range from 5.9–24.7/ha in unpolluted and nutrient rich ponds, respectively (Brown 1975). Densities range from 0.1–43.7/ha in Louisiana marshes and 0.5–21.4/ha in Maryland marshes, though dependent on season and salinity (Valentine et al. 1972, Willner et al. 1979, Linscombe et al. 1981, Kinler et al. 1987) whereas Texas marsh densities sometimes exceed 120/ha (Simpson and Swank 1979).

PRIMARY HABITATS

The nutria spends most of its life in or near water. Throughout the South, most significant populations inhabit coastal salt marshes, brackish tidal marshes, and estuaries, but they also inhabit a diversity of freshwater habitats including marshes, ponds, lakes, and rivers (Atwood 1950, Milne and Quay 1967, Chabreck 1970, Willner et al. 1979, Linscombe et al. 1981, Nichols and Chabreck 1981, Linscombe and Kinler 1984, Kinler et al. 1987). Nutria occur in the margins of bodies of water where there is abundant aquatic and semiaquatic vegetation. Burrows are constructed in the steep banks, under exposed roots and trees, and in masses of collapsed vegetation. Burrows of muskrats and armadillos (*Dasypus novemcinctus*) also are utilized. Nest platforms of variable size are constructed from available vegetation for use in

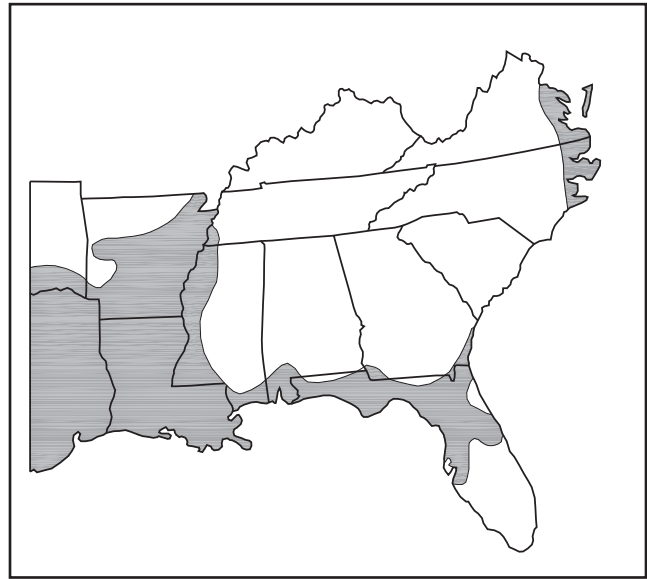


Figure 2. Distribution of *Myocastor coypus* in the South.

feeding, resting, and nesting (Willner 1982, Kinler et al. 1987, Woods et al. 1992).

REPRODUCTION

Breeding occurs continuously and 2–3 litters are produced each year. Gestation is 127–135 days. Litters have 1–11 offspring, with mean litter sizes ranging from 3.9–5.6. Young are precocial and capable of swimming and eating vegetation within 24 hours, but may nurse 7–8 weeks. Individuals reach sexual maturity in 4–7 months (Ashbrook 1948, Atwood 1950, Adams 1956, Newson 1966, Evans 1970, Brown 1975, Willner et al. 1979, Chabreck et al. 1981, Linscombe et al. 1981).

FOOD HABITS

This opportunistic herbivore consumes a wide variety of plants depending on season and habitat. Its diet consists primarily of the stems, leaves, and roots of aquatic vegetation, grasses, and sedges as well as cultivated crops including rice (*Oryza sativa*), soybeans (*Glycine* spp.), and sugarcane (*Saccharum ravennae*) (Atwood 1950, Swank and Petrides 1954, Milne and Quay 1967, Warkentin 1968, Willner et al. 1979, Chabreck et al. 1981, Shirley et al. 1981, Wilsey and Chabreck 1991, Wilsey et al. 1991, Taylor and Grace 1995). Nutria may eat 1.6 kg food/day. Their feeding habits have been described as wasteful of aquatic vegetation because of heavy use of roots compared to stems. Feeding may be so concentrated as to clear areas of emergent vegetation, creating “eat outs” (Harris and Webert 1962).

ASSOCIATED SPECIES

The nutria is associated with mammals that inhabit wetland habitats such as the marsh rice rat (*Oryzomys palustris*), muskrat, northern raccoon (*Procyon lotor*), mink (*Mustela vison*), and northern river otter (*Lontra canadensis*).

VULNERABILITY AND THREATS

In many areas the nutria has been viewed as a pest species due to its impact on agricultural crops, damage done to levees and banks, and overuse of marsh vegetation (Kinler et al. 1987, Bounds 2000, Carter and Leonard 2002). Most states in which nutria occur in high densities manage it as a furbearer. In Louisiana and Texas, where population densities are high, the species has significant local economic value. Elsewhere, most nutria populations are not sufficiently dense to warrant systematic harvest. Exploitation, winter freezes, hurricanes, winter droughts, and alligator predation may have significant impacts on localized populations (Kinler et al. 1987).

MANAGEMENT SUGGESTIONS

The introduction and successful colonization of nutria has upset the ecological balance of native wetland plant and animal species in many areas in the region (Hester et al. 1994, Taylor and Grace 1995, Bounds 2000). Continued research on the ecological impact on native communities, foraging behavior, and competition by this exotic species is warranted.

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Northern Flying Squirrel

Glaucomys sabrinus (Shaw, 1801)

W. Mark Ford and Jane L. Rodrigue

CONTENT AND TAXONOMIC COMMENTS

Twenty-five subspecies are currently recognized (Hall 1981, Wells-Gosling and Heaney 1984) and two occur in the South: the Carolina northern flying squirrel (*G. s. coloratus*) and the Virginia northern flying squirrel (*G. s. fuscus*). The literature was reviewed by Wells-Gosling and Heaney (1984).

DISTINGUISHING CHARACTERISTICS

The northern flying squirrel is the larger of the two flying squirrels in the South, both of which are characterized by a furred patagium extending from front to hind limbs and a long, flattened tail. The measurements are: total length, 256–274 mm; tail, 108–160 mm; hind foot, 34–39 mm; ear, 16–20 mm; weight, 90–140 g. The dental formula is: I 1 1/1, C 0 0/0, P 2 1/1, M 3 3/3 = 22 (Figure 1). The pelage is thick and silky, gray with brownish to rusty wash dorsally and grayish to buffy white ventrally, with the base of the hairs slate gray. The sides of the head are often grayish with a buffy wash. The tail is bicolored cinnamon to blackish dorsally, especially near the tip, and buffy to gray ventrally (Wells-Gosling and Heaney 1984). The northern flying squirrel is distinguished from the southern flying squirrel, *G. volans*, by its larger size, greater adult weight, and gray base of its ventral hairs. The northern flying squirrel cranium is larger (>36 mm) than that of the southern flying squirrel. Of the two extant subspecies in the South, *G. s. coloratus* tends to be larger and has a somewhat longer tail (134 mm) than *G. s. fuscus* (115 mm; Reynolds et al. 1999). See keys for details.

CONSERVATION STATUS

The U. S. Fish and Wildlife Service (U. S. Department of the Interior 1993, 2007) lists *G. s. coloratus* and *G. s. fuscus* as Endangered. The northern flying squirrel has a global rank of Secure (NatureServe 2007). However, the species is Imperiled in North Carolina and Critically Imperiled in Virginia. It is unranked in Tennessee. Significant status reviews are provided in Linzey (1983), Weigl (1987), Fies and Pagels (1991), and Weigl et al. (1999).

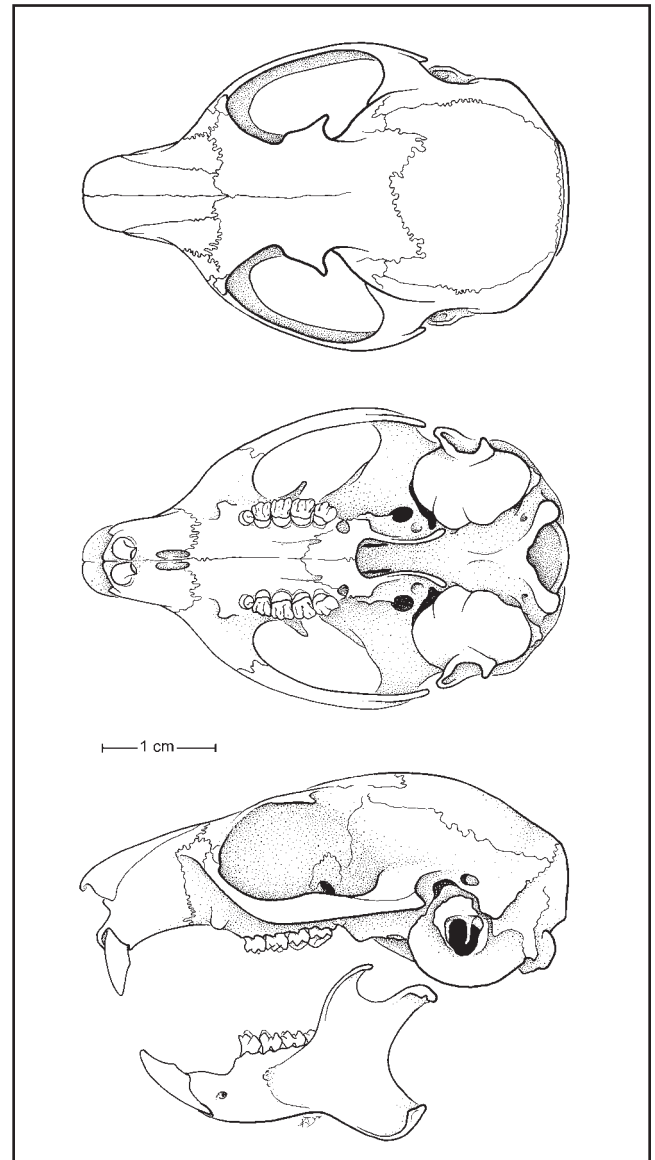


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Glaucomys sabrinus* from Fairbanks County, Alaska (USNM 242499, female).

DISTRIBUTION

The northern flying squirrel occurs throughout most of boreal and sub-boreal North America from Alaska east to Labrador and south to the Cascades, northern Rockies, Lake States and Upper mid-Atlantic to the Catskill Mountains in New York and the Pocono

Mountains and Allegheny Plateau in extreme northern Pennsylvania (Wells-Gosling and Heaney 1984, Merritt 1987). Significant southern distributional extensions include the Coast Ranges and Sierra Nevada through southern California. Disjunct populations occur in the Black Hills of South Dakota, the Allegheny Mountains of West Virginia and Virginia, and the southern Blue Ridge in North Carolina, Tennessee and Virginia (Miller 1936, Wells-Gosling and Heaney 1984, Merritt 1987, Whitaker and Hamilton 1998, Linzey 1995, Browne et al. 1999, Reynolds et al. 1999, Odom et al. 2001; Figure 2). Regionally, *G. s. fuscus* occurs at high elevations in the Allegheny Mountains of Highland County, Virginia on the George Washington National Forest and more widely in eastern West Virginia in Grant, Greenbrier, Pendleton, Pocahontas, Randolph, Tucker and Webster counties on or near the Monongahela National Forest (Pagels et al. 1990, Fies and Pagels 1991, Stihler et al. 1987, Stihler et al. 1995, Reynolds et al. 1999, Odom et al. 2001; S. Jones, U. S. Fish and Wildlife Service, personal communication). In the southern Blue Ridge, *G. s. coloratus* is distributed along several high elevation massifs in and around the Great Smoky Mountains National Park, Blue Ridge Parkway, and Pisgah, Nantahala, and Cherokee National Forests in Avery, Buncombe, Graham, Haywood, Jackson, McDowell, Mitchell, Swain, Transylvania, Watauga, and Yancey counties, North Carolina and Blount, Carter, Monroe, and Sevier counties, Tennessee. Potential high elevation habitat also occurs in Caldwell and Macon counties, North Carolina and Cocke, Johnson, and Unicoi counties, Tennessee although those areas have been unsurveyed or surveys have failed to document their presence (Linzey 1995, Weigl et al. 1999; M. Cantrell, U. S. Fish and Wildlife Service, personal communication; B. Stiver, National Park Service, personal communication; P. Wyatt, Tennessee Wildlife Resources Agency, personal communication). Northern flying squirrels also occur in and around the Mt. Rogers-Grayson Highlands area of Grayson and Smyth counties, Virginia on the Jefferson National Forest (Reynolds et al. 1999). These specimens are referable to *G. s. coloratus*, although Fies and Pagels (1991) and Sparks (2005) suggested they could be intergrades between *G. s. coloratus* and *G. s. fuscus*.

ABUNDANCE STATUS

The species is very rare in the South and population densities in occupied habitat are unknown. Prior to Federal listing, 30 specimens from eight localities were known from the central and southern Appalachians. The number of collection localities has been expanded greatly with approximately 1,200 individuals caught in West Virginia and > 1,000 individuals

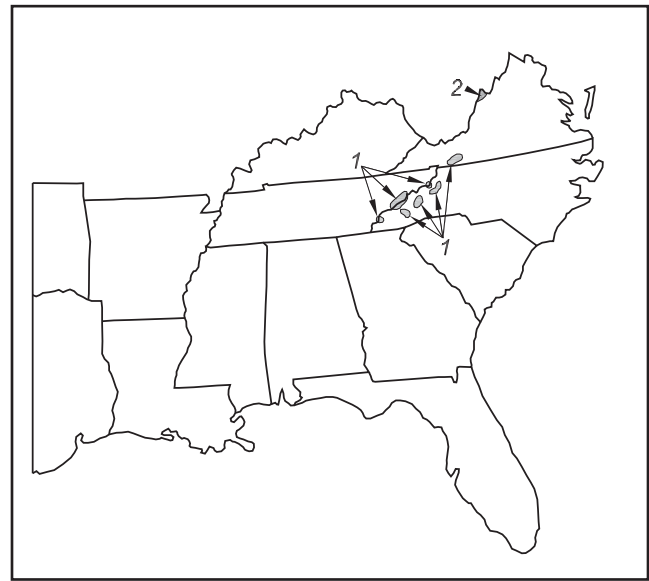


Figure 2. Distribution of *Glaucomys sabrinus* in the South: (1) *G. s. coloratus*; (2) *G. s. fuscus*.

caught in North Carolina, Tennessee, and Virginia since 1985 (Reynolds et al. 1999, Weigl et al. 1999; C. Stihler, West Virginia Department of Natural Resources, personal communication). However, Fies and Pagels (1991) suggest fewer than 250 individuals remain in Virginia. Both *G. s. coloratus* and *G. s. fuscus* have demonstrated yearly population fluctuations at some monitoring sites (Weigl et al. 1999; C. Stihler, West Virginia Department of Natural Resources, personal communication).

PRIMARY HABITATS

Throughout its range, the northern flying squirrel inhabits a variety of boreal coniferous and mixed hardwood forest habitats (Wells-Gosling and Heaney 1984). In the South, the species is a Pleistocene relict restricted to montane boreal or northern hardwoods communities at the highest elevations of the central and southern Appalachians. Montane boreal habitats in the southern Blue Ridge occupied by *G. s. coloratus* are dominated by conifers such as red spruce (*Picea rubens*) and Fraser fir (*Abies fraseri*), whereas those occupied by *G. s. fuscus* in the Allegheny Mountains are dominated by red spruce and eastern hemlock (*Tsuga canadensis*). Northern hardwood and northern hardwood-montane boreal forest ecotones occupied by northern flying squirrels in both areas are comprised of overstories of yellow birch (*Betula alleghaniensis*), sugar maple (*Acer saccharum*), red maple (*A. rubrum*), American beech (*Fagus grandifolia*), and black cherry (*Prunus serotina*), along with the aforementioned conifers. Captures of *G. s. fuscus* in the Alleghenies have ranged in elevation from approximately 900–1400 m (Stihler et al. 1995, Reynolds et al.

1999) whereas those of *G. s. coloratus* in the southern Blue Ridge generally have occurred at elevations above 1500 m (Browne et al. 1999). One individual was recorded in the Great Smoky Mountains National Park at 1230 m (Weigl 1968). Northern flying squirrels in the central and southern Appalachians have been documented in a variety of stand ages and conditions (Payne et al. 1989, Weigel and Osgood 1974), but mature forest conditions in the northern hardwood-montane boreal ecotone on north-facing slopes with large trees, numerous snags, high volumes of coarse wood debris, and abundant lichens and hypogaeal fungi are believed to be optimal (Reynolds et al. 1999, Weigl et al. 1999, Loeb et al. 2000, Schuler et al. 2002, Hackett and Pagels 2003). Odom et al. (2001) found that increased elevation and close proximity to the presence of red spruce and eastern hemlock were the best predictive variables between occupied and unoccupied habitat for *G. s. fuscus* in the Allegheny Mountains at the landscape-level. At the stand-level, forests with 35% montane conifer overstories, primarily red spruce, are very indicative of occupied habitat (Ford et al. 2004). Individual home ranges are variable, ranging from 1.2–51.4 ha for *G. s. coloratus* in North Carolina (Weigl et al. 1999, Weigl et al. 2002) and 5 ha to > 100 ha for *G. s. fuscus* in West Virginia (Urban 1988, Menzel et al. 2006b). Most northern flying squirrel activity occurs from shortly after sunset until midnight (Weigl et al. 2002). Home range size probably varies by habitat structure quality and seasonal food abundance. Males tend to have larger home ranges than females, particularly in late winter at the onset of breeding (Weigl et al. 1999). Both subspecies typically den in cavities in live hardwoods and snags. Yellow birch and American beech are two of the most preferred den tree species. Leaf or “drey” nests in conifers such as red spruce also are common (Weigl et al. 1999, Weigl et al. 2002, Menzel et al. 2004), particularly in the warmer months (Urban 1988). Both Hackett and Pagels (2003) and Menzel et al. (2004) reported that northern flying squirrels routinely utilize den trees near trails, old logging roads or railroad grades; occupied den trees and snags tended to be larger and taller than surrounding stems. Northern flying squirrels will readily occupy artificial nest boxes (Reynolds et al. 1999). Both natural and artificial dens are lined with bark, moss or grass, particularly shredded yellow birch bark. Unlike the southern flying squirrel, large aggregations of individuals sharing a single nest typically does not occur (Fies and Pagels 1991).

REPRODUCTION

Little is known about the reproductive habits of the northern flying squirrel in the South and most information is gleaned from trap or nest box capture data.

In both North Carolina and Virginia, testicular males have been reported from December into the spring months. Single litters of 2–4 are usually born March through June, but offspring can be born as late as September (Weigl et al. 1999). Gestation is approximately 37 days and weaning occurs at about 2 months, though young may remain with the female for some time. In part because year-to-year recapture rates are low in regional monitoring programs (Reynolds et al. 1999), actual longevity in the central and southern Appalachians is unknown. Elsewhere, it is thought to be < 4 years (Wells-Gosling and Heaney 1984).

FOOD HABITS

Throughout its range, northern flying squirrels feed on a diversity of conifer and hardwood seeds and cones, buds, fruits, insects, bird eggs and nestlings and even tree sap (Hall 1981, Carey et al. 1999, Weigl et al. 1999); however, the sporocarps of mycorrhizal fungi such as the genera *Elaphomyces*, and lichens are among the most important foods in the central and southern Appalachians (Weigl 1968, Weigl 1978, Wells-Gosling and Heaney 1984, Weigl et al. 1999, Mitchell et al. 2001). In North Carolina, hypogaeal fungal presence is closely correlated with the presence of red spruce in the overstory (Loeb et al. 2000). The year-round abundance of fungi and lichens may provide a steady food supply and reduce competition with other sciurids that depend upon hard mast such as northern red oak (*Quercus rubra*) acorns. Mitchell et al. (2001) found significant use of American beechnuts by *G. s. fuscus* in West Virginia during autumn. Northern flying squirrels spend large amounts of time on the ground indicative of foraging efforts for fungi during evening activity periods (Urban 1988).

ASSOCIATED SPECIES

The northern flying squirrel is frequently found in association with typical montane boreal small mammals in the central and southern Appalachians including the northern short-tailed shrew (*Blarina brevicauda*), smoky shrew (*Sorex fumeus*), masked shrew (*S. cinereus*), deer mouse (*Peromyscus maniculatus*), Allegheny woodrat (*Neotoma magister*), and red-backed vole (*Clethrionomys gapperi*). Other sciurids with which northern flying squirrel may be syntopic include the southern flying squirrel and red squirrel (*Tamiasciurus hudsonicus*).

VULNERABILITY AND THREATS

Despite numerous new collection records, the realization of larger populations than once believed, and as a potential candidate for down-listing, the northern

flying squirrel remains one of the rarest mammals in the southern United States. In part, the species rarity is an artifact of the natural contraction of northern forest communities in the Appalachians following the end of the last Ice Age to what are now relictual habitat islands (Browne et al. 1999, Arbogast et al. 2005). Regionally, many red spruce forest stands were destructively logged and altered by post-harvest fire at the turn of the 20th century, reducing this forest type's extent by up to 90% (Korstian 1937, Schuler et al. 2002), leading many to characterize this forest type as among the most endangered in North America (White et al. 1993). This habitat change and fragmentation undoubtedly has had serious negative impacts on northern flying squirrels. There is evidence of reduced genetic heterozygosity in *G. s. coloratus* from natural and human-induced isolation that has exposed sub-populations to a high risk of inbreeding depression (Browne et al. 1999). Current efforts are underway to fully assess the conservation genetic status of both subspecies in the region (J. Sparks, Virginia Commonwealth University, personal communication). Increased abundance of northern red oak and American beech following modification of these habitats probably has provided an infusion of cacheable, high-energy foods that allow southern flying squirrels to tolerate the climatically difficult and energetically expensive conditions at high elevations in the central and southern Appalachians (Weigl et al. 1999, Odom et al. 2001). In turn, range expansions of the competitively aggressive southern flying squirrel may have displaced northern flying squirrels locally at some sites (Weigl 1978, Weigl 1987, Fies and Pagels 1991). Moreover, many southern flying squirrels chronically harbor a parasitic nematode, *Strongyloides robustus*, which is believed to be seriously debilitating when passed to northern flying squirrels in these interspecific contact zones (Weigl 1978, Weigl 1987, Pagels et al. 1990). Weigl et al. (1999) noted a decline in northern flying squirrel numbers and reproductive activity following a steady increase in *S. robustus* infection and a series of mild winters in the Roan Mountain area on the North Carolina-Tennessee border. Fortunately, most identified northern flying squirrel habitat in the central and southern Appalachians occurs on public lands that are protected from further intentional anthropogenic perturbation. There are some indications that the ecotone between red spruce and northern hardwood forests is either stabilizing or decreasing in elevation to more approximate its former extent (Busing et al. 1988, Schuler et al. 2002). Still, infestations and spread of the balsam woolly aphid (*Adelges piceae*) and hemlock adelgid (*A. tsugae*) that threaten Fraser fir and eastern hemlock, as well as high atmospheric acid deposition, and continued global climate change continue to threaten the remaining structure, extent and integrity

of northern flying squirrel habitat patches in the region (Nicholas et al. 1992, Barker et al. 2002, Menzel 2006a). On private land, surface mining, forest management, second-home and recreational development, and wind-energy development are potential threats to northern flying squirrel populations and their habitats (Weigl et al. 1999, Schuler et al. 2002). Currently, *G. s. fuscus* is the focal species of several Habitat Conservation Plans on private lands in the Allegheny Mountains (S. Jones, U. S. Fish and Wildlife Service, personal communication).

MANAGEMENT SUGGESTIONS

Conditions that continue to promote or enhance mature forest conditions with an abundance of large trees, numerous snags, and mesic, micro-habitats in montane boreal and northern hardwood forests in the central and southern Appalachians should be maintained. Activities that convert or fragment occupied forest areas should be avoided, as should the use of prescribed fire in northern flying squirrel habitats. Techniques to effectively monitor population and demographic trends beyond current nest box programs also need to be developed (Odom et al. 2001). Odom and McNab (2000) showed that digital terrain modeling could successfully predict current and potential montane boreal and northern hardwood habitats in the southern Blue Ridge to guide habitat protection efforts and prioritize where habitat restoration activities should occur. Opportunities for ecological restoration of degraded former habitats through silvicultural manipulation of hardwoods to encourage red spruce release and eventual overstory/stand dominance to expand or link occupied patches have been proposed but remain untested (Schuler et al. 2002).

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Southern Flying Squirrel

Glaucomys volans (Linnaeus, 1758)

Joshua Laerm and Wm. David Webster

CONTENT AND TAXONOMIC COMMENTS

Eight subspecies of *Glaucomys volans* currently are recognized (Diersing 1980, Hall 1981), four of which occur in the South: *G. v. querceti*, *saturatus*, *texensis*, and *volans*. These have not been examined critically since Howell's (1918) revision; however, they are poorly defined (Lowery 1974) and patterns of morphological variation do not correspond to current subspecies boundaries (Braun 1988). The literature is reviewed by Dolan and Carter (1977) and Wells-Gosling (1985).

DISTINGUISHING CHARACTERISTICS

This is the smaller of the region's two flying squirrels, both of which are characterized by a furred patagium extending from front to hind limbs and a long flattened tail. External measurements are: total length, 198–255 mm; tail, 81–120 mm; hind foot, 28–33; ear, 12–23 mm; weight, 40–48 g. The pelage is soft and silky, gray with a cinnamon tinge above, grading into black at the edge of the patagium, and creamy-white below (including the bases of the individual hairs); the sides of the head are often grayish; the eyes are ringed in black; and the tail is grayish above, not tipped in black, and buffy to cinnamon below. The southern flying squirrel can be easily distinguished from the northern flying squirrel, *G. sabrinus*, by its smaller size and by the whitish bases of the hairs on its venter. The cranium is smaller (less than 36 mm) than that of *G. sabrinus*. The dental formula is I 1/1, C 0/0, P 2/1, M 3/3 = 22 (Figure 1). See keys for details.

CONSERVATION STATUS

The southern flying squirrel has a global rank of Secure (NatureServe 2007). It is considered Secure in Alabama, Georgia, Kentucky, Louisiana, Mississippi, North Carolina, Tennessee, and Virginia and Apparently Secure in Arkansas and Texas. It is Vulnerable in Oklahoma, and unranked in Florida and South Carolina.

DISTRIBUTION

The southern flying squirrel ranges throughout the eastern half of the United States and southern Quebec. In addition, a series of disjunct populations occur in

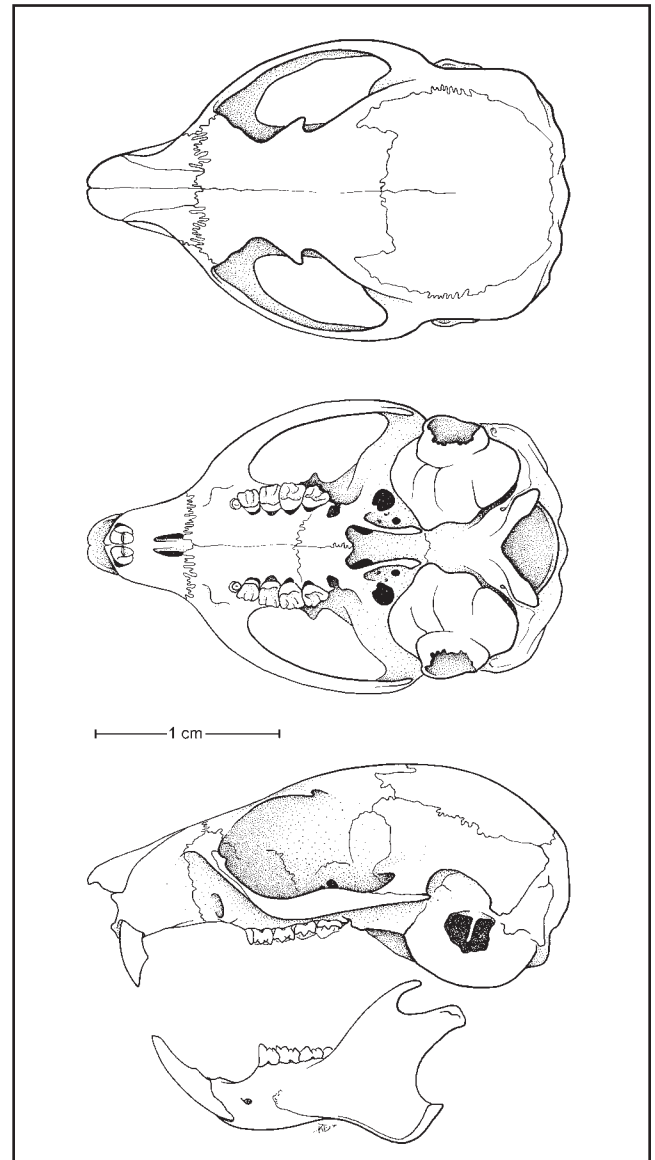


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Glaucomys volans* from Volusia County, Florida (USNM 72934, male).

Mexico and Central America (Diersing 1980). Regionally, it occurs widely throughout Virginia (Handley and Patton 1947, Handley 1979, 1992; Linzey 1998), North Carolina (Lee et al. 1982, Webster et al. 1985), South Carolina (Golley 1966, Schacher and Pelton 1979, Cothran et al. 1991), Georgia (Golley 1962, Neuhauser and Baker 1974, Laerm et al. 1982), Florida

(Rand and Host 1942, Moore 1946, Schwartz 1952, Pearson 1954, Sherman 1957, Ivey 1959, Layne 1974, Layne and Raymond 1994), Alabama (Howell 1921, Holliman 1963, Linzey 1970), Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989), Louisiana (Lowery 1974, Goertz et al. 1975), Tennessee (Calhoun 1941, Howell and Conaway 1952, Conaway and Howell 1953, Linzey and Linzey 1971, Smith et al. 1974, Kennedy 1991), Kentucky (Barbour and Davis 1974, Severinghaus et al. 1980), Arkansas (Sealander and Heidt 1990, Caster et al. 1994, Stone et al. 1996), eastern Texas (Schmidly 1983, Davis and Schmidly 1994), and eastern Oklahoma (Caire et al. 1989; Figure 2).

ABUNDANCE STATUS

The species is common to abundant throughout deciduous and pine (*Pinus* spp.) forests of the region. Most population estimates range from 2–13/ha (Burt 1940, Sollberger 1943, Jordan 1948, Jackson 1961, Muul 1968, Sonenshine et al. 1979), but population densities can reach 31–38/ha (Sawyer and Rose 1985). Individuals often live communally, particularly in winter, with up to 25 occupying a single cavity (Jackson 1961, Layne and Raymond 1994, Brady et al. 2000), a strategy that conserves energy (Merritt et al. 2001). Density apparently is not limited by the availability of nest sites (Brady et al. 2000).

PRIMARY HABITATS

The availability of nest sites and food resources appear to be the limiting factors in the distribution and density of the southern flying squirrel. Most hardwood cover types are the preferred habitat throughout the region. Pine and mixed pine-hardwood forests may be utilized, but usually only in the presence of mast producing species such as oak (*Quercus* spp.), hickory (*Carya* spp.), and American beech (*Fagus grandifolia*; Sollberger 1940, Moore 1946, Muul 1968, Weigl 1978, Sonenshine et al. 1979, Sonenshine and Levy 1981, Gilmore and Gates 1985, Taulman 1999). Natural tree cavities are preferred nesting sites, but woodpecker holes and abandoned nests of other squirrels may be utilized (Heidt 1977, Rudolph et al. 1990, Loeb 1993). The southern flying squirrel commonly nests in the attics of houses and abandoned buildings. Nests serve as food caches, refugia from predators, feeding stations, latrines, and sites for rearing young. The lower availability of suitable nesting cavities in coniferous trees explains their dependence upon hardwood stands. Moreover, although the southern flying squirrel has a varied diet, it is particularly dependent on hardwood mast in winter.



Figure 2. Distribution of *Glaucomys volans* in the South: (1) *G. v. querceti*; (2) *G. v. saturatus*; (3) *G. v. texensis*; (4) *G. v. volans*.

REPRODUCTION

Breeding is concentrated in the spring and fall (Brimley 1923, Svihla 1930, Hibbard 1935, Moore 1947, Goertz 1965, Sonenshine et al. 1979). One or two (and possibly more) litters may be produced each year (Hamilton 1943, Sollberger 1943, Jordan 1948, Linzey and Linzey 1979, Sonenshine et al. 1979). Gestation is 39–40 days; litter size is usually 3–4, although it ranges from 2–6. Young are weaned by 6–8 weeks and remain with the mother if a second litter is not produced (Svihla 1930, Hatt 1931, Rand and Host 1942, Sollberger 1943, Muul and Alley 1963, Muul 1968, 1969, Linzey and Linzey 1979, Sonenshine et al. 1979, Sawyer and Rose 1985, Raymond and Layne 1988, Pitts 1992).

FOOD HABITS

Southern flying squirrels feed on a diversity of plant matter including nuts, seeds, berries, fruits, buds, and fungi (Sollberger 1940, Jordan 1948, Muul and Alley 1963, Muul 1968, Weigl 1978, Harlow 1990). Furthermore, they are somewhat carnivorous and consume insects and other invertebrates as well as birds, eggs, nestlings, and carrion when available (Dolan and Carter 1977).

ASSOCIATED SPECIES

The species is associated with essentially all hardwood and mixed hardwood-coniferous forests and the small mammal species that inhabit these forest types.

It occurs in association with other sciurids including the eastern gray squirrel (*Sciurus carolinensis*), eastern fox squirrel (*S. niger*), red squirrel (*Tamiasciurus hudsonicus*), eastern chipmunk (*Tamias striatus*), and to a limited extent the northern flying squirrel (*G. sabrinus*). Unlike other sciurids, which are diurnal, the two species of *Glaucomys* are nocturnal. Southern flying squirrels can be cavity nest competitors to northern flying squirrels as well as vectors for the parasitic nematode, *Strongyloides robustus*, that are often lethal when passed to northern flying squirrels (Weigl et al. 1999).

VULNERABILITY AND THREATS

The southern flying squirrel is common throughout the eastern deciduous forest and nowhere does it appear to be vulnerable to threat. In some areas it is viewed as a pest species because of its use of houses and buildings. There is concern over the possible negative impact *G. volans* may have on the endangered red-cockaded woodpecker (*Picoides borealis*) throughout much of the South because flying squirrels usurp both active and inactive woodpecker cavities for nest sites (Rudolph et al. 1990, Loeb 1993). However, reproductive success of red-cockaded woodpeckers is similar in areas where squirrels have and have not been removed experimentally (Mitchell et al. 1999).

MANAGEMENT SUGGESTIONS

Southern flying squirrels require relatively large stands of mature conifer-hardwood and hardwood forests with adequate numbers of nest cavities and snags, especially forest stands dominated by mast producing species.

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Woodchuck

Marmota monax (Linnaeus, 1758)

Michael T. Mengak and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

The woodchuck or groundhog includes nine named subspecies, one of which occurs in the South, *Marmota m. monax* (Hall 1981, Lee et al. 1982, Whitaker, 1998). Kwiecinski (1998) reviewed the literature.

DISTINGUISHING CHARACTERISTICS

The woodchuck is the largest of the regional squirrels; it is a heavy-bodied, somewhat flat, squat rodent with short legs and tail. Its measurements are: total length, 418–665 mm; tail, 100–155 mm; hind foot, 66–68 mm; ear, 70–90 mm; weight, 2.3–5.4 kg. Its dorsal pelage is grizzled reddish brown to grayish brown overall. Its dense underfur is dark at the base and pale gray to buff at the tips. The guard hairs are tipped in gray or white, contributing to the grizzled appearance. The head, feet, and tail are dark. The under parts are somewhat pale. The woodchuck skull is characterized by an infraorbital opening that pierces the rostrum anterior to the zygomatic plate, broad postorbital process, that project at nearly right angles to long axis of the skull. The skull is concave between the postorbital processes. The dental formula is I 1/1, C 0/0, P 2/1, M 3/3=22 (Figure 1). See keys for details.

CONSERVATION STATUS

The woodchuck has a global rank of Secure (NatureServe 2007). It is Secure in Alabama, Kentucky, North Carolina, Tennessee, and Virginia. It is Apparently Secure in Arkansas and Mississippi. Both Georgia and Oklahoma classify the woodchuck as Vulnerable. It is unranked in South Carolina.

DISTRIBUTION

The woodchuck ranges throughout most of southeastern Canada and the eastern half of the United States, except for much of the southeastern Coastal Plain and Gulf Coast regions (Whitaker 1998). Figure 2 depicts the distribution of the woodchuck in the South. Robinson and Lee (1980) reviewed its recent range expansion in the southeastern Piedmont and Coastal Plain where it had not occurred in historical times. It is widely distributed throughout Virginia except for the Eastern Shore and lower southeastern Tidewater (Bailey 1946, Handley and Patton 1947,

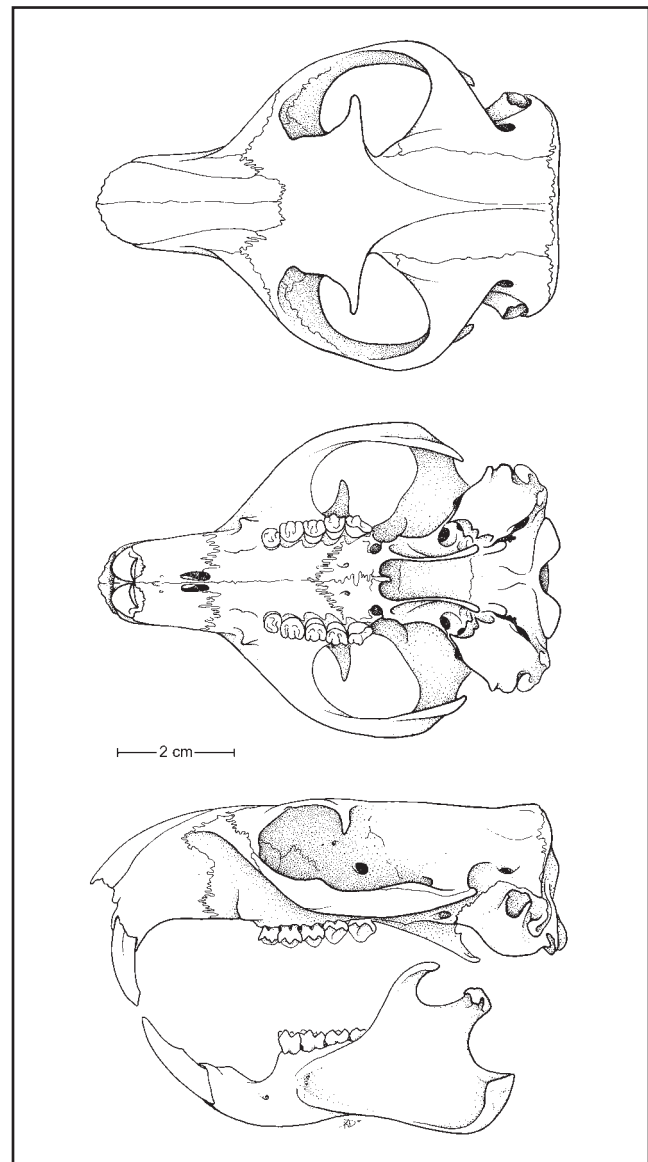


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Marmota monax* from Hendricks County, Indiana (USNM 347750, male).

Paul and Cordes 1969, Jackson et al. 1976, Robinson and Lee 1980, Handley 1992, English and Bowers 1994). The woodchuck is absent from the Tidewater area of North Carolina, but is expanding its range throughout the northern Coastal Plain and much of the Piedmont (Odum 1949, Johnston 1967, Robinson

and Lee 1980, Lee et al. 1982). South Carolina records are restricted to the Blue Ridge (Sherman 1937, Golley 1966, Robinson and Lee 1980). Golley (1962) and Laerm et al. (1982) described a distribution in the Blue Ridge and Upper Piedmont of Georgia; the species now ranges well into the middle and lower Piedmont in western Georgia. Populations inhabit the Upper Coastal Plain of Alabama (Howell 1921, Holt 1924, Holliman 1963, Hanlin and Russell 1978). The woodchuck is restricted to northeastern Mississippi (Ferguson 1962, Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989). The single record from Louisiana could represent a release (Lowery 1974); however, given the expanding range and proximity of Arkansas populations, it is likely that populations may occur in northwestern Louisiana. *Marmota monax* occurs throughout most of Tennessee (Calhoun 1941, Goodpaster and Hoffmeister 1952, Howell and Conaway 1952, Conaway and Howell 1953, Schultz 1954, Linzey and Linzey 1971, Smith et al. 1974, Kennedy 1991, Linzey 1995) and Kentucky (Barbour and Davis 1974, Fassler 1974, Schmeltz and Whitaker 1977), but to the west it is primarily restricted to the Interior Highlands of Arkansas (Hayes 1976, Sealander and Heidt 1990), and eastern Oklahoma (Long 1961, Caire et al. 1989).

ABUNDANCE STATUS

The woodchuck is common to abundant in appropriate habitat in the region. Population densities vary depending on habitat and season. In orchards and hayfields, densities of 3–8 burrows/ha have been reported (Henderson and Gilbert 1978, Swihart 1990a). It is a true hibernator, and in northern portions of its range remains in underground burrows throughout the colder months from November to February (Davis 1967). In southern portions of its range, the hibernation period is very short. In the Coastal Plain, individuals may remain active all year (Robinson and Lee 1980, Webster et al. 1985, Sealander and Heidt 1990).

PRIMARY HABITATS

Woodchucks are most common in open fields, oldfields, and brushy fencerows; they are often present in rocky slopes and at the base of limestone or sandstone escarpments. Although woodchucks use open woodlots and climb trees (Kwiecinski 1998), they rarely occur in dense woodlands. They inhabit transportation corridors and utility rights-of-way. Elimination of forest cover and conversion to grassland in the past century has permitted the expansion of its range.

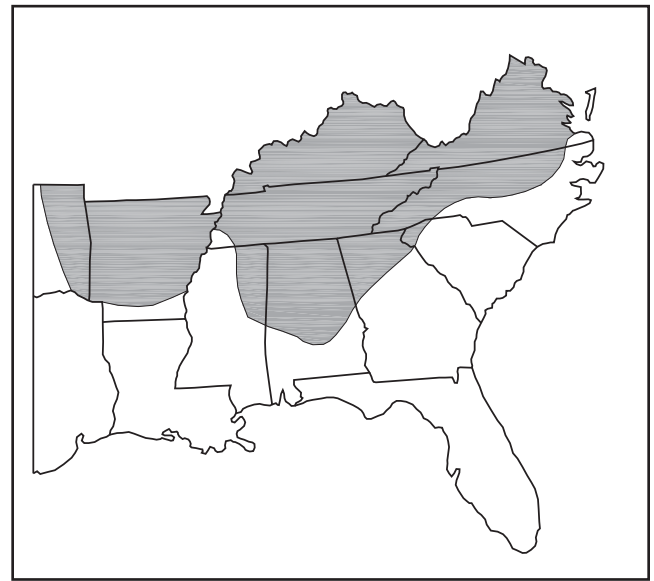


Figure 2. Distribution of *Marmota monax* in the South.

Woodchucks create extensive burrows in well-drained soils. Burrows are actively maintained; mounds of fresh soil are visible near the burrow entrance, which may be located beneath structural debris or in the open (Hamilton 1934, Grizzell 1955, Lee et al. 1982). Burrows rarely are shared with other woodchucks; however, an adult male and reproductive female may simultaneously use a burrow (Swihart 1992). Burrows serve as refugia for many wildlife species including the eastern cottontail (*Sylvilagus floridanus*), Virginia opossum (*Didelphis virginiana*), striped skunk (*Mephitis mephitis*), red fox (*Vulpes vulpes*), and the northern raccoon (*Procyon lotor*; Swihart and Picone 1995). In addition, the meadow vole (*Microtus pennsylvanicus*) and white-footed mouse (*Peromyscus leucopus*) use areas near the burrows extensively (Swihart and Picone 1995).

REPRODUCTION

Breeding is concentrated between January and March after emergence from hibernation. A single litter is produced each year. Gestation ranges from 31–32 days (Hamilton 1934, Grizzell 1955, Kwiecinski 1998). Average litter size is 4–6 (Kwiecinski 1998); individuals are weaned in approximately 4 weeks and disperse their first year. Most individuals do not breed until their second year (Hamilton 1934, Hoyt and Hoyt 1950, Grizzell 1955, Snyder and Christian 1960, Lee et al. 1982). Males have a home range that is almost twice as large as that of females (Swihart 1992).

FOOD HABITS

The woodchuck is primarily herbivorous; the diet consists of legumes, succulent herbs, leaves, twigs, fruits and vegetables (e.g., apples, peas, beans, carrots, cabbage, and corn (Grizzell 1955, Lee et al. 1982, Arsenault and Romig 1985, Swihart 1990b). Other foods include blackberry (*Rubus* spp.), serviceberry (*Amelanchier* spp.), chickweed (*Stellaria* spp.), and various grasses. Water requirements are usually satisfied by dew but they will occasionally drink free water (Grizzell 1955).

ASSOCIATED SPECIES

The species is associated with a number of grassland mammal species such as the northern short-tailed shrew (*Blarina brevicauda*), southeastern shrew (*Sorex longirostris*), least shrew (*Cryptotis parva*), harvest mice (*Reithrodontomys* spp.), deer mouse (*Peromyscus maniculatus*), hispid cotton rat (*Sigmodon hispidus*), meadow vole (*Microtus pennsylvanicus*), and prairie vole (*M. ochrogaster*).

VULNERABILITY AND THREATS

The woodchuck is common in grassland areas, oldfields, home sites, and early successional habitats. In many states it is viewed as a game animal as well as an agricultural pest. Nowhere does it appear to be vulnerable to threat.

MANAGEMENT SUGGESTIONS

Management that maintains oldfields and early successional habitats (e.g., planting food plots, mowing road banks) benefit woodchucks. Maintaining wildlife openings, utility corridors, and clearcuts near existing populations will benefit the woodchuck.

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Eastern Gray Squirrel

Sciurus carolinensis (Gmelin, 1788)

John W. Edwards and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Five subspecies are recognized, four of which occur in the South: *S. c. carolinensis*, *S. c. extimus*, *S. c. fuliginosus*, and *S. c. pennsylvanicus* (Hall 1981, Koprowski 1994). Lazell (1989) suggests that *S. c. matecumbei*, restricted to a few of the Florida Keys and long regarded a synonym of *S. c. extimus* (Hubbard and Banks 1970) may warrant recognition. Barkalow and Shorten (1973), Flyger and Gates (1982), Koprowski (1994), and Edwards et al. (2003) review the literature.

DISTINGUISHING CHARACTERISTICS

The gray squirrel is a medium-sized tree squirrel. Measurements are: total length, 380–525 mm; tail, 150–250 mm; hind foot, 54–76 mm; ear, 25–33 mm; weight, 300–750 g. There is no sexual dimorphism in size or pelage. Pelage most commonly is pale to dark grizzled gray above, with a “salt and pepper” appearance resulting from alternating bands of white, brown and black on hairs; mid-dorsum slightly more brownish; flanks and upper parts of feet with cinnamon to brownish wash. Ears are buff to gray to white (behind); chin, throat, and venter are white. The tail is elongated and flattened with brown and black bands and white tips. Pelage variants include melanistic black to sooty gray as well as yellowish to albino individuals and populations (Flyger and Gates 1982, Koprowski 1994). The gray squirrel is easily distinguished from the fox squirrel (*S. niger*) by its smaller size and presence of a pair of upper premolars. The dental formula is I 1/1, C 0/0, P 2/1, and M 3/3 = 22 (Figure 1). See keys for details.

CONSERVATION STATUS

The eastern gray squirrel has a global rank of Secure (NatureServe 2007). The species is listed as Secure in Alabama, Georgia, Kentucky, Louisiana, Mississippi, North Carolina, Oklahoma, Tennessee, Texas, and Virginia. Arkansas lists it as Apparently Secure, and it is unranked in Florida and South Carolina. The gray squirrel is considered a game animal with harvest seasons regulated in all states in the South.

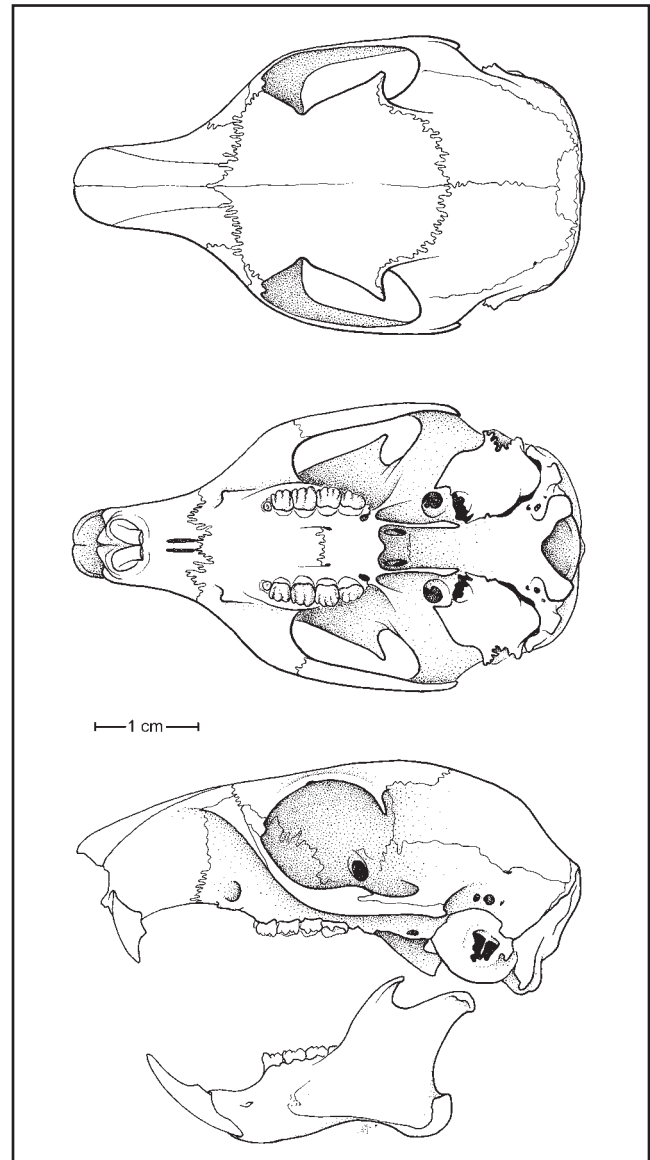


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sciurus carolinensis* from the Peaks of Otter, Bedford County, Virginia (USNM 85506, male).

DISTRIBUTION

The eastern gray squirrel ranges throughout the eastern and central United States and southern Canada west to the limits of deciduous forests and south to Mexico. It has been widely introduced in the western United States, Canada, England, Europe, and elsewhere

(Flyger and Gates 1982, Koprowski 1994). The species is common throughout Virginia (Patton 1939, Jackson et al. 1976, Dueser et al. 1979, Payne et al. 1987, Handley 1992), North Carolina (Odum 1949, Soots 1964, Smith 1967, Barkalow et al. 1970, Cordes and Barkalow 1972, Lee et al. 1982, Clark et al. 1985, Weigl et al. 1989), South Carolina (Sanders 1978), and Georgia (Neuhauser and Baker 1974, Laerm et al. 1982, Edwards and Guynn 1995, Conner et al. 1999), including most of the barrier islands of these states (Figure 2). They occur throughout Florida (Blair 1935, Moore 1946, 1949; Pournelle 1950, Lazell 1989), Alabama (Holliman 1963, Linzey 1970, Hotton 1978, Fischer and Holler 1991, 1994; Yarrow and Yarrow 1999), Mississippi (Jones and Carter 1989, Moncrief 1993, Chamberlain et al. 1999), Louisiana (Lowery 1974, Heuer and Perry 1976, Moncrief 1993), north throughout Tennessee (Calhoun 1941, Goodpaster and Hoffmeister 1952, Schultz 1957, Linzey and Linzey 1971, Smith et al. 1974, Fowler and Dimmick 1983, Kennedy 1991), Kentucky, and west throughout Arkansas (Sealander and Heidt 1990, Stone et al. 1996) into eastern Texas (Goodrum 1940, Baker 1944, Schmidly 1983, Davis and Schmidly 1994) and Oklahoma (Chesemore 1975).

ABUNDANCE STATUS

Gray squirrels are one of the most common mammals in the region. Although long-term densities appear to remain fairly constant (Gurnell 1987), short-term densities fluctuate irregularly (Uhlig 1956, 1957; Flyger 1959, Flyger and Gates 1982), typically in response to yearly fluctuations of mast production (Nixon and McClain 1969, Nixon and Hansen 1987). The effect of a good or poor mast crop is reflected in squirrel abundance the following year (Burns et al. 1954, Smith and Barkalow 1967). Fluctuations in densities also are attributed to weather conditions, predation, and disease. Actual densities are reported to range between 0.5–140/ha (Uhlig 1957, Flyger 1959, Mosby 1969, Nixon and McClain 1969, Barkalow et al. 1970, Doebel and McGinnis 1974, Fischer 1989).

PRIMARY HABITATS

The gray squirrel is most common in mature contiguous woodlands typically larger than 40 hectares in size, with a diverse woody understory and abundant den cavities (Koprowski 1994). Throughout its range, habitats and vegetational associations are highly varied. The range of the gray squirrel corresponds closely to that of the eastern deciduous forest, and highest densities occur in habitats dominated by mast producing hardwood tree species such as oaks (*Quercus* spp.), hickories (*Carya* spp.), and walnuts (*Juglans* spp.; Burns et al. 1954, Nixon and McClain 1969, Brown

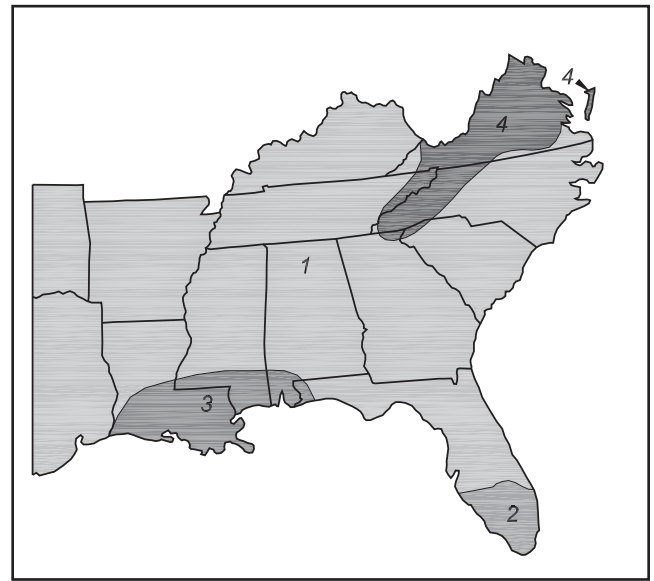


Figure 2. Distribution of *Sciurus carolinensis* in the South: (1) *S. c. carolinensis*; (2) *S. c. extimus*; (3) *S. c. fuliginosus*; (4) *S. c. pennsylvanicus*.

and Batzli 1984, Nixon and Hansen 1987). The gray squirrel is frequently associated with other mixed hardwood stands and conifers (Koprowski 1994), but rarely occurs at high densities in pure pine (*Pinus* spp.) stands. Landscapes containing at least 20 percent forest cover are generally required to support populations (Nixon et al. 1978); forested stands must be dense enough to permit travel between canopy trees without the necessity of descent to the ground. The species also is common in residential and urban areas.

Gray squirrels construct leaf nests and use tree cavities for protection from predators and inclement weather, escape, and rearing of young. Leaf nests occur in a variety of tree species including both hardwoods and pines. Vines such as grape (*Vitis* spp.) are an important component in nest site selection (Nixon and Donohue 1979, Sanderson et al. 1980). Gray squirrels use cavities in a variety of tree species. Cavity use is greatest during winter and spring (Fowler and Dimmick 1983, Ivey and Frampton 1987, Nupp 1992, Edwards and Guynn 1995). In areas where natural cavities are limited, the addition of artificial cavities can increase carrying capacity through increased survival of young and adults.

REPRODUCTION

In the South, females may breed twice annually (i.e., spring and summer), particularly following a good mast crop (Brauer and Dusing 1961, Nixon and McClain 1969). Gestation is 44–45 days (Webley and Johnson 1983), and litter size ranges from 1–8 with reported means of 1.8–3.7 (Brown and Yeager 1945,

Barkalow et al. 1970, Barkalow and Shorten 1973, Nixon et al. 1975). Weaning occurs between 8–10 weeks. Rarely, females may breed as early as 6 months of age (Smith and Barkalow 1967); most breeding occurs after 12 months of age (Brauer and Dusing 1961). Precocial breeding is typically associated with excellent mast crops (Smith and Barkalow 1967, Nixon and McClain 1969). Reproductive longevity averages 6–7 years up to a maximum of 12 years (Barkalow and Soots 1975).

FOOD HABITS

The gray squirrel diet is diverse and varies seasonally. Preferred foods are nuts, seeds, buds, and flowers of oaks, hickories, pecans (*Carya* spp.), walnuts, and several hardwood species such as beech (*Fagus grandifolia*), dogwood (*Cornus* spp.), maple (*Acer* spp.), and elm (*Ulmus* spp.). The seeds and catkins of conifers, including cedar, hemlock (*Tsuga* spp.), pine, and spruce (*Picea* spp.) are also consumed. In addition, fruits, herbaceous plants, and fungi are important summer foods. Cultivated crops also are consumed where available. Small vertebrates including bird eggs and nestlings are also preyed upon. See Flyger and Gates (1982), Koprowski (1994), and Edwards et al. (2003) for an extensive review of food habits.

ASSOCIATED SPECIES

The gray squirrel is associated with a diversity of small mammals that inhabit deciduous and mixed hardwood-pine forests. It occurs in association with other sciurids including the eastern fox squirrel (*S. niger*), southern flying squirrel (*Glaucomys volans*), red squirrel (*Tamiasciurus hudsonicus*), and eastern chipmunk (*Tamias striatus*).

VULNERABILITY AND THREATS

Survivorship in gray squirrel populations is highly dependent on mast production. Adult mortality ranges from 30 to 75 percent (Mosby 1969, Barkalow et al. 1970, Allen 1942); juvenile mortality ranges from 70 to 95 percent (Barkalow et al. 1970). Parasites, disease, and predation are generally considered minor factors in gray squirrel population dynamics. The gray squirrel is one of the most important game species in the eastern United States with annual harvests of approximately 40 million (Flyger and Gates 1982). Hunting mortality is considered compensatory to some extent, and is generally not thought of as a major factor controlling squirrel populations (Allen 1954, Uhlig 1956, Mosby 1969, Flyger and Gates 1982). In urban residential areas gray squirrels are frequently viewed as pests.

MANAGEMENT SUGGESTIONS

Habitat improvement for gray squirrels centers on forest structure and species composition. Changes in forest structure affect overstory and understory densities, and tree cavity availability; tree species composition markedly affects mast production. Nixon and Hansen (1987) discuss silvicultural options regarding harvest, stocking levels, and rotation lengths to maintain populations in the Midwest. The primary factor in maintaining populations in residual forest stands after harvest is sustaining adequate levels of winter-storable tree seeds (Nixon et al. 1975). Residual stands require 1.3–1.5 m²/ha basal area of mast producing trees, including a 50:50 mix of fall/spring germinating oaks, and a minimum of 0.4–0.6 m²/ha basal area of hickory (Nixon et al. 1975). Nixon and Hansen (1987) present guidelines for providing squirrels with 80 percent of their annual diet (e.g., 30 percent oak, 40 percent hickory, and 10 percent walnut). Nixon et al. (1980a) report that removal of 37–55 percent of stand basal area using single-tree or small group-selection harvests had no short-term (i.e., 1–2 years) effect on gray squirrel densities, breeding rates, or annual survival. In contrast, large-scale clearcutting of forested areas can result in a decline of squirrel populations (Nixon et al. 1980b). However, the use of small (<8 ha) and narrow (<160 m) clearcuts in forests where 40–60 percent is retained at seed-producing age should not substantially impact populations (Nixon et al. 1980b). Chamberlain et al. (1999) and Yarrow and Yarrow (1999) provide management guidelines specific to Mississippi and Alabama, respectively. Streamside management zones of hardwood and mixed-pine stands composed of mature timber of mixed age and species composition offer an option for providing high-quality habitat and travel corridors for gray squirrels in areas influenced by even-aged management.

Gray squirrels also benefit from the promotion and retention of cavity trees, and the provision of artificial cavities. Nixon and Hansen (1987) recommend 5–7 tree cavities/ha and 5–7 artificial cavities/ha for high squirrel densities. The addition of artificial cavities increases carrying capacity for gray squirrels (Burger 1969, Barkalow and Soots 1975). Moreover, artificial cavities may allow gray squirrels to recolonize a young stand 30–60 years after clearcutting, if food is not a limiting factor (Nixon and Donohoe 1979). Sanderson (1975) recognizes the difficulty in providing tree cavities in intensively managed forests and suggests the following options: (1) retention of existing trees with cavities; (2) retention of potential cavity trees; (3) option 2, plus treatment (e.g., stub-pruning) of selected trees to form

cavities; and (4) option 2 and 3, plus provision of artificial cavities. In the Midwest and Northeast, canopy-reaching vines serve as important support structures for leaf nests. Sanderson et al. (1980) and Nixon and Hansen (1987) recommend retention of 2–4 vines/ha for gray squirrels.

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Eastern Fox Squirrel

Sciurus niger (Linnaeus, 1758)

John W. Edwards and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

Ten subspecies of the eastern fox squirrel are recognized, nine of which occur in the South: *S. n. avicennia*, *S. n. bachmani*, *S. n. cinereus*, *S. n. ludovicianus*, *S. n. niger*, *S. n. rufiventer*, *S. n. shermani*, *S. n. subauratus*, and *S. n. vulpinus* (Hall 1981, Koprowski 1994). However, recent studies indicate that in certain areas patterns of morphological variation do not correspond to currently recognized subspecies boundaries (Turner and Laerm 1993, Roe 1994) while in other areas they do (Moncrief 1993). Limited genetic studies show similar problems (Moncrief 1993). Turner and Laerm (1993) and Roe (1994) suggest that populations referable to *S. n. shermani* occur in southern Florida north to the Piedmont of Georgia, well beyond their previously documented range, and that recognition of *S. n. bachmani* may not be warranted because these populations represent clinal variants of the more widespread *S. n. niger* (however, see Moncrief 1993). The life history of the fox squirrel is reviewed by Flyger and Gates (1982), Koprowski (1994), and Edwards et al. (2003).

DISTINGUISHING CHARACTERISTICS

Fox squirrels are large-sized tree squirrels. Measurements are: total length, 450–698 mm; tail, 200–330 mm; hind foot, 51–82 mm; ear, 19–30 mm; and weight, 0.5–1.2 kg. Pelage is highly variable, locally and regionally. The unusual polymorphism largely accounts for its subdivision into numerous subspecies (Hall 1981). Fox squirrel subspecies can be divided into two distinctive but intergrading coloration groups (Weigl et al. 1989). *S. n. vulpinus* (easternmost), *S. n. cinereus*, *S. n. niger*, *S. n. shermani*, *S. n. avicennia*, and *S. n. bachmani* are characterized by silver, gray, agouti, and melanistic forms with tan, gold, or reddish venter. Animals usually possess black head markings, white or gray noses, and white ears and feet. The other group consists of *S. n. vulpinus* (western), *S. n. subauratus*, *S. n. ludovicianus*, and *S. n. limitis* and is characterized by a distinctly reddish, orange, or tan agouti coloration, a grizzled or black nose, and no white markings on the head or feet. Melanism is common in the southern portions of the range (Lowery 1974, Kiltie 1989). However, Turner and Laerm (1993) conclude that pelage characteristics are too varied and subjective to permit consistent determination of subspecies in the

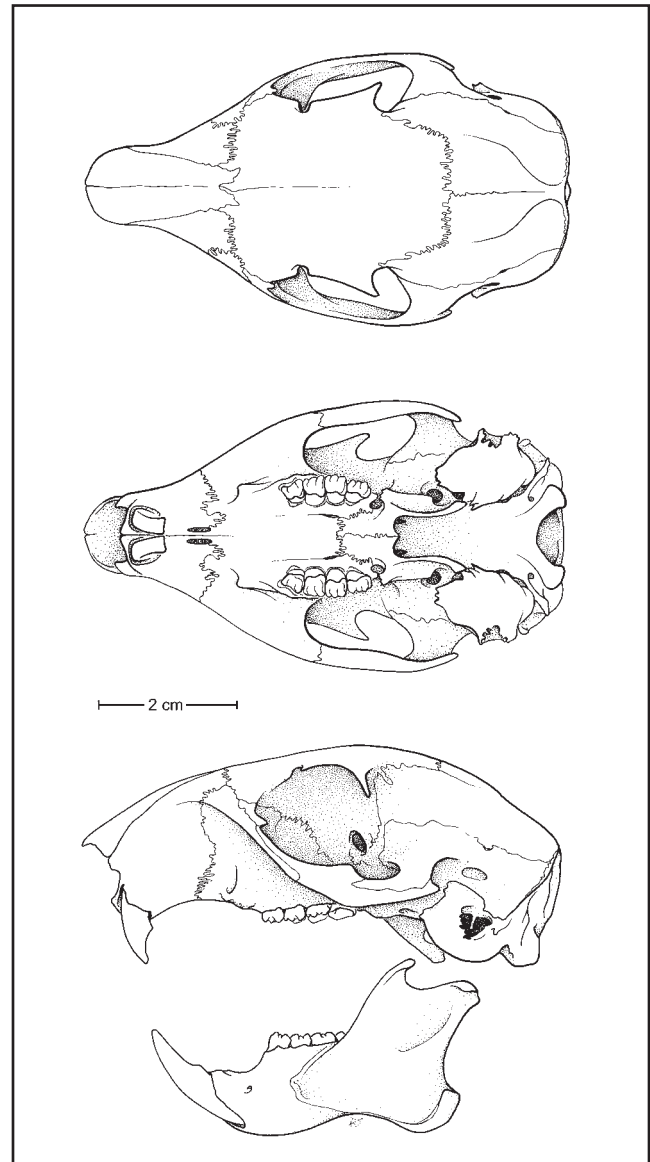


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sciurus niger* from Boston County, Georgia (USNM 276562, female).

southeastern portions of the range. The fox squirrel is readily distinguished from the smaller eastern gray squirrel (*S. carolinensis*) by its larger size and presence of only a single pair of premolars in upper and lower jaws. The dental formula is: I 1/1, C 0/0, P 1/1, and M 3/3 = 10 (Figure 1). See keys for details.

CONSERVATION STATUS

The Delmarva Peninsula fox squirrel (*S. n. cinereus*) is federally listed as Endangered (U. S. Department of Interior 2007).

The fox squirrel has a global rank of Secure (NatureServe 2007). However, it is listed as Vulnerable in North Carolina, Vulnerable/Apparently Secure in Alabama, Apparently Secure in Arkansas, South Carolina, and Virginia, and Secure in Florida, Georgia, Kentucky, Louisiana, Mississippi, Oklahoma, Tennessee, and Texas. The fox squirrel is considered a game animal; harvest seasons are regulated in all states in the South.

DISTRIBUTION

The fox squirrel ranges throughout the eastern and central United States into the High Plains as far north as Manitoba and Ontario and south into Mexico (Hall 1981). It has extended its range in the western United States along riverine corridors (Koprowski 1994) and has been widely introduced in the western United States (Flyger and Gates 1982). In Virginia, fox squirrels are rare in the Coastal Plain and Piedmont, but relatively common west of the Blue Ridge (Dueser and Handley 1991a; Figure 2).

A small, introduced population of *S. n. cinereus* is present on Assateague Island, Virginia (Dueser and Terwilliger 1987, Dueser and Handley 1991b). Fox squirrels in North Carolina are restricted to the southeastern Coastal Plain south of the Pamlico River (Weigl 1987), and although historical records indicate its former occurrence in the Piedmont and Blue Ridge (Lee et al. 1982), the species is now rare to absent (Weigl 1987). Populations are spotty throughout the Coastal Plain and Piedmont of South Carolina (Harrigal 1993) and rare to absent in the Blue Ridge. In Georgia the species is most common in the Coastal Plain and Piedmont and rare to absent in the Blue Ridge (Hillard 1979, Turner 1988, Turner and Laerm 1993). Populations are uncommon to rare on the Cumberland Plateau of eastern Kentucky but common elsewhere in the state (Barbour and Davis 1974, Fassler 1974). In Tennessee, fox squirrels are rare east of the Ridge and Valley, but common to the west (Goodpaster and Hoffmeister 1952, Schultz 1957, Linzey and Linzey 1971, Smith et al. 1974, Kennedy 1991). Fox squirrels occur throughout Florida (Moore 1956, 1957; Williams and Humphrey 1979, Humphrey and Jodice 1992, Kantola 1992), Alabama (Lowery and Davis 1942, Bakken 1952, Holliman 1963, Linzey 1970), Mississippi (Lowery and Davis 1942, Lowery 1974, Heuer and Perry 1976, Kiltie 1989, Moncrief 1993), Arkansas (Sealander and Heidt 1990), eastern Texas (Schmidly 1983, Davis and Schmidly 1994), and Oklahoma (Caire et al. 1989).

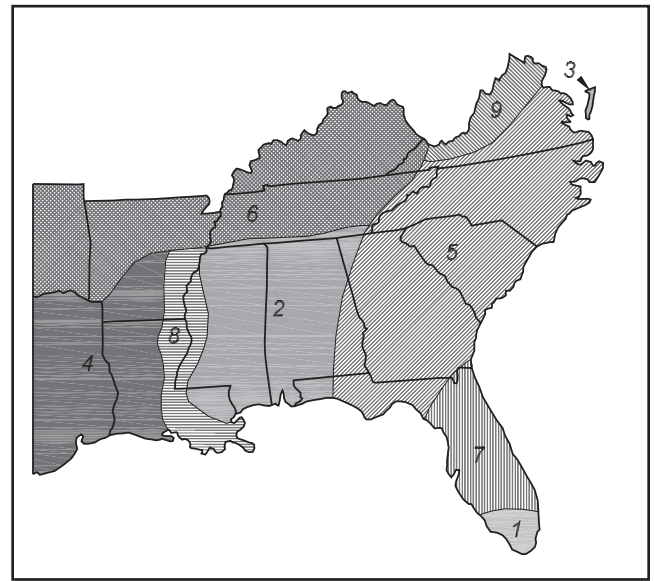


Figure 2. Distribution of *Sciurus niger* in the South: (1) *S. n. avicennia*; (2) *S. n. bachmani*; (3) *S. n. cinereus*; (4) *S. n. ludovicianus*; (5) *S. n. niger*; (6) *S. n. rufiventer*; (7) *S. n. shermani*; (8) *S. n. subauratus*; (9) *S. n. vulpinus*.

ABUNDANCE STATUS

Fox squirrel populations east of the Appalachians and along the eastern Gulf Coast have been declining dramatically over the past 100 years (Doutt et al. 1977, Webster et al. 1985, Loeb and Lennartz 1989, Weigl et al. 1989, Humphrey and Jodice 1992, Loeb and Moncrief 1993, Conner et al. 1999). However, west of the Appalachians in the midwestern and south-central states, they are considered common to abundant (Lowery 1974, Schmidly 1983, Caire et al. 1989, Sealander and Heidt 1990, Kennedy 1991). Density estimates in the South range up to 75/km² (Moore 1957, Hillard 1979, Humphrey et al. 1985, Weigl et al. 1989, Tappe 1991, Tappe et al. 1993, Lee 1999) while in the Midwest estimates range up to 510/km² (Brown and Yeager 1945, Hansen et al. 1986). Wooding (1997) suggested that differences in densities between southeastern and midwestern fox squirrel populations result from habitat quality, specifically the increased availability of hard mast.

PRIMARY HABITATS

Fox squirrels occupy a diversity of small patch, open canopy deciduous and mixed hardwood-pine habitats with an open understory (Koprowski 1994). In general, they are absent or rare in areas where forests constitute greater than 70% of available cover. Throughout the Midwest and south-central states, fox squirrels occur in mature open stands of oak-hickory (*Quercus-Carya*) and other hardwoods, mixed hardwood-pine

forests, and hardwood bottoms (Lowery 1974, Schmidly 1983, Caire et al. 1989, Sealander and Heidt 1990). Extensive agricultural development and resulting small woodlots and shelterbelt fencerows in many areas has created excellent fox squirrel habitat (Koprowski 1994). Southern fox squirrels differ substantially from midwestern subspecies in their ecological requirements; they select more pine-dominated habitats compared to deciduous habitats selected by fox squirrels in the Midwest (Weigl et al. 1989). In southern Florida, they occur in cypress (*Taxodium*) swamps, tropical hardwood, live oak (*Q. virginiana*), and mangrove forests (Humphrey and Jodice 1992). Throughout the lower Coastal Plain, fox squirrels occur in fire-maintained longleaf pine-turkey oak (*P. palustris*-*Q. laevis*) sandhills, pine flatwoods, and associated bottomland habitat (Moore 1957, Williams and Humphrey 1979, Kantola 1992, Conner et al. 1999). In the sandhills and Piedmont of the Carolinas, Georgia, and elsewhere, mixed stands of longleaf, loblolly (*P. taeda*) and shortleaf pine (*P. echinata*), hardwoods, and bottomlands are the preferred habitat (Taylor 1973, Hillard 1979, Dueser et al. 1988, Edwards et al. 1989, Loeb and Lennartz 1989, Weigl et al. 1989, Loeb and Moncrief 1993). In Louisiana and Mississippi, fox squirrels are most abundant in bottomland hardwood and streamside riparian habitats (Heuer and Perry 1976, Warren and Hurst 1980). Throughout its range, parks, golf courses, and residential areas may support substantial populations (Jodice and Humphrey 1992).

Fox squirrels use both tree cavities and leaf nests as refugia and for the rearing of young. Leaf nests occur in a variety of tree species. In midwestern populations, vines are an important component in nest site selection (Allen 1954, Sanderson et al. 1980), whereas fox squirrels in Georgia less frequently incorporate vines in their nests (Hilliard 1979, Edwards and Guynn 1995). Fox squirrels use cavities in a variety of tree species. Cavity use is greatest during winter and spring (Nixon and Hansen 1987, Edwards et al. 1989, Edwards and Guynn 1995). Hillard (1979) and Weigl et al. (1989) suggest that the absence of suitable cavity trees may be a critical factor in litter survival and subsequent recruitment of fox squirrels in Georgia and North Carolina. However, others have found no evidence that an absence of cavities was limiting fox squirrels in Maryland, Florida, or Georgia (Lustig and Flyger 1975, Kantola 1986, Edwards and Guynn 1995).

REPRODUCTION

Fox squirrels are dioestrus throughout their range with breeding concentrated in late winter/early spring and to a lesser extent in summer (Moore 1957, Hoffman and Kirkpatrick 1959, Harnishfeger et al.

1978, Weigl et al. 1989, Larson 1990). Gestation is 44 to 45 days and litter size ranges from 1–7 with reported means between 1.7–3.4 (Moore 1957, McCloskey and Vohs 1971, Harnishfeger et al. 1978, Weigl et al. 1989). Reproductive success is highly variable and dependent on demographic and environmental factors (Nixon and McClain 1969). Weaning occurs between 8–12 weeks (Allen 1942). Females reach sexually maturity at 10–14 months. Reproductive longevity averages 6–7 years up to a maximum of 12 years (Koprowski et al. 1988).

FOOD HABITS

The diet of the fox squirrel is diverse and varies seasonally by region. Nuts, seeds, buds, and flowers of oaks, hickories, beech (*Fagus grandifolia*), walnut (*Juglans* spp.), and other available hardwood species such as dogwood (*Cornus* spp.) or maple (*Acer* spp.) predominate in the diet as well as pine buds, flowers, and seeds. Soft mast such as grape (*Vitis* spp.), persimmon (*Diospyros virginiana*), cherry (*Prunus* spp.), various fruits (e.g., *Rubus* spp.), hypogeous and epigeous fungi, and insects are also eaten. See Flyger and Gates (1982), Koprowski (1994), and Edwards et al. (2003) for an extensive review of food habits.

ASSOCIATED SPECIES

The fox squirrel is associated with regionally diverse groups of deciduous and mixed hardwood-pine forest small mammals. It occurs in association with other sciurids including the gray squirrel, southern flying squirrel (*Glaucomys volans*), and eastern chipmunk (*Tamias striatus*).

VULNERABILITY AND THREATS

The fox squirrel is viewed as threatened throughout much of its range in the South particularly east of the Appalachians, due to widespread loss of preferred mature, open pine-oak forests and associated bottomland and swamp habitats. This has resulted from large-scale monocultural replacement of longleaf pine by loblolly pine, shortened stand rotation, loss of hardwoods, and fire suppression (Weigl 1987, Loeb and Lennartz 1989, Weigl et al. 1989, Dueser and Handley 1991a, 1991b; Humphrey and Jodice 1992, Kantola 1992). To the west of the Appalachians, however, past land use practices, especially conversion of closed canopy hardwood forest to woodlots, agricultural lands, fencerows, and shelterbelts as well as small patch prairie forestation and residential development has favored the fox squirrel.

The fox squirrel is a popular small game animal. Hunting mortality is considered compensatory to

some extent, and is generally not thought of as a major factor controlling squirrel populations. However, intensively hunted populations may be particularly vulnerable to over harvest depending on their level of isolation and potential for recolonization from nearby refuges or other lightly hunted areas (Allen 1943, Allen 1954, Nixon et al. 1974, Herkert et al. 1992). In many states, no distinction is made between hunting regulations for fox and gray squirrels. Tappe and Guynn (1998) suggested that it might be appropriate to manage southern fox squirrels as a relatively K-selected species when compared to the gray squirrel or other small game species. Fox squirrels in the South appear to be relatively long-lived, have smaller and fewer litters, and lower adult mortality. Ecological differences between gray and fox squirrels suggest the use of different strategies to manage harvest levels of each species in the South (Tappe and Guynn 1998, Conner 2001).

MANAGEMENT SUGGESTIONS

Habitat improvement practices for fox squirrels generally target forest structure and species composition. Changes in forest structure affect overstory and understory densities, and tree cavity availability; tree species composition markedly affects mast production. Specific management practices vary among regions. Nixon and Hansen (1987) discussed silvicultural options and management recommendations regarding harvest, intermediate practices, stocking levels, and rotation lengths to maintain populations of fox squirrels in the Midwest. The primary factor in maintaining populations in residual forest stands (i.e., post harvest) is sustaining adequate levels of winter-storable tree seeds (Nixon et al. 1975). Chamberlain et al. (1999) and Yarrow and Yarrow (1999) provide management guidelines specific to Mississippi and Alabama, respectively. Streamside management zones offer an option for providing high-quality habitat and travel corridors for fox squirrels in areas affected by even-aged management. Studies in the southeast suggest that management practices that reduce dense understory vegetation and promote retention of mature mast-producing hardwood will benefit fox squirrels. Such practices include the use of prescribed fire, mowing, and retention of hardwood stringers in pine-dominated habitats (Lustig and Flyger 1975, Kantola 1986, Edwards et al. 1989, Lee 1999, Chamberlain et al. 1999, Conner et al. 1999).

Fox squirrels also benefit from the promotion and retention of cavity trees. Although of benefit to both fox and gray squirrels, cavities are a more important habitat component for gray squirrels (Sanderson et al. 1980, Flyger and Gates 1982, Edwards and

Guynn 1995). In the Midwest and Northeast, canopy-reaching vines serve as important structures for fox squirrel leaf nests. Sanderson et al. (1980) and Nixon and Hensen (1987) recommend >4–6 vines/ha for fox squirrels.

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Eastern Chipmunk

Tamias striatus (Linnaeus, 1758)

Michael T. Mengak and Joshua Laerm

CONTENT AND TAXONOMIC COMMENTS

The taxonomy of the eastern chipmunk is unclear in the South. Hall (1981) recognized 11 subspecies. Jones and Sutkus (1979) examined morphological variation in Alabama, Florida, Georgia, Louisiana, and Mississippi and concluded that the distinction of *T. s. pipilans* (Lowery 1943) was unjustified and should be regarded a synonym of *T. s. striatus*. Ellis (1979) examined the systematics of the species throughout its range and recognized only five subspecies, regarding *T. s. pipilans* and *T. s. striatus* as distinct. Snyder (1982) commented on the contrasting views of Hall (1981) and Ellis (1979). Jones et al. (1992) followed Jones and Sutkus (1979). In view of the fact that the Jones and Sutkus (1979) study was regional in extent, and that Ellis (1979) examined the entire range of the species, we follow the arrangement of the former and Hall (1981). Thus, four subspecies occur regionally: *T. s. fisheri*, *T. s. pipilans*, *T. s. striatus*, and *T. s. venustus*. Ellis (1979) and Snyder (1982) review the life history of the eastern chipmunk.

DISTINGUISHING CHARACTERISTICS

The eastern chipmunk is a small sciurid, characterized by prominent stripes and flattened tail. The measurements are: total length, 215–299 mm; tail, 72–113 mm; hind foot, 34–38 mm; ear, 17–21 mm; weight, 80–130 g. The upper parts are grizzled gray to reddish brown with several diagnostic stripes: a narrow, dark brown mid dorsal stripe extends from head to rump, bordered on either side by wider grizzled grayish orange to brown stripes and two lateral narrow dark brown stripes enclosing slightly wider whitish stripe. A light facial stripe above each eye is bordered by a darker stripe. The sides are grizzled yellow to reddish brown and the underparts are whitish. The tail is well haired and flattened, dark above and gray below. The skull is characterized by an infraorbital opening piercing the zygomatic plate. The dental formula is $I\ 1\frac{1}{1}$, $C\ 0\ 0\ 0$, $P\ 1\frac{1}{1}$, $M\ 3\frac{3}{3}$ = 20 (Figure 1). See keys for details.

CONSERVATION STATUS

The eastern chipmunk has a global rank of Secure (NatureServe 2007). It is considered Secure in Alabama, Georgia, Kentucky, Mississippi, North Carolina, Tennessee, and Virginia. It is Apparently Secure in

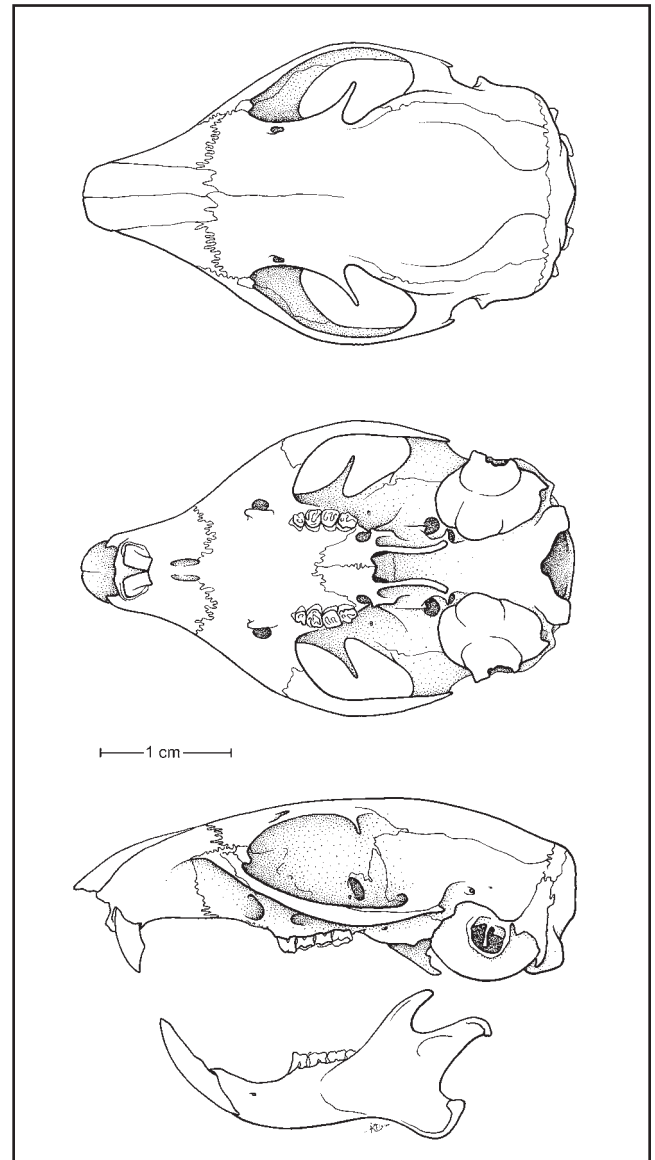


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Tamias striatus* from Russell County, Virginia (USNM 318716, male).

Arkansas and Oklahoma. Louisiana classifies it as Vulnerable and Florida lists the eastern chipmunk as Imperiled. It is unranked in South Carolina.

DISTRIBUTION

The eastern chipmunk ranges throughout the eastern half of North America, but is absent from much of

the Coastal Plain (Figure 2). It occurs throughout Virginia (Bailey 1946, Handley and Patton 1947, Handley 1979, 1992; Cranford and Maly 1986, Bowers 1995, Linzey 1998), but does not occur below the Piedmont of North Carolina (Odum 1949, Johnston 1967, Lee et al. 1982). South Carolina records are restricted to the upper Piedmont (Golley 1966). Populations range as far south as the Coastal Plain in western Georgia (Golley 1962, Laerm et al. 1982, Jones and Sutkus 1979) and into northern portions of the panhandle of Florida (Stevenson 1962, Jones and Sutkus 1979, Gore 1990, Jones et al. 1992). It occurs throughout all but immediate coastal regions of Alabama (Howell 1921, Holliman 1963, Jones and Sutkus 1979, Blackmore and Lishak 1985) and Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Sutkus 1979, Jones and Carter 1989). Louisiana populations are restricted to upland hardwoods of east Baton Rouge and west Feliciana parishes (Lowery 1943, 1974; Thomas 1974, Jones and Sutkus 1979). They occur throughout Tennessee (Calhoun 1941, Goodpaster and Hoffmeister 1952, Howell and Conaway 1952, Conaway and Howell 1953, Linzey and Linzey 1971, Smith et al. 1974, Dueser and Shugart 1978, 1979; Dueser and Hallett 1980, Kitchings and Levy 1981, Durden 1984, Kennedy 1991), Kentucky (Barbour and Davis 1974, Fassler 1974, Severinghaus et al. 1980, McComb and Rumsey 1982, McGehee-Marsh et al. 1992), Arkansas (Sealander and Heidt 1990, Tumblison et al. 1992), and range into eastern Oklahoma (Caire et al. 1989).

ABUNDANCE STATUS

Tamias striatus is common to abundant throughout its range in the South. Population densities vary depending upon time of year and habitat, but are reported as 0.3–37.6/ha (Yerger 1953, Snyder 1982). Throughout the winter, eastern chipmunks remain in their underground burrows in varying stages of torpor, but they may appear above ground in favorably warm weather, particularly in more southern portions of the range (Engles 1951, Stevenson 1962, Pivorun 1977, Yahner and Svendsen 1978, Jones and Sutkus 1979). Eastern chipmunk population densities are correlated to prior year acorn crop (Ostfeld et al. 1996, Wolff 1996).

PRIMARY HABITATS

Eastern chipmunks occur typically in mature woodlots at sites with a high tree density and a low shrub density. It is much more common in forest habitats including northern hardwoods, cove hardwood, oak-hickory (*Quercus-Carya*), and mixed pine (*Pinus* spp.)-hardwood cover types, than in grass or shrub habitats, although they occur here as well as brushy areas (Dueser and Shugart 1978, 1979; Svendsen and

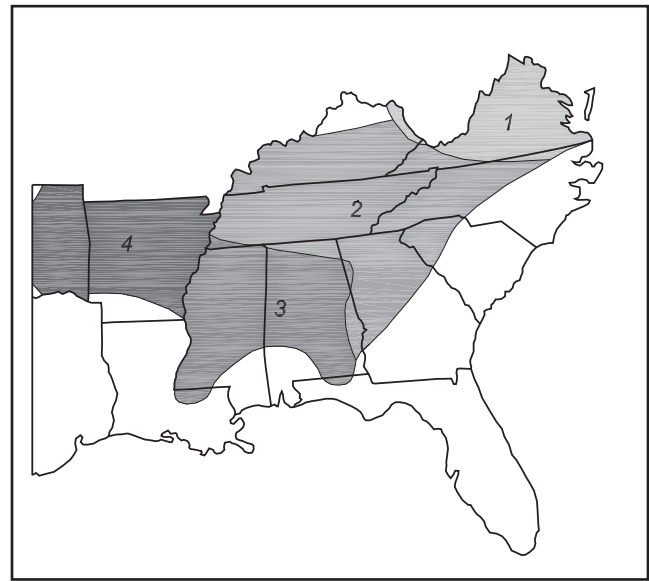


Figure 2. Distribution of *Tamias striatus* in the South: (1) *T. s. fisheri*; (2) *T. s. striatus*; (3) *T. s. pipilans*; (4) *T. s. venustus*.

Yahner 1979, Dueser and Hallett 1980). The negative association between eastern hemlock (*Tsuga canadensis*) and eastern chipmunk activity is presumably related to lack of ground cover, lack of food resources, or both (Pyare et al. 1993). It is less common in coniferous woodlands (Linzey and Linzey 1971), agricultural areas and ecotones (Henderson et al. 1985). It is often very common in residential and urban areas, wooded parks, and cemeteries (Ryan and Larsen 1976). Clearcutting had no influence on the demography of eastern chipmunks in a Pennsylvania study (Mahan and Yahner 1998); Nupp and Swihart (1998) suggested eastern chipmunks might be negatively affected by forest fragmentation due to increased predation. Eastern chipmunks readily use regenerating stands (Mahan and Yahner 1998) and forested corridors to travel between mature forests and agricultural landscapes (Mahan and Yahner 1998). Edges are important at times when hard mast is unavailable because edges provide soft mast and invertebrates.

Although not exclusively, the eastern chipmunk is associated with stands of mast species such as oak, hickory, and American beech (*Fagus grandifolia*). It is dependent upon habitats with abundant structural crevices for refugia and elevated observation and vocalization posts. Complex tunnels are created beneath rock piles, rubbish heaps, downed logs, roots, and stumps. Tunnels are often quite extensive (Panuska and Wade 1956, Thomas 1974, Elliott 1978, Yahner 1978). Eastern chipmunks rarely nest in trees. Eastern chipmunk home ranges in oak and mixed mesophytic forests of the central Appalachians typically were

less than 1 ha and with considerable conspecific overlap (Rowan et al. 2005).

REPRODUCTION

Breeding is concentrated between February–April and late June–early July. Females usually breed in spring but may breed again in summer (Smith and Smith 1972, Yerger 1955). Gestation ranges from 31–32 days (Allen 1938, Burt 1940). Average litter size is 4–5 with a range of 2–6. Sexual maturity occurs after the first winter, but some females may breed their first summer (Allen 1938, Smith and Smith 1972, Yerger 1955).

FOOD HABITS

Eastern chipmunks hoard foodstuffs in their burrows and they scatter hoard outside. Dominant overwinter foods include acorns, nuts, and other seeds; during spring and summer, invertebrates, fungi, and occasionally small vertebrates are consumed (Yerger 1955, Yahner 1975, Elliott 1978, Wrazen and Svendsen 1978, Blackmore and Lishak 1985, Ciraldeau 1994, Clarke 1994). Eastern chipmunks may prefer low tannin white oak (*Q. alba*) to northern red oak (*Q. rubra*) acorns; beechnuts are also preferred (Pyare et al. 1993).

Eastern chipmunks may consume ground-nesting songbird eggs (Ostfeld et al. 1996). However, Liemgruber et al. (1994) found little or no evidence of predation on artificial nests. Reitsma et al. (1990) noted that both eastern chipmunks and red squirrels (*Tamiasciurus hudsonicus*) were photographed preying on eggs.

ASSOCIATED SPECIES

Eastern chipmunk is associated with several hardwood and mixed hardwood-coniferous forest small mammals including the northern short-tailed shrew (*Blarina brevicauda*), masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), deer mouse (*Peromyscus maniculatus*), white-footed mouse (*P. leucopus*), cotton mouse (*P. gossypinus*), golden mouse (*Ochrotomys nuttalli*), Allegheny woodrat (*Neotoma magister*), and southern red-backed vole (*Clethrionomys gapperi*). Other sciurid associations include the southern flying squirrel (*Glaucomys volans*), eastern gray squirrel (*Sciurus carolinensis*), fox squirrel (*S. niger*), and red squirrel.

VULNERABILITY AND THREATS

The eastern chipmunk is common throughout eastern deciduous woodlands, as well as urban and residential areas. Nowhere does it appear to be vulnerable

to habitat loss or alteration. The hantavirus was not detected in individuals tested in Arkansas (Perry et al. 1997). Eastern chipmunks may be susceptible to infection by the northern raccoon (*Procyon lotor*) roundworm (*Baylisascariasis procyonis*; Page et al. 1999).

MANAGEMENT SUGGESTIONS

Management to retain mast-producing plants will benefit the eastern chipmunk. Refugia such as logs, stumps, and rock outcrops should not be disturbed. The complete ecological relationship of coarse woody debris within eastern chipmunk habitat has not been fully investigated. Limited evidence suggests that light prescribed fire that does not overly reduce coarse woody debris loadings does not impact eastern chipmunk behavior or habitat use (Rowan et al. 2005).

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Red Squirrel

Tamiasciurus hudsonicus (Erxleben, 1777)

Joshua Laerm and Wm. David Webster

CONTENT AND TAXONOMIC COMMENTS

Twenty-five subspecies of *Tamiasciurus hudsonicus* currently are recognized (Hall 1981), two of which occur in the South. *Tamiasciurus h. abieticola* is restricted to the mountainous portions of Virginia and West Virginia, and thence southward along the southern Appalachians, whereas *T. h. loquax* is distributed from the Piedmont of Virginia northward (Howell 1929, Kellogg 1937, Handley and Patton 1947, Hall 1981). Literature reviews are in Flyger and Gates (1982) and Obbard (1987).

DISTINGUISHING CHARACTERISTICS

This is the smallest of the North American tree squirrels, easily recognized by its distinct coloration, nervous scampering activity, and nearly constant chatter. External measurements are: total length, 300–350 mm; tail, 95–140 mm; hind foot, 42–55; ear, 20–25 mm; weight, 145–260 g. The red squirrel is readily distinguished from other tree squirrels by its pelage, which is reddish brown above, whitish below, and distinctly separated by a black lateral stripe in its summer pelage. A white ring encircles each eye. The cranium is wide and it typically lacks the interorbital notch, and the anterior border of the orbit is opposite the last premolar when viewed from beneath. The dental formula is I 1/1, C 0/0, P 1/1, M 3/3 = 20 (Figure 1), although another vestigial upper premolar may also be present. See keys for details.

CONSERVATION STATUS

The red squirrel has a global rank of Secure (NatureServe 2007). It is also ranked Secure in North Carolina and Virginia, and Apparently Secure in Tennessee. It is considered Vulnerable in Georgia and South Carolina.

DISTRIBUTION

The red squirrel is associated with boreal forests throughout much of Canada and the northern United States, with significant range extensions southward into the coniferous forests of the Rocky Mountains in the west and Appalachian Mountains in the east (Smith 1970; Figure 2). In the region, it is distributed widely in northern and western Virginia (Linzey

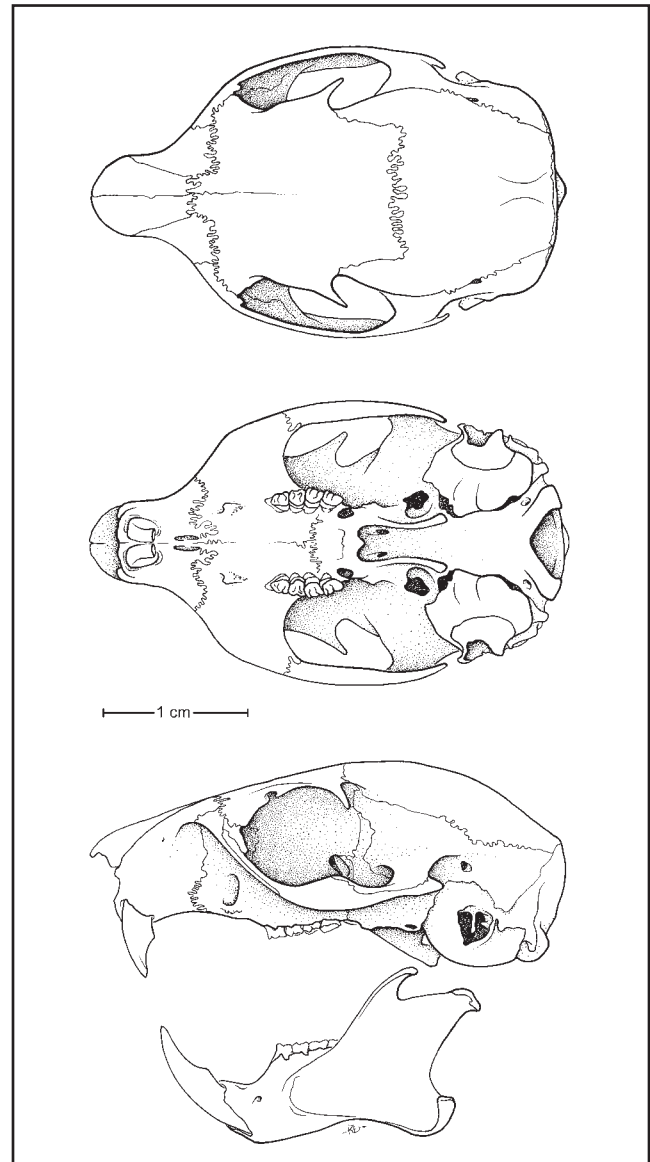


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Tamiasciurus hudsonicus* from Frederick County, Maryland (USNM 505585, male).

1998), but it also has been reported from the Piedmont as far east as Henrico County (Handley and Patton 1947, Webster et al. 1985, Handley 1992). In North Carolina, it generally is restricted to the western mountains (Lee et al. 1982, Webster et al. 1985); occasional records in the Piedmont apparently

do not represent established populations (Lee et al. 1982, Webster et al. 1985). The red squirrel is limited to higher elevations in extreme northwestern South Carolina (Sherman 1937, Golley 1966), northeastern Georgia (Johnston 1959, Golley 1962, Wharton 1968, Laerm 1981, Laerm et al. 1982), and eastern Tennessee (Conaway and Howell 1953, Komarek and Komarek 1938, Linzey and Linzey 1971, Smith et al. 1974).

ABUNDANCE STATUS

There are few population estimates available for the South; however, several local accounts indicate that the red squirrel is common to abundant in appropriate habitat (Conaway and Howell 1953, Wharton 1968, Linzey and Linzey 1971, Smith et al. 1974, Webster et al. 1985, Linzey 1998), reaching population densities of 1.3–1.5/km² in eastern Tennessee (Stevens and Kennedy 1999). In the northeastern United States and southeastern Canada, population densities range from 46–667 squirrels/km², depending upon habitat, with highest densities occurring (in decreasing density) in red spruce (*Picea rubens*), mixed conifer, and hardwood-conifer stands (Klugh 1927, Hatt 1929, Williams 1936, Layne 1954, Obbard 1987). Apparently the size of the cone or mast crop upon which red squirrels feed is the primary factor controlling density (Obbard 1987, Riege 1991), although red squirrels are quick to relocate when food resources are scarce (Wheatley et al. 2002).

PRIMARY HABITATS

In the eastern United States, red squirrels inhabit a diversity of vegetative communities (Hatt 1929, Layne 1954, Smith 1970, Obbard 1987), including spruce-fir (*Picea-Abies*), maple-beech-birch (*Acer-Fagus-Betula*), and white pine-eastern hemlock (*Pinus strobus-Tsuga canadensis*) forests. In some cases, they occupy mixed pine-hardwood and pure hardwood stands, but only if mature cone producing red spruce (*Picea rubens*) and American beech (*Fagus grandifolia*), oak (*Quercus* spp.), hickory (*Carya* spp.), black walnut (*Juglans nigra*) mast is present. In the southern Appalachians, red squirrels are most commonly associated with spruce-Fraser fir (*A. fraseri*) and white pine-hemlock stands (Howell 1929, Odum 1949, Wharton 1968, Linzey and Linzey 1971), although they occasionally are found in overgrown orchards, hedgerows, and parks. Adequate food supply, suitable nest sites, protective cover, and moisture and shade for cone storage are major habitat requirements (Layne 1954, Obbard 1987). In hardwood forests, tree cavities are the preferred nest sites; however, outside tree nests, including leaf nests and underground nests, are common in coniferous forests. Underground nests are typically in decaying stumps, rotting logs, fences,

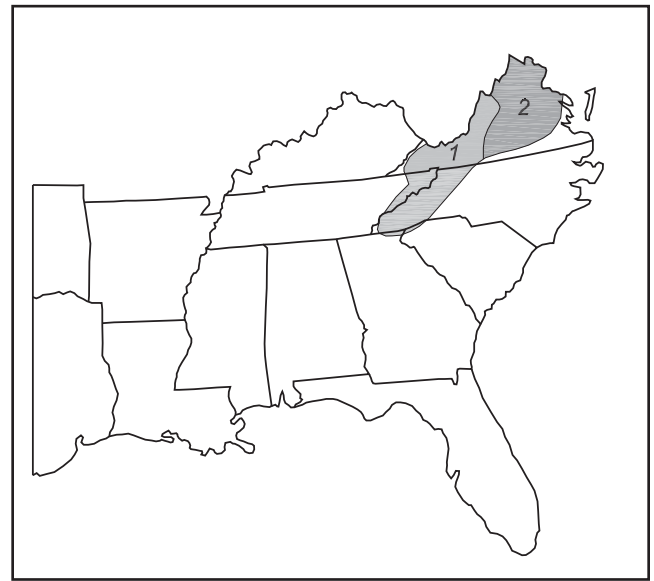


Figure 2. Distribution of *Tamiasciurus hudsonicus* in the South: (1) *T. h. abieticola*; (2) *T. h. loquax*.

and hedgerows (Hatt 1929, Layne 1954, Yahner 1980, Obbard 1987).

REPRODUCTION

Breeding extends from mid-January to late September with peaks in spring and late summer (Klugh 1927, Layne 1954). In the northernmost portions of its range there is a single litter per year, but in more southern populations two litters may be produced (Klugh 1927, Hamilton 1939, Layne 1954, Wrigley 1969, Obbard 1987, O'Donoghue and Boutin 1995). Gestation is approximately 35 days (Hamilton 1939, Ferron and Prescott 1977, Lair 1985a) and the first litters are born in late spring. Second litters may be conceived in June or July; young are born in September or October (Layne 1954, Lair 1985b). Litter size varies from 1–8 with means ranging from 4.2–5.4 in the eastern United States and Canada (Layne 1954, Millar 1970, Ferron and Prescott 1977, Lair 1985a). Young are weaned in 7–8 weeks, disperse by 3 months, and reach sexual maturity in 10–12 months (Layne 1954, Lair 1985a, Obbard 1987); survivorship is greater for young born earlier in the year (O'Donoghue and Boutin 1995).

FOOD HABITS

Red squirrels spend over half their active period collecting, consuming, and storing food (Benhamou 1996). In coniferous forests, they exist heavily on the conifer seeds. They cut, gather, and cache huge quantities of newly mature cones; these caches may be quite large and sufficient to sustain them during years of cone crop failure. In deciduous forests, red

squirrels gather and cache seeds and nuts of various species including oaks, hickories, maples, elm (*Ulmus* spp.), tulip tree (*Liriodendron tulipifera*), American beech, hazelnut (*Corylus* spp.), and walnut (Klugh 1927, Hatt 1929, Layne 1954, Smith 1968, Obbard 1987). Red squirrels will also gather and cache large species of fungi and drink the concentrated sap of sugar maples (Heinrich 1992).

ASSOCIATED SPECIES

This species is commonly found in association with other sciurids such as the eastern gray squirrel (*Sciurus carolinensis*), northern flying squirrel (*G. sabrinus*), and eastern chipmunk (*Tamias striatus*). In addition, red squirrels are found with several species of insectivores and rodents that inhabit the Southern Appalachian Mountains, including the masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), northern short-tailed shrew (*Blarina brevicauda*), hairy-tailed mole (*Parascalops breweri*), deer mouse (*Peromyscus maniculatus*), southern red-backed vole (*Clethrionomys gapperi*), and woodland jumping mouse (*Napaeozapus insignis*).

VULNERABILITY AND THREATS

The red squirrel is common throughout the boreal forests across much of northern North America. In the southern Appalachians it is largely restricted to high elevation boreomontane remnants including red spruce-Fraser fir communities, which is the region's most rare and rapidly declining forest cover type (White et al. 1993). While red squirrels are less abundant in mixed hardwood-conifer and pure hardwood stands, they appear to do well in these habitats (Obbard 1987). These cover types are more widely distributed and under less severe threats. However, the status of red squirrels in the southern Appalachians is poorly known, and information presented here is largely anecdotal and based on older descriptive surveys or drawn from studies conducted in other portions of the range of the species.

MANAGEMENT SUGGESTIONS

Conservation of red squirrels in the South depends on the existence of a variety of dense forest types that provide adequate forage (especially red spruce), nest sites, nesting material, cover, and the cool damp climate necessary for successful cone storage. This is most critical along the southern limit of the red squirrel's range, where it is restricted to higher elevations in the southern Appalachians.

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Woodland Jumping Mouse

Napaeozapus insignis (Miller, 1891)

Joshua Laerm and John F. Pagels

CONTENT AND TAXONOMIC COMMENTS

The ranges of the five subspecies of *Napaeozapus insignis* are depicted in Hall (1981). Only *N. i. roanensis* occurs in the South. Whitaker and Wrigley (1972) reviewed earlier literature on the woodland jumping mouse.

DISTINGUISHING CHARACTERISTICS

The woodland jumping mouse is characterized by a very long tail and large hind feet. These are adaptations for its saltatorial mode of locomotion. Its measurements are: total length, 205–255 mm; tail, 115–160 mm; hind foot, 28–34 mm; ear, 14–18 mm; weight, 15–26 gm. The pelage is characterized by a brown to blackish mid-dorsal stripe, orange to reddish brown sides, and a white venter. The tail is distinctly bicolor, brown above white below, and tipped with white hairs. The upper incisors are grooved as in the meadow jumping mouse, *Zapus hudsonius*. Unlike the meadow jumping mouse, it lacks small premolars. The dental formula is $I\ 1/1, C\ 0/0, P\ 0/0, M\ 3/3 = 16$ (Figure 1). See keys for details.

CONSERVATION STATUS

Napaeozapus insignis has a global rank of Secure (NatureServe 2007). It is considered Vulnerable in Georgia and Apparently Secure in Kentucky, North Carolina, South Carolina, and Tennessee. In Virginia, *N. insignis* is classified as Secure.

DISTRIBUTION

Figure 2 depicts the distribution of the woodland jumping mouse in the South. The woodland jumping mouse ranges throughout much of southeastern Canada and the northeastern United States south throughout higher elevations of the Appalachian Mountains of Virginia (Handley and Patton 1947, Cranford and Maly 1986, Mitchell et al. 1997, McShea et al. 2003), eastern Kentucky (Barbour and Davis 1974, Davis and Barbour 1979, Caldwell 1980, Houtcooper 1982, Meade 1992, Kiser and Meade 1993), eastern Tennessee (Linzey and Linzey 1968, Smith et al. 1974, Kennedy and Harvey 1980, Harvey et al. 1991, 1992; Todd 1992), western North Carolina (Johnston 1967, Gentry et al. 1968, Lee et al. 1982), extreme northwestern South Carolina (Coleman 1940, Golley 1966)

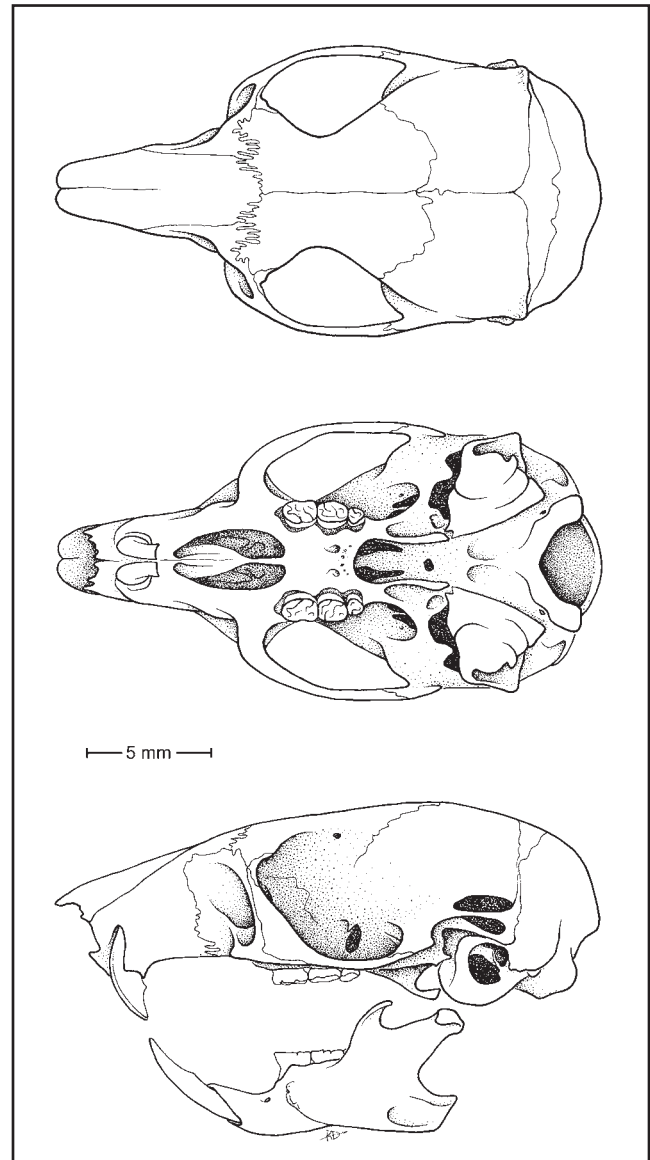


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Napaeozapus insignis* from Madison County, New York (USNM 155524, female).

and northeastern Georgia (Autrey and Odum 1949, Golley 1962, Laerm 1981, Ford et al. 1994, Laerm et al. 1996).

ABUNDANCE STATUS

Linzey and Linzey's (1968) observation that the woodland jumping mouse is not uncommon but that it occurs in localized populations seems to apply to the species throughout the region. Usually found above 800 m elevation, the woodland jumping mouse is widely distributed in the Appalachians and its often localized occurrence sometimes suggests that it is uncommon. Although population estimates are not available for the South, densities have been reported elsewhere from 0.6–12.8/ha (Whitaker and Wrigley 1972). Most faunal reports for the region indicate it to be uncommon, but not particularly rare in comparison to other small mammals (Johnston 1967, Gentry et al. 1968, Whitaker et al. 1975, Harvey et al. 1991, 1992; Ford et al. 1994, Laerm et al. 1996, McShea et al. 2003). Cranford and Maly (1986) found it was the most abundant small mammal in a Virginia study, and in another Virginia study of several habitats the numbers of the woodland jumping mice captured equaled those of the common white-footed mouse (*Peromyscus leucopus*), deer mouse (*P. maniculatus*), and northern short-tailed shrew (*Blarina brevicauda*; Mitchell et al. 1997). Wrigley (1972) reported its abundance might be negatively affected by the presence of the meadow jumping mouse in edge habitats and the red-backed vole (*Clethrionomys gapperi*) in woodlands. Cranford and Maly (1986) found higher numbers of woodland jumping mice than meadow jumping mice in a small wildlife clearing.

PRIMARY HABITATS

The woodland jumping mouse inhabits high-elevation forests that include red spruce-fir (*Picea rubens*-*Abies* spp.), maple-beech-birch (*Acer*-*Fagus*-*Betula*), cove hardwood, white pine-hemlock (*Pinus strobus*-*Tsuga* spp.) and oak-hickory (*Quercus*-*Carya*). It is often suggested to be restricted to cool moist environments and many studies have reported it in association with streams, bogs, and swamps (Wrigley 1972, Whitaker et al. 1975). In the southern portions of its range, its occurrence in mesic forests may confer the importance of nearby water. McShea et al. (2003) found that the closer non-mesic sampling sites were to mesic patches the more likely the presence of *N. insignis*. Other studies report no particular association with habitats near water (Connor 1960, Whitaker 1963) but suggested the importance of ground cover. Dense herbaceous ground cover, (e.g. sedges, grasses and low woody vegetation), which is often more abundant near water, as well as structural debris, including logs and moss covered rocks, appear to be important in its micro-habitat selection (Whitaker 1963, Brower and Cade 1966, Wrigley 1972). In a study of five forest types in Virginia, Mitchell et al.

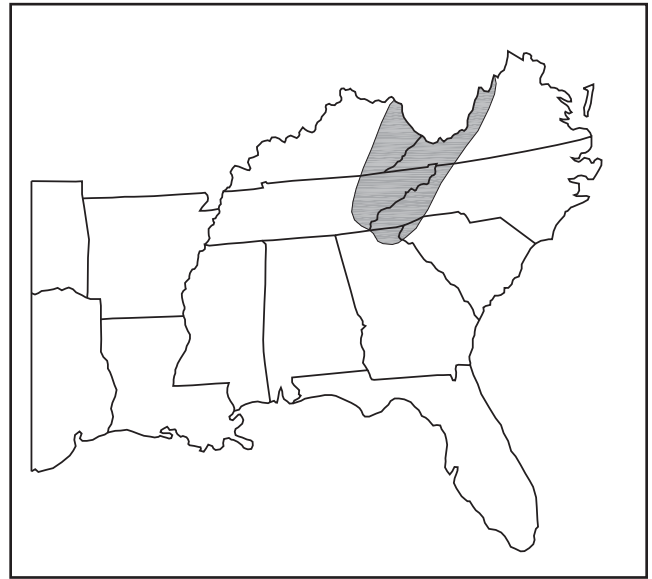


Figure 2. Distribution of *Napaeozapus insignis* in the South.

(1997) found greatest numbers of *N. insignis* at a site that included many of the aforementioned features—a forest of large hardwoods with mid- and understory vegetation, a summer cover of fern, and a nearby seep area. Cranford and Maly (1986) found relatively high numbers of the woodland jumping mouse in an oldfield community surrounded by forest at 1250 m in Virginia. Wrigley (1972) notes that in the South the species generally does not occur in areas where the mean air temperature during the warmest months exceeds 21°C. Nests of *N. insignis* are located in shallow underground burrows, brush piles, and beneath coarse woody debris and lined with grasses and leaves (Whitaker and Wrigley 1972).

REPRODUCTION

Little specific information on reproduction in the woodland jumping mouse is available for the region. Throughout most of its range it is known to hibernate from October until May, although in the southern region it enters hibernation later and emerges earlier, at least in some areas. Active individuals were observed in November and February in the Great Smoky Mountains (Linzey and Linzey 1968). Whitaker et al. (1975) reported capturing them in April in the mountains of North Carolina. The species is reproductively active in the period May through September after emerging from hibernation. In their overview, Whitaker and Wrigley (1972) reported females breed twice a year, and in the South breeding is concentrated in two peaks in early and late summer, May and August (Wrigley 1972). Gestation is uncertain, probably between 23–29 days, and the litter size ranges from 2–7 with an average of

about 4 (Wrigley 1972). Most individuals probably do not breed until their second year. Longevity is 3–4 years (Wrigley 1972).

FOOD HABITS

The food of the woodland jumping mouse is variable and may include invertebrates in addition to its main staples of fungi and various forms of plant material, including seeds and small nuts (Whitaker and Wrigley 1972, Linzey and Linzey 1973, Orrock et al. 2003). Summarizing earlier and new information, Orrock et al. (2003) noted that similar to the deer mouse, *N. insignis* consumed primarily glomelean fungi, unlike, for example, the southern red-back vole, which primarily consumes ectomycorrhizal fungi.

ASSOCIATED SPECIES

The woodland jumping mouse is usually segregated ecologically from the meadow jumping mouse, but the two may occur together in shrub or forest-edge stages of intermediate succession (Wrigley 1972), and as noted above, Cranford and Maly (1986) found higher numbers of *Napaeozapus* than *Zapus* in a small wildlife clearing. *Napaeozapus* has been found in association with most other small mammals that occur at relatively high elevations in the South, including the masked shrew (*Sorex cinereus*), smoky shrew (*S. fumeus*), long-tailed shrew (*S. dispar*), northern short-tailed shrew, deer mouse, white-footed mouse, and southern red-backed vole.

VULNERABILITY AND THREATS

Although limited in distribution in the region to the Appalachians, and restricted to relatively high elevation woodlands and forest-edge communities, *Napaeozapus* is sometimes abundant in appropriate high-elevation habitat. Because of the extent of high elevation woodland and forest-edge habitat, there do not appear to be threats to its survival anywhere in the region. Similar to other species with boreal affinities, climate change factors that could alter existing habitat conditions would negatively impact the species distribution in the South.

MANAGEMENT SUGGESTIONS

Although sometimes localized, because of its relative abundance the woodland jumping mouse should not require special management considerations in most of its range in the region. In the periphery of its range in the region, care should be given to maintenance of mesic, shaded conditions.

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Meadow Jumping Mouse

Zapus hudsonius (Zimmerman, 1780)

Joshua Laerm and John F. Pagels

CONTENT AND TAXONOMIC COMMENTS

Three of the 11 recognized subspecies of the meadow jumping mouse, *Zapus hudsonius*, occur in the South (Krutzsch 1954, Hall 1981): *Z. h. americanus*, *Z. h. intermedius*, and *Z. h. pallidus*. Whitaker (1972) reviewed the literature.

DISTINGUISHING CHARACTERISTICS

Similar to the woodland jumping mouse (*Napaeozapus insignis*), the meadow jumping mouse is characterized by a very long tail, and long hind legs and feet. Its measurements are: total length, 188–240 mm; tail, 108–145 mm; hind foot, 26–32 mm; ear, 12–15 mm; weight, 15–20 gm. Its pelage is characterized by a dark ochraceous to brownish mid-dorsal stripe, orange to reddish brown sides, and a white venter often suffused with yellowish-orange. Unlike *Napaeozapus*, the tip of the tail is not white, and it possesses four upper molariform teeth instead of three. The dental formula is I 1/1, C 0/0, P 1/0, M 3/3 = 18 (Figure 1). See keys for details.

CONSERVATION STATUS

Zapus hudsonius has a global rank of Secure (NatureServe 2007). However, it is classified as Critically Imperiled in Mississippi and Oklahoma. The species is considered Vulnerable in Alabama, Georgia, and North Carolina; Apparently Secure in Tennessee and Kentucky; and Secure in Virginia. It is unranked in Arkansas and South Carolina.

DISTRIBUTION

The meadow jumping mouse ranges throughout much of Canada from the Arctic treeline south into central and eastern United States, but there are several mistakes in earlier range maps. Distribution in the South is depicted in Figure 2. It occurs throughout most of Virginia including the barrier islands (Handley and Patton 1947, Porter and Dueser 1982, Cranford and Maly 1986, Dueser and Porter 1986, Handley 1992, Pagels et al. 1992, Kalko and Handley 1993, Bellows et al. 2001). Contrary to the range maps of Whitaker (1972) and Hall (1981), it is apparently absent from the extreme southeastern Virginia counties (Rose et al. 1990, Clark et al. 1993). Also, contrary to Handley and Patton (1947) and Webster et al.

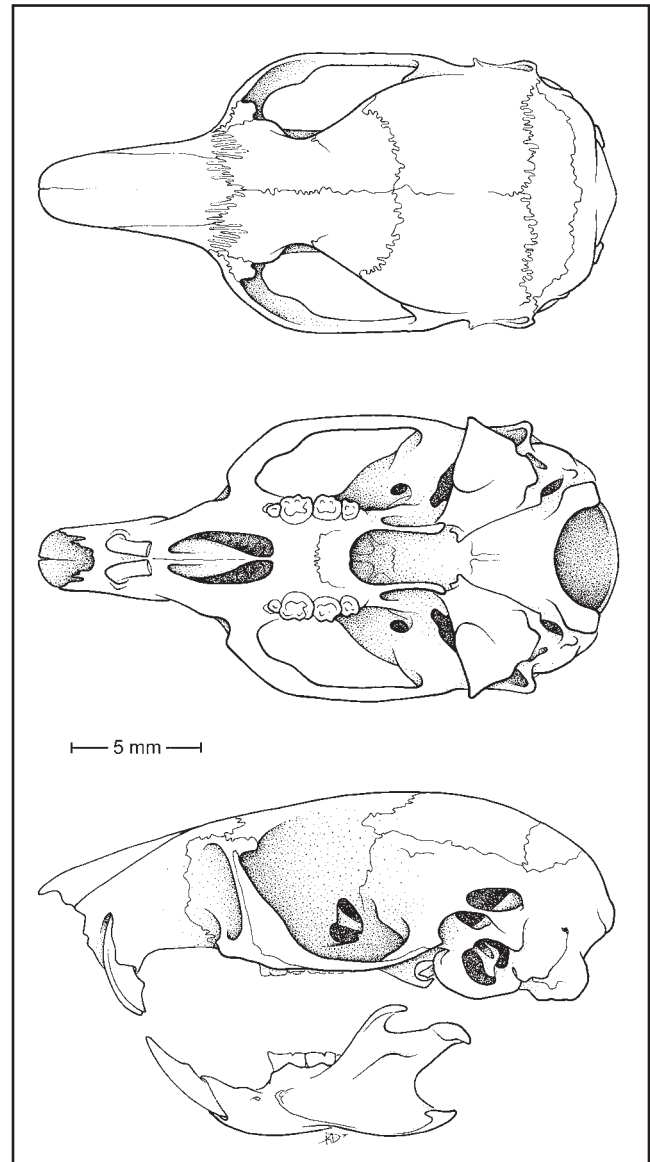


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Zapus hudsonius* from Schoolcraft County, Michigan (USNM 514454, male).

(1985), there are records of the meadow jumping mouse from extreme southwestern Virginia (Hooper and Cady 1941). Contrary to Whitaker (1972) and Hall (1981) it is restricted to the Piedmont and Blue Ridge provinces of North Carolina (Odum 1949, Johnston 1967, Linzey and Linzey 1968, Lee et al.

1982, Webster et al. 1985, W. D. Webster, University of North Carolina, personal communication) and Upper Piedmont and Blue Ridge provinces of South Carolina (Sherman 1937, Coleman 1941, 1948; Golley 1966). Laerm et al. (1996) summarized the scattered records from the Piedmont and Blue Ridge of Georgia. Alabama records are restricted to Lee County at the lower limits of the Piedmont (Sullivan 1954, Scott and French 1974, Dupree 1977, Dusi 1986). A single record has been reported from northern Mississippi (Kennedy et al. 1982). It possibly occurs statewide in Tennessee (Kennedy and Harvey 1980), but there are no records from southcentral and Ridge and Valley portions of the state (Goodpaster and Hoffmeister 1952, Linzey and Linzey 1966, 1968; Severinghaus and Beasley 1973, Smith et al. 1974, Kennedy and Harvey 1980, Harvey et al. 1991, 1992). In Kentucky, contrary to Taylor and Horn (1983), records are available only west of the Kentucky River (Barbour and Davis 1974, Houtcooper 1982, Rose and Seegert 1982); however, since it occurs in adjacent western West Virginia (Taylor and Horn 1983) and south-western Virginia (Hooper and Cady 1941) it is likely to be found in eastern Kentucky. Two specimens (referred to as *Z. h. pallidus* by Krutzsch 1954) were reported from the tallgrass prairie region of north-eastern Oklahoma by Blair (1938); however, the species was not reported from Oklahoma again until 1992 (Caire et al. 1989, Kasper et al. 1993). There have been no specimens reported from Arkansas (Sealander and Heidt 1990).

ABUNDANCE STATUS

In the northern portion of its range, meadow jumping mouse densities have been reported to range from 7.4–14.4/ha to as high as 48.3/ha (Quimby 1951). Quimby (1951) and Blair (1940) comment that numbers may fluctuate from year to year. In the southern portion of its range the meadow jumping mouse is often regarded as rare to uncommon (Goodpaster and Hoffmeister 1952, Severinghaus and Beasley 1973, Kennedy and Harvey 1980, Laerm 1981, Houtcooper 1982, Kennedy et al. 1982, Lee et al. 1982, Dusi 1986). Conversely, in several recent studies in Virginia that included the Coastal Plain (Bellows et al. 2001), Piedmont (Pagels et al. 1992), and mountains (Kalko and Handley 1993), the meadow jumping mouse was a significant component of the small mammal communities that were studied.

PRIMARY HABITATS

The meadow jumping mouse is associated with a variety of habitats that are nearly always characterized by a thick herbaceous ground cover. Habitats include moist wet lowlands, moist grassy fields,

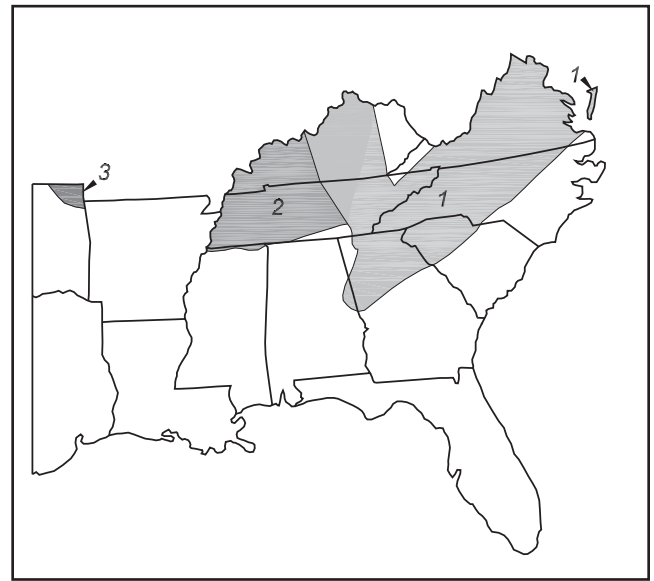


Figure 2. Distribution of *Zapus hudsonius* in the South: (1) *Z. h. americanus*; (2) *Z. h. intermedius*; (3) *Z. h. pallidus*.

lowland swamps, marshes, open woods floodplains, upland grassy fields, clearcuts, brushy woodlots, and heavy herbaceous vegetation in woodlands (Jenkins and Johnson 1950, Quimby 1951, Getz 1961, Whitaker 1963, 1972; Severinghaus and Beasley 1973, Scott and French 1974, Houtcooper 1982, Lee et al. 1982, Rose and Seegert 1982, Cranford and Maly 1986, Kalko and Handley 1993, Laerm et al. 1996) as well as mixed hardwood (Harvey et al. 1991, 1992) and mixed pine (*Pinus* spp.) and hardwood communities (Kennedy et al. 1982, Pagels et al. 1992). Nests, which are lined with grasses and leaves, are located in shallow underground burrows, clumps of vegetation, brush piles, within and under hollow logs and other woody debris.

REPRODUCTION

Only anecdotal data are available for the region. Quimby (1951) and later Whitaker (1972) summarized literature on *Z. hudsonius* as described below. Hibernation greatly influences the natural history of the meadow jumping mouse. It is considered a profound hibernator. In the northern portion of its range the species becomes dormant in October and emerges in April and May. In the South, it may enter dormancy later and emerge earlier. Breeding begins after emergence from hibernation. Female meadow jumping mice are polyestrous and 2–3 litters may be produced. Several peaks are reported, one in late June, another in mid-late July, and yet another in August. A pregnant female was reported in late September in Georgia (Petrides 1948). Gestation lasts 17–21 days and litter sizes range from 2–8 young

with a reported mean size between 4–5. Individuals probably do not breed their first year.

FOOD HABITS

Upon emergence from hibernation, the meadow jumping mouse feeds heavily on insects, particularly lepidopteran larvae and beetles. As the season advances, the species shifts from insects to grass seeds and fruits of shrubs. They apparently also feed heavily on the fungus *Endogone* (Quimby 1951, Whitaker 1963, 1972; Linzey and Linzey 1973).

ASSOCIATED SPECIES

The meadow jumping mouse is usually segregated ecologically from the woodland jumping mouse, but the two sometimes occur together in shrub, forest-edge stages of intermediate succession, and small oldfields surrounded by forest (Wrigley 1972, Cranford and Maly 1986). The meadow jumping mouse is often found in association with the south-eastern shrew (*Sorex longirostris*), least shrew (*Cryptotis parva*), northern short-tailed shrew (*Blarina brevicauda*), eastern harvest mouse, (*Reithrodontomys humulis*), white-footed mouse (*Peromyscus leucopus*), meadow vole (*Microtus pennsylvanicus*), and woodland vole (*M. pinetorum*).

VULNERABILITY AND THREATS

The meadow jumping mouse is common to abundant in appropriate habitat in the northern portions of its range. Through most of the South, it is regarded as uncommon as reflected in the relatively small number of museum records and its limited occurrence in numerous faunal studies. However, its rarity may be a reflection of several other factors. Some authors, for example Houtcooper (1982) and Taylor and Horn (1983), suggest the meadow jumping mouse is extremely trap shy and difficult to trap using traditional snap traps. However, with use of pitfall traps the meadow jumping mouse was second in abundance only to the very common white-footed mouse in captures of more than 700 small mammals (Pagels et al. 1992). The extremely long hibernation period (7–8 months) during which the species is not susceptible to sampling efforts also impacts collection efforts. Nonetheless, compared to other small mammals, records of the meadow jumping mouse are few in the region and its status and threats are uncertain, particularly in marginal portions of its range.

MANAGEMENT SUGGESTIONS

Despite the perceived uncommonness of the meadow jumping mouse in many areas, there are extensive

areas of appropriate habitat throughout the region. Unless there are management efforts in selected areas that are intended to increase early succession habitats to benefit numerous species, it would be impractical to manage for the meadow jumping mouse alone.

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Coyote

Canis latrans (Say, 1823)

Margaret K. Trani and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Young and Jackson (1951) recognized 19 subspecies of the coyote. However, the integrity of individual subspecies and their taxonomic utility are questionable (Nowak 1978, Bekoff and Gese 2003). The three eastern subspecies currently recognized appear to intergrade imperceptibly, as translocations of the species have confused the subspecific taxonomy in the South (Paradiso 1968, Hill et al. 1987). These include *C. l. frustror*, *C. l. latrans*, and *C. l. thamnias* (Whitaker and Hamilton 1998). The life history of the coyote is reviewed by Young and Jackson (1951), Bekoff (1977), Voight and Berg (1987), Bekoff and Gese (2003), and Gese and Bekoff (2004).

DISTINGUISHING CHARACTERISTICS

The coyote is a doglike carnivore with long legs. Measurements are: total length, 100–135 cm; tail, 27–42 cm; hind foot, 16.5–22.0 cm; ear, 9–12 cm; weight, 5–16 kg. The dorsal pelage is grizzled gray or buff overlaid with black-tipped hairs. The venter is pale cream or gray with a yellowish tint. The face is reddish brown or gray. The muzzle is narrow and the ears are erect. The tail, which is held down between the legs when running, is bushy and black-tipped. Melanistic individuals occur in some localities (Gipson 1976). Size variation is apparent in the South, with larger individuals in less humid areas of the region (Kennedy et al. 1986). The skull of the coyote is elongate with a narrow rostrum, and lacks the temporal ridges that distinguish the fox species. The coyote skull is differentiated from the red wolf (*C. rufus*) based on size; the coyote skull is usually less than 210 mm in total length whereas the red wolf skull is larger. Sealander and Heidt (1990) and Lawrence and Bossert (1967) provide keys to distinguish among the coyote, red wolf, and domestic dog (*C. familiaris*). The dental formula is $I\ 3/3, C\ 1/1, P\ 4/4, M\ 2/3 = 42$ (Figure 1). See keys for details.

CONSERVATION STATUS

The coyote has a global rank of Secure (NatureServe 2007). The species is also considered Secure in those states where it occurs within the region except for Arkansas, Georgia and North Carolina, where it is classified as Apparently Secure. It is unranked in Florida and South Carolina. Many states consider the

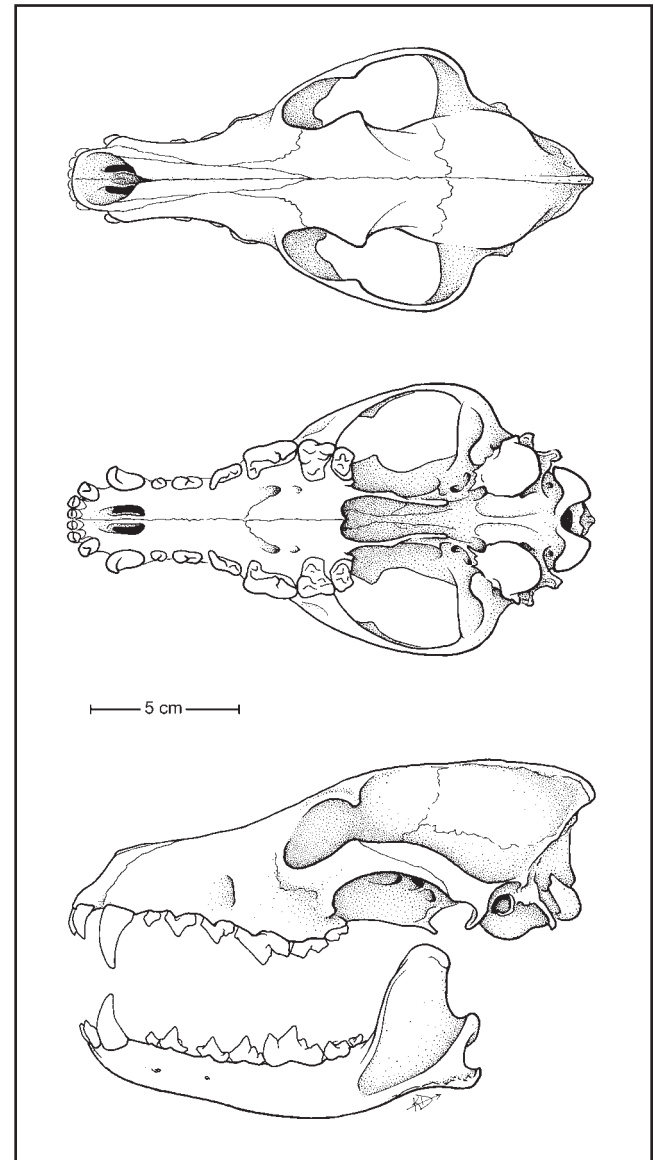


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Canis latrans* from Sterling County, Texas (USNM 211666, male).

coyote a furbearing species with varying regulations on method of take and bag limit. For the majority of southern states, the coyote can be harvested throughout the year on private lands. Although variable by state, restrictions on season dates and harvest method occur on some public lands.

DISTRIBUTION

The original distribution of the coyote included the plains and mountains of western and central North America from eastern Alaska to Indiana and southward to Costa Rica (Young and Jackson 1951, Paradiso 1968). In the last four decades, the coyote has expanded eastward across the United States due to the elimination of the red wolf and other large carnivores from their former ranges (Leopold and Chamberlain 2001). Some range expansion in the South was hastened by translocation efforts by sportsmen groups (Hill et al. 1987).

The species was introduced into scattered locations in Virginia (Handley 1992, Linzey 1998) and now occurs statewide (R. Farrar, Virginia Game and Inland Fisheries, personal communication). Expansion has occurred steadily across North Carolina (DeBow et al. 1998) and South Carolina (Cothran et al. 1991, Baker and Carmichael 1997; J. Kilgo, U. S. Forest Service, personal communication). Currently, the coyote occurs statewide in Georgia (Holzman et al. 1992, Whitaker and Hamilton 1998), Alabama (Sumner et al. 1984, Hoerath and Causey 1991), Florida (Brady and Campbell 1983, Wooding and Hardisky 1990, J. Gore, Florida Fish and Wildlife Conservation Commission, personal communication), Mississippi (Jones and Carter 1989, Chamberlain and Leopold 1999, 2001), Louisiana (Lowery 1974, Morrison et al. 1981, Mullin and Williams 1987, Giordano and Pace 2000), Tennessee (Kennedy 1991, Crawford et al. 1993, Linzey 1995), Kentucky (Barbour and Davis 1974, Hill et al. 1987, Crossett and Elliott 1991), Arkansas (Sealander and Heidt 1990), eastern Texas (Davis and Schmidly 1994), and eastern Oklahoma (Caire et al. 1989).

ABUNDANCE STATUS

Coyote density varies with food abundance, season, and habitat (Gese and Bekoff 2004). Densities between 0.35–0.6/km² were reported in Tennessee (Babb and Kennedy 1989, Stephenson and Kennedy 1993, Crawford et al. 1993), whereas estimates ranged between 0.8–2.3/km² in Texas (Knowlton 1972, Andelt 1985). Flather et al. (1999) presented regional trends in coyote abundance based upon projections from state agencies. For those southern states reporting on the coyote, populations are at or near carrying capacity and are expected to remain stable. Based upon statewide surveys in Mississippi, Lovell et al. (1998) reported that the coyote had experienced a 7.5-fold increase since 1980. State biologists currently report that coyote population levels are continuing to increase in Arkansas (B. Sasse, Arkansas Game and Fish Commission, personal communication), Kentucky

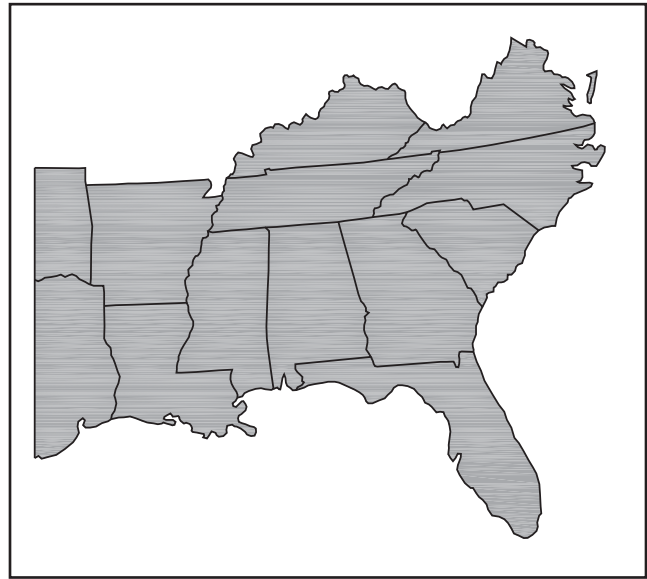


Figure 2. Distribution of *Canis latrans* in the South.

(L. Patton, Kentucky Department of Fish and Wildlife Resources, personal communication), and Texas (J. Young, Texas Parks and Wildlife Department, personal communication).

PRIMARY HABITATS

Habitat use by the coyote is diverse, and reflects its opportunistic feeding habits (Andelt 1977). In the South, the species inhabits almost every habitat present from forests to grasslands, from uplands to coastal areas and bottomland swamps, from rural settings to urban environments (Gese and Bekoff 2004). Male and female habitat use does not appear to differ significantly.

Home range size varies with habitat, food distribution, and social organization (Bekoff and Wells 1986, Gese and Bekoff 2004). Kamler et al. (2005) reported that transient coyotes in Texas use larger home range (84 km²) areas, while resident coyotes occupy smaller territories (10 km²). Annual home range size was reported as 6–19 km² in Georgia (Holzman et al. 1992), 13–33 km² in Arkansas (Gipson and Sealander 1972), and 31–60 km² in western Tennessee (Babb and Kennedy 1989).

REPRODUCTION

The coyote is monoestrus, and pair bonds may last for several years. Breeding season occurs between January–March (Kennelly and Johns 1976, Kennelly 1978). Approximately 60–90% of adult females and up to 70% of yearling females produce litters (Knowlton et al. 1999); gestation lasts 58–65 days (Leopold and Chamberlain 2001). Mean litter size is 6 pups, which is influenced by food availability (Bekoff and Gese 2003) and population density (Knowlton 1972).

during the previous winter. Maternal dens are constructed on brush-covered slopes, steep banks, rock ledges, and under hollow logs. Dens of other animals such as the American badger (*Taxidea taxus*) may also be used (Whitaker and Hamilton 1998). The young are born blind and helpless, and remain in the den until they are weaned at 5–7 weeks (Gier 1968). The pups reach adult weight by 9 months and are capable of breeding before 1 year of age (Gese and Bekoff 2004). Juvenile dispersal usually occurs during autumn and early winter; increased mortality is associated with dispersal as the coyote moves into unfamiliar areas (Knowlton et al. 1999).

FOOD HABITS

The coyote is opportunistic, consuming food in relation to availability (Hoerath and Causey 1991). Common prey include voles (*Microtus* spp.), rats (*Rattus* spp.), pocket gophers (*Geomys* spp.), hispid cotton rat (*Sigmodon hispidus*), and the woodchuck (*Marmota monax*; Hilton 1978, Smith and Kennedy 1983, Wooding et al. 1984, Hoerath and Causey 1991, Chamberlain and Leopold 1999, Hodges 2001). Thornton et al. (2004) reported that common food items eaten in Florida include white-tailed deer (*Odocoileus virginianus*), feral pig (*Sus scrofa*), eastern cottontail (*Sylvilagus floridanus*), marsh rabbit (*S. palustris*), marsh rice rat (*Oryzomys palustris*), eastern gray squirrel (*Sciurus carolinensis*), cotton mouse (*Peromyscus gossypinus*), and saw palmetto (*Serenoa repens*) fruit. Eastern wild turkey (*Meleagris gallopavo*) and northern bobwhite (*Colinus virginianus*) was reported as prey in Tennessee, Mississippi, Georgia, and Florida (Lee and Kennedy 1986, Wagner and Hill 1994, Edwards 1996, Staller et al. 2005); striped skunk (*Mephitis mephitis*) was reported in Kentucky (Crossett and Elliott 1991). Predation on white-tailed deer fawns in some areas can be high (Blanton and Hill 1989). Predation on adult deer may be correlated with winter severity (Gese and Bekoff 2004), and hence may rarely occur in the South except for localized areas in the Central and Southern Appalachian Mountains. Other food items include various reptiles, amphibians, fish, snails, invertebrates, common persimmon (*Diospyros virginiana*) and other fruits, and carrion (Michaelson and Goertz 1977, Litvaitis and Shaw 1980, Stratman and Pelton 1997, Leopold and Chamberlain 2001). The coyote will hunt small mammals alone; when preying on larger prey such as ungulates, cooperation among pack members may occur (Gese et al. 1996). Excess food is cached by burying it in the ground. The coyote in suburban areas is adept at exploiting human-made food resources and readily consumes pet food.

ASSOCIATED SPECIES

Where the coyote and red fox (*Vulpes vulpes*) are sympatric, home ranges do not overlap and strong avoidance occurs (Leopold and Chamberlain 2001). The coyote may suppress fox population growth by aggression and competition for food (Whitaker and Hamilton 1998). Litvaitis and Harrison (1989) observed avoidance behavior between the coyote and bobcat (*Lynx rufus*); Thornton et al. (2004) reported an ecological separation between these two species in Florida facilitated by dietary differences. Gese and Bekoff (2004) observed that the coyote will kill smaller canids such as the common gray fox (*Urocyon cinereoargenteus*), although their coexistence may be mediated by resource partitioning due to the arboreal nature of the gray fox (Neale and Sacks 2001). In red wolf reintroduction sites, the introgression of coyote genes into the wolf population through hybridization is a threat to recovery success (DeBow et al. 1998, Stoskopf et al. 2005). Fertile hybrids are also known between the domestic dog and coyote (Gese and Bekoff 2004).

VULNERABILITY AND THREATS

Disease can be a substantial mortality factor, especially among pups. Serological analyses show that the species can be infected by canine parvovirus enteritis, distemper, hepatitis, rabies, and brucellosis (Bekoff and Gese 2003, Krebs et al. 2003). Sarcoptic mange may be locally common in some populations, resulting in reduced fecundity and increased mortality rates (Pence and Windberg 1994). The coyote hosts numerous ectoparasites including ticks (*Dermacentor variabilis*, *Amblyomma maculatum*), and biting lice (*Trichodectes canis*; Bekoff 1977). In addition, the coyote is susceptible to a variety of internal parasites including heartworm (*Dirofilaria immitis*), hookworm (*Ancylostoma caninum*), tapeworm (*Taenia pisiformis*), and various nematodes including *Physaloptera rara* (Foster et al. 2003).

Most studies indicate that human activity contributes to a high proportion of the deaths of coyotes (Bekoff and Gese 2003, Gese and Bekoff 2004). Human exploitation can be substantial in some coyote populations (Knowlton et al. 1999), with protection of livestock and game species constituting the motives for removal. The coyote does prey on domestic sheep, cattle, and poultry (Armstrong 1991, Philip and Armstrong 1994).

MANAGEMENT SUGGESTIONS

Conservation measures have not been needed to enhance habitat quality for the coyote or to maintain

viable populations (Gese and Bekoff 2004). In areas of overpopulation, management centers on damage prevention and population control. Knowlton et al. (1999) reviewed many of the techniques for reducing coyote depredation including protective fencing, frightening devices, and use of repellents. Coyote control efforts promote sound husbandry methods that prevent livestock losses to predation. Research needs include development of non-lethal depredation techniques and methods of fertility control, and resolution of potential conflicts at the urban interface.

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Red Wolf

Canis rufus (Audubon and Bachman, 1851)

Margaret K. Trani and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Because many hybrids with the gray wolf (*Canis lupus*) and the coyote (*C. latrans*) were known, the validity of the red wolf as a distinct species was questioned by McCarley (1962), Lawrence and Bossert (1967), and Clutton-Brock et al. (1976). Paradiso and Nowak (1972a) suggested that hybridization occurred as a result of habitat disruption caused by human development. Nowak (1970), Atkins and Dillon (1971), Paradiso and Nowak (1973), and Ferrell et al. (1980) provided evidence to show validity of the species. However, Hall (1981) and Wayne and Jenks (1991) noted that specimens examined had genotypic characteristics related to either gray wolves or coyotes. Wilson and others (2000) suggested that the red and gray wolf share a common lineage with the coyote in North America and are best considered conspecifics. Nowak (2002) conducted morphometric analyses of red wolf skulls and presented the case for retaining the red wolf as a separate species. Baker and others (2003) recognized the red wolf as a separate species, whereas Wilson and Reeder (2005) listed the red wolf as a subspecies of the gray wolf. Although the taxonomic status remains unclear, there is evidence to support *C. rufus* as a unique species (Kelly et al. 2004). Currently, the U. S. Fish and Wildlife Service recognizes the red wolf as a distinct species under the Endangered Species Act, and afford it full protection under federal law.

Three subspecies were recognized by Goldman (1937, 1944) and Hall (1981): *C. r. gregoryi*, *C. r. floridanus*, and *C. r. rufus*. The subspecies used for the reintroduction effort into North Carolina is *C. r. gregoryi* (Kelly et al. 2004). The life history of the red wolf is reviewed by Paradiso and Nowak (1972b, 1982) and Carbyn (1987).

DISTINGUISHING CHARACTERISTICS

The red wolf is intermediate in size between the gray wolf and coyote. Measurements are: total length, 135–165 cm; tail, 34.5–43.0 cm; hind foot, 21–25 cm; ear, 12–13 cm; weight, 16–41 kg. The dorsal pelage is variable in coloration and generally is reddish-buff interspersed with gray and black. Melanism is common (Whitaker and Hamilton 1998). The tail is bushy and tipped with black; the belly, nose, and throat are whitish-buff. The muzzle, ears, nape, and outer

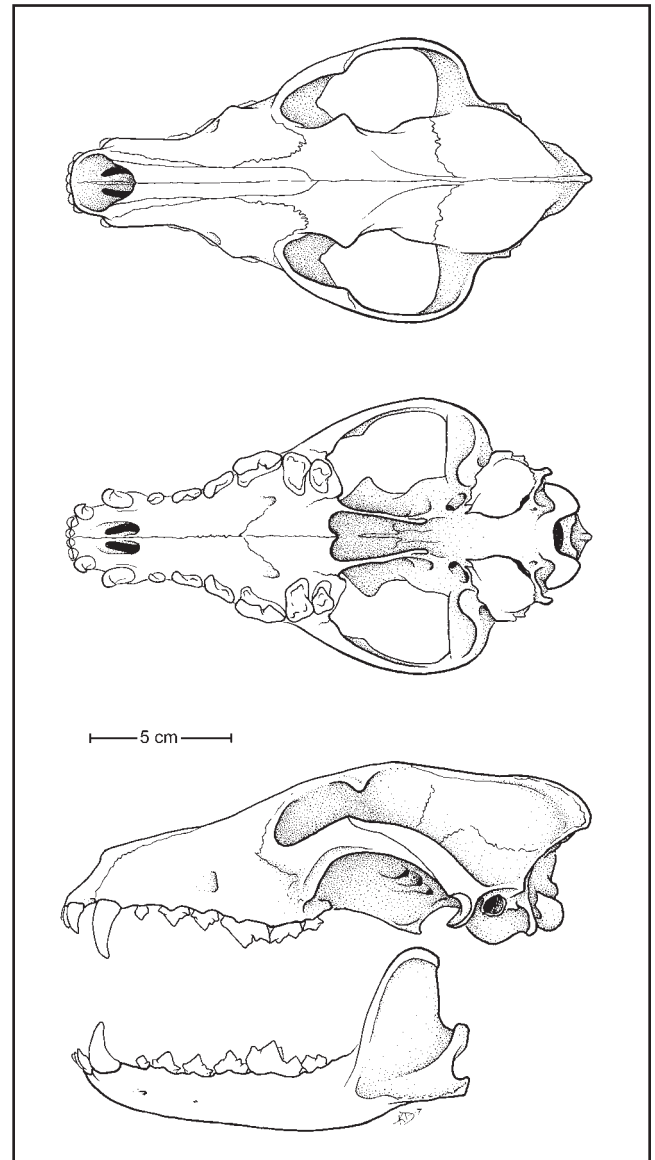


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Canis rufus* from Newton County, Arkansas (USNM 235599, female).

surfaces of the legs are tawny to cinnamon-buff. The skull is elongate with a narrow rostrum, and can be distinguished from a coyote skull based on size. The coyote skull usually is less than 210 mm long and 110 mm wide, whereas the wolf skull often exceeds those dimensions. The dental formula is I 3/3, C 1/1, P 4/4, M 2/3 = 42 (Figure 1). See keys for details.

CONSERVATION STATUS

The red wolf has a global rank of Critically Imperiled. (NatureServe 2007). It is considered Critically Imperiled in North Carolina and South Carolina. It is Presumed Extirpated in the remaining states in the South; however, an island propagation site occurs on an barrier island in Florida. The U. S. Fish and Wildlife Service (U. S. Department of the Interior 2007) lists the red wolf as Endangered, with a recovery plan approved for the species (USFWS 1989). The reintroduced population in North Carolina is considered Experimental Non-essential, a designation that permits the U. S. Fish and Wildlife Service to manage the population and promote recovery in a manner respectful of the needs and concerns of local citizens (Parker and Phillips 1991).

DISTRIBUTION

The red wolf historically roamed an extensive range including southern Illinois, Indiana and Pennsylvania south to Florida and west to central and southern Texas (Harper 1942, Young and Goldman 1944, Nowak 1979, Hall 1981, Choate et al. 1994, Wilson et al. 2000). From the early 1800s through the middle 1900s, indiscriminate predator control, extensive land clearing, expanding human populations, and declining prey populations (Crawford et al. 2001) reduced the range of the red wolf to the forested bottoms and coastal marshes of southeastern Texas and southwestern Louisiana (Paradiso 1965, 1968; Pimlott and Joslin 1968, Nowak 1970). The resulting changes to the landscape favored the eastern expansion of the coyote into historic red wolf territory, fostering a breakdown of the red wolf social structure. As a result, hybridization occurred as the wolf found it increasingly difficult to locate conspecifics (Nowak 1979, Crawford et al. 2001).

By 1976, the remaining red wolves in coastal Louisiana had been removed from the wild with the goal of establishing a captive breeding program and eventually restoring the species to portions of their historic range (Carley 1979). Point Defiance Zoo and Aquarium in Tacoma, Washington was established as the red wolf captive breeding center (Crawford et al. 2001). The program was successful and the first mainland repatriation of red wolves occurred in 1987 on Alligator River National Wildlife Refuge and adjacent U. S. Department of Defense land in northeastern North Carolina (Parker 1987, Phillips and Parker 1988, Parker and Phillips 1991). The 5-county area covering 688,259 hectares of public and private land includes Dare, Hyde, Tyrrell, Beaufort, and Washington counties. Island propagation sites were subsequently established on Bulls Island, South

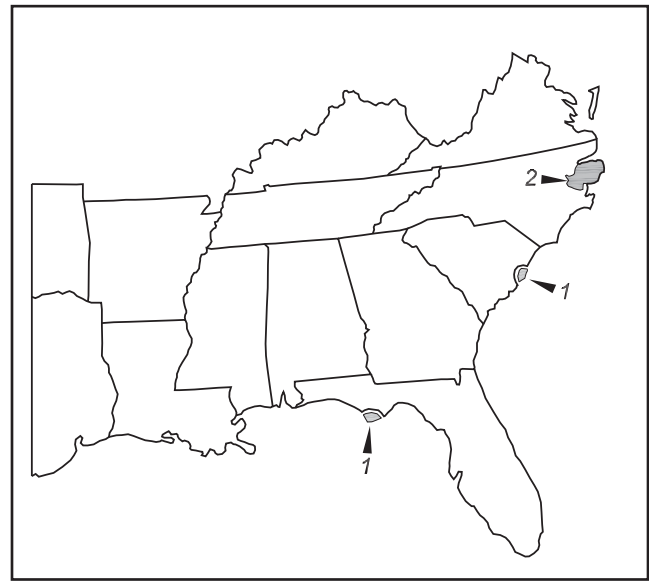


Figure 2. Distribution of *Canis rufus* in the South: (1) Island propagation sites; (2) Reintroduction counties.

Carolina (1987); Horn Island, Mississippi (1989); and St. Vincent Island, Florida (1990) to supply wild-reared wolves for the mainland restoration effort (van Manen et al. 2000). Horn Island was eventually dropped as a propagation site (B. Fazio, U. S. Fish and Wildlife Service, personal communication). A second mainland repatriation effort initiated in 1991 within the Great Smoky Mountains National Park, Tennessee was terminated seven years later due to the high rate of pup mortality from disease, predation from the coyote, and malnutrition (Lucash et al. 1998).

ABUNDANCE STATUS

The current free ranging populations are a result of these restoration efforts (Figure 2). There are over 100 red wolves in the wild population in northeastern North Carolina, comprising 18–22 packs (B. Fazio, U. S. Fish and Wildlife Service, personal communication). Each wolf pack typically includes a dominant breeding pair and offspring from previous years. Bulls Island, Cape Romain National Wildlife Refuge in South Carolina and St. Vincent Island National Wildlife Refuge, Florida remain as island propagation sites. Each island retains one family of wolves; the size ranges from 2–6 animals depending on field management decisions. Young born on the islands are either fostered as pups or translocated at 18 months of age into the wild population in North Carolina. Approximately 160–180 wolves are in captivity at 38 facilities throughout the United States and Canada (B. Fazio, U. S. Fish and Wildlife Service, personal communication) for reproduction and

conservation genetics research. The breeding program maintains genetic diversity and provides wolves for release into the wild population.

PRIMARY HABITATS

Given the historical distribution of the red wolf, it is probable that the species used a variety of habitats. The former population in Texas and Louisiana inhabited bayous, fallow fields, coastal prairies, and marshes (Carley 1979, Kelly et al. 2004). There is evidence that the species inhabited densely vegetated habitats such as the bottomland hardwood forests and swamps in the South (Paradiso and Nowak 1972a). Reintroduced wolves in North Carolina have made extensive use of habitats ranging from agricultural lands, pocosins, and loblolly pine (*Pinus taeda*) and pond pine (*P. serotina*) forests. The wolf is a habitat generalist, occurring where sufficient prey occur along with their basic needs for shelter and breeding (B. Fazio, U. S. Fish and Wildlife Service, personal communication). The red wolf is territorial; home range size varies from 46–226 km² depending upon habitat and prey density (Phillips et al. 2003). Red wolves require large tracts of land relatively free from human activity, heavily used highways, and livestock.

REPRODUCTION

Age at sexual maturity for the red wolf has been reported at 3 years (Paradiso and Nowak 1972b) and as young as 22 months (Phillips et al. 2003). Breeding occurs in February and March with gestation lasting 61–63 days; average litter size is 1–8 pups (Crawford et al. 2001). Peak whelping dates occur from mid-April to mid-May (Paradiso and Nowak 1972b). Pregnant females may establish several dens. Some dens are shallow surface depressions located in dense vegetation where the water table is high, while other dens are deep burrows in wind rows between agricultural fields or in canal banks; dens have also been found in hollowed out bases of large trees (Phillips et al. 2003). One litter per year is produced by the dominant pair (Kelly et al. 2004). Dispersal typically occurs before individuals reach two years of age (Phillips et al. 2003).

FOOD HABITS

The red wolf is an opportunistic predator and will take prey items that are available (Crawford et al. 2001). Common prey species in Louisiana and Texas included nutria (*Myocastor coypus*), eastern cottontail (*Sylvilagus floridanus*), swamp rabbit (*S. aquaticus*), marsh rice rat (*Oryzomys palustris*), hispid cotton rat (*Sigmodon hispidus*), common muskrat (*Ondatra zibethicus*), small domestic animals, and carrion (Stutzenbaker 1968, Russell and Shaw 1971, Riley and

McBride 1972). Red wolves in South Carolina consume fox squirrel (*Sciurus niger*), American coot (*Fulica americana*), and other birds (Carley 1979). In North Carolina, white-tailed deer (*Odocoileus virginianus*), northern raccoon (*Procyon lotor*), and marsh rabbit (*S. palustris*) comprise 86% of the diet (Phillips et al. 2003); frogs and turtles are also taken (Venters 1989). The red wolf is primarily a nocturnal hunter with crepuscular peaks of activity (Kelly et al. 2004).

ASSOCIATED SPECIES

In addition to its primary prey, the red wolf occurs in habitats occupied by a variety of species. These include the Virginia opossum (*Didelphis virginiana*), American beaver (*Castor canadensis*), eastern gray squirrel (*Sciurus carolinensis*), common gray fox (*Urocyon cinereoargenteus*), northern river otter (*Lontra canadensis*), American mink (*Mustela vison*), and bobcat (*Lynx rufus*). It is preyed upon by the American alligator (*Alligator mississippiensis*) on St. Vincent Island, Florida (Whitaker and Hamilton 1998). The red wolf has been observed killing young American black bears (*Ursus americanus*; Kelly et al. 2004). Research is underway to determine the degree of competition for prey and habitat between the red wolf and coyote.

VULNERABILITY AND THREATS

The introgression of coyote genes into the red wolf population is the principal threat to recovery (Stoskopf et al. 2005). Although hybridization with coyotes was a factor in the red wolf's initial demise in the wild, it was not detected as a problem in the North Carolina population until the early 1990s (Phillips et al. 1995). Vehicle mortality accounts for 18% of known red wolf deaths (Kelly et al. 2004). Human-induced mortality (e.g., gunshot, traps, and poison) in the reintroduced population accounts for approximately 17% of red wolf deaths (Kelly et al. 2004). The red wolf is susceptible to viral diseases including rabies, canine distemper, parvovirus, and hepatitis (Crawford et al. 2001). Commonly occurring helminths include canine heartworm (*Dirofilaria immitis*) and hookworm (*Ancylostoma caninum*); neither appears to be a significant source of mortality (Phillips and Scheck 1991). Mortality related to demodectic mange and infestation from the American dog tick (*Dermacentor variabilis*), lone star tick (*Amblyomma americanum*), and black-legged tick (*Ixodes scapularis*) have also occurred (Kelly et al. 2004).

MANAGEMENT SUGGESTIONS

Continued success has established northeastern North Carolina as a permanent red wolf recovery site. (Crawford et al. 2001). The outlook continues to be

positive; coyote genetic intrusion into the red wolf population has been largely controlled (Stoskopf et al. 2005). Improved genomic testing protocols are available for assessing alleles (Miller et al. 2003). There is no evidence that inbreeding depression has been a major obstacle to the success of the recovery effort (Kalinowski et al. 1999). The primary recovery focus is protecting and promoting the growth of a self-sustaining, non-hybridizing population in the wild and sustaining an active captive component (Stoskopf et al. 2005).

Biological factors such as prey abundance, habitat availability and socioeconomic factors such as agricultural practices and land ownership patterns are important considerations for future reintroduction sites. Those limiting the restoration of the red wolf include conflicts with humans, political opposition, potential livestock depredation, increasing coyote populations, and uncertain taxonomic status (Crawford et al. 2001). Although the red wolf can flourish in a wide variety of habitats, the support of the public is a requisite for its successful recovery.

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Common Gray Fox

Urocyon cinereoargenteus (Schreber, 1775)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Although the common gray fox (*Urocyon cinereoargenteus*) once was placed in the genus *Vulpes* by Clutton-Brock et al. (1976) and the genus *Canis* by Van Gelder (1978), most authorities agree upon the present generic assignment (Jones et al. 1992, Wozencraft 1993). There are 21 recognized subspecies of *U. cinereoargenteus*, three occur in the South: *U. c. cinereoargenteus*, *U. c. floridanus*, and *U. c. ocythous* (Hall 1981). The life history of the gray fox was reviewed by Trapp and Hallberg (1975), Fritzell and Haroldson (1982), Samuel and Nelson (1982), and Fritzell (1987).

DISTINGUISHING CHARACTERISTICS

The common gray fox is a medium-sized, dog-like carnivore. Measurements are: total length, 80.0–102.5 cm; tail, 28.0–37.5 cm; hind foot, 12.5–15.0 cm; ear, 6–8 cm; weight, 2.5–7.0 kg. The dorsal pelage is red-dish-gray with salt-and-pepper overtones and buff-colored underfur. A distinct zone of black-tipped hairs runs down the back and upper surface of the tail. The tail is bushy with a black tip. The sides of the muzzle are marked with black and the chin and throat are white. The sides of the neck, the backs of the ears, the upper chest, the legs, and the feet are rusty-yellow. The venter from the lower chest to the anus is buff-colored. The skull of the common gray fox can be distinguished by the prominent cranial ridges. Two distinct ridges begin at the postorbital processes, converge at the junction of the temporal and parietal bones, diverge at mid-parietal, then join at the occipital forming a lyre-shaped pattern. The skull of the red fox (*V. vulpes*) also possesses ridges, but the ridges converge toward the rear of the skull forming a narrow V-shaped pattern. The dental formula of the fox is $I\ 3/3, C\ 1/1, P\ 4/4, M\ 2/3 = 42$ (Figure 1). See keys for details.

CONSERVATION STATUS

The common gray fox has a global rank of Secure (NatureServe 2007). The species is also considered Secure in those states where it occurs within the region except for Arkansas, Kentucky, Louisiana and Oklahoma, where it is Apparently Secure. It is unranked in Florida and South Carolina.

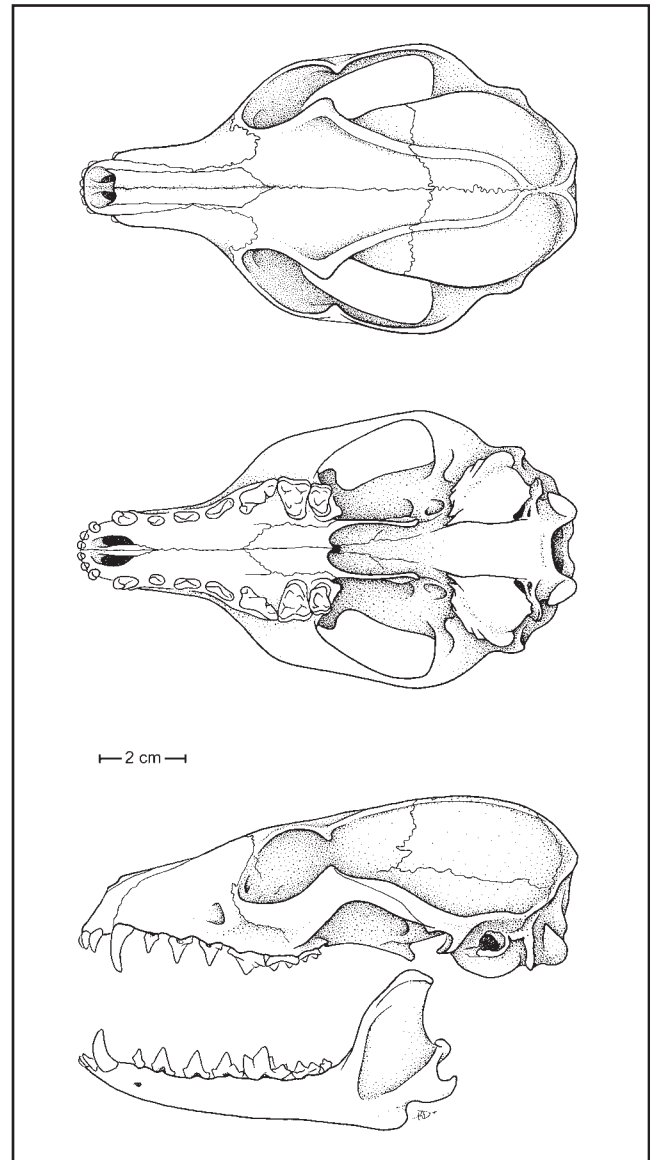


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Urocyon cinereoargenteus* from Gavito Pass, California (USNM 43802, female).

DISTRIBUTION

The gray fox ranges from southern Canada to northern Venezuela and Columbia, but is absent from parts of the Great Plains and the higher elevations of the Rocky Mountains (Hall 1981, Choate et al. 1994). The distribution of the species in the South is depicted in

Figure 2. The species is found statewide in Virginia (Nelson 1933, Bailey 1946, Handley and Patton 1947, Carey et al. 1978, Handley 1979, 1992; Linzey 1998), North Carolina (Odom 1949, Johnston 1967, Linzey and Linzey 1971, Lee et al. 1982, Clark et al. 1985, Webster et al. 1985, Webster 1988), South Carolina (Penny 1950, Wood and Odom 1964, Golley 1966, Sanders 1978, Schacher and Pelton 1979, Jeselnik 1980, Sawyer and Fendley 1990, 1994; Cothran et al. 1991), Georgia (Harper 1927, Wood 1958, Wood et al. 1958, Golley 1962, Laerm et al. 1980, 1981), Florida (Hamilton 1941, Moore 1946, 1949; Schwartz 1952, Ivey 1959, McKeever 1959, Pearson 1960, Lord 1961, Layne 1974, Progulske 1982, Wassmer 1984, Sunquist 1989), Alabama (Howell 1921, Sullivan 1956, Holliman 1963, Linzey 1970, Sumner and Hill 1980, Nicholson and Hill 1984, Wooding 1984, Nicholson et al. 1985), and Mississippi (Wolfe 1971, Kennedy et al. 1974, Wooding 1984, Jones and Carter 1989, Tucker et al. 1993, Chamberlain and Leopold 2000, 2002). The fox is found outside of the coastal marshes of Louisiana (Lowery 1974, Foote 1984, Mullin and Williams 1987). It is found throughout eastern Texas (Wood 1952, McCarley 1959, Schmidly 1983, 1984; Davis and Schmidly 1994) and eastern Oklahoma (Caire et al. 1989). It is distributed statewide in Arkansas (Heidt et al. 1984, Tumblison and McDaniel 1984, Peck et al. 1985, Steward et al. 1989, Sealander and Heidt 1990), Tennessee (Howell and Conaway 1952, Goodpaster and Hoffmeister 1952, Conaway and Howell 1953, Linzey and Linzey 1971, Yoho and Henry 1972, Smith et al. 1974, Greenberg and Pelton 1991, 1994; Kennedy 1991, Linzey 1995), and Kentucky (Barbour and Davis 1974, Fassler 1974). The common gray fox has been stocked in some areas (Hall 1983).

ABUNDANCE STATUS

Estimates of population density vary from 1.2–2.1/km², and are influenced by location and season (Gier 1948, Lord 1961, Connor et al. 1983). Richmond (1952) found that the population density of gray foxes in Pennsylvania increased during years when January–March were wetter and warmer than normal, and decreased when the same months were colder and drier than average. Lovell et al. (1998) found that common gray fox populations in Mississippi remained stable during 1980–1995. Reported home ranges of the common gray fox are slightly less than 4 km² and do not vary with sex, age, or season (Greenberg and Pelton 1994).

PRIMARY HABITATS

In parts of the South, the gray fox usually is associated with deciduous forest communities (Fritzell 1987) and pine (*Pinus* spp.) forests that are 30 years

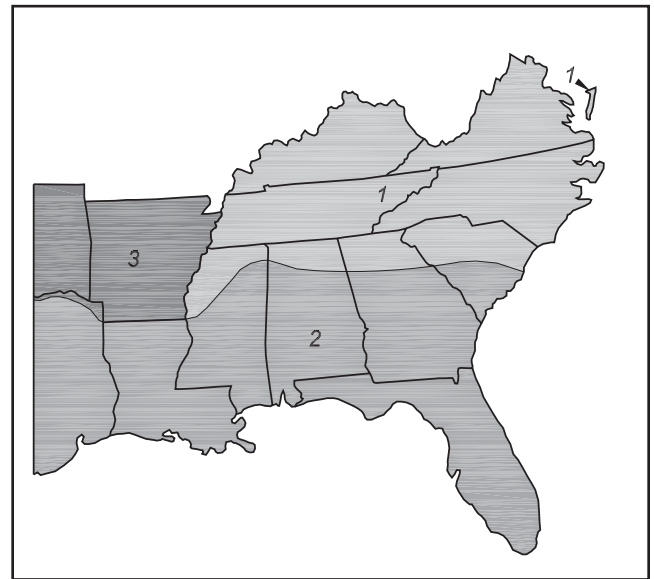


Figure 2. Distribution of *Urocyon cinereoargenteus* in the South: (1) *U. c. cinereoargenteus*; (2) *U. c. floridanus*; (3) *U. c. ocythous*.

old or older (Chamberlain and Leopold 2000). Woodland-farmland edges provide the best habitat in Virginia (Carey et al. 1978), Georgia (Wood et al. 1958, Jeselnik 1980), and eastern Texas (Wood 1952). Patterns of habitat use may change seasonally (Foote 1984, Sawyer and Fendley 1990, 1994). Follman (1973 in Fritzell 1987) reported that the fox preferred oldfields during summer and fall, but shifted most activities to woodlands during winter and spring. The common gray fox consistently selects habitats that have an abundance of small mammals (Chamberlain and Leopold 2000). Heavy use of habitats containing fruit-bearing trees and shrubs also has been documented (Yearsley and Samuel 1980). During the daylight hours, the fox rests in wooded habitats with dense, brushy cover (Yearsley and Samuel 1980, Haroldson and Fritzell 1984). The species may climb trees to escape predators and hunt birds (Trapp and Hallberg 1975). Dens may be used at any time of the year, but females consistently use dens when whelping. In Alabama, all dens were located underground (Nicholson et al. 1985) but many other den locations have been reported. Hollow logs and trees, rock outcrops and abandoned dens of other animals have been used as dens (Sullivan 1956, Jackson 1961, Schmeltz and Whitaker 1977). Grinnell et al. (1937) reported a whelping den in a 7.6 m high hollow tree.

REPRODUCTION

The breeding season is from January–March with the peak in February (Sullivan 1956, Wood 1958). The female fox is monestrous. The length of the gestation

period is estimated to be from 53 days (Wood 1958) to 63 days (Grinnell et al. 1937, Asdell 1964). In captivity, the fox has a gestation period of 59 days (Fritzell 1987). The mean litter size is 4; however, 1–7 young are reported (Sheldon 1949, Sullivan 1956, Layne 1958). The behavioral development of the young is poorly known. Most females reach sexual maturity at 10 months old (Wood 1958), but some yearlings may reach sexual maturity later in the year (Layne 1958). Although Seton (1929) suggested that gray foxes can live for 14–15 years in the wild, Lord (1961) found that few live beyond 5 years. In a sample of 435 common gray foxes collected in Georgia, 89.4% were less than two years old (Wood 1958).

FOOD HABITS

The diet of the gray fox varies seasonally. Rabbits (*Sylvilagus* spp.), voles (*Microtus* spp.), mice (*Peromyscus* spp.), eastern woodrats (*Neotoma floridana*), and hispid cotton rats (*Sigmodon hispidus*) are common prey during the winter months (Nelson 1933, Wood 1954, Wood et al. 1958, Yoho and Henry 1972, Nicholson 1982, Greenberg and Pelton 1991). Fruits and insects (primarily orthopterans) are important components of the diet during summer and fall (Llewellyn and Uhler 1952, Wood et al. 1958, Pils and Klimstra 1975). Other foods consumed include squirrel (*Sciurus* spp.), Virginia opossum (*Didelphis virginiana*), and white-tailed deer (*Odocoileus virginianus*) carrion.

ASSOCIATED SPECIES

The gray fox is often sympatric with the red fox. The gray fox often uses the dens of striped skunk (*Mephitis mephitis*), nine-banded armadillo (*Dasypus novemcinctus*), and woodchuck (*Marmota monax*). Because the species is so widespread, the common gray fox is found in association with numerous species in the region.

VULNERABILITY AND THREATS

Trapping, hunting, and vehicular traffic are the major sources of mortality in the common gray fox. Outbreaks of canine distemper may reduce local populations and it sometimes takes up to six years for the population to recover. The gray fox may carry rabies (Davidson and Nettles 1997).

MANAGEMENT SUGGESTIONS

Management centers on the provision of a mix of early seral conditions, mature forest, and dense cover. Allen (1987) suggests activities that promote understory vegetation diversity and encourage establishment of fruit-producing shrubs such as

blackberry (*Rubus* spp.) and blueberry (*Vaccinium* spp.). Prescribed burning may be used to maintain oldfields and forest openings in a desirable condition that enhance productivity of rodent prey.

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Red Fox

Vulpes vulpes (Linnaeus, 1758)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Whether or not the red fox (*Vulpes vulpes*) is native to North America is a matter of debate. Churcher (1959) argues that the red fox was native to North America north of latitude 40°, but was rare or absent in the forests of the South. Others suggest that the red fox was introduced into the southeastern United States from European stock around 1750 (Godin 1977, Kamler and Ballard 2002). The North American red fox originally was described as a separate species, *V. fulva*, but several authors demonstrated that the European and North American red foxes are a single species (Rausch 1953, Vogt and Arakaki 1971). Most authorities (Hall 1981, Jones et al. 1992, Wozencraft 1993) have accepted this arrangement but some widely used references (Burt and Grossenheider 1980) retain *V. fulva*. Ten subspecies are recognized in North America; one subspecies (*V. v. fulva*) occurs in the South. The life history of the red fox is reviewed by Ables (1975), Samuel and Nelson (1982), Voigt (1987), and Lariviere and Pasitschniak-Arts (1996).

DISTINGUISHING CHARACTERISTICS

The red fox is a medium-sized, dog-like carnivore. Measurements are: total length, 90–114 cm; tail, 30–43 cm; hind foot, 13.5–17.5 cm; ear 7.5–9.5 cm; weight, 2.7–7.8 kg. The dorsal pelage is reddish yellow and the legs, feet, and the backs of the ears are black. The long, bushy tail is tipped with white. The cheeks, throat, and belly are whitish or grayish. The upper surface of the skull possesses two distinct ridges that begin at the postorbital processes and converge toward the rear where they join at the occipital. The ridges do not converge, diverge, and converge again as in the common gray fox (*Urocyon cinereoargenteus*). The dental formula is $I\ 3/3, C\ 1/1, P\ 4/4, M\ 2/3 = 42$ (Figure 1). See keys for details.

CONSERVATION STATUS

The red fox has a global rank of Secure (NatureServe 2007). It is also considered Secure in Georgia, Kentucky, North Carolina, and Virginia. It is Apparently Secure in Alabama, Arkansas, Louisiana, Mississippi, and Texas. It is classified as Vulnerable in Oklahoma. It is unranked in Florida and South Carolina. Tennessee has assigned a conservation status rank of Not

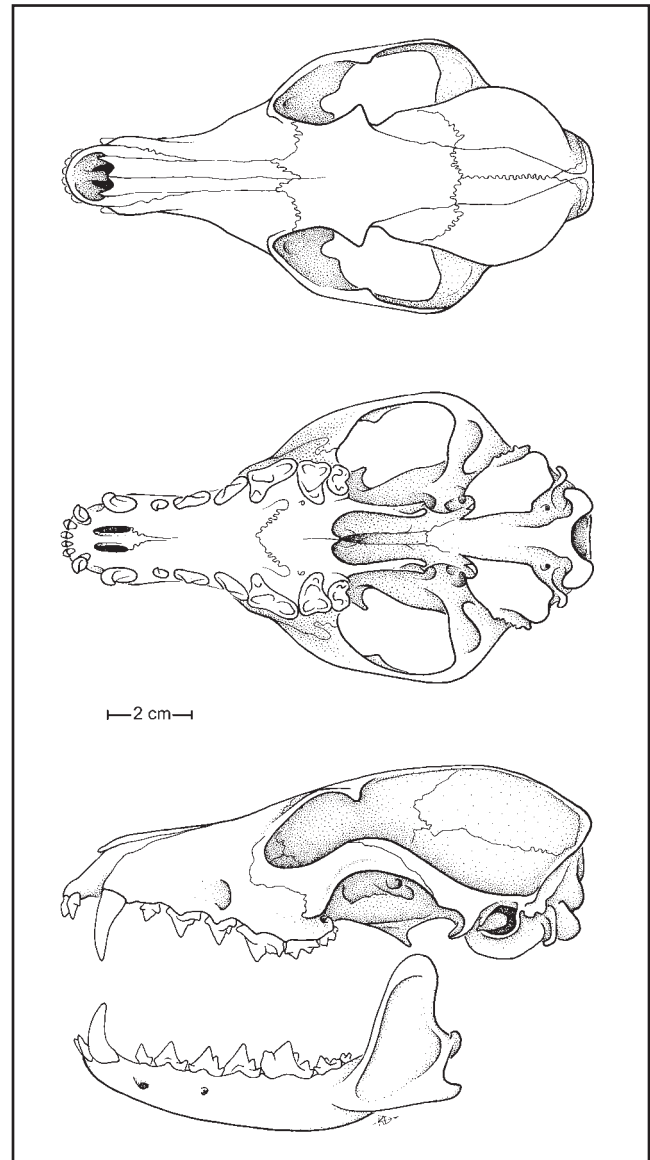


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Vulpes vulpes* from Howard County, Maryland (USNM 271206, male).

Applicable; the species is not considered a suitable target for conservation activities.

DISTRIBUTION

The distribution of the red fox in North America includes most of Canada and the United States, but the species does not occur in parts of the Rocky

Mountains and it is absent from much of the arid Southwest (Hall 1981, Lariviere and Pasitschniak-Arts 1996). The species occurs throughout Virginia, but no specimens are known from the southeastern corner of the state (Bailey 1946, Handley and Patton 1947, Jackson et al. 1976, Dueser et al. 1979, Handley 1979, 1992; Webster et al. 1985, Linzey 1998). The red fox occurs throughout North Carolina (Conaway and Howell 1953, Johnston 1967, Linzey and Linzey 1971, Lee et al. 1982, Clark et al. 1985, Webster et al. 1985, Linzey 1995), South Carolina (Penny 1950, Wood and Odom 1964, Golley 1966, Sanders 1978, Cothran et al. 1991), Georgia (Wood 1958, Pearson 1960, Golley 1962, Laerm et al. 1981), Florida (Wood 1958, McKeever 1959, Pearson 1960, Layne 1974, Hall 1981, Sunquist 1989), Alabama (Howell 1921, Pearson 1960, Holliman 1963, Linzey 1970), and Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989). The species may be rare or absent in the coastal marshes of southern Louisiana (Lowery 1974, Mullin and Williams 1987) and southeastern Texas (Schmidly 1983, 1984; Davis and Schmidly 1994), but occurs throughout the northern portions of Louisiana and eastern Texas (Figure 2). *Vulpes vulpes* is distributed in most of eastern Oklahoma (Caire et al. 1989) and throughout Arkansas (Steward et al. 1989, Sealander and Heidt 1990), Tennessee (Howell and Conaway 1952, Goodpaster and Hoffmeister 1952, Conaway and Howell 1953, Linzey and Linzey 1971, Smith et al. 1974, Greenberg and Pelton 1991, Kennedy 1991), and Kentucky (Barbour and Davis 1974, Fassler 1974, Cobb and Frederick 1985, Crossett and Elliott 1991). Red fox populations in most southern states were periodically augmented (Lowery 1974, Lee et al. 1982, Schmidly 1983, Zumbaugh and Choate 1985, Webster et al. 1985).

ABUNDANCE STATUS

Because the species is secretive and elusive, it is difficult to determine the density of red fox populations (Voigt 1987). Where the food supply is abundant in urban areas of Great Britain, red fox densities reach 30/km² (Harris 1977, Harris and Rayner 1986). In southern Ontario, densities of about 1/km² have been reported (Voigt 1987). Population densities of the red fox in the southeastern United States are not known. However, between 21,000 and 36,000 red foxes were trapped annually in Texas from 1973–1976 (Samuel and Nelson 1982).

PRIMARY HABITATS

The red fox inhabits many habitats (Voigt 1987). In the South, the species occurs in pine-oak (*Pinus-Quercus* spp.) forests that are juxtaposed with agricultural fields or pastures (Golley 1962, 1966; Lowery

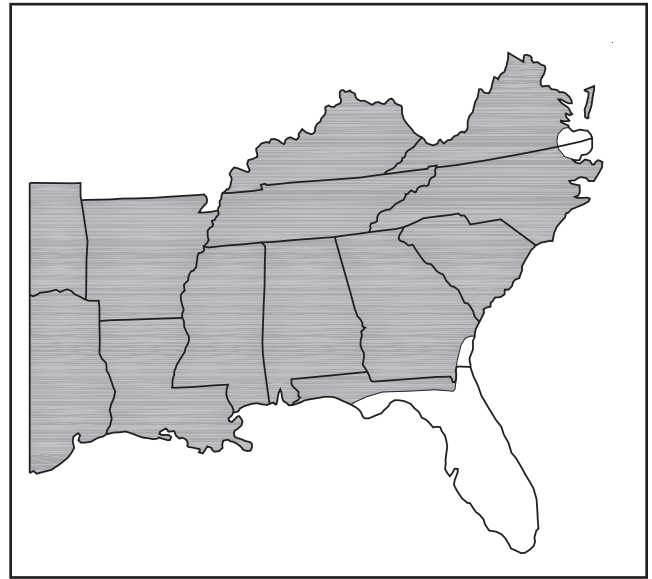


Figure 2. Distribution of *Vulpes vulpes* in the South.

1974, Schmidly 1983, Sealander and Heidt 1990). In eastern Oklahoma, the red fox is abundant in oak-hickory (*Quercus-Carya*) forests (Caire et al. 1989). The species also inhabits suburban areas (Harris 1977). During spring and summer, activity centers on the whelping den. Red foxes dig their own dens or use abandoned dens of other mammals such as the woodchuck (*Marmota monax*; Sheldon 1950, Samuel and Nelson 1982). Some dens may be used for several generations (Stanley 1953).

REPRODUCTION

Female red foxes are monestrous; breeding occurs from December–March (Rowlands and Parkes 1935). The gestation period is 50–53 days (Asdell 1964) and the mean litter size is 5 (Storm et al. 1976, Voigt 1987). Pups remain at the den for at least a month, and may be moved from one den to another several times before they are 6 weeks old (Scott 1943, Sheldon 1949, Storm et al. 1976). The pups leave the den for short forays at 10 weeks of age, and disperse September–October (Storm et al. 1976). Female red foxes are capable of breeding at 10 months old, but some do not mate during the first estrous (Wood 1958). Red foxes can live about 5 years in the wild (Wood 1958, Storm et al. 1976).

FOOD HABITS

The red fox is an opportunistic predator and a scavenger. Predominant prey include voles (*Microtus* spp.), mice (*Peromyscus* spp.), rabbits (*Sylvilagus* spp.), hispid cotton rats (*Sigmodon hispidus*), and woodchucks. The red fox may be an important nest predator of ducks and other birds (Sargent 1978, Crossett and Elliott

1991). It also consumes fruits, berries, insects, earthworms, and carrion (Scott and Klimstra 1955, Korschgen 1959, Schofield 1960, Hockman and Chapman 1983, Voigt 1987, Crossett and Elliott 1991, Greenberg and Pelton 1991).

ASSOCIATED SPECIES

The red fox interacts with several mammal species of the region. It sometimes occupies the same habitats as the common gray fox and coyote (*Canis latrans*).

VULNERABILITY AND THREATS

Red foxes have high annual mortality rates (Voigt 1987). Shooting, trapping, and vehicular accidents are contributing factors (Storm et al. 1976). Local populations also may be periodically decimated by outbreaks of rabies and other diseases (Barker et al. 1983, Addison et al. 1987, Little et al. 1998). In the presence of coyotes, red fox populations may decline (Johnson and Sargeant 1977, Sargeant and Allen 1989).

MANAGEMENT SUGGESTIONS

The red fox benefits from prescribed burning and timber harvest activities that enhance edge habitat, vegetation diversity, and interspersed of seral stages. Allen (1987) recommends maintaining travel corridors and the establishment of fruit producing trees and shrubs. Population control is implemented by regulated harvest seasons. Damage prevention and control methods include fencing, toxicants, den hunting, and trapping.

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Mountain Lion

Puma concolor (Linnaeus, 1771)

Margaret K. Trani and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The taxonomic classification of the *Felis concolor* group was revised and described by Nelson and Goldman (1929) and Young and Goldman (1946). Nowell and Jackson (1996) reviewed the taxonomy and placed the mountain lion into the genus *Puma* (Clark 2001, Baker et al. 2003). The mountain lion includes 15 recognized subspecies (Wilson and Ruff 1999); however, Culver et al. (2000) proposed a reduction of the genus to six subspecies based on genetic diversity. The only known reproducing population of mountain lions in the South today is that of the subspecies *P. c. coryi*, the Florida panther (Whitaker and Hamilton 1998, U. S. Fish and Wildlife Service 2006). The life history of the mountain lion is reviewed by Currier (1983) and Lindzey (1987). Beier et al. (2003) and the U. S. Fish and Wildlife Service (2006) reviewed the literature on the Florida panther.

DISTINGUISHING CHARACTERISTICS

The mountain lion is the largest native North American cat. Measurements are: total length, 150–274 cm; tail, 53.5–90.0 cm; hind foot, 22.0–29.5 cm; ear, 7.5–10.0 cm; weight, 35–100 kg. Adult dorsal pelage is tan but may appear grayish, reddish, or brownish. The cylindrical tail is long and is usually tipped with black. The ears are rounded and lack tufts or black coloration. The pads of the feet have a distinctive three-lobed appearance and the toes are equipped with long, sharp, retractile claws. The skull is short and rounded dorsally with a blunt rostrum and is distinguished from that of the bobcat (*Lynx rufus*) by size and number of teeth. The mountain lion skull exceeds 130 mm in length and has four cheek teeth in the upper jaw whereas the bobcat skull is smaller and has only three upper cheek teeth. The skull of the Florida panther is unique, with a flat, frontal region with broad, high-arched nasal bones (Young and Goldman 1946). The dental formula is I 3/3, C 1/1, P 3/2, M 1/1 = 30 (Figure 1). See keys for details.

The Florida panther is unspotted and typically rusty reddish-brown on the back, tawny on the sides, and pale gray underneath. A right angle crook (kink) near the end of the tail and a hair whorl (cowlick) in the middle of the back were commonly observed in Florida panthers through the early 1990s (U. S. Fish and Wildlife Service 2006). These were considered

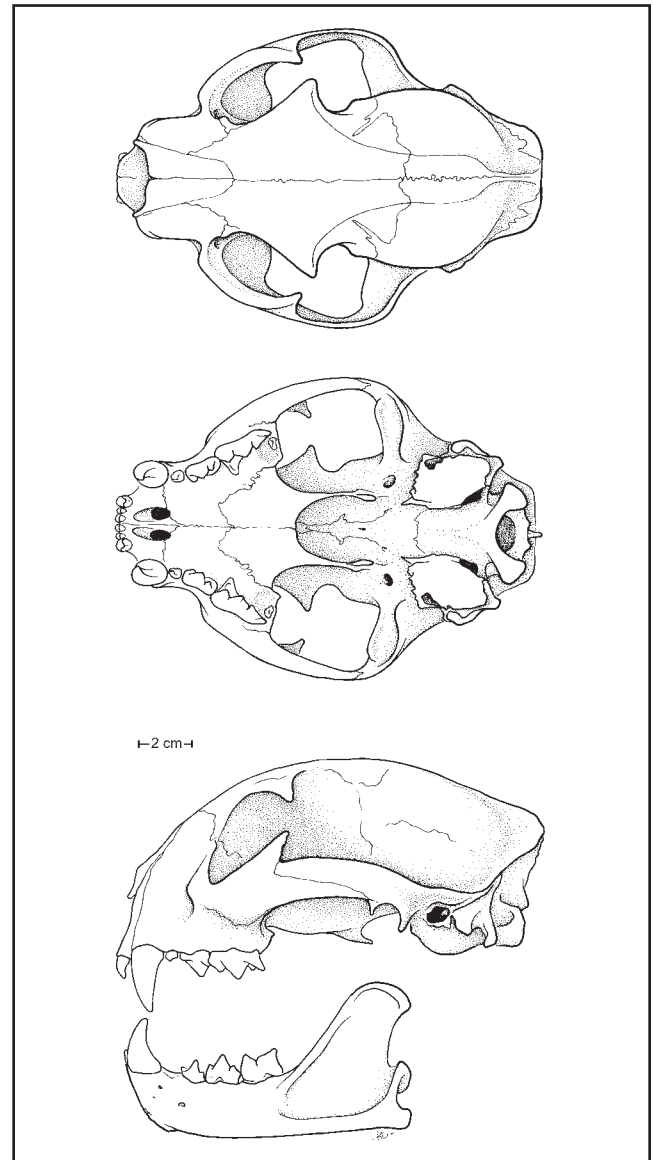


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Puma concolor* from Gila County, Arizona (USNM 271971 female).

expressions of inbreeding (Seal 1994). Since genetic introgression with the mountain lion (*P. c. stanleyana*) released into Florida from west Texas in 1995, these characteristics have dramatically decreased (Land et al. 2004).

CONSERVATION STATUS

The mountain lion has a global rank of Secure (NatureServe 2007). In Florida, it is considered Apparently Secure. It is Presumed Extirpated in Alabama and Kentucky and Possibly Extirpated in Georgia, North Carolina, and South Carolina. It is Imperiled in Texas and Critically Imperiled in Arkansas, Louisiana, Mississippi, and Oklahoma. It is unranked in Virginia. The U. S. Fish and Wildlife Service (U. S. Department of the Interior 2007) lists *P. c. coryi* as Endangered.

DISTRIBUTION

The historical distribution of the mountain lion extended from southern Canada through most of the United States into South America (Hall 1981, Choate et al. 1994, Whitaker and Hamilton 1998). The species disappeared from much of the eastern United States during the latter half of the last century (Downing 1984, McBride et al. 1993). Although generally considered unreliable (U. S. Fish and Wildlife Service 2006), sightings of mountain lions occasionally occur throughout the South. These include areas throughout the central Appalachians in Virginia (Handley 1991) and the southern Appalachians of North Carolina and Tennessee in or near the Great Smoky Mountains National Park (Linzey 1995). Sealander and Heidt (1990) believed that mountain lions might be present in the Ozark-Ouachita Mountains of Arkansas. Recent observations include Oklahoma (Pike et al. 1999), Louisiana (Leberg et al. 2004), and Arkansas (Witsell et al. 1999, Clark et al. 2002). However, the origins of these reported animals could not be determined. They may have been released captive animals (Pike et al. 1997, Heck 1998) or dispersing subadult males from northern and western populations (C. Belden, U. S. Fish and Wildlife Service, personal communication). The dispersal of mountain lion over long distances (1067 km) recently described in the Black Hills of South Dakota (Thompson and Jenks 2005) and reports of verified sightings in the Badlands of North Dakota (North Dakota Game and Fish Department 2006) suggest that these are potential source populations in addition to those in western and southern Texas.

Currently, the only known reproducing population of mountain lions in the South is in Florida (Belden et al. 1988, Maehr 1992, Foster and Humphrey 1995, Land et al. 1998, Main and Richardson 2002). Historically, the Florida panther ranged from Arkansas and Louisiana east to portions of South Carolina and south to Florida (Clark 2001). The panther's current breeding range includes the Big Cypress Swamp and Everglades physiographic region in Collier, Lee,

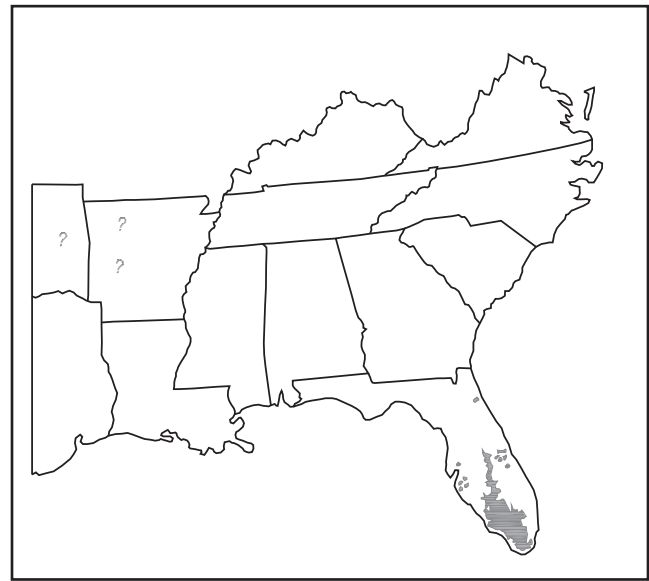


Figure 2. Distribution of *Puma concolor* in the South.

Hendry, Dade, and Monroe counties south of the Caloosahatchee River (Belden et al. 1991). Radio-collared male panthers and uncollared males killed by vehicles (McBride 2002, Belden and McBride 2005) have also been recorded in 11 counties within central and northeastern Florida (Figure 2). Between 1988–1995, 26 Texas mountain lions were released near Okefenokee National Wildlife Refuge and Osceola National Forest to evaluate the feasibility of reintroducing panthers into unoccupied areas of their historic range. Although the animals successfully established territories in the reintroduction areas (U. S. Fish and Wildlife Service 2006), they were subsequently removed from the wild at the end of the study.

ABUNDANCE STATUS

The Florida panther is secretive and occurs at low densities. In the early 1970s, Nowak and McBride (1974) estimated the population from Lake Okeechobee southward at 20–30 animals. After this initial research, additional surveys on public and private lands were completed (Belden et al. 1991). Maehr et al. (1991a) estimated a density of 1 panther/110 km² based on extrapolation of areas occupied by radio-collared panthers during the period 1985–1990 for southwest Florida. McBride (2003) reported a minimum population estimate of 87 panthers based upon a count of adult and subadult panthers. Today, the panther is restricted to a breeding population of less than 100 animals (U. S. Fish and Wildlife Service 2006).

PRIMARY HABITATS

The Florida panther uses a mosaic of habitats for denning, hunting, and dispersal. These include cypress (*Taxodium distichum*) swamps, live oak (*Quercus virginiana*) hammocks, pine flatwoods, cabbage palm (*Sabal palmetto*) forests, flooded prairies, sawgrass (*Cladium jamaicense*) marshes, and agricultural lands (Belden 1986, Maehr 1992). Kautz et al. (in press) reported that forest patches of variable size are important components of the panther landscape, although the species will also use non-forested habitats (Comiskey et al. 2002). Den sites are often located in dense, understory vegetation such as saw palmetto (*Serona repens*; Land et al. 1998). Shindle et al. (2003) reported that 73% of panther dens were located in palmetto thickets. Panther habitat use is also related to prey availability (Janis and Clark 1999, Dees et al. 2001). Although daytime habitat use from radio-telemetry indicates that panthers use the majority of habitats available to them, additional nocturnal research is required to provide a comprehensive understanding of panther habitat relationships (Beier et al. 2003).

REPRODUCTION

Age at first reproduction for the female panther ranges between 1.8–3.2 years; successful reproduction has occurred up to 11 years of age (U. S. Fish and Wildlife Service 2007). Mating may not occur until a territory is established (Maehr et al. 1989). Male panthers are polygynous, maintaining large, overlapping home ranges containing several adult females and their offspring. The average age at sexual maturity is 3 years (Maehr et al. 1991), although some males may breed as early as 17 months (U. S. Fish and Wildlife Service 2007). The breeding activity peaks between December–March (Shindle et al. 2003). Gestation lasts 90–96 days (Maehr 1992). Prior to giving birth, females establish den sites which may include rock crevices, brush piles, thickets, and uprooted trees (Maehr et al. 1989; 1990b). Although litters are produced throughout the year, most births occur between May–June (Jansen et al. 2005). The average litter size ranges from 2–3 kittens; the mean birth interval between litters is 20 months (Lotz et al. 2005). Females that lose their litters generally produce another within an average of 10.4 months (Land 1994). Den sites are used for up to two months by female panthers and their litters. Dispersal of young typically occurs at 18 months, but may occur as early as 12 months (Maehr 1992).

FOOD HABITS

The white-tailed deer (*Odocoileus virginianus*) and feral pig (*Sus scrofa*) are the dominant prey species of the Florida panther (Maehr et al. 1990, Dalrymple and Bass 1996). In the northern portion of its range, feral pigs comprise the bulk of the diet, whereas white-tailed deer are more important in the southern portion (Clark 2001). The northern raccoon (*Procyon lotor*), nine-banded armadillo (*Dasypus novemcinctus*), and marsh rabbit (*Sylvilagus palustris*) are of secondary importance (Dalrymple and Bass 1996). Seasonal variation in the diet has not been detected (U. S. Fish and Wildlife Service 2007). An adult male consumes the equivalent of one white-tailed deer every 8–11 days; this frequency is 14–17 days for a female (Ackerman et al. 1986). Maehr et al. (1990a) documented domestic livestock infrequently in scats or kills, although cattle were readily available on their study area.

ASSOCIATED SPECIES

In addition to its primary prey species, the panther commonly occurs in habitats occupied by the swamp rabbit (*Sylvilagus aquaticus*), round-tailed muskrat (*Neofiber alleni*), marsh rice rat (*Oryzomys palustris*), coyote (*Canis latrans*), common gray fox (*Urocyon cinereoargenteus*), northern river otter (*Lontra canadensis*), striped skunk (*Mephitis mephitis*), and American mink (*Mustela vison*). Potential competitors of the Florida panther include the American black bear (*Ursus americanus*), bobcat, and American alligator (*Alligator mississippiensis*; Beier et al. 2003).

VULNERABILITY AND THREATS

Habitat loss and degradation are significant threats to the continued survival of the panther throughout its range (Belden 1986, Maehr 1992, U. S. Fish and Wildlife Service 2007). Intolerant of human disturbance, panthers are sensitive to habitat fragmentation stemming from road construction, agricultural development, and urban expansion (Beier et al. 2003). Illegal shooting and highway collision also are major problems; off-road vehicle traffic has increased in South Florida, making accessible large areas that formerly had been isolated wilderness (Clark 2001). Due to habitat alteration, a reduced prey base also is a concern. The Florida panther is susceptible to infectious disease such as feline leukemia and pseudorabies virus and environmental contaminants such as mercury, polychlorinated biphenyls, and organochlorines (Land et al. 2004, Newman et al. 2004). Finally, the problems associated with being a small, isolated population have resulted in the loss of genetic variability. Panther populations are losing genetic diversity

by 3–7 percent per generation (Clark 2001). Data on polymorphism and physiological abnormalities suggest that the panther population has experienced inbreeding depression (Roelke et al. 1993, Barone et al. 1994). To address this problem, eight female Texas puma were released into South Florida in 1995 (U. S. Fish and Wildlife Service 2006). The initial results have been promising: atrial septal defects and cryptorchidism have not been found in introgressed panthers, while the occurrence of kinked tails and cowlicks in intercross progeny has been reduced (Land et al. 2004).

MANAGEMENT SUGGESTIONS

Because the Florida panther range includes public and private lands, coordination of management and proactive land use planning is essential. The key to panther conservation is the protection and acquisition of large, interconnected blocks of suitable habitat. Objectives of the recovery plan focus on achieving long-term viability (U. S. Fish and Wildlife Service 2007). These include: (1) maintaining and expanding the panther population and its habitat in South Florida; (2) providing for the expansion of the breeding population into south-central Florida; (3) facilitating panther recovery through public awareness and education; and (4) establishing viable populations of the panther in potential reintroduction areas in other parts of the South. Thatcher et al. (2006) identified prospective sites for panther reintroduction within the historic range based on several landscape parameters (e.g., natural and urban land cover, contagion, patch density, human population density, and road density). Of the nine areas identified, the Okefenokee National Wildlife Refuge in Georgia, and the Ozark National Forest and Felsenthal National Wildlife Refuge in Arkansas rank as the top prospective reintroduction sites based on effective habitat area and expert opinion (Thatcher et al. 2006).

Additional conservation measures include implementing a genetics management plan to monitor physical and physiological characteristics correlated with the depletion of genetic variability; devising appropriate biomedical strategies to limit population disease threats; and actively managing white-tailed deer and feral pig populations and their habitats. The construction of wildlife crossings, road signs, and fencing can ameliorate highway fatalities where roadways intersect panther habitat; incorporating habitat connectivity and dispersal corridors into road expansion projects is key. Public support is critical to attainment of recovery goals and reintroduction efforts; political and social issues must be resolved before these efforts are initiated.

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Bobcat

Lynx rufus (Schreber, 1777)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Disagreement has existed for many years about the nomenclature of the bobcat (Jones et al. 1986, 1992). The arguments have centered around whether short-tailed cats such as the bobcat should be included in the genus *Felis* or *Lynx*. This guide follows Jones et al. (1997). There are 12 recognized subspecies of bobcat; three occur in the South: *L. r. rufus*, *L. r. floridanus*, and *L. r. texensis*. The literature is reviewed by Young (1958), McCord and Cardoza (1982), Boyle and Fendley (1987), and Rolley (1987).

DISTINGUISHING CHARACTERISTICS

The bobcat is a medium-sized carnivore, twice the size of a large domestic cat (*Felis catus*). Measurements are: total length, 810–1,050 mm; tail, 130–185 mm; hind foot, 160–185 mm; ear, 60–78 mm; weight, 5.7–20 kg. The dorsal pelage is reddish-brown, grayish-brown, or buffy-brown; however, melanistic bobcats are reported from Florida (Ulmer 1941, Paradiso 1973, Regan and Maehr 1990). The ventral pelage is white or whitish. Black spots are usually present on both the dorsum and venter; there are conspicuous black bars on the insides of the front legs. The backs of the ears have a large white patch, surrounded by black, near the base and sometimes terminate in a black tuft at the tip. The large eyes have elliptical pupils. The hair on the sides of the face forms a long, sideburn-like ruff. The tail is short, usually shorter than the length of the hind foot, with a black tip dorsally and white fur underneath. An elevated fifth toe is present on the front feet. Bobcats have retractile claws. The broad, rounded skull is distinguished by the presence of two upper premolars on each side. The skull of the mountain lion (*Puma concolor*) and domestic cat have three upper premolars per side. The dental formula is: I 3/3, C 1/1, P 2/2, M 1/1 = 28 (Figure 1). See keys for details.

CONSERVATION STATUS

The bobcat has a global rank of Secure (NatureServe 2007). It is considered Secure in Georgia, Mississippi, Tennessee, and Texas and Apparently Secure in Alabama, Arkansas, Kentucky, Louisiana, North Carolina, Oklahoma, and Virginia. It is unranked in Florida and South Carolina. The species has status as

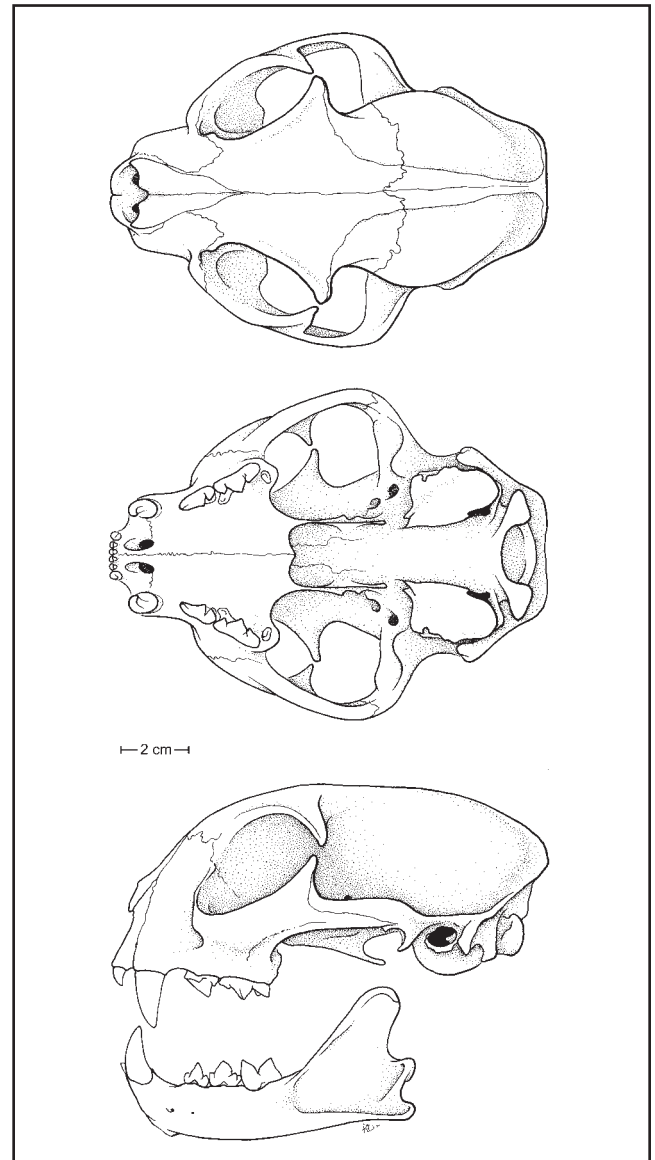


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Lynx rufus* from Birch Creek, Idaho (USNM 30729, gender unknown).

a furbearer (Woolf and Hubert 1998). In Virginia, *L. r. floridanus* is at the northern limits of its range with uncertain population status (Handley 1991).

DISTRIBUTION

The bobcat occurs across southern Canada, throughout the United States, extending southward into Mexico (Hall 1981, Choate et al. 1994). Bobcat distribution in the South is depicted in Figure 2. It occurs throughout Virginia (Bailey 1946, Handley and Patton 1947, Handley 1979, 1991, 1992; Linzey 1998), North Carolina (Johnston 1967, Linzey and Linzey 1971, Hair et al. 1979, Lee et al. 1982, Miller et al. 1983, Clark et al. 1985, Webster et al. 1985, Lancia et al. 1986, Murray et al. 1992, Linzey 1995), South Carolina (Penny 1950, Golley 1966, Marshall and Jenkins 1966, Provost et al. 1973, Buie et al. 1979, Webster et al. 1985, Griffith and Fendley 1986, Heller and Fendley 1986, Cothran et al. 1991), and Georgia (Golley 1962, Hon 1979, Laerm et al. 1980, 1981; James 1992, Baker et al. 1993). The species was extirpated from Georgia barrier islands (Johnson et al. 1974); it has been reintroduced on Cumberland Island (Warren et al. 1990, James 1992, Baker et al. 1993, 2001; Diefenbach et al. 1993, 1994; Brooks et al. 1999). The bobcat ranges throughout Florida (Layne 1974, Brady 1979, Conner et al. 1983, Maehr and Brady 1986, Wassmer et al. 1988, Cames and Forrester 1998, Labisky and Boulay 1998), Alabama (Howell 1921, Holliman 1963, Miller and Speake 1978, 1979; Miller 1980, Sumner and Hill 1980, Cames and Forrester 1998), Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989, Conner et al. 1992, 1999; van Domelen et al. 1992, Stys and Leopold 1993, Conner and Leopold 1996, Lovell et al. 1998, Chamberlain et al. 1999, Chamberlain and Leopold 1999), Louisiana (Lowery 1974, Hall and Newsom 1976, Morrison et al. 1981, Linscombe et al. 1983, Shiflet 1984, Zwank et al. 1985), Texas (Blankenship and Swank 1979, Schmidly 1983, Stapper et al. 1989, Davis and Schmidly 1994), Oklahoma (Rolley 1985a,b; Rolley and Warde 1985, Caire et al. 1989), Arkansas (Fritts and Sealander 1978a,b; Rucker et al. 1989, Sealander and Heidt 1990), Tennessee (Linzey and Linzey 1971, Smith et al. 1974, Story et al. 1982, Kitchings and Story 1984, Kennedy 1991, Gabor et al. 1994, Linzey 1995), and Kentucky (Barbour and Davis 1974, Whitaker et al. 1987).

ABUNDANCE STATUS

Populations of bobcat in the South are considered to be stable or increasing. Densities of the bobcat vary considerably related to habitat type and prey density (McCord and Cardoza 1982). In the Ouachita Mountains, density ranges from 1/9.6 km² (Rucker et al. 1989) to 1/11 km² (Rolley 1985a). Population densities fluctuate between 14–42 bobcats/100 km² in south-central Florida (Wassmer et al. 1988). Estimates of population densities elsewhere in the region include 0.58/km² in South Carolina (Provost et al.

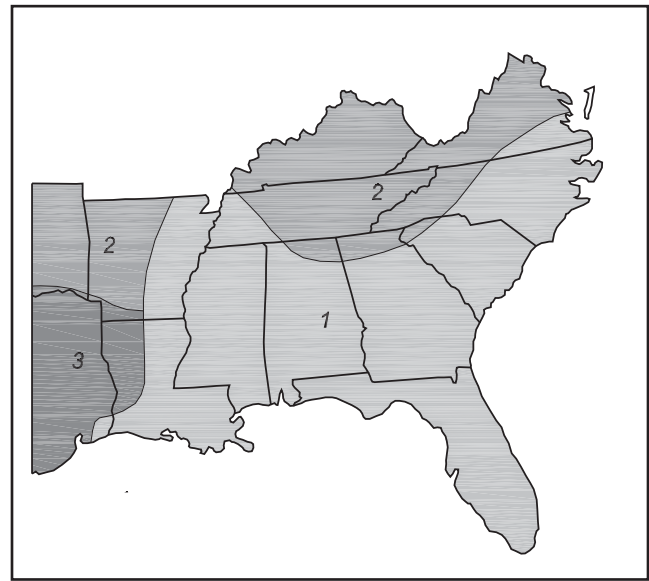


Figure 2. Distribution of *Lynx rufus* in the South: (1) *L. r. floridanus*; (2) *L. r. rufus*; (3) *L. r. texensis*.

1973) and 0.8–1.2/km² in Alabama (Miller and Speake 1978).

PRIMARY HABITATS

The bobcat uses many, if not most, terrestrial habitat types. It prefers areas with dense understory vegetation and high prey densities (Boyle and Fendley 1987, Rolley 1987). In the South, the bobcat inhabits both mature hardwood forests and pine (*Pinus* spp.) regeneration sites less than 20 years old (Hall and Newsom 1976, Rucker et al. 1989, Conner and Leopold 1993, 1996). Bottomland hardwood forests are preferred habitats in South Carolina (Heller and Fendley 1986) and Florida (Wassmer et al. 1988). Such areas often support relatively high densities of rodents and lagomorphs (Rolley and Warde 1985). The bobcat generally avoids mature closed-canopy stands of pine and agricultural areas lacking brushy cover. Home ranges and movement patterns are altered to avoid paved roads (Lovallo and Anderson 1996). When available, the bobcat uses rocky outcrops and ledges for daytime retreats and for den sites (Rolley 1987). In areas where rocks are not present, the species may use brush piles, depressions at the base of stumps, or hollow logs and trees as den sites (Gashwiler et al. 1961, Kitchings and Story 1984). In Florida, Winegarner (1985) observed a bobcat with three kittens using a gopher tortoise (*Gopherus polyphemus*) burrow.

REPRODUCTION

Although breeding may occur throughout the year, most breeding activity takes place from December to

March (Fritts and Sealander 1978a, Stys and Leopold 1993). Reported gestation periods range from 50–70 days (McCord and Cardoza 1982). The species produces a single litter per year during the summer months, but 2 litters/year have been reported (Winegarner and Winegarner 1982). Litter size ranges from 1–6 (Gashwiler et al. 1961); the mean litter size is 3. At birth, kittens are furred and blind. Their eyes open within 11 days and they nurse for approximately 2 months (Young 1958, Jackson et al. 1989). The young remain with their mother until the following spring (Griffith et al. 1980, Kitchings and Story 1984). Females are capable of breeding at 9–12 months of age (Crowe 1975, Fritts and Sealander 1978b); males may not reach sexual maturity until 1.5 years (Fritts and Sealander 1978b). Maximum longevity in the wild is 10–14 years (Young 1958).

FOOD HABITS

Rabbits (*Sylvilagus* spp.) and hispid cotton rats (*Sigmodon hispidus*) are important prey foods. The species also consumes a variety of rodents and medium-sized mammals such as the Virginia opossum (*Didelphis virginiana*) and nutria (*Myocastor coypus*). It also takes Florida sandhill cranes (*Grus canadensis pratensis*), gallinaceous and passerine birds, snakes, and insects (Beasom and Moore 1977, Fritts and Sealander 1978a, Miller and Speake 1978, Buttrey 1979, Kight 1982, Story et al. 1982, Miller et al. 1983, Kitchings and Story 1984, Rolley 1985b, Rolley and Warde 1985, Bennett and Bennett 1990, Murray et al. 1992, Van Domelen et al. 1992, Chamberlain and Leopold 1999, Baker et al. 2001). The bobcat occasionally kills white-tailed deer (*Odocoileus virginianus*), but the species is not regarded as a major deer predator (Fritts and Sealander 1978a, Kight 1982, Miller et al. 1983, Labisky and Boulay 1998).

ASSOCIATED SPECIES

The bobcat is widely distributed and uses a diversity of habitats. It associates with most mammals in the region.

VULNERABILITY AND THREATS

The bobcat is not susceptible to many parasites or diseases. However, the species can carry rabies, toxoplasmosis, and feline distemper (Davidson and Nettles 1997). Extensive clearing of forests that fragment bobcat habitat may be a localized threat in some instances.

MANAGEMENT SUGGESTIONS

Habitat management centers on activities such as timber harvest and prescribed fire that maintain a mosaic of cover types that include early to mature successional seres. Forest interspersed with oldfields and brushy areas are particularly beneficial. In addition, Allen (1987) suggests protecting vegetation in riparian areas to promote dispersal. Population levels are monitored through annual harvest statistics by state wildlife agencies in the South.

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Striped Skunk

Mephitis mephitis (Schreber, 1776)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The striped skunk (*Mephitis mephitis*) has 13 recognized subspecies (Miller and Kellogg 1955, Hall 1981). Three subspecies occur in the South: *M. m. elongata*, *M. m. mesomelas*, and *M. m. nigra*. The natural history of the striped skunk is reviewed by Verts (1967), Godin (1982), Wade-Smith and Verts (1982), and Rosatte (1987).

DISTINGUISHING CHARACTERISTICS

Mephitis mephitis is a distinctly patterned skunk about the size of a large domestic cat (*Felis domesticus*). Measurements are: total length, 520–770 mm; tail, 170–280 mm; hind foot, 65–85 mm; ear, 25–30 mm; weight, 1.2–5.5 kg. Males are larger than females. The striped skunk is recognized easily by the presence of two white stripes on its black or brownish-black dorsal pelage. The stripes originate from a narrow white stripe on the forehead that expands on the nape and divides into two dorso-lateral stripes. The extent of the stripe is variable and some individuals are mostly black whereas others are mostly white dorsally (Verts 1967). Several color morphs (brown, white, and yellow) also are documented (Detlefsen and Holbrook 1921). The fur is dense and oily. The legs are short and there are long, curved claws on the forefeet. The head is small and triangular. The tail is long and bushy, frequently tipped with white. The skull usually exceeds 70 mm in length and is rounded when viewed from the side. The braincase is elongate but the rostrum is short. When viewed from above, the mastoid region appears inwardly curved. The dental formula is: I 3/3, C 1/1, P 3/3, M 1/2 = 34 (Figure 1). See keys for details.

CONSERVATION STATUS

The striped skunk has a global rank of Secure (NatureServe 2007). It is also considered Secure in Georgia, Kentucky, Mississippi, Oklahoma, Tennessee, Texas, and Virginia. It is Apparently Secure in Alabama, Arkansas, Louisiana, and North Carolina. It is unranked in Florida and South Carolina.

DISTRIBUTION

Mephitis mephitis is distributed widely in North America; the species occurs from southern Canada to

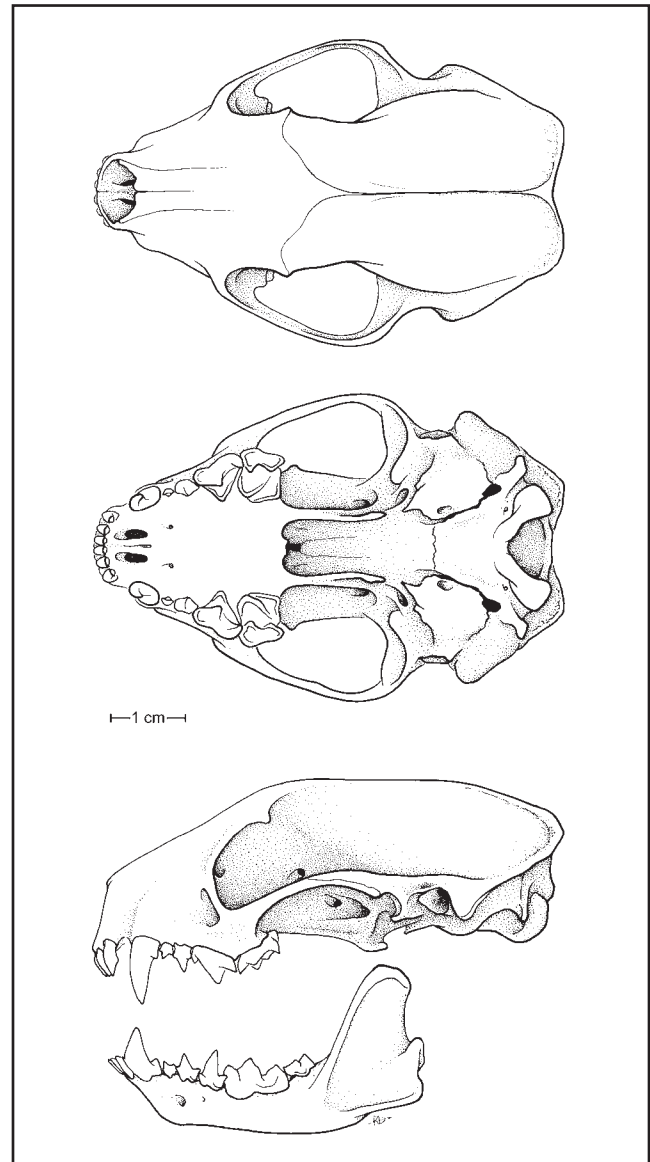


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Mephitis mephitis* from Arlington County, Virginia (USNM 275125, female).

northern Mexico (Hall 1981, Choate et al. 1994). The distribution of the striped skunk in the South is depicted in Figure 2. It is found throughout Virginia (Bailey 1946, Handley and Patton 1947, Stout and Shonenshine 1974, Handley 1992, Linzey 1998) but it is rare in the Coastal Plain of North Carolina (Calhoun

1941, Conaway and Howell 1953, Johnston 1967, Linzey and Linzey 1971, Lee et al. 1982, Clark et al. 1985, Webster et al. 1985, Linzey 1995). It is locally common throughout South Carolina (Penny 1950, Golley 1966, Cothran et al. 1991), Georgia (Golley 1962, Landers et al. 1980, Laerm et al. 1981, Parker et al. 1992), Florida (Hall 1981), Alabama (Howell 1921, Holliman 1963), and Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989). Although it is not recorded from the Mississippi Delta of southeastern Louisiana, it occurs elsewhere in the state (Lowery 1974). It is common in eastern Texas (Wood 1954, McCarley 1959, Schmidly 1984, Davis and Schmidly 1994) and eastern Oklahoma (Blair 1939, Caire et al. 1989) and it is distributed statewide in Arkansas (Heidt et al. 1982, Sealander and Heidt 1990), Tennessee (Howell and Conaway 1952, Goodpaster and Hoffmeister 1952, Conaway and Howell 1953, Linzey and Linzey 1971, Smith et al. 1974, Weller and Pelton 1987, Kennedy 1991, Linzey 1995, Bixler 2000), and Kentucky (Barbour and Davis 1974, Fassler 1974).

ABUNDANCE STATUS

The striped skunk is considered abundant throughout its range. Density estimates have been recorded from 0.7–26/km²; most average between 2–5/km² (Jones 1939, Scott and Selko 1939, Allen and Shapton 1942, Burt 1946, Ferris and Andrews 1967, Verts 1967, Bailey 1971, Stout and Shonenshine 1974). Density estimates are influenced by several factors including disease (e. g., rabies, distemper), winter weather, availability of den sites, and food supply. Density also varies with season and geographic area. In Illinois the minimum-area home range calculated for striped skunks was 511 ha for adult males, 378 ha for adult females, 284 ha for juvenile males, and 234 ha for juvenile females (Storm 1972). Home ranges tend to be linear (Verts 1967, Storm 1972).

PRIMARY HABITATS

The striped skunk occurs in all habitat types in the region. The habitats that support the greatest population density consist of interspersed woodlands, open fields with wooded ravines, brushy zones such as fencerows, and rock outcrops (Hamilton 1943, Burt 1946, Schwartz and Schwartz 1959, Verts 1967, Storm 1972). However, the striped skunk also may be abundant in urban environments, pastures, hayfields, and intensively cultivated areas (Verts 1967, Storm 1972, Rue 1981, Parker et al. 1992). Underground dens are used from late fall to early spring, and by lactating females in late spring and summer (Verts 1967, Houseknecht 1969, Storm 1972, Houseknecht and Tester 1978). Dens excavated by other mammals such

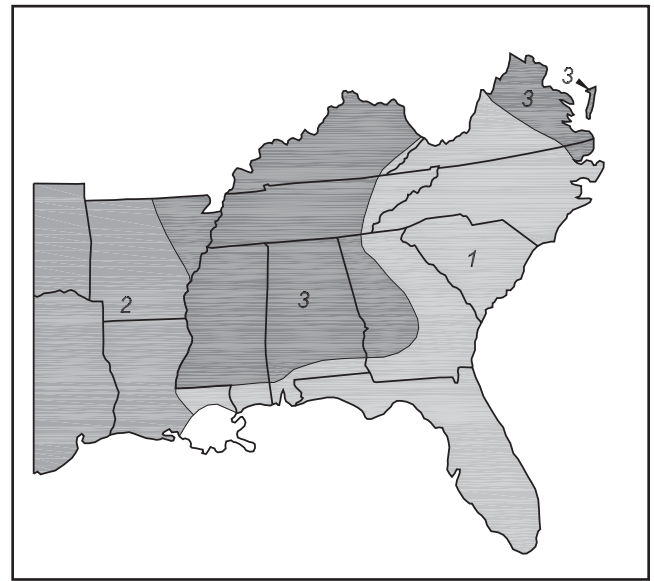


Figure 2. Distribution of *Mephitis mephitis* in the South: (1) *M. m. elongata*; (2) *M. m. mesomelas*; (3) *M. m. nigra*.

as the woodchuck (*Marmota monax*) are used when available, but the striped skunk digs its own den when necessary (Selko 1938, Allen and Shapton 1942, Verts 1967). During the winter, several female striped skunks may share a den but males rarely den communally (Allen and Shapton 1942, Verts 1967, Houseknecht 1969). In spring and summer, the species uses hollow logs and brush piles, stumps, bases of live and dead trees, and culverts (Houseknecht and Tester 1978, Weller and Pelton 1987, Lariviere et al. 1999).

REPRODUCTION

The mating season for the striped skunk is from February–April (Wade-Smith et al. 1980). The gestation period ranges from 59–77 days, which suggests that delayed implantation may occur (Wade-Smith and Richmond 1978, Wade-Smith et al. 1980). From 5–9 young are born in May–June (Wade-Smith et al. 1980, Rue 1981, Godin 1982, Greenwood and Sargeant 1994). Although the striped skunk is usually monestrous, second litters are known from July (Shadle 1956, Parks 1967). Young are born helpless and blind and are nursed for 6–7 weeks (Hamilton and Whitaker 1979). The young remain with their mother until August or September. Few wild individuals live beyond 3 years, but the age potential is 10 years (Linduska 1947).

FOOD HABITS

The striped skunk is opportunistic and the diet varies seasonally. Grasshoppers (Orthoptera) and beetles (Hemiptera) are major components of the diet (Verts

1967). The striped skunk also preys upon crawfish, frogs, small mammals, and bird eggs. It also consumes fruits and grains (Selko 1937, Landers et al. 1980, Rue 1981). The consumption of vertebrates and vegetation increases during fall (Cuyler 1924).

ASSOCIATED SPECIES

The striped skunk is distributed widely and associates with most small mammals in the region. It often uses the dens of the common gray fox (*Urocyon cinereoargenteus*), red fox (*Vulpes vulpes*), nine-banded armadillo (*Dasypus novemcinctus*), American badger (*Taxidea taxus*), and woodchuck.

VULNERABILITY AND THREATS

There are no threats to the survival of *Mephitis mephitis* in the region. Mortality on paved roadways is common (Whitaker and Hamilton 1998). The species undoubtedly benefits from a mosaic of land-use patterns.

MANAGEMENT SUGGESTIONS

The striped skunk provides many ecological benefits, including control of rodents and insects (Whitaker and Hamilton 1998). The skunk has been managed more as a nuisance species than as a furbearer. The skunk is not harvested as intensely as the northern raccoon (*Procyon lotor*) for its pelt; fur prices and market fluctuations are unlikely to affect population viability (Chamberlain and Leopold 2001). Management usually centers on removal in agricultural and urban settings. Because the skunk is a major carrier of rabies, direct contact requires care (Davidson and Nettles 1997).

In localities where the support of striped skunk populations is desirable, maintaining suitable habitat centers on maintenance of early seral conditions, fields, forest openings, as well as provision of brushy cover. In addition, Allen (1987) recommends providing activities that promote understory vegetation diversity and encourage establishment of fruit-producing trees and shrubs. The availability of den sites may effectively limit striped skunk populations. Additional research is needed on rabies acquisition, reproductive requirements, and population dynamics.

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Eastern Spotted Skunk

Spilogale putorius (Linnaeus, 1758)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The North American spotted skunks, genus *Spilogale*, were the subjects of taxonomic controversy. Hall and Kelson (1959) recognized four species of spotted skunks in North America. Following Van Gelder's (1959) revision of the genus, Hall (1981) recognized only two species, the spotted skunk (*S. putorius*) and the pygmy spotted skunk (*S. pygmaea*). However, Mead (1968b) demonstrated that the eastern spotted skunk (*S. putorius*) and the western spotted skunk (*S. gracilis*) were geographically and reproductively isolated. Both Jones et al. (1992) and Wozencraft (1993) treat these as separate species. *Spilogale putorius* is the only spotted skunk that occurs east of the Continental Divide in North America. Three described subspecies occur in the South: *S. p. putorius*, *S. p. ambarvalis*, and *S. p. interrupta*. The life history of the eastern spotted skunk is reviewed by Howard and Marsh (1982), Rosatte (1987), and Kinlaw (1995).

DISTINGUISHING CHARACTERISTICS

Spilogale putorius is a squirrel-sized skunk with spots (rather than distinct longitudinal stripes) on the dorsum. Measurements are: total length, 470–585 mm; tail length, 175–240 mm; hind foot, 40–55 mm; ear, 20–26 mm; weight, 325–650 g. The pelage of the eastern spotted skunk is black but the dorsum has four to six broken stripes and several white spots. A large white spot also is present between the eyes. The tail is bushy and tipped with white. The claws on the forefeet are twice as long as those of the hind feet. The skull is flat dorsally and the rostrum at the anterior insertion of the zygomatic arch is almost wide as the braincase. When viewed from above, the mastoid region appears to be flared outwardly. The skull is less than 70 mm in length. The skull of the striped skunk (*Mephitis mephitis*) exceeds 70 mm in length, is rounded dorsally, and the mastoid regions appears to be inwardly-curved. The dental formula of the eastern spotted skunk is $I\ 3/3, C\ 1/1, P\ 3/3, M\ 1/2 = 34$ (Figure 1). See keys for details.

CONSERVATION STATUS

The eastern spotted skunk has a global rank of Secure (NatureServe 2007). However, it is classified as Apparently Secure in Arkansas, Georgia, South Carolina, and Texas. It is Vulnerable in North Carolina,

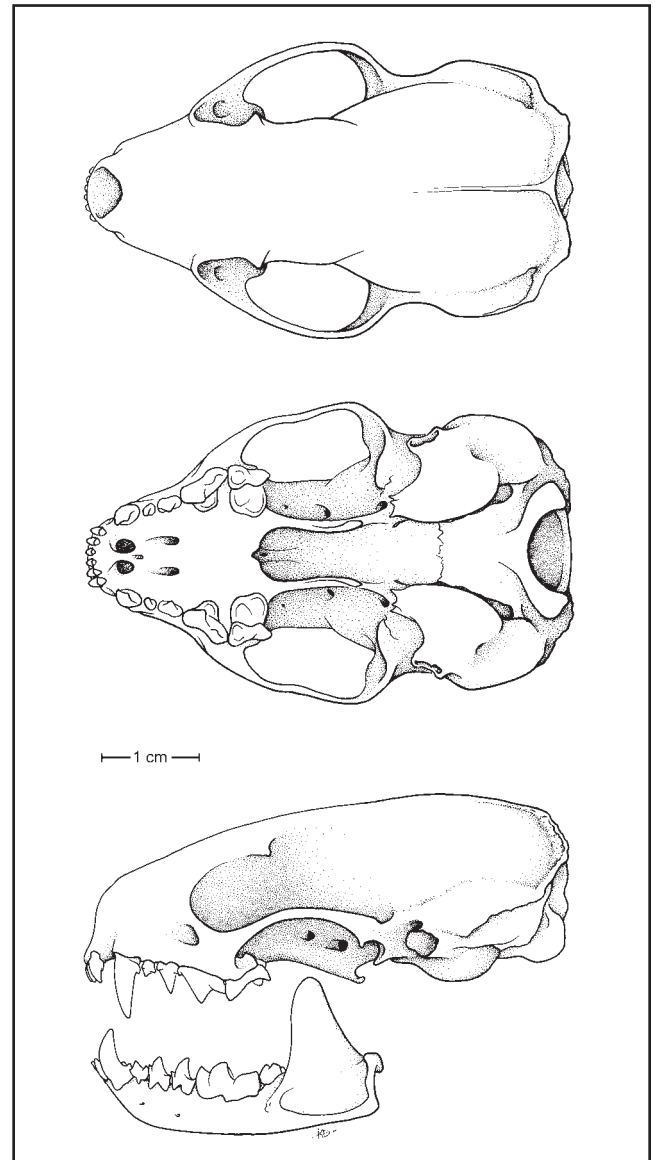


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Spilogale putorius* from Brevard County, Florida (USNM 70306, male).

Virginia, and Tennessee. It is Imperiled in Alabama, Kentucky, Mississippi, and Oklahoma. In Louisiana, it is Critically Imperiled. It is unranked in Florida. Two subspecies are of special concern in several states: *S. p. putorius* and *S. p. interrupta*.

DISTRIBUTION

Spilogale putorius is distributed from the Continental Divide eastward through most of the United States and southward into northeastern Mexico along the Gulf Coast (Hall 1981, Choate et al. 1994). The distribution of the spotted skunk in the South is depicted in Figure 2. The species occurs in the Appalachian Mountains of Virginia (Bailey 1946, Handley and Patton 1947, Handley 1992, Linzey 1998) and North Carolina (Conaway and Howell 1953, Van Gelder 1959, Johnston 1967, Linzey and Linzey 1971, Lee et al. 1982, Webster et al. 1985, Linzey 1995). In South Carolina, the species occurs in the Appalachian Mountains and in the Piedmont region on the western edge of the state (Burnett 1851, Howell 1906, Pickens 1938, Penny 1950, Golley 1966, Webster et al. 1985, Cothran et al. 1991). The eastern spotted skunk is absent from the Atlantic Coastal Plain but is present elsewhere in Georgia (Emmons et al. 1949, Golley 1962, Laerm et al. 1981). The species occurs statewide in Alabama (Howell 1921, Holliman 1963) and almost statewide in Florida (Bangs 1898, Howell 1920, Hamilton 1941, Schwartz 1952, Van Gelder 1959, Manaro 1961, Ehrhart 1974, Layne 1974, Frank and Lips 1989, Kinlaw et al. 1995). The species occurs in most of Mississippi but is absent from the Mississippi Alluvial Valley (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989). It is rare in the Atchafalaya Basin and the northern half of Louisiana (Lowery 1974). It occurs in eastern Texas (Van Gelder 1959, Schmidly 1983, 1984; Davis and Schmidly 1994) and eastern Oklahoma (Caire et al. 1989). The spotted skunk may occur statewide in Arkansas, but there have been few sightings in the western third of the state or along the Mississippi River (Sealander and Heidt 1990). The species is distributed in portions of central and eastern Tennessee (Kellogg 1939, Conaway and Howell 1953, Linzey and Linzey 1971, Smith et al. 1974, Kennedy 1991, Linzey 1995, Reed and Kennedy 2000), and eastern Kentucky (Barbour and Davis 1974).

ABUNDANCE STATUS

The subspecies found in peninsular Florida, *S. p. ambarvalis*, is abundant (Kinlaw et al. 1995). When data collected by Ehrhart (1974) were re-analyzed, a population density of 40/km² at Canaveral National Seashore was reported (Kinlaw et al. 1995). The other two subspecies have declined in numbers (Kinlaw 1995). Throughout much of the South, the eastern spotted skunk is rare and probably extirpated in many localized areas.

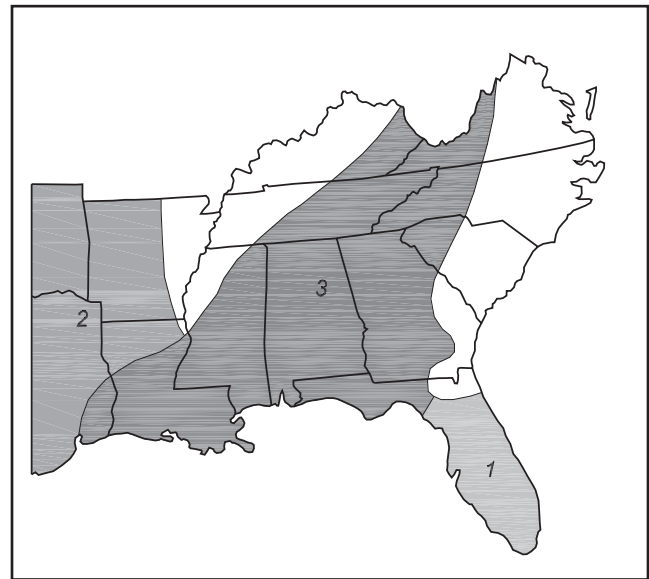


Figure 2. Distribution of *Spilogale putorius* in the South: (1) *S. p. ambarvalis*; (2) *S. p. interrupta*; (3) *S. p. putorius*.

PRIMARY HABITATS

The eastern spotted skunk may occupy brushy, rocky, and wooded habitats, but it tends to avoid wetland areas (Ehrhart 1974, Kinlaw 1995). It often is captured in areas with dense vegetative cover, but rarely inhabits open habitats (Crabb 1948, Manaro 1961, McCullough and Fritzell 1984). In Missouri, it uses oak-hickory (*Quercus-Carya*) forests (McCullough and Fritzell 1984). Populations of eastern spotted skunks are dense in saw palmetto (*Serenoa repens*) thickets along the Atlantic coast of Florida (Bangs 1898, Kinlaw et al. 1995) and individuals occasionally wander onto beaches (Howell 1906). The species dens within any natural cavity or crevice (Crabb 1948, Lee et al. 1982), the burrows of other mammals (Crabb 1948; Lowery 1974), and the burrows of the gopher tortoise (*Gopherus polyphemus*; Frank and Lips 1989). In agricultural or urban areas, the eastern spotted skunk dens in haystacks, woodpiles, and abandoned buildings (Crabb 1948).

REPRODUCTION

The breeding season is from March–April. The eastern spotted skunk does not exhibit delayed implantation. Young are born in late May–June after a gestation period of 50–65 days. The mean litter size is 5.5 (Mead 1968a). The young are born blind with a thin coat of distinctly spotted fur. They are weaned at about 54 days of age (Crabb 1944, 1948). Females of *S. p. ambarvalis* may have a second estrus in July or August, and produce a second litter (Mead 1968a).

FOOD HABITS

The eastern spotted skunk is primarily insectivorous, consuming Orthoptera (crickets, grasshoppers), Hemiptera (beetles), Oligochaeta (earthworms), and grubs. It will often catch small mammals and birds, especially during winter and spring. The species will also consume fruit, corn, and other plant material (Selko 1937, McCullough and Fritzell 1984). The eastern cottontail (*Sylvilagus floridanus*) and voles (*Microtus* spp.) are important prey during winter in Iowa (Crabb 1941).

ASSOCIATED SPECIES

The eastern spotted skunk occupies the same habitat as the striped skunk, long-tailed weasel (*Mustela frenata*), least weasel (*M. nivalis*), eastern cottontail, and several rodents. The skunk sometimes uses the burrow of the gopher tortoise, nine-banded armadillo (*Dasypus novemcinctus*), eastern woodrat (*Neotoma floridana*), Allegheny woodrat (*N. magister*) striped skunk, and long-tailed weasel (Crabb 1948, Lowery 1974, Frank and Lips 1989, Kinlaw 1995).

VULNERABILITY AND THREATS

The reason for the apparent decline in the two subspecies is unclear. Human activities are commonly suggested as factors associated with the population declines; these include roadkills (Rosatte 1987), trapping, and poisoning (Howard and Marsh 1982). However, Polder (1968) notes that habitat alteration associated with modern agricultural practices also may be a significant decimating factor. The feral dog (*Canis familiaris*) is another potential enemy of the spotted skunk (Crabb 1948).

MANAGEMENT SUGGESTIONS

The skunk provides an important role in the control of rodents and insects harmful to agriculture. The pelt of the spotted skunk is not considered as valuable as the striped skunk; Chamberlain and Leopold (2001) consider the species economically insignificant as a southern furbearer. Because the skunk may carry rabies, caution is warranted.

In localities where the support of skunk populations is desirable, management centers on maintenance of early seral conditions, fields, forest openings, as well as provision of brushy cover. Allen (1987) suggests activities that promote understory vegetation diversity, maintain shelterbelts and travel corridors, and encourage establishment of fruit-producing shrubs. Additional research is needed on specific habitat and reproductive requirements, and on their ectoparasites.

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Northern River Otter

Lontra canadensis (Schreber, 1777)

Margaret K. Trani and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The Nearctic northern river otter was recognized as distinct from Eurasian genera by van Zyll de Jong (1972, 1987). Wozencraft (1993) and Baker et al. (2003) followed van Zyll de Jong in using *Lontra* as the generic name. However, some authors (e.g., Whitaker and Hamilton 1998) continue to place the species in genus *Lutra*. Seven subspecies currently are recognized (Hall 1981, Lariviere and Walton 1998); one subspecies (*L. c. lataxina*) occurs in the South. The life history of the northern river otter is reviewed by Toweill and Tabor (1982), Melquist and Dronkert (1987), Lariviere and Walton (1998), and Melquist et al. (2003).

DISTINGUISHING CHARACTERISTICS

The northern river otter has a large, long body with short legs and a hydrodynamic shape that distinguishes it from other mustelids. Feet are pentadactyl and plantigrade with interdigital webbing pronounced on the longer toes of the hind foot (Melquist et al. 2003). The tail is about one-third of total length and tapered from base to tip. Measurements are: total length, 890–1200 mm; tail, 350–520 mm; hind foot, 100–140 mm; ear, 20–30 mm; weight, 4.5–15 kg. Females are 3–21% smaller than males (Blundell et al. 2002). The short, thick, and glossy pelage ranges from dark brown to dark reddish-brown dorsally, and pale brown to silver-gray ventrally. The throat and muzzle often are silvery gray to brownish-white. The ears are round and inconspicuous. The small eyes are positioned anteriorly (Lariviere and Walton 1998). The muzzle is broad with stiff vibrissae bordering the nose. The skull is relatively flat with a short, broad rostrum; the cranium narrows at the eye sockets. The auditory bullae are flattened. The teeth are adapted for crushing and cutting. The dental formula is I 3/3, C 1/1, P 4/3, M 1/2 = 36 (Figure 1). See keys for details.

CONSERVATION STATUS

The northern river otter has a global rank of Secure (NatureServe 2007). It is classified Secure in Georgia and Apparently Secure in Alabama, Arkansas, Louisiana, Mississippi, North Carolina, and Virginia. Kentucky, Tennessee, and Texas consider the species Vulnerable. It is Imperiled in Oklahoma and is

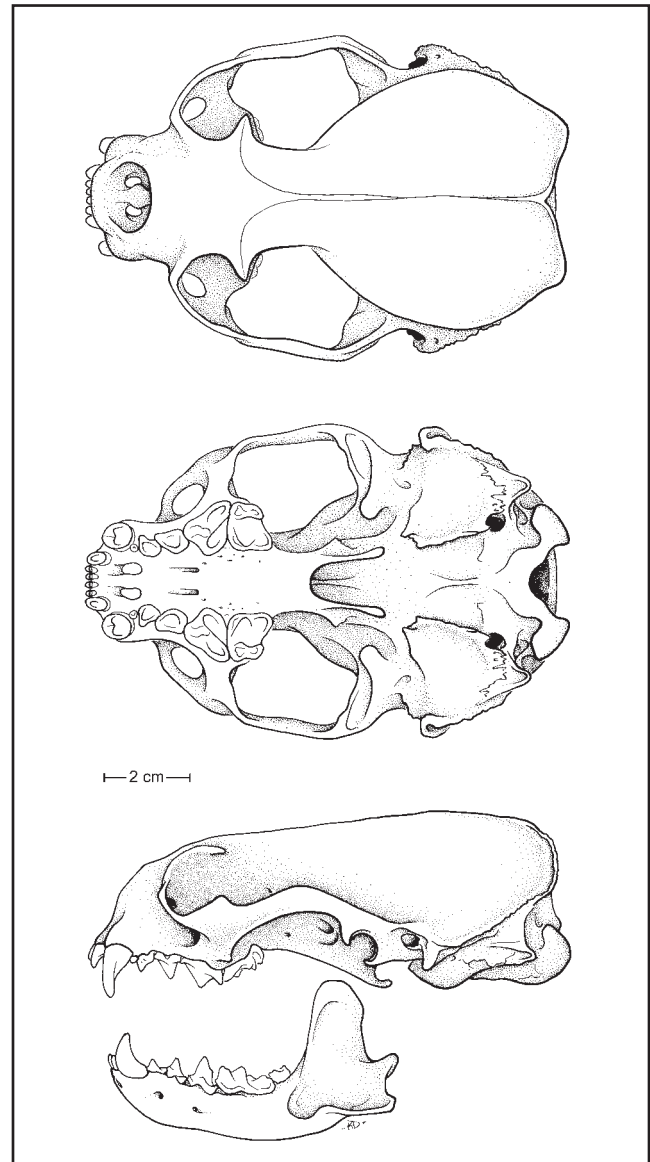


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Lontra canadensis* from Eleuthera, Bahama Islands (USNM 122018,

Unranked in Florida and South Carolina. The otter is protected under the Convention for the International Trade of Endangered Species of Fauna and Flora (CITES) because of concerns regarding trade in the European otter. The otter is closely monitored by states that allow harvest (Leopold and Chamberlain 2001). With the exception of Oklahoma, all southern

states consider the northern river otter a furbearing species with varying regulations on method of take and bag limit.

DISTRIBUTION

The northern river otter historically occurred throughout most major drainages and riverine habitats of the continental United States and Canada (Hall 1981, Choate et al. 1994). Excessive harvest, water pollution (including pesticide bio-magnification from fish consumption), and wetland drainage for agriculture and other development resulted in widespread population decline (Melquist et al. 2003). Between 1982–1995, otter reintroduction and population enhancement efforts were undertaken in Kentucky, Oklahoma, Virginia, Tennessee (including Great Smoky Mountains National Park), and North Carolina (Tango et al. 1991, Raesly 2001). Currently, the river otter is distributed throughout the South (Figure 2), including all of Virginia (Handley 1992, Linzey 1998; M. Fies, Virginia Game and Inland Fisheries, personal communication), North Carolina (Lee et al. 1982, Webster et al. 1985), Georgia (Laerm et al. 1981; J. Ozier, Georgia Department of Wildlife Resources, personal communication) and most waterways in South Carolina (Cothran et al. 1991, J. Butfiloski, South Carolina Department of Natural Resources, personal communication). The otter also is present throughout the waterways of Florida (Whitaker and Hamilton 1998; K. Hodges, Florida Fish and Wildlife Conservation Commission, personal communication), Alabama (Lauhachinda 1978, Whitaker and Hamilton 1998), Mississippi (Kennedy et al. 1974, Jones and Carter 1989), Louisiana (Lowery 1974, Leopold and Chamberlain 2001, Scognamillo et al. 2003), eastern Texas (Davis and Schmidly 1994, Jackson et al. 1998; J. Young, Texas Parks and Wildlife Department, personal communication), eastern Oklahoma (Caire et al. 1989, Shackleford and Whitaker 1997, White and Hoagland 1997), Arkansas (Tumlison and Karnes 1987, Sealander and Heidt 1990), Kentucky (Raesly 2001; L. Patton, Kentucky Department of Fish and Wildlife Resources, personal communication), and Tennessee (Griess and Anderson 1987, Kennedy 1991; G. Anderson, Tennessee Wildlife Resources Agency, personal communication).

ABUNDANCE STATUS

Reliable census procedures for the river otter have not been developed, and few states have reliable estimates of population numbers. Biologists in Kentucky, Texas, and Virginia note stable to increasing populations (M. Fies, Virginia Department of Game and Inland Fisheries; J. Young, Texas Parks and Wildlife Department; and L. Patton, Kentucky

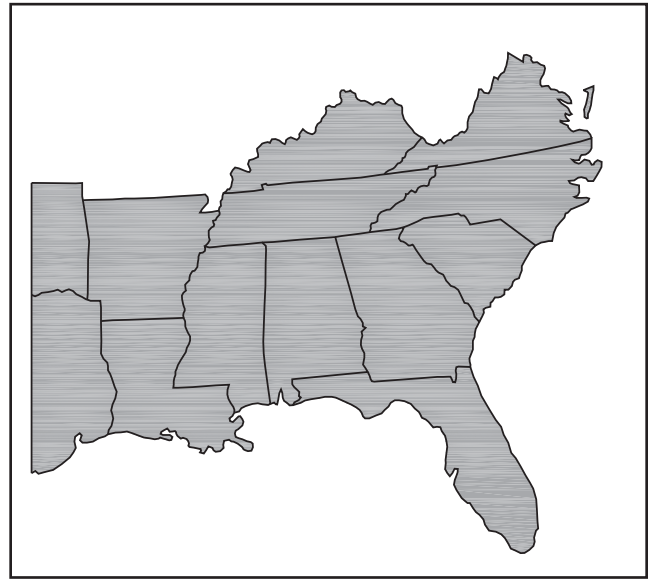


Figure 2. Distribution of *Lontra canadensis* in the South.

Department of Fish and Wildlife Resources, personal communication). Expanding populations are reported in Arkansas, Alabama, Georgia, South Carolina, Oklahoma, and Tennessee (Melquist and Dronkert 1987, Raesly 2001). The species is relatively common statewide in Florida (K. Hodges, Florida Fish and Wildlife Conservation Commission, personal communication), Georgia (J. Ozier, Georgia Department of Wildlife Resources, personal communication), Alabama (M. Sievering, Alabama Department of Conservation and Natural Resources), Mississippi (R. Flynt, Mississippi Department of Wildlife, Fisheries, and Parks), and Arkansas (B. Sasse, Arkansas Game and Fish Commission, personal communication). Leopold and Chamberlain (2001) report stable populations in Louisiana and North Carolina. Average density throughout the river otter range is 1 otter/4 km of waterway (Lariviere and Walton 1998, Whitaker and Hamilton 1998). In Texas, Foy (1984) reported 1 otter/71–106 ha in coastal marshlands, while Shirley et al. (1988) observed 1 otter/86 ha in similar habitat in Louisiana.

PRIMARY HABITATS

The northern river otter uses a variety of aquatic habitats including coastal estuaries, marshes, wetlands, rivers, and streams (Melquist and Dronkert 1987). Individuals are less common in brackish and salt-water areas, except in the tidal zones of large rivers (Brown 1997). In Florida, the otter inhabits mixed hardwood swamps, wet prairies, coastal scrub, sloughs, and canals (Cox and Kautz 2000). Habitat selection is influenced by water quality, availability of forage fish, and suitable denning sites

(Toweill and Tabor 1982). Important habitat components include slow moving water with deep pools, abundant fish, and shoreline vegetation (Chapman and Feldhamer 1982, Allen 1987). American beaver (*Castor canadensis*) ponds on intermittent streams provide habitat conducive to the river otter; lodges, submerged trees, and logjams provide shelter and foraging areas (Rosell et al. 2005). The otter uses dens dug by other animals, as well as natural shelters such as hollow trees, tall marsh grasses, and riverbank thickets (Chapman and Feldhamer 1982). Mech (2003) provided an unusual record of an otter inhabiting a small pond in a golf course within an urban area. Home range size averages between 8–78 km² (Bischof 2003) and is a function of habitat, prey species, and social status (Nowak 1999). The northern river otter may travel >80 km of stream course throughout the year (Cox and Kautz 2000).

REPRODUCTION

The northern river otter is polygamous. The breeding season is December–April with estrus lasting 42–46 days (Lauhachinda 1978, Nowak 1999). Delayed implantation lengthens gestation by 288–375 days (Leopold and Chamberlain 2001). Time of development, when corrected for delayed implantation, ranges between 60–63 days (Humphrey and Zinn 1982). The peak birthing period occurs between March–April (Leopold and Chamberlain 2001). Mean litter size is 2.9 young, with a range of 1–6 (Toweill and Tabor 1982). Neonates are blind, helpless, and covered with fine fur. They are weaned at 3 months; dispersal typically occurs at 12–13 months of age. Sexual maturity is reached at 2 years for females (Melquist and Dronkert 1987). Male otters, although capable of breeding their second year, may not breed until 5–7 years old when they are large enough to defend a territory (Leopold and Chamberlain 2001). Family groups include an adult female and her offspring. Longevity in the wild is estimated at 15 years; some animals have lived 25 years in captivity (Melquist and Dronkert 1987).

FOOD HABITS

The northern river otter feeds primarily on fish and other aquatic vertebrates and invertebrates (Cooley 1983, Melquist and Dronkert 1987). Fish are preyed upon in proportion to their occurrence and density, and in inverse proportion to their swimming ability (Melquist et al. 2003). Slow moving fish such as suckers (*Catostomus* spp.), sunfish (*Lepomis* spp.), carp (*Cyprinus* spp.), shiner (*Notropis* spp.), and bullhead (*Ictalurus* spp.) are common prey (Leopold and Chamberlain 2001). In Alabama and Georgia,

Lauhachinda and Hill (1977) reported 12 families of fish consumed including bass (Centrarchidae), bowfin (*Amia calva*), and perch (*Perca* spp.). Crayfish also comprise a major portion of the diet (Griess 1987). Noordhuis (2002) found that crayfish (*Procambarus* and *Cambarus* spp.) comprised significant portions of the summer and winter diet in Georgia, while Tumblison and Karnes (1987) reported crayfish in 73% of otter scats in Arkansas when water levels were high. Other prey taken include frogs (*Rana* spp.), salamanders (*Amphiuma* and *Ambystoma* spp.), mudpuppy (*Necturus maculosus*), and water snakes (*Nerodia* spp.; Chabreck et al. 1982, Lizotte and Kennedy 1997). Turtles rarely are found as a dietary item (Toweill and Tabor 1982). Although reported infrequently, mammalian prey includes the common muskrat (*Ondatra zibethicus*), American mink (*Mustela vison*), nutria (*Myocastor coypus*), eastern cottontail (*Sylvilagus floridanus*), and white-tailed deer (*Odocoileus virginianus*; Lizotte and Kennedy 1997, Leopold and Chamberlain 2001). Mollusks (e.g., bivalves and aquatic gastropods), aquatic insects, and various waterfowl are consumed opportunistically (Lauhachinda and Hill 1977, Logsdon 1989, Miller 1992, Lizotte 1994). Carnion is rarely consumed (Melquist et al. 2003).

ASSOCIATED SPECIES

The northern river otter inhabits a diversity of aquatic habitats and is associated with numerous mammals in the South, including the long-tailed weasel (*M. frenata*), American mink, northern raccoon (*Procyon lotor*), common muskrat, and nutria. American beaver and river otter territories frequently overlap. The otter benefits from beaver activities that provide den sites and improve fish habitat by impounding streams (Melquist et al. 2003); population levels are correlated for the two species in many locations (Rosell et al. 2005). The American alligator (*Alligator mississippiensis*) and American crocodile (*Crocodylus acutus*) are the only aquatic predators of the river otter (Lariviere and Walton 1998). Terrestrial predators of young otter include the coyote (*Canis latrans*), red fox (*Vulpes vulpes*), bobcat (*Lynx rufus*), and domestic dog (*C. familiaris*; Bischof 2003). In South Carolina, Bergan (1990) observed a bobcat taking an American coot (*Fulica americana*) from a river otter. Competition between the American mink and river otter is minimal due to differential foraging strategies (Leopold and Chamberlain 2001); the former is primarily non-piscivorous, while the otter is piscivorous (M. Ford, U. S. Forest Service, personal communication). The river otter is proficient at capturing fish and may influence non-game fish populations; game fish are not an important part of the diet (Toweill and Tabor 1982).

VULNERABILITY AND THREATS

Otter reintroduction combined with regulatory protection and improved furbearer management have proven successful in restoring populations throughout the South (Serfass et al. 1998). Continued threats include the pollution of aquatic environments (e.g., siltation, pesticide residues), clearing of bottomland forest and riparian habitat, and wetland modification (Sealander and Heidt 1990, Handley 1991, Choate et al. 1994). The river otter is susceptible to aquatic pollution and serves as an indicator of ecosystem health (Lariviere and Walton 1998). Fish and crayfish are known methylmercury vectors; their dominance in the diet has potential for bioaccumulation levels of toxicological concern (Basu et al. 2005, Yates et al. 2005). Exposure to other environmental contaminants, including petroleum products, heavy metals, polychlorinated biphenyls, and aluminum silicates is well documented (Kimber and Kollias 2000). The disappearance of the otter in portions of Tennessee and Kentucky was attributed to increased acid drainage from coal-mining operations (Lauhachinda 1978).

The river otter is susceptible to a variety of diseases including canine distemper, rabies, respiratory tract disease, jaundice, hepatitis, and feline panleucopenia (Davidson and Nettles 1988, Bischof 2003). Based on pathogenicity and prevalence, important endoparasites include several trematodes, nematodes, cestodes, and acanthocephalans (Kimber and Kollias 2000). Ectoparasites are rare and few cause disease (Kimber and Kollias 2000); species that may be most significant for reintroduction programs include the ticks *Ixodes banksi*, *Amblyomma americanum*, and *Dermacentor variabilis*. Other parasites include fleas and sucking lice (Lariviere and Walton 1998).

MANAGEMENT SUGGESTIONS

The otter can cause damage to fish hatchery operations and commercial catfish (*Ictalurus* spp.) farms in the South. The relatively long distances the northern river otter travels and the linear nature of its habitats provide challenges for management. Strict harvest and population monitoring are needed, along with the development of accurate and reliable census procedures. Because the species cannot persist in polluted aquatic systems, management actions to improve or maintain water quality are necessary to ensure viable populations (Allen 1987). The facultative association between the otter and the American beaver suggests that management plans consider the two species in synchrony (Sealander and Heidt 1990, Chillelli et al. 1996). Other management activities include the provision of vegetative cover and natural debris adjacent to wetlands, structural diversity

along shorelines, permanent surface water, and enhanced pool to riffle ratios (Allen 1987). The conservation of wetlands and riparian areas are critical to the future success of the northern river otter (Melquist et al. 2003).

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Fisher

Martes pennanti (Erxleben, 1777)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The fisher (*Martes pennanti*) is the largest member of its genus (Anderson 1970). Goldman (1935) described three subspecies, but the validity of these taxa was disputed (Hagmeier 1959, Coulter 1966). Nevertheless, Hall (1981) and Powell (1981) continue to recognize these subspecies. Only one subspecies, *M. p. pennanti*, occurs in the South. The life history of the fisher is reviewed by Powell (1981, 1982), Strickland et al. (1982), and Douglas and Strickland (1987). A Habitat Suitability Index model for the fisher was developed by Allen (1983) and tested by Thomasma et al. (1991).

DISTINGUISHING CHARACTERISTICS

The fisher is a fox-sized mammal with an elongate, stocky body. Measurements are: total length, 800–1050 mm; tail, 280–400 mm; hind foot, 90–130 mm; ear, 40–46 mm; weight, 1.8–5.4 kg. Males are larger than females. The dorsal pelage is long and usually black. The fur on the face, neck, and shoulders is short and burnished with gold or silver. The ventral pelage is black, but there are patches of white on the forelegs and genital area. The fisher has a long, bushy tail and rounded ears. Its feet have retractable claws, but the claws are not in sheaths as in cats. The skull is long and tapers only slightly from the cranium to the rostrum. The fisher may be confused with one other species in the region, the mink (*Mustela vison*). The mink is approximately two-thirds the size of a fisher and has three premolars rather than four on both upper and lower jaws. The dental formula is: I 3/3, C 1/1, P 4/4, M 1/2 = 38 (Figure 1). See keys for details.

CONSERVATION STATUS

The fisher has a global rank of Secure (NatureServe 2007). It is considered Critically Imperiled in Virginia and Tennessee and Presumed Extirpated in North Carolina.

DISTRIBUTION

Prior to 1900, the fisher was distributed widely across Canada and the northern United States, extending southward to the mountains of California and the Appalachians (Choate et al. 1994). The fisher was known from Virginia (Audubon and Bachman 1846, Bailey 1946, Handley and Patton 1947, Handley 1991,

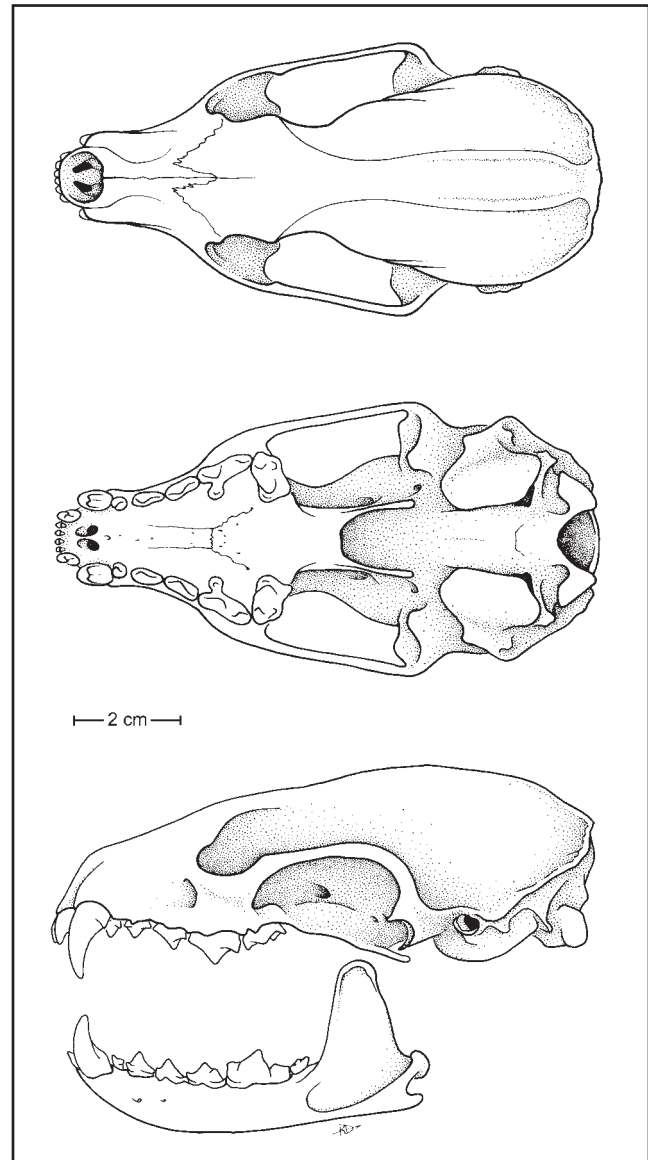


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Martes pennanti* from Oswego County, New York (USNM 188238, gender unknown).

1992), North Carolina (Miller and Kellogg 1955), Georgia (Parmalee 1960), and Tennessee (Audubon and Bachman 1846). Fisher populations were extirpated or greatly reduced over much of the original range between 1800 and 1940 as a result of overtrapping and deforestation (Irvine et al. 1964, Brander and Books 1973).

Fishers are appearing along the eastern Virginia border in Augusta, Bath, Highland, and Rockingham counties (Virginia Game and Inland Fisheries 2006); there are also reports of observations in surrounding counties. These animals are in contact with a known population that originated from a restocking program in West Virginia (Handley 1991, 1992; Linzey 1998) that was initiated in 1969 (Pack and Cromer 1981, Berg 1982).

Reintroduction of the fisher began during 2001–2002 in the Catoosa Wildlife Management Area of Tennessee (Tennessee Wildlife Resources Commission 2003). The animals were captured from the Chequamegon National Forest and surrounding lands in Wisconsin (Copas 2004). Forty animals were released over a two-year period (B. Anderson, Tennessee Wildlife Resources Agency, personal communication) into the Cumberland, Morgan, and Fentress counties (Figure 2). The current status is unclear; the population is being monitored to determine feasibility of continuing reintroduction efforts (Tennessee Wildlife Resources Commission 2006).

ABUNDANCE STATUS

Elsewhere in its distributional range, the fisher is regarded as common. Increasing harvests of the fisher have been recorded during the past three decades in northern North America (Obbard et al. 1987). In the South, however, the fisher is extremely rare.

PRIMARY HABITATS

The fisher prefers mixed forests with extensive, dense canopies (DeVos 1951, Clem 1977, Kelly 1977). Forests with little or no canopy closure are avoided (Clem 1977, Coulter 1966, Kelly 1977, Powell 1978). Many forest types are used including red spruce–eastern hemlock (*Picea rubens*–*Tsuga canadensis*), mature conifers, mixed conifer–deciduous, northern hardwoods, and forested wetlands with conifer undergrowth (DeVos 1951, Coulter 1966, Kelly 1977, Allen 1983). During winter, *M. pennanti* prefers coniferous ridges (Raine 1983). The fisher uses temporary sleeping sites that may be located in hollow trees, logs and stumps, brush piles, rocks, abandoned beaver lodges, holes, and snow dens (Hamilton and Cook 1955, Eadie and Hamilton 1958, Coulter 1966). The fisher sometimes moves through the tops of trees, but DeVos (1951) and Powell (1980) suggest such arboreal activities are unusual.

REPRODUCTION

The breeding season is from March to May (Eadie and Hamilton 1958, Coulter 1966); the fisher exhibits

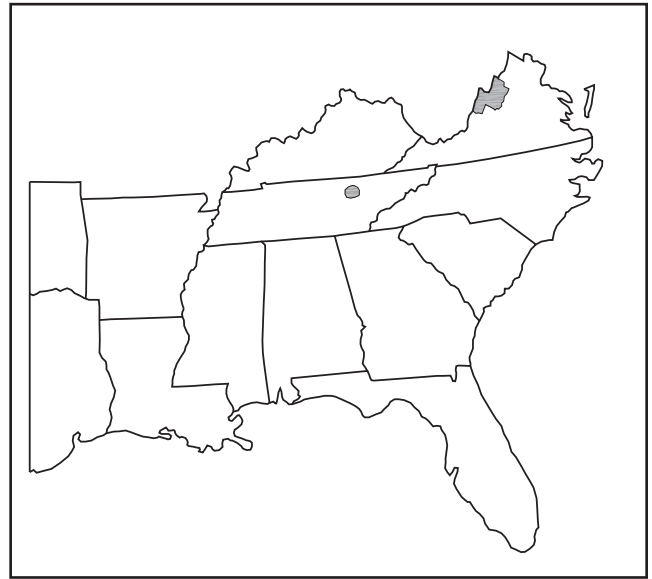


Figure 2. Distribution of *Martes pennanti* in the South.

delayed implantation (Enders and Pearson 1943). Implantation occurs from January to early April the following year (Ewer 1973). The period of active pregnancy is unknown but is estimated to be from 30 days (Coulter 1966) to 60 days (Hamilton and Cook 1955). The fisher produces 1 litter/year and the litter size varies from 1–6 with a mean of 2.8 (Hall 1942, Hamilton and Cook 1955, Coulter 1966). Maternity dens are located in hollow trees (Hamilton and Cook 1955, Coulter 1966, Arthur and Krohn 1991). Young are born naked, blind, and helpless, and become mobile at about eight weeks of age. The young remain with the female until they disperse in early fall (Coulter 1966). Both males and females may become reproductively mature at 1 year of age (Eadie and Hamilton 1958, Wright and Coulter 1967).

FOOD HABITS

Although it usually preys on small mammals, the fisher is an opportunistic forager that consumes carrion, fruits, nuts, and berries (Coulter 1966, Kelly 1977, Douglas and Strickland 1987). Many researchers report that the snowshoe hare (*Lepus americanus*) is the primary prey of the fisher (Coulter 1966, Bulmer 1975, Powell 1982). However, Kelly (1977) reported that mice (*Peromyscus* spp.) and squirrels (*Tamiasciurus hudsonicus* and *Sciurus* spp.) were the primary prey in New Hampshire. Shrews (*Blarina* and *Sorex* spp.), voles (*Microtus* spp. and *Clethrionomys gapperi*), and birds also are important components of fall and winter diets. Bird eggs, reptiles, amphibians, and insects are eaten in spring and summer (DeVos 1951, Hamilton and Cook 1955, Coulter 1966, Clem 1977, Brown and Will 1979, Raine 1987, Arthur et al. 1989, Giuliano et al. 1989, Kuehn 1989).

ASSOCIATED SPECIES

The fisher uses the same habitats as its prey species. The species may occur in the same habitats as the long-tailed weasel (*M. frenata*), least weasel (*M. nivalis*), northern flying squirrel (*Glaucomys sabrinus*) and snowshoe hare.

VULNERABILITY AND THREATS

Extensive timber harvest and thinning of closed forest canopies are forest management practices that may be detrimental to the fisher (Handley 1991). Virginia is at the extreme southern limit of the fisher's geographical range; the species may persist if individuals from reintroduced populations in West Virginia continue to move southward and eastward. The species is vulnerable to over-trapping. The long-term status in Tennessee remains unclear.

MANAGEMENT SUGGESTIONS

The fisher is capable of moving long distances, but movement may be restricted in landscapes with large, nonforested openings. Reducing the size of openings may facilitate contact between subpopulations and the recolonization of unoccupied habitat. The creation of small forest openings also enhances vegetative diversity and prey abundance. The retention of down woody debris, cavity trees, and dense patches of understory cover would benefit the fisher. Isolated populations are of special concern and require monitoring. Additional research needs include investigation of dispersal capabilities and characterization of habitat and geographic features that facilitate or inhibit movement.

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Long-tailed Weasel

Mustela frenata (Lichtenstein, 1831)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Before the revision of the weasels by Hall (1951), considerable confusion in taxonomy and nomenclature existed. The names of European weasels (e.g., *Putorius*) were applied to many North American forms. Most of the subspecies of the long-tailed weasel (*Mustela frenata*) previously were classified as distinct species. Worldwide, there are 51 recognized subspecies of *M. frenata* (Wozencraft 1993). Only five of the 35 subspecies that occur in North America occur in the South (Hall 1981). These include: *M. f. noveboracensis* in the north-central part of the region; *M. f. olivacea* in the south-central area of the region; *M. f. peninsulae* in peninsular Florida; *M. f. arthuri* in southern Louisiana and southeastern Texas; and *M. f. primulina* in northeastern Texas, eastern Oklahoma, and Arkansas. Svendsen (1982) and Fagerstone (1987) review the life history of the species.

DISTINGUISHING CHARACTERISTICS

The long-tailed weasel is the largest of the North American weasels. Measurements are: total length, 285–430 mm; tail, 85–140 mm; hind foot, 30–50 mm; ear, 18–25 mm; weight, 285–430 g. Males are larger than females. The long-tailed weasel possesses a characteristically long and slender body form with short legs and the longest tail of the North American weasels. The dorsal pelage is brown and the tail has a black tip. Although the more northern forms of this species molt into a white winter pelage, long-tailed weasels in the South do not. The dorsal pelage may get longer, denser, and lighter in color during winter, however. The ventral pelage remains white, yellowish, or buff-colored all year. In some areas, long-tailed weasels have white or yellow mask-like facial markings. The skull is long and narrow with an elongated braincase and a short rostrum. Skull length can be used to distinguish the skull of the long-tailed weasel (less than 55 mm, but greater than 30 mm) from the American mink (*M. vison*; greater than 55 mm), and the least weasel (*M. nivalis*; less than 30 mm). In all weasels, the last upper molar is shaped like a dumbbell. The dental formula of the long-tailed weasel is: I 3/3, C 1/1, P 3/3, M 1/2 = 34 (Figure 1). See keys for details.

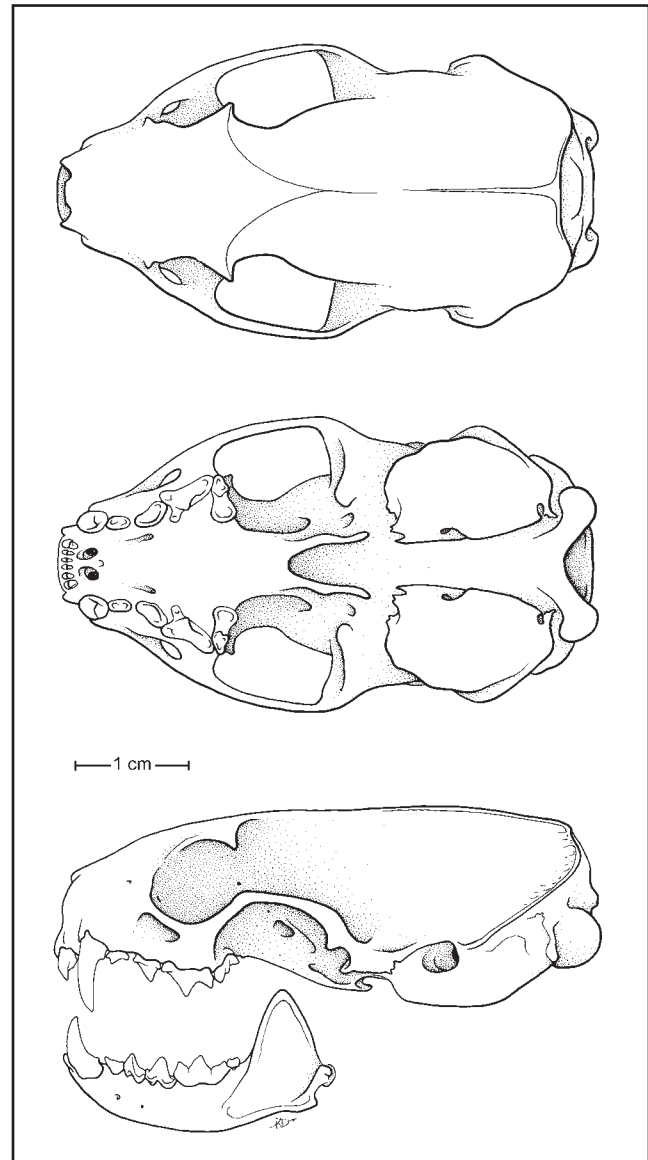


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Mustela frenata* from Delaware County, Delaware (USNM 253934, male).

CONSERVATION STATUS

The long-tailed weasel has a global rank of Secure (NatureServe 2007). It is Secure in Florida, Georgia, Tennessee, Texas, and Virginia. It is considered Apparently Secure in Kentucky. Alabama, North Carolina and South Carolina classify the species as Vulnerable. Arkansas and Louisiana both consider

it Imperiled, and in Oklahoma it is Critically Imperiled. It is unranked in Mississippi. The Florida Fish and Wildlife Conservation Commission consider *M. f. peninsulæ* and *M. f. olivacea* protected furbearers.

DISTRIBUTION

Mustela frenata is distributed from southern Canada into northern South America but is absent from arid zones of the southwestern United States and northern Mexico (Hall 1981, Choate et al. 1994). The distribution of the long-tailed weasel in the South is depicted in Figure 2. The weasel occurs throughout Virginia (Bailey 1946, Handley and Patton 1947, Webster et al. 1985, Handley 1979, 1992; Linzey 1998), North Carolina (Calhoun 1941, Conaway and Howell 1953, Johnston 1967, Linzey and Linzey 1971, Lee et al. 1982, Clark et al. 1985, Webster et al. 1985, Linzey 1995), South Carolina (Burnett 1851, Sherman 1937, Penny 1950, Golley 1966, Webster et al. 1985, Cothran et al. 1991), Georgia (Golley 1962, Laerm et al. 1980, 1981) and all but southern most Florida (Moore 1944, Humphrey 1992, Whitaker and Hamilton 1998, Wilson and Ruff 1999). The long-tailed weasel occurs throughout Alabama (Howell 1913, 1921; Holliman 1963) and Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989), and is absent from the low coastal areas of southern Louisiana (Lowery 1974). The species is known from many localities in eastern Texas (Baker 1944, McCarley 1959, Schmidly 1983, Davis and Schmidly 1994). There are no collection records from eastern Oklahoma although it is presumed to occur there (Caire et al. 1989, White and Hoagland 1997). The weasel occurs throughout Arkansas (Sealander and Heidt 1990), Tennessee (Kellogg 1939, Howell and Conaway 1952, Goodpaster and Hoffmeister 1952, Conaway and Howell 1953, Linzey and Linzey 1971, Smith et al. 1974, Kennedy 1991, Linzey 1995), and Kentucky (Barbour 1951, Barbour and Davis 1974).

ABUNDANCE STATUS

Data on the abundance of the long-tailed weasel in the South is lacking. Populations are considered locally variable throughout the South and often rare in many areas (Leopold and Chamberlain 2001). Because of its secretive habits, the long-tailed weasel may be more common in some areas than suspected (Sealander and Heidt 1990). The population density of the long-tailed weasel was 2–18/km² in Kentucky (DeVan 1982). In Iowa, population densities on agricultural lands were estimated to be from 1/ha to 1/5.7 ha (Polderboer et al. 1941). The long-tailed weasel does not hibernate and is active year-round.



Figure 2. Distribution of *Mustela frenata* in the South: (1) *M. f. arthuri*; (2) *M. f. noveboracensis*; (3) *M. f. olivacea*; (4) *M. f. peninsulæ*; (5) *M. f. primulina*.

PRIMARY HABITATS

The long-tailed weasel is known from most habitat types within the South. The species occurs in all forest types, seral stages, and varied elevations. The weasel inhabits brushland, open woodlands, field edges, riparian grasslands, swamps, and marshes. In Florida, they inhabit sand pine (*Pinus clausa*) scrub, sandhills, pine flatwoods, cypress (*Taxodium* spp.) swamps, and tropical hammocks (Humphrey 1992). Dens may be located in a rock crevice, brushpile, or an abandoned burrow (Polderboer et al. 1941, Quick 1951, Fitzgerald 1977, Frank and Lips 1989). It readily occupies barns and other structures frequented by humans (Lowery 1974, Sealander and Heidt 1990).

REPRODUCTION

Breeding occurs in July or August (Svendsen 1982). After fertilization, the embryo develops for 8 days and then ceases development for the following 7.5 months. Implantation is delayed until the following spring (Wright 1948a). After implantation, the gestation period is 23–27 days (Wright 1947). A litter of 4–9 young is born in April or May. The mean litter size is 6.8 (Heidt 1970). The newborn is blind and covered with a fine, thin, white fur that is replaced by adult fur in 3–4 weeks (Hamilton 1933). After six to seven weeks, the young begin to accompany their mother while hunting (Sanderson 1949); within 12 weeks the young disperse (Wright 1947). Juvenile females may mate in their first summer, but juvenile males do not breed until they are 15 months old.

(Wright 1948b). The adult female enters estrus 65–104 days after giving birth and may remain in estrus for several months (Wright 1948b). Adult males are sexually active from April to August (Wright 1947). Males and females frequently are observed together during the nonbreeding season (Hamilton 1933, Quick 1951). Hamilton (1939) speculated that mated weasels remain together for long periods, possibly for life. The longevity of the long-tailed weasel is not known (Fagerstone 1987).

FOOD HABITS

Mustela frenata is a generalist, preying upon a wide variety of small vertebrates. The primary prey include voles (*Microtus* spp. and *Clethrionomys gapperi*), jumping mice (*Napaeozapus insignis* and *Zapus hudsonius*), harvest mice (*Reithrodontomys* spp.), rabbits (*Sylvilagus* spp.), and eastern chipmunk (*Tamias striatus*; Hamilton 1933, Errington 1936, Polderboer et al. 1941, Quick 1951, Simms 1979). Occasionally, the weasel consumes bats, birds and their eggs, snakes, frogs, and insects (Errington 1936, Quick 1951, Mumford 1969, Simms 1979). The average daily food requirement for the weasel is approximately 1.5 voles per day (Powell 1973). The long-tailed weasel sometimes kills more food than it can eat, and it caches the extra food in burrows for later consumption (Fagerstone 1987).

ASSOCIATED SPECIES

Because of its wide distribution over several habitats, the long-tailed weasel is found in association with numerous species in the region. The weasel is preyed upon by the red fox (*Vulpes vulpes*), gray fox (*Urocyon cinereoargenteus*), coyote (*Canis latrans*), bobcat (*Lynx rufus*), barred owl (*Strix varia*), feral dog (*Canis familiaris*), and several snakes (Whitaker and Hamilton 1998, Leopold and Chamberlain 2001). The species is sympatric with other mustelids including the fisher (*Martes pennanti*), least weasel, and American mink.

VULNERABILITY AND THREATS

Canine distemper and tularemia can affect the long-tailed weasel. The reproductive potential of the weasel is limited, and the species could be negatively impacted by high harvest or trapping pressure (Leopold and Chamberlain 2001). Indirect effects associated with habitat loss and modification are likely to affect the species (Humphrey 1992).

MANAGEMENT SUGGESTIONS

The maintenance of a mosaic of vegetation types can benefit the weasel by supporting the prey populations

that comprise the diet of this species. The long-tailed weasel is associated with early to mid-successional vegetation and the ecotones between these communities. Management that maintains fencerows, shelterbelts, and riparian vegetation is beneficial (Allen 1987). In addition, the protection of preferred foraging areas adjacent to water sources is beneficial (Dickson 2001). The lack of basic knowledge on weasel ecology in the South limits the ability to effectively manage this species through harvest and habitat management (Leopold and Chamberlain 2001).

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Least Weasel

Mustela nivalis (Linnaeus, 1766)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The least weasel (*Mustela nivalis*) is a widespread species with four subspecies in North America (Hall 1981, Sheffield and King 1994). Only one subspecies, *M. nivalis allegheniensis*, occurs in the South. The life history of the species is reviewed by Svendsen (1982), Fagerstone (1987), and Sheffield and King (1994).

DISTINGUISHING CHARACTERISTICS

The least weasel is the smallest carnivore in North America. Measurements are: total length, 180–215 mm; tail, 25–45 mm; hind foot, 20–28 mm; ear, 10–15 mm; weight, 30–65 g. Males are approximately twice the size of females. During summer, the dorsal pelage is brown; during winter, the pelage is white at high altitudes or mottled with white at lower elevations in the region. The ventral pelage is white all year. The tail is short and lacks a black tip. The skull is long and narrow with a long braincase and a short rostrum. The width of the palate, which extends beyond the upper molars, is approximately equal to the distance between the tympanic bullae (Sheffield and King 1994). The dental formula of the least weasel is: I 3/3, C 1/1, P 3/3, M 1/2 = 34 (Figure 1). The small size, length of tail, and absence of a black tip on the tail distinguish the least weasel from the long-tailed weasel (*M. frenata*). See keys for details.

CONSERVATION STATUS

The least weasel has a global rank of Secure (NatureServe 2007). However, it is considered Vulnerable in Virginia and Imperiled in Kentucky, North Carolina, and Tennessee. It is Critically Imperiled in Georgia. It is unranked in South Carolina.

DISTRIBUTION

The least weasel is found throughout northern Eurasia including Japan, and much of northern North America (Allen 1933, Sheffield and King 1994). In North America, the species occurs from Alaska and northern Canada southward in the central United States and in the eastern United States to the southern Appalachians (Hall 1981, Choate et al. 1994). The distribution of the least weasel in the South is depicted in Figure 2. The weasel occurs in the mountains and northern Piedmont of Virginia (Llewellyn 1942, Bailey 1946,

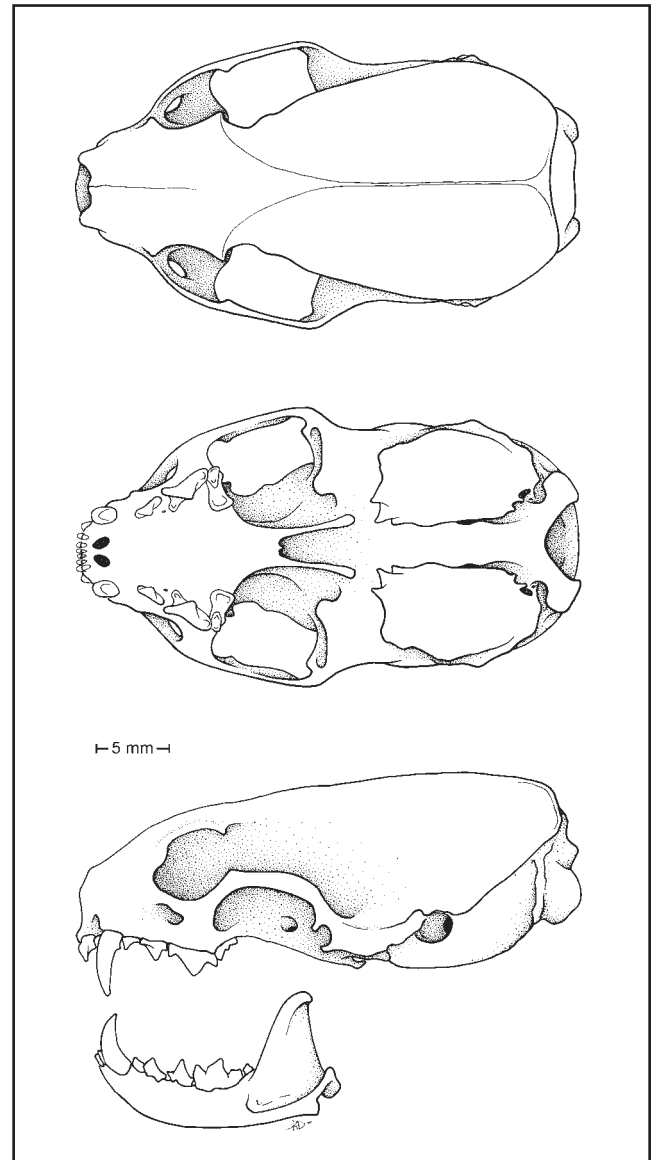


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Mustela nivalis* from Newton County, Indiana (USNM 319834, male).

Handley and Patton 1947, Handley 1949, 1991, 1992; Webster et al. 1985, Linzey 1998). A recent record from the Coastal Plain indicates that the species may be more widely distributed in Virginia than previously believed (Bellows et al. 1999). In North Carolina, all records of the species are from the Southern Appalachian Mountains above 500 m in elevation

(Church 1925, Stupka 1960, Edwards 1963, Barkalow 1967, Linzey and Linzey 1971, Cooper et al. 1977, Lee et al. 1982, Webster et al. 1985, Linzey 1995). The species is reported in Tennessee from a few locations on the eastern Cumberland Plateau and in the Blue Ridge Mountains in the northeastern corner of the state (Tuttle 1968, Nagel 1972, Smith et al. 1974, Kennedy and Harvey 1980, Anderson 1988, Copeland and Caldwell 1991, Cushing and Knight 1991). The least weasel is known from central and eastern Kentucky (Davis and Barbour 1979, Prather 1984, David 1988, Meade 1992). A specimen recently collected in the southern Appalachians of Georgia represents the southern limit of the known range (J. M. Wentworth, U. S. Forest Service, personal communication). Although it has not been recorded from South Carolina, the least weasel has been collected 16 km from the South Carolina border (Barkalow 1967) and may occur in the southern Appalachians in that state (Webster et al. 1985).

ABUNDANCE STATUS

Population densities of the least weasel in the South are unknown. Although the species is considered rare in North America (Hall 1951), the least weasel may be locally common in the Appalachians when populations of the meadow vole (*Microtus pennsylvanicus*) or other rodents are high (Handley 1991). Local populations are vulnerable to changes in prey density. When rodent populations decline, least weasels may disappear. They can recolonize quickly once prey populations increase again (King 1983). There is no published information on home range size in the South. Outside of the region home ranges of least weasels range from 0.8–15 ha (Leopold and Chamberlain 2001).

PRIMARY HABITATS

The least weasel may occupy many habitats, and commonly occurs in open areas associated with water. It is found in damp meadows, grasslands, marshes, and river bottoms (Soper 1946, Gunderson and Beer 1953, Nagel 1972, Chapman and Harman 1977). Specimens from Virginia and West Virginia were collected from pastures, brushy fence rows, oldfields with dense vegetation, in and around buildings, and deep within oak-hickory-eastern hemlock (*Quercus-Carya-Tsuga canadensis*; Bailey 1946, Handley 1991). The least weasel often uses abandoned dens (Criddle 1947). Nests inside the dens are improved by the addition of bits of fur plucked from the prey, skulls, and prey carcasses (Criddle 1947, MacLean et al. 1974). The entrance to an active den, which may be located in burrows or rock piles, is usually marked by fecal deposits (Polderboer 1942). The least weasel is active

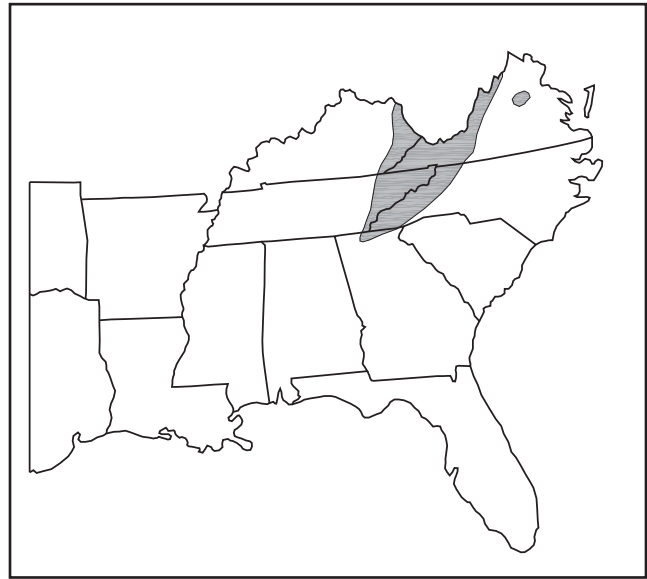


Figure 2. Distribution of *Mustela nivalis* in the South.

all year and snow does not impede its activity patterns (Simms 1979).

REPRODUCTION

When food is abundant, the least weasel is capable of breeding year-round (Hall 1951). A female can produce 2–3 litters/year and the testes of the male, normally active only from March to August, are active all year under optimum nutritional conditions (Heidt et al. 1968). Unlike other weasels, the species does not exhibit delayed implantation (Heidt 1970). The gestation period is 34–37 days and the litter size ranges from 1–6 with a mean of 4.8 (Heidt 1970). The young are born naked, blind, and deaf. They are weaned within 56 days and reach adult weight within 15 weeks (Heidt et al. 1968, Heidt 1970). Females reach sexual maturity at 3 months of age (Deanesly 1944, King 1980). The life expectancy for the least weasel is less than 1 year, but some individuals survive up to 3 years (King 1980). In captivity, some have lived for 7 years (Hill 1939).

FOOD HABITS

The least weasel specializes in small prey, primarily rodents (Fagerstone 1987). In North America, voles (*Microtus* and *Clethrionomys* spp.), mice (*Peromyscus* spp.), harvest mice (*Reithrodontomys* spp.), and the hispid cotton rat (*Sigmodon hispidus*) are the primary prey (Polderboer 1942, Criddle 1947, Hall 1951). When prey is scarce, the least weasel may take shrews (*Sorex* and *Blarina* spp.), bird eggs, insects, lizards, salamanders, fish, worms, and carrion (Hall 1951, Tapper 1979, Sheffield and King 1994). Least

weasel nests often contain caches of prey carcasses (Bishop 1923, Winecoff 1930, Polderboer 1942, MacLean et al. 1974). Least weasels cannot eat more than a few grams of food at a time, and they eat 9–10 times/day (Gillingham 1984). The caching behavior provides a ready source of food. The least weasel hunts by random searches, and Heidt (1972) describes its searching and killing behavior.

ASSOCIATED SPECIES

The least weasel occupies the same habitats as its prey species. In the southern Appalachians, the geographical range of the species overlaps with the long-tailed weasel and American mink (*M. vison*).

VULNERABILITY AND THREATS

Several roundworms, biting lice, mites, and ticks affect the least weasel. Hawks and owls are important predators of this species; predation can be heavy at times (Whitaker and Hamilton 1998). The least weasel has historically not been very abundant throughout the South, and thus trapping has not seriously impacted its abundance (Leopold and Chamberlain 2001).

MANAGEMENT SUGGESTIONS

The maintenance of a diversity of understory vegetation can enhance least weasel habitat by supporting the prey populations that comprise the diet of this species. The maintenance of fencerows and shelterbelts for cover and travel corridors are also beneficial where agricultural land use is high (Allen 1987). Little is known about least weasel ecology, limiting sound management throughout its southern range. However, least weasel populations often track local rodent populations.

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American Mink

Mustela vison (Schreber, 1777)

Margaret K. Trani and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The American mink (*Mustela vison*) has 15 recognized subspecies (Hall 1981). Six subspecies occur in the South: *M. v. evergladensis*, occurring in the Everglades and Big Cypress Swamp of southern Florida; *M. v. halilimnetes*, inhabiting coastal areas along the Gulf coast of northwestern Florida from Pasco County north to Ochlockonee Bay, Franklin County; *M. v. vison*, occurring in the Appalachian Mountains and northwards to Quebec; *M. v. mink*, the most widespread subspecies in the region; *M. v. lutensis*, found along the Atlantic coastal zone of extreme northeastern Florida, Georgia, and southern South Carolina; and *M. v. vulgivaga*, found in western Mississippi, Louisiana and central Arkansas. There is disagreement, however, on the taxonomy of these six subspecies. Humphrey and Setzer (1989) consider *M. v. evergladensis* as a disjunct population of *M. v. mink*, and suggest that the distribution of the latter contracted since the 1930s by the loss of the northern Everglades. The Florida Fish and Wildlife Conservation Commission follows this convention (Cox and Kautz 2000). In contrast, Whitaker and Hamilton (1998) and Wilson and Ruff (1999) recognize *M. v. evergladensis* as a separate subspecies. *M. v. halilimnetes* was described as a new subspecies by Humphrey and Setzer (1989) and is recognized by state Natural Heritage agencies (NatureServe 2007) and Whitaker and Hamilton (1998). However, Wilson and Ruff (1999) and Cox and Kautz (2000) do not recognize *M. v. halilimnetes*; the latter considers this as a disjunct population of *M. v. lutensis*. Linscombe et al. (1982), Eagle and Whitman (1987), and Lariviere (1999) describe the life history of the mink. A Habitat Suitability Index Model for the species was developed by Allen (1984) and tested by Loukmas and Halbrook (2001).

DISTINGUISHING CHARACTERISTICS

The American mink is a small, elongate mammal with short legs. Measurements are: total length, 510–580 mm; tail, 135–210 mm; hind foot, 50–75 mm; ear, 19–27 mm; weight, 0.7–1.6 kg. Males average 10% larger than females. The pelage is soft, lustrous, and composed of thick underfur with long, shiny guard hairs. The color varies from dark brown to almost black, but the ventral surface may be paler. Some animals have white patches on the chin, chest,

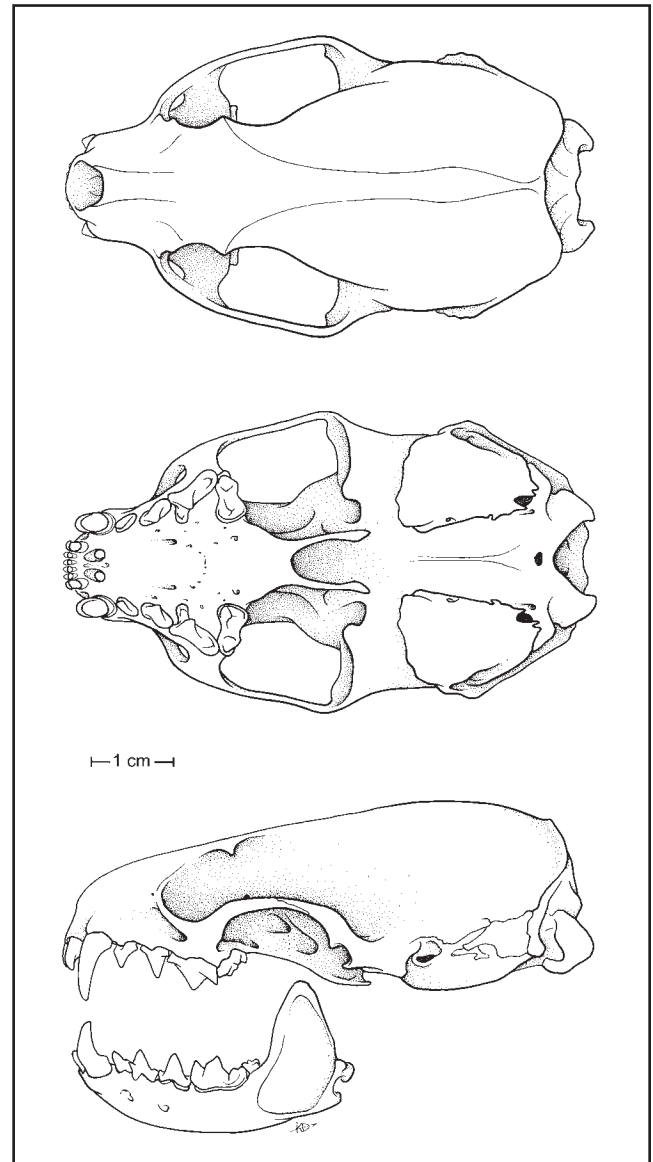


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Mustela vison* from, Godbout, Quebec, Canada (USNM 188249, female).

and abdomen. The thickly furred tail is darker toward the tip. The feet are fully furred except for the pads, and the toes are semi-webbed. The skull is narrow with a long braincase and a short rostrum. The American mink skull can be distinguished from other mustelids on the basis of size. The mink skull exceeds 55 mm in length whereas the skull of the long-tailed

weasel (*M. frenata*) and the least weasel (*M. nivalis*) are less than 55 mm long. The dental formula is: I 3/3, C 1/1, P 3/3, M 1/2 = 34 (Figure 1). See keys for details.

CONSERVATION STATUS

The American mink has a global rank of Secure (NatureServe 2007). It is also considered Secure in Alabama, Florida, Georgia, Kentucky, Louisiana, Mississippi, Tennessee, and Virginia. It is Apparently Secure in Arkansas, North Carolina, Oklahoma, and Texas. It is unranked in South Carolina. Two subspecies, *M. v. evergladensis* and *M. v. lutensis*, are of special concern in Florida.

DISTRIBUTION

Mustela vison is found throughout northern North America except for the extreme northern part of central Canada. The southern limits of its range are in the southwestern United States from northern California to the central Texas coast (Hall 1981, Choate et al. 1994). The distribution of the mink in the South is depicted in Figure 2. The species occurs throughout Virginia (Bailey 1946, Handley and Patton 1947, Webster et al. 1985, Handley 1992, Linzey 1998), North Carolina (Conaway and Howell 1953, Lee et al. 1982, Clark et al. 1985, Webster et al. 1985, Linzey 1995), and Georgia (Golley 1962, Laerm et al. 1981). The mink inhabits most of South Carolina (Penny 1950, Golley 1966, Cothran et al. 1991); however, recent survey data indicate mink are essentially absent in the northern coastal marshes (South Carolina Department of Natural Resources 2005). The species occurs in the coastal zones of northern peninsular Florida as well as the Everglades and Big Cypress Swamp (Allen and Neill 1952, Humphrey and Zinn 1982, Humphrey 1992, Cox and Kautz 2000). A sighting along the Suwannee River in Levy County suggests the mink may occur inland, but Humphrey (1992) and Cox and Kautz (2000) note a general absence of the species from the wetlands of interior Florida. The mink occurs throughout Alabama (Howell 1921, Holliman 1963), Mississippi (Wolfe 1971, Kennedy et al. 1974, Jones and Carter 1989), Louisiana (Svilha 1931, Lowery 1974, Leopold and Chamberlain 2001), eastern Texas (Schmidly 1983, Davis and Schmidly 1994), eastern Oklahoma (Caire et al. 1989), Arkansas (Smith and McDaniel 1982, Sealander and Heidt 1990), Tennessee (Goodpaster and Hoffmeister 1952, Conaway and Howell 1953, Smith et al. 1974, Kennedy 1991, Linzey 1995), and Kentucky (Barbour 1951, Barbour and Davis 1974).



Figure 2. Distribution of *Mustela vison* in the South: (1) *M. v. evergladensis*; (2) *M. v. halilimnetes*; (3) *M. v. lutensis*; (4) *M. v. mink*; (5) *M. v. vison*; (6) *M. v. vulgivaga*.

ABUNDANCE STATUS

Densities of the American mink throughout the South are poorly understood; their secretive habits contribute to the paucity of population data. Population densities vary with the permanence of aquatic habitat, climate, and intraspecific interaction (Linscombe et al. 1982). Trapping returns often provide indices to relative abundance. The greatest mink harvests occur in Louisiana and Arkansas. Leopold and Chamberlain (2001) report that during the 1991–1992 trapping season, these states comprised 37% and 21% respectively, of the total mink harvest in the region. In Louisiana, reported mink densities (1 mink/2–4 ha) were highest in swamps whereas freshwater marshes had lower mink densities (Leopold and Chamberlain 2001). Linscombe et al. (1982) reported 1 mink/10–12 ha in cypress (*Taxodium* spp.) and tupelo (*Nyssa* spp.) swamps. For several states including Florida (Cox and Kautz 2000), there are no estimates for the density of mink populations. Female home ranges average 8 ha (Layne 1978), while male home ranges can exceed 769 ha (Schwartz and Schwartz 1981). In Tennessee, male mink ranged from 6–11 km along streams in the winter (Stevens et al. 1997).

PRIMARY HABITATS

The American mink requires wetland habitats such as bottomland swamps, riverbanks, and streams. The species is also found near riparian areas, lakeside

zones, and coastal marshes (Eagle and Whitman 1987). Habitat use varies by geographic area and season. In the Everglades, the mink retreats from drying marshlands to long hydroperiod swamp forests as the dry season progresses (Cox and Kautz 2000). During wet seasons, salt marshes found between mangroves and freshwater habitats are used. Permanence of water and emergent shoreline vegetation are important factors for evaluating wetland habitat suitability for mink (Allen 1984). Racey and Euler (1983) determined that mink activity decreased along a shoreline in Ontario after trees and emergent vegetation were removed. Cypress-tupelo swamps provide quality habitat in Louisiana (Linscombe et al. 1982). The mink often dens in streambank burrows constructed by common muskrats (*Ondatra zibethicus*) and other animals (Yeager 1943, Errington 1961, Schladweiler and Storm 1969). The species also uses tree cavities, brush piles, and abandoned American beaver (*Castor canadensis*) lodges as den sites (Grinnell et al. 1937, Eagle and Whitman 1987).

REPRODUCTION

Onset of the breeding season is related to photoperiod (Hammond 1951, Duby and Travis 1972). Breeding begins in late January in Louisiana and may continue through March in the South (Svilha 1931, Enders 1952, Sealander and Heidt 1990). The breeding season lasts 3 weeks in most localities; females are receptive at 7–10 day intervals during that period (Enders 1952). Ovulation is induced by mating and occurs within 33–72 hours after copulation (Venge 1959). Delayed implantation may occur early in the breeding season, but does not occur in females that are fertilized late in the season (Mead 1981). Consequently, the average period from mating to birth is 51 days, with a range of 40–79 days (Enders 1952, Mead 1981). The gestation period (after implantation of the embryo) may last 28–30 days (Enders 1952). Births occur from April to May (Svilha 1931). The litter size varies from 1–8 young (Hansson 1947, Enders 1952). The young are born naked and blind, and grow rapidly and reach adult weight by autumn. Females reach sexual maturity the following spring and may reproduce once a year in successive years (Enders 1952).

FOOD HABITS

Mink characteristically have diets in which animal material exceeds 95 percent. Muskrats (*O. zibethicus* and *Neofiber alleni*), mice (*Peromyscus* spp.), and lagomorphs (*Sylvilagus* spp.) are preferred prey (Goodpaster and Hoffmeister 1950, Smith and McDaniel 1982). Hispid cotton rats (*Sigmodon hispidus*), often are prey of the mink in the South (Leopold and

Chamberlain 2001). The mink diet also includes ground-nesting birds, frogs, reptiles, crayfish, fish, and mollusks (Hamilton 1936, Sealander 1943, Korschgen 1958, Gerell 1967, Cowan and Reilly 1973). During winter, fewer birds and more fish are taken (Sealander 1943, Gerell 1967, Smith and McDaniel 1982, Casson and Klimstra 1983). Caching of prey is common during all seasons (Svilha 1931, Yeager 1943). The amount of consumed insects, fruits, and seeds varies with seasonal availability.

ASSOCIATED SPECIES

The American mink is distributed widely in the region and may be found in association with several wetland mammals. It is often found in association with the American beaver, common muskrat, nutria (*Myocastor coypus*), and northern river otter (*Lontra canadensis*). The interaction between the mink and other terrestrial carnivores is minimal because of the aquatic nature of the species (Leopold and Chamberlain 2001). The mink and river otter are not competitors for resources (mink are primarily non-piscivorous and otter are primarily piscivorous). They are preyed upon by the coyote (*Canis latrans*), bobcat (*Lynx rufus*), and great-horned owl (*Bubo virginianus*). Linscombe et al. (1982) indicate the mink may occasionally fall victim to fisher (*Martes pennanti*), red fox (*Vulpes vulpes*), gray fox (*Urocyon cinereoargenteus*), and alligator (*Alligator mississippiensis*).

VULNERABILITY AND THREATS

Wetland loss is of special concern, because these areas provide important habitat for the mink. In the last two centuries, substantial wetland losses have occurred along the southern Coastal Plain and along the lower reaches of the Mississippi River (Trani 2002a). Florida alone has lost 46 percent (3.6 million hectares) of its wetlands (Stein et al. 2000). The mink is sensitive to hydrologic manipulation; the increased demand on surface water resources to support a growing human population is a concern. Industrial and residential water pollution can render habitat unsuitable, as the species is vulnerable to environmental contaminants (e.g., mercury and pesticide residues) concentrated in prey foods. In Florida, the conversion of privately owned portions of the Big Cypress Swamp to citrus production has the potential to result in loss of habitat (elimination of potential denning areas) and pollution of surface waters with excess fertilizer nitrates (Humphrey 1992). Roadkills may be a significant cause of mortality where roads cross wetlands.

MANAGEMENT SUGGESTIONS

Successful management of the American mink requires a combination of population and habitat management. Harvest levels are regulated by state wildlife management agencies. The maintenance of wetland habitat is critical for viable mink populations (Dickson 2001) and is important to other species of wildlife. Stream channelization, and the removal of aquatic shoreline vegetation and woody debris, should be avoided as it reduces prey availability (e.g., crayfish and fish). The prevention of high levels of environmental contaminants also is needed to maintain habitat quality for this species (Trani 2002*b*). Additional management centers on maintaining vegetative cover adjacent to wetlands, providing aquatic structural diversity (e.g., downfall, log jams), increasing pool to riffle ratios, and ensuring water permanence (Allen 1987). Finally, the ecology of the mink in the South is poorly understood; further research is warranted.

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American Badger

Taxidea taxus (Schreber, 1778)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The genus *Taxidea* contains only one recent species, the American badger (*T. taxus*). The American badger is confined entirely to North America and is distinct from the Old World badger (*Meles meles*). There are four recognized subspecies of *T. taxus*; two occur in the South: *T. t. berlandieri* and *T. t. taxus* (Long 1973, Hall 1981). The life history of the American badger is reviewed by Long (1973), Lindzey (1982), Long and Killingley (1983), and Messick (1987).

DISTINGUISHING CHARACTERISTICS

The American badger is a medium-sized mammal with a broad and flattened head, depressed body, short legs, and long, recurved foreclaws. Measurements are: total length, 680–780 mm; tail, 110–135 mm; hind foot, 100–130 mm; ear, 40–58 mm; weight, 5–12 kg. A white stripe extends mid-dorsally from just behind the eyes, over the head, and onto the middle of the back. The ears are short and rounded and the eyes are small. A large white patch on each cheek is bordered posteriorly by a black patch in front of each ear. The dorsal pelage appears shaggy because the fur is grizzled grayish or brownish, and the hairs are frosted with long white tips. The ventral pelage is yellowish-white. The short, bushy tail is yellowish-brown and the lower legs and feet are black. The hind feet are equipped with short, shovel-like claws. The skull is broad and flattened and the cranium is triangular when viewed from above. The last tooth in the upper tooth rows is triangular and large. The dental formula is $I\ 3/3, C\ 1/1, P\ 3/3, M\ 1/2 = 34$ (Figure 1). See keys for details.

CONSERVATION STATUS

The American badger has a global rank of Secure (NatureServe 2007). It is considered Secure in Texas and Apparently Secure in Oklahoma.

DISTRIBUTION

The American badger is found from northern Alberta, Canada, to central Mexico, and occurs throughout the western, midwestern, and north-central United States (Hall 1981, Choate et al. 1994). The species has been expanding its range in the northeastern United States (Lyon 1932, Leedy 1947, Nugent and Choate

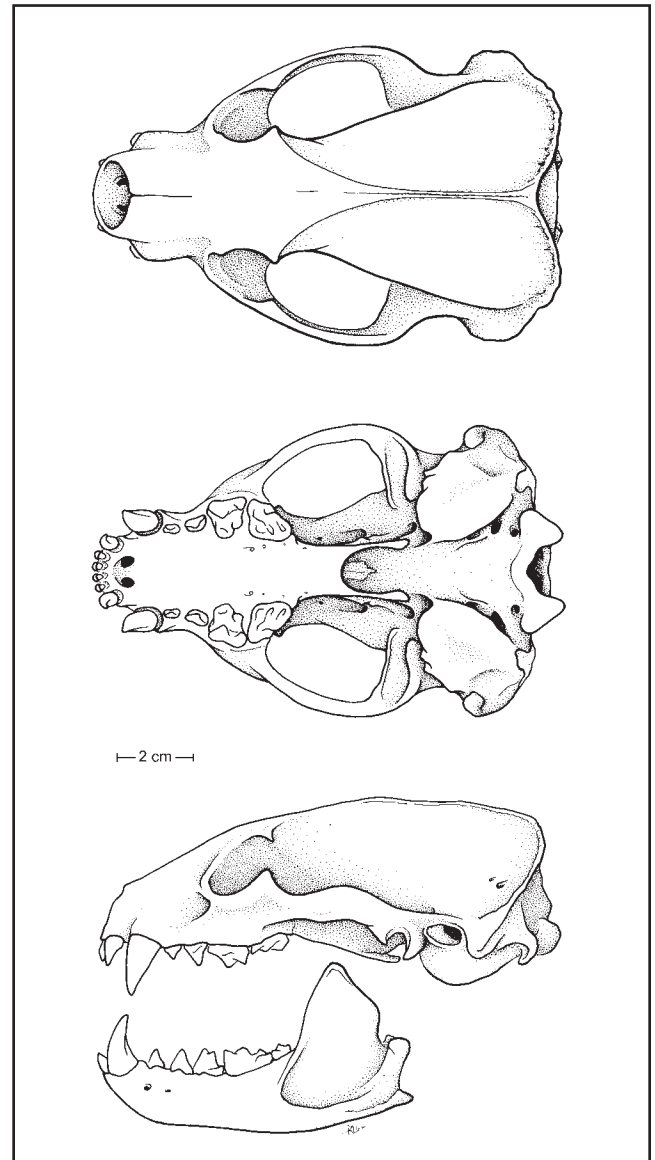


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Taxidea taxus* from Cameron County, Texas (USNM 36351, male).

1970). The distribution of the badger in the South is depicted in Figure 2. The American badger is more common in western Oklahoma than it is at the periphery of the distributional range in the eastern part of the state (Caire et al. 1989). There are a few reports of the American badger from northeastern Oklahoma (Taylor 1965), but the species apparently does not occur in the southeastern third of the state

(Caire et al. 1989). The species is known in Arkansas only from one specimen taken from the northwestern corner of the state (Sealander and Forsyth 1966), although there are undocumented reports of other badgers observed in the area (Sealander and Heidt 1990). Natural Heritage agencies (NatureServe 2007) no longer recognize the presence of badger in Arkansas. During the Pleistocene, the badger occurred in Kentucky (Guilday 1968, Long 1973) and there are recent records of the species just north of the Kentucky border in Indiana (Lyon 1932, Long 1973, Mumford and Whitaker 1982). The species may be expanding its range in southern Illinois (Gremillion-Smith 1985, Hoffmeister 1989). Wilson and Ruff (1999) show the range of the badger extending throughout central and western Texas.

ABUNDANCE STATUS

The American badger is known from few specimens in eastern Oklahoma and Arkansas and is considered rare in those areas. Near the center of their range in the western United States, the population density of the American badger is 0.4/km² (Seton 1929, Lindzey 1978). In areas of abundance, the species reaches densities of 3–5/km² (Wilson and Ruff 1999).

PRIMARY HABITATS

Throughout most of its range, the American badger occupies open, grassy areas with loose, sandy, or loamy soils. It sometimes occupies forested areas (Sargeant and Warner 1972, Lampe and Sovada 1981, Messick 1987). The species occurs in desert, savanna, and shrubland habitats. Soil type is an important factor influencing habitat use as *T. taxus* digs its own burrows, and avoids rocky or dense substrates (Messick and Hornocker 1981). Cultivated areas are avoided (Messick 1987). Local distribution and abundance may depend upon the occurrence of semi-fossorial prey, such as ground squirrels (*Spermophilus* spp.) or prairie dogs (*Cynomys* spp.) which are absent in the South (Clark et al. 1982).

REPRODUCTION

Breeding occurs in late July and August (Wright 1966, 1969; Messick and Hornocker 1981); implantation is delayed until December or January (Hamlett 1932). The single annual litter of 1–4 young is produced in March or early April (Wright 1966, Messick 1987). The young are weaned at 8 weeks. Females are capable of breeding at 4–5 months but apparently few do so (Wright 1969). Males and most females do not breed until they are at least 1 year old. Longevity of captive individuals approaches 13–15 years (Jackson 1961); few live longer than 2 years in the wild.

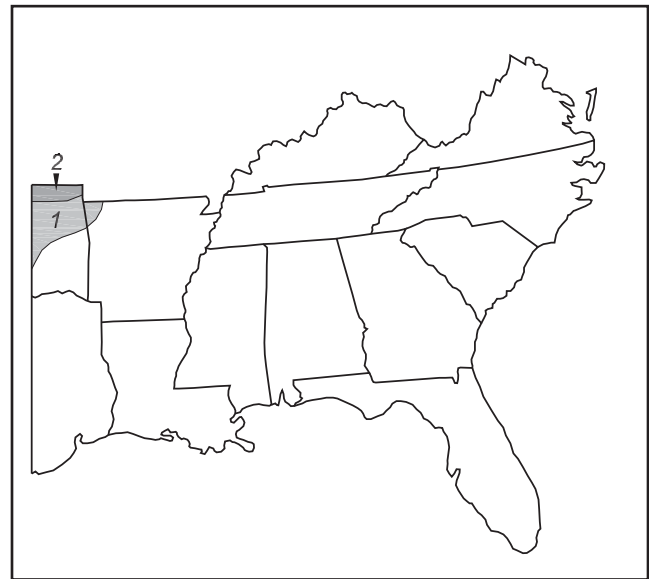


Figure 2. Distribution of *Taxidea taxus* in the South: (1) *T. t. berlandieri*; (2) *T. t. taxus*.

FOOD HABITS

The badger is an efficient predator of semi-fossorial and fossorial species (Messick 1987). Their primary prey includes pocket gophers (*Geomys* spp.), mice (*Peromyscus* spp.), ground squirrels and prairie dogs (Errington 1937, Snead and Hendrickson 1942, Lampe 1982). However, the American badger also supplements its diet with birds, reptiles, eggs, amphibians, fish, invertebrates, carrion, and plant material (Potter 1924, Errington 1937, Snead and Hendrickson 1942, Drake and Presnall 1950, Long and Killingley 1983). Seton (1929), Calahane (1950), and Minta et al. (1992) describe hunting associations between the American badger and the coyote (*Canis latrans*).

ASSOCIATED SPECIES

The mammalian associates of the American badger include the coyote, striped skunk (*Mephitis mephitis*), and weasels (*Mustela* spp.). Rodents and other furbearers may use abandoned American badger burrows for dens.

VULNERABILITY AND THREATS

The badger has become common in many areas of the East such as Illinois, Indiana and Ohio, though still largely absent from the South (Whitaker and Hamilton 1998). Still, the species may have declined in areas subject to the control of colonial rodents. Internal parasites of badgers include nematodes, cestodes, and trematodes.

MANAGEMENT SUGGESTIONS

In locations where habitat enhancement is an objective, Allen (1987) recommends maintaining grassland communities of sufficient size interspersed with agricultural land to support the prey base of the badger. Conversely, rodent control may make habitat less suitable for the badger; the species may forage elsewhere to search for food. Badger damage is often extensive, creating a hazard to livestock, horseback riders, and machinery. Other damage prevention and control methods include trapping and shooting; exclusion fencing is not effective.

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American Badger (*Taxidea taxus*)

Wright, P. L. 1966. Observations on the reproductive cycles of the American badger (*Taxidea taxus*). Symposium of the Zoological Society of London 15:27–45.

Wright, P. L. 1969. The reproductive cycle of the male American badger (*Taxidea taxus*). Journal of Reproduction and Fertility Supplement 6:435–445.

Ringtail

Bassariscus astutus (Lichtenstein, 1830)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

The ringtail (*Bassariscus astutus*) is a member of the family Procyonidae along with the northern raccoon (*Procyon lotor*). Only one subspecies (*B. a. flavus*), of the 14 recognized subspecies, occurs in the South (Hall 1981). The species also is known as the ring-tailed cat, cacomixtl, or cacomistle. The life history of the ringtail is reviewed by Kaufmann (1982, 1987) and Poglayen-Neuwall and Toweill (1988).

DISTINGUISHING CHARACTERISTICS

The ringtail is a small, cat-like carnivore with a long, black and white banded tail. Measurements are: total length, 615–810 mm; tail, 310–440 mm; hind foot, 60–78 mm; ear, 45–50 mm; weight, 725–1,100 g. The dorsal pelage is pale buff with black tipped hairs and the ventral pelage is white or buffy-white. The muzzle is narrow and pointed. The eyes are large. A black eye ring surrounded by prominent white patches above and below the eyes accentuate the size of the eyes. The ears are large, erect, and rounded. The long tail with 6–9 black bands is the most distinctive feature. The skull is elongate and somewhat flattened with slender zygomatic arches. The braincase expands laterally; the auditory bullae are inflated. The ringtail skull resembles that of a raccoon, but can be distinguished by the blade-like premolars. Raccoon premolars have multiple cusps. The dental formula of the ringtail is: $I\ 3/3, C\ 1/1, P\ 3/4, M\ 3/2 = 40$ (Figure 1). See keys for details.

CONSERVATION STATUS

The ringtail has a global rank of Secure (NatureServe 2007). It is Apparently Secure in Texas and Vulnerable in Oklahoma. It is unranked in Louisiana.

DISTRIBUTION

The ringtail occurs from southern Oregon eastward to the Mississippi River and as far south as central Mexico (Hall 1981, Choate et al. 1994). The distribution of the species in the South is depicted in Figure 2. The ringtail is found in Louisiana (Lowery 1974), eastern Texas (Blair 1949, Taylor 1954, Wood 1954, Schmidly 1983, 1984; Davis and Schmidly 1994), and south central Arkansas (Sealander and Gipson 1972, Sealander and Heidt 1990). The single record of the

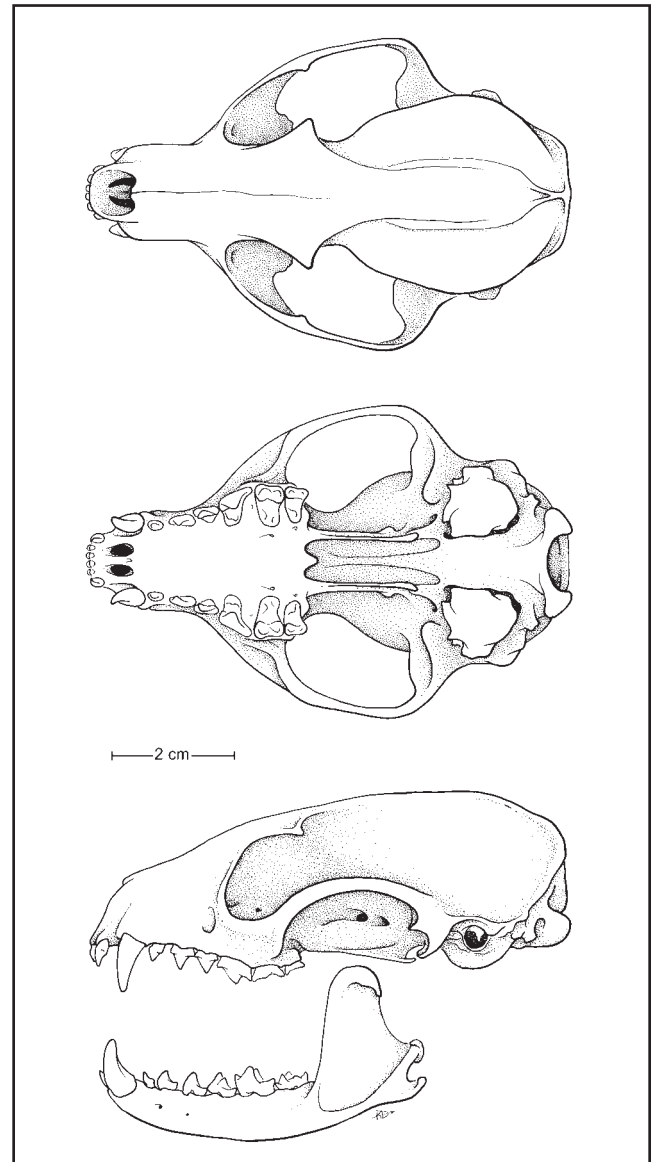


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Bassariscus astutus* from Cuernavaca, Mexico (USNM 51131, male).

ringtail in Alabama (Brannon 1922) likely represented an escaped captive animal (Poglayen-Neuwall and Toweill 1988).

ABUNDANCE STATUS

Population densities of the ringtail in the South are unknown. The species is known from only a few

records in Louisiana (Lowery 1974) and Arkansas (Sealander and Heidt 1990) and is considered uncommon in eastern Texas (Schmidly 1983, 1984). Wilson and Ruff (1999) indicate that ringtail density in central Texas may be 1/25 ha.

PRIMARY HABITATS

Throughout its range, the ringtail commonly inhabits brushy or rocky slopes and rock outcroppings (Poglayen-Neuwall and Toweill 1988). It may be found in all available habitats in eastern Texas but prefers wooded habitats near water (Schmidly 1983). The ringtail does not construct dens, but occupies rock crevices, abandoned burrows, tree hollows, brush piles, and rural buildings as temporary den sites (Grinnell et al. 1937, Toweill and Teer 1977, Trapp 1978, Davis and Schmidly 1994). The ringtail climbs trees with ease and can rotate its hind feet 180 degrees, an ability which allows it to run down a tree trunk head first (Trapp 1972). It is active at night and twilight (Kavanau 1971).

REPRODUCTION

Although the breeding season extends from February to May, most mating occurs in March and April (Taylor 1954, Bailey 1974). Females are monestrous and are receptive to the male for a maximum of 36 hours (Bailey 1974). The gestation period is 51–54 days (Poglayen-Neuwall and Poglayen-Neuwall 1980); parturition occurs in May or June when a litter of 1–4 young are born (Fry 1926, Grinnell et al. 1937, Richardson 1942). The newborn are blind and sparsely haired (Richardson 1942, Bailey 1974, Toweill and Toweill 1978). The young are weaned at 10 weeks of age and attain full size at 30 weeks (Richardson 1942, Toweill and Toweill 1978). The ringtail attains sexual maturity near the end of its second year but successful mating has been reported for younger individuals (Poglayen-Neuwall and Poglayen-Neuwall 1980). Life span in the wild is unknown but captive individuals have lived for 16.5 years (Poglayen-Neuwall 1987).

FOOD HABITS

The ringtail is omnivorous, but it tends to be more carnivorous than herbivorous. The animal portion of its diet includes a variety of arthropods, small mammals, birds, lizards, snakes, frogs, fish, and carrion (Taylor 1954, Wood 1954, White and Lloyd 1962, Toweill and Teer 1977, Trapp 1978, Davis and Schmidly 1994). It also consumes the fruits of many plants including persimmon (*Diospyros* spp.), hackberry (*Celtis* spp.), and prickly pear (*Opuntia* spp.).

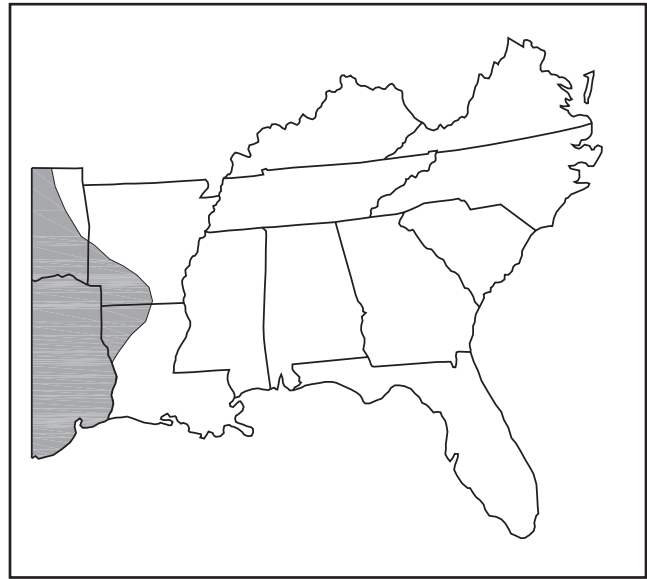


Figure 2. Distribution of *Bassariscus astutus* in the South.

ASSOCIATED SPECIES

The ringtail sometimes shares a den with the nine-banded armadillo (*Dasypus novemcinctus*; Toweill and Price 1976). The ringtail apparently competes for food with the northern raccoon, Virginia opossum (*Didelphis virginiana*), common gray fox (*Urocyon cinereoargenteus*), and striped skunk (*Mephitis mephitis*; Wood 1954). The species is preyed upon by the coyote (*Canis latrans*), bobcat (*Lynx rufus*), great horned owl (*Bubo virginianus*), and other carnivores.

VULNERABILITY AND THREATS

Although it is not regarded as a common animal in the South, there are no known threats to the survival of the ringtail in the region. It is harvested for its fur, but there is no evidence that trapping has affected its population density.

MANAGEMENT SUGGESTIONS

Allen (1987) recommends the maintenance of dense riparian vegetation in association with draws and ridgelines as travel corridors. The maintenance of denning cavities in proximity to a water source also is beneficial.

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Northern Raccoon

Procyon lotor (Linnaeus, 1758)

Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Hall (1981) recognizes 31 subspecies of the northern raccoon (*Procyon lotor*). Thirteen of these subspecies occur in the South: *P. l. auspicatus*, *P. l. elucus*, *P. l. fuscipes*, *P. l. hirtus*, *P. l. incautus*, *P. l. inesperatus*, *P. l. litoreus*, *P. l. lotor*, *P. l. marinus*, *P. l. maritimus*, *P. l. megalodous*, *P. l. solutus*, and *P. l. varius*. It is difficult to distinguish most subspecies of *P. lotor* if the collection locality is unknown (Lotze and Anderson 1979). Lotze and Anderson (1979), Kaufmann (1982), and Sanderson (1987) summarize the life history of the raccoon.

DISTINGUISHING CHARACTERISTICS

The raccoon is a stocky, medium-sized mammal with a prominent black mask. Measurements are: total length, 730–950 mm; tail, 220–260 mm; hind foot, 100–125 mm; ear, 50–65 mm; weight, 5–15 kg. The dorsal pelage is gray to blackish-brown and the venter is lighter. Some raccoons have undertones of yellow, brown, or cinnamon in their fur. The head is broad and the nose is pointed. The face is white but black patches surround each eye forming a conspicuous mask. The tail is bushy and is marked by four to six distinct black rings. Each foot has five toes. The skull is elongate and usually exceeds 100 mm in length. Northern raccoon skulls are distinguished from ringtail (*Bassariscus astutus*) skulls in two ways. The canines of raccoons are laterally flattened whereas they are rounded in ringtails. The palate of the raccoon extends beyond the posterior edge of the last molar for a distance that exceeds the width of the palate. In the ringtail, the palate ends near the posterior edge of the molars. The dental formula for the raccoon is $I\ 3/3, C\ 1/1, P\ 4/4, M\ 2/2 = 40$ (Figure 1). See keys for details.

CONSERVATION STATUS

The northern raccoon has a global rank of Secure (NatureServe 2007). The species is also considered Secure in those states where it occurs within the region except for Arkansas, where it is Apparently Secure. It is unranked in South Carolina. Two subspecies were under consideration for federal listing: *P. l. auspicatus* and *P. l. incautus* (Whitaker and Hamilton 1998).

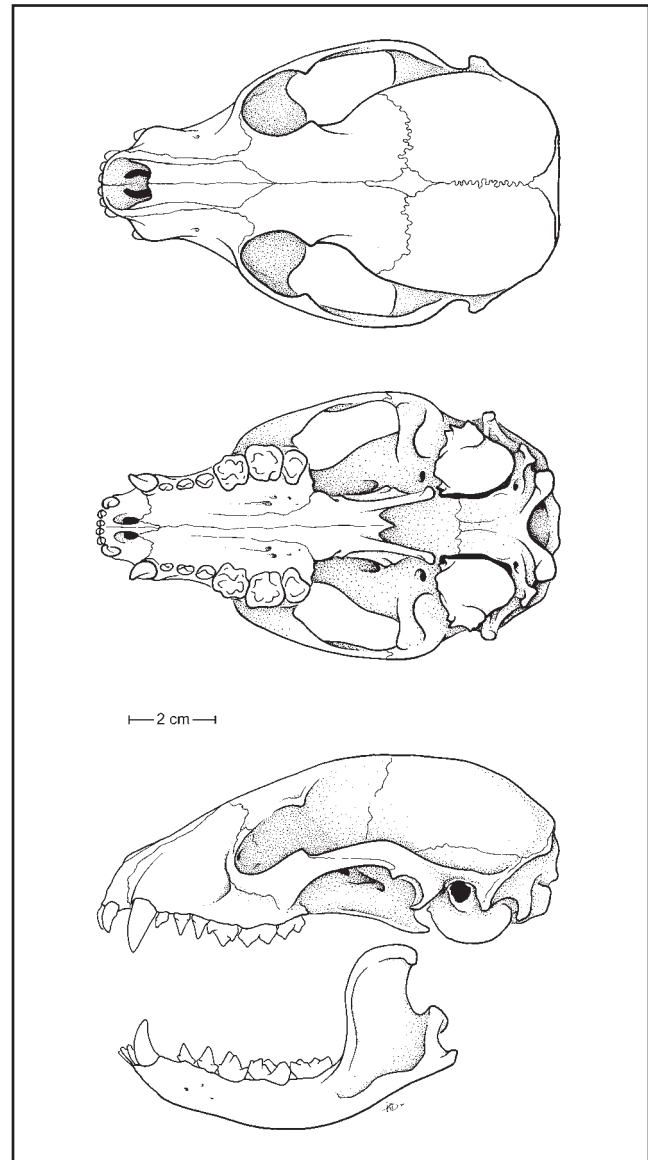


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Procyon lotor* from Baldwin County, Alabama (USNM 178079, male).

DISTRIBUTION

The northern raccoon has a broad geographical range in North America. It occurs from British Columbia to Nova Scotia and southward throughout most of the United States except for some portions of the Rocky Mountains and the Great Basin. It also is distributed throughout Mexico except part of Baja California,

and extends southward to Panama (Goldman 1950, Hall 1981, Choate et al. 1994). The northern raccoon is found throughout the South (Figure 2). The species occurs statewide in Virginia (Bailey 1946, Sonenshine and Winslow 1972, Webster et al. 1985, Handley 1991, 1992; Linzey 1998), North Carolina (Conaway and Howell 1953, Lee et al. 1982, Clark et al. 1985, Webster et al. 1985, Linzey 1995), and South Carolina (Penny 1950, Golley 1966, Webster et al. 1985, Cothran et al. 1991). The species inhabits Georgia (McKeever 1958, Golley 1962, Fountain 1975, Hudson 1978, Laerm et al. 1981, Diefenbach et al. 1994), Florida (McKeever 1958, Conner et al. 1983, McKeever et al. 1989, Ratnaswamy et al. 1997, Walker and Sunquist 1997, Kramer et al. 1999), Alabama (Howell 1921, Johnson 1970, Sumner and Hill 1980), Mississippi (Yeager 1941, Kennedy et al. 1974, Jones and Carter 1989, Henner et al. 2000, Hodges et al. 2000, Chamberlain et al. 2002), and Louisiana (Goldman 1950, Lowery 1974, Morrison et al. 1982, Lindscombe et al. 1983). It is abundant in eastern Texas (Schmidly 1983, Strapper et al. 1989, Davis and Schmidly 1994) and eastern Oklahoma (Caire et al. 1989). The raccoon occurs statewide in Arkansas (Sealander and Heidt 1990), Tennessee (Howell and Conaway 1952, Goodpaster and Hoffmeister 1952, Conaway and Howell 1953, Smith et al. 1974, Hatcher and Shaw 1981, Moore and Kennedy 1985a,b; Leberg and Kennedy 1987, 1988; Nottingham et al. 1989, Kennedy et al. 1991, Kissell and Kennedy 1992, Linzey 1995, White et al. 1998), and Kentucky (Barbour and Davis 1974, Fassler 1974, White et al. 1998).

ABUNDANCE STATUS

Accurate population densities are difficult to determine for the northern raccoon (Sanderson 1987). One of the greatest densities was reported by Twichell and Dill (1949), who removed 100 raccoons from a 41 ha waterfowl refuge in Missouri. In what they considered average raccoon habitat in Virginia, Sonenshine and Winslow (1972) estimated the population density to be 1/5.8 ha. Kaufmann (1982) summarized the reported home range sizes of the northern raccoon and found that maximum home range diameters were within 1–3 km. Some home ranges approached 6.4 km in size. Males maintain larger home ranges than females (Gehrt and Fritzell 1997, Walker and Sunquist 1997) and males move faster and farther each night than females (Walker and Sunquist 1997).

PRIMARY HABITATS

The northern raccoon occurs in many habitat types and is most abundant in areas near water (Sanderson 1987). Populations are highest in bottomland

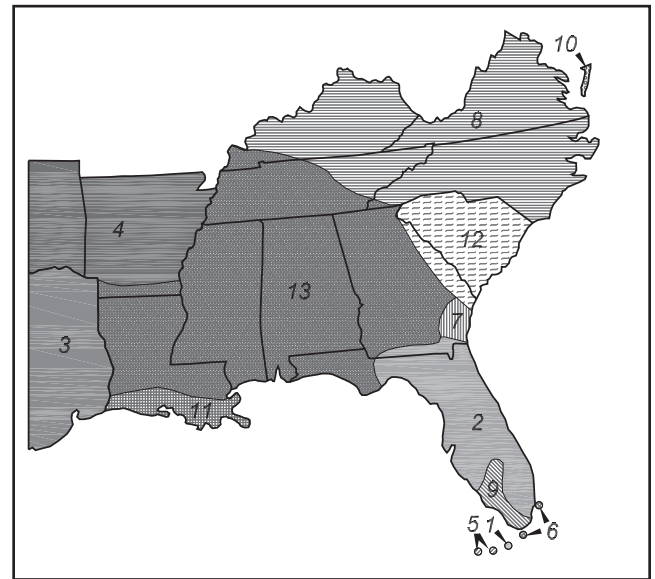


Figure 2. Distribution of *Procyon lotor* in the South: (1) *P. l. auspicatus*; (2) *P. l. elucus*; (3) *P. l. fuscipes*; (4) *P. l. hirtus*; (5) *P. l. incautus*; (6) *P. l. inesperatus*; (7) *P. l. litoreus*; (8) *P. l. lotor*; (9) *P. l. marinus*; (10) *P. l. maritimus*; (11) *P. l. megalodous*; (12) *P. l. solutus*; (13) *P. l. varius*.

hardwood forests associated with streams or rivers, hardwood swamps, and the habitats surrounding reservoirs, marshes, and mangroves. Populations are low in much of the southern Appalachians and in southern pine (*Pinus* spp.) forests (Kaufmann 1982). Managed pine forests are used seasonally (Chamberlain et al. 2000, 2002). Hollow trees are valued as den sites; where these are not available, the northern raccoon may den in abandoned burrows of other mammals, piles of brush, lumber, or other structures (Berner and Gysel 1967). Because of its tendency to use human-influenced habitats, the northern raccoon is a common nuisance animal reported by wildlife agencies in urban and suburban areas (De Almeida 1987).

REPRODUCTION

The breeding season in southwestern Georgia and northwestern Florida extends from February–August; most females mate in March (McKeever 1958). Johnson (1970) found the peak of the northern raccoon mating season in Alabama was from March–June. Ovulation probably occurs after copulation (Llewellyn and Enders 1954) and the gestation period is approximately 63 days (Sanderson and Nalbandov 1973). In Alabama, birth dates range from May–August and the mean number per litter varies from 1.9–5.0 (Johnson 1970). Late litters are sometimes born in September (Gehrt and Fritzell 1996). Young northern raccoons are weaned between 7–16 weeks of age (Montgomery 1969), but the young may remain for at least 9 months

(Sharp and Sharp 1956, Gehrt and Fritzell 1998a). Some northern raccoons may breed in their first year but most do not breed until their second year (Stuewer 1943, Pope 1944, Sanderson 1951, Wood 1955). Most females reach adult size as yearlings, but males do not reach adult size until they approach two years of age (Gehrt and Fritzell 1998b). The longevity record for the northern raccoon in the wild is 12 years (Haugen 1954, Johnson 1970); most live less than 5 years (Johnson 1970, McKeever et al. 1989).

FOOD HABITS

The northern raccoon is omnivorous and tends to be opportunistic when food is scarce but selective when it is abundant (Wood 1955, Johnson 1970). The northern raccoon eats seeds, berries, nuts, arthropods, and small vertebrates. In eastern Texas, acorns (*Quercus* spp.) and crayfish constituted more than half the diet of the raccoon during all seasons (Baker et al. 1945). Wood (1954) found that plants made up over 55% and insects, (mainly Orthoptera), comprised 27% of the diet in eastern Texas. Principal plant foods were acorns, grapes (*Vitis* spp.), yaupon berries (*Ilex vomitoria*), holly (*Ilex* spp.) and hackberry (*Celtis occidentalis*). Raccoons also consume rabbits (*Sylvilagus* spp.), hispid cotton rats (*Sigmodon hispidus*), crabs, and snakes (*Agkistrodon* and *Nerodia* spp.). Similar food habits were reported from Alabama (Johnson 1970). Wilson (1953) reported that the raccoon preys on the common muskrat (*Ondatra zibethicus*) in North Carolina marshes. The raccoon can be a major egg predator of sea turtles (Shabica et al. 1978, Ratnaswamy et al. 1997), colonially nesting waterbirds (Kadlec 1971), and waterfowl (Llewellyn and Webster 1960).

ASSOCIATED SPECIES

The raccoon commonly frequents marshes and the margins of lakes, ponds, and watercourses. In these habitats, it frequently is in close association with the American beaver (*Castor canadensis*), common muskrat, northern river otter (*Lontra canadensis*), and weasel (*Mustela* spp.). The northern raccoon sometimes uses the abandoned dens of the woodchuck (*Marmota monax*), common gray fox (*Urocyon cinereoargenteus*), red fox (*Vulpes vulpes*), and striped skunk (*Mephitis mephitis*; Butterfield 1954, Gysel 1961). The northern raccoon is a primary host of the ascarid roundworm, *Baylisascaris procyonis*, and has been implicated in the decline of the Allegheny woodrat (*Neotoma magister*) (LoGiudice 2006).

VULNERABILITY AND THREATS

The northern raccoon is so widely distributed and ecologically tolerant that there are no threats to its

survival in the region. In mountain habitats, the species was scarce in much of the 20th century due to over hunting (Minser and Pelton 1982, Cantrell 1989).

MANAGEMENT SUGGESTIONS

Active habitat management includes the provision of denning trees and enhancement of mast producing trees and shrubs. Aquatic systems such as wetlands, marshes, and streams should be protected from pollution. Due to the variety of foods the species consumes, food shortage is rarely a problem. Low fur prices and reductions in demand for pelts have caused a decline in raccoon harvest. With such declines, population management may be required to control damage to agricultural and urban areas. In selected areas, conflicts with humans and concerns regarding disease transmission (e. g., rabies) may create conditions when removal of individual animals is necessary. Trapping and hunting are effective control methods (Atkenson and Hulse 1953).

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American Black Bear

Ursus americanus (Pallas, 1780)

Margaret K. Trani and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Hall (1981) and Lariviere (2001) recognize 16 subspecies of the American black bear (*Ursus americanus*). Three subspecies occur in the region: *U. a. americanus*, Florida black bear (*U. a. floridanus*), and Louisiana black bear (*U. a. luteolus*). The life history of the black bear is reviewed by Kolenosky and Strathearn (1987), Powell et al. (1997), Lariviere (2001), and Pelton (2003).

DISTINGUISHING CHARACTERISTICS

The American black bear is a large-bodied, powerfully built species. Measurements are: total length, 125–180 cm; tail, 8.0–12.5 cm; hind foot, 18.5–28.0 cm; ear 13.0–14.5 cm; weight, 100–250 kg. Males typically are 10–70% heavier than females. Most black bears in the South are uniformly black with a tapered, brown muzzle and an occasional white blaze on the chest (D. Hightower, Quantitative Ecological Services, personal communication). The eyes and erect ears are small. The legs and feet are massive; the plantigrade feet possess strong, recurved claws. The skull is massive (may exceed 30 cm in length) with a large cranium, prominent sagittal crest and zygomatic arches, and broad rostrum. Auditory bullae are flat, depressed, and inconspicuous. Nares are large and expose well-developed turbinate bones (Lariviere 2001). In an analysis of morphological characters from eastern black bear, Kennedy et al. (2002a) reported that the largest measurements of the cranium and mandible occurred from animals collected in the southernmost states. The teeth are bunodont and the first three premolars are rudimentary; the last upper molar is elongate rather than round. The dental formula is $I\ 3/3$, $C\ 1/1$, $P\ 4/4$, $M\ 2/3 = 42$; the number of premolars may vary on each side of the upper and lower jaws (Figure 1). See keys for details. Kennedy et al. (2002b) provide additional measurements for subspecific recognition including dentition, height of frontal region, and skull length and width.

CONSERVATION STATUS

The American black bear has a global rank of Secure (NatureServe 2007). It is also Secure in Florida and Apparently Secure in Georgia, North Carolina and Virginia. The black bear is listed as Vulnerable in Arkansas, South Carolina, Tennessee and Texas. It is Imperiled in Alabama, Kentucky, Louisiana, and

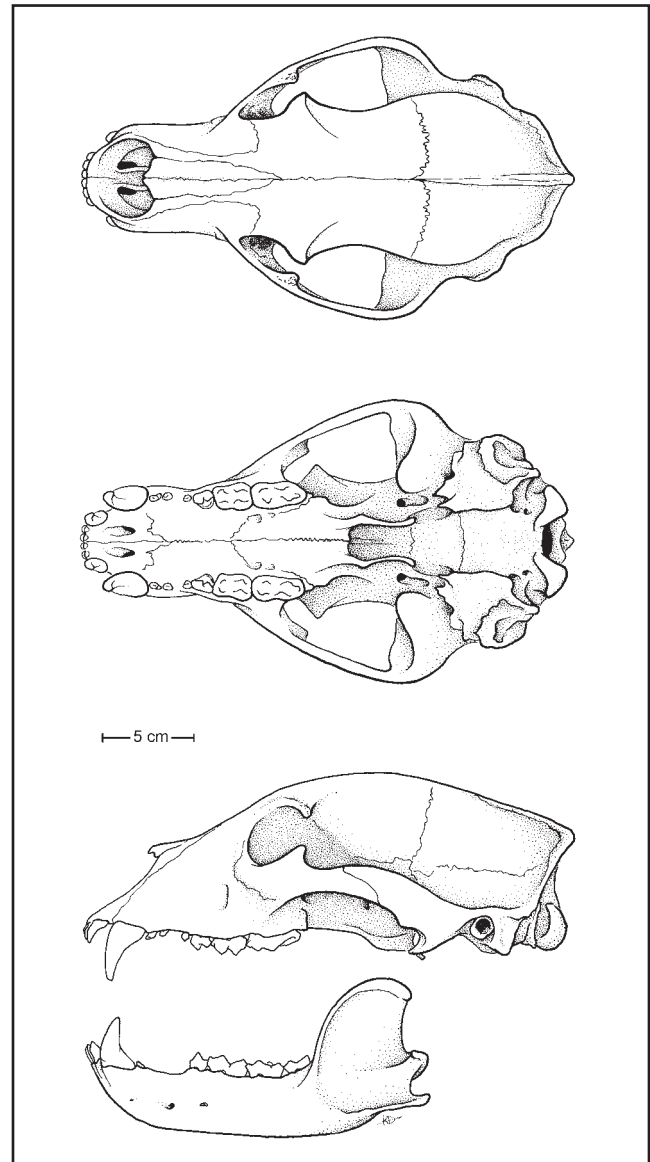


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Ursus americanus* from “Rocky River”, Virginia (USNM 288946, gender unknown)

considered Critically Imperiled in Mississippi and Oklahoma. Regulated hunting seasons occur in Virginia, Tennessee, North Carolina, Georgia, South Carolina, and Arkansas. The timing, length of season, and methods allowed varies; harvests within each state where hunting is allowed are based on bear population levels. No other game species or its

harvest is as closely monitored in the South. There is currently no hunting season in Kentucky, Oklahoma, or Texas.

The U. S. Fish and Wildlife Service (U. S. Department of the Interior 2007) lists the Louisiana black bear as Threatened, with a recovery plan approved for the species (USFWS 1995). Other bears within the historic range of *U. a. luteolus* are designated as Threatened by Similarity of Appearance and are protected in eastern Texas, southern Mississippi, and Louisiana. The Florida black bear (*U. a. floridanus*) is listed as Threatened by the Florida Fish and Wildlife Conservation Commission. The subspecies was a candidate for listing under the U. S. Endangered Species Act (Federal Register, 28 February 1996) and subsequently removed from candidacy (Federal Register, 8 December 1998). The subspecies is protected in Alabama.

DISTRIBUTION

The American black bear historically ranged throughout most of North America from Alaska to the Atlantic Ocean and south to central Mexico. The species was largely absent only from portions of the Great Basin Desert and the arid Southwest (Hall 1981, Choate et al. 1994). Habitat loss, fragmentation, and unrestricted harvest have significantly reduced the present distribution of the American black bear (Hellgren and Vaughan 1994, Lariviere 2001). Since the 1970s, active state management programs have resulted in expanding populations in some portions of the South (Pelton 2001).

U. a. americanus. Martin and Steffen (2005) report that the black bear has been observed in all but seven counties in the easternmost portion of Virginia (Figure 2). The highest populations are found in the western mountains (e.g., Shenandoah National Park in the Blue Ridge Mountains) and in the southeast tidewater area of the Great Dismal Swamp bordering North Carolina, while populations in the eastern counties are sparse (Hellgren et al. 1991, Handley 1992, Garshelis and Hellgren 1994, Kasbohm et al. 1996, Linzey 1998, D. Martin, Virginia Game and Inland Fisheries, personal communication). The species occurs in two disjunct populations in North Carolina: the first occurs in the Appalachian Mountains in the western third of the state and the second occurs throughout the Coastal Plain in the eastern portion of the state (Clark et al. 1985, Warburton et al. 1993, Pelton and van Manen 1996, Beringer et al. 1998, Mitchell et al. 2002, Jones 2005). In South Carolina, the mountain population of American black bear occurs in Oconee, Pickens, and Greenville counties, and appears to be expanding their range southward

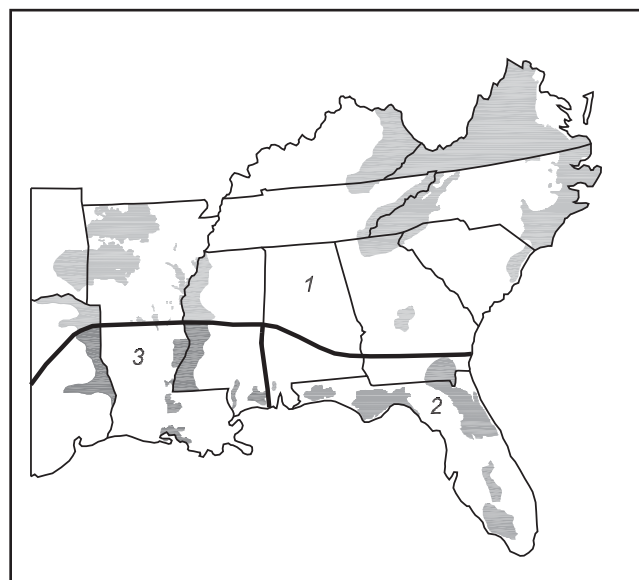


Figure 2. Distribution of *Ursus americanus* in the South: (1) *U. a. americanus*; (2) *U. a. floridanus*; (3) *U. a. luteolus*.

(S. Still, South Carolina Department of Natural Resources, personal communication). The Coastal Plain population occurs primarily in Georgetown and Horry counties (Golley 1966, Webster et al. 1985, Cothran et al. 1991, Willey et al. 1996, Still 2005). The two disjunct populations of *U. a. americanus* in Georgia occur in the Appalachian Mountains and portions of the upper Piedmont in the northern portion of the state and the Ocmulgee River drainage in the upper Coastal Plain (Golley 1962, Laerm et al. 1981, Cantrell et al. 2005). The species is known from 11 counties within the Blue Ridge portion of the southern Appalachians in eastern Tennessee (Eiler et al. 1989, Kennedy 1991, Pelton 2001, 2003). Following an experimental repatriation of black bear during 1996–1998 in the Big South Fork National River and Recreation Area and Daniel Boone National Forest in Tennessee and Kentucky (Eastridge and Clark 1998), a second disjunct population now occurs in the Cumberland Mountains and Plateau region within Campbell, Fentress, Morgan, and Scott counties (Brandenburg 2005). American black bears also occur in other portions of eastern Kentucky (Day et al. 2005) that include dispersing animals from Virginia and West Virginia (Pelton 2001). The bear was reintroduced into the Arkansas Interior Highlands (Ozark and Ouachita Mountains) between 1958–1968 from Minnesota and Manitoba, Canada (Smith et al. 1991). Today, the Interior Highlands populations have expanded their range into Missouri, Oklahoma, and southward along the Ouachita and Saline river drainages (Eastridge 2005). It also occurs in portions of the Mississippi Alluvial Valley in the White River National Wildlife Refuge and along the southern border in

Ashley, Bradley, and Union counties (Clark and Smith 1994, Bowman et al. 1996, Oli et al. 1997, Clark et al. 2005). There are questions over the subspecific taxonomy of bears occurring within the refuge; Csiki et al. (2003) present DNA data indicating that these animals may be Louisiana black bear. The U. S. Fish and Wildlife Service formally recognize the Arkansas/Louisiana state border as the range limit for the subspecies (M. Harney, U. S. Fish and Wildlife Service, personal communication). Although the black bear was extirpated from eastern Oklahoma (Caire et al. 1989), the species now occurs in the in the southeastern portion of the state in LeFlore, McCurtain, Latimer, Haskell, and Pushmataha counties (Bales 2003; R. Bastarache, U. S. Forest Service, personal communication). The black bear also occurs in the vicinity of Broken Bow Lake north through the Ouachita National Forest and Kiamichi, Winding Stair, and San Bois Mountains (J. Ford, Oklahoma Department of Wildlife, personal communication).

U. a. luteolus. In Louisiana, black bears are found in the Upper and Lower Atchafalaya River Basin in the south central part of the state and on the Tensas National Wildlife Refuge and surrounding private lands in the Tensas River Basin in northeast Louisiana (Weaver et al. 1990, Marchinton 1995, Pace et al. 2000, Hightower et al. 2002, Clark et al. 2005, Davidson 2005). Translocated bears from Minnesota augmented the Upper Atchafalaya Basin and Tensas River Basin populations during the 1960s (Triant et al. 2003). There has been a recent range expansion within the Three Rivers Wildlife Management area in central Louisiana (D. Telesco, Black Bear Conservation Committee, personal communication). Currently in the sixth year of a repatriation project, 168 bears have been moved from the northern population into suitable, unoccupied habitat in east-central Louisiana (D. Telesco, Black Bear Conservation Committee, personal communication). A remnant population exists in Mississippi (Jones and Carter 1989, Stinson 1996, Oli et al. 1997). Black bear sightings are concentrated along the western border, primarily in the Mississippi, Pearl, and Pascagoula River drainages (Mississippi Department of Wildlife, Fisheries, and Parks 2005). Reproducing bears have recently been reported in the southwestern corner of the state (B. Young, Mississippi Department of Wildlife, Fisheries, and Parks, personal communication). The species was believed extirpated in eastern Texas during the period between 1900–1940 (Davis and Schmidly 1994). Currently, there are 28 counties with confirmed bear sightings although they likely are juvenile or sub-adult males dispersing from Arkansas, Oklahoma (Maxey and Lange 2005), and Louisiana (R. Maxey, Texas Parks and Wildlife, personal communication). The majority of sightings are near major river basins

including the Angelina, Cypress, Neches, Red, Sabine, San Jacinto, Trinity, and Sulphur River basins.

U. a. floridanus. The six primary black bear populations within Florida occur on the Apalachicola National Forest, Big Cypress National Preserve, Eglin Air Force Base, Ocala and Osceola National Forest, and St. Johns River region (Maehr and Wooding 1992, Roof 1997, Seibert et al. 1997, Dobey et al. 2005, Dixon et al. 2006). Two smaller, restricted populations occur in the Greater Chassahowitzka Ecosystem and in the Glades and Highland counties area (Maehr et al. 2003, Simek 2005). In southeast Georgia, the subspecies inhabits the Okefenokee National Wildlife Refuge and the Dixon Memorial State Forest (Wilson and Ruff 1999, Cantrell et al. 2005). A small, remnant population of Florida black bear also exists in southwestern corner of Alabama near the Mississippi border (K. Guyse, Alabama Department of Conservation and Natural Resources, personal communication).

ABUNDANCE STATUS

Populations appear to be stable or increasing in Virginia, Tennessee, North Carolina, Georgia, South Carolina, and Arkansas. Martin and Steffen (2005) estimate the number of bears in Virginia at 7000–9000. In North Carolina, the mountain population (4000) is increasing whereas the coastal population (7000) appears stable (M. Jones, North Carolina Wildlife Resources Commission, personal communication). Black bear abundance in northern and central Georgia is stable or slightly increasing (Cantrell et al. 2005) with population estimates between 1400–1500 (B. Bond and W. Abler, Georgia Department of Natural Resources, personal communication). Population estimates for Tennessee range between 1500–2000 (D. Ratajczak, Tennessee Wildlife Resources Agency, personal communication); the Great Smoky Mountains National Park population shared with North Carolina is estimated at approximately 1250 bears (W. Stiver, National Park Service, personal communication). South Carolina reports the mountain population (900) is increasing whereas the Coastal Plain population (250) is stable (S. Still, South Carolina Department of Natural Resources, personal communication). The Interior Highlands population in Arkansas has grown to 3,000 bears (Eastridge 2005); the White River population is estimated between 300–500 animals (R. Hines, U. S. Fish and Wildlife Service, personal communication). Black bear abundance in eastern Oklahoma is estimated at 250–275 (J. Ford, Oklahoma Department of Wildlife, personal communication). Bear abundance for the small, recolonizing population in Kentucky is unknown (Day et al. 2005); research is underway to develop a population estimate using hair snare and infrared

surveys (D. Maehr, University of Kentucky, personal communication).

There are between 2000–3200 Florida black bears in Florida (Simek 2005), approximately 50 in Alabama (K. Guyse, Alabama Department of Conservation and Natural Resources, personal communication), and 700–800 in South Georgia (W. Abler, Georgia Department of Natural Resources, personal communication). The population of Louisiana black bear is estimated at almost 600 in the Tensas and Atchafalaya River basins (D. Telesco, Black Bear Conservation Committee, personal communication) and 60 in Mississippi (B. Young, Mississippi Department of Wildlife, Fisheries, and Parks, personal communication). In Texas, sightings of black bear have increased 60–70% in the past 10–15 years (Maxey and Lange 2005); the current estimate of bear abundance is less than 50 animals (R. Maxey, Texas Parks and Wildlife, personal communication). Research is underway by state personnel and cooperators to determine the extent of range expansion.

PRIMARY HABITATS

In the Appalachians, Ozark, and Ouachita mountains, the black bear inhabits several communities including red spruce-fir (*Picea rubens-Abies*) forest, northern hardwoods (*Acer-Fagus-Betula*); mixed mesophytic forest including blackgum (*Nyssa sylvatica*), black walnut (*Juglans nigra*), and eastern hemlock (*Tsuga canadensis*); oak-hickory (*Quercus-Carya*) forest, mixed pine (*Pinus* spp.) forest, and others (Garshelis and Pelton 1981, Pelton 2003). Large tracts of suitable habitat are important (Kolenosky and Strathearn 1987, Pelton 2003). Areas of at least 389–778 km² with relatively low human habitation have been suggested for sustaining harvestable, viable populations (Hellgren and Vaughan 1984, Rudis and Tansley 1995). In the Coastal Plain, habitat includes pocosins, bottomland hardwoods, Carolina bays, mixed hardwood hammocks, bald cypress (*Taxodium distichum*) swamps, cabbage palm (*Sabal palmetto*) forests, pine flatwoods dominated by slash pine (*P. elliotii*) and longleaf pine (*P. palustris*), loblolly bay (*Gordonia lasianthus*), and sand pine (*P. clausa*) scrub (Landers et al. 1979, Clark et al. 1985, Webster et al. 1985, Mykytko and Pelton 1990, Weaver et al. 1990, Hellgren et al. 1991, Maehr et al. 2003, Dobey et al. 2005). The black bear requires an impenetrable understory for escape cover as a habitat component (Pelton 2001); dense thickets of mountain laurel (*Kalmia latifolia*) and rhododendron (*Rhododendron maxima*) are important in the Appalachians whereas greenbrier (*Smilax* spp.), titi (*Cliftonia monophylla*), and other evergreen shrubs are important in the Coastal Plain. Winter denning habitat includes cavities in large trees, hollow logs and

stumps, standing snags, piles of woody debris, caves, rock outcroppings, and thickets (Hellgren and Vaughan 1989b, Wooding and Hardisky 1992, Pelton 2003). Elevated den sites are important in areas with significant winter flooding (Oli et al. 1997). The American black bear requires secure corridors for obtaining food, dispersal, and travel (White et al. 2000, Pelton 2001).

Sex, population density, and changing food resources influence the size of the home range (Pelton 2003). Average home ranges are quite variable with sizes reported of 2–11 km² in the White River area of Arkansas (Oli et al. 2002) and 30–78 km² in the Okefenokee National Wildlife Refuge-Osceola National Forest area of Georgia and Florida (Dobey et al. 2005). Home range estimates from Appalachian populations include 7–51 km² in Tennessee (van Manen 1994) and 27–112 km² in Virginia (Hellgren and Vaughan 1989a). Seasonal changes in habitat use occur in response to food availability; following poor acorn and other mast production in the fall, black bears disperse into habitats that increase their chances of contact with humans (Pelton 2001).

REPRODUCTION

Breeding occurs in summer and peaks between June–July (Eiler et al. 1989, Pelton 2003). Depending on nutritional status, female black bears reach sexual maturity between 2–7 years with 3–4 years being common (Pelton 2003). Black bears are polygynous and dominant males may mate with several females (Pelton 2001). During years of poor mast production, suboptimal nutrition results in failed implantation of the blastocysts or death of the neonates (Hellgren et al. 1990). If a female attains sufficient body weight by fall, implantation occurs (Samson and Huot 1995). The gestation period for the American black bear is 7–8 months with the majority of fetal development occurring during the last 6–8 weeks. Cubs are born in winter dens during January–February (Pelton 2003). The average litter size is 2 (range 1–5); reported litter sizes in Louisiana range between 1.5–2.4 (Hightower et al. 2002). The adult female normally breeds every other year; older females show the greatest productivity (Eiler et al. 1989). Dispersal occurs at approximately 16 months (Seibert et al. 1997); most mortality occurs in this subadult group up through 4–5 years of age. Years with poor food availability can cause reproductive failure, initiating population-wide breeding synchrony (McLean and Pelton 1994, Pelton 2003). With declining nutrition, litter size may decline, age at first reproduction rises, juvenile and first-year cub survival diminish, and litter frequency declines (Lariviere 2001). The American black bear can live for 20–25 years (Kolenosky and Strathearn 1987).

FOOD HABITS

The American black bear is an opportunistic feeder with a diet that changes seasonally. During the spring, newly emerging grasses, sedges, forbs, catkins, leaves of trees and shrubs, and squawroot (*Conopholis americana*) comprise much of the diet. In summer, the bear feeds on blackberries (*Rubus* spp.), huckleberries (*Gaylussacia* spp.), blueberries (*Vaccinium* spp.), wild grapes (*Vitis* spp.), red mulberry (*Morus rubra*), hawthorn (*Crataegus* spp.), sassafras (*Sassafras albidum*), pokeberry (*Rivina humilis*), and persimmon (*Diospyros virginiana*; Landers et al. 1979, Beeman and Pelton 1980, Eagle and Pelton 1983, Clark et al. 1987, Pelton 2003). Other foods include saw palmetto berries (*Serenoa repens*), cabbage palm, swamp tupelo (*Nyssa biflora*), black gum fruits, gallberry (*Ilex glabra*), honey bees (*Apis mellifera*) and their honey, yellow jackets (*Vespula* spp.), bumble bees (*Bombus bimaculatus*), and carpenter ants (*Camponotus* spp.; Maehr and Brady 1984, Maehr and DeFazio 1985, Dobey et al. 2005). Oak mast becomes a staple food during the fall and winter, with other foods such as pecans (*Carya* spp.) and hickory nuts consumed as available (Pelton 2001). The bear adapts readily to abundant agricultural crops such as sugarcane (*Saccharum officinarum*), corn (*Zea mays*), and soybeans (*Glycine max*; Jones 2005). Vertebrate prey reported include Virginia opossum (*Didelphis virginiana*), nine-banded armadillo (*Dasypus novemcinctus*), feral pig (*Sus scrofa*), northern raccoon (*Procyon lotor*), and white-tailed deer (*Odocoileus virginianus*; Roof 1997).

ASSOCIATED SPECIES

The American black bear inhabits a diversity of habitats and is associated with numerous mammals in the South. Comparison of diet, habitat use, and movement indicated a low probability for competitive interactions with strict predatory carnivores such as bobcat (*Lynx rufus*) and Florida panther (*Puma concolor*; Maehr 1997). However, competition for food with the coyote (*Canis latrans*) may influence bear numbers in Florida (Maehr 1997). The bobcat, coyote, and adult bear may kill young black bears (Lariviere 2001). The bear is an opportunistic predator that occasionally preys upon young white-tailed deer or newborn livestock (Pelton 2001); however, they are more likely to feed on the carrion of these and other species.

VULNERABILITY AND THREATS

Habitat loss continues to threaten the American black bear. Forest fragmentation and the conversion of forest to agriculture, commercial and urban development, and pine monoculture restricts available habitat or limits additional range expansion (Pelton 2001, Jones

2005). The disjunct nature of black bear populations in the Coastal Plain is exacerbated by several major highway projects (J. Dozier, South Carolina Department of Natural Resources, personal communication) that may eventually result in the loss of genetic diversity (Hellgren and Vaughan 1994). As the human population continues to expand into bear habitat, increased incidents of road mortality are reported in Alabama, North Carolina, Tennessee, and Florida. Advances in hunting techniques and increased access capabilities can result in over-exploitation (Lee and Vaughan 2005). The illegal trade in bear gall bladders used for the alternative medicine markets overseas also is a concern.

Twenty-two different parasites have been reported including lice, fleas, and ticks (Lariviere 2001). Severe cases of mange have been reported in Florida populations (Forrester et al. 1993). A number of neoplastic, rickettsial, viral, and bacterial diseases have been reported (Foster et al. 2004); none appears to contribute greatly to the natural regulation of populations (Cook and Pelton 1979). Few significant diseases are reported for black bears (Pelton 2001). Diseases include fluke fever, rabies, bronchopneumonia, dental caries, osteomyelitis, and periodontal disease (Pelton 2003). Most mortality is human-induced through legal hunting, poaching, depredation control, and collision with vehicles.

MANAGEMENT SUGGESTIONS

The components of black bear management include a mix of hunting access, habitat protection, nuisance control, research, and education. Access can be restricted through road gating, designation of no-hunting zones, and provision of escape cover. Roadless and wilderness area designation on public lands can reduce vehicle-based hunting where it serves as an important management tool to sustain bear numbers. Habitat management includes mast enhancement and provision of key habitat components such as foraging and denning cover (Hellgren and Vaughan 1994, Oli et al. 1997). Management practices that enhance suitable cover in areas of inundation may mitigate for lack of den trees in flood-prone landscapes. The bear has a relatively low biological potential, and changes in food availability can significantly alter population stability, becoming an important consideration for management (Pelton 2003). In the Gulf Coastal Plain, incentive programs encourage the replanting of marginal farmland into bottomland hardwood forest. The provision of dispersal corridors between bear populations has proven successful (Vaughn and Pelton 1995, Pelton 2001, Davidson 2005, Dixon et al. 2006). Sanctuaries established in North Carolina protect female bears and foster population

recruitment (Beringer et al. 1998). Texas has proposed the establishment of bear “recovery zones” through a partnership of federal and state agencies, forest industry, and other landowners. Stringent law enforcement also is required to continue to combat illegal hunting. Additional research can improve monitoring programs, providing accurate predictions of population responses to various perturbations (Pelton and van Manen 1996). Finally, education is essential to successful nuisance control programs, acceptance of regulations such as those that discourage feeding of bears, and for mitigation of human-bear conflicts.

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Elk

Cervus elaphus (Linnaeus, 1758)

David S. Maehr, John J. Cox, and Jeffery L. Larkin

CONTENT AND TAXONOMIC COMMENTS

Although the elk has been historically considered a subspecies of the red deer (*Cervus elaphus elaphus*), recent DNA analyses suggested that the elk is sufficiently divergent from its Eurasian ancestors to warrant specific status as *C. canadensis* (Polziehn and Strobeck 1998). The elk has been historically classified into six subspecies on the basis of morphological characteristics: *C. e. canadensis*, Eastern elk; *C. e. nelsoni*, Rocky Mountain elk; *C. e. manitobensis*, Manitoba elk; *C. e. merriami*, Merriam's elk; *C. e. roosevelti*, Roosevelt elk; and *C. e. nannodes*, Tule elk (Murie 1951). *C. e. canadensis* and *C. e. merriami* are extinct. Mitochondrial analysis supported the recognition of *C. e. nannodes* and *C. e. roosevelti* as valid subspecies, suggesting that *C. e. canadensis* and *C. e. manitobensis* be combined (Polziehn and Strobeck 1998). *C. e. nelsoni* is the only free-ranging subspecies in the South because of its use as a reintroduction surrogate for the extinct eastern elk. The common name, wapiti, is used in technical publications to distinguish the species in North America, avoiding confusion with names for the European moose (O'Gara 2002a).

DISTINGUISHING CHARACTERISTICS

The elk is a large grazer and cursorialist (Geist 1998) with long legs, conspicuous ears, and rump patch. Elk exhibit noticeable sexual dimorphism. Males are typically 20% larger and grow antlers, while females are smaller and rarely grow antlers (Murie 1951). The range of measurements across all subspecies are: total length, 198–262 cm; tail, 9–19 cm; hind foot, 60–74 cm; ear, 18–23 cm; weight, 171–497 kg (Peek 2003). *C. e. nelsoni*, used for reintroduction in the South, typically weighs between 225 (females) and 315 kg (males). The summer adult pelage is reddish to dark brown. The winter coat is dimorphic: males have a tan coat and a dark brown, shaggy neck mane; females have a light brown coat that fades as winter progresses. The head, neck, and lower regions of the body and extremities of both sexes are typically darker than the rest of the body throughout the year. A conspicuous, cream-colored to tan rump patch surrounds a short, stubby tail that is broad at the base and tapers to a marginal fringe. The pelage of juveniles is tan to reddish brown dappled with conspicuous cream to white-colored spots. *C. e. nelsoni* has light and spreading antlers with straight branches.

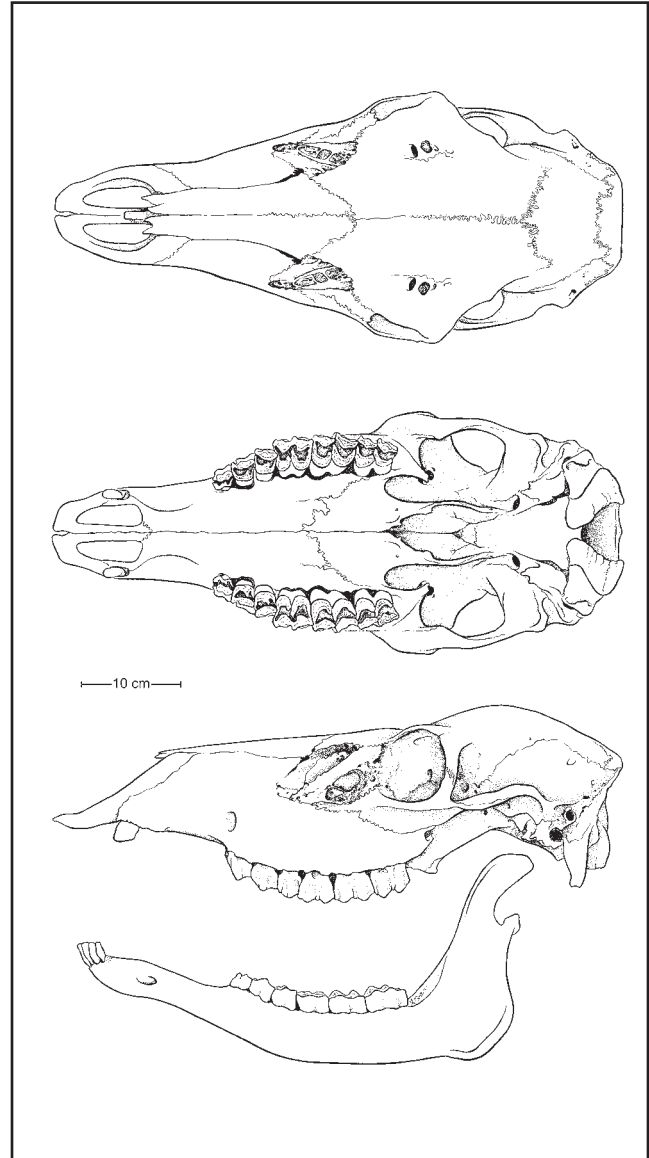


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Cervus elaphus* from Jackson, Wyoming (USNM 265054, female).

This contrasts with other subspecies that can have more curved branches (*C. e. nannodes*) or with *C. e. roosevelti* that supports antlers with a crowning shape with heavy, short branches (Peek 2003). Yearling and adult males begin antler growth between late March–early April. These are typically shed between late February–early April the following year. Yearling elk often grow single spike antlers or have fewer than 4

tines per antler, while adults usually have 5–7 tines per antler. Similar to other ruminants, elk dentition reflects its herbivorous lifestyle. Elk have hypsodont molars and premolars, and the upper incisors are replaced with a dental pad. The dental formula of the elk is: I 0/3, C 1/1, P 3/3, M 3/3 = 34 (Figure 1). See keys for details.

CONSERVATION STATUS

The elk has a global rank of Secure (NatureServe 2007). Overhunting resulted in the extirpation of the eastern subspecies by the middle of the 19th century (Murie 1951). Alabama, Georgia, Louisiana, Oklahoma, South Carolina, and Virginia classify the elk as Presumed Extirpated. It is classified as Critically Imperiled in North Carolina and Imperiled-Vulnerable in Texas. It is not ranked in Florida and Mississippi. Limited hunting is allowed in Arkansas, Kentucky, and Oklahoma where reintroductions have occurred.

DISTRIBUTION

Elk ancestors likely evolved in Eurasia during the Pliocene (O’Gara and Dundas 2002). They appear in the North American fossil record during the Wisconsin glacial stage of the late Pleistocene approximately 40,000 years ago when sea levels were considerably lower and suitable habitat existed between Alaska and Siberia (Guthrie 1966, O’Gara and Dundas 2002). Fossil remains dramatically increased during the Holocene and have been found throughout North America between 30 and 60 degrees latitude (O’Gara and Dundas 2002).

The elk was restored to portions of its historical range during the early twentieth century. Elk were introduced to Alabama and Louisiana in 1916; both efforts failed within a few years (O’Gara and Dundas 2002). Virginia translocated approximately 193 elk between 1917–1935; the animals disappeared following conflicts with agricultural interests and after limited harvests were conducted in the 1960s (O’Gara and Dundas 2002). In 1933, the United States Forest Service introduced elk to the Black Mountain Refuge in Arkansas. During the next two decades, this population grew to approximately 200, and then disappeared due to illegal hunting and habitat loss (Cartwright et al. 2001). Although the elk is not found in the Florida fossil record (Webb 1974), it was introduced to south central Florida in the late 1960s as part of the production of a television series, and is believed to persist in this area (Layne 1993). The current status of the Florida herd is unknown.

Arkansas, Kentucky, Oklahoma, and Tennessee have established populations of *C. e. nelsoni* through

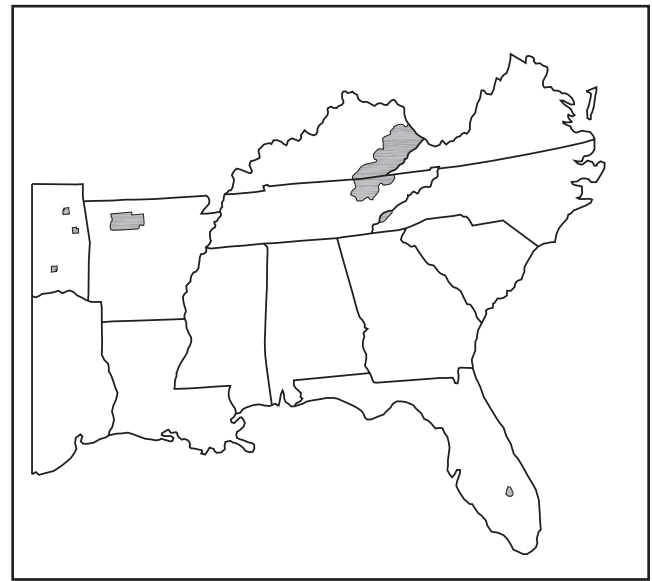


Figure 2. Distribution of *Cervus elaphus* in the South.

translocation efforts (Figure 2). Between 1969–1972, 391 elk were translocated into 6 wildlife management areas in eastern Oklahoma. They still inhabit the Cookson Hills, Spavinaw, and Pushmataha Wildlife Management Areas (Walter and Leslie 2002). Between 1981–1985, the Arkansas Game and Fish Commission in cooperation with Newton County citizens translocated 112 elk from Colorado and Nebraska to 5 sites on or adjacent to the Buffalo National River. Although elk have been reported in surrounding counties, most occur along the upper and middle Buffalo National River corridor in Newton and Searcy counties, and on private land in Boone and Carroll counties (Cartwright et al. 2001). The Kentucky Department of Fish and Wildlife Resources translocated 1543 elk from 6 western source states to a 14-county zone in southeastern Kentucky from 1997–2002. This area was expanded to include McCreary and Whitley counties during 2003 to make the zone contiguous with the Tennessee elk restoration zone. The Tennessee Wildlife Resources Agency translocated 167 elk from Elk Island National Park in Alberta, Canada and Land between the Lakes National Recreation Area to a 5-county zone in northeastern Tennessee during 2000–2003. The National Park Service translocated 52 elk from Land between the Lakes to the North Carolina portion of Great Smoky Mountains National Park during 2001–2002; this herd is considered an experimental release (J. Dobey, University of Tennessee, personal communication). Several other states have considered elk reintroduction (McClafferty and Parkhurst 2001, J. Zysik and W. Porter, Syracuse University, personal communication); however, concerns about chronic wasting disease, human safety, and crop depredation have halted attempts to do so.

An unknown number of elk have moved from Kentucky to Virginia, West Virginia, and Tennessee, and may have established there. Virginia allows the harvest of elk during the white-tailed deer (*Odocoileus virginianus*) season to reduce the chances that an elk population will become established. For the most part, however, free-ranging elk primarily occur in or near designated zones where they have been reintroduced.

ABUNDANCE STATUS

The pre-European elk population in North America has been estimated at 10 million (O’Gara 2002b). Elk populations increased from 90,000 in the early 1900s (Seton 1953) to nearly 1 million at the end of the century (Bunnell 1997). In the South, elk populations have increased wherever they have been recently reintroduced. Presently, the reintroduced populations in Arkansas are estimated at 400–450 (M. Cartwright, Arkansas Game and Fish Commission, personal communication). In Kentucky, the estimate is over 5000 animals. Elk populations in Tennessee (Mueller et al. 2004) and Great Smoky Mountains National Park are conservatively estimated at 165 (L. Mueller, University of Tennessee, personal communication) and 55 animals (J. Dobey, University of Tennessee, personal communication), respectively. Approximately 160 elk occur among 3 wildlife management areas in eastern Oklahoma. The number in Florida is unknown, but likely less than 30.

PRIMARY HABITATS

Elk in North America occupied a wide range of habitats prior to European settlement. In the west, these habitats included temperate coniferous rainforest, non-forested inland valleys, dry forest, chaparral mountains, cool shrub forests, and prairies. In the East, elk also inhabited mixed conifer/hardwood forests; only western deserts and humid ecosystems of the South were unoccupied (Skovlin et al. 2002). A specific pattern of habitat use in the South prior to settlement is speculation due to dramatic changes in the structure and species composition of the region’s landscapes. Grassland savannas, such as the Bluegrass Region of Kentucky (Wharton and Barbour 1991), and river floodplains were likely important as permanent habitat and as movement corridors through this forested region.

Reintroduced elk in Kentucky use forested landscapes with large (up to 5000 ha) openings created by strip mining and subsequent reclamation (Larkin et al. 2001, 2004; Wichrowski 2001, Cox 2003, Seward 2003). Home range size varies from 9–276 km² and increases with the percentage of available contiguous

openings (Cox 2003). Smaller home ranges in areas dominated by forest may be attributed to high concentrations of palatable forage (Cox 2003). Kentucky elk do not migrate in response to snowfall and food availability; little seasonal variation in habitat use has been observed. This population primarily uses forested ridgetops and upper side slopes for resting and ruminating during the day, and grassy openings and shrubby edge in floodplains or reclaimed surface mines for feeding at crepuscular and nocturnal hours (Wichrowski 2001). Cox (2003) found that Kentucky elk preferred mined habitats whenever it was available. Elk use of grasslands and edge habitats is also reflected in the foods they consume (see Food Habits). The small elk population in south central Florida has been observed to utilize sawgrass marsh, cabbage palm-live oak (*Sabal palmetto-Quercus virginiana*) hammock, and dry palmetto prairie (Layne 1993). Elk are capable of tolerating a variety of habitats, development, and human disturbance in order to meet their nutritional and reproductive needs.

REPRODUCTION

Like other North American cervids, elk are seasonal breeders (Raedeke et al. 2002). In Kentucky, peak breeding occurs between mid-September and mid-October, which is consistent with western elk populations. However, breeding behavior often begins in mid-August and can extend into early December (Larkin et al. 2002). During the breeding season (rut), bulls compete for mating opportunities via antler display, bugling, and sparring. Elk are polygynous; each dominant bull maintains a group of reproductively active cows in a harem. Conception dates are influenced by several factors including forage quality and quantity, bull age, and cow condition (Trainer 1971, Noyes et al. 1996, Larkin et al. 2002). The rut is usually longer and conception dates later when yearling bulls do the majority of the breeding. Larkin et al. (2002) reported that calving season length was reduced from 67 days when yearling bulls dominated the breeding, to 37 days when adult bulls were responsible for most of the breeding. Once females come into estrous they are receptive to males for 12–24 hours (Frandsen and Whitten 1981, Raedeke et al. 2002). If insemination does not occur during this period, the cow will cycle into estrous every 21–22 days (Raedeke et al. 2002). As many as 6 recurrent estrus have been reported (Wishart 1981). Cows between 2.5–7.5 years contribute the most to reproduction (Raedeke et al. 2002, Larkin et al. 2003b). However, yearling cows in peak physical condition can become pregnant. Larkin et al. (2003b) reported a 50% calving rate for yearling cows in the reintroduced Kentucky herd. Gestation ranges from 247–268 days (Raedeke et al. 2002). Cows usually

give birth to one calf; twinning occasionally occurs (Larkin et al. 2003b). Most calves are born between the last week of May and first week of June (Larkin et al. 2003b, Seward 2003). The earliest and latest documented births in Kentucky were 13 May and on or about 5 October, respectively. Prior to parturition, pregnant cows isolate themselves by moving several kilometers from the herd and then restricting their movements (Seward 2003). In Kentucky, parturition sites were within 150 meters of the interface between remnant forests and reclaimed surface mine grasslands (Seward 2003). These areas provided security and thermal cover, adequate forage, and were within 180 meters of a water source (Seward 2003). The cow and the neonate calf will usually rejoin the herd when the calf is approximately 7–10 days old.

FOOD HABITS

Elk are intermediate feeders that consume a variety of species depending on season and availability (Murie 1951, Hofman 1985). Although nutrition can be a primary influence on the distribution, abundance, and productivity of elk, the influence of dietary quality on herd dynamics is poorly understood (Cook 2002), especially in the South. The Kentucky herd has twinning rates that are higher than expected, and individuals not infected by the meningeal worm (*Parelaphostrongylus tenuis*) appear to reach adult weight quickly. This suggests that nutrition is not limiting in the central Appalachians. In eastern Kentucky, common summer foods include red clover (*Trifolium pratense*), serecea (*Lespedeza cuneata*), brome grass (*Bromus* spp.), black locust (*Robinia pseudoacacia*), fescue (*Festuca arundinacea*), ragweed (*Ambrosia artemisiifolia*), broom-sedge (*Andropogon virginicus*), autumn olive (*Elaeagnus umbellata*), square-stemmed rose pink (*Sabatia angularis*), rye grass (*Lolium perenne*), and sourwood (*Oxydendrum arboreum*; J. Schneider, Morehead State University, unpublished data). The winter diet is less diverse and includes rye grass, silverberry, fescue, orchard grass (*Dactyli glomerata*), Christmas fern (*Polystichum acrostichoides*), serecea, and red clover. Elk also consume cane (*Arundinaria gigantea*), big bluestem (*Andropogon gerardii*), wheat (*Triticum aestivum*), switch grass (*Panicum virgatum*), sorghum (*Sorghum* spp.), foxtail (*Setaria* spp.), little bluestem (*Schizachyrium scoparium*), meadow timothy (*Phleum pratense*), corn (*Zea mays*), aster (*Aster* spp.), butterfly pea (*Centrosema virginianum*), goldenrod (*Solidago* spp.), violet (*Viola* spp.), cattail (*Typha* spp.), common pokeweed (*Phytolacca americana*), multi-flora rose (*Rosa multiflora*), blackberry (*Rubus* spp.), bird's foot trefoil (*Lotus corniculatus*), common crown vetch (*Coronilla varia*), eastern white pine (*Pinus strobus*), American holly (*Ilex opaca*), red maple (*Acer rubrum*), tulip poplar (*Liriodendron tulipifera*), magnolia

(*Magnolia* spp.), American basswood (*Tilia americana*), common elderberry (*Sambucus canadensis*), white mulberry (*Morus alba*), oak (*Quercus* spp.), hickory (*Carya* spp.), cherry (*Prunus* spp.), Bradford pear (*Pyrus calleryana*), and southern crab-apple (*Malus angustifolia*; Mueller et al. 2004; J. Cox, University of Kentucky, unpublished data). The grazing habits of elk in the central Appalachians reflect a landscape altered by surface mining then reclaimed to large expanses of grassland. Elk food habits elsewhere in its eastern range are poorly documented.

ASSOCIATED SPECIES

The elk is associated with most terrestrial species in the South. Prior to European settlement, the elk was subject to mortality and behavioral modifications caused by large predators such as mountain lion (*Puma concolor*) and gray wolf (*Canis lupus*). The coyote (*C. latrans*) preys on elk calves in the South, while the American black bear (*Ursus americanus*) is a potential predator (Cox 2003; J. Dobey, University of Tennessee, personal communication). The white-tailed deer occurs throughout the occupied elk range in the South, and is the definitive host of the meningeal worm, a nematode that can be fatal in elk (Raskevitz et al. 1991, Cartwright 1995, Walter and Leslie 2002, Cox 2003, Larkin et al. 2003a, Alexy 2004). The absence of this parasite in central Florida may help to explain the persistence of elk in this area (Forrester 1992, Layne 1993). White-tailed deer are typically subordinate to elk when using a similar resource; close interactions without aggression are common (Cox 2003). Elk browsing and grazing promotes vegetative conditions that benefit songbirds that nest in woodland edges such as the yellow-breasted chat (*Icteria virens*) and grassland birds such as the grasshopper sparrow (*Ammodramus savannarum*; Ciuzio 2002). Elk have also benefited other bird species. The common raven (*Corvus corax*), a state-endangered bird in Kentucky, scavenges on elk carcasses (Cox et al. 2003); the eastern phoebe (*Sayornis phoebe*) uses elk hair as nesting material (Seward et al. 2005). Dense populations of elk have the potential to alter soil and groundcover conditions that may subsequently alter successional patterns of forested plant communities (Ter Beest 2005), and the habitat of forest amphibians (Secrist et al. 2004).

VULNERABILITY AND THREATS

The elk has a high probability of persisting in areas of its former range (Griffith et al. 1989). Although the elk is vulnerable to the kinds of pressures that exterminated it from the East in the 19th century, these pressures are mostly absent today. Support for reintroduced populations (Maehr et al. 1999), controlled

hunts, and a landscape mosaic of second growth forest and reclaimed grasslands bode well for the persistence of elk in the South. However, the failure of introduced herds in Alabama, Louisiana, and Virginia suggest that legal harvest has the potential to cause population declines that lead to local extirpation. Disease and poaching inhibit population growth in small, isolated herds (Walter and Leslie 2002, Mueller et al. 2004). Despite these factors, habitat conditions appear suitable for elk over most of its former range. The future of the elk relies upon maintenance of landscape connectivity that ensures sufficient reproduction and genetic health among subpopulations, habitat mosaics that the species prefers, hunting seasons that assure stable or expanding herds, and continued public support.

Continued research will examine the role of the meningeal worm in elk population dynamics, a factor that may be a consideration for managers where climatic conditions facilitate high rates of infection throughout the year (Larkin et al. 2003a, Alexy 2004). Managers should also continue to monitor the epidemiology of chronic wasting disease in the eastern United States where it has caused widespread public concern, economic damage, and localized population declines of white-tailed deer because of eradication efforts intended to eliminate the disease. An outbreak of chronic wasting disease has the potential to jeopardize all reintroduced elk populations in the region; the smaller populations in Arkansas, Florida, Oklahoma, Tennessee, and the Great Smoky Mountain National Park are of special concern.

MANAGEMENT SUGGESTIONS

The elk appears to be well-adapted to a wide range of habitat conditions in the South. The challenge for management agencies is the reduction of human-elk conflicts, and the maintenance of ecologically-based hunting seasons. These issues are not unrelated. In the absence of free-ranging large predators, suitable harvest strategies will be necessary to prevent elk from becoming an economic and public relations burden in agricultural lands and the urban-wildland interface. This requires consistent population monitoring of a species that has considerable dispersal abilities and a tolerance for human activities and cultural landscapes. Management agencies should develop monitoring protocols that permit the estimation of population sizes and trends. Such estimates will be critical in driving adaptive management that is sensitive to subtle changes in the demographics and distribution of this adaptable species. Future management of elk habitat in the east will consider the complex arrangement of vegetation, terrain, access, hunting regulations, and land owner

intentions (Lyon and Christensen 2002). In addition, the potential for infection by brucellosis and chronic wasting disease requires the annual monitoring of animals handled during research and harvest to assure the public that southern herds remain disease free, and a rapid response to the discovery of infection should it occur. Finally, natural resource managers should carefully weigh the promotion of elk restoration in the face of declining forest biodiversity. While the return of this generalist herbivore has increased species diversity in the region, the changes associated with its preferred habitats have occurred with concomitant declines in other species.

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White-tailed Deer

Odocoileus virginianus (Zimmerman, 1780)

Margaret K. Trani and Brian R. Chapman

CONTENT AND TAXONOMIC COMMENTS

Since the original description of the species, taxonomists have assigned 12 different names to the North American deer (Baker 1984, Smith 1991). Although Hershkovitz (1948) proved that the name *Dama* had priority over *Odocoileus*, the International Commission on Zoological Nomenclature used its plenary powers to validate *Dama* as the generic name for the fallow deer of Europe, rejecting it for the North American deer species (China 1960). Consequently, *Odocoileus* became available for the North American deer. *Odocoileus virginianus* includes 30 recognized subspecies in North and Central America and eight in South America (Hall 1981, Baker 1984, Smith 1991). Eleven subspecies occur in the South: *O. v. clavium*; *O. v. hiltonensis*; *O. v. macrourus*; *O. v. mcilhennyi*; *O. v. nigribarbis*; *O. v. osceola*; *O. v. seminolus*; *O. v. taurinsulae*; *O. v. texanus*; *O. v. venatorius*; and *O. v. virginianus*. Smith (1991), Gerlach et al. (1994), and Miller and Marchinton (1995) review the life history.

DISTINGUISHING CHARACTERISTICS

The white-tailed deer is a large, long-legged ungulate with conspicuous ears. Measurements are: total length, 134–206 cm; tail, 15–33 cm; hind foot, 36–52 cm; ear, 14–22 cm; weight, 25–150 kg. The dorsal pelage is light brownish gray to reddish brown. There is a white band around the eye and muzzle; a white throat patch is present. The ventral pelage, insides of the legs, and linings of the ears are white. The dorsal pelage of juveniles is tan to reddish brown and dappled with conspicuous white spots. The tail of the adult, often carried erect when the animal is disturbed or running, is broad at the base and brown dorsally with a white marginal fringe and venter. Females are approximately 25% smaller than males. Adult males carry antlers, which begin growth in late April and are shed January–March. The dental formula is: I 0/3, C 0/1, P 3/3, M 3/3 = 32 (Figure 1). See keys for details.

The endangered Key deer (*O. v. clavium*) is much smaller than its mainland counterpart; maximum shoulder height is 76 cm. Females weigh up to 28 kg, while males may reach 36 kg (Whitaker and Hamilton 1998). The skull is as broad as that of mainland deer (i.e., greatest width of the skull across the orbits

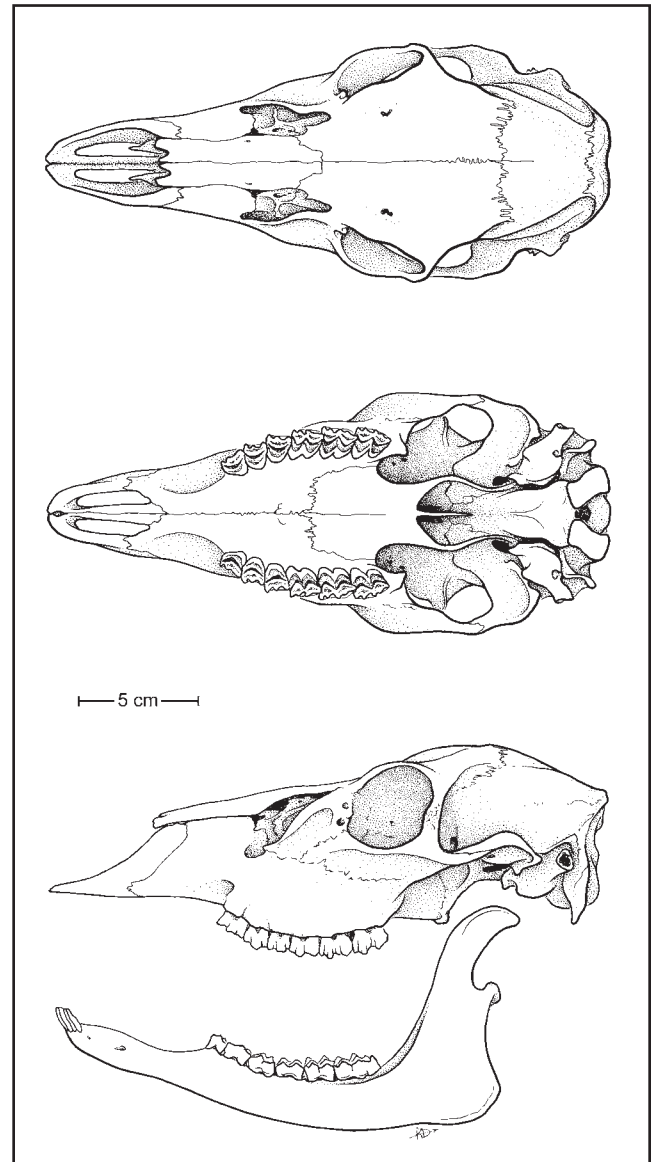


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Odocoileus virginianus* from Swanquarter Wildlife Refuge, Pamlico County, North Carolina (USNM 266377, female).

is 90–119 mm), but the molariform tooth row is shorter; the maximum length is 66 mm (Lazell 1989). Key deer also have proportionately longer tails, and although variable in color, they do not have the summer red and winter gray phases that characterize mainland deer.

CONSERVATION STATUS

The white-tailed deer has a global rank of Secure (NatureServe 2007). The species is also considered Secure in those southern states where it occurs with the exception of Arkansas, where it is Apparently Secure. The deer is unranked in South Carolina. Regulated hunting seasons are supported throughout the South.

The U. S. Fish and Wildlife Service (U. S. Department of the Interior 2007) classifies the Key deer as Endangered. Four other subspecies of concern include *O. v. nigribarbis*, found on Sapelo and Blackbeard Islands, Georgia; *O. v. hiltonensis*, known from Hilton Head Island, South Carolina; *O. v. taurinsulae*, known from Bulls Island, South Carolina; and *O. v. venatorius*, found on Hunting Island, South Carolina (Whitaker and Hamilton 1998).

DISTRIBUTION

Odocoileus virginianus ranges from south-central Canada throughout most of the United States and southward into South America (Hall 1981, Smith 1991, Choate et al. 1994). The species is common throughout the southern United States (Figure 2). The deer ranges throughout Virginia (Dueser et al. 1979, Gaudette and Stauffer 1988, Keiper 1990, Handley 1992, Knox 1997, Linzey 1998) and North Carolina (Clark et al. 1985, Webster 1988, Linzey 1995, Murray and Webster 1995). The deer inhabits South Carolina (Webster et al. 1985, Cothran et al. 1991, Caudell and Warren 1997, Scribner et al. 1997), Georgia (Sawyer et al. 1990, Osborne et al. 1992, Ford et al. 1993, 1994), and Florida (Folk and Klimstra 1991, Shea et al. 1992, Smith et al. 1996, Labisky and Fritzen 1998, Labisky et al. 1999). It occurs throughout Alabama (Ivey and Causey 1984, Dyess et al. 1994), Mississippi (Guynn et al. 1983, Jacobson 1984, Jones and Carter 1989), Louisiana (Lowery 1974), and eastern Texas (Schmidly 1983, Davis and Schmidly 1994). The deer ranges across eastern Oklahoma (Carlile and Lowry 1975, Caire et al. 1989), Arkansas (Nelson et al. 1988, Wigley and Garner 1988, Sealander and Heidt 1990, Kluvender et al. 1992), Tennessee (Bratton 1979, Kennedy 1991, Feldhamer et al. 1992), and Kentucky (Davidson et al. 1985, Paris et al. 1991).

The current range of the Key deer includes approximately 26 islands (330 km²) from Big Pine Key to Sugarloaf Key, Florida (U. S. Fish and Wildlife Service 1998). The National Key Deer Refuge and Great White Heron National Wildlife Refuge encompass much of this territory. The type locality is Big Pine Key, which is the center of the deer's range and supports two-thirds of the population.



Figure 2. Distribution of *Odocoileus virginianus* in the South: (1) *O. v. clavium*; (2) *O. v. hiltonensis*; (3) *O. v. macrourus*; (4) *O. v. mcilhennyi*; (5) *O. v. nigribarbis*; (6) *O. v. osceola*; (7) *O. v. seminulus*; (8) *O. v. taurinsulae*; (9) *O. v. texanus*; (10) *O. v. venatorius*; (11) *O. v. virginianus*.

ABUNDANCE STATUS

Deer are widespread and relatively abundant throughout the South, although populations on some islands have declined. Deer numbers have fluctuated dramatically since European settlement due to intensive hunting, widespread agricultural clearing, and other habitat alteration. Populations nationwide plummeted to less than a million animals by 1900 (Dickson 2001). Remnant populations were restricted to rugged mountainous terrain and hardwood swamps (Knox 1997). Decades later, state agencies initiated harvest restrictions and restocking programs (McDonald and Miller 1993). Populations have rebounded during the last several decades due to farm abandonment (Trani et al. 2001), lower hunting pressure, and the extirpation of large predators (Trani 2002). In some locations, populations have risen to the level of pest status.

Flather et al. (1999) present regional trends in white-tailed deer abundance in the Renewable Resources Planning Act (RPA) Wildlife Report. The RPA is a periodic assessment of natural resources on the nation's forests; population estimates and projections originate from state agencies based on species expert surveys. For states reporting on white-tailed deer, populations have increased approximately four-fold since 1975 (2 million–8 million). Currently, deer population estimates exceed one million

animals in Alabama, Georgia, Louisiana, Mississippi, North Carolina, and Texas (Miller 2001). There is concern among state personnel that deer increasingly may become a management problem during the next decade. Seven states expect deer numbers to decline slightly over the next 50 years (Flather et al. 1999). Relative densities of white-tailed deer have been recorded from less than 5.8/km² in marginal habitat to over 11.6/km² in optimal habitat of the South.

PRIMARY HABITATS

The deer uses a wide variety of habitats, and benefits from a mosaic of wetlands, forests, farmland, and early-successional seres. The deer occurs in numerous forest types in the region (Baker 1984, Hardin et al. 1984, Smith 1991, Shea et al. 1992). Important factors affecting habitat use include soil fertility and productivity; in general, the largest deer are found on fertile soils. Soil phosphorus is a useful predictor of potential physiological condition (Miller 2001). The most productive soils (and therefore the high quality habitats) occur in the agricultural areas of the Piedmont, Upper Coastal Plain, and bottomland habitat (Miller 2001). Forage quality and quantity are also important factors. Deer occupy the same home range year after year (Whitaker and Hamilton 1998); they are not territorial (Labisky and Fritzen 1998, Labisky et al. 1999) but will defend bedding sites. Home ranges of individual deer vary from 16–135 ha, although winter ranges may be larger. Bucks usually have greater home ranges than does, and may extend their range during the rut. Home range tends to be larger in open country than in thick vegetation (Whitaker and Hamilton 1998). The degree of fragmentation on a forested landscape may change home range extent (Inglis et al. 1979, Marchinton and Hirth 1984). Seasonal changes in home range are related to food availability.

REPRODUCTION

Females come into estrus in fall (Sauer 1984) and are receptive to males for 72 hours (White et al. 1995). If they are not inseminated, estrus may reoccur every 21–30 days (Knox et al. 1988). Plotka et al. (1977) suggest the deer is capable of estrous cycling until March of the following year; some captive females have had seven estrous periods between October–April (Knox et al. 1988). Hirth (1977) and Miller et al. (1987) describe behaviors associated with mating. The gestation period ranges from 187–222 days (Verme and Ullrey 1984). Many females produce twins, however litter size can range from 1–3. (Note: For the Key deer, twinning is infrequent and triplets have not been documented). Weaning is complete by 10 weeks of age (Marchinton and Hirth 1984).

Summer nutritional stress may reduce lactation, fawn growth, and body weight. In poor mast years, reproduction rates may be low and conception delayed.

FOOD HABITS

Although the white-tailed deer is often characterized as a browser (eating twigs, shoots, and leaves), research across the South has indicated that only a moderate portion of the diet consists of browse (Miller 2001). The deer is an opportunistic herbivore, consuming hard mast forbs, fruits, grasses, flowers, and fungi (Kirkpatrick et al. 1969, Harlow and Hooper 1972). When conditions allow, they are highly selective, choosing the most palatable, succulent, and nutritious portions of plants (Miller 2001). Browse, forbs, and native grasses are consumed during spring. Typical woody browse species include red maple (*Acer rubrum*), American beautyberry (*Callicarpa americana*), Japanese honeysuckle (*Lonicera japonica*), greenbrier (*Smilax* spp.), trumpet creeper (*Campsis radicans*), and Carolina jessamine (*Gelsemium sempervirens*). Herbaceous species include aster (*Aster* spp.), sweet clover (*Melilotus* spp.), white clover (*Trifolium repens*), vetch (*Vicia* spp.), goldenrod (*Solidago* spp.), and wintergreen (*Gaultheria procumbens*). Soft mast consumption peaks during the summer (Harlow and Hooper 1972, Johnson et al. 1995). Foods include blueberry (*Vaccinium* spp.), sumac (*Rhus* spp.), grape (*Vitis* spp.), hawthorn (*Crataegus* spp.), persimmon (*Diospyros virginiana*), plum (*Prunus* spp.), saw palmetto (*Serenoa* spp.), yaupon (*Ilex vomitoria*), and blackberry (*Rubus* spp.).

Hard mast is an important component of the fall and winter diet, often comprising over 70% of the foods consumed. Nutrition, reproduction, weight, and antler characteristics are influenced by acorn availability (Wentworth et al. 1990, Ford et al. 1994). Postnatal survival may decline following years of minimal acorn production. Agricultural grasses are often planted for winter food plots and include wheat (*Triticum aestivum*), oats (*Avena sativa*), and fescue (*Festuca* spp.). Food plots can improve physical condition and reproductive rates (Johnson et al. 1987, Kammermeyer and Moser 1990).

ASSOCIATED SPECIES

The white-tailed deer is associated with numerous vertebrates in the region. Predators include the Florida panther (*Puma concolor*), red wolf (*Canis rufus*), American black bear (*Ursus americanus*), coyote (*C. latrans*), and bobcat (*Lynx rufus*). Most predation by bobcats and coyotes is on young fawns; predation rates are low and insignificant when compared to other causes of mortality. Other than competition for acorns with eastern wild turkey (*Meleagris gallapavo*), squirrels

(*Sciurus* spp.), feral pigs (*Sus scrofa*), and other vertebrates, white-tailed deer do not compete directly for forage with native species (Miller 2001). Wild pigs can severely impact food plot plantings for winter deer food. Grazing of woodlands by domestic livestock can eliminate deer forage, reduce escape cover, and change plant species composition. Overabundant deer populations can have significant effects on forest regeneration (DeCalesta 1997, Stromayer and Warren 1997), and vegetation community stability (Bratton 1979, Miller et al. 1992). This provides the species with the ability to influence both plant and animal communities wherever it occurs.

The Key deer occurs in association with the Lower Keys marsh rabbit (*Sylvilagus palustris hefneri*) and silver rice rat (*Oryzomys argentatus*). These species make use of similar habitats including salt marshes, transitional areas, and fresh-water marshes (U. S. Fish and Wildlife Service 1998).

VULNERABILITY AND THREATS

There are no threats to the survival of the white-tailed deer in the region. The deer is susceptible to a number of diseases (such as hemorrhagic and chronic wasting disease) and is host to over 100 species of parasites including ticks, keds, lice, and bot flies (Miller 2001). Extensive networks of roads can have negative impacts on white-tailed deer (Graham 2002), stemming from increased hunter accessibility and vehicular mortality. Increases in the number of roads and vehicles are factors contributing to deer-vehicle collisions where deer density is high.

Populations of Key deer have improved in response to decreased illegal hunting (Lopez et al. 2004) and the establishment of a federal refuge. Urban development and habitat fragmentation continue to threaten the Key deer's recovery (Lopez et al. 2003). Fencing restricts deer movements, creating bottlenecks that interfere with the ability to reach permanent water and feeding areas (White et al. 1998). Highway mortality accounts for the majority of total deer mortality. Fire suppression has resulted in the deterioration of important rockland communities.

MANAGEMENT SUGGESTIONS

Mainland deer management centers on population control and habitat enhancement to maintain ecological balance. Regulated annual harvests are the primary tool of population management; harvest guidelines target specific sex and age classes to maintain populations within the carrying capacity of the habitat. Management programs differ across the region because habitat quality and population

dynamics vary dramatically between states. The overabundance of deer populations has become so prevalent, it represents an important wildlife management problem during the current decade.

Forested landscapes with a mosaic of habitat types and seral stages benefit white-tailed deer. Early successional communities provide herbaceous forage and soft mast, whereas late successional forests provide fall and winter mast and limited browse (Miller 2001). Timber harvest and prescribed fire create a diversity of age classes, providing forage and cover (Ford et al. 1993). In pine stands, fire can increase browse production, palatability, and nutrition. Burning in late winter or early spring promotes resprouting of vegetation in the warmer months. Stand thinning can enhance understory browse and release mast-producing trees. The maintenance of mature oak-pine stands is also beneficial. Food plots can be an important management tool; choice of food plantings target the winter and late summer nutritional stress periods. The contribution of agricultural food plots is reviewed by Kammermeyer and Thackston (1995) and Weeks (1995). Management of the endangered Key deer centers on the acquisition of additional land (U. S. Fish and Wildlife Service 1998).

Management and restoration of habitat includes prescribed burning, mowing clearings, planting native vegetation, removing exotic species, providing roadway bypasses, and protecting travel corridors. The use of prescribed fire as a management tool is hampered by urbanization. A low reproductive potential, coupled with intense residential and commercial development, suggests management and recovery of the Key deer will be difficult.

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Feral Pig

Sus scrofa (Linnaeus, 1758)

Brian R. Chapman and Margaret K. Trani

CONTENT AND TAXONOMIC COMMENTS

The feral pig (*Sus scrofa*) is native to northern Africa, Europe and Asia. The species was first introduced into the West Indies by Columbus in 1493 (Groves 1981) and Florida by DeSoto in 1593 (Gipson et al. 1998). Populations that now occur in the United States represent hybrid combinations of the European wild boar (*S. s. scrofa*) and domestic pig (*S. s. domesticus*; Rary et al. 1968, Wood and Lynn 1977). Common names include feral hog, feral swine, razor-back, wild boar, and wild hog. Sweeney and Sweeney (1982) and Mayer and Brisbin (1991) review the life history of the feral pig in the United States.

DISTINGUISHING CHARACTERISTICS

The feral pig is a large, hoofed mammal with a flattened snout. Measurements are: total length, 153–240 cm; tail, 21–38 cm; hind foot, 23–35 cm; ear, 24–26 cm; weight, 66–272 kg. Male pigs are somewhat larger than females; these differences are evident at 15 months and increase with age (Dickson et al. 2001). The most common pelage color is black but considerable variation is present. Some are brown, reddish brown, spotted black or brown, black and white, or all white. The pelage is usually coarse and dense. The elongated, mobile snout has a flattened terminal surface punctuated by terminal nares. The feet have four toes terminating in hooves, but the weight is borne by the two larger, central digits. The upper canine teeth are triangular in cross section, recurved, and visible externally as tusks; they are larger than the lower canines. The dental formula is: I 3/3, C 1/1, P 4/4, M 3/3 = 44 (Figure 1). A complete description of morphological variation is found in Mayer and Brisbin (1991). The only other species in the South that resembles the feral pig is the collared peccary or javelina (*Pecari tajacu*) in southern Texas. See keys for details.

CONSERVATION STATUS

The feral pig has a global rank of Secure (NatureServe 2007). A conservation status rank of Not Applicable has been assigned by each state in the South where it occurs; the species is not a suitable target for conservation activities. In many areas, the pig is considered an important recreational resource as a big game species. Florida, North Carolina, Texas, Tennessee, and

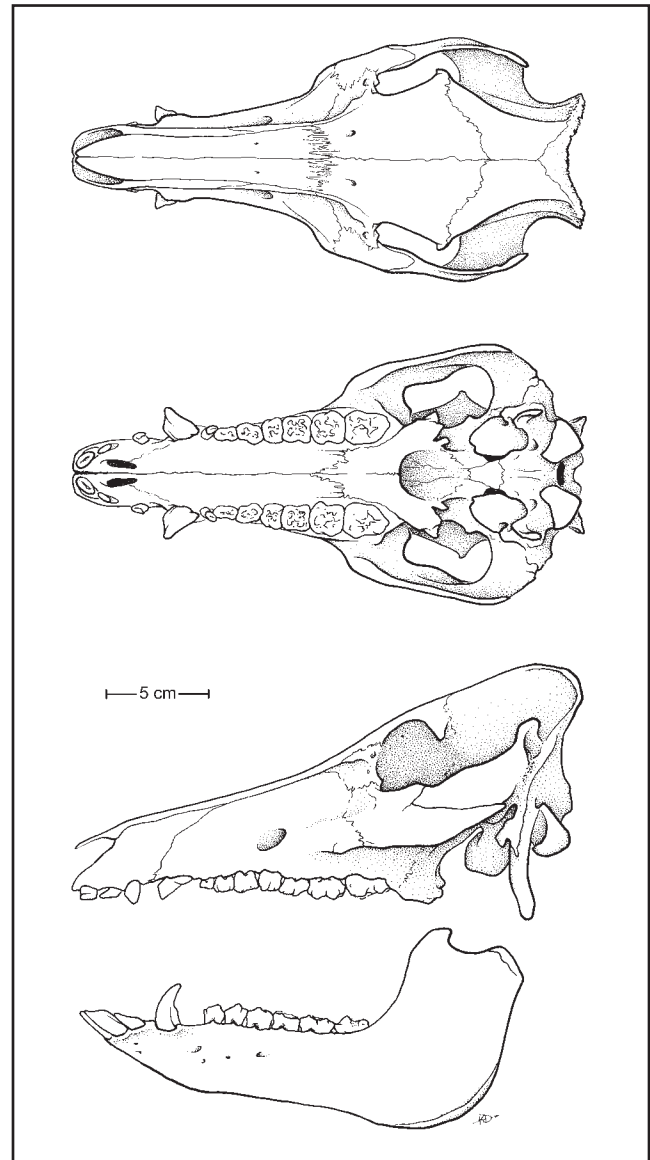


Figure 1. Dorsal, ventral, and lateral view of cranium and lateral view of mandible of *Sus scrofa* from Beaufort County, South Carolina (USNM 256035, male).

other southern states allow hunting during specified seasons (Whitaker and Hamilton 1998).

DISTRIBUTION

The feral pig is found in California, Arizona, New Mexico, Missouri and throughout the southern United States in scattered locations (Sweeney and

Sweeney 1982, Gipson et al. 1998). Figure 2 depicts the distribution of the pig in the South. Once widespread and numerous in Virginia (Bailey 1946, Handley 1979, Linzey 1998), the species now occurs only in the southeastern corner of the state (Mayer and Brisbin 1991) and along the coast (Duncan and Schwab 1986). The feral pig occurs along the coast and barrier islands of North Carolina (Whitaker and Hamilton 1998) and within the Great Smoky Mountain National Park (Johnston 1967, Bratton et al. 1982, Johnson et al. 1982, Lacki and Lancia 1986, Linzey 1995, Lancia et al. 1996). Populations in South Carolina occur along the Savannah River and the Coastal Plain (Golley 1966, Kurz and Marchinton 1972, Schacher and Pelton 1979, Wood and Brennenman 1980, Wood and Roark 1980, Lipscomb 1989, Cothran et al. 1991). In Georgia, disjunct populations occur on barrier islands (Warren and Ford 1997) and throughout the Coastal Plain, Piedmont, and southern Appalachians (Golley 1962, Laerm et al. 1981). It occurs widely throughout Florida (Hamilton 1941, Layne 1974, Maehr et al. 1990). The feral pig inhabits lowland areas in southwestern Alabama (Mayer and Brisbin 1991). In Mississippi, it occurs on coastal islands and in the southeastern corner and south-central part of the state (Baron 1982, Jones and Carter 1989). Isolated populations occur in many areas of Louisiana (Lowery 1974, Mayer and Brisbin 1991), eastern Texas (Davis and Schmidly 1994), and southeastern Oklahoma (Caire et al. 1989, Mayer and Brisbin 1991). Populations are discontinuous in southern Arkansas (Sealander and Heidt 1990). There are feral pigs in Tennessee in the Blue Ridge (Henry and Conley 1972, Singer et al. 1981, Johnson et al. 1982), Cumberland Plateau, and along the Mississippi River (Rary et al. 1968, Lacki and Lancia 1986, Kennedy 1991, Linzey 1995). In Kentucky, they primarily occur in the southeastern corner of the state (Mayer and Brisbin 1991, Whitaker and Hamilton 1998).

ABUNDANCE STATUS

There are few estimates of feral pig populations in the South. An estimated 1500 pigs roam the Cherokee National Forest and adjacent lands; there are approximately 800 individuals in the Great Smoky National Park (Whitaker and Hamilton 1998). It is estimated that 800 inhabit western North Carolina where 100 are taken annually by hunters. In Kentucky, an estimated 300 are found in the Cumberland Plateau (Whitaker and Hamilton 1998). In Texas, there have been recorded densities of 9.5/km² (Ilse and Hellgren 1995). Home ranges of 123–799 ha have been reported; however, the ranges of sows during the farrowing period averaged between 17–30 ha (Whitaker and Hamilton 1998). According to a survey conducted by

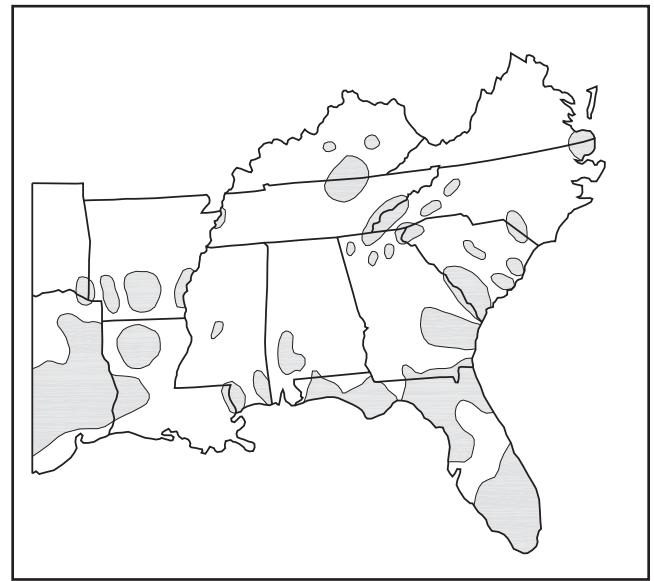


Figure 2. Distribution of *Sus scrofa* in the South.

the Southeastern Cooperative Wildlife Disease Study (SCWDS 1988), most states in the region have scattered populations of feral pigs with densities greater than 3.9/km². Landscape pattern (i.e., degree of fragmentation) may be a determinant of feral pig abundance in patchy landscapes (Virgos 2002).

PRIMARY HABITATS

In the South, the pig occurs in a variety of habitat types including bottomland forests, brushlands, dry ridges, swamps, and marshes. In mountain habitats, feral pigs use mixed hardwood forests associated with a permanent water source (Lacki and Lancia 1986). In Texas, preferred habitat is open brush-savanna (Ilse and Hellgren 1995). The pig requires adequate cover, both for protection from disturbance and to help reduce heat loss (Whitaker and Hamilton 1998). Seasonal changes in habitat use are related to food availability and dietary shifts (Dickson et al. 2001).

REPRODUCTION

The species matures at an early age and breeds throughout the year (Sweeney et al. 1979). In Tennessee and South Carolina, young are born during January–February and May–June (Henry 1966, Sweeney et al. 1979, Warren and Ford 1997, Taylor et al. 1998). At farrowing time, the female prepares a nest of vegetation (e. g., pine straw and broom sedge) within a shallow depression. The gestation period averages 112–115 days (Whitaker and Hamilton 1998); litter size ranges from 1–16 with a mean between 5–6 (Dickson et al. 2001). Reproductive success is influenced by food availability, particularly annual mast crops. The piglets remain with the sow until they

weigh approximately 25–35 kg (Kurz and Marchinton 1972). Males usually reach puberty within 5–7 months; females attain puberty at 10 months (Sweeney et al. 1979). The pig reaches full growth at 5–6 years of age.

FOOD HABITS

Sus scrofa is an opportunistic species (Henry and Conley 1972, Wood and Roark 1980, Warren and Ford 1997). In South Carolina, grasses and other herbaceous material represent 51% of the diet in spring and 9.5–35.8% during other seasons (Roark 1977). Fruits (e.g., berries) are an important component of the summer and fall diet; acorns and hickory nuts are important foods during fall and winter. Stems, leaves, roots, tubers, forbs, and mushrooms are also consumed. Insect larvae, snails, centipedes, and earthworms are included in the diet. Other components include reptiles, birds, small mammals, crustaceans, and the eggs of ground nesting birds such as ruffed grouse (*Bonasa umbellus*), wild turkey (*Meleagris gallopavo*), and northern bobwhite (*Colinus virginianus*). Carrion is also consumed (Roark 1977). On coastal islands, the species will consume the eggs and hatchlings of loggerhead (*Caretta caretta*) and green sea (*Chelonia mydas*) turtles. The pig will prey on newborn lambs, kids, and calves as well as adult sheep and goats (Dickson et al. 2001). The opportunistic food habits of this species often result in the depredation of a variety of commercial grain and vegetable crops.

ASSOCIATED SPECIES

The feral pig competes for mast foods with white-tailed deer (*Odocoileus virginianus*) and several other species of native wildlife (Wood and Lynn 1977). The species may out-compete the sympatric peccary, resulting in declines of density, herd size, and home range of the peccary (Ilse and Hellgren 1995, Gabor and Hellgren 2000). The pig is preyed upon by American alligator (*Alligator mississippiensis*), bobcat (*Lynx rufus*), American black bear (*Ursus americanus*), coyote (*Canis latrans*), and Florida panther (*Puma concolor*; Maehr et al. 1990).

VULNERABILITY AND THREATS

The feral pig is in no danger of extirpation in the region. In many states, the species can be hunted throughout the year on private land (Mayer and Brisbin 1991). In some national parks, natural areas, and other public lands, active removal programs have been implemented to reduce feral pig populations. Additional research is needed on the

competitive relationship between the pig and the native species occurring in its habitat.

MANAGEMENT SUGGESTIONS

Management of the feral pig centers on long-term control programs aimed at reducing population size. The species is very prolific and becomes wary with hunting pressure; once populations are established, they are difficult to control (Dickson et al. 2001). Control techniques include fencing, snares, trapping, shooting, and hunting with dogs. Feral pigs can cause extensive environmental degradation (Bratton et al. 1982, Lacki and Lancia 1986, Lipscomb 1989). Many agencies consider the species to be a serious economic pest due to its feeding habits (depredation on agricultural crops and forest plantations) and behavior. Wallowing can degrade water quality via siltation and contamination of riparian streams. Rooting may cause environmental damage in areas where these animals occur, resulting in disruption of soil surface (erosion and destabilization) and damage to vegetation (tree roots, sprouts, and sensitive communities). Because of these problems, further reintroduction of the feral pig into new areas is not recommended (Dickson et al. 2001).

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