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The Forest Ecosystem Study: Background, Rationale, Implementation, Baseline Conditions, and Silvicultural Assessment

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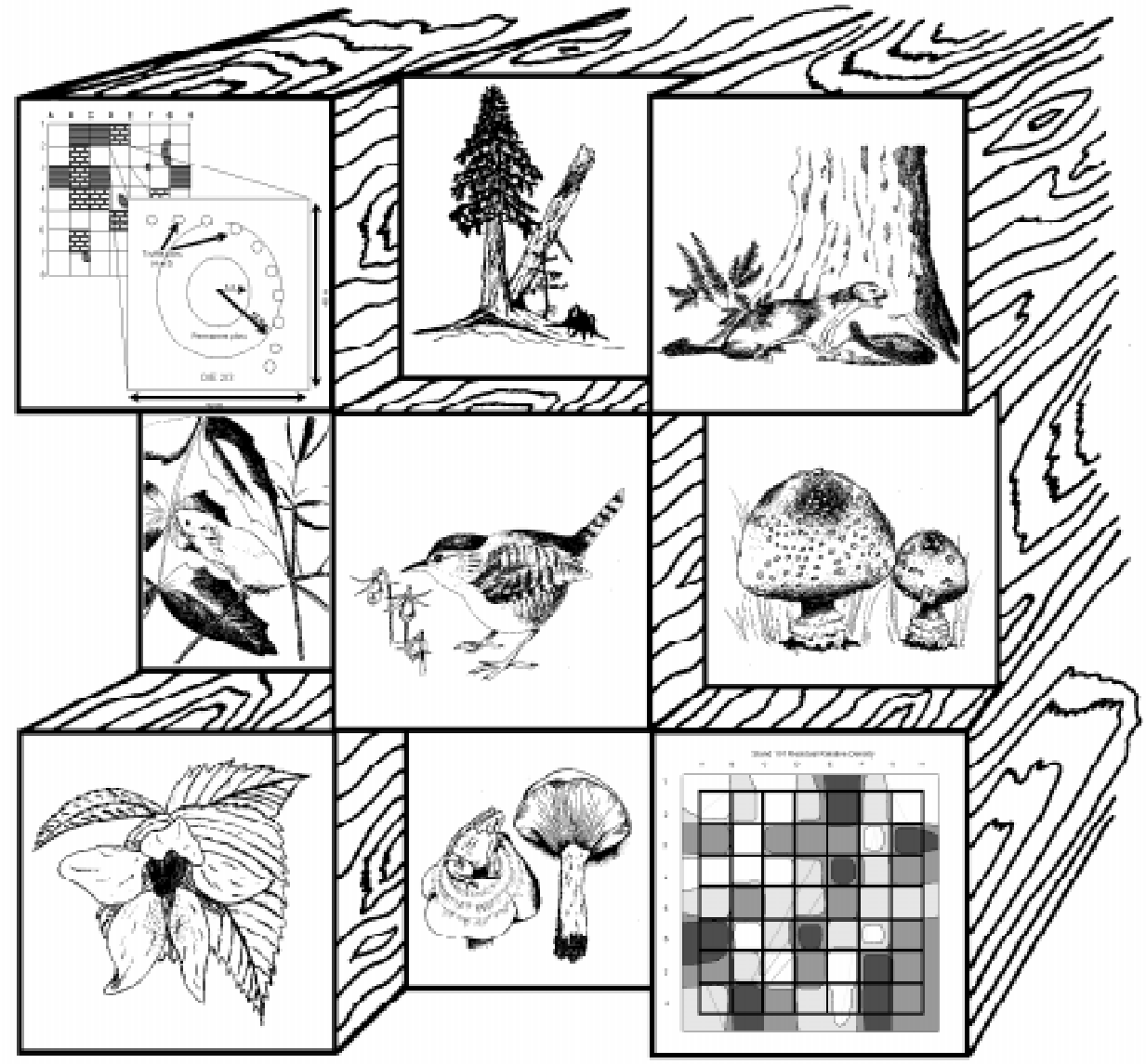

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Abstract

Carey, Andrew B.; Thysell, David R.; Brodie, Angus W. 1999. The Forest Ecosystem Study: background, rationale, implementation, baseline conditions, and silvicultural assessment. Gen. Tech. Rep. PNW-GTR-457. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 129 p.

The Forest Ecosystem Study (FES) came about as an early response to the need for innovative silvicultural methods designed to stimulate development of late-successional attributes in managed forests—a need ensuing from the exceptional and long-standing controversies over old-growth forests and endangered species concerns in the Pacific Northwest. In 1991, scientists with the FES applied experimental, variable-density thinning to even-aged Douglas-fir (*Pseudotsuga menziesii* (Mirbel) Franco) forests on the Fort Lewis Military Reservation in western Washington after having first accumulated extensive baseline data on arboreal rodents, small mammals, trees and other vascular plants, and fungi. Since thinning, further research elements have been incorporated into the FES, including top rot fungal inoculation and soil food web response to thinnings, in addition to the ongoing prey base, vegetation, fungal, and silvicultural assessment investigations. We present study background, rationale, baseline conditions, and selected preliminary responses, as well as a silvicultural assessment of the variable-density thinning.

Keywords: Variable-density thinning, Pacific Northwest, Douglas-fir, biodiversity, northern flying squirrel, truffle, Forest Ecosystem Study, experimental silviculture.

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Introduction

Ancient forests have achieved preeminence in conservation issues in North America because of their dwindling area, value as habitat for threatened species of plants and animals, contributions as reservoirs of biodiversity, and aesthetic qualities. In the Pacific Northwest, old-growth Douglas-fir forests have been at the center of exceptional public controversy because of their importance to spotted owls, marbled murrelets, and other species.[1] This controversy has resulted in numerous appeals of proposed timber sales, production of several environmental impact statements followed by challenges in courts, and finally a succession of three major interagency reports: the Interagency Scientific Committee (ISC) to address the conservation of the northern spotted owl (Thomas and others 1990); the Scientific Analysis Team (SAT; Thomas and others 1993) that addressed other wildlife associated with late seral forests; and the Forest Ecosystem Management Assessment Team (FEMAT 1993) that provided a comprehensive ecological, economic, and social assessment of forest management in the Pacific Northwest.

The opportunity to maintain biodiversity in managed forests and the need to recreate late-seral ecosystems was recognized more than a decade ago (Carey 1985, Ruggiero and Carey 1984, Thomas and others 1990). Thomas and others (1990) called for experimental approaches to forest management to determine if suitable habitat for spotted owls could be created at the same time as commodity values are extracted from managed forests. Substantial information on biodiversity in managed and natural Douglas-fir forests had accumulated, including Carey (1989, 1995), Carey and Johnson (1995), Carey and others (1990, 1992, 1996a), Forsman and others (1984), Ruggiero and others (1991), and Spies (1991). By 1991, sufficient correlational data on biodiversity and elements of forest structure existed to design an experiment in ecosystem manipulation, the Forest Ecosystem Study (FES).

The FES was initiated (Carey and Miller 1991) to address development of spotted owl habitat through experimental manipulation of managed stands. Both silvicultural treatments and direct interventions to enhance biodiversity were incorporated into the experimental design. After a review of potential study locations, the Fort Lewis Military Reservation was chosen because (1) the forests were predominantly Douglas-fir, thus maximizing both potential for positive prey-base responses (Carey 1995) and applicability to the most common forest type in western Washington and Oregon; (2) the forests were 50- to 70-year-old, even-aged, closed-canopy stands regenerated after clearcutting and representative of commonly occurring second growth and thus presented opportunities for ecosystem management; and (3) Fort Lewis had large, forested areas that allowed selection of homogeneous blocks for replicates of treatments that would minimize confounding effects of heterogeneity and forest fragmentation on ecosystem processes.

Following site selection in 1991, a memorandum of understanding (MOU) between the Pacific Northwest Research Station and Fort Lewis was developed that protected the study sites for a minimum of 20 years. In 1992, Fort Lewis became a Designated Conservation Area (DCA) for the northern spotted owl with publication of the final draft

[1] Scientific names, authorities, and sources of all species not provided in the body of the report are given in appendix 1. Where common names have not been recognized or assigned, the scientific name is used throughout.

Table 1—Schedule of Forest Ecosystem Study activities

	Year and quarter[a]																							
	1991				1992				1993				1994				1995				1996			
FES activity	1	2	3	4	1	2	3	4	1	2	3	4	1	2	3	4	1	2	3	4	1	2	3	4
Treatments and assessments:																								
Site selection/MOU		•																						
Grid establishment			•																					
Arboreal rodent trap placement			•																					
Small mammal trap placement						•																•		
Cavity and nestbox installation						•															•			
Silvicultural treatment									•	•														
Underplanting														•										
Top rot inoculation														•										
Silvicultural assessment																				•	•			
Sampling of response variables:																								
Prethinning vegetation				•	•																			
Postthinning vegetation															•								•	
Cavity tree age and growth											•	•			•	•								
Den-site use by flying squirrels									•		•		•		•		•		•		•		•	
Epigeous fungi								•								•		•		•		•		•
Hypogeous fungi (truffles)										•	•	•	•	•	•	•	•	•	•	•		•		
Soil food webs														•		•		•		•				
Prethinning coarse woody debris						•																		
Postthinning coarse woody debris										•								•						
Den-site availability											•													
Arboreal rodent trapping				•		•		•		•		•		•		•		•		•		•		•
Small mammal trapping							•				•				•								•	
Owl survey						•																		
Winter bird survey																					•			

[a] Calendar quarters: 1 = January-March; 2 = April-June; 3 = July-September; 4 = October-December.

of the northern spotted owl recovery plan, because it was “an important location to assist in reestablishing demographic interchange between owls in the Cascade Mountains and owls on the Olympic Peninsula” (Lujan and others 1992).[2] Subsequently, a consultation (under Section 7 of the Endangered Species Act) was undertaken with the Olympia, Washington, office of the U.S. Fish and Wildlife Service, as to the suitability of the proposed habitat manipulation. The experiment was deemed compatible with management of Fort Lewis as a DCA, and thinning operations were performed during January-April 1993. Variable-density thinning (VDT), a cornerstone of our experiment, was a new silvicultural technique (Carey 1994b, Carey and Curtis 1996, Carey and Miller 1991) devised to create spatial heterogeneity in composition and structure that would mimic conditions found in old-growth forests. Supplemental funding that allowed expansion of research was obtained from the U.S. Army and the U.S. Department of Agriculture, National Research Initiative.

In this report, we provide the background, historical context, and objectives of the FES; an outline of the experimental design, implementation schedule (table 1), and sampling methods; pretreatment and baseline conditions; lists of species identified during the project (appendix 1); an assessment of the posttreatment silvicultural conditions; and opportunities for potential research collaboration. We hope this information will be useful to future research collaborators, managers seeking to design or implement adaptive management programs, and readers of related publications who want further detail on some aspect of the study.

Statistics and statistical tests are presented for illustrative purposes only. Because presented data are preliminary, we primarily use the nonparametric Mann-Whitney U-test and Wilcoxon matched-pairs signed-rank test (Norušis 1993) to contrast, respectively, two independent samples or two paired samples. We also use the two-sample t-test where necessary assumptions were met. We recognize many of our data are amenable to the use of analysis of variance (ANOVA), but because this publication is intended as an establishment report rather than a report of research results, such analyses are not presented herein. More refined and detailed analyses and discussions of specific study components will be published in the future.

Study Objectives

The FES is an application of experimental management techniques to second-growth forests. The techniques are intended to hasten development of late-seral “composition, structure, function, ecologic products, and economic products” (Carey 1994a). The study was designed to test whether late-seral forest attributes[3] can be developed from stem-exclusion-phase (Oliver and Larson 1990) forests through silviculture, and whether providing supplemental dens can increase the density of northern flying squirrels (the primary prey of the northern spotted owl) in second-growth forests. The experiment was formulated to address specific applied and basic objectives relative to forest structure, composition, and ecological function.

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[2] Currently, northern spotted owls are not known to occur within Fort Lewis.

[3] Late-seral attributes include large-diameter trees, well-developed understories, high vegetation site type diversity, and associated characteristics of high fungal diversity and abundant small mammal populations.

The applied research objectives of the study are to determine if:

1. Woody plant species diversity, spatial heterogeneity in vegetation, and vertical diversity in vegetation can be manipulated through variable-density thinning and underplanting that do not require replacing the existing stand.
2. Enhancing the growth and diversity of woody plants also will be accompanied by increased abundance and diversity of ectomycorrhizal fungi.[4]
3. Increasing den availability through direct intervention (creating cavities in live trees and adding nest boxes) will increase flying squirrel populations.
4. Inoculation of live trees with stem decay fungi will hasten their use by primary excavators (woodpeckers) and thus hasten the development of cavities useful to other secondary cavity dwellers (including the northern flying squirrel).
5. Increasing flying squirrel numbers and woody plant diversity will then have a synergistic effect in promulgating an abundance and diversity of ectomycorrhizal fungi and concurrently increase forest productivity.
6. Wood production is compatible with advancing late-seral forest conditions.
7. Variable-density thinning compares favorably with traditional commercial thinning in promoting tree growth and yield.

The basic research objectives include:

1. Elucidating the community ecology of ectomycorrhizal fungi in relation to seasonal and annual climatic variability, composition and abundance of the rodent community, composition and abundance of the understory vascular plant community, coarse woody debris, and silvicultural treatments.
2. Evaluating the relative influence of den availability, abundance and diversity of food, abundance of protective cover (understory vegetation), and abundance and diversity of predators on populations of northern flying squirrels, Douglas' squirrels, and Townsend's chipmunks to determine factors limiting sciurid populations and regulating arboreal rodent communities.
3. Elucidating factors controlling composition and abundance of the forest-floor mammal communities, especially the influence of understory vegetation and coarse woody debris.
4. Testing hypotheses about the utility of vertebrate communities as ecosystem indicators. The indices, developed and explained by Carey and others (1996a), include:
   a. biomass of arboreal rodents as an indicator of ecosystem productivity; and
   b. integrity of the forest-floor mammal community as an index of forest-floor function.

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[4] The hypogeous sporocarps of ectomycorrhizal fungi (truffles) are the primary food of the northern flying squirrel. The flying squirrel disseminates the spores of the fungi and serves as the primary prey of the northern spotted owl. The sporocarps also are important food for several other animals (e.g., voles, chipmunks, other squirrels).

The following tabulation indicates which methods will be used to meet specific objectives and the pages on which the methods are discussed:

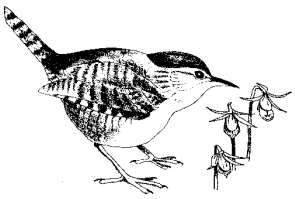
Objective	Method to meet specific objectives (with page nos.)
	Applied
1	Variable-density thinning (11, 26); underplanting (20, 24); vegetation sampling (25, 27).
2	Vegetation sampling (25, 27); fungal sampling (29-31); fecal pellet analysis (33).
3	Supplemental den establishment and use (18, 23, 32); arboreal rodent trapping (22, 32).
4	Top-rot inoculation, monitoring (21, 24).
5	Arboreal rodent trapping (22, 32); vegetation sampling (25, 27); fungal sampling (29-31); fecal pellet analysis (33).
6	Vegetation sampling (25, 27); fungal sampling (29-31); trapping (22, 23, 32, 33).
7	Stand assessment (21, 26); vegetation sampling (25, 27).
	Basic
1	Vegetation sampling (25, 27); fungal sampling (29-31); trapping (22, 23, 32, 33); coarse woody debris sampling (31); fecal pellet analysis (33).
2	Supplemental den establishment and use (18, 23, 32); vegetation sampling (25, 27); fungal sampling (29-31); arboreal rodent trapping (22, 32).
3	Vegetation sampling (25, 27); small mammal trapping (23, 33); coarse woody debris sampling (31).
4	Arboreal rodent and small mammal trapping (22, 23, 32, 33).

The increases in productivity of plants, foliage, fruits, and seeds predicted to result from VDTs should increase abundance and diversity of invertebrates, insectivores, amphibians, birds, and mammals. Many small mammals, especially Townsend's chipmunks, northern flying squirrels, and Douglas' squirrels, are important prey for predators such as weasels, marten, owls (e.g., spotted and great-horned), and raptors (e.g., northern goshawk). Spotted owl abundance, requirements for late-seral forests, and use of second-growth forests differ with prey diversity and total prey biomass (Carey and others 1992, Carey and Peeler 1995). Thus, experimental treatments should affect a hierarchy of ecosystem processes from symbioses between fungi and plant roots to predation at the highest trophic level (the spotted owl, marten, fisher, great-horned owl, and northern goshawk). Response variables representative of this hierarchy of ecosystem biodiversity that will be evaluated during the study include:

1. Composition and diversity of selected soil microbial communities (including fungi, bacteria, and nematodes)
2. Abundance and diversity of hypogeous and epigeous sporocarps of ectomycorrhizal fungi in or on the forest floor
3. Diversity of fungi in the diets of the primarily mycophagous northern flying squirrel and the facultatively mycophagous Townsend's chipmunk and southern red-backed vole
4. Abundance and diversity of understory vegetation
5. Abundance of Townsend's chipmunks and northern flying squirrels
6. Space and den use by northern flying squirrels
7. Diversity and abundance of the forest-floor community of insectivorous, frugivorous, granivorous, herbivorous, and mycophagous small mammals (shrews, shrew-moles, voles, and deer-mice)
8. Frequency of encounter of predators of small mammals (e.g., weasels, owls, hawks)
9. Tree diameter at breast height (d.b.h.), height to live crown, total height, crown width, and growth rates
10. Stand basal area and volume of wood

These response variables comprise the basis for an ecosystem study based on trophic pathways supporting spotted owls and other predators and will provide a template for evaluating the degree to which our applied and basic research objectives are met.

Meeting many FES objectives will be contingent on a thorough understanding of actual postthinning forest conditions, including stand density, relative density, and basal area. The FES is primarily a stand-level experiment, and eventually all response variables will be assessed in relation to actual posttreatment conditions. Some response variables (understory vegetation, hypogeous and epigeous fungi) will further be evaluated in relation to postthinning conditions at a specific plot or at plots of similar thinning intensity. Thus, we incorporated a complete, posttreatment assessment of stand conditions.



Finally, the study site, experimental design, and sampling framework meet our operational objective of creating a foundation and stimulus for interdisciplinary studies of ecosystem processes.

Study Area

Location

The FES is in the Rainier Training Area, Fort Lewis Military Reservation, south of the Nisqually River, approximately 30 km[5] east of Olympia, Washington, in the southern Puget Trough Physiographic Province (Franklin and Dyrness 1973). The present-day landforms and substrate were deposited when the Puget Lobe of the Vashon Stade of the continental glacier retreated 13,000-15,000 years ago (Kruckeberg 1991). Topographic relief is moderate to rolling, with occasional steeper areas surrounding small kettle depressions. The experimental sites contain no year-round creeks, ponds, or wetlands. The study blocks are on glacial uplands, terminal moraines, and glacial till

[5] Metric units are used except where English units were the operational units. A conversion table is given on p. 69.

plains; root penetration into the soil is moderately shallow, and drainage is classified as somewhat excessive (Pringle 1990). The maximum elevation at Fort Lewis is 168 m above sea level; the study blocks range from 120 to 165 m in elevation. Annual precipitation for Fort Lewis is 800-900 mm with only 10 to 15 percent of the annual precipitation falling during the peak growing months of June-September (Pringle 1990). Soils are coarse-textured gravelly and gravelly sandy loams (of glacial till and glacial outwash origin) of Tenino and Everett soil series (Pringle 1990). The excessively drained soils combine with low summer rainfall to create droughty conditions where Douglas-fir typically dominates and also regenerates in the understory (Franklin and Dyrness 1973, Pringle 1990).

Vegetation

Fort Lewis is an area of relatively isolated forests, prairies, and wetlands bounded on the northwest by Puget Sound, and on the north, east, and south by agricultural and urban areas. The forests of Fort Lewis provide the only forested connection between the Black Hills and Olympic Mountains to the west, and the Cascade Range to the east; its unique position is the primary reason for its being a Designated Conservation Area for the northern spotted owl (Lujan and others 1992).

Large portions of Fort Lewis forests were clearcut in the early 20th century and regenerated from natural seeding; the second growth is now actively managed. Some areas contain residual Douglas-fir as legacies from the previous stand. Where present, these large, dominant Douglas-fir project above a second, dense layer of codominant Douglas-fir. Many stands at Fort Lewis are now in the stem exclusion phase of forest development (Oliver and Larson 1990) or competitive exclusion stage of forest ecosystem development (Carey and others 1996a). In this stage of development there are abundant dying suppressed trees, small-diameter fallen trees, and standing dead trees throughout the stands. In contrast, intensively managed stands at Fort Lewis are uniform in tree diameter and canopy height, contain few old-growth legacy trees, contain few small diameter, suppressed, or dead trees, and are in the understory reinitiation stage (Oliver and Larson 1990, Carey and others 1996a).

The FES is divided between two forested tracts that have experienced widely disparate management histories. The two portions of the study are about 3 km apart and are referred to as the Star/Stellar (SS) forest (fig. 1) and the Farley/Hill (FH) forest (fig. 2). The FH forest, comprised of the Farley and Hill blocks, was clearcut in 1925 and, following natural regeneration, was lightly thinned in 1972 and again between 1979 and 1989. During these commercial thinnings, live, recently dead but salvageable, and fallen trees were routinely removed; 3.0-5.8 thousand board feet (MBF) per acre were removed per entry. In 1991 the FH blocks were dominated by 55- to 65-cm d.b.h. Douglas-fir (39-43 m tall) with small amounts of black cottonwood, red alder, and bigleaf maple (3 to 8 percent total stems). Trees in FH appeared to have been grown under moderate stocking levels since stand initiation, with many trees having ample crowns and lateral branches to near ground level. The low understory was well developed and was dominated by salal, California hazel, swordfern, and bracken fern. Other common Douglas-fir series associates (Henderson and others 1989), such as serviceberry, western fescue, baldhip rose, and creeping snowberry, were common.

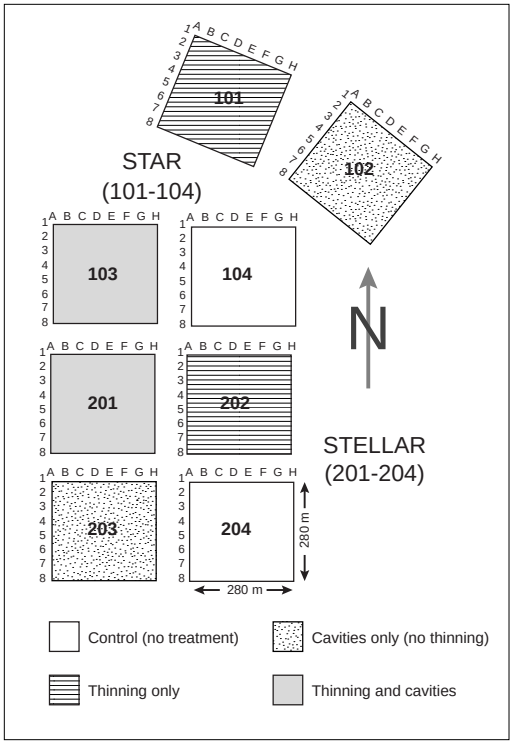


Figure 1—Blocks 1 (Star 101-104) and 2 (Stellar 201-204) show the spatial arrangement of the four main treatments. Each of the eight stands is a 280- by 280-m square, with 80 m between stands.

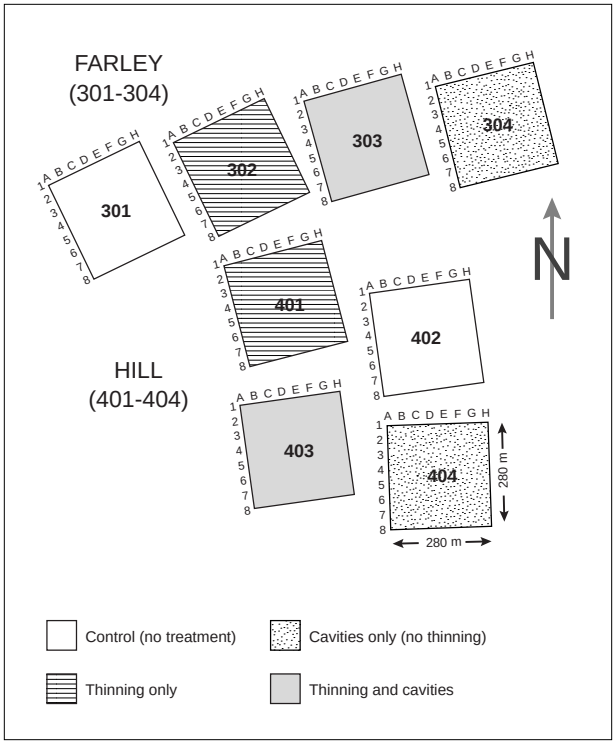


Figure 2—Blocks 3 (Farley 301-304) and 4 (Hill 401-404) show the spatial arrangement of the four main treatments. Each of the eight stands is a 280- by 280-m square, with 80 m between stands.

Though the likelihood of laminated root rot (*Phellinus weirii* [6]) in these blocks was high, actual occurrence was difficult to judge because diseased or infected trees were probably removed during the thinnings. Because of past thinning entries, and because very few trees were left standing during the 1925 harvest, the FH forest had few residual Douglas-fir trees and snags and minimal cover of coarse woody debris (Carey and others 1996b).

The SS forest, consisting of the Star and Stellar blocks, was harvested in 1937 and had not been further manipulated. In 1991, SS was a closed-canopy forest dominated by 30- to 45-cm d.b.h. Douglas-fir (30-35 m tall), with a few western hemlocks, western redcedars, and Pacific yews. Trees killed by suppression and trees with small crowns were abundant, suggesting a very high stocking level since stand initiation. Understory vascular vegetation was sparse, with only small amounts of salal and Cascade Oregongrape. Douglas-fir series associates (Henderson and others 1989), such as serviceberry, western fescue, baldhip rose, and creeping snowberry, also were found in this forest. The forest floor typically was dominated by mosses, particularly *Kindbergia oregana* (Sull.) Ochyra and *Hylocomium splendens* (Hedw.) B.S.G.[7] Exceptions to the high canopy closure and sparse understory were found in areas of laminated root rot infection. In these root rot centers, understory shrubs, forbs, and ferns were well developed. In SS, some Douglas-fir trees and snags were left during the 1937 entry, as was a substantial amount of large coarse woody debris (Carey and others 1996b).

Experimental Design and Rationale

Overview

We used a complete randomized block design of four blocks with four treatments per block, to which were applied the three principle manipulative components of the experiment: (1) a VDT treatment (with three different thinning density subtreatments); (2) den augmentation with cavities and nestboxes; and (3) underplanting to increase species and structural diversity. The 13-ha stands, each containing a 280- by 280-m, 8 by 8 grid (with 40- by 40-m, 0.16-ha grid cells), were the operational units for application of treatments. The four main treatments, each randomly assigned to one of the four stands per block (figs. 1 and 2) in all blocks, were:

1. no treatment (control = CT);
2. VDT with 3 thinning intensities and underplanting;
3. den-augmentation-only (cavities in live trees and nest boxes);[8] and
4. den augmentation plus VDT and underplanting.

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[6] Larson and others (1994) have proposed the name, *Inonotus sulphurascens* (Pilat) M. Larsen and others, for Douglas-fir-infecting laminated root rot. We recognize the distinction between the Douglas-fir form and the western redcedar form of laminated root rot (Thies and Sturrock 1995); however, to avoid confusion, we use the name *Phellinus weirii* for the Douglas-fir form.

[7] Moss names and authorities are from Schofield (1992).

[8] Because only baseline and preliminary responses of selected variables are presented, we do not herein present or evaluate the effect of den-augmentation-only. As such, unless specifically noted, control will refer to both no treatment (1) and den-augmentation-only (3).

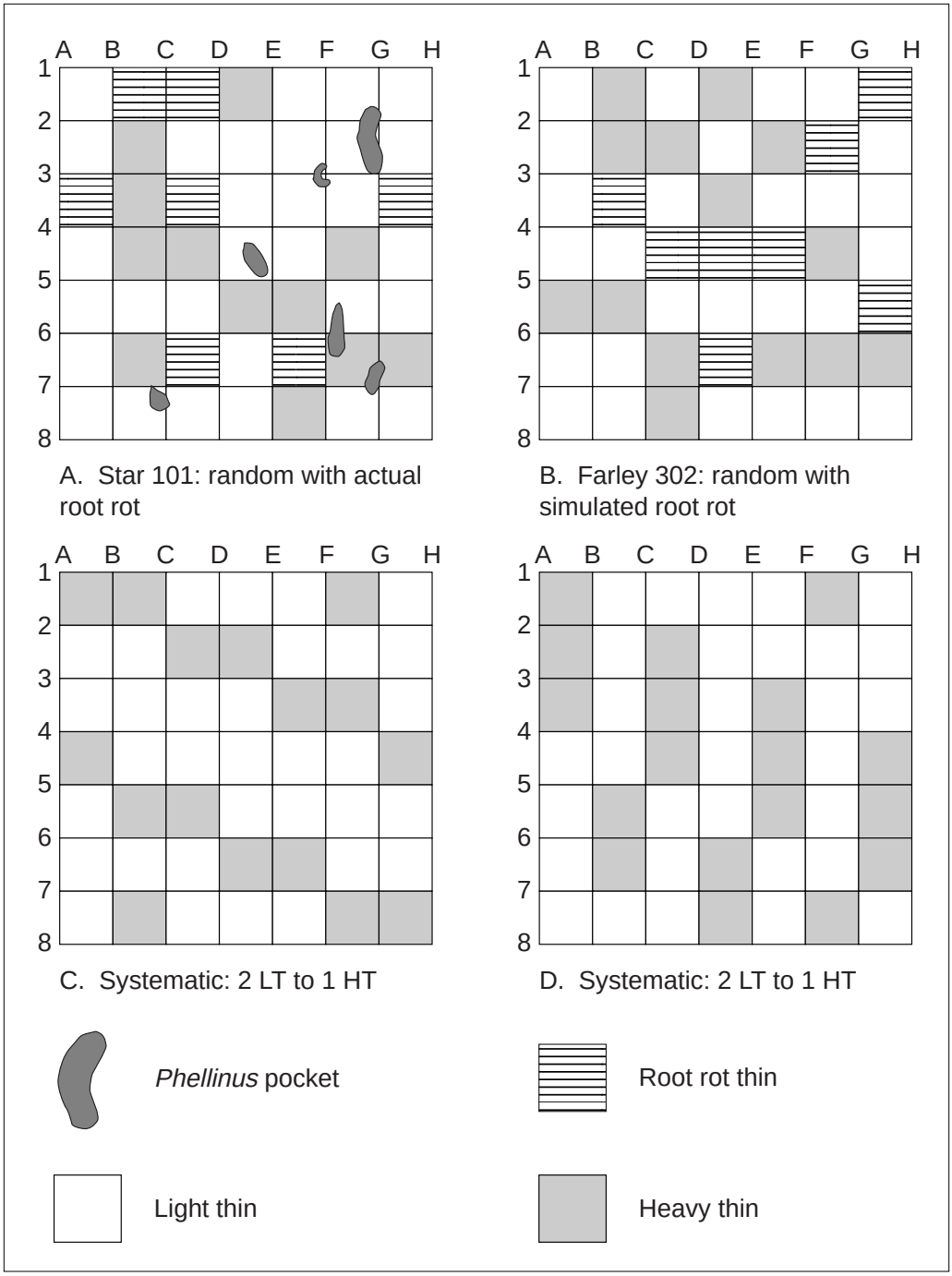


Figure 3—Actual and potential applications of variable-density thinning. Star 101 (A) shows random assignment of subtreatments with actual root rot pockets. Farley 302 (B) shows random assignment of subtreatments with simulated root rot pockets. Systematic assignment (C. & D.) of light and heavy thins (as possible operational applications) in a 2 to 1 ratio of LT to HT.

Subtreatments were randomly assigned to the forty-nine 40- by 40-m grid cells in each of the two treated stands (thinning and thinning plus dens) per block (see fig. 3 for example applications). Thus, the entire study consisted of 16 stands, 13 ha each, for a total study area of 208 ha. Grids were templates for treatments (figs. 1, 2 and 3), for cavity and nestbox installation (fig. 4), for sampling arboreal rodents and forest-floor mammals (fig. 5), fungi, soil food webs, and vegetation (fig. 6), and for evaluating prethinning and postthinning stand conditions.

Variable-Density Thinning

Background—Development of the silvicultural prescription took place in two stages. First, a desired future condition was defined that would provide a suitable environment for foraging and roosting by spotted owls. Second, the study areas were surveyed to determine the steps necessary to achieve the desired future condition in the shortest time possible. The desired future condition was based on information summarized in Carey and others (1991b, 1992), Gutiérrez and Carey (1985), Ruggiero and others (1991), USDA Forest Service (1988), and Thomas and others (1990), and research on characteristics of the habitat of spotted owls and their prey (Carey 1995, Carey and Johnson 1995, Carey and Peeler 1995).

The desired future condition was an environment suitable for spotted owl foraging. Suitability for spotted owl foraging encompasses both an abundance of prey sufficient to provide owls with a reasonable return per unit effort of foraging and vegetation structure amenable to the sit-and-wait foraging strategy used by spotted owls. The structure also should be amenable to spotted owl roosting. Silvicultural manipulation has the potential to produce the structure desired and to benefit prey by providing a diversity of woody plants that support ectomycorrhizal fungi of the types consumed by flying squirrels and other small mammals. Both herbaceous and woody understory vegetation also contribute other food types (foliage, seeds, fruits, nuts, associated fungi) that are used by ancillary prey of owls (hares, rabbits, mice, and voles).

The desired future structure consists of the spatial arrangement of vegetation; spatial scale is also important. Areas of high vertical diversity of vegetation should alternate with areas of sparse understory. Sparse understory allows owls to spot, track, and attack prey; a column of vegetation provides a vertical array of perches from which owls can pounce or fly short distances to prey. Data collected from a variety of plot radii ranging from 5.6 to 25 m, and grids and transects with distances between stations ranging from 20 to 100 m in old forests (Carey 1995; Carey and others 1991b, 1992; Carey and Johnson 1995), suggested patterning in the overstory should be on a scale of 80 m with areas of sparse and variable understory twice as abundant as areas of dense understory.

The spatial heterogeneity observed in natural old forests could have resulted from a combination of three different processes: suppression of subordinate trees in densely stocked stands; gap formation resulting from the breakage of trees with top rot infestation or death of trees from senescence, windthrow, lightning strikes, disease, insects, or other causes; and gaps resulting from locally intense, laminated root rot infestations or small-scale catastrophic disturbances (fire, windthrow). Variable-density thinning was designed to simulate these processes. Light thinning simulates suppression

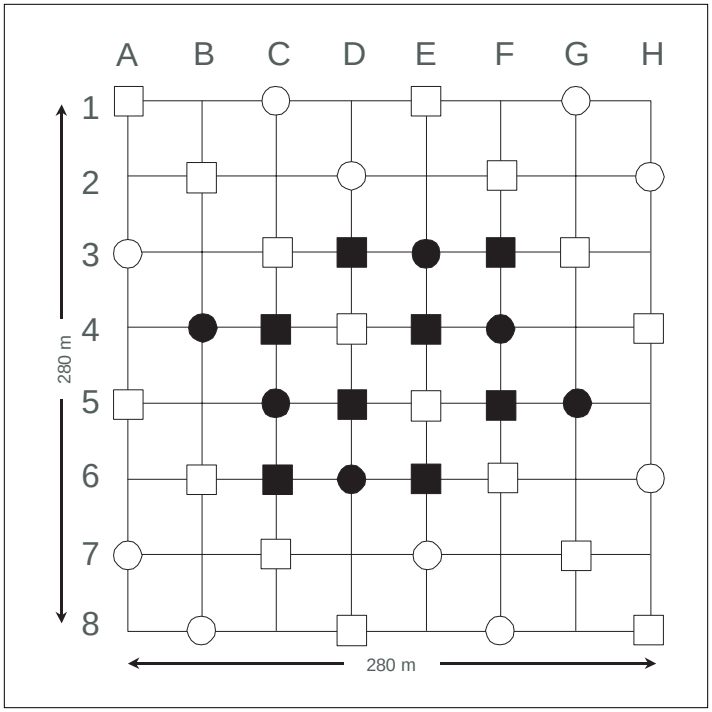


Figure 4—Nestbox, cavity, and top rot inoculation locations in Star 102, 103; Stellar 201, 203; Farley 303, 304; and Hill 403,404. Squares (nestboxes) and circles (cavities) are locations of supplemental den sites. Open boxes and circles are cavities and nestboxes from fall 1991. Solid boxes are the additional eight nestboxes added in winter 1995. Filled circles mark locations of top rot inoculations in addition to cavities. All grid corners also served as stations for trapping arboreal mammals.

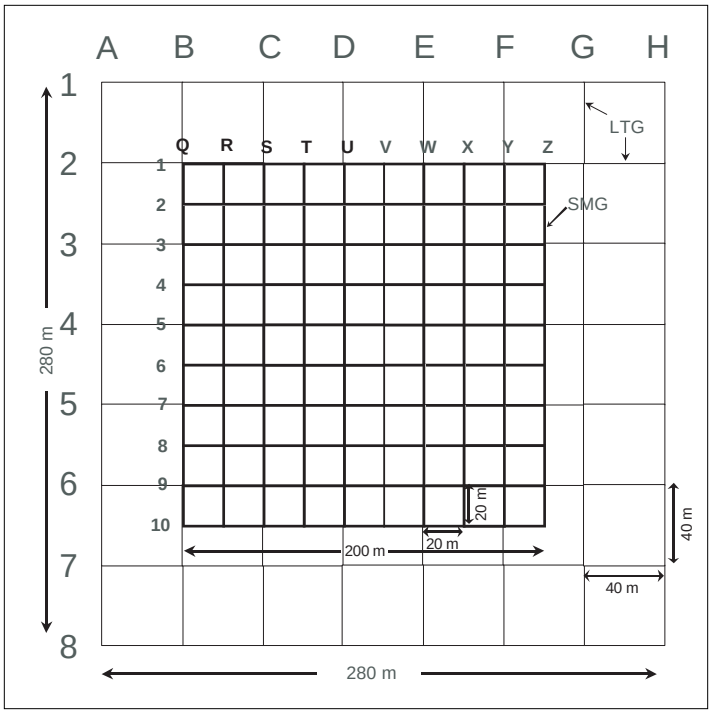


Figure 5—Design of live trapping grids (LTG) used for trapping arboreal rodents (A-H, 1-8), and small mammal grids (SMG, in bold) used for trapping forest floor small mammals (Q-Z, 1-10). Both grid types are located on all 16 stands.

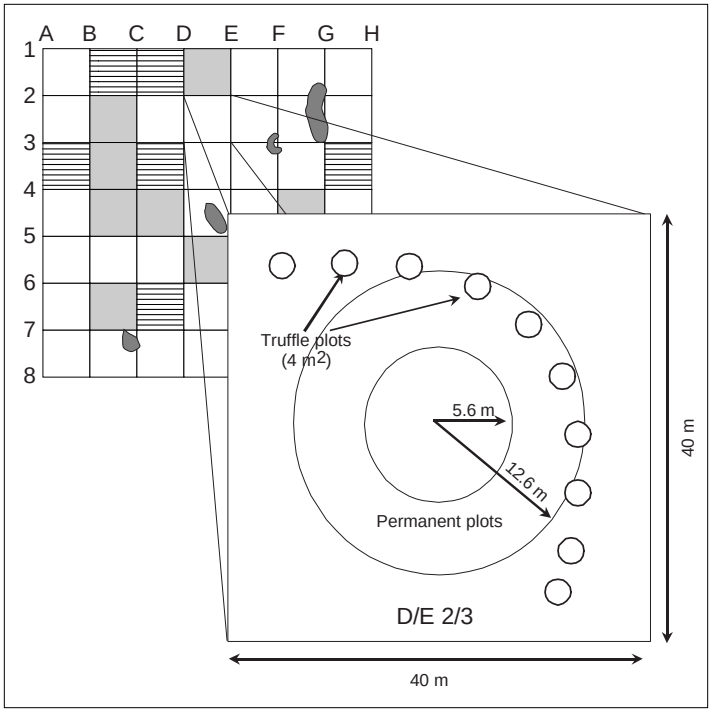
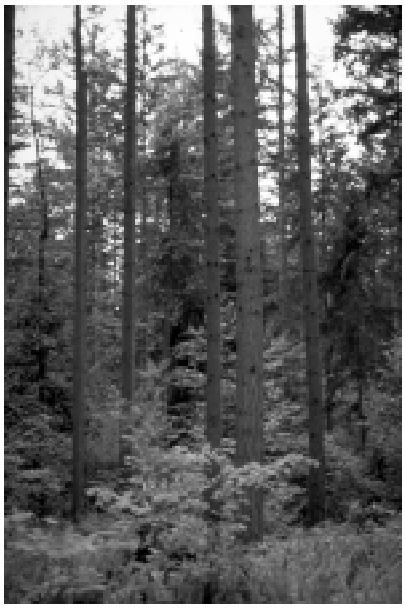


Figure 6—Permanent vegetation plot configuration. Plots were marked with a 1-m-long PVC pipe inserted 66 cm in the ground at plot center. The inner 5.6-m-radius plot was used for understory composition, cover, and structure and for epigeous fungal composition and biomass. The outer, 12.6-m-radius plot was used for overstory tree composition and tally. Truffle plots were located systematically, 10 m apart after a random start. Plots were identified by stand and cell number, in this case D/E 2/3.

mortality; heavy thinning simulates gap formation as a result of individual tree breakage or death; and the root rot treatment, contributing to, but not essential to our goals of creating spatial heterogeneity, mimics small-scale catastrophic disturbance (e.g., root rot, fire, windthrow).



Natural or management-induced canopy openings can produce a vertical array of vegetation—a column of vegetation from the forest floor to the canopy. The vegetation can consist of low shrubs and ferns (e.g., salal, Oregongrape, swordfern), tall shrubs (e.g., huckleberry, oceanspray, hazel, vine maple), conifer seedlings and midstory trees (e.g., western hemlock, western redcedar), deciduous trees (e.g., red alder, bigleaf maple, Scouler's willow), and lower branches of overstory conifers with deep crowns. Low understory shrubs provide food and cover for spotted owl prey; tall shrubs, saplings, understory trees, midstory trees, and overstory trees provide the array of foraging perches owls use to exploit the prey base. The column of vegetation and perches provides protected roost sites for owls. Undesirable structure includes large vertical gaps between the lower crown and understory vegetation,

Table 2—Block descriptions of live trees in the Star, Stellar, Farley, and Hill blocks based on prethinning prism plots

Block name	Year of origin	Harvest activities	Basal area	Douglas-fir	Height to live crown ± S.E.	Other tree species[a]
			m^2/ha(ft^2/acre)	*Percent*	*Meters*	
Star	1937	Unthinned since 1937 harvest	42.2 (184)	98	18.8 ± 0.3	Western redcedar, western hemlock, Pacific yew
Stellar	1937	Unthinned since 1937 harvest	50 (218)	99	18.7 ± 0.5	Western hemlock, big-leaf maple
Farley	1925	Clearcut 1925. Thinned twice: 1972, 1979-89	45 (196)	97	20.9 ± 0.6	Red alder, black cottonwood, big-leaf maple, western hemlock
Hill	1925	Clearcut 1925. Thinned twice: 1972, 1979-89	43.6 (190)	92	21.4 ± 0.6	Red alder, black cottonwood

[a] Scientific names are provided in appendix 1.

an absence of understory vegetation, and a continuous, dense cover of low shrubs. The most difficult aspect of producing the desired future structure is stimulating vegetation to fill intermediate heights (2 m to base of live crown). Height to live crown for dominant Douglas-fir trees averaged 16 to 21 m in Star/Stellar stands and 18 to 24 m in Farley/Hill stands (tables 2 and 3), suggesting shade-tolerant conifers or understory deciduous trees would be required to fill this gap where subordinant and codominant trees were removed by thinning. Our goals for the desired future structure in areas of high vertical vegetative diversity were:

1. Vertical diversity of vegetation using the Berger-Parker Index[9] > 2 (old-growth averages 1.7; spotted owl roosts, 2.3)
2. Shrub cover (≤ 2 m tall) > 40 percent
3. Understory cover (subcanopy, tall shrubs > 2 m tall) > 24 percent

[9] A measure of foliage-height diversity. See Magurran (1988) and Carey and others (1991b). In this case, four strata are considered: forbs, ferns, and trailing vines; low shrubs; subcanopy trees and tall shrubs; and canopy trees. During forest development (after the stem exclusion phase), canopy cover becomes less dominant in relation to the cover of the other strata, and thus the index becomes larger. We use the reciprocal form of the B.P. Index so that it increases with increasing evenness among the strata.

Table 3—Mean height, mean d.b.h., and mean height to live crown for Douglas-fir site trees from pretreatment prism plots[a]

Block name	Stand number	Height ± S.E.	D.b.h. ± S.E.	Height to live crown ± S.E.
		Meters	*Centimeters*	*Meters*
Star	101	31.5 ± 0.6	38.6 ± 2.0	16.5 ± 0.9
	102	34.1 ± 0.7	43.4 ± 1.5	20.7 ± 0.7
	103	32.3 ± 0.7	46.5 ± 1.8	18.5 ± 0.5
	104	32.9 ± 0.7	45.0 ± 2.0	18.4 ± 0.7
Stellar	201	31.9 ± 0.6	42.0 ± 1.7	16.4 ± 0.7
	202	34.5 ± 0.8	46.6 ± 1.3	17.9 ± 0.8
	203	32.3 ± 0.8	37.3 ± 1.8	19.7 ± 0.9
	204	33.1 ± 0.2	41.5 ± 1.6	20.8 ± 1.0
Farley	301	43.1 ± 0.8	61.6 ± 1.9	19.1 ± 0.7
	302	42.0 ± 0.5	59.7 ± 1.5	23.6 ± 1.1
	303	41.8 ± 0.8	63.8 ± 1.8	22.0 ± 1.1
	304	41.4 ± 0.6	62.3 ± 1.3	17.9 ± 1.1
Hill	401	39.1 ± 1.6	56.2 ± 3.4	21.7 ± 1.3
	402	40.9 ± 1.4	62.6 ± 2.3	19.9 ± 1.3
	402	43.2 ± 0.8	65.7 ± 1.9	22.7 ± 1.1
	404	41.6 ± 1.3	60.2 ± 2.1	21.2 ± 0.9

[a] 30 site trees per stand were sampled.

Relative density—We adopted Curtis' (1982) relative density (RD) as a broadly applicable index of stand density that is also useful in designing silvicultural prescriptions. Relative density varies along a continuum from zero (no trees) to a species-specific biological maximum in extremely heavily stocked stands (Curtis 1982). Though empirically derived data relating understory vegetative development (and other forest conditions) to stand relative density are not extant, the range of relative density for a given age stand encompasses recognizable stages of forest development and of responses to natural forest disturbance (Carey and others 1996a, Curtis 1997, Oliver and Larson 1990). For 50- to 60-year-old, even-aged Douglas-fir stands, we suggest the following RD ranges as being indicative of particular forest conditions.[10]

1. RD < 3.25 (metric) represents larger gaps in the forest canopy, which, under natural conditions, could be formed by multiple tree blowdown, small fires, localized insect outbreaks, or root rot pockets. In such areas, there is little intertree competition, and abundant insolation is available to understory vegetation. Depending on the size of such areas, and on age and species composition of the canopy trees, these gaps may fill by lateral branch growth, by growth of shade-tolerant trees and shrubs, or by shade-intolerant trees such as Douglas-fir.

2. RD 3.25-4.75. Within this range, intertree competition increases rapidly. Subcanopy insolation is reduced, thereby resulting in less understory development. Natural canopy gaps within this range of RD may be the result of small-scale endogenous disturbances such as the death, defoliation, tree-fall, or lightning strike of one or a few trees. Stand-level tree growth within this range is somewhat sacrificed to understory development, though forest floor vegetation is less well developed than when RD is < 3.25. The major source of light below the canopy is from short-duration sunflecks through a moderately open canopy.

3. RD 4.75-6.75. Within this range, the canopy is increasingly closed, with little direct light reaching the forest floor. Intertree canopy competition increases, resulting in near-suppression mortality conditions at the upper end of this range. Understory vegetation is typically sparse and poorly developed. Many even-aged stands under commercial management for timber production are managed within this range; stand tree growth is maximized and tree form and development are both quite uniform.

4. RD > 6.75. Above RD 6.75-7.00, forest stands enter a state of suppression (Curtis 1982), where severe crowding results in reduction of tree growth and significant tree mortality. Individual trees are under extreme intertree competition, crown development is restricted, and understory vegetation is typically quite sparse.

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[10] Continued sampling and analysis on the Forest Ecosystem Study will evaluate and further refine these suggested relative density categories.

Subtreatment descriptions—Two thinning entries were scheduled as part of the FES. The first entry was made during January-April 1993; the second is scheduled for 2003 but is provisional. Prescription targets for the first entry were formulated for forest stands under "normal"[11] conditions for 50- to 60-year-old, even-aged, unmanaged Douglas-fir forests. An average stand-level residual tree density target was specified as 220 trees/ha, representing a relative density of between RD 4 and RD 5 (metric) for such stand conditions. To achieve this stand density and the desired spatial diversity of densities within stands, the prescription for the first entry had two primary components (subtreatments):

1. A thinning of subordinant and codominant trees > 20 cm d.b.h. to reduce the density of overstory to RD 6 (metric) with a thinning ratio d/D[12] of 0.8-1.0. For a 50- to 60-year-old forest under such normal conditions, this represents a tree density of approximately 310 trees/ha with an average spacing[13] of 5.8 m between trees (= light thin, LT).

2. A thinning of subordinant and codominant trees > 20 cm d.b.h. to reduce the density of overstory trees to RD 4 (metric), or about 185 trees/ha with an average spacing of 7.3 m between trees (= heavy thin, HT).

The prescription specified the removal of suppressed and subdominant trees of the current cohort (e.g., thinning from below); this equates to a thinning ratio (d/D) of 0.8-1.0. Marking guidelines specified retention of all large standing dead trees and all deciduous trees (trees retained included black cottonwood, bigleaf maple, red alder, Pacific madrone, and Scouler's willow). Similarly, deciduous shrubs were favored for retention; these included red huckleberry, oceanspray, and California hazel. Existing roads and skid trails were used wherever possible to reduce impacts on stands; where skid trails were lacking, they were preferentially located in the more heavily thinned areas.

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[11] A "normal" forest is described as a stand where every tree is using all its available space, and where competition, in terms of individual tree growth, represents an "average maximum" (Curtis 1970). The choice of the term "normal" may mislead some, because it does not refer to modal or average conditions in naturally regenerated forests of the Pacific Northwest, and does not refer to the normal or Gaussian distribution. Actual thinning densities were applied by using foresters' professional judgment to avoid severely reduced wind resistance in dense stands and to create variable-density in the more open stands.

[12] d/D is the ratio of the quadratic mean d.b.h. of the cut trees (d) to the quadratic mean d.b.h. of the original stand of trees (D). The ratio, d/D, indexes the type of thinning by comparing the original stand diameter distribution with that of the cut trees. When d/D = 1.0, this indicates a systematic thinning; when < 1.0, a thinning from below; and when > 1.0, a crown thinning (Bennett and Maguire 1995).

[13] Our tree spacing targets do not imply that we sought to achieve even or regular spacing conditions. Even at the 40- by 40-m scale, we attempted to capitalize on naturally occurring clumps and openings. As such, spacing values are averages only.

Prethinning forest cruise surveys of the study area revealed that pockets of laminated root rot occupied 7 to 15 percent of the area of the younger, unmanaged stands (Star and Stellar); root rot may have been present in the older, managed stands (Farley and Hill) but infection pockets were not apparent. Because root rot can lead to canopy openings, we incorporated a root rot treatment in the prescription. This became a third component (subtreatment) of the thinning prescription:

3. Removal of low-vigor trees from root rot infection centers to 10 m beyond the perimeter of the pocket of dead and dying trees; apparently healthy, dominant trees were retained. Approximate residual tree density for the root rot areas was 40 trees/ha > 20 cm d.b.h., or an RD 1.5 (= root rot thin, RT).[14]

Because this is an experimental study, we applied the root rot treatment equally (15 percent of stand area) to all stands selected for thinning. Where root rot pockets were apparent, they were mapped and treated; where not apparent, the treatment was randomly assigned. Thus, the same overall treatment was applied to all treated stands.

The spatial arrangements of the treatments consisted of first treating existing root rot pockets in the most heavily infected stand—15 percent of the area. Natural root rot pocket size averaged 0.07 ha. Where root rot pockets comprised < 15 percent of the remaining stands, root rot treatments were randomly applied to the 0.16-ha blocks to achieve consistency in the experimental treatment. Next, the light and heavy thinnings were applied to the 0.16-ha blocks (40 by 40 m) in a 2:1 ratio over the remaining area by random assignment.[15] Additional entries (at about 10-year intervals) will be necessary to achieve the desired future condition: eventually the 2:1 ratio will be reversed to heavy:light.

Alternating the heavy and light thinnings should produce highly variable light regimes on the forest floor that result in a mosaic of vegetation site types, some with a high degree of vertical layering, and some with simple structure. Variable-density thinning has the additional advantage of maintaining stand wind resistance while increasing the amount of light available to the understory and maintaining the stand composition necessary for wood production. The multiple entries that are a part of VDT are necessary to achieve the final desired future condition (Curtis and Carey 1996).

Cavities

The amount of old forest used by spotted owls in the Pacific Northwest reflects regional variation in prey abundance; selection of stands for foraging also reflects prey abundance (Carey and others 1992). The primary prey of the spotted owl in the Douglas-fir and western hemlock forests of western Washington is the northern flying squirrel (Carey 1995; Forsman and others 1984, 1991), which uses cavities in trees

[14] Our choice of root rot treatment was guided by Thies and Sturrock (1995). Due to site-specific windthrow considerations and the 40- by 40-m size of our operational thinning units, we chose 10-m buffers rather than their suggested 15 m.

[15] Figure 3 illustrates examples of VDT application with actual RT pockets (3a), VDT with simulated RT (3b), and systematic HT and LT applications, such as could be used in an operational, rather than an experimental, setting (3, c and d).

(both live and dead), stick nests, and moss-leaf-twig nests as dens (Carey 1991, Carey and others 1997). Previous research shows that both large snags and trees with natural cavities are much less abundant in young, managed stands than in old growth (Carey 1995, Carey and others 1991a). Numerous studies of cavity-using squirrels outside the Pacific Northwest have shown that addition of supplemental dens (usually nest boxes) to managed stands can increase squirrel populations (see Carey and Sanderson 1981 for a review). Thus, adding supplemental dens to managed stands allows testing of the hypothesis that dens are limiting to flying squirrels. If providing dens can raise flying squirrel populations, the potential exists for attracting owls to managed stands, thus increasing available owl foraging habitat.

Supplemental dens can be provided in two forms: nestboxes and cavities created in trees (Carey and Gill 1983). Nestboxes increase the number of flying squirrels in other areas (see Carey and Sanderson 1981 for a review) but have the disadvantages of being relatively susceptible to decay and damage and, to some observers, appearing unnatural. Nestboxes generally benefit only a few species besides target species. Cavities created in live trees have the advantage of potentially being available for many years and of introducing top rot that will eventually provide a condition suitable for excavation by woodpeckers and subsequently be used by a great variety of cavity-using birds and mammals. Because nestboxes increase flying squirrel abundance whereas excavated cavities, as designed for this study, have not, both forms were used in this study. High flying squirrel densities are those > 3/ha (Carey 1995, Carey and others 1992), with flying squirrels using multiple dens and dens being used by several flying squirrels simultaneously (Carey and others 1997). Thus, we initially added sufficient cavities and nestboxes so that with natural dens we would ensure that dens were not a limiting factor for squirrel populations.

Cavities and nestboxes were systematically and alternately placed over the treatment areas at a density of 3/ha. They were placed in half of the experimentally thinned stands and half of the unthinned stands (fig. 4). Nestbox and cavity designs are shown in Carey and Gill (1983), in a video (Carey 1993), and in appendix 2. Sizes of boxes and cavities were chosen to be the minimum that could contain three or more flying squirrels.[16] Interior dimensions (below the shelf) of Flyger nestboxes (Carey and Gill 1983) were 6 by 7 by 7 inches (height, width, depth) and 7 by 7 by 6 inches for the cavities. Cavity depth was a minimum of 6 inches but often extended to 10 inches if the tree was sufficiently large. The nestboxes are more predator-proof and more thermally beneficial (because of the shelf) than the created cavities. If nestboxes are used more than created cavities, cavities may be modified by adding a shelf below the entrance as was done for nestboxes. Because addition of a shelf to a cavity reduces interior volume, the cavity might need to be deepened or lengthened. We placed boxes and cavities 6 to 7 m aboveground for ease of inspection. Application in a management scenario would call for placing cavities as high as practical with maximum height being limited by tree diameter; trees should be at least 30 cm in diameter at the cavity site. Increasing the height of cavities aboveground is desirable not only because of increased attractiveness to flying squirrels and reduced access to predators

[16] Up to five flying squirrels have used the boxes simultaneously.

but also because of timber values of cavity trees destined for harvest. Cavities will induce top rot, but trees should compartmentalize the rot and one or two 16-foot logs could be kept clear of rot if the cavity is placed high enough.

Underplanting

Reconnaissance showed insufficient tree regeneration and insufficient seed sources to provide a midstory of shade-tolerant trees to fill gaps between lower crowns of dominant and codominant trees and upper layers of the crowns of tall shrubs. Accordingly, we incorporated underplanting for species and architectural diversity into our experimental design. Because part of the underplanting of seedlings was to go into *Phellinus weirii* infection centers, we included species immune to, highly resistant to, or tolerant of root rot (red alder, western redcedar, and white pine, respectively) and grand fir because of its shade tolerance, seed production, and retention of a dense crown (to provide architectural diversity) (Burns and Honkala 1990a, 1990b; Thies and Sturrock 1995).[17] We limited our choice to species that commonly occur or were

[17] Western hemlock regeneration was found infrequently in both forests. Though western hemlock occurs in other areas with similar glacial soils, the combination of excessively permeable soils (and concurrent low water storage capacity) and low summer rainfall found at Fort Lewis creates substantially greater annual soil moisture deficit than in other areas where western hemlock dominates (Packee 1976). Fort Lewis is also an area of historically short fire-return intervals, and the thin-barked western hemlock is very susceptible to fire (Burns and Honkala 1990a). Because western hemlock also is frequently infected with *Phellinus weirii,* especially where Douglas-fir is or was a major component (Burns and Honkala 1990a), we chose not to underplant western hemlock. Grand fir was chosen as an ecological (shade-tolerant) equivalent to western hemlock; grand fir is common in the Puget Trough and it was thought that it would grow better in such a dry environment.

historically common in west-side forests. We chose to plant only root rot thin and heavy thin cells to retain the desired open understory conditions in the remaining light thin cells.

These four tree species will enhance the tree diversity of the study site and in the long term will begin to fill the intermediate canopy spaces between the tall shrubs and the base of the overstory canopy. Natural regeneration of Douglas-fir also will occur, but its growth in the understory probably will be limited (Oliver 1995). The production of abundant Douglas-fir cone crops is highly variable, and thus the mix of underplanted species will in the long term provide a more diverse and more reliable source of seed for small mammals and birds (Burns and Honkala 1990a, 1990b; USDA Forest Service 1974).

Top Rot Fungi Inoculation

Flying squirrels, other arboreal mammals, and birds make substantial use of woodpecker holes for nest sites. Our work has shown natural nest sites (cavities and others) to be of limited availability in many managed Douglas-fir forests, including those of the study site (Carey 1995, Carey and others 1997). Artificial inoculation of decay fungi in managed stands has been successful in inducing decay (Conner and others 1983, Parks and others 1995); fungal decay is a prerequisite for excavation by many primary cavity nesters such as woodpeckers (Neitro and others 1985). To hasten development of trees and snags with acceptable decay conditions for woodpecker use, we incorporated tree inoculation with stem-decaying fungi into our experimental design.[18] We isolated two commonly occurring wind-dispersed fungi, *Fomitopsis cajanderi* and *Phellinus pini*, from the study site; *Fomitopsis* is generally found at greater heights in the stem than is *Phellinus*. Both are widespread, naturally occurring, and weakly pathogenic fungi. Isolates were obtained from decaying Douglas-fir trees, grown in pure culture, and inoculated into sterilized softwood dowels to facilitate later placement in trees.

Postthinning Stand Assessment

Because response variables will be related to specific forest conditions as they exist after experimental thinning, an evaluation was performed to describe actual, postthinning variability at the stand and grid-cell level. Specific goals of the assessment were:

1. To estimate prethinning and current stand stocking densities[19] and basal area by sampling each thinning subtreatment in each stand
2. To estimate logging damage and stand health
3. To estimate incidence of root grafting
4. To estimate stocking density of underplanted seedlings and natural regeneration

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[18] The FES inoculations are part of a much larger and more widespread series of field trials throughout different forest conditions in the Western United States. Background, methods, and locations of the larger study are available from: Catherine Parks, Pacific Northwest Research Station, 1401 Gekeler Lane, La Grande, OR 97850.

[19] The terms “stocking density,” “stocking,” and “tree density” are used interchangeably and refer to number of trees per hectare (N/ha).

5. To provide a database of estimates of forest parameters (e.g., retained basal area, retained relative density, current density) for each grid cell within thinned stands
6. To reclassify thinning subtreatments based on retained relative density and to provide estimates of areas occupied by each category in each thinned stand
7. To provide forest parameter maps for each of the thinned stands
8. To provide the basis for designing the 2003 thinning

Implementation

A schedule of the major activities, treatments, and sampling efforts from the inception of the Forest Ecosystem Study in 1991 until the end of 1996 is shown in table 1.

Grid Establishment

After on-the-ground reconnaissance identified sufficiently large areas of homogeneous vegetation to contain the study, the four stands per block were located with a minimum of 80 m between the stands. Blocks and stands were given numerical designations: the Star stands were designated 101-104; Stellar, 201-204; Farley, 301-304; and Hill, 401-404. Maps of the four blocks (Farley, Hill, Star, and Stellar) showing the arrangement of treatments and the grid coordinate designations are shown in figures 1 and 2. Grids were surveyed and established in early fall 1991; each grid consisted of 64 stations aligned in an 8 by 8 matrix with 40-m spacing between stations. Each station was identified by a letter (A through H, west to east) and a number (1 through 8, north to south). For example, the northwestmost station of every grid was always "A1." The grids were 280- by 280-m squares, with each station point (= grid cell corner) marked with a 1-m length of white plastic PVC pipe (1-3/4 in OD, Schedule 40, inserted 66 cm in the ground). The aboveground portion of the stake was painted fluorescent orange. Each station also was marked with two strips of orange fluorescent flagging, and the grid station was visibly marked at the base of both strips with permanent ink. In addition to the 64 station points, the north-south 20-m points between stations were identified by a pin flag inserted into the ground and with a single strip of fluorescent orange flagging. The resulting forty-nine 40- by 40-m grid cells in each surveyed stand were the operational units to which the thinning subtreatments were randomly applied (fig. 3).

Arboreal Rodent Trap Placement

The 8 by 8 matrices were used as a template for live trapping grids (LTG), which determined the placement of arboreal rodent traps (fig. 5). In 1991, two Tomahawk[20] 201 live traps were placed at each of 64 grid stations in each of the 16 stands (2,048 traps total)—one trap was placed on the ground and one was attached to a tree as outlined in Carey and others (1991a). A 4-in-square aluminum tag with the grid and station number etched on it was nailed to each trap tree. Traps were placed in the grids in early fall 1991; trap locations were identified by stand (i.e., 201, 303, 404) and grid station (i.e., A1, B3, G7). Trapping of all grids has taken place twice yearly (spring and fall) since 1991.

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[20] The use of trade or firm names in this publication is for reader information only and does not imply endorsement by the U.S. Department of Agriculture of any product or service.

Small Mammal Trapping Grid and Trap Placement

In 1992, 10 by 10 small mammal grids (SMG) with 20-m spacing between stations were established within the center of each of the eight stands designated as not-to-be-thinned (e.g., control and den-augmentation only). Each SMG was located within the LTG, with the northwestmost SMG station being identical with the LTG point B-2, and the SMG extending grid-east and -south for 200 m from that point. Because of the 20-m spacing, every second SMG station coincided with an arboreal rodent trapping station. To avoid confusion with the arboreal rodent trapping station designations, a system of identification was used for the SMG stations whereby each station was identified by a letter (Q through Z, west to east) and a number (1 through 10, north to south). Because many of the grid points were both an arboreal mammal trap station and an SMG station, the station number used for recording data was based on the type of trapping being performed at the time (fig. 5).

Two Sherman live traps were positioned on the ground within 2 m of each SMG station; one large 3- by 3.5- by 9-in trap, and a smaller 2- by 2.5- by 6.5-in trap. Trap placement was prioritized by proximity to large, fallen, decayed logs and other coarse woody debris.

Trapping was performed at the eight unthinned SMGs in 1992, 1993, and 1994. In summer 1996, SMGs were placed in the eight experimentally thinned grids as well (fig. 5). Trapping of all 16 SMGs began in July 1996.

Cavity and Nestbox Placement

In April 1992, 16 nestboxes and 16 cavities (illustrated in appendix 2) were systematically distributed at 80-m intervals throughout eight grids. A cavity or nestbox was placed on the largest tree (> 30 cm d.b.h.) within 5 m of the selected gridpoint, or on the nearest tree > 30 cm d.b.h. if no large tree could be found within 5 m. Beginning with the A1 station, alternating cavities and nestboxes were placed at every other grid station on the north-south lines (fig. 4). Construction of nestbox and cavity faceplates, installation of cavities, and placement of nestboxes were performed by contractors. Nestboxes were secured with aluminum nails at a height of 6 m. Cavities were created at this same height with the use of a chainsaw equipped with a special tip. The faceplate was then nailed over the opening of each cavity.

Because most supplemental nestboxes showed signs of use by spring 1995, an additional eight nestboxes per den-augmented stand (64 boxes total) were added in December 1995. The additional eight nestboxes were placed 12 m above the ground rather than at the original 6-m height. They were placed at the C4, C6, D3, D5, E4, E6, F3, and F5 grid stations on den-augmented stands (fig. 4). At that time, modifications were made to the cavity size and the faceplate door placement (appendix 2).

Silvicultural Treatment

Experimentally thinned stands (thin, thin plus den-augmentation) were cruised during tree marking. We employed cut-tree marking; only trees > 20 cm d.b.h. were marked for harvest. Thinning operations took place between January and April 1993 with tractor skidders. Net harvested volumes from SS stands ranged from 104 to 154 MBF per stand; FH net volumes ranged from 192 to 222 MBF per stand. For Star and Stellar, the harvested volumes ranged from 5.4 to 8.1 MBF per acre; Farley and Hill harvested volumes ranged from 8.7 to 11.1 MBF per acre.

The locations of skid trails and landings were planned to minimize disturbance in thinned stands. In Star and Stellar blocks, skid trails from the 1937 harvest were used to the greatest extent possible; otherwise, new skid trails were located in the heavy thins, root rot thins, and along the margins of *Phellinus* infection centers. In Farley and Hill blocks, skid trails and landings from previous thinning entries were used.

We employed one of the standard slash disposal treatments that the Fort Lewis Forestry Division uses during their thinning operations. After trees were felled, they were pulled to the landing with crowns intact. At the landing, trees were bucked into logs and their branches trimmed; the accumulated debris was piled and burned after harvest. By doing so, a relatively constant amount of coarse woody debris (CWD) was maintained in the treated stands, with the net intended result being that no additional CWD would accumulate from the thinning operations.

Underplanting

Western redcedar, western white pine, and grand fir were planted at equal densities within each root rot and heavy thin subtreatment cell. Planting took place in May and June 1994. Conifer seedlings were planted on a 9 by 9 matrix with 3.7-m spacing, a density equal to about 510 trees/ha.[21] Three planters, each with a different species, planted three sets of three parallel rows. Thus, three lines of nine seedlings (27 total per subtreatment cell) were planted for each conifer species (for a total of 81 conifer seedlings per cell). The 20 red alder seedlings were planted every 11 m, between the lines created by the planted conifers. Because substantial natural alder regeneration was expected, red alder was planted only in the RT cells, and was planted at lower densities than the conifers—20 red alder seedlings per grid cell vs. 27 per conifer species per grid cell. Planting guidelines for all species were as follows:

- To avoid the relatively higher shade along the edge of the treatment cells, planting lines began about 7 m from the treatment boundary. This produced a small buffer around the planted seedlings and afforded space to plant seedlings not planted on a line because of their planting location falling within 2.5 m of an existing overstory tree.
- If a planting site fell within 2.5 m of an existing tree, the seedling was placed at the end of that planting row. As many as two additional seedlings could be planted at the end of any planting row. If three planting sites were displaced by existing trees, the third seedling could be planted 3.7 m in from the treatment boundary on the next line. The 7-m buffer around seedlings allowed space for planting trees that were displaced by existing trees. Planting at the end or beginning of a planting line allowed the planting of an equal number of seedlings in each planted grid cell.

Top Rot Inoculation

Fungal inoculum was placed above cavities in selected Douglas-fir cavity trees. The insertion points were holes drilled to a depth of 10 to 15 cm. The holes were cleaned of chips and an inoculated dowel placed within. A PVC pipe was then inserted in the hole at a horizontal or slightly downturned angle to prevent water entry and to prevent closure of the hole. Five to eight cm of pipe was left exposed from the side of the tree

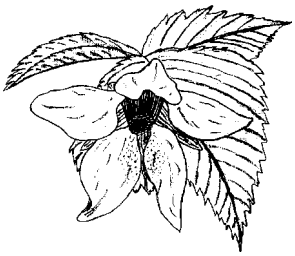
[21] These figures refer to planting densities at the grid cell level. This resulted in a stand level planting density of about 210 conifer seedlings/ha.

to mark the location of the inoculum. Tree d.b.h., aspect of inoculum hole, and any tree defects (broken, forked, dead tops) were noted, as were color and texture of chips removed from the drill hole (to assess presence of preexisting decay).

A systematic subset of six of the artificial cavity trees in each of the eight den-augmented stands was selected for inoculation. Inoculation was performed in summer 1994. Locations of the inoculation holes and specific treatments of the inoculated trees were as follows (fig. 4):[22]

Grid points B4 and G5—*Phellinus pini* at 25 cm above created cavities

Grid points C5 and F4—*Phellinus pini* at 25 cm above and *Fomitopsis cajanderi* at 3 m above created cavities



Grid point D6—*Phellinus pini* at 3 m above created cavities

Grid point E3—*Fomitopsis cajanderi* at 3 m above created cavities

Thus, each den-augmented stand contained six inoculated trees with a total of eight inoculation holes. Evaluation of the success of fungal inoculation will be made by observing bird excavation and use at or near the inoculation site and sampling to attempt to reisolate the original fungal strains.

Sampling Methods

Vegetation

Pretreatment tree and understory woody plant abundance—Fifteen interior gridpoints, a minimum of 56 m apart in each of the 16 stands (240 points total), were selected systematically as reference points for measurement of trees and understory woody plants before the thinning treatments. Alternating N-S grid lines had two or three sampling plots, respectively. Tree sampling was performed in fall and winter 1991. Understory vegetation was sampled in June-August 1992.

At each selected grid point, a variable-radius prism plot count (basal area factor 30 [English]) was made. Species and d.b.h. were determined for each tallied tree as well as whether the tree originated with the current or previous stand. For the two dominant trees (site trees) nearest to plot center, species, d.b.h., height, height to live crown, crown width, and current-year horizontal and terminal growth were recorded. Crown width was determined by measuring the extent of the crown or dripline of the foliage for each measured tree. Estimates of lateral growth were made on lower or mid-crown branches; an average of the past 3 years of growth was recorded.

Woody shrub and other understory vegetation cover were sampled on 2.82-m-radius circular plots (25-m^2), offset 5 m at a randomly determined bearing from the grid point. We recorded (1) lifeform of plant species (e.g., evergreen shrub; deciduous shrub; fern or forb; grass, sedge, or rush; moss; or lichen); (2) vascular plant species; (3) percentage of cover by species; and (4) mean height of shrub species. In addition, moss and lichen species and moss cover were noted. We defined “cover” as that portion of the ground covered by the vertical projection onto the ground of the vegetation of interest.

The incidence and areal extent of root rot pockets (particulary laminated root rot) was mapped by using grid lines as line-intercepts. When root rot pockets were intersected, length and width of the disease pocket were recorded.

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[22] The exact positions of the inocula and descriptions of the host trees are on file with: Pacific Northwest Research Station, Forestry Sciences Laboratory, 3625 93d Ave SW, Olympia, WA 98512.

Posttreatment stand conditions—We used a stratified random sample to assess the silvicultural prescription. Initially, six grid cells per target subtreatment per stand were randomly selected and sampled with fixed-radius plots. Within each thinned stand were the three target subtreatments:

1. RT cells and *Phellinus weirii* pockets with underplanting
2. HT cells with underplanting
3. LT cells without underplanting

After the initial fixed-plot sampling, we sampled every additional grid cell with variable-radius plots. Sample plots were located in the center of grid cells to prevent overlap with adjacent cells and to minimize edge effects. Fixed-plot radii ranged from 4 to 15 m, according to the variable being measured (tree, stump, or seedling density) and the subtreatment.

In each fixed-radius plot, we counted standing trees (live and dead) by species, measured d.b.h. (cm) outside the bark of all trees > 10 cm d.b.h., and ranked tree health by using a modification of the Pacific Northwest Region stand exam procedure (USDA Forest Service 1989). For stumps, we measured diameter outside bark (cm), height (m), and presence or absence of callus formation (presence of callus was used as an indicator of root grafting). Stump diameters were converted to tree d.b.h. after forest-specific regression equations were developed. The equations are as follows:

$$\text{Star/Stellar: } Y = 0.81X + 1.43 \quad (r^2 = 0.94) \qquad (1)$$

$$\text{Farley/Hill: } Y = 0.88X + 0.99 \quad (r^2 = 0.96) \qquad (2)$$

where Y = d.b.h. in cm, and

X = stump diameter at 30-cm height.

We measured density of the three underplanted conifer species and density of natural tree regeneration by species and height class (table 4).

Table 4—Height class categories and descriptions for natural tree regeneration

Height class	Approximate age	Description
Centimeters	*Years*	
0-10	1 or less	Emergent seedling
11-50	1-2	Established seedling
51-100	≥ 2	Well-established seedling, emerging above ground vegetation
> 100 (and < 10 cm d.b.h.)	5 or more	Well-established sapling, with most of crown above ground vegetation

In the variable-radius plots, trees were counted with a wedge prism from the center of each cell. A basal-area-factor (BAF) 20 (English) prism was used in SS, and BAF 30 (English) in FH. Regression equations calculated from fixed-radius plots were used to estimate tree density from basal area:

$$\text{Star/Stellar: } Y = 9.23\,X + 19.06 \quad (r^2 = 0.74) \qquad (3)$$

$$\text{Farley/Hill: } Y = 3.70\,X + 11.41 \quad (r^2 = 0.73) \qquad (4)$$

where Y = residual tree density (N/ha), and

X = residual basal area ($m^{2/}$ha).

Data collected from both fixed- and variable-radius plots reflected current (1995-96) tree diameters for live trees and prethin (1992) diameters for stumps. Due to tree growth during that 3- to 4-year period, current live tree density of trees > 20-cm d.b.h. was greater than that of 1992 (assuming minimal tree mortality), thus resulting in a slight overestimation of the ratio of current tree density to target tree density. Because no growth and yield data were available for these sites, the current assessment made no correction for growth and mortality during this period.

Posttreatment vegetation permanent plots—After the experimental thinnings, permanent vegetation plots were established in the centers of 191 randomly selected grid cells (fig. 6; appendix 3). During summer 1994, six permanent plots were established in each target subtreatment in each stand[23] and in each control and den-augmentation-only stand by using a stratified random sampling design. In each stand of 49 cells, 100 percent of the root rot thin cells were sampled; about 50 percent of the heavy thin cells were sampled; and about 25 percent of the light thin cells were sampled. About 12 percent (6 of 49) of the cells in control stands and den-augmentation-only stands[24] were sampled. Twelve permanent plots were established in each target subtreatment in each block and 12 total in the unthinned stands in each block.

Plots were marked with 1-m lengths of 1-3/4-in diameter, schedule 40, white PVC pipe inserted 66 cm into the ground at plot center. At each plot we measured slope and aspect. In August 1995, each plot was photodocumented with a 24-mm wide-angle lens on a 35-mm camera (84-degree field of view) from a point 1.5 m high and 8.4 m south of the plot center (thus providing full lateral coverage of the 5.6-m-radius plot). A 2-m-tall photo board with alternating 0.25-m white and black sections was placed at plot center for reference. Plots will be rephotographed every 3 to 5 years.

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[23] The RT subtreatment in stand 103 contained only five cells of sufficient size to contain permanent plots, resulting in 191 rather than 192 permanent plots.

[24] Because no data are presented for experimental responses to the den-augmentation-only treatment, we also refer to them as control stands unless it is necessary to differentiate between the two unthinned stands (control and den-augmentation-only) per block.

Vegetation was sampled by using nested circular plots of 5.6-m radius (100 m^2) and 12.6-m radius (500 m^2). Understory composition, cover, and forest floor cover were measured on 5.6-m-radius plots centered on the PVC pipe. For cover values, we used octave scale categories modified from Gauch (1982).[25] Within 5.6 m, we described the forest floor cover of bare ground, moss, fine litter (needles, leaves, twigs < 2 cm diameter), coarse litter (2 to 10 cm diameter), fine woody debris (10 to 30 cm diameter), coarse woody debris > 30 cm diameter, and all vascular plant species. Computer-generated maps depicting a range of cover values were used to train observers; training also served to ensure consistency among observers in regard to species identifications and cover estimates. Identification of all vascular plant species (including grasses, sedges, and rushes) was facilitated because of involvement by the authors in a previous project that produced a vouchered floristic inventory of the Fort Lewis Military Reservation (Thomas and others 1994). Because of this earlier effort, vouchers from the FES were collected only for previously uncollected species. Vouchers from the current project will be deposited in the University of Washington Herbarium, Seattle.

Overstory composition, cover, and structure, and understory vegetation groups were described on 12.6-m-radius plots also centered on the PVC pipe at plot center. We described cover of forbs and ferns < 0.5 m tall, low shrubs 0.5-2 m, midstory shrubs and understory trees > 2 m, and overstory trees. We counted live and dead trees (> 2 m tall) by species and by size class within 12.6 m. Size classes were 5 to 20 cm d.b.h., >20 to 40 cm d.b.h., >40 to 60 cm d.b.h., and >60 cm d.b.h. Within the 12.6-m-radius plots, we used a four-category subset of the octave scale (where 1 = 0 to 12 percent; 2 = > 12 to 24 percent; 3 = > 24 to 48 percent; 4 = > 48 to 100 percent) to estimate the percentage of volume filled by forbs, ferns, low shrubs, midstory shrubs, and trees in cylinders 0 to 0.5 m, 0.5 to 2 m, and 2 to 10 m in height.

Vegetation plots were established and variables first measured during summer 1994, 16 to 18 months after completion of the experimental thinnings. Permanent plots were revisited in summer 1996 and will be sampled every 3 to 5 years thereafter for the duration of the study.

Tree age and growth response—The largest tree[26] within 5 m of each grid point was described: tree age, height, crown width, crown ratio, d.b.h., and species were recorded. In 1993 and 1994, all 64 grid-point trees in each of the 16 grids (1024 total; 985 live trees) were cored with an increment borer at breast height. Tree height and height to live crown were measured with a tree laser (Laser Technology Inc., Criterion 400). Tree increments were used to assess the age distribution of the dominant trees in the stands. These dominant and codominant trees were used to characterize the site class of each forest and stand and will be used to assess growth of the retained overstory trees during the life of the experiment.

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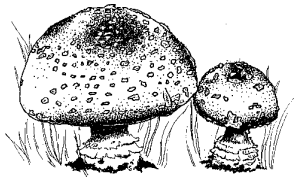
[25] The following scale was used for cover values: 1 = 0-1 percent cover; 2 = >1-3 percent; 4 = >3-6 percent; 8 = >6-12 percent; 16 = >12-24 percent; 32 = >24-48 percent; 64 = >48-100 percent.

[26] These are the same trees on which arboreal mammal traps were placed.

Den Use

Supplemental nestboxes and cavities were checked for use twice per year beginning in winter 1993. Arboreal mammals were captured and processed as per Carey and others (1991a). Animals were handled minimally and immediately placed back in the box when all data had been obtained. In addition, nest materials, food items, or den use by nontarget animal species were noted. Summer den checks were conducted to roughly coincide with the presence of northern flying squirrel young of the year. If young were found that did not have fur developed, they were noted, but left untouched.

Fungi and Soil Food Webs



Pretreatment epigeous fungi—Epigeous fungi were initially sampled during fall 1992 (November 8-24) to assess effects of previous forest management practices on epigeous fungal diversity and to establish preliminary fungal biodiversity data. No attempt was made at that time to assess fungal biomass production. In each of the eight stands designated as not-to-be-thinned, six 4-m^2 plots were selected for fungal sampling. Plots were 5 m northwest of randomly selected grid station points. Within sample plots, all exposed fungi were removed, tagged, and placed in wax paper bags for transport to the laboratory, where specimens were identified.

Posttreatment epigeous fungi—A randomly selected subset (n = 128) of the 191 permanent plots was sampled yearly to evaluate effects of experimental thinning on species composition and biomass production of sporocarps of epigeous fungi. All subtreatments and nonthinned stands were sampled with equal intensity (four plots per subtreatment per stand and four plots per unthinned stand per session). Species composition and biomass of all macrofungi (> 1 cm diameter) were recorded on the 100-m^2 plots during the peak of the fall fungal seasons in 1994, 1995, and 1996. We identified fungi in the field or collected specimens of all unknown taxa for later identification; identification was with fresh specimens where possible. Voucher specimens were kept for most species; characteristics of fresh sporocarps, including color, odor, staining, etc., were noted, as was the substrate from which they grew. Voucher specimens will be deposited at the Oregon State University Herbarium, Corvallis.

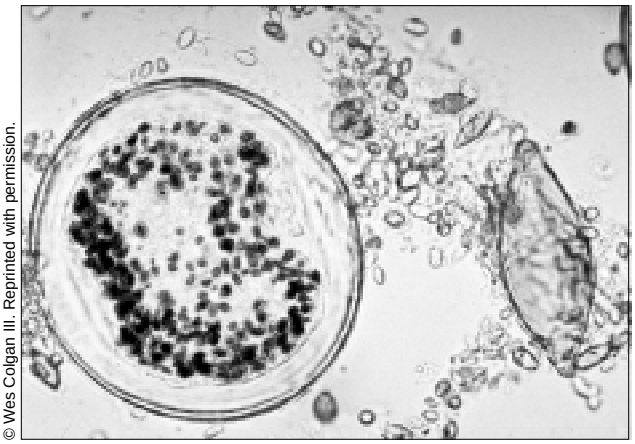
Epigeous fungal sporocarp biomass production was estimated by developing fungal cap diameter to dry weight regression equations for groups of fungi (e.g., groups of similar life form fungi such as gilled, stalked mushrooms, *Russulas* only, or chanterelles). In this way fungal biomass production was estimated without repeated destructive sampling. We anticipate this study will continue yearly. When possible, a second yearly sampling will take place during spring; however, pilot studies have indicated that epigeous sporocarp production during spring is minimal compared to fall.

Identification of many species of epigeous fungi to the species level is difficult, if not impossible, especially for complex genera such as *Cortinarius*, *Inocybe*, and *Russula*. We report trends in diversity of fungal taxa; we used genus and species names from regional guides, including Arora (1986), Phillips (1991), and Tylutki (1979, 1987). Because of the uncertainty surrounding species concepts in fungi[27] and the preliminary

[27] For those fungal taxa listed in table C-3 of USDA and USDI (1994) (e.g., "survey and manage" species), much of the taxonomic and nomenclatural uncertainty has recently been clarified by Castellano and O'Dell (1997). For other taxa, questions of synonomy and nomenclature will require further investigation and will be reported in the future.

nature of our report, we do not purport to present all synonyms for, or the complete nomenclatural history of, all epigeous fungal taxa included in this report. In general, fungal authorities for epigeous fungi follow Phillips (1991) and Tylutki (1979, 1987). Additional authorities and some commonly encountered synonyms were provided by Dr. J.M. Trappe of Oregon State University. For fungal taxa found in table C-3 of USDA and USDI (1994), nomenclature follows Castellano and O'Dell (1997). The list in appendix 1 is a preliminary summary of epigeous fungal diversity on the study sites. Future research will clarify many species concepts and nomenclatural questions; if possible, we will amend our findings as these results are published.

We distinguished between two groups of epigeous fungi: saprobic and mycorrhizal.[28] We assigned mycorrhizal status to genera for which mycorrhizal potential has been reported in, Miller (1983), Singer (1986), and Trappe (1962). We classified as mycorrhizal all species of the genera *Amanita*, *Boletus*, *Cantharellus*, *Clavaria*, *Cortinarius*, *Dentinum*, *Entoloma sensu stricto*, *Gomphidius*, *Hebeloma*, *Hygrophorus, Inocybe*, *Laccaria*, *Lactarius*, *Lycoperdon*, *Russula*, *Suillus*, *Thelephora*, and *Tricholoma*. We report average ectomycorrhizal species richness per plot and total ectomycorrhizal species richness for experimentally thinned and unthinned sites in SS and FH for the fall sampling sessions of 1994-96.



Hypogeous fungi (truffles)—Truffles (hypogeous sporocarps of the fungal subdivisions Zygomycotina, Ascomycotina, and Basidiomycotina) were sampled every 5 to 7 weeks from April 1993 (immediately after the thinnings) until December 1995. Truffles were collected from each of 10 circular 4-m^2 plots systematically located 10 m apart along randomly placed transects in each thinning subtreatment and in nonthinned stands (160 truffle plots per sampling period). Stands with and stands without supplemental dens were sampled in alternating sessions. At each truffle plot, vegetation was removed, plots were raked to

[28] Mycorrhizal fungal types can be described in relation to two axes or gradients: from lack of penetration of root cortical cells to penetration, and from root enclosure by hyphae to nonenclosure (Allen 1991). Root enclosure by fungal hyphae and lack of root cortical cell penetration characterize the ectomycorrhizae, and nonenclosure and cortical cell penetration characterize the endomycorrhizae, of which the vesicular-arbuscular (VA) mycorrhizae are a common form. The family Pinaceae, and Douglas-fir in particular, are characterized by a dominance of ectomycorrhizal fungi—the hypogeous truffles and epigeous mushrooms. Where we refer to mycorrhizal fungi, we imply ectomycorrhizal fungi. Otherwise, we refer to endomycorrhizal or VA fungi where we need to distinguish between them and ectomycorrhizae.

mineral soil depth (about 5 cm), and all fungal sporocarps were collected, identified to species, and dried for biomass determinations. Truffle species identifications were provided by Dr. J.M. Trappe of Oregon State University. Voucher specimens for all hypogeous species were deposited at the Oregon State University Herbarium, Corvallis.

Soil food webs—Soil food web samples were taken in spring and fall 1994 and 1995 from a subset (n = 100) of 4-m^2 plots offset from permanent plot centers (to avoid disturbance of permanent plots). The percentage of cover of recognizable ectomycorrhizal mat types (*Piloderma* sp., *Hysterangium* sp., *Gautieria* sp.) was measured and mapped, and about 100 g of soil was collected from each plot. Soil was placed in plastic bags and stored in an insulated cooler during travel. Samples were then stored at 4 °C and microbial activity analyzed within 5 days of collection. The following parameters were evaluated: (1) grams dry weight (gdw) per gram of soil; (2) active and total bacterial and fungal biomass (µg/gdw); (3) total nematodes (no./ gdw); and (4) percent fungal-feeding, percent bacterial-feeding, percent plant-feeding, and percent predatory nematodes. Active and total bacterial and fungal biomass (AB, AF, TB, TF) were determined as described by Babiuk and Paul (1970) and Ingham and Klein (1984) and were used to compute the ratios, TF/TB and AF/AB. Nematodes were extracted as described by Anderson and others (1978) and were separated into trophic groups based on morphological characteristics used in taxonomic classification (Bongers 1989).

Coarse Woody Debris, Snags, and Potential Dens

Pretreatment coarse woody debris and snags—Grid lines in each of the 16 stands were used as a template to sample CWD cover by using a line-intercept method (Van Wagner 1968, Van Wagner and Wilson 1976). Snags were sampled in every other grid cell in all stands. For sampling fallen trees, 4 to 6 interior lines in each stand (lines B through G, rows 2 through 7) were sampled, with a total of 800 to 1200 m of line sampled per stand. Fallen trees were classified by origin (originating with the previous or current stand), size class (trees with a minimum large end diameter of 15 cm were recorded), and decay class. For fallen trees greater than 15 cm d.b.h., the length of transect covered by the fallen tree was recorded—if the line intercepted the log on a diagonal, the diagonal measurement was recorded. If numerous logs were piled on each other, only the top log was recorded. For large fallen trees (greater than 30 cm diameter), the following information was recorded: species, diameter in centimeters at each end, length (meters), decay class, and origin (originating from the previous or current stand). For large

fallen trees with roots still attached, the large-end diameter was recorded at a height above the base equal to breast height. We used a three-decay-class system for fallen logs, modified from the five-decay-class classification system developed by Fogel and others (1973) and modified by Sollins (1982). Decay classes 1 and 2 of Fogel and others (1973) were treated as class I; classes 3 and 4 were treated as class II; and decay class 5 was treated as class III.

All snags greater than 15 cm d.b.h. and taller than 1.4 m were inventoried in alternating grid cells in each stand. All standing trees 15 to 30 cm d.b.h. were tallied by using a simple dot-count method. Detailed measurements were recorded for standing dead trees greater than 30 cm diameter. For larger snags, we recorded species, d.b.h., height, and decay class from the five-class system of Cline and others (1980).

Postthinning coarse woody debris—Grid lines used in the prethinning CWD survey were resampled after thinning to assess effects of thinning operations on CWD cover and distribution. The three-decay-class classification system was used, and the same size of CWD was sampled as during the prethinning sampling. Grid lines were assigned one of six designations, depending on which type of thinning subtreatments the lines separated. The six categories were (1) light thin-light thin; (2) light thin-heavy thin; (3) light thin-root rot thin; (4) heavy thin-heavy thin; (5) heavy thin-root rot thin; and (6) root rot thin-root rot thin.

Den availability—After completion of the experimental thinnings, we inventoried 100 percent of all 16 stands (plus a surrounding 20-m buffer) for residual snags, residual stumps, residual live trees, conifers with sticknests, conifer snags with cavities, conifer snags without cavities, deciduous snags with cavities, and deciduous snags without cavities. Residual stumps were those with sapwood in an advanced state of decay and were ≥ 1 m high and ≥ 25 cm diameter. Snags were ≥ 2 m high, ≥ 25 cm diameter, and characterized as residual, current, or deciduous. Stands were sampled in summer 1993.

Forest Floor and Arboreal Mammals

Arboreal rodents—Arboreal rodents were trapped on all 16 grids twice yearly (fall 1991, and spring and fall 1992-96). We trapped for two consecutive weeks in each session, opening and baiting traps on Monday morning, checking traps each of the following four mornings (Tuesday-Friday), and closing traps on Fridays. Arboreal rodent trapping data will be reported here only for the baseline period (fall 1991 and spring and fall 1992). We summed captures and counted individuals for the three arboreal species (flying squirrel, Townsend's chipmunk, and Douglas' squirrel) for 1991-92. Chipmunks were not sampled in 1991. We estimated mean densities of flying squirrels (three seasons) and chipmunks (two seasons) per stand (Carey and others 1991a), combining the Chapman modification of the Lincoln-Peterson Index with grouping captures by week and estimating area sampled with site-specific mean maximum distances moved. Forest means were calculated from stand means. All reported data are from prethinning sampling and do not consider the effects of supplemental dens.

During trapping, flying squirrels were radio-collared to document movement patterns and den use; telemetry and den site data will be reported elsewhere. Den site characteristics and abundance are reported in Carey and others (1997). Concurrent with flying squirrel trapping, extensive descriptive notes were taken on captured animals and were used to assess flying squirrel age and reproductive status (Villa and others 1999) as well as foraging activity and use of nontruffle foods (Thysell and others 1997). During trapping, fecal pellets were collected and placed in ethanol and later were analyzed for dietary components. The protocol for trapping is outlined in Carey and others (1991a); the protocol for fecal pellet analysis follows McIntire and Carey (1989) and Colgan and others (1997).

Forest floor small mammals—Forest floor small mammals were trapped for 2-week sessions during summers of 1992, 1993, and 1994 in the eight randomly selected, not-to-be-thinned stands. In summer 1996 and 1997, all 16 stands (thinned and unthinned) were trapped for forest floor small mammals. We report small mammal trapping data from 1992 and 1993 only. Trapping effort was 6,400 trap nights in each forest (FH and SS) in each reported year. Small mammal abundance was calculated as captures per 100 corrected trap nights (CPUE) (Nelson and Clarke 1973). Abundance was calculated for each of the four trapped stands per forest per year; we report the mean CPUE $\pm$ SE for each forest in each year based on these four stand values.

For both arboreal and forest floor mammals, we made extensive use of necropsies to evaluate field identification of species, sex, age, and reproductive condition. Additionally, species identifications were confirmed by the Burke Museum, University of Washington, and voucher specimens were deposited in that museum.

Predators and Birds

Owl survey—In spring 1992, we conducted an owl survey to establish baseline data on owl diversity and abundance. In each forest (FH and SS) we surveyed for owls in an area extending about 0.5 mile out from the experimental stands. We established a network of calling stations within each forest—stations that allowed an appreciable amount of audible overlap between adjacent stations when hooting conditions were fair or better. Calling stations were visited three or four times during April and May 1992. At these times, we broadcast calls for the great-horned owl, barred owl, northern spotted owl, barn owl, northern pygmy owl, saw-whet owl, and screech owl.

Since the spring 1992 formal owl survey, we also have noted the presence of owls on the study site by recording the observations of night telemetry personnel. We include a list of owl and other bird species observed on the study site (appendix 1).

Mammals—Though the FES does not incorporate a formal mammalian predator sampling component, we have recorded presence and numbers of mustelids caught while we were trapping for arboreal mammals. To date, we have radio-collared two weasels caught during trapping sessions. We report mammalian predators and all other mammalian, amphibian, and reptilian species observed on the study site (appendix 1). A report on short- and long-tailed weasels, drawn from experiences at the FES site and other Pacific Northwest arboreal mammal study sites, can be found in Wilson and Carey (1996).

Winter bird survey—Beginning in winter 1996, surveys were initiated to determine the effects of the experimental thinnings on winter bird populations. Four experimentally thinned and four nonthinned stands were selected for a study designed to determine the feasibility of using standard bird census techniques to assess experimental silvicultural prescriptions. The study used a simple point-count method (Manuwal and Carey 1991), with nine point-count stations at least 80 m apart and at least 80 m from any habitat edge in each stand. Point-count stations were located at existing grid stations along established trails to facilitate replication and movement between stations by field observers. The method yielded 27 point counts per stand, totaling 108 counts each in thinned and unthinned stands. It is anticipated that the winter bird study will continue yearly for the next 3 to 5 years. A list of all bird species observed on the study site is presented (appendix 1).

Baseline Conditions

Because data collection from the FES is in progress, and because most analyses are preliminary, we present primarily prethinning, between-forest comparisons and summaries for those response variables discussed. In other cases (i.e., coarse woody debris, vascular plant species richness), we present preliminary, postthinning response data where relevant. Because we do not yet consider the effects of supplemental dens on the reported responses, we refer to both den-augmentation-only stands and

Table 5—Mean d.b.h. in centimeters for all Douglas-fir trees; mean d.b.h. in centimeters, mean height in meters, and mean height to live crown in meters (± S.E.) for Douglas-fir site trees from pretreatment prism plots, by forest

Variable	Star/Stellar	Farley/Hill[a]
All Douglas-fir prism plot trees:		
Diameter at breast height [d.b.h. in centimeters (sample size)]	33.7 ± 0.5 (763)	**53.4** ± 0.6 (600)
Douglas-fir site trees:		
Diameter at breast height [d.b.h. in centimeters (sample size)]	42.8 ± 0.7 (236)	**60.5** ± 1.0 (230)
Height (m)	33.7 ± 0.4	**41.8** ± 0.4
Height to live crown (m)	18.8 ± 0.3	**21.2** ± 0.4

[a] Bold numerals indicate significant difference between forests for that variable (Mann-Whitney U-test [Norušis 1993]; $p < 0.01$).

control stands as controls unless it is necessary to distinguish between the two. Thus, contrasts are presented between experimentally thinned and unthinned stands without regard to supplemental dens. We present a comprehensive checklist of all vascular plants, fungi, birds, mammals, reptiles, and amphibians observed on FES stands to date (appendix 1).

Prethinning and Control Stand Conditions

Overstory—The overstory vegetation for all stands was dominated by Douglas-fir (table 2). Tree species other than Douglas-fir were rarely encountered in either the prism plots or permanent plots or during the postthinning stand assessment. Stands in the SS blocks were similar to one another in tree size and understory vegetation, but differed from the FH stands, which were also similar to one another (table 3). The average diameter of all sampled Douglas-fir prism plot trees was significantly greater in FH than in SS, and the dominant (site tree) prism-plot Douglas-fir trees in FH were significantly larger in d.b.h., height, and in height to live crown than those in SS (tables 3 and 5). The same between-forest differences were evident in the larger sample of permanently marked trees at gridpoints (table 6).

Prethin relative densities (Curtis 1982) in Star and Stellar[29] closely matched normal (see footnote 11) forest stocking; tree densities were high and basal areas were low to moderate. Star/Stellar values were typical for single cohort, closed canopy stands in the stem-exclusion phase of forest development (Oliver and Larson 1990). However, FH stands exhibited conditions more typical of stands managed intensively for wood production; individual tree d.b.h. was greater as a result of thinning-induced reduction in tree density. Farley/Hill exhibited pretreatment and control stand RD values of about 7 metric (50 English)—suggested by Curtis (1982) as a maximum RD to avoid

[29] Stellar 204 was an exception, probably because the stand contained a number of small *Phellinus weirii* openings.

Table 6—Characteristics of trap-placement trees by block[a]

Block name	Douglas-fir	Site index[b]	Site class[c]	Age at breast height ± S.E.	Height ± S.E.	D.b.h. ± S.E.	Crown ratio ± S.E.
	Percent[d]	*Feet (meters)*		*Years*	*Meters*	*Centi-meters*	*Percent*
Star	99.2 (250)	119 (36.3)	Low II	46.9 ± 0.3	33.6 ± 0.2	40.5 ± 0.6	39.0 ± 0.7
Stellar	100 (248)	118 (36.0)	Low II	47.4 ± 0.3	33.2 ± 0.3	40.2 ± 0.6	41.7 ± 0.7
Farley	98.8 (245)	130 (39.6)	High II	59.6 ± 0.4	42.6 ± 0.2	58.3 ± 0.7	37.4 ± 0.7
Hill	97.5 (242)	133 (40.5)	High II	59.6 ± 0.4	42.0 ± 0.3	58.5 ± 0.9	41.1 ± 0.7

[a] Site Index, tree age, tree height, tree d.b.h., and tree crown ratio are for Douglas-fir trees only.

[b] The site index calculations are based on 50-year site curves developed by King (1966).

[c] The site classes are from King (1966).

[d] The number in parentheses is the total number of live, sampled, trap-placement trees per block. The figure preceeding it is the percentage of these live trees that were Douglas-fir.

significant tree mortality from suppression and restriction of crown development. Carey and others (1996a) also suggest RD 7 as a target at age 30 years (given precommercial thinning at 15 years) when managing for biodiversity. Stands would then receive a commercial thinning.

The SS pretreatment tree d.b.h. distribution curve (fig. 7a) indicated a skewed-normal distribution typical of naturally regenerating, single cohort, even-aged forests (Oliver and Larson 1990). The FH stands (fig. 7b) showed a normal d.b.h. distribution characteristic of intensively managed stands although the distribution is somewhat bimodal. The subdominant curve contained smaller diameter class (10-25 cm d.b.h.), younger trees that have established in the more open FH stand conditions. The bimodal distribution illustrates, in part, stand regenerative processes that occur as a result of thinnings or canopy gaps (Oliver and Larson 1990).

Trap-placement trees (table 6) were similar in species, age, and size distribution to those measured in the prism-plots (see tables 2 and 5 for a comparison). Ninety-nine to 100 percent of SS trap trees were Douglas-fir; 97-99 percent of the FH trap trees were Douglas-fir. Average age of nonresidual Douglas-fir tagged trees was 54 ± 0.3 years in SS and 67 ± 0.4 years in FH,[30] indicating essentially even-aged cohorts of trees with stand initiation occurring 3 to 5 years after previous harvests.

[30] Total age for site index II trees was determined by adding 7 to the age at breast height (King 1966). We counted an average of 47 rings and 60 rings at breast height, respectively, for trap trees in SS and FH (table 6).

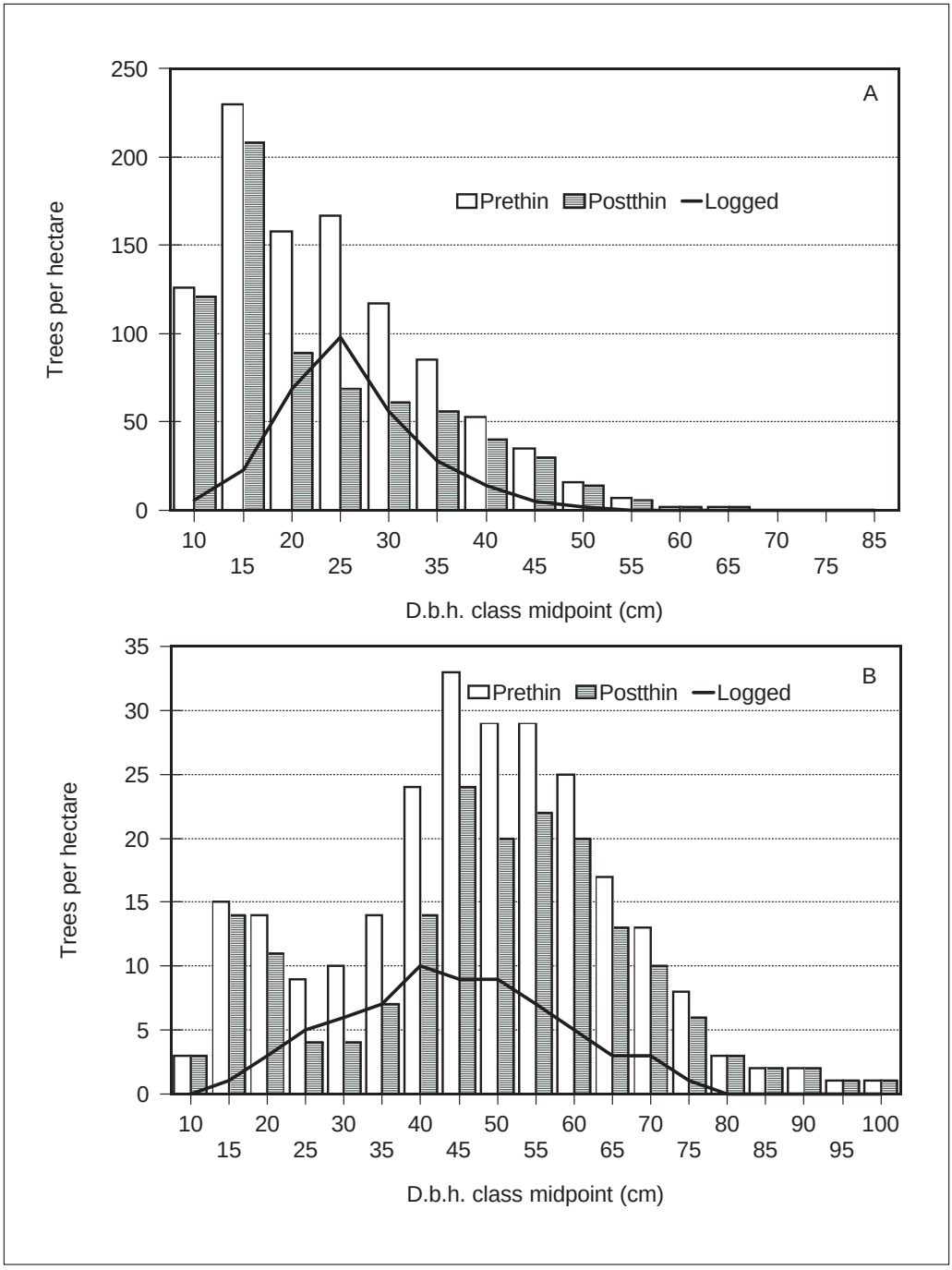


Figure 7—Distribution of d.b.h. classes of prethin, postthin, and logged trees over 7.5 cm d.b.h. in the Star/ Stellar stands (A) and Farley/Hill stands (B). D.b.h. classes are midpoints of 5-cm ranges (e.g., the 40-cm class contains trees from 37.5 to 42.49 cm d.b.h.) Number of trees are the average number per hectare for each d.b.h. class and category.

Table 7—Prethin and postthin vascular plant species richness per plot by *post facto* relative density (RD) class in 1994, 1 year after thinning

		Postthin[b]					
				Post facto RD class[d]			
Forest	Prethin[a]	Control	All thinned[c]	RD8	RD6	RD4	RD2
	----------	*Mean number of species per plot ± S.E. (sample size)*					----------
Star/Stellar	8.6	14.8	24.1	23.7	19.8	24.1	29.7
	± 0.3	± 0.6	± 0.9*	± 1.5	± 1.3	± 1.5	± 1.4
	(120)	(24)	(71)	(6)	(27)	(17)	(21)
Farley/Hill	**15.1**[e]	**18.5**	**34.6**	29.4	**31.4**	26.9	**44.5**
	± 0.4	± 1.2	± 1.2*	± 2.1	± 1.5	± 2.0	± 1.4
	(120)	(24)	(72)	(12)	(22)	(14)	(24)

[a] Plot size = 25 m^2.

[b] Plot size = 100 m^2.

[c] Asterisks(*) indicate significant difference between all thinned plots and CT plots for that forest (t-test; $p < 0.01$).

[d] Classes are defined as follows: RD2 = retained RD of < 3.25 (metric); RD4 = retained RD of 3.25 ≤ RD < 4.75; RD6 = retained RD of 4.75 ≤ RD ≤ 6.75; RD8 = retained RD > 6.75.

[e] Bold numerals indicate significant difference between SS and FH for that treatment (Mann-Whitney U-test (Norušis 1993); $p < 0.05$).

Prior to experimental thinning, western hemlock regeneration occurred infrequently on all study stands; Douglas-fir regeneration dominated in both forests. The relatively dry climate and soils of the region permit regeneration of Douglas-fir in the understory. Given adequate time and absence of disturbance, the stands may develop western hemlock associations.[31] The study stands closely resemble the *Tsuga heterophylla/ Gaultheria shallon/Polystichum munitum* and *Pseudotsuga menziesii/Gaultheria shallon* associations described by Henderson and others (1989), and the *Pseudotsuga menziesii/Tsuga heterophylla* and *Pseudotsuga* types of the Olympic montane forests described by Fonda and Bliss (1969). Puget lowland forests are, however, distinctly different from forests of the Olympic Peninsula and eventually may be recognized as such (Franklin and Dyrness 1973).

Understory—Pretreatment understory vegetation at SS was highly variable and reflected the size of overstory openings. Star/Stellar stands generally had a very dense overstory and, subsequently, had minimal understory development. Ground mosses were abundant, averaging over 50 percent cover in these stands, with dominant mosses being *Kindbergia oregana* (Sull.) Ochyra (= *Eurhynchium oreganum*) and *Hylocomium splendens* (Hedw.) B.S.G. Other mosses occurring in these stands

[31] Nonetheless, the stumps remaining from the clearcut entries in the 1920s and 1930s are almost entirely Douglas-fir, suggesting presettlement history of fires or other disturbances that maintained Douglas-fir as the dominant tree species.

Table 8—Mean cover of vascular plant subcanopy layers by stratum by *post facto* relative density (RD) class for each forest type in 1994, 1 year after thinning

Treatment class[a]	Farley/Hill stratum[b c]				Star/Stellar stratum			
	Sample size	1	2	3	Sample size	1	2	3
		--Percent cover ± S.E.--				*--Percent cover ± S.E.--*		
Control	24	**28.3** ± 4.3	**48.3** ± 4.7	**10.0** ± 2.0	24	17.7 ± 4.6	25.8 ± 4.1	6.3 ± 1.9
RD8	12	**61.3** ± 2.7	25.3 ± 6.2	**8.3** ± 2.6	6	5.3 ± 0.8	18.0 ± 4.8	0.7 ± 0.4
RD6	22	**49.5** ± 3.9	**36.6** ± 4.6	**7.2** ± 1.0	27	8.4 ± 2.5	11.3 ± 2.0	2.4 ± 0.7
RD4	14	**48.0** ± 5.3	24.7 ± 4.6	**15.9** ± 3.1	17	10.9 ± 1.8	15.1 ± 1.9	2.9 ± 1.0
RD2	24	**62.7** ± 1.3	14.5 ± 1.8	**5.9** ± 1.5	21	26.5 ± 4.0	21.4 ± 4.0	3.3 ± 1.5

[a] Classes are defined as follows: RD2 = retained RD of < 3.25 (metric); RD4 = retained RD of 3.25 ≤ RD < 4.75; RD6 = retained RD of 4.75 ≤ RD ≤ 6.75; RD8 = retained RD > 6.75.

[b] Stratum 1 included forbs and ferns < 0.5 m tall; stratum 2 included low shrubs 0.5 to 2.0 m tall; stratum 3 included midstory shrubs and understory trees > 2.0 m tall.

[c] Bold numerals indicate significant difference between Farley/Hill and Star/Stellar for a particular treatment and RD class (Mann-Whitney U-test [Norušis 1993]; $p < 0.05$).

included *Rhytidiadelphus loreus* (Hedw.) Warnst, *R. triquetrus* (Hedw.) Warnst, *Plagiothecium undulatum* (Hedw.) B.S.G., *Plagiomnium insigne* (Mitt.) Kop., *Leucolepis acanthoneuron* (Schwaegr.) Lindb. (= *L. menziesii*), *Isothecium stoloniferum* Brid., and *Dicranum scoparium* Hedw. (see footnote 7). Where openings occurred, understory vegetation was dominated by salal, Oregongrape, swordfern, hazel, Scouler's willow, and oceanspray.

Pretreatment understory vegetation of FH was more diverse than at SS. The former stands were more open, incident light was higher in the understory, and species richness was significantly greater. Of 120 pretreatment vegetation plots in each forest, FH plots averaged 15.1 understory vascular plant species (range = 5 to 31) per 25-m^2 plot, compared to an average of 8.6 species in SS (range = 3 to 16) (table 7). Control and den-augmentation-only stand permanent plots in FH also had significantly higher vascular plant species richness than did those in SS (table 7). Individual plants were often more robust in FH, and dominants (salal, swordfern, Oregongrape, oceanspray, hazel, and brackenfern) had higher cover values than at SS (table 8). Moss cover was substantially less in FH than in SS, probably due to the extensive development of deciduous understory shrubs and forbs. Altogether we found about 90 species of vascular plants on FES stands during our pretreatment and control stand sampling (table 9).

Table 9—Vascular plant species found in control permanent plots and in prethinning understory vegetation plots, by block

Plant taxon[a]	Farley (species richness = 60)	Hill (species richness = 73)	Star (species richness = 38)	Stellar (species richness = 37)
Acer macrophyllum	•	•		
Achlys triphylla	•	•	•	•
Actaea rubra	•			
Adenocaulon bicolor	•	•		
Alnus rubra	•	•		
Amelanchier alnifolia	•	•	•	•
Anemone deltoidea	•		•	
Anaphalis margaritacea		•		
Arbutus menziesii	•			
Asarum caudatum		•		
Athyrium filix-femina	•			
Blechnum spicant	•	•		
Bromus vulgaris	•	•		
Carex deweyana	•	•		
Campanula scouleri	•	•		•
Chimaphila menziesii			•	
Chimaphila umbellata		•	•	
Corylus cornuta* var. *californica	•	•	•	•
Collomia heterophylla				•
Corallorhiza maculata	•		•	•
Cornus nuttallii	•	•		
Crepis capillaris		•		
Cytisus scoparius		•		
Disporum hookeri			•	•
Dryopteris campyloptera	•		•	
Elymus glaucus var. *glaucus*	•	•		
Epilobium angustifolium		•		
Festuca occidentalis	•	•	•	•
Festuca subulata		•		
Festuca subuliflora	•			•
Fragaria virginiana var. *platypetala*		•		
Frangula purshiana	•	•	•	•

Table 9—Vascular plant species found in control permanent plots and in prethinning understory vegetation plots, by block (continued)

Plant taxon[a]	Farley (species richness = 60)	Hill (species richness = 73)	Star (species richness = 38)	Stellar (species richness = 37)
Galium aparine	•	•	•	•
Gaultheria shallon	•	•	•	•
Geum macrophyllum var. *macrophyllum*		•		
Goodyera oblongifolia	•	•	•	•
Hieracium albiflorum		•		
Holodiscus discolor	•	•	•	
Holcus lanatus		•		
Hypochaeris radicata	•	•		
Ilex aquifolium	•	•	•	•
Lathyrus polyphyllus	•	•	•	
Leucanthemum vulgare	•	•		
Linnaea borealis	•	•	•	•
Listera caurina	•	•		•
Lonicera ciliosa	•	•	•	•
Lotus micranthus		•		
Luzula parviflora		•		
Mahonia aquilinum		•		
Mahonia nervosa	•	•	•	•
Maianthemum stellatum				•
Melica subulata	•	•		
Moehringia macrophylla				•
Monotropa uniflora			•	
Mycelis muralis	•	•		
Nemophila parviflora	•	•		
Oemlaria cerasiformis	•	•	•	
Osmorhiza berteroi	•	•		
Polypodium glycyrrhiza		•		•
Polystichum munitum	•	•	•	•
Prunella vulgaris		•		
Pseudotsuga menziesii	•	•	•	•
Pteridium aquilinum	•	•	•	•
Pyrola aphylla			•	•

Table 9—Vascular plant species found in control permanent plots and in prethinning understory vegetation plots, by block (continued)

Plant taxon[a]	Farley (species richness = 60)	Hill (species richness = 73)	Star (species richness = 38)	Stellar (species richness = 37)
Pyrola asarifolia		•		•
Ribes sanguineum		•		
Rosa gymnocarpa	•	•		•
Rubus discolor		•		
Rubus laciniata		•		
Rubus leucodermis	•	•		
Rubus parviflora		•		•
Rubus spectabilis	•	•		
Rubus ursinus	•	•	•	•
Sambucus racemosa	•	•	•	
Salix scouleriana	•	•		
Senecio jacobea	•	•		
Senecio sylvaticus		•		
Stellaria crispa	•	•		•
Symphoricarpos alba	•	•		•
Symphoricarpos hesperius	•	•	•	•
Taxus brevifolia			•	
Thuja plicata			•	
Tiarella trifoliata	•	•		
Trientalis borealis* ssp. *latifolia	•	•	•	•
Trillium ovatum	•	•	•	•
Tsuga heterophylla			•	
Urtica dioica	•			
Vancouveria hexandra	•	•	•	•
Vaccinium ovatum	•	•	•	
Vaccinium parvifolium	•	•	•	•
Vicia gigantea	•			
Viola sempervirens	•	•	•	•

[a] Species found in all 4 blocks are in bold. Introduced species are underlined. Authorities and common names are found in appendix 1.

Prethinning Coarse Woody Debris

The fallen tree components of the two forests differed markedly. Star/Stellar stands averaged 7 to 8 percent CWD cover (over 15 cm diameter large end), compared to 2 to 3 percent cover in the eight FH stands (table 10). The subset of stands (101, 103, 201, 202, 302, 303, 401, 403) selected for thinning differed similarly (table 11).

Some large trees were retained during the 1937 regeneration harvest in SS. Although SS had not been thinned or harvested since that entry, about one-third of residual standing trees were felled in 1986, because they were thought to pose a hazard to low-flying helicopters. Thus, SS contained three distinct populations of fallen trees: (1) small-diameter fallen trees from the current stand (trees that died from suppression or root rot); (2) large-diameter and well-decayed fallen trees from the previous stand; and (3) larger diameter, relatively undecayed trees from felling operations of 1986.

Farley/Hill stands contained minimal coarse woody debris; little remained in the stands except for what had fallen since the last thinning entry (table 10). At the time of the last thinnings, the entire, harvested trees were yarded to landings and bucked into logs; tree crowns and cull material were burned on site. Debris in the stand was piled and burned.

Standing Dead Trees

Total standing dead tree (snag) density was significantly greater in SS than in FH (table 12). On average, there were 34 snags/ha (> 15 cm d.b.h.) in SS vs. 19 snags/ha in FH. Standing dead trees also had three distinct size and decay class distributions: (1) small current-stand material (e.g., part of the cohort of trees currently in place); (2) large-diameter and tall residual standing dead trees from the previous stand (e.g., trees not cut during the harvest entries of the 1920s and 1930s); and (3) well-decayed and short (< 2 m), standing dead stumps remaining from the harvest of the 1920s or 1930s. Density of large snags (> 30 cm d.b.h.) was greater in SS than in FH, but not significantly so. Large snags in SS were significantly larger in diameter than in FH, but FH large snags were significantly taller than those in SS. Snags retained from the preceding stand (bark sloughed, stems well decayed, all but the largest branches completely missing) in SS were similar in diameter to, but significantly taller than, those in FH. Residual snags also were significantly more abundant in SS than in FH. Snags from the current cohort of trees were significantly larger and significantly taller in FH than in SS. Density of current snags was greater in FH than in SS (3.6 snags/ha vs. 1.6 snags/ha); however, this difference was not statistically significant (table 12).

Arboreal Rodents

Arboreal rodent communities in both forests were composed of the northern flying squirrel, Townsend's chipmunk, and Douglas' squirrel. Northern flying squirrels were more abundant in SS than in FH, and the Townsend's chipmunk was more abundant in the FH. Douglas' squirrels were caught infrequently in both forests (table 13).

Forest-Floor Small Mammals

Forest-floor small mammal communities in both forests included two shrew species (Trowbridge's and montane), the deer mouse, the Oregon creeping vole, the shrew-mole, and the southern red-backed vole. Two additional species, the Columbian deer mouse and the vagrant shrew, occurred occasionally in both forests. Other infrequently caught species included the Pacific jumping mouse, the coast mole, and the Pacific water shrew. Although species richness was similar in both forests, overall abundance of forest-floor small mammals was significantly higher in FH than in SS in 1992 and in 1993 (table 14).

Table 10—Prethinning and postthinning coarse woody debris cover

Forest	Prethinning[a b]		Postthinning[c]
	All stands	Stands to be thinned	
	Mean percent cover ± S.E. (sample size[d])		
Star/Stellar	**7.4** ± 0.5 (32)	**7.1** ± 0.5 (16)	**5.2** ± 0.3 (24)
Farley/Hill	2.5 ± 0.2 (48)	2.9 ± 0.4 (24)	1.6 ± 0.1 (24)

[a] Data are from all 16 stands and for the 8 stands designated as to-be-thinned (e.g., 101, 103, 201, 202, 302, 303, 401, 403).

[b] Bold figures indicate a significant difference between forests for that category or subset (Mann-Whitney U-test [Norušis 1993]; $p < 0.01$).

[c] Data are from thinned stands only (e.g., 101, 103, 201, 202, 302, 303, 401, 403).

[d] Number of sampled 200-m lines.

Table 11—Coarse woody debris (CWD) cover before and after thinning by decay class by forest, thinned stands only

CWD decay class[a]	Star/Stellar[b] (sample size = 16[c])		Farley/Hill[b] (sample size = 24[c])	
	Prethin	Postthin	Prethin	Postthin
	---------- *Mean percent cover ± S.E.* ----------			
Decay class I	0.4 ±0.1	0.6 ± 0.1	**0.6** ± 0.1	0.8 ± 0.1
Decay class II	3.9 ± 0.4	4.1 ± 0.4	**1.4** ± 0.2	0.7 ± 0.1
Decay class III	**2.8** ± 0.4	0.5 ± 0.2	**0.9** ± 0.3	0.2 ± 0.1
Total CWD	**7.1** ± 0.5	5.2 ± 0.4	**2.9** ± 0.4	1.6 ± 0.1

[a] Our decay classes are modified from the 5-decay-class system of Fogel and others (1973) and modified by Sollins (1982). Decay classes 1 and 2 of Fogel and others (1973) were treated as class I; classes 3 and 4 were treated as class II; and decay class 5 was treated as class III.

[b] Bold figures indicate significant difference between prethinning and postthinning cover for that category (Wilcoxon matched-pairs signed-rank [Norušis 1993]; $p < 0.01$).

[c] Number of sampled 200-m lines.

Table 12—Snag characteristics (mean ± S.E), by forest (prethinning conditions)

Forest and attribute	Snag category[a]			
	All snags >15 cm d.b.h.[b]	All snags >30 cm d.b.h.[c]	Residual >30 cm d.b.h.[c d]	Current >30 cm d.b.h.[c d]
Star/Stellar:				
Density (no./ha ± S.E.)	**34.1** ± 2.4	5.6 ± 0.8	**3.9** ± 0.5	1.6 ± 0.4
D.b.h. (cm) ± S.E.	-	**70.7** ± 2.8	83.5 ± 3.2	38.2 ± 1.1
Height (m) ± S.E.	-	8.7 ± 0.7	**6.0** ± 0.6	15.0 ± 1.6
Farley/Hill:				
Density (no./ha ± S.E.)	18.9 ± 4.5	4.6 ± 1.3	1.0 ± 0.5	3.6 ± 1.0
D.b.h. (cm) ± S.E.	-	54.8 ± 2.3	87.3 ± 6.7	**45.4** ± 1.4
Height (m) ± S.E.	-	**20.6** ± 1.2	4.6 ± 1.0	**25.3** ± 1.2

- = not applicable; d.b.h. and height were not measured on snags < 30 cm d.b.h.

[a] Bold figures indicate significant difference between forests for that attribute (Mann-Whitney U-test [Norušis 1993]; $p < 0.05$).

[b] Densities of all snags > 15 cm are based on complete snag counts in every other 40- by 40-m grid cell in each of the 8 stands per forest (hence 196 grid cells per forest).

[c] For all snags > 30 cm d.b.h, sample size = 170 in SS and 141 in FH; for residual snags, sample size = 120 in SS and 31 in FH; for current snags, sample size = 50 in SS and 110 in FH.

[d] Residual snags show evidence of well-decayed sapwood (and heartwood), typically have lost their bark, and have lost most or all of their branches. Current snags have most of their bark intact, are less decayed, and retain more branches (see Cline and others 1980 for a discussion of snag decay).

Table 13—Total summed captures, total captured individuals, mean density, and range of density of arboreal mammals, by forest[a]

Species	Farley/Hill		Star/Stellar	
	Total captures/ total individuals	Density	Total captures/ total individuals	Density
		No./ha ± S.E. (range)		*No./ha ± S.E. (range)*
Flying squirrel	207/126	0.3 ± 0.1 (0.18 - 0.46)	384/214	0.6 ± 0.1 (0.28 - 1.04)
Townsend's chipmunk	641/312	0.7 ± 0.1 (0.29 - 1.43)	100/34	0.1 ± 0.02 (0.0 - 0.28)
Douglas' squirrel	29/20	-	21/20	-

- = not calculated.

[a] Data are summaries (captures, individuals) or means (density) of fall 1991 and spring and fall 1992 trapping sessions, and represent prethinning conditions only.

Table 14—Mean captures per 100 corrected trap nights (± S.E.) for all forest floor small mammal species combined, and for selected species, by forest[a]

	1992		1993	
Species	Farley/Hill[b]	Star/Stellar	Farley/Hill[b]	Star/Stellar
All species	13.9 ± 1.1**	9.5 ± 0.3	11.4 ± 1.5*	7.8 ± 0.7
Peromyscus maniculatus (deer mouse)	2.5 ± 0.4***	0.6 ± 0.2	0.7 ± 0.1**	0.15±0.03
Microtus oregoni (Oregon creeping vole)	2.1 ± 0.6**	0.2 ± 0.1	1.8 ± 0.4***	0.07±0.04
Sorex monticolus and *S. trowbridgii*	3.1 ± 0.4	2.6 ± 0.4	3.5 ± 0.3	2.7 ± 0.4

[a] Data are from unthinned (control) stands only (102, 104, 203, 204, 301, 304, 402, 404).

[b] FH captures in a given year differ significantly from SS captures (*t* - test): * = $p < 0.10$; ** = $p < 0.05$; *** = $p < 0.01$.

The deer mouse and Oregon creeping vole accounted for most of the between-forest difference in overall small mammal abundance in both years (table 14). In 1992, deer mice and Oregon creeping voles were significantly more abundant in FH than in SS. In 1993, both taxa were again significantly more abundant in FH than in SS. Other taxa were trapped consistently and frequently (e.g., southern red-backed vole, Trowbridge's and montane shrews, shrew-mole), or infrequently (e.g., Columbian deer mouse, vagrant shrew) in both forests in both years, and did not greatly contribute to differences in overall abundance between forests. Trowbridge's shrew and the montane shrew were numerically the most abundant forest-floor small mammals in both forests in both years (table 14).

Postthinning Conditions

Silvicultural Conditions

All descriptions provided for stand and subtreatment forest parameters considered standing trees, both live and dead, with a d.b.h. > 20 cm (the exception being forest-level d.b.h. distributions where smaller trees were also measured—see below). Stand and forest-level descriptions are based on weighted means derived from the actual distribution of *post facto* thinning classes for each stand. Subtreatment-level descriptions are based on forestwide averages for each *post facto* thinning class.

Stand and forest level stocking conditions—Thinning operations reduced the heavily stocked SS stands from an average of 454 stems/ha to 291 stems/ha and removed about 30 percent of the basal area from SS stands. This represented a reduction from a forestwide average RD of 7.4 before thinning, to RD 4.8 after thinning (table 15). In FH stands, stocking levels were reduced from an average of 203 stems/ha to 147 stems/ha; basal area was reduced by about 24 percent. Relative density in FH treated stands was reduced from an average of 7.9 to a residual mean of 4.7 (table 15).

Table 15—Prethinning and postthinning silvicultural conditions by forest and *post facto* relative density (RD) class

Forest	Treatment class[a]	Sample size[b]	Prethinning			Posttreatment[c]		
			Basal area	Tree density	Relative density (metric)	Basal area	Tree density	Relative density (metric)
			m^2/ha	*No./ha*		*m^2/ha*	*No./ha*	
Star/Stellar	RD2	36	31.5 ± 2.2	351.1 ± 26.3	5.6 ± 0.4	11.4 ± 0.9	108.9 ± 9.6	1.9 ± 0.1
	RD4	68	37.3 ± 1.2	410.1 ± 17.7	6.4 ± 0.2	25.3 ± 0.3	263.0 ± 6.6	4.1 ± 0.1
	RD6	66	49.1 ± 1.0	503.2 ± 13.5	8.3 ± 0.2	35.9 ± 0.5	353.6 ± 6.3	5.8 ± 0.1
	RD8	26	60.4 ± 1.9	589.4 ± 24.3	10.2 ± 0.3	49.1 ± 1.3	457.6 ± 14.9	7.9 ± 0.2
	All thinned	196	43.2 ± 1.0	454.4 ± 11.0	7.4 ± 0.2	**29.5** ± 0.9	**291.0** ± 8.7	4.8 ± 0.1
	Control	14	42.7 ± 5.1	515.1 ± 60.3	7.5 ± 0.9	42.7 ± 5.1	515.1 ± 60.3	7.5 ± 0.9
Farley/Hill	RD2	42	43.2 ± 3.1	199.4 ± 15.9	7.7 ± 0.6	14.7 ± 0.9	59.1 ± 4.8	2.0 ± 0.1
	RD4	50	35.6 ± 1.1	152.4 ± 6.8	5.4 ± 0.3	30.9 ± 0.5	130.2 ± 3.8	4.0 ± 0.1
	RD6	77	52.8 ± 1.1	216.9 ± 5.6	8.5 ± 0.2	44.1 ± 0.6	178.4 ± 3.4	5.6 ± 0.1
	RD8	27	67.6 ± 1.6	259.2 ± 8.2	10.8 ± 0.4	59.7 ± 0.9	223.5 ± 6.3	7.8 ± 0.1
	All thinned	196	48.4 ± 1.1	202.5 ± 5.1	7.8 ± 0.2	**36.6** ± 1.1	**146.8** ± 4.4	4.7 ± 0.1
	Control	12	44.3 ± 2.5	259.7 ± 15.7	6.5 ± 0.3	44.3 ± 2.5	259.7 ± 15.7	6.5 ± 0.3

[a] Classes are defined as follows: RD2 = retained RD of < 3.25 (metric); RD4 = retained RD of 3.25 ≤ RD < 4.75; RD6 = retained RD of 4.75 ≤ RD ≤ 6.75; RD8 = retained RD > 6.75.

[b] Number of sampled grid cells.

[c] Bold figures indicate significant difference between posttreatment and control stand conditions for that variable and forest (t-test; $p < 0.001$).

Residual stems per hectare and basal area in thinned stands in both forests differed significantly from those of their respective unthinned stands (table 15). In SS, basal area in thinned stands was 29.5 ± 0.9 m^2/ha vs. 42.7 ± 5.1 m^2/ha in SS control stands (t-test, $p < 0.001$). Stem density in SS thinned stands was 291.0 ± 8.7 trees/ha vs. 515.0 ± 60.3 trees/ha in SS control stands (t-test, $p < 0.01$). In FH, basal area in thinned stands was 36.6 ± 1.1 m^2/ha in thinned stands vs. 44.3 ± 2.5 m^2/ha in control stands (t-test, $p < 0.01$). Stem density in FH thinned stands was 146.8 ± 4.4 trees/ha vs. 259.7 ± 15.7 trees/ha in control stands (t-test, $p < 0.001$).

Forest-level d.b.h.-class distributions—Owing to disparate management histories in SS and FH, experimental thinnings had different effects on the resulting d.b.h.-class distributions in the two forests. In SS, with its pretreatment skewed-normal distribution typical of a naturally regenerated stand (Oliver and Larson 1990), thinnings were clearly weighted towards removal of smaller d.b.h. classes (fig. 7a). In contrast, treatment of the previously thinned FH forest achieved a more equitable removal of trees from all d.b.h. classes (fig. 7b). However, some large-diameter trees (> 70 cm d.b.h.) were removed in FH, even though the prescription stipulated thinning from below and leaving large, dominant trees. These large trees probably were removed from RT thinning cells in FH, where only the largest diameter trees or hardwoods were selected for retention. Such trees also may have exhibited low vigor (e.g., thin crowns) or may have been in severe competition with other leave trees at the time of marking.

Subtreatment level descriptions—The postthinning stand assessment was performed to document within-stand distribution of cells of known stocking levels. Having adopted Curtis' (1982) relative density as an index of stand density, and having determined relative density within each treated cell, we propose four *post facto* relative density classes to describe and encompass the range of variability found in the post-VDT stands. The *post facto* classes are presented to characterize the actual variability resulting from our thinning regime and to facilitate the use of RD as a covariate in the analysis of other experimental variables. Stated ranges are provisional and will be further refined during the course of this study. *Post facto* RD classes were delineated and designated as follows, with designations representing the approximate midpoint of the suggested range of relative densities:

1. RD 2: retained relative density of < 3.25. These were the most heavily thinned cells and correspond to RT target areas.
2. RD 4: retained relative density of 3.25 ≤ RD < 4.75. Areas of relatively heavy thinning, cells of this class correspond to target HT areas.
3. RD 6: retained relative density of 4.75 ≤ RD ≤ 6.75. Areas of relatively light thinning, cells of this class correspond to target LT cells.
4. RD 8: retained relative density > 6.75. These cells had insufficient trees removed during VDT for them to substantially depart from suppression mortality conditions. Such cells had some trees removed during the experimental thinnings, but remained in an overstocked, closed-canopy state.

Distribution of cells by *post facto* class, and average pretreatment and posttreatment basal area, tree density, and relative density are summarized by forest in table 15. Distribution of *post facto* thinning cells by stand and forest is summarized in table 16. Descriptive statistics of each *post facto* RD class for each treated stand and a cell-by-cell summary of forest conditions are provided in appendix 4. Stand maps of each treated stand are found in appendix 5.

Root grafting—Of 1,224 stumps with a d.b.h. equivalent of > 20 cm, 7 percent (n = 83) had some callus growth on the cut surface.[32] The mean d.b.h. equivalent of callused stumps was 34 cm (of those > 20 cm d.b.h.). Incidence of callusing was slightly higher in Star (8 percent, or 34 of 413) and Hill (9 percent, or 13 of 150) than in Stellar (5 percent, or 25 of 472) or Farley (6 percent or 11 of 189).

Forest health and logging damage—A total of 128 of 1,417 sampled trees (excluding the 65 dead trees) had some visual damage or reduced vigor. Logging damage was the most commonly observed type of damage, but it had low overall incidence (7 percent) among the total tree population. Typically, logging damage occurred at or near ground level, suggesting that it was the result of skidding operations. At the time of sampling, most damaged trees (98 percent) showed no outward signs of reduced vigor. Other damage or diseases (including *Phellinus weirii*) were present at very low levels (table 17).

Logging error—A total of 1,482 trees > 20 cm d.b.h. were sampled on all stands; these included 65 dead trees and 9 standing live trees that had been marked to be cut. Among sampled trees > 10 cm d.b.h. (n = 2,597), 32 standing trees were marked as cut-trees but remained uncut.

Underplanting—Although four tree species were planted (*Thuja plicata, Pinus monticola, Abies grandis*, and *Alnus rubra*), only the three conifer species were sampled because of difficulty in determining whether red alder seedlings and saplings had been planted or had resulted from natural regeneration. Because of difficulty in locating many dead (and live) underplanted seedlings, we report current observed seedling density rather than percentage of survival.

All blocks demonstrated a general trend of decreasing seedling presence with increasing tree cover. Seedling density ranged from stand averages of 189 to 263 seedlings/ha in the RD2 cells and from 120 to 225 seedlings/ha in other thinned cells. Average density of grand fir in RD2 cells in both forests was substantially greater than density of the other two conifer species. Grand fir densities also were higher in RD2 cells than in any other thinned cells. In other thinned cells, approximately even numbers of each of the three underplanted species were found in both forests. The average numbers per hectare of each underplanted conifer species in RD2 and other-thinned cells in each forest are shown in figure 8.[33]

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[32] Callus growth is used to indicate root-grafting.

[33] Underplanting initially was stratified by target subtreatments, with only RT and HT cells to be planted. However, following the thinning assessment, we determined that all four *post facto* thinning classes (RD2, RD4, RD6, RD8) contained cells that had been underplanted. Because few cells of the RD6 and RD8 *post facto* classes contained planted trees, we present RD2 seedling density in contrast to all other thinned cells that had been underplanted (RD4, RD6, and RD8).

Table 16—Distribution of *post facto* relative density (RD) classes by stand and forest, thinned stands only

	Post facto thinning class[a]			
Stand	RD2	RD4	RD6	RD8
	---------- *Number of cells (percentage of total)* ----------			
Star/Stellar:				
101[b]	13 (26)	15 (31)	13 (27)	8 (16)
103	6 (12)	17 (35)	25 (51)	1 (2)
201	8 (16)	18 (37)	12 (24)	11 (22)
202	9 (18)	18 (37)	16 (33)	6 (12)
Total, Star/Stellar	36 (18)	68 (35)	66 (34)	26 (13)
Farley/Hill:				
302	11 (22)	5 (10)	26 (53)	7 (14)
303	13 (26)	14 (29)	17 (35)	5 (10)
401	8 (16)	20 (41)	10 (20)	11 (22)
403	10 (20)	11 (22)	24 (49)	4 (8)
Total, Farley/Hill	42 (21)	50 (26)	77 (39)	27 (14)

[a] Classes are defined as follows: RD2 = retained RD of < 3.25 (metric); RD4 = retained RD of 3.25 ≤ RD < 4.75; RD6 = retained RD of 4.75 ≤ RD ≤ 6.75; RD8 = retained RD > 6.75.

[b] 49 cells per stand; 196 per forest.

Table 17—Percentage of sampled trees showing evidence of damage, by stand

	Stand								
Damage type	101	103	201	202	302	303	401	402	403
Animal damage (species unknown)	0	0.2	0	0	0	0	0	0	0
Bark beetle (species unknown)	0	0.3	0	0.1	0	0	0.4	2.7	0
Douglas-fir beetle[a]	0	0	0	0	3.5	0.8	3.0	4.1	0
Laminated root rot	0	0	0.2	0.1	0	0	0.2	0	0
Logging damage	7.3	1.3	2.3	5.9	7.5	4.5	4.0	0	4.8
Wind	0	0	0.1	0	0	0	0	0	0
Other damage (unspecified)	1.4	0.4	0	0.1	0.4	0	1.2	2.7	0.6

[a] *Dendroctonus pseudotsugae* Hopkins (name from Furniss and Carolin 1977).

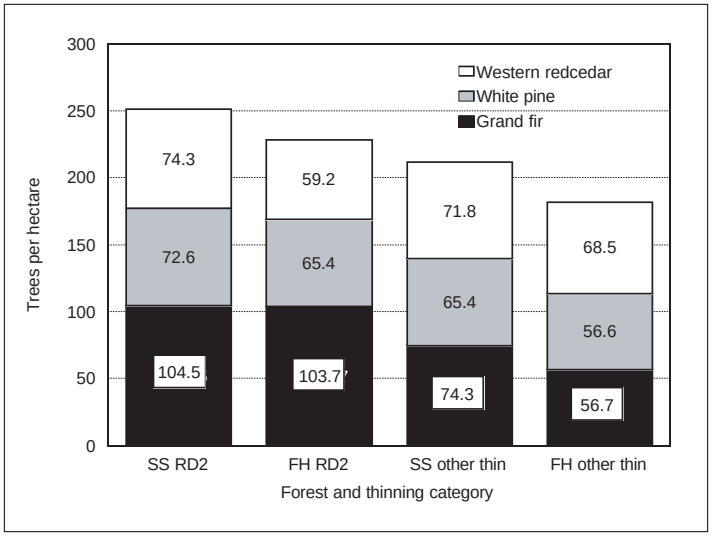


Figure 8—Density of underplanted conifer species by forest for *post facto* root rot thinned cells and all other thinned cells that had been underplanted. Figures are averages of the four treated stands per forest.

Natural tree regeneration—Seven tree species were commonly found regenerating at the time of sampling: Douglas-fir, western hemlock, western redcedar, Scouler's willow, black cottonwood, red alder, and bigleaf maple. In addition, *Quercus garryana* and *Ilex aquifolium* were found infrequently. Summaries of regeneration data indicated the sample size was too small to draw any significant inferences. Only 1.24 percent of the area of any treated stand was sampled for tree regeneration, with many plots containing no regenerating trees.

Natural regeneration in all stands was dominated by Douglas-fir, which comprised 60 to 100 percent of regenerating trees. Substantial numbers (25 to 40 percent of total seedlings) of Scouler's willow and red alder seedlings were found in FH thinned stands. About 30 percent of seedlings were western hemlock in the Star 104 control stand, and about 20 percent of seedlings in the Star 101 thinned stand were western redcedar. No further sampling or analysis was performed to examine within-stand, between-stand, or between-treatment variation across blocks or forests.

Most young, naturally regenerating trees belonged to the lowest height class (fig. 9). In SS, up to 90 percent of natural regeneration consisted of young (less than 1 year old), newly emergent Douglas-fir seedlings. Many more small seedlings were seen in RD2 cells than in other thinned cells, with a total of 6,000 to 7,000 seedlings/ha (< 50 cm tall) found in some SS RD2 cells. Very few seedlings of the larger height classes were found in thinned SS cells (fig. 9a).

The FH treated stands contained fewer total naturally regenerating seedlings and saplings than SS treated stands, but exhibited a more even distribution of size classes (fig. 9b). More small (< 50 cm) seedlings were found in FH RD2 cells than in other treated cells; the larger two regeneration height classes were found in approximately

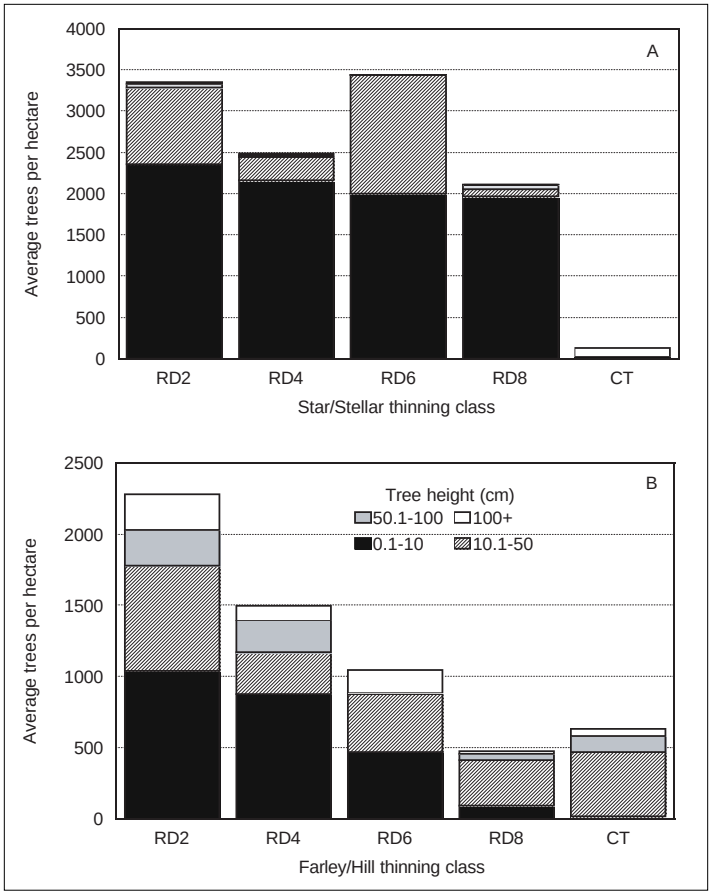


Figure 9—Densities of naturally regenerating seedlings by size class by *post facto* thinning class for Star/Stellar (A) and Farley/Hill (B) forests. Figures are averages of the four treated stands per forest or from the two sampled control stands per forest.

equal numbers in all FH thinned cells. Very few naturally regenerating seedlings were found in SS control stands; of those found in FH control stands, most were in the 10- to 50-cm height class (fig. 9).

Postthinning Understory Vegetation

Permanent vegetation plots were initially stratified by target subtreatments. After the silvicultural assessment, retained relative density values for each grid cell (and consequently each permanent plot location) were computed; vegetation summaries are presented in relation to the *post facto* RD classes based on retained RD and described above. Locations of all vegetation permanent plots are provided in appendix 3, along with retained relative density values and *post facto* RD class designations for each plot. One year after experimental thinnings, FH stands had significantly more plant species per plot than SS in both control and thinned stands. Thinning resulted in significant increases in species richness in both forests, with the greatest increases in species richness occurring in areas of lowest retained RD (table 7). Of the 146

vascular plant species found during permanent plot sampling in 1994, 138 were found in all treatments combined in FH (only 69 species in control stands), and 101 species were found in all treatments combined in SS (only 44 species in control stands). A complete list of all vascular plant species found during the study is given in appendix 1.

Cover values in FH were significantly higher than in SS for forbs-ferns and tall shrubs-understory trees in all RD classes. Though cover of low shrubs in RD2 plots in FH was less than in SS, differences were not statistically significant. The RD2 treatment cover for low shrubs was less in FH than in SS, probably because some SS RD2 areas were in actual root rot pockets (table 8).

Of 34 vascular plant species unique to FH control plots (e.g., not found in SS controls), 12 were alien (nonnative) species (none was a graminoid), 4 were native graminoids, 8 were native woody understory or midstory species, and 10 were native forbs and ferns. All 9 species unique to SS controls were native forb, fern, or shade-tolerant tree species. Of 44 species found only in FH (all treatments combined), 14 were aliens (4 were graminoids), 7 were native graminoids, 7 were native woody understory or midstory species, and 12 were native forbs and ferns. No species was unique to SS (all treatments combined).

By summer 1994, average per plot species richness in both forests had increased by 7 to 26 species in thinned areas compared to controls (table 7). The increase was most evident in the forb-fern layer, where about 40 percent of the new species in both forests (all treatments combined) were aliens. In both forests, weed species were mostly composites (family Asteraceae), grasses, and legumes. Of 65 plant species found only in thinned areas (not in control plots), 33 (51 percent) were alien species (7 were grasses), and 14 were graminoids. Of the 20 plant species found only in RD2 permanent plots, 16 (80 percent) were aliens (5 were grasses). All 20 species were found infrequently and at low cover values.

Weeds had a low percentage of cover in both forests, except for occasional thick patches of wall lettuce, bull thistle, and clovers that were mostly confined to RD2 treatments. Farley/Hill also had higher understory and midstory woody species richness and cover values than SS, where the lengthy stem-exclusion phase had precluded understory development (tables 7 and 8).

Postthinning Coarse Woody Debris

After thinning, forestwide averages of CWD cover in SS remained significantly greater than in FH (table 10), with all experimentally thinned stands having suffered reductions in CWD cover. These reductions were statistically significant in both forests (table 11). In SS, prethinning and postthinning cover of decay classes I and II were not significantly different, but decay class III cover was significantly reduced after thinning. In FH, overall CWD cover was significantly reduced after thinning; cover of decay classes II and III was also significantly reduced. Cover of decay class I increased significantly, though the difference did not appear to be biologically significant (table 11).

Den Site Availability and Snags

Stands differed in availability of dens for northern flying squirrels. Densities of residual stumps and live conifers with top rot were significantly less in the SS thinned stands than in SS unthinned stands (table 18). Other den categories did not differ significantly between experimentally thinned and unthinned stands in either forest (data not shown). Of den types not differing significantly in abundance between thinned and unthinned stands (within a forest), residual snags, residual live trees, and conifers with stick nests were significantly more abundant in SS than in FH; coniferous snags without cavities and deciduous snags without cavities were significantly more abundant in FH than in SS. Other den types did not differ significantly in abundance between forests (table 18). Thinning subtreatments within forests (FH, SS) did not differ in abundance of dens.

Supplemental Den Use

In both forests, use of supplemental nestboxes and cavities (defined by the presence of nest material) steadily increased after their installation (fig. 10). By the fifth nestbox check in March 1995, 80 percent (102 of 128) of all nestboxes exhibited use; three stands had ≥ 93 percent use of nestboxes (all in FH), one of which (303) had 100 percent use. During March 1996 (after the addition of 32 nestboxes per forest in December 1995), both nestboxes (70 percent) and cavities (35 percent) were more frequently used in FH than in SS (45 percent and 25 percent respectively). Nest materials in both forests were predominantly moss (primarily *Isothecium stoloniferum*

Table 18—Mean densities per hectare (± S.E.) of den site types by forest and in thinned and unthinned stands within forests[a]

Den site type	Star/Stellar	Farley/Hill
Residual snags	**3.5 ± 0.7**[b]	0.4 ± 0.1
Residual live trees	**2.7 ± 0.8**	0.4 ± 0.4
Conifers with sticknest	**1.7 ± 0.3**	0.6 ± 0.1
Conifer snags with cavity	0.4 ± 0.1	0.4 ± 0.1
Conifer snags without cavity	1.7 ± 0.5	**4.1 ± 1.0**
Deciduous snags with cavity	0.0 ± 0.0	0.2 ± 0.1
Deciduous snags without cavity	0.1 ± 0.1	**2.1 ± 0.8**

	Unthinned stands	Thinned stands	Unthinned stands	Thinned stands
Residual stumps	**62.6 ± 2.0**	33.9 ± 4.4	21.9 ± 3.1	20.5 ± 3.8
Live conifers with top rot	**1.3 ± 0.2**	0.5 ± 0.1	0.9 ± 0.4	0.6 ± 0.1

[a] Figures are means from inventories of all grid cells in every stand (n = 8 stands per forest for between-forest comparisons; n = 4 stands each for thinned or unthinned stands).

[b] Bold figures indicate significant difference between forests or between thinned and unthinned stands in Star/Stellar for that attribute (Mann-Whitney U-test [Norušis 1993]; $p < 0.05$).

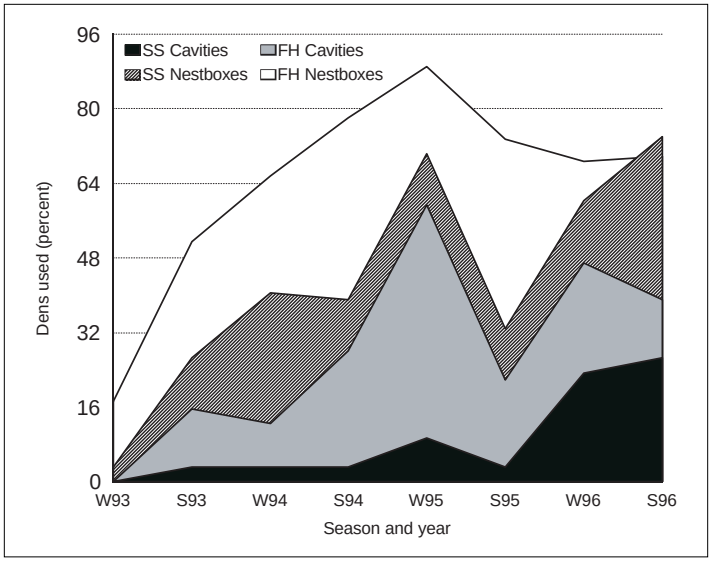
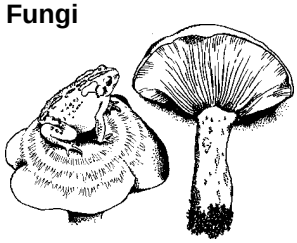


Figure 10—Seasonal changes in percentage of use of supplemental den sites, by forest, from winter (W) 1993 through summer (S) 1996. Dens were cleaned of all nest material during winter 1995. An additional eight nestboxes per den-augmented stand (32 boxes per forest) were added in December 1995. Figures represent percentage of use of 64 possible (SS and FH cavities and nestboxes, W93-S95, and SS/FH cavities, W96 and S96) or 96 possible (SS and FH nestboxes, W96 and S96).

and *Kindbergia oregana*), with some shredded cambium, fronds of brackenfern, and fruticose lichen (*Usnea* sp.). By March 1996, 214 sciurids had been found in supplemental dens--209 in nestboxes and 5 in cavities. Of these, 72 percent (154) were in FH, and 28 percent (60) were in SS. Flying squirrels (94 in FH vs. 30 in SS), Douglas' squirrels (57 in FH vs. 30 in SS), and Townsend's chipmunks (3 in FH vs. 0 in SS) were all found more frequently in FH than in SS.

Fungi



Epigeous fungi—Sixty-four mushroom species were found during sampling of SS and FH forests in October and November 1992; 37 in SS (19 mycorrhizal) and 44 in FH (15 mycorrhizal). Over 260 species[34] of fungi (124 mycorrhizal) were observed on permanent plots during fall sampling periods in 1994, 1995, and 1996 (appendix 1). Epigeous fungal communities in both forests were dominated by the ectomycorrhizal genera *Russula*, *Cantharellus*, *Cortinarius*, *Inocybe*, *Laccaria*, *Lactarius*, *Boletus*, and *Amanita*, and the saprobic genera *Clitocybe*, *Collybia*, *Hypholoma*, and *Mycena*.

[34] Some of what we refer to as species are actually groups containing one or more similar taxa. Further investigation into these groups (including such difficult genera as *Inocybe*, *Cortinarius*, and *Russula*) will undoubtedly expand the number of well-circumscribed taxa. As such, our current estimate of epigeous fungal species richness is considered to be conservative.

Table 19—Mean mycorrhizal mushroom species per permanent vegetation plot by forest and year for all plots, control plots, and all thinned plots, and control stand and total mycorrhizal species richness by forest and year

	Star/Stellar[a b]				Farley/Hill			
Year	Species richness total/ control[c]	All plots	Control plots[d]	Thinned plots	Species richness total/ control[c]	All plots	Control plots[d]	Thinned plots
	No. species	*No. species ± S.E. -------(sample size)-------*			*No. species*	*No. species ± S.E. -------(sample size)-------*		
1994	37/26	**2.7**[b] ± 0.4 (63)	4.0 ± 1.1 (15)	**2.3** ± 0.3 (48)	30/20	1.3 ± 0.2 (64)	2.1 ± 0.4** (16)	1.1 ± 0.2 (48)
1995	65/49	**4.8** ± 0.5 (63)	7.9 ± 1.4** (16)	3.8 ± 0.4 (47)	55/39	3.3 ± 0.4 (64)	4.8 ± 0.6** (16)	2.9 ± 0.4 (48)
1996	86/65	**8.9** ± 0.6 (79)	**12.7** ± 0.9* (20)	**7.6** ± 0.6 (59)	56/45	5.3 ± 0.4 (81)	8.0 ± 0.7* (22)	4.3 ± 0.4 (59)
All	108/89	**5.7** ± 0.3 (205)	**8.6** ± 0.8* (51)	**4.8** ± 0.3 (154)	78/65	3.5 ± 0.2 (210)	5.3 ± 0.5* (54)	2.8 ± 0.2 (156)

[a] Bold figures indicate significant differences between forests (t-test, $p < 0.05$).

[b] Underlined figures indicate significant differences between forests (t-test, $p < 0.10$).

[c] The number of mycorrhizal species found in all sampled plots (total) or control-only plots.

[d] Control and thinned plots for a forest and year or years differ significantly (t-test): * = $p < 0.01$; ** = $p < 0.05$).

Notwithstanding substantial year-to-year variation in ectomycorrhizal species richness, average ectomycorrhizal species richness per plot was significantly greater in SS than in FH for all years and categories except for control plots in 1994. Total ectomycorrhizal species richness was greater in SS than in FH for each year and for all years combined (table 19). Average ectomycorrhizal species richness per plot was significantly greater in control plots than in thinned plots in each forest and year except in SS in 1994.[35] Of 78 ectomycorrhizal taxa found in all years combined in FH, 16 were not found in SS. Of 108 ectomycorrhizal taxa found during all years combined in SS, 45 were not found in FH. Of those 45, 28 were in the genus *Cortinarius.* On the FES study sites, the genus *Cortinarius* was represented by 40 or more distinct taxa—about one-third of the total ectomycorrhizal species richness we have observed to date.

[35] 1994 was a low fungal production year. Though average species richness values in SS CT plots were substantially greater than for CT plots in FH and for SS thinned plots, the inherent variability and relatively small sample size precluded detection of significant differences.

Hypogeous fungi: *in situ* and in fecal pellets—Overall species richness and number of species unique to a particular forest (SS or FH) were similar in each forest. Thirty-seven species were found in FH; 16 of those were found only in FH. Thirty-five species were found in SS; 16 only there. Of the 51 identified species of hypogeous fungi, 4 to 6 species and 2 genera were undescribed previously (appendix 1). Truffles were found on 15 percent (550 of 3,680) of truffle plots (Colgan 1997). The most frequently encountered genera were *Rhizopogon*, *Melanogaster*, *Hysterangium*, *Endogone*, and *Leucogaster*.

Analysis of flying squirrel fecal pellets collected during the 1991-94 arboreal trapping sessions showed that the genera *Rhizopogon*, *Gautieria*, *Leucogaster*, and *Melanogaster* were common in squirrel diets in both forests, with *Rhizopogon* sp. being the most frequently encountered spore.[36] *Gautieria* sp. was more commonly consumed in the SS forest, and *Melanogaster* spp. were more commonly consumed in the FH forest. Fecal pellets from both forests also commonly contained spores of 10 to 12 other truffle genera, as well as amorphous plant material (not identified), and fungal spores of the mushroom genera *Russula* and *Peziza* and the families Agaricaceae and Boletaceae. All genera identified in fecal pellets also were collected during truffle sampling. Truffles were the primary food for flying squirrels throughout the year in the study area, with seasonal variation in taxa consumed.

Discussion

The FES has two aspects: retrospective and prospective or experimental. The retrospective portion determines how, and to what extent, forest vertical and horizontal structure and plant, animal, and fungal species composition differ between two forests with widely different management histories. The experimental component seeks to answer questions about how such forests will respond to thinning treatments designed to accelerate forest development. Here, we summarize our preliminary, tentative conclusions from these two views.

Retrospective Aspects

Vegetation—Both forests were established after large-scale clearcutting in the 1920s and 1930s. Their subsequent management practices have differed substantially, though, with FH having intensive management similar to many other managed forests in western Washington and Oregon, and SS having retained legacies but receiving little or no subsequent intervention. Thus the differences we observed between the two forests may illustrate long-term, but poorly understood, management-induced changes relevant to other managed west-side forests.

The Farley/Hill forest developed a substantially richer and more complex vascular plant community than SS, with more species and greater abundances (cover and volume) in all subcanopy vegetation layers (e.g., forbs and ferns, low shrubs, and midstory shrubs and low trees). Control stands in FH commonly contained weed species not found in SS (table 9), with introduced species such as *Hypochaeris*

[36] This spore type also includes the closely related genera *Alpova* and *Truncocolumella*, spores of which are indistinguishable from *Rhizopogon* (Castellano and others 1989). In western Washington, however, the common species of *Alpova* is associated only with alder, and *Truncocolumella* has been collected only in late summer and fall, and only in SS. Thus, we refer to all these spore types as *Rhizopogon* sp.

radicata, Ilex aquifolium, and *Rubus laciniata* well established in these systematically thinned stands. Other alien species, including *Rubus discolor*, *Mycelis muralis*, *Senecio jacobea*, and *S. sylvaticus* also were found in FH, but not SS,[37] control stands. Graminoids were almost completely absent from the closed-canopy SS control stands (two native grass taxa, three occurrences on 24 plots), but the more open conditions in FH control stands supported a graminoid flora of nine species (eight of which were native) occurring 28 times.

Another group, the mycotrophic vascular plants (including members of the families Orchidaceae and Ericaceae *sensu lato*), were represented by more species and were more frequently encountered in SS than in FH. Members of the genera *Chimaphila*, *Coralorrhiza*, *Goodyera*, *Monotropa*, and *Pyrola* were rarely encountered in FH control stands (3 species, 6 occurrences) compared to 6 species and 23 occurrences in SS control stands.

Salal was the dominant understory woody component in both forests; low shrub cover in control stands was about 50 percent in FH (60 percent of that salal), and 25 percent in SS (80 percent salal). Farley/Hill also had higher species richness in low-shrub and mid-shrub-understory tree layers (Carey and others 1996a). Low stocking and thinnings in FH stands also produced significantly larger diameter trees than in SS, where suppression was uniformly evident.

In contrast to the uniform canopy (and understory) that are produced by light and systematic thinnings, VDT generated a mosaic of overstory and understory conditions, thus creating an admixture of habitats. In so doing, VDT increased stand structural and biological diversity by ensuring habitat availability for most plant species and species groups.

Coarse woody debris and snags—Intensive management of FH reduced CWD to exceptionally low levels. Retention of legacies provided moderate levels of CWD in SS. This unplanned experiment and previous research are helping us understand the role of CWD.

Coarse woody debris is an important correlate of small mammal abundance in the Pacific Northwest (Carey and Johnson 1995), mycorrhizal colonization and truffle abundance (Amaranthus and others 1994), and diversity of mycorrhizal fungi in flying squirrel diets (Carey, unpublished data). Vertebrates and invertebrates that inhabit and consume coarse woody debris and snags carry decomposer bacteria and fungi as well as mycorrhizal fungi (Harmon and others 1986, Li and others 1986). Other organisms seek shelter, nests, or forage in these woody structures. Wood-boring beetles enter dead wood and while doing so, inoculate wood with a rich assemblage of fungi, bacteria, protozoans, and nematodes; different functional groups of insects also carry different assemblages of decomposer organisms, many of which are symbiotic

[37] The only weed species found in SS control stands was *Ilex aquifolium*. *Ilex* is bird dispersed, very shade tolerant, and widely established in the understory of western Washington forests (Thysell 1997).

with their specific insect host (Schowalter and others 1992). Numerous other predators, parasites, and fungal-feeding insects can then enter CWD via insect galleries, and in doing so, disperse their own complement of fungi, bacteria, protozoans, nematodes, and microarthropods (Harmon and others 1986, Schowalter and others 1992).

Small mammals and birds also are drawn to such woody structures for protection or for foraging (for insects, fungi, and seeds), and while there deposit fecal pellets containing mycorrhizal fungal spores (Harmon and others 1986; Maser and others 1985, 1986), other microorganisms (Li and others 1986), or seeds. Vascular plants, such as red huckleberry and western hemlock, mosses, liverworts and lichens (e.g., *Tetraphis pellucida* Hedw., *Lepidozia reptans* (L.) Dum., *Cladonia* spp.[38]), and mycorrhizal and decomposer fungi (e.g., *Piloderma fallax*, *Inocybe lanuginosa*, *Pleurotus ostreatus*), establish preferentially or are found nearly exclusively on woody debris (Arora 1986, Christy and Mack 1984,[39] Hitchcock and Cronquist 1973, Maser and Trappe 1984, Vitt and others 1988). Thus, CWD plays important, though incompletely understood, roles in the maintenance of biodiversity[40] in managed forests. Preliminary results from the FES are suggesting a degree of substitutability of understory vegetation for CWD in the arthropod (Burgess 1997), fungal, and forest floor mammal communities, but not for flying squirrels; for example, understory vegetation provides cover from predation, seasonal contributions of organic matter from the foliage, and cooler microclimate through shading the forest floor. Fruits and seeds also may substitute for CWD-inhabiting fungi in small mammal diets.

Both large and small snags were more abundant in SS than in FH; abundant small-d.b.h. snags in SS were a result of stem exclusion and of root rot infestations. Many small d.b.h. trees in SS were suppressed and dying; these structures, however, are of little value to cavity-nesting wildlife (Carey and others 1997). Residual large snags in SS were a legacy left from the preceding stand. Residual snags and other den sites of northern flying squirrels (especially live trees) have been suggested as a limiting factor for flying squirrels and other arboreal mammals in managed stands (Carey 1995). These snags contribute not only to abundance of dens for arboreal rodents and cavity

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[38] Names and authorities are from Vitt and others (1988) (lichen, liverwort), and Schofield (1992) (moss).

[39] They found 98 percent of western hemlock seedlings in an old-growth Douglas-fir/western hemlock forest were rooted on rotten wood, which covered only 6 percent of the forest floor.

[40] Fungi and insects are two of the most species-rich groups of organisms in the world, with new insect and fungal taxa being regularly found in the conifer-dominated forests of the Pacific Northwest. Hundreds of species of insects are associated with fallen Douglas-fir, many of which may be undescribed (Deyrup 1981, Marra and Edmonds 1995); numerous undescribed truffle taxa have been found during the course of the FES project. Many more undescribed truffles and other mycorrhizal fungi also have been collected in the Pacific Northwest (Trappe 1996). Insects frequently harbor obligate mutualist fungal symbionts, parasites, or commensals; these fungal groups are very poorly studied (Hawksworth 1991). Thus insects and fungi, many found on or in standing or down coarse woody debris in temperate forests, contribute greatly to overall biodiversity as well as to crucial forest processes (Parks and Shaw 1996).

nesting birds but also to roost sites for raptors, recruitment of large CWD, and to increased biological, structural, and functional diversity within the forest (Parks and Shaw 1996). Many residual snags in our study area, however, were well-decayed, short structures of marginal value to cavity-using wildlife (Carey and others 1991b, 1997).

Arboreal and forest floor mammals—Small mammal communities in our study were composed of the same characteristic species but had a different structure than those reported by Carey and Johnson (1995) in their analysis of managed and old-growth forests of the Olympic Peninsula. In particular, FH and SS communities were dominated by fewer species, showed a reversal of relative abundances of Columbian mice and deer mice, and SS contained fewer creeping voles when compared to managed stands on the Olympic Peninsula.

Carey and Johnson (1995) found prevalence of herbaceous cover and shrub abundance (with CWD) to be the best predictors, respectively, of creeping vole and deer mouse abundance in managed stands, with Columbian mice increasing with total understory vegetation in managed stands. Deer mice and creeping voles accounted for most of the differences in abundance between FH and SS (table 14). Creeping voles are primarily foliage eaters, and deer mice are primarily seed and fruit eaters but will consume large quantities of insects; both are also known to consume truffles and mushrooms (Carraway and Verts 1985, Maser and others 1978, Rhoades 1986, Van Horne 1982). As such, both benefit from the increased vascular plant species richness and cover observed in FH, which are attributable to FH management history. Thus, we believe the greater abundance of deer mice and Oregon voles in FH to be a further validation of Carey and Johnson's (1995) predictors. These same species may be limited by the paucity of understory plant species richness and cover that characterizes SS and other closed canopy, stem-exclusion phase stands. The rarity of the Columbian mouse in SS and FH may be related to lack of tall understory development and lack of tree species diversity. Thus, plant and small mammal biodiversity seem more likely to be preserved if understory vegetation enhancement and avoidance of stem-exclusion conditions are explicit goals in managed forests.

The arboreal rodent community, represented by the same three species in both forests, exhibits taxon-specific differences in abundance that we believe are attributable, in part, to past management practices. The northern flying squirrel is strictly nocturnal and relies almost entirely on truffles for food in our study sites. We identified 16 truffle genera from squirrel fecal pellets out of the 21 genera identified during truffle sampling (these 16 genera contain 40 of the 50+ truffle species we have identified to date), validating the important role flying squirrels have in truffle dispersal. Unlike in other portions of their range (Maser and others 1985), flying squirrels did not use lichens as a food source.

In SS with its larger component of residual trees and snags (potential den sites), CWD, and an absence of human disturbance since stand origin, we found a greater abundance of flying squirrels than in FH. Recently, Carey and others (1997) confirmed the significantly higher availability of potential natural den sites (large trees, snags) in SS than in FH and a substantially greater use of supplemental dens in FH, thus suggesting a need to proactively manage for such structures if maintenance of flying squirrel and other sciurid populations is desired.

In contrast, the Townsend's chipmunk is diurnal and is more general in its diet and use of dens (Carey 1991a, Maser and others 1981). Townsend's chipmunks eat seeds and fruits as well as fungi (truffles and mushrooms) and insects (Maser and others 1981). Food is thought to be the most limiting factor for Townsend's chipmunks, with annual differences in conifer seed and fungi production leading to large year-to-year variance in population density. Their dens are widely variable, being found both underground in burrows and in trees (Carey 1991a). The greater abundance of Townsend's chipmunks in FH is, we believe, primarily a response to the better developed and more diverse understory (Carey 1995) with its greater variety of fruits, nuts, and seeds, with den availability being less constraining for this species.

Fungi—Overall truffle species richness was similar in FH and SS control stands; further analysis of truffle data is currently underway by collaborators and will not be reported herein. Average epigeous mycorrhizal species richness per plot and total epigeous mycorrhizal species richness were higher in SS control stands than in FH in every sampled year and for all years combined. In spite of substantial year-to-year variation in overall and per-plot richness, the consistently higher species richness in SS suggests that different management practices could be partly responsible for such between-forest differences. Mycorrhizal and nonmycorrhizal fungi may be strongly associated with CWD, which is also much less abundant in FH. Many weedy plant species found in FH are also nonectomycorrhizal; thus, the combination of less substrate for some fungi (e.g., woody debris) and an abundance of nonectomycorrhizal host plant species could reduce ectomycorrhizal diversity in such stands.

Prospective and Experimental Aspects

Vegetation—There was a rapid increase in vascular plant species richness after experimental thinnings and as a function of thinning intensity within stands. The increase in species richness after thinning was evident in both forests, with most new species being found in the lowest stratum (ferns, forbs, trailing vines, herbaceous annuals and perennials). After thinning, rapidly dispersing and colonizing composites (family Asteraceae), grasses, and legumes were encountered in both forests, especially in RD2 cells. Increased species richness also resulted from growth of residual species and of other native forest floor species having the capacity to rapidly establish by runners, suckering, or by germination from a soil seed bank. Extensive or long-term weed colonization does not appear to be a problem because of the intentionally small scale of the thinnings and because of our effort to capitalize on preexisting shrub patches.

Conversely, small areas of root rot or of RT treatment in closed-canopy forests provide sites for ruderals, shade-intolerant species, and other early- to mid-successional organisms, and so increase forestwide diversity. Similar responses are seen with other localized tree mortality events and canopy gap formation processes (Spies and others 1990) that are a part of natural forest vegetative development.

Seedlings of species that contribute to development of understory and subcanopy layers (e.g., willow, alder, maple, salal, huckleberry, hazelnut, and cascara) were found in most of the permanent plots in thinned cells (93 percent of plots had one or more species; 65 percent had three or more species). Gaps such as those created by VDT may become long-lasting shrub or herb patches, or they may fill with trees (Spies and others 1990)—either naturally regenerating Douglas-fir or shade-tolerant species, or planted grand fir, western white pine, western redcedar (of which about 225 total planted conifer seedlings/ha survive in planted cells), or red alder. In any case, architectural and spatial complexity has the potential to quickly develop on thinned sites, resulting in an increase in community-level diversity in variably thinned stands.

Though shrub cover in each forest was substantially less in thinned than in control stands one year after FES thinnings (table 8), we believe this to be a short-duration response to VDT thinning disturbances. Recent work by Klinka and others (1996) has quantified the inverse relation between canopy cover and cover of salal and other shrubs in similar forests on Vancouver Island. Their work suggests that salal and other shrub cover on FES sites should increase dramatically in response to thinnings.

Salal is one of a number of woody plants that may form mycorrhizae with some of the same fungal symbionts as Douglas-fir (Amaranthus and Perry 1994, Largent and others 1980, Molina and Trappe 1982, Smith and others 1995[41]). If so, salal and other hosts could facilitate the transition from thinned to restocked stands by stimulating rapid mycorrhizal recolonization and establishment on Douglas-fir (and other Pinaceae) in thinned stands where salal is present. The presence of salal, other potential mycorrhizal host species, and CWD (Amaranthus and others 1994) in thinned sites may be important because nonmycorrhizal weeds and annuals (Trappe 1987) could gain competitive advantages if mycorrhizal fungi and other essential rhizosphere species decline in abundance (Amaranthus and Perry 1994, Perry and others 1989)—changes that could alter soil food webs and make developing a native plant community difficult (Amaranthus and Perry 1994, Ingham and Thies 1996). By capitalizing on preexisting openings with intentionally small-scale thinning units and by retaining substantial cover of overstory trees, practices such as the VDT thinnings have the potential to maintain and even accelerate natural forest processes and interactions, while minimizing the establishment of introduced and noxious weeds.

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[41] In greenhouse studies, Smith and others (1995) found limited ectomycorrhizal development on salal of two ectomycorrhizal types also commonly found on Douglas-fir, but they point out that the degree to which salal forms ectomycorrhizae in the field is not known. For some other members of Betulaceae, Ericaceae, Fagaceae, and Salicaceae, the sharing of ectomycorrhizae with Douglas-fir and other Pinaceae is clearer (Amaranthus and Perry 1994, Borchers and Perry 1990, Molina and Trappe 1982).

Prethinning and early postexperimental thinning vegetative conditions have affected abundance and size of naturally regenerating Douglas-fir seedlings and saplings in both forests. In SS control stands, the understory was depauperate in species and cover. Following VDT operations, tree seedlings (almost all Douglas-fir) quickly emerged; many seedlings (mostly < 50 cm height) were present in RD2 to RD8 cells. Emergent tree seedlings in SS experienced much less competition from herbaceous and shrub species than in FH. The RD6 and RD8 overstories in SS were still very heavily stocked after thinning and it is unlikely that shade-intolerant Douglas-fir would survive for long under such dense canopies, as evidenced by the near absence of tall seedlings.

Although FH stands contained fewer Douglas-fir seedlings and saplings than did SS, FH had a more equitable distribution of seedlings over the four size classes. This pattern is consistent with the management history of FH; previous thinning entries in FH provided opportunities not only for increased herbaceous and shrub growth, but also for Douglas-fir seedling emergence (hence the bimodal d.b.h. distribution evident in fig. 7b). Consequently, understory development, especially the abundant shrub cover, resulted in reduced survivorship of newly emergent Douglas-fir seedlings. Because of the near absence of appropriate seed sources in both forests, naturally regenerating shade-tolerant tree seedlings were infrequent.

Because SS RD2 subtreatments were applied to actual root rot areas, shrub cover there was greater than in the RD2 cells of FH as a result of previous tree mortality and increased incident light. The presence of such root rot areas (and associated understory) in stands to be managed for both commodities and biodiversity may provide an opportunity to capitalize on preexisting spatial, structural, and species diversity in stem exclusion stands.

Fungi—Preliminary data on hypogeous fungi support the importance of maintaining salal and other native species in place during thinning operations. The two forests were similar in patterns of truffle species richness and standing crop production; community structural differences have not yet been evaluated. Intensive management in FH did not change diversity and abundance of truffles compared to SS. In both forests, the effect of VDT on the productivity and biodiversity of truffles was limited temporally and spatially, with even the most aggressive treatments showing only short-term reduction in truffle standing crop (Colgan and others 1994, Colgan 1997). We believe this to be a result of the tempering effect of the deliberately small spatial scale of variable-density thinnings. Such reductions may have been longer lasting if entire blocks had received RD2 and RD4 treatments. In addition, systematic, heavy thinnings could have resulted in more widespread and persistent weed populations and in difficulty in tree regeneration, because of alterations to the soil microbial community (Ingham and Thies 1996, Perry and others 1989).

The FES sites contain substantial ectomycorrhizal species richness, both hypogeous and epigeous. Trappe (1977) reports Douglas-fir as having 2,000 or more fungal associates throughout its range, but little guidance is available for predicting total ectomycorrhizal biodiversity within a stand or more limited geographical region. Our evidence of 175 or more ectomycorrhizal symbionts in a relatively small area

(ca. 200 ha) of even-aged Douglas-fir forests lends credence to the extreme complexity within ectomycorrhizal guild structure and function (Molina and others 1992), while also demonstrating the rather remarkable diversity[42] to be found in just one organismal group within a small area of even-aged, managed forest.

Our work indicates at least a moderate-term (10-year) reduction in epigeous ectomycorrhizal species richness (as evidenced by sporocarp production) after thinning. Species richness values in FH control plots were significantly less than SS CT values and were about the same as SS thinned stand values. Species richness also was significantly less in FH thinned plots than in FH CT plots, thereby suggesting the possibility of cumulative deleterious effects of repeated disturbance on ectomycorrhizal communities.

In contrast, abundant endomycorrhizal plant host species in heavily thinned sites should increase the abundance of mycobionts such as *Glomus*, sporocarps of which are widely consumed by forest-floor small mammals (Maser and others 1978, McIntire 1984), as well as being used by arboreal rodents (Colgan 1997; Maser and others 1985, 1986). Thus, changes in plant species composition will alter mycorrhizal communities in several ways. Increased abundance of herbaceous vegetation, maples, western redcedars, and other endomycorrhizal hosts will support *Glomus* and other VA mycorrhizae. An increase in the abundance, diversity, and age-class distribution of ectomycorrhizal hosts, such as willow, poplar, hazelnut, white pine, and grand fir, can augment ectomycorrhizal diversity within a community otherwise dominated by a single host species and age class (e.g., Douglas-fir).

Our results tend to support the hypothesis that “disturbing the forest to remove commodities does not necessarily conflict with maintaining spatial and temporal linkages between plants via ectomycorrhizal fungi” (Amaranthus and Perry 1994). The intentionally small size of our thinning subtreatments (0.16 ha), coupled with the root grafting evident in these and other (Eis 1972) essentially monospecific Douglas-fir stands and the protective effect salal and other understory shrubs may have on ectomycorrhizal fungi (Amaranthus and Perry 1994, Largent and others 1980, Perry and others 1989, Smith and others 1995), should temper and ameliorate soil microbial community disturbances that accompany VDT. Thus, VDT should not produce the long-term alterations in soil structure and function known to be deleterious to tree establishment and which have occurred in some large clearcuts that were burned or had herbicides applied before replanting (Ingham and Thies 1996, Perry and others 1989). Indeed, our results suggest that VDT is a management activity that “can mimic natural disturbance regimes and help assure that ectomycorrhizal linkages among plants are not severed and that trees and soil ecosystem continue to operate as a coherent and dynamic unit” (Amaranthus and Perry 1994).

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[42] Sporocarp diversity typically underestimates total ectomycorrhizal diversity as seen on the actual root tips, and sporocarp abundance or dry weight may not accurately reflect actual hyphal biomass dominance (Molina 1997). As such, ectomycorrhizal diversity is probably higher than indicated by sporocarp diversity.

Variable-Density Thinning

The FES is the first application of VDT and the first deliberate attempt to alter the trajectory of managed second-growth stands from homogeneity, simplification, and wood production to heterogeneity, complexity, and biodiversity, as well as production of economic goods and services. Because the FES was a first attempt, little guidance was available on thinning targets or on operational application of VDT. Although simple in concept, VDT is complex in application. First, it is a departure from traditional thinning in spacing (even vs. varying between patches), residual density (VDT removes more trees than traditional thinning), and marking (root rot infestations and existing understory development are capitalized on). Second, because the objective is to grow large trees and a diverse understory concurrently, care must be exercised to avoid creating salal brushfields separated from a high canopy (an undesirable condition). Shade-tolerant regeneration must be promoted to fill canopy-to-shrub layer gaps; thus, judgment must temper density or RD targets. Underplanting may be necessary to reach forest structure targets. Third, many opportunities exist now in 40- to 70-year-old stands that have an accumulated history of management or lack thereof. These stands may have risk factors (susceptibility to wind throw, root rot infestation, insect infestations) that younger stands with consistent density and spacing management do not.

In designing prescriptions for accelerating forest development in second-growth stands, theoretical targets based on retrospective studies (Carey 1995, Carey and Johnson 1995), simulations using growth and yield models (Carey and others 1996a), and models derived from experimental silvicultural plots (Curtis 1970, 1982; Curtis and Carey 1996) must be adjusted to take into account management history and existing site conditions. Factors such as tree vigor (e.g., as determined by the ratio of live crown to total tree height), risk of high winds and degree of wind resistance, presence of disease (e.g., root rot or insect infestations), and site productivity, especially as it influences understory development and potential natural vegetation, must be considered. In FH, only light thinning was necessary to further understory development; limited heavy thinning was necessary to promote shade-tolerant regeneration and spatial heterogeneity in the stands. Because shade-tolerant natural regeneration was lacking, underplanting also was necessary. In SS, even heavy thinning was inadequate to achieve the desired theoretical spacing; however, heavier thinning in SS would have substantially increased the risk of windthrow. In both forests, multiple entries will be needed to obtain the desired future condition.

In our application of VDT, we met with Fort Lewis foresters and discussed our goals and approach in general terms by using density targets conceptually, but stressed the development of patchy understory. Because we were about to treat stands with disparate management histories, we asked these foresters, with their substantial experience (> 15 years) with traditional thinnings on Fort Lewis, to exercise their professional judgment. Their task was formidable, given the constraint of thinning from below (subordinate trees), very high stocking levels, obvious root rot infestations, and lack of understory in SS, and the moderate stocking levels, inconspicuous root rot

infestations, and well-developed, low (< 2 m tall) understory in FH. The root rot treatments, an addition to the VDT concept, would result in substantial addition of light to adjacent grid cells and create open overstory that could potentially "trap" high winds during storms.

Though specific cell-by-cell thinning targets (trees per hectare) were not always reached, the overall goal of creating a mosaic of variably stocked cells while retaining wind firmness was achieved—few trees blew down during an extreme wind storm in winter 1995. Many SS cells remained overstocked in relation to target densities, and some cells in FH were understocked relative to targets. Nonetheless, wide variation in tree density and basal area was achieved at the cell level in both forests, resulting in substantial within-stand diversity in habitat conditions (see appendices 4 and 5).

The ranges of RDs in *post facto* classes were quite large, with root rot cells having the least variable range of relative density values. Cells selected as root rot subtreatment areas in the original prescription largely coincided with the lowest tree density (RD2) cells shown on the stand maps. Remaining cells included a wide variation in retained tree densities and relative densities (RD3-RD10 in SS; RD2-RD9 in FH). After thinning, each treated stand nonetheless contained an admixture of densities sufficient to stimulate variable, cell-specific responses in understory and tree development. Variability in vegetation response is both a central goal of the FES and, we believe, a direct result of initiating the creation of a mosaic canopy in these managed stands. This variability is already manifested by increases in species richness and cover values in thinned stands; existing variability will be further capitalized on in future entries.

Random assignment of subtreatments provided the ability to examine subtreatment effects (for example, on fruiting of ectomycorrhizal fungi and on soil food webs) but was not necessary for stand-level analyses. However, irregular patterns of subtreatment assignment may have made marking more difficult and may have led to difficulty in interpretation of cell prescriptions.

Operational Aspects of Variable-Density Thinning

Several steps could be taken to refine application of VDT. Application could be systematic (fig. 3, c and d), and a repeating pattern (light-light-heavy) could be applied to consecutive 0.5-acre squares or to strips (but only if strips were perpendicular to prevailing winds). A stand density measure such as Curtis' (1982) RD is easy to use and is perhaps the best guide for applying thinnings; RD is most directly applicable when density is controlled beginning with stand initiation. Interventions to alter the developmental pathway of existing stands must consider existing conditions of understory development and forest health in addition to stocking. Application of RD is most straightforward with regular spacing between trees. Biodiversity pathways[43] for forest management, however, require developing heterogeneity in spacing between small

[43] The theoretical and empirical background of such a management approach is provided in Carey and Curtis (1996) and Carey and others (1996a). A biodiversity pathway for forest management involves specific steps intended to conserve biological legacies, minimize area and time in the competitive-exclusion stage, make use of extended rotations, minimize site preparation, and make use of alternative types of thinnings such as VDT.

patches (ca. 0.16 ha). Wide spacing in small patches (heavy thins and root rot thins) changes light conditions (admits more light) in adjacent patches, introducing a confounding effect of variable-density thinning; thus, RD calculated as a weighted average could be conservative (underestimate reduction of competition among trees) due to effects of sun-angle (a relatively small proportion of light is received perpendicular to the canopy). Thus, professional judgment about the many factors potentially influencing stand development must be used in choosing exact RD targets.

Some general rules for multiple-entry VDTs might be (1) thin 30- to 40-year-old stands (with current RD 8.5 to 10.0 [metric]) to RD 7 by using patches of RD 8.5 and RD 5.75; (2) thin previously thinned stands to RD 4.3 to 5.0 by using a combination of RD 5.75 and RD 4.3; (3) in all cases, incorporate site-specific information on risk of windthrow, disease, site quality, vegetation series, stocking, understory vegetation, and advanced regeneration of shade-tolerant species, while also taking advantage of naturally occurring openings, groups of snags, or other forest features.

Tree marking is applied to individual trees and is therefore operationally a density control. But when there was substantial cell-to-cell variation, we found markers tended to undermark or mark based on a perceived average stand condition (thus overmarking sparse cells and undermarking the dense cells). Particularly in the more dense cells, markers tended to undermark generally, probably due to a perception that too many trees were being marked. Thus, tree marking in heavily stocked stands, especially with abundant suppressed trees, may be best achieved by conspicuously marking leave trees, and by basing marking guidelines on relative density as opposed to other forest parameters. Then, RD can be checked easily by using a wedge prism. In lesser stocked stands, trees to be cut or trees to be left could be marked. In either case, consideration of the stand d.b.h. distribution is important, so that understory trees not a part of the main cohort are not removed or damaged.

Our initial application of VDT has set the trajectory of the stands toward development of late-seral forest. Star/Stellar will move from competitive exclusion to understory reinitiation and FH will move from understory reinitiation, to developed understory. The second VDT entry will be tailored specifically to conditions found in the two forests. In SS, heavy VDT will be used to further accelerate understory development. In FH, lighter VDT and coarse woody debris-augmentation will move FH toward the niche diversification stage (Carey and others 1996a). Both groups of stands will require additional underplanting of shade-tolerant conifers and, perhaps, seeding of bigleaf maple to add to midstory development.

Summary

As an integrated, multidisciplinary study of accelerating forest development of Douglas-fir forests in western Washington, the FES seeks to evaluate the extent to which, and the time frame within which, the development of late-seral attributes can be hastened through management of young, even-aged stands. By dividing the study between two forests with different management histories, the study also seeks to evaluate effects of past forest practices on forest communities, structures, and processes in forests not far removed temporally from pre-European-settlement conditions. Our studies indicate substantial differences between the two forests in terms of their overall biodiversity, CWD cover and snag density, tree growth, invasive species establishment, and

vegetative and mammalian community composition. Our studies also suggest that both forests contain much of the documented fungal, plant, and small mammal diversity found in later-seral stands (Carey 1989, 1995; Carey and Johnson 1995; Ruggiero and others 1991; Thomas and others 1993). Variable-density thinnings that aim to recreate the patchiness of late-seral forests when applied to stem-exclusion forests are an opportunity to manage for biodiversity, prevent stand degradation, and accelerate forest development (Carey and others 1996a, Carey and Johnson 1995).

Our choices of experimental treatments and response variables were developed to evaluate ecosystem processes that include photosynthetically and fungally supported trophic pathways. Mycorrhizal fungi comprise a large portion of the fungal community in forest soils (Allen and Allen 1992) and assimilate large amounts of net primary productivity in Douglas-fir forests (Fogel and Hunt 1983), while enhancing primary productivity by augmenting plant uptake of water and minerals, improving resistance to drought, and protecting against pathogens (Molina and Amaranthus 1991). Fungi (both mycorrhizal and saprobic) are food for a large diversity of consumers including arthropods, nematodes, and mammals (Perry and others 1989; Thysell and others 1997). Fungal sporocarps are the primary food for northern flying squirrels (Carey 1995; Carey and others 1992, 1996b) and are widely consumed by numerous other small mammals as well (Carey 1991a, Carey and Johnson 1995, Maser and others 1981). These small mammals, in turn, are important prey for predators, including the northern spotted owl, the northern goshawk, mustelids, and other animals at the highest trophic level.

The coniferous overstory species and other ectomycorrhizal understory species are hypothesized to “preserve ectomycorrhizal fungi during periods of rapidly changing above ground community structure and that mycorrhizal links between hardwoods and conifers facilitate conifer establishment by providing a ready source of inoculum, nutrients, and water” (Amaranthus and Perry 1994). Natural ecosystems, driven by photosynthetic and fungal processes and symbioses, maintain a “balanced, stable disequilibrium through the buffering capacity of biodiversity” (Perry and others 1989). We now recognize that a great deal of biodiversity is to be found within the plant, fungal, microbial, and animal communities and that these communities are highly interdependent. The various communities are associated with one another via numerous functional connections and pathways that include crucial forest processes such as (1) response to changing soil arthropod populations by forest insectivores; (2) diaspore dispersal by sciurids, forest floor mammals, and arthropods; (3) decay and recycling of recalcitrant organic compounds; (4) mutualisms resulting in enhanced mineral and water use, carbon storage, and ultimately, fitness; and (5) fungal- and photosynthetic-based trophic pathways supporting large numbers of herbivores, fungivores, and their predators (Carey and others 1996b). The species-, physiological-, and genetic-diversity of the plant-fungal-microbial mutualisms (along with their predators and consumers), and the redundancy in shared rhizosphere flora, should stabilize the plant-soil system (Perry and others 1990), and provide numerous benefits to plants, soil microorganisms, and soil structure (Molina and Amaranthus 1991). Retention of

such linkages and mutualisms (Amaranthus and Perry 1994, Molina and Amaranthus 1991) and protection of soil through the maintenance of vigorous tree and shrub cover (Perry and others 1990) should be an explicit goal during any planned management disturbance.

We believe these functions, relations, and links can be maintained and enhanced in managed forests, and that "many of these functions can be served simultaneously as well as sustainably" (Myers 1995) through proactive and innovative management approaches, such as those we are testing with the Forest Ecosystem Study. By taking such approaches, we believe long-term site productivity can be protected; plant, fungal, and small mammal and predator populations can be maintained; establishment of invasive or noxious weeds can be minimized; threatened species conflicts can be avoided; development of late-seral forest and landscape conditions can be hastened; and future management options can be preserved.

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Metric and English Conversions

1 inch (in) = 2.54 centimeters

1 foot (ft) = 0.3048 meter

1 square foot (ft^2) = 0.09290 square meter

1 acre = 0.40469 hectare

1 square foot per acre (ft^2/acre) = 0.22957 square meters per acre

1 mile (mi) = 1.6093 kilometer

1,000 board feet (MBF) per acre = 5.808 cubic meters per hectare

1 meter (m) = 3.28084 feet

1 square meter (m^2) = 10.764 square feet

1 hectare (ha) = 2.471 acres

1 board foot (BF) = 0.00236 cubic meter

1 kilometer (km) = 0.6214 mile

1 millimeter (mm) = 0.04 inch

1 centimeter (cm) = 0.39 inch

Celsius (°C) = (°F - 32) ÷ 1.8

References

Allen, M.F. 1991. The ecology of mycorrhizae. Cambridge, England: Cambridge University Press. 184 p.

Allen, M.F.; Allen, E.B. 1992. Mycorrhizae and plant community development: mechanisms and patterns. In: Carrol, G.C.; Wicklow, D.T., eds. The fungal community: its organization and role in the ecosystem. New York: Marcel Dekker: 455-480.

Amaranthus, M.; Trappe, J.M.; Bednar, L.; Arthur, D. 1994. Hypogeous fungal production in mature Douglas-fir forest fragments and surrounding plantations and its relation to coarse woody debris and animal mycophagy. Canadian Journal of Forest Research. 24: 2157-2165.

Amaranthus, M.P.; Perry, D.A. 1994. The functioning of ectomycorrhizal fungi in the field: linkages in space and time. Plant and Soil. 159: 133-140.

Anderson, R.V.; Elliot, E.T.; McClellan, J.F. [and others]. 1978. Trophic interactions in soils as they affect energy and nutrient dynamics. III: Biotic interactions of bacteria, amoebae and nematodes. Microbial Ecology. 4: 361-371.

Arora, D. 1986. Mushrooms demystified. Berkely, CA: Ten Speed Press. 959 p.

Babiuk, L.A.; Paul, E.A. 1970. The use of fluorescein isothiocyanate in the determination of the bacterial biomass of a grassland soil. Canadian Journal of Microbiology. 16: 57-62.

Banks, R.C.; McDiarmid, R.W.; Gardner, A.L., eds. 1987. Checklist of vertebrates of the United States, the U.S. Territories, and Canada. Resour. Publ. 166. Washington, DC: U.S. Fish and Wildlife Service. 79 p.

Bennett, W.S.; Maguire, D.A. 1995. A field procedure for thinning by relative density specifications: development, implementation, and relationship to stand structure. Western Journal of Applied Forestry. 10: 85-90

Bongers, T. 1989. De Nematoden van Nederland: The nematodes of The Netherlands. van de Haar, J., translator. London, England: British Museum (Natural History).

Borchers, S.L.; Perry, D.A. 1990. Growth and ectomycorrhizal formation of Douglas-fir seedlings grown in soils collected at different distances from pioneering hardwoods in southwest Oregon clear-cuts. Canadian Journal of Forest Research. 20: 712-721.

Burgess, C. 1997. Unpublished data. On file with: The Evergreen State College, 2700 Evergreen Parkway NW, Olympia, WA 98505.

Burns, R.M.; Honkala, B.H., tech. coords. 1990a. Silvics of North America: vol. 1, conifers. Agric. Handb. 654. Washington, DC: U.S. Department of Agriculture, Forest Service. 675 p.

Burns, R.M.; Honkala, B.H., tech. coords. 1990b. Silvics of North America: vol. 2, hardwoods. Agric. Handb. 654. Washington, DC: U.S. Department of Agriculture, Forest Service. 877 p.

Carey, A.B. 1985. A summary of the scientific basis for spotted owl management. In: Gutiérrez, R.; Carey, A.B., eds. Ecology and management of the spotted owl in the Pacific Northwest. Gen. Tech. Rep. PNW-185. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station: 100-114.

Carey, A.B. 1989. Wildlife associated with old-growth forests of the Pacific Northwest. Natural Areas Journal. 9: 151-162.

Carey, A.B. 1991. The biology of arboreal rodents in Douglas-fir forests. Gen. Tech. Rep. PNW-276. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station.

Carey, A.B. 1993. Creating cavities in trees—one facet of ecosystem management. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 1 tape cassete; 20 min.

Carey, A.B. 1994a. Biodiversity pathways: accelerating ecosystem development. In: Carey, A.B.; Elliot, C., eds. Washington forest landscape management—progress report. Proj. Rep. 1. Olympia, WA: Washington Department of Natural Resources, Washington Forest Landscape Management Project: 20-29.

Carey, A. B. 1994b. The Forest Ecosystem Study: summary, background, and rationale. Unpublished report. On file with: Pacific Northwest Research Station, Forestry Sciences Laboratory, 3625 93d Ave. SW, Olympia, WA 98512.

Carey, A.B. 1995. Sciurids in Pacific Northwest managed and old-growth forests. Ecological Applications. 5: 648-661.

Carey, A.B. 1997. Unpublished data. On file with: Pacific Northwest Research Station, Forestry Sciences Laboratory, 3625 93d Ave. SW, Olympia, WA 98512.

Carey, A.B.; Biswell, B.; Witt, J.W. 1991a. Methods for measuring populations of arboreal rodents. Gen. Tech. Rep. PNW-273. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 24 p.

Carey, A.B.; Curtis, R.O. 1996. Conservation of biodiversity: a useful paradigm for forest ecosystem management. Wildlife Society Bulletin. 24: 610-620.

Carey, A.B.; Elliot, C.; Lippke, B.R. [and others]. 1996a. A pragmatic, ecological approach to small landscape management: final report of the biodiversity pathways working group of the Washington Landscape Management Project. Proj. Rep. 2. Olympia, WA: Washington State Department of Natural Resources, Washington Forest Landscape Management Project. 153 p.

Carey, A.B.; Gill, J.D. 1983. Direct habitat improvement—some recent advances. In: Davis, J.W.; Goodwin, G.A.; Ockenfels, R.A., tech. coords. Snag habitat management—proceedings of the symposium; 1983 June 7-9; Flagstaff, AZ. Gen. Tech. Rep. RM-99. Fort Collins, CO: U.S. Department of Agriculture, Forest Service, Rocky Mountain Experiment Station: 80-87.

Carey, A.B.; Hardt, M.M.; Horton, S.P.; Biswell, B.L. 1991b. Spring bird communities in the Oregon Coast Range. In: Ruggiero, L.F.; Aubry, K B.; Carey, A.B.; Huff, M.H., tech. coords. Wildlife and vegetation of unmanaged Douglas-fir forests. Gen. Tech. Rep. PNW-GTR-285, Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station: 123-144.

Carey, A.B.; Horton, S.P.; Biswell, B.L. 1992. Northern spotted owls: influence of prey base and landscape character. Ecological Monographs. 62: 223-250.

Carey, A.B.; Johnson, M.L. 1995. Small mammals in managed, naturally young, and old-growth forests. Ecological Applications. 5: 336-352.

Carey, A.B.; Miller, R. 1991. Experimental manipulation of managed stands to provide habitat for spotted owls and to enhance plant and animal diversity. Study plan. On file with: Pacific Northwest Research Station, Forestry Sciences Laboratory, 3625 93d Ave. SW, Olympia, WA 98512.

Carey, A.B.; Peeler, K.C. 1995. Spotted owls: resource and space use in mosaic landscapes. Journal of Raptor Research. 29: 223-239.

Carey, A.B.; Reid, J.A.; Horton, S.P. 1990. Spotted owl home range and habitat use in southern Oregon Coast Ranges. Journal of Wildlife Management. 54: 11-17.

Carey, A.B.; Sanderson, H.R. 1981. Routing to accelerate tree-cavity formation. Wildlife Society Bulletin 9: 14-21.

Carey, A.B.; Thysell, D.R.; Villa, L.J. [and others]. 1996b. Foundations of biodiversity in managed Douglas-fir forests. In: Peterson, D.L.; Climas, C.V., eds. Proceedings of the 2d symposium for ecological restoration. 1995 Sept. 15; Seattle, WA. Madison, WI: Society for Ecological Restoration: 68-82.

Carey, A.B.; Wilson, T.M.; Maguire, C C.; Biswell, B.L. 1997. Dens of northern flying squirrels in the Pacific Northwest. Journal of Wildlife Management. 61: 684-699.

Carraway, L.N.; Verts, B.J. 1985. *Microtus oregoni*. Mammalian Species. 233: 1-6.

Castellano, M.A.; Maser, Z.; Trappe, J.M.; Maser, C. 1989. Key to the spores of the genera of hypogeous fungi of north temperate forests with special reference to animal mycophagy. Eureka, CA: Mad River Press. 186 p.

Castellano, M.A.; O'Dell, T. 1997. Management recommendations for survey and manage fungi: version 2.0. Unpublished report. On file with: Pacific Northwest Research Station, Forestry Sciences Laboratory, 3200 Jefferson Way, Corvallis, OR 97331.

Christy, E.J.; Mack, R.N. 1984. Variation in demography of juvenile *Tsuga heterophylla* across the substratum mosaic. Journal of Ecology. 72: 75-91.

Cline, S.P.; Berg, A.B.; Wight, H.M. 1980. Snag characteristics and dynamics in Douglas-fir forests, western Oregon. Journal of Wildlife Management. 44: 773-786.

Colgan, W., III. 1997. Diversity, productivity, and mycophagy of hypogeous mycorrhizal fungi in a variably thinned Douglas-fir forest. Corvallis, OR: Ph.D. dissertation. 95 p.

Colgan, W., III; Carey, A.B.; Trappe, J.M. 1997. A reliable method for analyzing dietaries of mycophagous small mammals. Northwestern Naturalist. 78: 65-69.

Colgan, W., III; Trappe, J.M.; Molina, R. [and others]. 1994. Production of hypogeous fungal sporocarps in a variably thinned Douglas-fir forest. In: Proceedings of the 4th European symposium on mycorrhizae. 1994 July 11-14. Grenada, Spain: [publisher unknown].

Conner, R.N.; Dickson, J.G.; Howard, J.H. 1983. Potential woodpecker nest trees through artificial inoculation of heart rots. In: Davis, J.W.; Goodwin, G.A.; Ockenfels, R.A., tech. coords. Snag management: proceedings of a symposium; 1983 June 7-9; Flagstaff, AZ. Gen. Tech. Rep. RM-99, Fort Collins, CO: U.S. Department of Agriculture, Forest Service, Rocky Mountain Experiment Station.

Curtis, R.O. 1970. Stand density measures: an interpretation. Forest Science. 16: 403-414.

Curtis, R.O. 1982. A simple index of stand density for Douglas-fir. Forest Science. 28: 92-94.

Curtis, R.O. 1997. Personal communication. Pacific Northwest Research Station, Olympia Forestry Sciences Laboratory, Olympia, WA.

Curtis, R.O.; Carey, A.B. 1996. Timber supply in the Pacific Northwest: managing for economic and ecological values. Journal of Forestry. 94: 4-7, 35-37.

Deyrup, M. 1981. Deadwood decomposers. Natural History. 90: 84-91.

Eis, S. 1972. Root grafts and their silvicultural implications. Canadian Journal of Forest Research. 2: 111-120.

Fogel, R.; Hunt, G. 1983. Contribution of mycorrhizae and soil fungi to nutrient cycling in a Douglas-fir ecosystem. Canadian Journal of Forest Research. 13: 219-232.

Fogel, R.; Ogawa, M.; Trappe, J.M. 1973. Terrestrial decomposition: a synopsis. Conif. For. Biome Int. Rep. 135. Seattle, WA: College of Forest Resources, University of Washington.

Fonda, R.W.; Bliss, L.C. 1969. Forest vegetation of the montane and subalpine zones, Olympic Mountains, Washington. Ecological Monograph. 39: 271-301.

Forest Ecosystem Management Assessment Team [FEMAT]. 1993. Forest ecosystem management: an ecological, economic and social assessment. Portland, OR: U.S. Department of Agriculture; U.S. Department of the Interior [and others]. [Irregular pagination].

Forsman, E.D.; Meslow, E.C.; Wright, H.M. 1984. Distribution and biology of the spotted owl in Oregon. Wildlife Monographs. 87: 1-64.

Forsman, E.D.; Otto, I.; Carey, A.B. 1991. Diet of spotted owls on the Olympic Peninsula, Washington, and the Roseburg District of the Bureau of Land Management. In: Ruggiero, L.F.; Aubry, K B.; Carey, A.B.; Huff, M.H., tech. coords. Wildlife and vegetation of unmanaged Douglas-fir forests. Gen. Tech. Rep. PNW-GTR-285, Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station: 527.

Franklin, J.F.; Dyrness, C.T. 1973. Natural vegetation of Oregon and Washington. Gen. Tech. Rep. PNW-8. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Forest and Range Experiment Station. 417 p.

Furniss, R.L.; Carolin, V.M. 1977. Western forest insects. Misc. Publ. 1339. Washington, DC: U.S. Department of Agriculture. 654 p.

Gauch, H.G. 1982. Multivariate analysis in community ecology. Cambridge, England: Cambridge University Press. 298 p.

Gutiérrez, R.J.; Carey, A.B. 1985. Ecology and management of the spotted owl in the Pacific Northwest. Gen. Tech. Rep. PNW-GTR-185. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station.

Harmon, M.E.; Franklin, J.F.; Swanson, F.J. [and others]. 1986. Ecology of coarse woody debris in temperate ecosystems. Advances in Ecological Research. 15: 133-302.

Hawksworth, D.L. 1991. The fungal dimension of biodiversity; magnitude, significance, and conservation. Mycological Research. 95: 641-655.

Henderson, J.A.; Peter, D.H.; Lesher, R.B.; Shaw, D.C. 1989. Forested plant associations of the Olympic National Forest. Ecol. Tech. Pap. 001-88. Portland, OR: U.S. Department of Agriculture, Forest Service, Region 6 [Pacific Northwest Region].

Hitchcock, C.L.; Cronquist, A. 1973. Flora of the Pacific Northwest: an illustrated manual. Seattle, WA: University of Washington Press. 730 p.

Ingham, E.R.; Klein, D.A. 1984. Soil fungi: relationships between hyphal activity and staining with fluorescein diacetate. Soil Biology and Biochemistry. 16: 273-278.

Ingham, E.R.; Thies, W. 1996. Responses of soil foodweb organisms one year following clearcutting and chloropicrin application to stumps to control laminated root-rot. Applied Soil Ecology. 3: 35-47.

Jones, J.K.; Hoffman, R.S.; Rice, D.W. [and others]. 1992. Revised checklist of North American mammals north of Mexico, 1991. Occasion Papers, The Museum Texas Tech University. 146: 1-23.

Kartesz, J.T. 1994. A synonymized checklist of the vascular flora of the United States, Canada, and Greenland. Volume 2: Thesaurus. 2d ed. Portland, OR: Timber Press. 816 p.

King, J.E. 1966. Site index curves for Douglas-fir in the Pacific Northwest. Weyerhaeuser For. Pap. 8. Centralia, WA: Weyerhaeuser Forestry Research Center.

Klinka, K., Chen, H.Y.H.; Wang, Q.; de Montigny, L. 1996. Forest canopies and their influence on understory vegetation in early-seral stands on west Vancouver Island. Northwest Science. 70: 193-200.

Kruckeberg, A.R. 1991. The natural history of Puget Sound country. Seattle, WA: University of Washington Press.

Largent, D.L.; Sugihara, N.; Wishner, C. 1980. Occurrence of mycorrhizae on ericaceous and pyrolaceous plants in northern California. Canadian Journal of Botany. 58: 2274-2279.

Larson, M.J.; Lombard, F.F.; Clark, J.W. 1994. *Phellinus sulphurascens* and the closely related *P. weirii* in North America. Mycologia. 86: 121-130.

Li, C.Y.; Maser, C.; Maser, Z.; Caldwell, B.A. 1986. Role of three rodents in forest nitrogen fixation in western Oregon: another aspect of mammal-mycorrhizal fungus-tree mutualism. Great Basin Naturalist. 46: 411-414.

Lujan, M., Jr.; Knowles, D.R.; Turner, J.; Plenart, M., tech. coords. 1992. Recovery plan for the northern spotted owl. Vol. 1: Final draft. [Place of publication unknown]: U.S. Department of the Interior. 322 p.

Magurran, A.E. 1988. Ecological diversity and its measurement. Princeton, NJ: Princeton University Press. 179 p.

Manuwal, D.A.; Carey, A.B. 1991. Measuring populations of small, diurnal forest birds. Gen. Tech. Rep. PNW-GTR-278. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 23 p.

Marra, J.L.; Edmonds, R.L. 1995. Pre-treatment sampling of coarse woody debris invertebrates in 50 year old Douglas-fir/western hemlock stands on the Olympic Peninsula. Seattle, WA: College of Forest Resources, University of Washington; report to the USDA Forest Service.

Maser, C.; Maser, Z.; Witt, J.W.; Hunt, G. 1986. The northern flying squirrel: a mycophagist in southwestern Oregon. Canadian Journal of Zoology. 64: 2086-2089.

Maser, C.; Mate, B.R.; Franklin, J.F.; Dyrness, C.T. 1981. Natural history of Oregon coast mammals. Gen. Tech. Rep. PNW-GTR-133. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station.

Maser, C.; Trappe, J.M., tech. eds. 1984. The seen and unseen world of the fallen tree. Gen. Tech. Rep. PNW-GTR-164. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 56 p.

Maser, C.; Trappe, J.M.; Nussbaum, R.A. 1978. Fungal-small mammal interrelationships with emphasis on Oregon coniferous forests. Ecology. 59: 799-809.

Maser, Z.; Maser, C.; Trappe, J.M. 1985. Food habits of the northern flying squirrel (*Glaucomys sabrinus*) in Oregon. Canadian Journal of Zoology. 63: 1084-1088.

McIntire, P.W. 1984. Fungus consumption by the Siskiyou chipmunk within a variously treated forest. Ecology. 65: 137-146.

McIntire, P.W.; Carey, A.B. 1989. A microhistological technique for analysis of food habits of mycophagous rodents. Res. Pap. PNW-RP-404. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 16 p.

Miller, O.K., Jr. 1983. Ectomycorrhizae in the Agaricales and Gasteromycetes. Canadian Journal of Botany. 61: 909-916.

Molina, R. 1997. Personal communication. Pacific Northwest Research Station, Forestry Sciences Laboratory, Corvallis, OR.

Molina, R.; Amaranthus, M. 1991. Rhizosphere biology: ecological linkages between soil processes, plant growth, and community dynamics. In: Harvey, A.E.; Neuenschwander, L.F., comps. Proceedings—management and productivity of western-montane forest soils; 1990 April 10-12; Boise, ID. Gen. Tech. Rep. INT-280. Ogden UT: U.S. Department of Agriculture, Forest Service, Intermountain Research Station: 51-58.

Molina, R.; Massicotte, H.B.; Trappe, J.M. 1992. Ecological role of specificity phenomena in ectomycorrhizal plant communities: potentials for interplant linkages and guild development. In: Read, D.J.; Lewis, D.H.; Fitter, A.H.; Alexander, I.J., eds. Mycorrhizas in ecosystems. Wallingford, United Kingdom: C.A.B. International: 106-112.

Molina, R.; Trappe, J.M. 1982. Patterns of ectomycorrhizal host specificity and potential among Pacific Northwest conifers and fungi. Forest Science. 28: 423-458.

Myers, N. 1995. The world's forests: need for a policy appraisal. Science. 268: 823-824.

Neitro, W.A.; Mannan, R.W.; Taylor, D. [and others]. 1985. Snags (wildlife trees). In: Brown, E.R., ed. Management of wildlife and fish habitats in forests of western Oregon and Washington. Part 1: Chapter narratives. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Region: 129-169.

Nelson, L.J.; Clark, F.W. 1973. Correction for sprung traps in catch/effort calculations of trapping results. Journal of Mammalogy. 54: 295-298.

Norušis, M.J. 1993. SPSS for Windows, base system user's guide: release 6.0. Chicago, IL: SPSS Inc. 828 p.

Oliver, C.D. 1995. Personnel communication. College of Forest Resources, University of Washington, Seattle, WA.

Oliver, C.D.; Larson, B.C. 1990. Forest stand dynamics. New York: McGraw-Hill, Inc. 467 p.

Packee, E.C. 1976. The ecology of western hemlock. In: Atkinson, W.A.; Zasoski, R.J., eds. Western hemlock management. Contrib. 34. Seattle, WA: University of Washington, Institute of Forest Products: 10-25.

Parks, C.G.; Bull, E.L.; Filip, G.M. 1995. Creating wildlife trees in managed forests using decay fungi. In: Aguirre-Bravo, C.; Eskew, L; González-Vicente, C.E.; Villa-Salas, A.B., tech. eds. Partnerships for sustainable forest ecosystem management: 5th Mexico/U.S. biennial symposium; 1994 October 17-20; Guadalajara, Mexico. Gen. Tech. Rep. RM-GTR-266. Fort Collins, CO: U.S. Department of Agriculture, Forest Service, Rocky Mountain Forest and Range Experiment Station: 175 [English]; 217 [Spanish].

Parks, C.G.; Shaw D.C. 1996. Death and decay: a vital part of living canopies. In: Shaw, D.C.; Rose, C.L.; Sillett, S.C.; Carey, A.B., eds. Special issue on Northwest forest canopies. Northwest Science. 70: 46-53.

Perry, D.A.; Amaranthus, M.P.; Borchers, J.G. [and others]. 1989. Bootstrapping in ecosystems. Bioscience. 39: 230-237.

Perry, D.A.; Borchers, J.G.; Borchers, S.L. 1990. Species migrations and ecosystem stability during climate change: the belowground connection. Conservation Biology. 4: 266-274.

Phillips, R. 1991. Mushrooms of North America. Boston, MA: Little, Brown and Company. 319 p.

Pringle, R.F. 1990. Soil survey of Thurston County, Washington. [Place of publication unknown]. U.S. Department of Agriculture, Soil Conservation Service. 283 p.

Rhoades, F. 1986. Small mammal mycophagy near woody debris accumulations in the Stehekin River Valley, Washington. Northwest Science. 60: 150-153.

Ruggiero. L.F.; Aubry, K.B.; Carey, A.B.; Huff, M.H., tech. coords. 1991. Wildlife and vegetation of unmanaged Douglas-fir forests. Gen. Tech. Rep. PNW-GTR-285. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 533 p.

Ruggiero, L.F.; Carey, A.B. 1984. A programmatic approach to the study of old-growth forest-wildlife habitat relationship. In: Proceedings of the 1983 convention of the Society of American Foresters; 1983 October 16-20; Portland, OR. Washington, DC: Society of American Foresters: 340-345.

Schofield, W.B. 1992. Some common mosses of British Columbia. Victoria, BC: Royal British Columbia Museum. 394 p.

Schowalter, T.D.; Caldwell, B.A.; Carpenter, S.E. [and others]. 1992. Decomposition of fallen trees: effects of initial conditions and heterotroph colonization rates. In: Singh, K.P.; Singh, J.S., eds. Tropical ecosystems: ecology and management. New Delhi, India: Wiley Eastern Limited: 373-383.

Singer, R. 1986. The Agaricales in modern taxonomy. 4th ed. Koenigstein [Königstein], Germany: Koelts Scientific Books. 990 p.

Smith, J.E.; Molina, R.; Perry, D.A. 1995. Occurrence of ectomycorrhizas on ericaceous and coniferous seedlings grown in soils from the Oregon Coast Range. New Phytologist. 129: 73-81.

Sollins, P. 1982. Input and decay of coarse woody debris in coniferous stands in western Oregon and Washington. Canadian Journal of Forest Research. 12: 18-28.

Spies, T.A. 1991. Plant species diversity and occurrence in young, mature, and old-growth Douglas-fir forests in western Oregon and Washington. In: Ruggiero, L.F.; Aubry, K.B.; Carey, A.B.; Huff, M.H., tech. coords. Wildlife and vegetation of unmanaged Douglas-fir forests. Gen. Tech. Rep. PNW-GTR-285. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station: 111-121.

Spies, T.A.; Franklin, J.F.; Klopsch, M. 1990. Canopy gaps in Douglas-fir forests of the Cascade mountains. Canadian Journal of Forest Research. 20: 649-658.

Thies, W.G.; Sturrock, R.N. 1995. Laminated root-rot in western North America. Gen. Tech. Rep. PNW-GTR-349. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 32 p.

Thomas, J.W.; Forsman, E.D.; Lint, J.B. [and others]. 1990. A conservation strategy for the northern spotted owl: report of the interagency scientific committee to address the conservation of the northern spotted owl. Portland, OR: U.S. Department of Agriculture; U.S. Department of the Interior [and others]. 427 p.

Thomas, J.W.; Raphael, M.G.; Anthony, R.G. [and others]. 1993. Viability assessments and management considerations for species associated with late-successional and old-growth forests of the Pacific Northwest. Washington, DC: U.S. Government Printing Office. 530 p.

Thomas, T.B.; Thysell, D.R.; Carey, A.B. 1994. Floristic inventory of Fort Lewis, Washington: final report. Olympia, WA: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station; unpublished report to the Environmental and Natural Resources Division, Fort Lewis Military Reservation, U.S. Army; [contract no. unknown].

Thysell, D.R. 1997. Personnal communication. Pacific Northwest Research Station, Forestry Sciences Laboratory, Olympia, WA.

Thysell, D.R.; Carey, A.B.; Ball, S. 1997. Noteworthy collection: *Pityopus californicus* (Eastw.) H.F. Copel. (Ericaceae). Madroño. 44: 398.

Thysell, D.R.; Villa, L.J.; Carey, A.B. 1997. Observations of northern flying squirrel feeding behavior: use of non-truffle food items. Northwestern Naturalist. 78: 87-92.

Trappe, J.M. 1962. Fungus associates of ectomycorrhizae. Botanical Review. 28: 538-606.

Trappe, J.M. 1977. Selection of fungi for ectomorrhizal inoculation in nurseries. Annual Review of Phytopathology. 22: 203-222.

Trappe, J.M. 1987. Phylogenetic and ecological aspects of mycotrophy in the Magnoliophyta (angiosperms) from an evolutionary standpoint. In: Safir, G., ed. Ecophysiology of vesicular-arbuscular mycorrhizal plants. Boca Raton, FL: CRC Press: 5-25.

Trappe, J.M. 1996. Personal communication. Oregon State University, Corvallis, OR.

Tylutki, E.E. 1979. Mushrooms of Idaho and the Pacific Northwest. Volume 1: Discomycetes. Moscow, ID: University of Idaho Press. 133 p.

Tylutki, E.E. 1987. Mushrooms of Idaho and the Pacific Northwest. Volume 2: Non-gilled hymenomycetes. Moscow, ID: University of Idaho Press. 232 p.

U.S. Department of Agriculture, Forest Service. 1974. Seeds of woody plants in the United States. Agric. Handb. 450. Washington, DC.

U.S. Department of Agriculture, Forest Service. 1988. Final supplement to the environmental impact statement for an amendment to the Pacific Northwest Regional guide: spotted owl guidelines. Portland, OR: Pacific Northwest Region.

U.S. Department of Agriculture, Forest Service. 1989. Stand exam program: field procedures guide. Portland, OR: Pacific Northwest Region.

U.S. Department of Agriculture, Forest Service; U.S. Department of the Interior, Bureau of Land Management. 1994. Record of decision for amendments to Forest Service and Bureau of Land Management planning documents within the range of the northern spotted owl. [Place of publication unknown]. 74 p. [plus attachment A: standards and guidelines].

U.S. Department of Agriculture, Natural Resoures Conservation Service. 1997. The PLANTS database [online] . Available HTTP: http://plants.usda.gov. Baton Rouge, LA: National Plant Date Center.

Van Horne, B. 1982. Niches of adult and juvenile deer mice (*Peromyscus maniculatus*) in seral stages of coniferous forest. Ecology. 63: 992-1003.

Van Wagner, C.E. 1968. The line intersect method in forest fuel sampling. Forest Science. 14: 20-26.

Van Wagner, C.E.; Wilson, A.L. 1976. Diameter measurement in the line intersect method. Forest Science. 22: 230-232.

Villa, L.J.; Carey, A.B.; Wilson, T.M.; Glos, K.E. 1999. Maturation and reproduction of northern flying squirrels in Pacific Northwest forests. Gen. Tech. Rep. PNW-GTR-444. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station.

Vitt, D.H.; Marsh, J.E.; Bovey, R.B. 1988. Mosses, lichens, and ferns of northwest North America. Seattle, WA: University of Washington Press. 296 p.

Wilson, T.M.; Carey, A.B. 1996. Observations of weasels in second-growth Douglas-fir forests in the Puget Trough, Washington. Northwestern Naturalist. 77: 35-39.

Appendix 1
Forest Ecosystem Study Species List

Scientific name[1]	Code[2]	Common name[3]
Vascular Plants:		
Abies grandis (Dougl. Ex D. Don) Lindl.	ABGR	Grand fir
Acer circinatum Pursh	ACCI	Vine maple
Acer macrophyllum Pursh	ACMA3	Big-leafed maple
Achlys triphylla (Sm.) DC.[4]	ACTR	Vanillaleaf
Actaea rubra (Ait.) Willd.	ACRU2	Baneberry
Adenocaulon bicolor Hook.[4]	ADBI	Trail-plant
Adiantum aleuticum (Rupr.) Paris (=*A. pedatum* L.)[4]	ADAL	Maidenhair fern
Agrostis capillaris L. (= *A. tenuis* Sibth.)	AGCA5	Colonial bentgrass
Agrostis exarata Trin.	AGEX	Spike bentgrass
Aira caryophyllea L.	AICA	Silver hairgrass
Aira praecox L.	AIPR	Little hairgrass
Alnus rubra Bong.	ALRU2	Red alder
Amelanchier alnifolia (Nutt.) Nutt. ex M. Roemer	AMAL2	Western serviceberry
Anaphalis margaritacea (L.) Benth. & Hook. f.	ANMA	Pearly-everlasting
Anemone deltoidea Hook.[4]	ANDE3	Western anemone
Anthoxanthum odoratum L.	ANOD	Sweet vernalgrass
Arbutus menziesii Pursh	ARME	Madrone
Arctostaphyllos uva-ursi (L.) Spreng.	ARUV	Kinninnick
Arrhenatherum elatius (L.) J. & K. Presl	AREL3	Tall oatgrass
Artemisia suksdorfii Piper	ARSU4	Suksdorf's sagebrush
Asarum caudatum Lindl.[4]	ASCA2	Wild ginger
Athyrium filix-femina (L.) Roth	ATFI	Lady-fern
Blechnum spicant (L.) Roth	BLSP	Deer-fern
Bromus vulgaris (Hook.) Shear	BRVU	Columbia brome
Campanula scouleri Hook. ex A. DC.	CASC7	Scouler's harebell
Cardamine pensylvanica Muhl. ex Willd.	CAPE3	Pacific bittercress
Carex deweyana Schwein.	CADE9	Dewey's sedge

Footnotes are on p. 102.

Scientific name[1]	Code[2]	Common name[3]
Carex pachystachya Cham. ex Steud.	CAPA14	Thick-headed sedge
Cerastium fontanum Baumg. ssp. *vulgare* (Hartman) Greuter & Burdet (= *C. vulgatum* L.)	CEFOV2	Common chickweed
Chimaphila menziesii (R. Br. ex D. Don) Spreng.[4]	CHME	Pipsissewa
Chimaphila umbellata (L.) W. Bart.[4]	CHUM	Common pipsissewa
Circaea alpina L.	CIAL	Enchanter's nightshade
Cirsium arvense (L.) Scop.	CIAR4	Canada thistle
Cirsium brevistylum Cronq.	CIBR2	Short-styled thistle
Cirsium vulgare (Savi) Ten.	CIVU	Bull thistle
Claytonia perfoliata Donn ex Willd. ssp. *perfoliata* (= *Montia perfoliata* (Donn.) Howell)	CLPEP	Miner's lettuce
Claytonia sibirica L. var. *sibirica* (= *Montia siberica* (L.) Howell)	CLSIS	Candyflower
Collomia heterophylla Dougl. ex Hook.	COHE2	Varied-leafed collomia
Conyza canadensis (L.) Cronq.	COCA5	Canada fleabane
Corallorhiza maculata (Raf.) Raf.[4]	COMA4	Spotted coral-root
Cornus nuttallii Audubon ex Torr. & Gray	CONU4	Pacific dogwood
Corylus cornuta Marsh. var. *californica* (A. DC.) Sharp	COCOC	California hazel
Crepis capillaris (L.) Wallr.	CRCA3	Smooth hawksbeard
Cytisus scoparius (L.) Link	CYSC4	Scot's broom
Deschampsia elongata (Hook.) Munro	DEEL	Slender hairgrass
Dicentra formosa (Andr.) Walp.	DIFO	Pacific bleedingheart
Digitalis purpurea L.	DIPU	Foxglove
Disporum hookeri (Torr.) Nichols.[4]	DIHO3	Hooker's fairy-bell
Dryopteris campyloptera Clarkson[4] (= *D. austriaca* (Jacq.) Woynar)	DRCA3	Spreading woodfern
Elymus glaucus Buckl. ssp. *glaucus*	ELGLG	Blue wild-rye
Epilobium angustifolium L.	EPAN2	Fireweed

Scientific name[1]	Code[2]	Common name[3]
Epilobium glaberrimum Barbey ssp. *glaberrimum*	EPGLG	Smooth willow-herb
Epilobium minutum Lindl. ex Lehm.	EPMI	Small-flowered willow-herb
Erechtites minima (Poir.) DC.	ERMI6	Toothed coast burnweed
Festuca occidentalis Hook.	FEOC	Western fescue
Festuca subulata Trin.	FESU	Bearded fescue
Festuca subuliflora Scribn.	FESU2	Coast Range fescue
Fragaria crinita Rydb. (= *F. vesca* L.var. *crinita* (Rydb.) C.L. Hitchc.)	FRCR	Pacific strawberry
Fragaria virginiana Duchesne ssp. *platypetala* (Rydb.) Staudt	FRVIP2	Broadpetal strawberry
Frangula purshiana (DC.) Cooper (= *Rhamnus purshiana* DC.)	FRPU7	Pursh's buckthorn
Galium aparine L.	GAAP2	Cleavers
Galium triflorum Michx.	GATR3	Sweetscented bedstraw
Gamochaeta purpurea (L.) Cabrera (= *Gnaphalium purpureum* L.)	GAPU3	Spoonleaf purple everlasting
Gaultheria shallon Pursh	GASH	Salal
Geranium robertianum L.	GERO	Herb Robert
Geum macrophyllum Willd.var. *macrophyllum*	GEMAM	Oregon avens
Gnaphalium microcephalum Nutt.	GNMI	Slender cudweed
Goodyera oblongifolia Raf.[4]	GOOB2	Rattlesnake-plantain
Hieracium albiflorum Hook.	HIAL2	White-flowered hawkweed
Holcus lanatus L.	HOLA	Common velvet-grass
Holodiscus discolor (Pursh) Maxim.	HODI	Oceanspray
Hypericum perforatum L.	HYPE	Klamath weed
Hypochaeris radicata L.	HYRA3	Spotted cats-ear
Ilex aquifolium L.	ILAQ80	English holly
Juncus bufonius L.	JUBU	Toad rush
Juncus effusus L. var. *gracilis* Hook.	JUEFG	Common rush
Juncus tenuis Willd.	JUTE	Slender rush

Scientific name[1]	Code[2]	Common name[3]
Lactuca biennis (Moench) Fern.	LABI	Tall blue lettuce
Lapsana communis L.	LACO3	Nipplewort
Lathyrus polyphyllus Nutt.[4]	LAPO3	Leafy peavine
Leucanthemum vulgare Lam. (= *Chrysanthemum leucanthemum* L.)	LEVU	Oxeye-daisy
Lilium columbianum hort. ex Baker	LICO	Tiger lilly
Linnaea borealis L.	LIBO3	Twinflower
Listera caurina Piper[4]	LICA10	Western twayblade
Lonicera ciliosa (Pursh) Poir. ex DC.	LOCI3	Trumpet honeysuckle
Lonicera hispidula (Lindl.) Dougl. ex Torr. & Gray	LOHI2	Hairy honeysuckle
Lotus corniculatus L.	LOCO6	Birdsfoot-trefoil
Lotus micranthus Benth.	LOMI	Small-flowered deervetch
Lupinus rivularis Dougl. ex Lindl.	LURI	Stream lupine
Luzula campestris (L.) DC.	LUCA2	Woodrush
Luzula parviflora (Ehrh.) Desv.	LUPA4	Small-flowered woodrush
Mahonia aquifolium (Pursh) Nutt. (= *Berberis aquifolium* Pursh)	MAAQ2	Tall Oregongrape
Mahonia nervosa (Pursh) Nutt. (= *Berberis nervosa* Pursh)	MANE2	Cascade Oregongrape
Maianthemum stellatum (L.) Link[4] (= *Smilacina stellata* (L.) Desf.)	MAST4	Starry false Solomon's seal
Melica subulata (Griseb.) Scribn.[4]	MESU	Alaska oniongrass
Mentha canadensis L.	MECA7	Field mint
Moehringia macrophylla (Hook.) Fenzl (= *Arenaria macrophylla* Hook.)	MOMA3	Big-leafed sandwort
Monotropa uniflora L.[4]	MOUN3	Indian-pipe
Mycelis muralis (L.) Dumort. (= *Lactuca muralis* (L.) Fresen.)	MYMU	Wall lettuce
Nemophila parviflora Dougl. ex Benth.	NEPA	Small-flowered nemophila
Oemlaria cerasiformis (Torr. & Gray ex Hook. & Arn.) Landon	OECE	Indian plum

Scientific name[1]	Code[2]	Common name[3]
Osmorhiza berteroi DC. (= *O. chilensis* H. & A.)	OSBE	Sweet-cicely
Petasites frigidus (L.) Fries var. *palmatus* (Ait.) Cronq.	PEFRP	Coltsfoot
Phalaris arundinacea L.	PHAR3	Reed canarygrass
Philadelphus lewisii Pursh	PHPR3	Mockorange
Phleum pratense L.	PHLE3	Timothy
Pinus monticola Dougl. ex D. Don	PIMO3	White pine
Pityopus californicus (Eastw.) H.F. Copel.[4 5]	PICA9	California pine-foot
Plantago lanceolata L.	PLLA	English plantain
Plantago major L.	PLMA2	Common plantain
Polypodium glycyrrhiza D.C. Eat.	POGL8	Licorice fern
Polystichum munitum (Kaulfuss) K. Presl	POMU	Swordfern
Populus balsamifera L. ssp. *trichocarpa* (Torr. & Gray ex Hook.) Brayshaw (= *P. trichocarpa* T. & G.)	POBAT	Black cottonwood
Prunella vulgaris L.	PRVU	Self-heal
Pseudotsuga menziesii (Mirbel) Franco var. *menziesii*	PSMEM	Douglas-fir
Pteridium aquilinum (L.) Kuhn.	PTAQ	Brackenfern
Pyrola asarifolia Michx.[4]	PYAS	Common pink wintergreen
Pyrola picta Sm.[4] (includes *P. aphylla*)	PYPI2	White vein pyrola
Quercus garryana Dougl. ex Hook.	QUGA4	Oregon white oak
Ranunculus occidentalis Nutt.	RAOC	Western buttercup
Ranunculus repens L. var. *repens*	RARER	Creeping buttercup
Ranunculus uncinatus D. Don ex G. Don var. *parviflorus* (Torr.) L. Benson	RAUNP	Little buttercup
Ribes sanguineum Pursh	RISA	Red currant
Rosa gymnocarpa Nutt.	ROGY	Baldhip rose
Rubus discolor Weihe & Nees	RUDI2	Himalayan blackberry
Rubus laciniatus Willd.	RULA	Evergreen blackberry

Scientific name[1]	Code[2]	Common name[3]
Rubus leucodermis Dougl. ex Torr. & Gray	RULE	Blackcap
Rubus parviflorus Nutt.	RUPA	Thimbleberry
Rubus spectabilis Pursh	RUSP	Salmonberry
Rubus ursinus Cham. & Schlecht.	RUUR	California blackberry
Rumex acetosella L.	RUAC3	Sheep sorel
Rumex crispus L.	RUCR	Curly dock
Salix scouleriana Barratt ex Hook.	SASC	Scouler willow
Sambucus racemosa L.	SARA2	Red elderberry
Satureja douglasii (Benth.) Briq.[4]	SADO5	Yerba buena
Senecio jacobaea L.	SEJA	Tansy ragwort
Senecio sylvaticus L.	SESY	Wood groundsel
Solanum dulcamera L.	SODU	Bittersweet
Solidago canadensis L. var. *salebrosa* (Piper) M. E. Jones	SOCAS	Canadian goldenrod
Sonchus asper (L.) Hill	SOAS	Prickly sow-thistle
Sonchus oleraceus L.	SOOL	Common sow-thistle
Spiraea douglasii Hook. var. *douglasii*	SPDOD	Douglas' spiraea
Stellaria crispa Cham. & Schlecht.	STCR2	Crisped starwort
Stellaria longipes Goldie	STLO2	Longstalk starwort
Symphoricarpos albus (L.) Blake	SYAL	Common snowberry
Symphoricarpos hesperius G. N. Jones (= *S. mollis* Nutt. ssp. *hesperius* (G.N. Jones) Abrams ex Ferris)	SYHE	Creeping snowberry
Taxus brevifolia Nutt.[4]	TABR2	Pacific yew
Thuja plicata Donn ex D. Don[4]	THPL	Western redcedar
Tiarella trifoliata L.[4]	TITR	Foamflower
Trientalis borealis Raf. ssp. *latifolia* (Hook.) Hulten (= *T. latifolia* Hook.)	TRBOL	Western starflower
Trifolium campestre Schreb. (= *T. procumbens* L.)	TRCA5	Hop clover
Trifolium pratense L.	TRPR2	Red clover

Scientific name[1]	Code[2]	Common name[3]
Trifolium repens L.	TRRE3	White clover
Trillium ovatum Pursh[4]	TROV2	Western wake-robin
Trisetum cernuum Trin.	TRCE2	Nodding trisetum
Tsuga heterophylla (Raf.) Sarg.	TSHE	Western hemlock
Urtica dioica L.	URDI	Stinging nettle
Vaccinium ovatum Pursh	VAOV2	Evergreen huckleberry
Vaccinium parvifolium Sm.[4]	VAPA	Red huckleberry
Vancouveria hexandra (Hook.) Morr. & Dcne.[4]	VAHE	Inside-out flower
Vicia americana Muhl. ex Willd.	VIAM	American vetch
Vicia nigricans Hook. & Arn. ssp. *gigantea* (Hook.) Lassetter & Gunn. (= *V. gigantea* Hook.)	VINIG	Giant vetch
Viola glabella Nutt.[4]	VIGL	Stream violet
Viola howellii Gray	VIHO	Howell's violet
Viola sempervirens Greene	VISE3	Evergreen violet

Amphibians and reptiles:

Mole salamanders—

Ambystoma gracile (Baird, 1859)	Northwestern salamander
Ambystoma macrodactylum Baird, 1849	Long-toed salamander

Lungless salamanders—

Ensatina escholtzii Gray, 1850	Ensatina
Plethodon vehiculum (Cooper, 1860)	Western red-backed salamander

Frogs and toads—

Bufo boreas Baird & Girard, 1852	Western toad
Pseudacris regilla Baird & Girard, 1852	Pacific tree frog
Rana aurora Baird & Girard, 1852	Red-legged frog
Rana catesbeiana Shaw, 1802	Bullfrog

Newt—

Taricha granulosa (Skilton, 1849)	Rough-skinned newt

Scientific name[1]	Common name[3]
Lizard—	
Gerrhonotus coeruleus Wiegmann, 1828	Northern alligator lizard
Snakes—	
Charina bottae (Blainville, 1835)	Rubber boa
Thamnophis ordinoides (Baird & Girard, 1852)	Northwestern garter snake
Mammals:	
Marsupial—	
Didelphis virginiana Kerr, 1792	Virginia opossum
Insectivores—	
Neurotrichus gibbsii (Baird, 1858)	Shrew mole
Scapanus orarius True, 1896	Coast mole
Scapanus townsendii (Bachman, 1839)	Townsend's mole
Sorex bendirii (Merriam, 1884)	Pacific marsh shrew
Sorex monticolus Merriam, 1890	Montane shrew
Sorex trowbridgii Baird, 1858	Trowbridge's shrew
Sorex vagrans Baird, 1858	Vagrant shrew
Bats—	
Myotis californicus (Audubon & Bachman, 1842)	California myotis
Plecotus townsendii Cooper, 1837	Townsend's big-eared bat
Rabbits—	
Lepus americanus Erxleben, 1777	Snowshoe hare
Sylvilagus floridanus (J.A. Allen, 1890)	Eastern cottontail
Rodents—	
Aplodonia rufa (Rafinesque, 1817)	Mountain beaver
Erethizon dorsatum (Linnaeus, 1758)	Porcupine
Squirrels	
Glaucomys sabrinus (Shaw, 1801)	Northern flying squirrel
Sciurus carolinensis Gmelin, 1788	Eastern gray squirrel

Scientific name[1]	Common name[3]
Tamias townsendii Bachman, 1839	Townsend's chipmunk
Tamiasciurus douglasii (Bachman, 1839)	Douglas' squirrel
Mice and voles	
Clethrionomys gapperi (Vigors, 1830)	Southern red-backed vole
Microtus longicaudus (Merriam, 1888)	Long-tailed vole
Microtus oregoni (Bachman, 1839)	Oregon creeping vole
Mus musculus Linnaeus, 1758	House mouse
Peromyscus maniculatus (Wagner, 1845)	Deer mouse
Peromyscus oreas Bangs, 1898	Columbian deer mouse
Zapus trinotatus Rhoads, 1895	Pacific jumping mouse
Carnivores—	
Canis latrans Say, 1823	Coyote
Mustela erminea Linnaeus, 1758	Short-tailed weasel
Mustela frenata Lichtenstein, 1831	Long-tailed weasel
Procyon lotor (Linnaeus, 1758)	Racoon
Ursus americanus Pallas, 1780	Black bear
Vulpes vulpes (Linnaeus, 1758)	Red fox
Ungulate—	
Odocoileus hemionus (Rafinesque, 1817)	Black-tailed deer
Birds:	
Heron—	
Ardea herodias Linnaeus, 1758	Great blue heron
Ducks and geese—	
Anas platyrhynchos Linnaeus, 1758	Mallard
Branta canadensis (Linnaeus, 1758)	Canada goose
Hawks and vultures—	
Accipiter cooperii (Bonaparte, 1828)	Cooper's hawk
Accipiter gentilis (Linnaeus, 1758)	Northern goshawk
Accipiter striatus (Vieillot, 1807)	Sharp-shinned hawk

Scientific name[1]	Common name[3]
Buteo jamaicensis (Gmelin, 1788)	Red-tailed hawk
Cathartes aura (Linnaeus, 1758)	Turkey vulture
Haliaeetus leucocephalus (Linnaeus, 1766)	Bald eagle
Grouse and quail—	
Bonasa umbellus (Linnaeus, 1766)	Ruffed grouse
Dendragapus obscurus (Say, 1823)	Blue grouse
Pigeons and doves—	
Columba fasciata Say, 1823	Band-tailed pigeon
Zenaida macroura (Linnaeus, 1758)	Mourning dove
Owls—	
Aegolius acadicus (Gmelin, 1788)	Northern saw-whet owl
Bubo virginianus (Gmelin, 1788)	Great-horned owl
Glaucidium gnoma Wagler, 1832	Northern pygmy owl
Otus kennicotti (Elliot, 1867)	Western screech owl
Strix varia Barton, 1799	Barred owl
Tyto alba (Scopoli, 1769)	Barn owl
Goatsucker—	
Chordeiles minor (Forster, 1771)	Common nighthawk
Hummingbird—	
Selasphorus rufus (Gmelin, 1788)	Rufous hummingbird
Woodpeckers and allies—	
Colaptes auratus (Linnaeus, 1758)	Northern flicker
Dryocopus pileatus (Linnaeus, 1758)	Pileated woodpecker
Picoides pubescens (Linnaeus, 1766)	Downy woodpecker
Picoides villosus (Linnaeus, 1766)	Hairy woodpecker
Sphyrapicus ruber (Gmelin, 1788)	Red-breasted sapsucker
Flycatchers—	
Contopus borealis (Swainson, 1832)	Olive-sided flycatcher
Empidonax difficilis (Baird, 1858)	Pacific slope flycatcher
Empidonax traillii (Audubon, 1828)	Willow flycatcher

Scientific name[1]	Common name[3]
Swallows—	
Hirundo rustica Linnaeus, 1758	Barn swallow
Tachycineta bicolor (Vieillot, 1808)	Tree swallow
Jays, crows, and ravens—	
Corvus brachyrhynchos Brehm, 1822	American crow
Corvus corax Linnaeus, 1758	Common raven
Cyanocitta stelleri (Gmelin, 1788)	Steller's jay
Perisoreus canadensis Linnaeus, 1766	Gray jay
Chickadees, nuthatches, and creepers—	
Certhia americana Bonaparte, 1838	Brown creeper
Parus atricapillus Linnaeus, 1766	Black-capped chickadee
Parus rufescens Townsend, 1837	Chestnut-backed chickadee
Psaltriparus minimus (Townsend, 1837)	Common bushtit
Sitta canadensis Linnaeus, 1766	Red-breasted nuthatch
Wren—	
Troglodytes troglodytes (Linnaeus, 1758)	Winter wren
Kinglets—	
Regulus calendula (Linnaeus, 1766)	Ruby-crowned kinglet
Regulus saltrapa Lichtenstein, 1823	Golden-crowned kinglet
Thrushes—	
Catharus guttatus (Pallas, 1811)	Hermit thrush
Catharus ustulatus (Nuttall, 1840)	Swainson's thrush
Ixoreus naevius (Gmelin, 1789)	Varied thrush
Myadestes townsendi (Audubon, 1838)	Townsend's solitaire
Sialia mexicana Swainson, 1832	Western bluebird
Turdus migratorius Linnaeus, 1766	American robin
Waxwing—	
Bombycilla cedrorum Vieillot, 1808	Cedar waxwing

Scientific name[1]	Common name[3]
Vireos—	
Vireo gilvus (Vieillot, 1808)	Warbling vireo
Vireo huttoni (Cassin, 1851)	Hutton's vireo
Vireo solitarius (Wilson, 1810)	Solitary vireo
Warblers—	
Dendroica coronata (Linnaeus, 1766)	Yellow-rumped warbler
Dendroica nigrescens (Townsend, 1837)	Black-throated gray warbler
Dendroica occidentalis (Townsend, 1837)	Hermit warbler
Dendroica townsendi (Townsend, 1837)	Townsend's warbler
Oporornis tolmiei (Townsend, 1837)	MacGillivray's warbler
Vermivora celata (Say, 1823)	Orange-crowned warbler
Wilsonia pusilla (Wilson, 1811)	Wilson's warbler
Tanager and grosbeak—	
Pheucticus melanocephalus (Swainson, 1827)	Black-headed grosbeak
Piranga ludoviciana (Wilson, 1811)	Western tanager
Sparrows—	
Junco hyemalis (Linnaeus, 1758)	Dark-eyed junco
Melospiza melodia (Wilson, 1810)	Song sparrow
Pipilo erythrophthalmus (Linnaeus, 1758)	Rufous-sided towhee
Spizella passerina (Bechstein, 1798)	Chipping sparrow
Zonotrichia leucophrys (Forster, 1772)	White-crowned sparrow
Blackbirds—	
Agelaius phoeniceus (Linnaeus, 1766)	Red-winged blackbird
Molothrus ater (Boddaert, 1783)	Brown-headed cowbird
Finches—	
Carduelis pinus (Wilson, 1810)	Pine siskin
Carpodacus purpureus (Gmelin, 1789)	Purple finch
Loxia curvirostra Linnaeus, 1758	Red crossbill

Scientific name[1]

Hypogeous fungi (truffles):

Alpova diplophloeus (Zeller & Dodge) Trappe & Smith

Elaphomyces granulatus Fr.[4]

Elaphomyces muricatus Fr.[4]

Endogone lactiflua Bk. & Bk.

Endogone pisiformis Link:Fr.

Gautieria monticola Harkn.

Gautieria sp. nov.[6]

Genabea cerebriformis (Harkn.) Trappe

Genea harknessii Gilkey

Genea intermedia Gilkey

Glomus sp. nov.[6]

Glomus macrocarpum Tul. & Tul.

Glomus microcarpum Tul. & Tul.

Glomus mosseae (Nicol. & Gerd.) Gerd. & Trappe

Hydnotrya variiformis Gilkey

Hymenogaster sublilacinus Smith

Hymenogaster sp. (to be identified)

Hysterangium coriaceum Hesse

Hysterangium crassirhachis Zeller & Dodge

Hysterangium setchellii Fischer

Leucangium carthusianum (Tul. & Tul.) Paoletti

Leucogaster candidus (Harkn.) Fogel comb. ined.

Leucogaster citrinus (Harkn.) Zeller & Dodge [4 7]

Leucogaster gelatinosus Fogel nom. ined.

Leucogaster rubescens Zeller & Dodge

Leucogaster sp. nov.

Leucophleps magnata Harkn.

Leucophleps spinispora Fogel

Melanogaster ambiguus (Vitt.) Tul. & Tul.

Scientific name[1]
Melanogaster euryspermus (Zeller & Dodge) Zeller
Melanogaster natsii Wang, Trappe, & Castellano sp. nov.
Melanogaster thiersii Wang, Trappe, & Castellano sp. nov.
Melanogaster trappei Wang sp. nov.
Melanogaster tuberiformis Corda in Sturm
Melanogaster variegatus (Vitt.) Tul. & Tul.
Pachyphloeus thysellii Colgan & Castellano, nom. ined.[6]
Piloderma fallax (Lib.) Stalp.
Radiigera fuscogleba Zeller
Rhizopogon hawkerae Smith
Rhizopogon rogersii Smith
Rhizopogon subareolatus Smith
Rhizopogon villosulus Zeller
Rhizopogon vinicolor Smith
Rhizopogon vulgaris (Vittad.) M. Lange
Scleroderma hypogaeum Zeller
Truncocolumella citrina Zeller
Tuber anniae Colgan & Trappe
Tuber gibbosum Harkn.
Tuber monticola Harkn.
Genus & species nov. # 1[6]
Genus & species nov. # 2[6]
Epigeous fungi (mushrooms):
Agaricus albolutescens Zeller
Agaricus diminutivus Pk.
Agaricus micromegathus Pk.
Agaricus silvicola (Vitt.) Pk.
Agrocybe praecox (Fr.) Fayod
Albatrellus pescaprae (Fr.) Pouzar

Scientific name[1]

Aleuria aurantia (Fr.) Fuckel

Amanita gemmata var. *gemmata* (Fr.) Bert.[4]

Amanita gemmata var. *exannulata* Lange

Amanita pantherina (DC:Fr.) Schum.[4]

Amanita porphyria (A.& S. ex Fr.) Secr.[4]

Amanita silvicola Kauffm.

Amanita smithiana Bas.[4]

Armillaria mellea (Vahl ex Fr.) Karsten **grp.**[8]

Auriscalpium vulgare S.F. Gray

Bolbitius sp.

Boletus chrysenteron Fr.[4]

Boletus zelleri Murr.[4]

Cantharellus formosus Corner[4 7 9]

Cantharellus subalbidus Smith & Morse[4 7]

Cantharellus tubaeformis Fr.[4 7]

Clavaria sp.

Clavulina cinerea (Fr.) Schroet.[4 7]

Clavulina cristata (Fr.) Schroet.[4 7]

Clavulinopsis corniculata (Schaeff.:Fr.) Corner (= *Clavaria corniculata* Fr.)

Clavulinopsis laeticolor (Berk. & Curt.) R.H. Petersen[4]

Clitocybe coniferophila H.E. Bigelow **grp.**[8]

Clitocybe deceptiva H.E. Bigelow

Clitocybe dilatata Pers. ex Karsten

Clitocybe inversa (Fr.) Gill.

Collybia acervata (Fr.) Kummer[4]

Collybia alcalivirens Singer

Collybia butyracea (Fr.) Quel.[4]

Collybia confluens (Pers. ex Fr.) Kummer

Collybia dryophila (Bull. ex Fr.) Kummer

Collybia fuscopurpurea (Pers.:Fr.) Kumm. **grp.**[8]

Scientific name[1]

Collybia oregonensis A.H. Smith

Collybia racemosa (Pers.:Fr.) Quél. [4 7]

Coltricia cinnamomea (Jacq.:Pers.) Murr.

Conocybe cyanopus (Atk.) Kühner

Conocybe tenera (Schaeff. ex Fr.) Kühner

Coprinus micaceus (Bull. ex Fr.) Fr.

Coprinus plicatilis (Curt.:Fr.) Fr.

Coprinus sylvaticus Pk.

Cortinarius acutus Fr. **grp.**[4 8]

Cortinarius alboviolaceus (Pers. ex Fr.) Fr.

Cortinarius angulosus Fr.

Cortinarius brunneus Fr.

Cortinarius californicus A.H. Smith

Cortinarius cinnamomeus (Fr.) Fr. **grp.**[8]

Cortinarius corrugatus Peck

Cortinarius cotoneus Fr. **grp.**[8]

Cortinarius crassus Fr.[4]

Cortinarius crystallinus Fr.

Cortinarius decipiens Fr.

Cortinarius duracinus Fr.

Cortinarius evernius Fr.[4]

Cortinarius glaucopus (Schaeff.:Fr.) Fr.[4]

Cortinarius hemitrichus Fr.

Cortinarius infractus (Pers.:Fr.) Fr.[4]

Cortinarius laniger Fr. **grp.**[4 8]

Cortinarius malachius Fr.

Cortinarius mucosus (Bull.:Fr.) Fr.

Cortinarius multiformis (Fr.) Fr.

Cortinarius nigrocuspidatus

Cortinarius obtusus Fr. **grp.**[4 8]

Scientific name[1]

Cortinarius olympianus A.H. Smith [4 7]

Cortinarius paleaceus Fr.[4]

Cortinarius percomis Fr.

Cortinarius phoeniceus (Bull.) R. Maire var. *occidentalis* Smith.[4]

Cortinarius pinetorum (Fr.) Kauffman[4]

Cortinarius plumiger (Fr.) Fr.

Cortinarius prasinus Fr.

Cortinarius pseudobolaris Maire *sensu* Smith

Cortinarius rubripes Kauffman

Cortinarius sanguineus (Fr.) Fr.[4]

Cortinarius scaurus Fr. **grp.**[8]

Cortinarius semisanguineus (Fr.) Gillet [4]

Cortinarius subfoetidus A.H. Smith

Cortinarius subg. *bulbopodium*

Cortinarius subg. *phlegmacium*

Cortinarius subg. *sericeocybe*

Cortinarius subg. *telemonia*

Cortinarius superbus A.H. Smith

Cortinarius triformis Fr.

Cortinarius uliginosus Berk.

Cortinarius uraceus Fr.

Cortinarius vibratilis Fr.[4]

Crepidotus herbarum (Pk.) Sacc.

Crepidotus mollis (Fr.) Stde.

Crucibulum laeve (Huds.) Kamb.

Cystoderma amianthinum (Scop.:Fr.) Fr.

Cystoderma fallax Smith & Singer

Cystoderma granulosum (Fr.) Fayod

Cudonia monticola Mains [4 7]

Dacrymyces palmatus (Schw.) Bres.

Scientific name[1]

Entoloma nidorosum (Fr.) Quél. **grp.**[8]

Entoloma rhodopolium (Fr.) Kumm. **grp.**[8]

Fomitopsis cajanderi (Karsten) Kotlaba & Pouz.

Fomitopsis officinalis (Fr.) Bond. & Sing.

Fomitopsis pinicola (Fr.) Karst.

Galerina autumnalis (Pk.) Smith & Singer

Galerina heterocystis (Atk.) A.H. Smith [4 7]

Galerina marginata (Fr.) Kühner

Gomphidius glutenosus (Fr.) Fr.

Gomphidius oregonensis Peck

Gomphidius smithii Miller

Gomphidius subroseus Kauffman

Gomphus clavatus (Fr.) S.F. Gray [4 7]

Guepiniopsis alpinus (Tracy & Earle) Bres.

Gymnopilus bellulus (Peck) Murr.[4]

Gymnopilus liquiritiae (Pers.:Fr.) Karst.

Gymnopilus penetrans (Fr. ex Fr.) Murr.

Gymnopilus sapineus (Fr.) Maire

Hebeloma crustuliniforme (Bull. ex St. Amams) Quel.[4]

Hebeloma sinapizans (Paulet:Fr.) Gill.

Helvella crispa Scop. ex Fr.

Helvella elastica Bulliard:Fr.[4 7]

Helvella lacunosa Afz. ex Fr.

Hydnum repandum L. ex Fr. (= *Dentinum repandum* (Fr.) S.F. Gray)[4 7]

Hydnum umbilicatum Pk. (= *Dentinum umbilicatum* (Pk.) Pouzar)[4 7]

Hygrocybe miniata (Fr.) Kumm.

Hygrophoropsis aurantiaca (Wulf. ex Fr.) Maire[4]

Hygrophorus calophyllus Karst.

Hygrophorus conicus (Fr.) Fr. (= *Hygrocybe conica* Fr.)[4]

Hygrophorus glutinosus (Schff.:Fr.) Fr.

Scientific name[1]

Hypholoma capnoides (Fr. ex Fr.) Kummer (= *Naematoloma capnoides* (Fr.) Karsten)[4]

Hypholoma dispersum (Fr.) Quel. (= *Naematoloma dispersum* (Fr.) Karsten)[4]

Hypholoma fasciculare (Huds. ex Fr.) Kummer (= *Naematoloma fasciculare* (Huds. ex Fr.) Karsten)

Inocybe albodisca Pk.

Inocybe calamistrata (Fr.) Gillet[4]

Inocybe cincinnatula Kühner

Inocybe cookei Bres.

Inocybe eutheles Berk. & Br.

Inocybe fastigiata (Schaeff. ex Fr.) Quel.

Inocybe fuscodisca (Peck) Massae[4]

Inocybe geophylla (Sow. ex Fr.) Kummer

Inocybe lanatodisca Kauffman

Inocybe lanuginosa (Bull. ex Fr.) Kummer[4]

Inocybe lilacina (Boud.) Kauffman

Inocybe maculata Boud.

Inocybe mixtilis Britz.

Inocybe olympiana A.H. Smith

Inocybe pudica Kühner

Inocybe sororia Kauffman[4]

Inocybe subcarpta Kühner & Boursier

Laccaria amethysteo-occidentalis Mueller[4]

Laccaria laccata (Scop. ex Fr.) Cke.[4]

Lactarius affinis Pk.

Lactarius deliciosus (Fr.) S. F. Gray[4]

Lactarius fragilis var. *rubidus* Hels. & Smith

Lactarius pseudomucidus Smith & Hesler[4]

Lactarius rubrilacteus Smith & Hesler

Lactarius subflammeus Smith & Hesler

Lactarius uvidus Fr.

Scientific name[1]

Laetiporus sulphureus (Bull. ex Fr.) Murr.[4]

Lentinus sulcatus Berk.

Leotia lubrica Fr.

Lepiota clypeolaria (Bull. ex Fr.) Kummer

Lepiota rhacodes Pilat

Leptonia gracilipes Peck

Leptonia parva Peck

Leptonia serrulata (Fr.:Fr.) Kumm.

Leptonia undulatella (Peck) Sacc.

Limacella glioderma (Fr.) R. Maire[4]

Lycoperdon foetidum Bon.

Lycoperdon perlatum Pers.

Lycoperdon pyrifome Schaeff.:Pers.[4]

Lyophyllum decastes (Fr.) Singer

Marasmius candidus (Bolt.:Fr.) Fr.

Marasmius copelandii Peck

Marasmius umbilicatus Kauffman

Melanoleuca melaleuca (Pers. ex Fr.) Murr.

Mycena acicula (Schaeff. ex Fr.) Kummer

Mycena amabilissima (Peck) Sacc.[4]

Mycena atroalboides (Peck) Sacc.

Mycena aurantiomarginata (Fr.) Quél.[4]

Mycena capillaris (Schum.:Fr.) Kumm

Mycena citrinomarginata Gillet

Mycena delicatella (Pk.) Smith

Mycena elegantula Peck[4]

Mycena epipterygia (Fr.) S. F. Gray[4]

Mycena maculata Karst.[4]

Mycena murina Murrill **grp.**[8]

Mycena occidentalis Murrill

Scientific name[1]

Mycena oregonensis Smith

Mycena pura (Pers. ex Fr.) Kummer

Mycena purpureofusca (Peck) Sacc.

Mycena rorida (Fr.) Quél.

Mycena scabripes (Murrill) Singer

Mycena subcana A.H. Smith

Nidula candida (Pk.) White

Nidula niveotomentosa (Henn.) Lloyd

Nolanea mammosa (L.) Quél. **grp.**[8]

Omphalina luteicolor Murrill

Otidea leporina var. *leporina* (Batsch:Fr.) Fuckel [4, 7]

Paxillus atrotomentosus (Fr.) Fr.[4]

Peziza badia Pers.

Peziza sp.

Phaeolus schweinitzii (Fr.) Pat.

Phellinus pini (Fr.) Ames

Pholiota astragalina (Fr.) Singer[4]

Pholiota decorata (Murr.) Smith & Kessler[4]

Pholiota mutabilis (Schaeff. ex Fr.) Kummer

Pholiota terrestris Overholts

Pleurocybella porrigens (Pers. ex Fr.) Singer (= *Pleurotus porrigens* (Pers. ex Fr.) Kummer[4]

Pleurotus ostreatus (Jacq. ex Fr.) Kummer

Pluteus cervinus (Fr.) Kummer

Polyporus badius (S.F. Gray) Schw.

Polyporus hirtus Quél. (=*Jahnoporus hirtus* (Quél.) Nuss.)[4]

Polyporus volvatus Peck (=*Cryptoporus volvatus* (Peck) Shear)

Psathyrella gracilis (Fr.) Quél.

Psathyrella hydrophila (Fr.) Maire

Psathyrella longistriata (Murre) Smith

Scientific name[1]
Pseudohydnum gelatinosum (Scop. ex Fr.) Karsten
Pseudoplectania melaena (Fr.) Boud.
Ramaria stricta (Fr.) Quél.
Ramariopsis kunzei (Fr.) Donk.
Rhodocybe hirneola (Fr.) Orton
Russula aeruginea Lindl.[4]
Russula albonigra (Krombh.) Fr.[4]
Russula alutacea (Pers.:Fr.) Fr.
Russula bicolor Burl.[4]
Russula brevipes Pk.
Russula cremoricolor Earle
Russula cyanoxantha (Schw.) Fr. **grp.**[8]
Russula densifolia (Secr.) Gillet
Russula emetica Fr.
Russula fragrantissima Rom.
Russula gracilis Burlingham
Russulas nigricans Fr.[4]
Russula placita Burl **grp.**[8]
Russula sororia (Fr.) Romell **grp.**[8]
Russula xerampelina (Schaeff. ex Secr.) Fr.[4]
Sparassis crispa Wulf:Fr.[4] [7]
Stereum complicatum (Fr.) Fr.
Strobilurus trullisatus (Murr.) Lennox
Stropharia ambigua (Pk.) Zeller
Stropharia hornemannii (Fr.) Lundell[4]
Suillus lakei (Murr.) Smith & Thiers
Suillus ponderosus Smith & Thiers
Suillus tomentosus (Kauff.) Singer, Snell, & Dick
Thelephora americana Lloyd
Thelephora palmata Scop.:Fr.

Scientific name[1]

Thelephora terrestris Fr.

Trametes hirsuta (Wulf.:Fr.) Pilat

Trametes versicolor (L. ex Fr.) Pilat (= *Coriolus versicolor* (L. ex Fr.) Quél.)

Trichaptum abietinus (Fr.) Donk.

Trichoglossum hirsutum (Fr.) Boudier

Tricholoma flavovirens (Pers. ex Fr.) Lundell[4]

Tricholoma imbricatum (Fr.:Fr.) Kumm.[4]

Tricholoma sulphureum (Bull.:Fr.) Kumm.

Tricholoma terreum (Schaeff.:Fr.) Kumm.

Tricholomopsis rutilans (Schaeff. ex Fr.) Singer

Tubaria furfuracea (Pers. ex Fr.) Gillet

Tyromyces caesius (Fr.) Murr.

Tyromyces chioneus (Fr.) Karsten

Xeromphalina campanella (Bat. ex Fr.) Kuhner & Maire

Xeromphalina fulvipes (Murr.) Smith[4]

Xylaria hypoxylon (L. ex Hooker) Grev.

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[1] Vascular plant names and authorities are from Kartesz (1994), with synonomy (in parentheses) to names found in Hitchcock and Cronquist (1973), where different. Mammal, reptile, amphibian, and bird names and authorities are from Banks and others (1987) and Jones and others (1992). Hypogeous fungi identifications were made and authorities assigned by J.M. Trappe of Oregon State University, Corvallis, OR. Epigeous fungal names are from Arora (1986), Castellano and O'Dell (1997), Phillips (1991), and Tylutki (1979, 1987).

[2] Alphacode abbreviations for plants are from the PLANTS database (USDA NRCS 1997).

[3] Common names are provided for plants and animals. Because many fungi do not have common names, they are not provided for any of the included fungi.

[4] Species closely associated with late-successional or old-growth forests within the range of the northern spotted owl (FEMAT 1993).

[5] First record for this vascular plant in Washington (Thysell and others 1997).

[6] Previously undescribed hypogeous fungal species collected during the course of the Forest Ecosystem Study.

[7] Species to be protected through survey and management guidelines as outlined in the standards and guidelines for management of habitat for late-successional and old-growth forest related species within the range of the northern spotted owl (USDA and USDI 1994).

[8] Those fungal taxa ending in **grp.** have been placed in a group of related or similar species pending further investigation.

[9] This is the common yellow chantarelle of the Pacific Northwest. For a discussion of its nomenclatural and taxonomic history, see Castellano and O'Dell (1997).

Appendix 2

Nestbox and Cavity Design

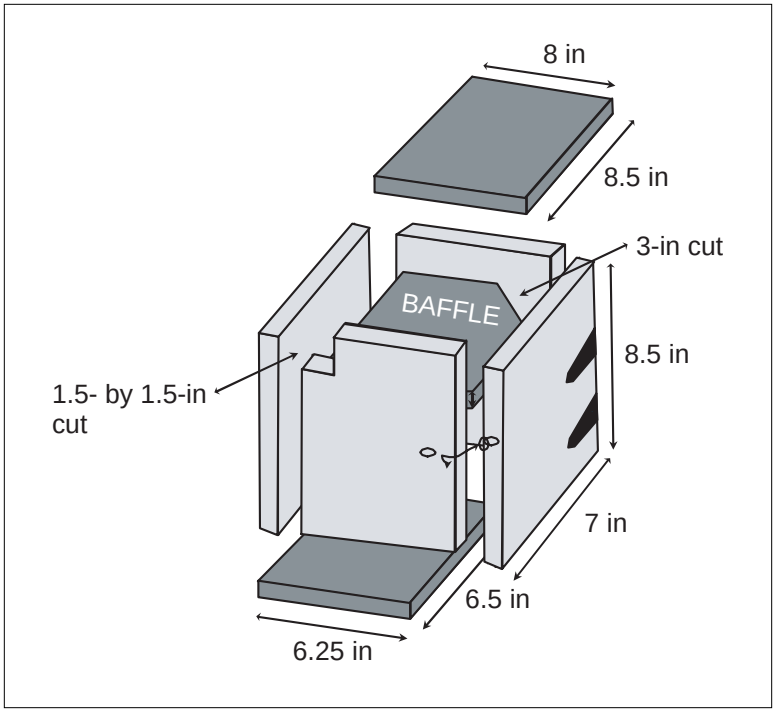


Figure 11—Construction of the Flyger nestboxes (Carey and Gill 1983).

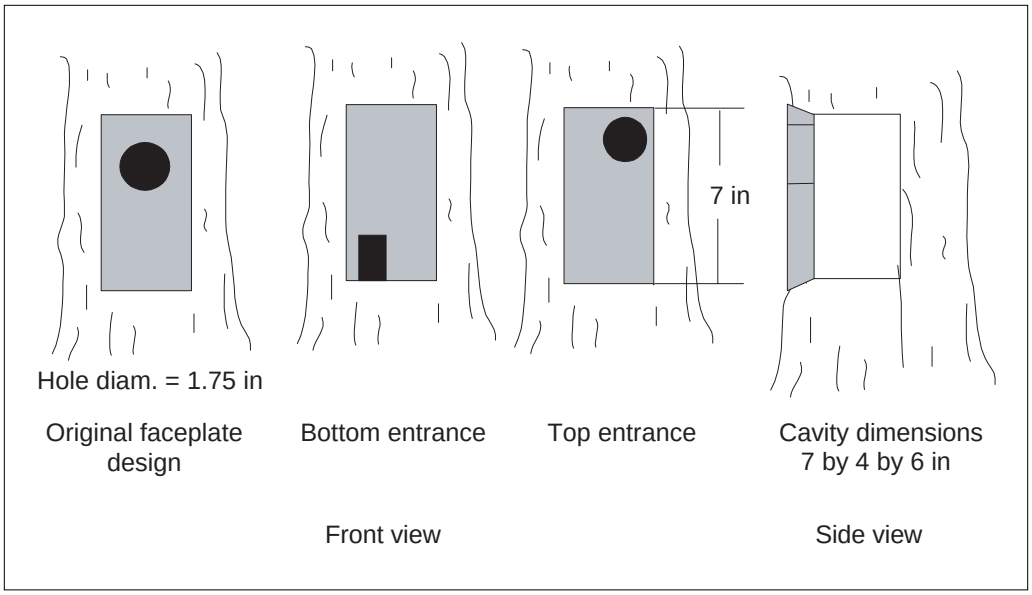


Figure 12—Construction and dimensions of the original cavities and faceplates.

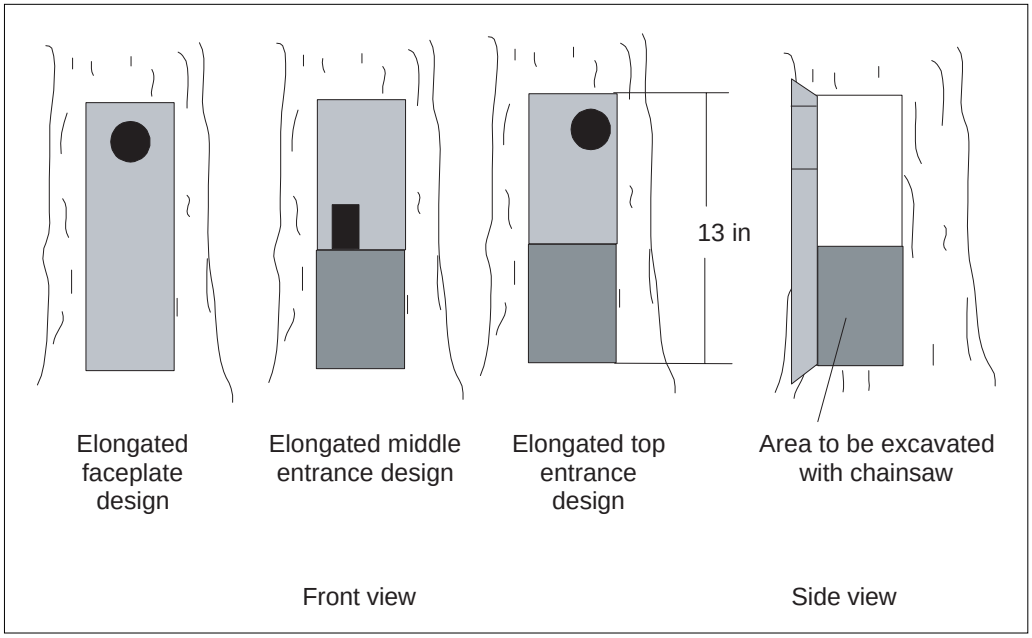


Figure 13—Construction and dimensions of the modified cavities and faceplates.

Appendix 3
Permanent Plot Locations

Star/Stellar permanent plots					Farley/Hill permanent plots				
Stand	**Cell ID[1]**	**Target treatment**	**Retained RD**	***Post facto* RD class[2]**	**Stand**	**Cell ID[1]**	**Target treatment**	**Retained RD**	***Post facto* RD class[2]**
101	A/B 3/4	RT	1.9	RD2	301	A/B 6/7	CT	-[3]	-
101	B/C 1/2	RT	3.4	RD4	301	B/C 4/5	CT	-	-
101	C/D 1/2	RT	1.1	RD2	301	E/F 3/4	CT	-	-
101	C/D 3/4	RT	2.5	RD2	301	E/F 4/5	CT	-	-
101	C/D 6/7	RT	1.2	RD2	301	F/G 1/2	CT	-	-
101	G/H 3/4	RT	5.1	RD6	301	G/H 5/6	CT	-	-
101	B/C 3/4	HT	3.0	RD2	302	B/C 3/4	RT	1.3	RD2
101	B/C 4/5	HT	6.1	RD6	302	C/D 4/5	RT	1.0	RD2
101	B/C 6/7	HT	3.8	RD4	302	D/E 6/7	RT	1.9	RD2
101	D/E 5/6	HT	7.0	RD8	302	E/F 4/5	RT	.6	RD2
101	F/G 4/5	HT	4.0	RD4	302	F/G 2/3	RT	3.0	RD2
101	G/H 6/7	HT	6.5	RD6	302	G/H 5/6	RT	2.9	RD2
101	A/B 6/7	LT	6.7	RD6	302	B/C 2/3	HT	5.5	RD6
101	C/D 2/3	LT	5.3	RD6	302	B/C 5/6	HT	6.4	RD6
101	E/F 4/5	LT	5.3	RD6	302	C/D 6/7	HT	2.9	RD2

Star/Stellar permanent plots					Farley/Hill permanent plots				
Stand	Cell ID[1]	Target treatment	Retained RD	*Post facto* RD class[2]	Stand	Cell ID[1]	Target treatment	Retained RD	*Post facto* RD class[2]
101	F/G 3/4	LT	4.5	RD4	302	C/D 7/8	HT	4.8	RD6
101	G/H 5/6	LT	6.7	RD6	302	D/E 3/4	HT	6.4	RD6
101	G/H 7/8	LT	6.0	RD6	302	F/G 6/7	HT	7.2	RD8
102	A/B 3/4	CT	-	-	302	A/B 1/2	LT	5.9	RD6
102	B/C 6/7	CT	-	-	302	A/B 7/8	LT	6.4	RD6
102	C/D 2/3	CT	-	-	302	D/E 2/3	LT	5.5	RD6
102	D/E 5/6	CT	-	-	302	E/F 3/4	LT	8.3	RD8
102	F/G 2/3	CT	-	-	302	F/G 7/8	LT	5.5	RD6
102	F/G 6/7	CT	-	-	302	G/H 3/4	LT	6.4	RD6
103	C/D 7/8	RT[4]	1.7	RD2	303	B/C 2/3	RT	1.9	RD2
103	D/E 6/7	RT	2.3	RD2	303	C/D 3/4	RT	1.0	RD2
103	E/F 2/3	RT	1.7	RD2	303	E/F 3/4	RT	1.6	RD2
103	E/F 7/8	RT	3.3	RD4	303	F/G 1/2	RT	1.9	RD2
103	G/H 1/2	RT	1.5	RD2	303	F/G 5/6	RT	1.8	RD2
103	A/B 3/4	HT	6.0	RD6	303	G/H 5/6	RT	1.6	RD2
103	A/B 7/8	HT	6.0	RD6	303	A/B 3/4	HT	2.8	RD2
103	D/E 1/2	HT	5.7	RD6	303	C/D 1/2	HT	5.4	RD6
103	D/E 3/4	HT	6.2	RD6	303	C/D 4/5	HT	6.3	RD6
103	E/F 6/7	HT	6.0	RD6	303	C/D 6/7	HT	5.5	RD6
103	F/G 3/4	HT	7.6	RD8	303	E/F 6/7	HT	7.3	RD8
103	A/B 4/5	LT	4.7	RD4	303	F/G 3/4	HT	4.2	RD4
103	B/C 2/3	LT	6.3	RD6	303	A/B 6/7	LT	5.4	RD6
103	B/C 5/6	LT	6.3	RD6	303	A/B 7/8	LT	5.4	RD6
103	B/C 6/7	LT	6.0	RD6	303	C/D 7/8	LT	3.6	RD4
103	C/D 3/4	LT	6.0	RD6	303	D/E 1/2	LT	4.6	RD4
103	E/F 3/4	LT	5.1	RD6	303	E/F 2/3	LT	4.6	RD4
104	B/C 4/5	CT	-	-	303	F/G 2/3	LT	3.9	RD4
104	C/D 6/7	CT	-	-	304	A/B 3/4	CT	-	-
104	D/E 2/3	CT	-	-	304	B/C 3/4	CT	-	-
104	E/F 7/8	CT	-	-	304	B/C 7/8	CT	-	-

Star/Stellar permanent plots					Farley/Hill permanent plots				
Stand	Cell ID[1]	Target treatment	Retained RD	*Post facto* RD class[2]	Stand	Cell ID[1]	Target treatment	Retained RD	*Post facto* RD class[2]
104	F/G 3/4	CT	-	-	304	C/D 5/6	CT	-	-
104	F/G 5/6	CT	-	-	304	D/E 7/8	CT	-	-
201	B/C 2/3	RT	2.4	RD2	304	F/G 2/3	CT	-	-
201	B/C 7/8	RT	1.0	RD2	401	B/C 2/3	RT	2.7	RD2
201	C/D 5/6	RT	.8	RD2	401	C/D 5/6	RT	1.2	RD2
201	C/D 7/8	RT	.4	RD2	401	D/E 1/2	RT	3.5	RD4
201	E/F 4/5	RT	.5	RD2	401	E/F 4/5	RT	3.7	RD4
201	F/G 4/5	RT	1.8	RD2	401	E/F 7/8	RT	1.4	RD4
201	A/B 1/2	HT	4.5	RD4	401	F/G 4/5	RT	1.6	RD4
201	A/B 2/3	HT	7.0	RD8	401	A/B 4/5	HT	4.3	RD4
201	E/F 6/7	HT	4.7	RD4	401	C/D 2/3	HT	3.6	RD4
201	F/G 3/4	HT	3.7	RD4	401	C/D 3/4	HT	6.4	RD6
201	G/H 2/3	HT	4.3	RD4	401	E/F 5/6	HT	7.7	RD8
201	G/H 3/4	HT	3.7	RD4	401	F/G 7/8	HT	4.5	RD4
201	A/B 3/4	LT	6.0	RD6	401	G/H 5/6	HT	8.4	RD8
201	A/B 5/6	LT	8.4	RD8	401	A/B 3/4	LT	3.5	RD4
201	C/D 3/4	LT	3.7	RD4	401	A/B 6/7	LT	7.4	RD8
201	C/D 4/5	LT	6.0	RD6	401	B/C 1/2	LT	7.1	RD8
201	F/G 6/7	LT	5.2	RD6	401	D/E 4/5	LT	6.4	RD6
201	F/G 7/8	LT	5.2	RD6	401	E/F 3/4	LT	3.6	RD4
202	A/B 1/2	RT	1.7	RD2	401	F/G 6/7	LT	8.3	RD8
202	A/B 3/4	RT	1.4	RD2	402	A/B 5/6	CT	-	-
202	B/C 4/5	RT	3.7	RD4	402	B/C 3/4	CT	-	-
202	E/F 4/5	RT	1.2	RD2	402	C/D 7/8	CT	-	-
202	F/G 2/3	RT	.7	RD2	402	D/E 2/3	CT	-	-
202	F/G 6/7	RT	.8	RD2	402	D/E 5/6	CT	-	-
202	B/C 3/4	HT	5.9	RD6	402	F/G 4/5	CT	-	-
202	B/C 7/8	HT	5.9	RD6	403	A/B 6/7	RT	1.3	RD2
202	D/E 3/4	HT	4.4	RD4	403	D/E 1/2	RT	1.5	RD2
202	E/F 2/3	HT	5.2	RD6	403	E/F 2/3	RT	.7	RD2

Star/Stellar permanent plots					Farley/Hill permanent plots				
Stand	Cell ID[1]	Target treatment	Retained RD	*Post facto* RD class[2]	Stand	Cell ID[1]	Target treatment	Retained RD	*Post facto* RD class[2]
202	F/G 7/8	HT	3.9	RD4	403	E/F 6/7	RT	1.8	RD2
202	G/H 2/3	HT	4.4	RD4	403	F/G 3/4	RT	2.8	RD2
202	A/B 4/5	LT	7.1	RD8	403	F/G 7/8	RT	1.4	RD2
202	D/E 2/3	LT	3.1	RD2	403	A/B 2/3	HT	6.5	RD6
202	E/F 3/4	LT	4.0	RD4	403	B/C 4/5	HT	5.5	RD6
202	F/G 3/4	LT	7.1	RD8	403	B/C 6/7	HT	7.3	RD8
202	F/G 4/5	LT	5.2	RD6	403	C/D 2/3	HT	6.9	RD8
202	G/H 3/4	LT	5.9	RD6	403	C/D 5/6	HT	3.9	RD4
203	A/B 7/8	CT	-	-	403	D/E 3/4	HT	6.5	RD6
203	C/D 3/4	CT	-	-	403	B/C 1/2	LT	7.3	RD8
203	C/D 5/6	CT	-	-	403	C/D 4/5	LT	7.3	RD8
203	D/E 1/2	CT	-	-	403	D/E 2/3	LT	4.3	RD4
203	D/E 6/7	CT	-	-	403	F/G 4/5	LT	5.4	RD6
203	F/G 5/6	CT	-	-	403	F/G 6/7	LT	6.5	RD6
204	A/B 7/8	CT	-	-	403	G/H 4/5	LT	6.5	RD6
204	B/C 2/3	CT	-	-	404	A/B 3/4	CT	-	-
204	B/C 6/7	CT	-	-	404	B/C 3/4	CT	-	-
204	D/E 2/3	CT	-	-	404	D/E 2/3	CT	-	-
204	D/E 5/6	CT	-	-	404	D/E 5/6	CT	-	-
204	F/G 5/6	CT	-	-	404	F/G 4/5	CT	-	-
					404	F/G 6/7	CT	-	-

[1] Cell ID provides the alphanumeric identity of the four corners of the 40- by 40-m cell in the center of which is the permanent plot.

[2] *Post facto* RD classes are delineated as follows: RD2 = RD < 3.25; RD4 = 3.25 < RD < 4.75; RD6 = 4.75 < RD < 6.75; RD8 = RD > 6.75.

[3] - = not applicable.

[4] All target subtreatments or treatments in every stand were sampled with six plots, except for the RT subtreatments in stand 103 which contained only five total RT cells of sufficient size in which to establish permanent plots.

Appendix 4
Cell-by-Cell Estimates of Stand Parameters (From Postthinning Assessment)

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter[1]	Residual quadratic mean diameter[1]	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----Centimeters----		*-------m^2/ha-------*		*-------No./ha-------*			
101	A1	34.0	34.0	18.4	18.4	188.6	188.6	3.1	3.1
101	A2	34.4	36.9	41.3	36.7	400.5	358.1	7.0	6.0
101	A3	31.1	34.0	24.9	11.0	273.5	155.6	4.5	1.9
101	A4	34.4	36.9	41.6	23.7	746.7	353.7	7.1	3.9
101	A5	34.6	37.5	59.7	50.5	570.1	485.3	10.1	8.2
101	A6	34.6	37.5	59.7	41.3	570.1	400.5	10.1	6.7
101	A7	34.4	36.9	43.3	22.5	746.7	353.7	7.4	3.7
101	B1	31.1	34.0	48.3	19.8	495.1	141.5	8.7	3.4
101	B2	34.0	34.0	13.8	13.8	146.2	146.2	2.4	2.4
101	B3	31.1	34.0	22.1	17.3	243.2	176.8	4.0	3.0
101	B4	34.4	36.9	59.1	36.8	641.0	331.6	10.1	6.1
101	B5	31.1	34.0	18.4	13.8	188.6	146.2	3.3	2.4
101	B6	34.4	36.9	50.5	23.0	485.3	231.0	8.6	3.8
101	B7	34.6	37.5	55.1	50.5	527.7	485.3	9.4	8.2
101	C1	31.1	34.0	43.0	6.4	542.3	70.7	7.7	1.1
101	C2	34.4	36.9	64.3	32.1	612.4	315.7	11.0	5.3
101	C3	31.1	34.0	46.8	14.9	545.4	113.2	8.4	2.5
101	C4	34.4	36.9	43.1	26.1	508.4	221.0	7.4	4.3
101	C5	34.4	36.9	37.5	27.7	589.5	393.0	6.4	4.6
101	C6	31.1	34.0	47.2	6.7	620.9	70.7	8.5	1.2
101	C7	34.4	36.9	55.1	32.1	527.7	315.7	9.4	5.3
101	D1	34.4	36.9	45.9	27.6	442.9	273.4	7.8	4.5
101	D2	34.6	37.5	68.9	41.3	654.8	400.5	11.7	6.7
101	D3	36.9	36.9	27.6	27.6	273.4	273.4	4.5	4.5
101	D4	34.4	36.9	50.5	27.6	485.3	273.4	8.6	4.5
101	D5	34.6	37.5	61.3	42.8	795.8	486.3	10.4	7.0
101	D6	34.6	37.5	53.2	38.0	786.0	510.9	9.0	6.2
101	D7	34.4	36.9	41.3	27.6	400.5	273.4	7.0	4.5

Footnote is on p. 121.

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
101	E1	34.6	37.5	73.5	64.3	697.2	612.4	12.5	10.5
101	E2	34.4	36.9	41.3	36.7	400.5	358.1	7.0	6.0
101	E3	34.6	37.5	53.9	42.9	589.5	393.0	9.2	7.0
101	E4	34.4	36.9	45.9	32.1	442.9	315.7	7.8	5.3
101	E5	36.9	36.9	23.0	23.0	231.0	231.0	3.8	3.8
101	E6	31.1	34.0	45.7	6.8	396.1	42.4	8.2	1.2
101	E7	34.0	34.0	18.4	18.4	188.6	188.6	3.1	3.1
101	F1	34.4	36.9	32.1	23.0	315.7	231.0	5.5	3.8
101	F2	31.1	34.0	36.7	18.4	358.1	188.6	6.6	3.1
101	F3	36.9	36.9	27.6	27.6	273.4	273.4	4.5	4.5
101	F4	34.4	36.9	36.6	24.4	464.2	265.3	6.2	4.0
101	F5	34.0	34.0	18.4	18.4	188.6	188.6	3.1	3.1
101	F6	34.6	37.5	50.5	45.9	485.3	442.9	8.6	7.5
101	F7	34.6	37.5	59.7	45.9	570.1	442.9	10.1	7.5
101	G1	34.0	34.0	4.6	4.6	61.4	61.4	.8	.8
101	G2	34.6	37.5	49.5	43.8	353.7	275.1	8.4	7.2
101	G3	34.4	36.9	59.3	30.7	633.5	240.5	10.1	5.1
101	G4	36.9	36.9	23.0	23.0	231.0	231.0	3.8	3.8
101	G5	34.6	37.5	55.1	41.3	527.7	400.5	9.4	6.7
101	G6	34.6	37.5	45.8	39.7	486.3	353.7	7.8	6.5
101	G7	36.9	36.9	36.7	36.7	358.1	358.1	6.0	6.0
103	A1	34.4	37.2	52.4	27.2	530.5	198.9	8.9	4.5
103	A2	34.4	37.2	27.2	24.1	353.7	275.1	4.6	3.9
103	A3	34.4	37.2	52.7	36.8	397.9	221.0	9.0	6.0
103	A4	34.4	37.2	28.0	28.7	393.0	353.7	4.8	4.7
103	A5	34.6	43.7	50.5	41.3	485.3	400.5	8.6	6.3
103	A6	34.6	43.7	49.8	37.5	746.7	471.6	8.5	5.7
103	A7	34.4	37.2	59.7	36.7	570.1	358.1	10.2	6.0

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
103	B1	34.4	37.2	59.7	36.7	570.1	358.1	10.2	6.0
103	B2	34.4	37.2	36.7	32.1	358.1	315.7	6.3	5.3
103	B2	34.6	43.7	50.5	41.3	485.3	400.5	8.6	6.3
103	B4	34.4	37.2	36.7	27.6	358.1	273.4	6.3	4.5
103	B5	34.6	43.7	64.3	41.3	612.4	400.5	10.9	6.3
103	B6	34.4	37.2	45.9	36.7	442.9	358.1	7.8	6.0
103	B7	34.6	43.7	50.5	41.3	485.3	400.5	8.6	6.3
103	C1	34.4	37.2	36.7	32.1	358.1	315.7	6.3	5.3
103	C2	34.4	37.2	50.5	36.7	485.3	358.1	8.6	6.0
103	C3	34.4	37.2	50.5	36.7	485.3	358.1	8.6	6.0
103	C4	34.4	37.2	50.5	27.6	485.3	273.4	8.6	4.5
103	C5	43.7	43.7	41.3	41.3	400.5	400.5	6.3	6.3
103	C6	37.2	37.2	23.0	23.0	231.0	231.0	3.8	3.8
103	C7	31.1	35.2	39.2	10.3	382.0	56.6	7.0	1.7
103	D1	34.6	43.7	50.4	37.9	508.4	331.6	8.6	5.7
103	D2	34.4	37.2	41.3	27.6	400.5	273.4	7.0	4.5
103	D3	34.6	43.7	53.2	40.9	618.9	397.9	9.0	6.2
103	D4	34.4	37.2	36.7	23.0	358.1	231.0	6.3	3.8
103	D5	34.4	37.2	32.1	27.6	315.7	273.4	5.5	4.5
103	D6	31.1	35.2	52.1	13.8	622.5	113.2	9.3	2.3
103	D7	34.4	37.2	27.6	23.0	273.4	231.0	4.7	3.8
103	E1	34.6	43.7	55.1	41.3	527.7	400.5	9.4	6.3
103	E2	31.1	35.2	40.3	10.3	466.9	56.6	7.2	1.7
103	E3	34.4	37.2	40.9	31.3	589.5	432.3	7.0	5.1
103	E4	31.1	35.2	27.6	18.4	273.4	188.6	4.9	3.1
103	E5	34.4	37.2	52.8	28.5	746.7	353.7	9.0	4.7
103	E6	34.6	43.7	53.2	39.8	464.2	265.3	9.0	6.0
103	E7	31.1	35.2	53.8	19.5	495.1	84.9	9.7	3.3

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
103	F1	34.4	37.2	41.3	32.1	400.5	315.7	7.0	5.3
103	F2	34.6	43.7	50.5	41.3	485.3	400.5	8.6	6.3
103	F3	34.6	43.7	55.1	50.5	527.7	485.3	9.4	7.6
103	F4	34.4	37.2	36.7	32.1	358.1	315.7	6.3	5.3
103	F5	34.4	37.2	32.1	23.0	315.7	231.0	5.5	3.8
103	F6	34.4	37.2	52.2	29.3	746.7	353.7	8.9	4.8
103	F7	34.4	37.2	32.1	27.6	315.7	273.4	5.5	4.5
103	G1	31.1	35.2	49.4	8.8	650.8	70.7	8.9	1.5
103	G2	31.1	35.2	25.2	19.3	397.9	287.4	4.5	3.3
103	G3	35.2	35.2	13.8	13.8	146.2	146.2	2.3	2.3
103	G4	34.4	37.2	45.9	23.0	442.9	231.0	7.8	3.8
103	G5	34.4	37.2	45.9	32.1	442.9	315.7	7.8	5.3
103	G6	34.4	37.2	36.7	27.6	358.1	273.4	6.3	4.5
103	G7	34.4	37.2	50.5	32.1	485.3	315.7	8.6	5.3
201	A1	34.4	38.0	32.1	27.6	315.7	273.4	5.5	4.5
201	A2	34.6	36.5	48.4	42.1	464.2	397.9	8.2	7.0
201	A3	34.4	38.0	45.9	36.7	442.9	358.1	7.8	6.0
201	A4	34.4	38.0	50.5	27.6	485.3	273.4	8.6	4.5
201	A5	34.6	36.5	55.1	50.5	527.7	485.3	9.4	8.4
201	A6	34.4	38.0	59.7	36.7	570.1	358.1	10.2	6.0
201	A7	34.4	38.0	36.7	27.6	358.1	273.4	6.3	4.5
201	B1	34.6	36.5	71.2	49.3	685.3	375.8	12.1	8.2
201	B2	31.1	33.1	42.5	13.6	460.7	84.9	7.6	2.4
201	B3	34.4	38.0	59.7	27.6	570.1	273.4	10.2	4.5
201	B4	34.4	38.0	45.8	32.9	668.1	432.3	7.8	5.3
201	B5	34.6	36.5	78.1	64.3	739.6	612.4	13.3	10.6
201	B6	34.6	36.5	50.5	41.3	485.3	400.5	8.6	6.8
201	B7	31.1	33.1	19.1	5.8	263.5	42.4	3.4	1.0

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
201	C1	34.4	38.0	32.1	23.0	315.7	231.0	5.5	3.7
201	C2	34.4	38.0	49.0	33.3	746.7	432.3	8.4	5.4
201	C3	38.0	38.0	23.0	23.0	231.0	231.0	3.7	3.7
201	C4	34.4	38.0	55.1	36.7	527.7	358.1	9.4	6.0
201	C5	31.1	33.1	20.8	4.6	313.9	70.7	3.7	.8
201	C6	38.0	38.0	32.1	32.1	315.7	315.7	5.2	5.2
201	C7	31.1	33.1	25.4	2.4	426.2	28.3	4.6	.4
201	D1	34.6	36.5	53.3	48.6	550.2	471.6	9.1	8.0
201	D2	31.1	33.1	41.3	18.4	400.5	188.6	7.4	3.2
201	D3	34.6	36.5	82.7	64.3	782.0	612.4	14.1	10.6
201	D4	34.4	38.0	45.9	32.1	442.9	315.7	7.8	5.2
201	D5	38.0	38.0	27.6	27.6	273.4	273.4	4.5	4.5
201	D6	34.4	38.0	32.1	23.0	315.7	231.0	5.5	3.7
201	D7	38.0	38.0	27.6	27.6	273.4	273.4	4.5	4.5
201	E1	31.1	33.1	13.8	9.2	146.2	103.8	2.5	1.6
201	E2	38.0	38.0	23.0	23.0	231.0	231.0	3.7	3.7
201	E3	34.4	38.0	42.2	35.6	530.5	397.9	7.2	5.8
201	E4	31.1	33.1	33.6	2.7	448.3	28.3	6.0	.5
201	E5	34.4	38.0	45.0	25.8	628.8	314.4	7.7	4.2
201	E6	34.4	38.0	45.4	28.7	707.4	331.6	7.7	4.7
201	E7	34.6	36.5	64.3	45.9	612.4	442.9	10.9	7.6
201	F1	34.4	38.0	41.3	23.0	400.5	231.0	7.0	3.7
201	F2	34.6	36.5	64.3	45.9	612.4	442.9	10.9	7.6
201	F3	34.4	38.0	27.6	23.0	273.4	231.0	4.7	3.7
201	F4	31.1	33.1	30.1	10.3	372.2	84.9	5.4	1.8
201	F5	34.6	36.5	68.9	50.5	654.8	485.3	11.7	8.4
201	F6	34.4	38.0	50.5	32.1	485.3	315.7	8.6	5.2
201	F7	34.4	38.0	45.9	32.1	442.9	315.7	7.8	5.2

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
201	G1	34.4	38.0	36.7	23.0	358.1	231.0	6.3	3.7
201	G2	34.4	38.0	40.1	26.3	397.9	221.0	6.8	4.3
201	G3	31.1	33.1	38.6	21.2	596.8	287.4	6.9	3.7
201	G4	34.6	36.5	52.2	45.9	550.2	393.0	8.9	7.6
201	G5	34.4	38.0	36.7	23.0	358.1	231.0	6.3	3.7
201	G6	34.4	38.0	43.8	30.9	707.4	432.3	7.5	5.0
201	G7	34.4	38.0	45.9	32.1	442.9	315.7	7.8	5.2
202	A1	31.1	35.5	39.7	9.9	353.7	99.0	7.1	1.7
202	A2	31.1	35.5	23.0	13.8	231.0	146.2	4.1	2.3
202	A3	31.1	35.5	41.3	8.3	481.0	84.9	7.4	1.4
202	A4	34.6	42.2	55.1	45.9	527.7	442.9	9.4	7.1
202	A5	34.4	38.3	32.1	27.6	315.7	273.4	5.5	4.4
202	A6	34.4	38.3	41.3	36.7	400.5	358.1	7.0	5.9
202	A7	34.6	42.2	52.7	38.2	596.8	331.6	9.0	5.9
202	B1	34.4	38.3	36.7	32.1	358.1	315.7	6.3	5.2
202	B2	34.6	42.2	50.5	45.9	485.3	442.9	8.6	7.1
202	B3	34.6	42.2	48.9	38.2	464.2	331.6	8.3	5.9
202	B4	31.1	35.5	60.3	22.1	608.3	155.6	10.8	3.7
202	B5	38.3	38.3	27.6	27.6	273.4	273.4	4.4	4.4
202	B6	34.4	38.3	36.7	27.6	358.1	273.4	6.3	4.4
202	B7	34.4	38.3	57.8	36.5	530.5	265.3	9.9	5.9
202	C1	34.4	38.3	68.2	35.2	596.8	309.5	11.6	5.7
202	C2	34.4	38.3	36.7	27.6	358.1	273.4	6.3	4.4
202	C3	34.4	38.3	41.3	32.1	400.5	315.7	7.0	5.2
202	C4	34.4	38.3	32.1	27.6	315.7	273.4	5.5	4.4
202	C5	34.4	38.3	41.3	36.7	400.5	358.1	7.0	5.9
202	C6	34.6	42.2	50.5	41.3	485.3	400.5	8.6	6.4
202	C7	34.4	38.3	36.7	27.6	358.1	273.4	6.3	4.4

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
202	D1	31.1	35.5	32.1	18.4	315.7	188.6	5.8	3.1
202	D2	35.5	35.5	18.4	18.4	188.6	188.6	3.1	3.1
202	D3	34.4	38.3	50.5	27.6	485.3	273.4	8.6	4.4
202	D4	31.1	35.5	56.7	13.1	523.4	84.9	10.2	2.2
202	D5	34.4	38.3	50.5	32.1	485.3	315.7	8.6	5.2
202	D6	34.6	42.2	66.7	54.5	668.1	471.6	11.3	8.4
202	D7	34.4	38.3	32.1	27.6	315.7	273.4	5.5	4.4
202	E1	34.6	42.2	74.5	46.7	903.8	471.6	12.7	7.2
202	E2	34.4	38.3	45.9	32.1	442.9	315.7	7.8	5.2
202	E3	34.4	38.3	27.3	24.5	235.8	196.5	4.7	4.0
202	E4	31.1	35.5	47.6	7.4	339.5	42.4	8.5	1.2
202	E5	34.4	38.3	47.2	28.6	530.5	287.4	8.0	4.6
202	E6	34.6	42.2	55.1	50.5	527.7	485.3	9.4	7.8
202	E7	34.4	38.3	59.7	36.7	570.1	358.1	10.2	5.9
202	F1	34.4	38.3	41.3	23.0	400.5	231.0	7.0	3.7
202	F2	31.1	35.5	26.6	4.0	268.8	14.1	4.8	.7
202	F3	34.6	42.2	51.9	46.3	432.3	353.7	8.8	7.1
202	F4	34.4	38.3	36.7	32.1	358.1	315.7	6.3	5.2
202	F5	34.4	38.3	36.7	32.1	358.1	315.7	6.3	5.2
202	F6	31.1	35.5	39.9	5.0	622.5	70.7	7.2	.8
202	F7	34.4	38.3	50.9	24.0	840.0	331.6	8.7	3.9
202	G1	34.4	38.3	23.8	23.8	235.8	235.8	4.1	3.8
202	G2	34.4	38.3	45.9	27.6	442.9	273.4	7.8	4.4
202	G3	34.4	38.3	50.5	36.7	485.3	358.1	8.6	5.9
202	G4	34.4	38.3	27.6	23.0	273.4	231.0	4.7	3.7
202	G5	34.4	38.3	30.6	24.1	628.8	471.6	5.2	3.9
202	G6	34.4	38.3	36.7	27.6	358.1	273.4	6.3	4.4
202	G7	42.2	42.2	41.3	41.3	400.5	400.5	6.4	6.4

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
302	A1	34.6	56.3	51.8	44.1	397.9	309.5	8.8	5.9
302	A2	56.3	56.3	48.2	48.2	189.8	189.8	6.4	6.4
302	A3	34.4	51.9	55.1	34.4	215.3	138.8	9.4	4.8
302	A4	34.4	51.9	48.2	34.4	189.8	138.8	8.2	4.8
302	A5	34.6	56.3	48.4	37.5	265.3	176.8	8.2	5.0
302	A6	56.3	56.3	41.3	41.3	164.3	164.3	5.5	5.5
302	A7	56.3	56.3	48.2	48.2	189.8	189.8	6.4	6.4
302	B1	34.6	56.3	48.7	38.7	331.6	243.2	8.3	5.2
302	B2	34.6	56.3	48.2	41.3	189.8	164.3	8.2	5.5
302	B3	31.1	51.6	40.5	9.5	241.4	42.4	7.3	1.3
302	B4	34.6	56.3	55.1	41.3	215.3	164.3	9.4	5.5
302	B5	34.6	56.3	55.1	48.2	215.3	189.8	9.4	6.4
302	B6	31.1	51.6	34.6	20.7	221.0	132.6	6.2	2.9
302	B7	51.6	51.6	20.7	20.7	87.9	87.9	2.9	2.9
302	C1	56.3	56.3	55.1	55.1	215.3	215.3	7.3	7.3
302	C2	34.6	56.3	75.8	48.2	291.8	189.8	12.9	6.4
302	C3	34.6	56.3	82.7	68.9	317.2	266.3	14.1	9.2
302	C4	31.1	51.6	55.2	6.9	285.6	42.4	9.9	1.0
302	C5	51.9	51.9	27.6	27.6	113.4	113.4	3.8	3.8
302	C6	51.6	51.6	20.7	20.7	87.9	87.9	2.9	2.9
302	C7	34.4	51.9	41.3	34.4	164.3	138.8	7.0	4.8
302	D1	34.6	56.3	58.2	47.0	309.5	243.2	9.9	6.3
302	D2	34.6	56.3	62.0	41.3	240.8	164.3	10.5	5.5
302	D3	34.6	56.3	62.0	48.2	240.8	189.8	10.5	6.4
302	D4	31.1	51.6	51.5	18.4	269.7	70.7	9.2	2.6
302	D5	51.9	51.9	34.4	34.4	138.8	138.8	4.8	4.8
302	D6	31.1	51.6	30.4	13.5	130.9	42.4	5.5	1.9
302	D7	51.9	51.9	34.4	34.4	138.8	138.8	4.8	4.8

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
302	E1	34.4	51.9	34.4	27.6	138.8	113.4	5.9	3.8
302	E2	34.6	56.3	51.9	46.2	243.2	198.9	8.8	6.2
302	E3	34.6	56.3	68.9	62.0	266.3	240.8	11.7	8.3
302	E4	31.1	51.6	53.0	4.0	213.1	14.1	9.5	.6
302	E5	34.4	51.9	36.9	28.2	198.9	154.7	6.3	3.9
302	E6	34.6	56.3	75.8	55.1	291.8	215.3	12.9	7.3
302	E7	56.3	56.3	64.8	64.8	265.3	265.3	8.6	8.6
302	F1	34.4	51.9	26.3	25.3	198.9	176.8	4.5	3.5
302	F2	31.1	51.6	56.0	21.6	290.0	113.2	10.0	3.0
302	F3	56.3	56.3	62.0	62.0	240.8	240.8	8.3	8.3
302	F4	34.4	51.9	39.0	36.2	132.6	110.5	6.7	5.0
302	F5	34.6	56.3	48.2	41.3	189.8	164.3	8.2	5.5
302	F6	56.3	56.3	53.6	53.6	221.0	221.0	7.2	7.2
302	F7	56.3	56.3	41.3	41.3	164.3	164.3	5.5	5.5
302	G1	31.1	51.6	57.2	15.8	336.0	70.7	10.3	2.2
302	G2	34.4	51.9	48.2	34.4	189.8	138.8	8.2	4.8
302	G3	34.6	56.3	55.1	48.2	215.3	189.8	9.4	6.4
302	G4	34.6	56.3	51.8	43.3	176.8	154.7	8.8	5.8
302	G5	31.1	51.6	69.7	20.6	153.0	42.4	12.5	2.9
302	G6	51.9	51.9	27.6	27.6	113.4	113.4	3.8	3.8
302	G7	51.9	51.9	34.4	34.4	138.8	138.8	4.8	4.8
303	A1	57.2	57.2	27.6	27.6	113.4	113.4	3.6	3.6
303	A2	57.2	57.2	34.4	34.4	138.8	138.8	4.6	4.6
303	A3	53.0	53.0	20.7	20.7	87.9	87.9	2.8	2.8
303	A4	34.6	58.2	62.0	41.3	240.8	164.3	10.5	5.4
303	A5	58.2	58.2	41.3	41.3	164.3	164.3	5.4	5.4
303	A6	34.6	58.2	55.1	41.3	215.3	164.3	9.4	5.4
303	A7	34.6	58.2	48.2	41.3	189.8	164.3	8.2	5.4

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
303	B1	34.6	58.2	48.2	41.3	189.8	164.3	8.2	5.4
303	B2	31.1	53.0	52.8	13.5	261.7	84.9	9.5	1.9
303	B3	34.6	58.2	62.3	47.4	265.3	198.9	10.6	6.2
303	B4	34.6	58.2	82.7	62.0	317.2	240.8	14.1	8.1
303	B5	53.0	53.0	6.9	6.9	36.9	36.9	.9	.9
303	B6	57.2	57.2	27.6	27.6	113.4	113.4	3.6	3.6
303	B7	53.0	53.0	6.9	6.9	36.9	36.9	.9	.9
303	C1	34.6	58.2	62.0	41.3	240.8	164.3	10.5	5.4
303	C2	34.6	58.2	55.1	48.2	215.3	189.8	9.4	6.3
303	C3	31.1	53.0	49.8	7.1	168.9	14.1	8.9	1.0
303	C4	34.6	58.2	62.0	48.2	240.8	189.8	10.5	6.3
303	C5	57.2	57.2	34.4	34.4	138.8	138.8	4.6	4.6
303	C6	34.6	58.2	48.4	42.1	265.3	221.0	8.2	5.5
303	C7	34.4	57.2	34.4	27.6	138.8	113.4	5.9	3.6
303	D1	34.4	57.2	41.3	34.4	164.3	138.8	7.0	4.6
303	D2	31.1	53.0	27.6	20.7	113.4	87.9	4.9	2.8
303	D3	57.2	57.2	34.4	34.4	138.8	138.8	4.6	4.6
303	D4	34.6	58.2	55.3	53.2	243.2	221.0	9.4	7.0
303	D5	58.2	58.2	41.3	41.3	164.3	164.3	5.4	5.4
303	D6	34.6	58.2	62.0	48.2	240.8	189.8	10.5	6.3
303	D7	34.6	58.2	54.6	41.2	221.0	154.7	9.3	5.4
303	E1	57.2	57.2	34.4	34.4	138.8	138.8	4.6	4.6
303	E2	57.2	57.2	34.4	34.4	138.8	138.8	4.6	4.6
303	E3	31.1	53.0	54.7	11.4	307.7	42.4	9.8	1.6
303	E4	31.1	53.0	55.2	17.8	247.6	70.7	9.9	2.4
303	E5	34.6	58.2	71.8	54.4	287.4	198.9	12.2	7.1
303	E6	34.6	58.2	68.7	56.0	265.3	198.9	11.7	7.3
303	E7	57.2	57.2	35.9	35.9	154.7	154.7	4.8	4.8

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
303	F1	31.1	53.0	48.3	13.8	203.4	70.7	8.7	1.9
303	F2	57.2	57.2	29.4	29.4	110.5	110.5	3.9	3.9
303	F3	34.4	57.2	38.7	31.9	176.8	132.6	6.6	4.2
303	F4	34.6	58.2	58.9	44.0	287.4	198.9	10.0	5.8
303	F5	31.1	53.0	63.5	12.8	241.4	42.4	11.4	1.8
303	F6	34.6	58.2	52.2	44.0	265.3	176.8	8.9	5.8
303	F7	31.1	53.0	27.6	20.7	113.4	87.9	4.9	2.8
303	G1	31.1	53.0	26.8	21.2	114.9	70.7	4.8	2.9
303	G2	58.2	58.2	65.0	65.0	176.8	176.8	8.5	8.5
303	G3	34.4	57.2	53.8	31.2	309.5	198.9	9.2	4.1
303	G4	34.4	57.2	48.2	34.4	189.8	138.8	8.2	4.6
303	G5	31.1	53.0	65.7	11.9	344.0	56.6	11.8	1.6
303	G6	34.6	58.2	68.9	48.2	266.3	189.8	11.7	6.3
303	G7	34.4	57.2	34.4	27.6	138.8	113.4	5.9	3.6
401	A1	59.4	59.4	34.4	34.4	138.8	138.8	4.5	4.5
401	A2	34.6	56.2	55.9	50.8	243.2	198.9	9.5	6.8
401	A3	34.4	59.4	28.3	26.7	154.7	132.6	4.8	3.5
401	A4	34.4	59.4	37.8	33.2	265.3	221.0	6.4	4.3
401	A5	34.6	56.2	62.0	48.2	240.8	189.8	10.5	6.4
401	A6	34.6	56.2	62.0	55.1	240.8	215.3	10.5	7.4
401	A7	59.4	59.4	34.4	34.4	138.8	138.8	4.5	4.5
401	B1	34.6	56.2	62.6	53.5	287.4	221.0	10.6	7.1
401	B2	31.1	54.6	46.4	19.6	223.7	113.2	8.3	2.7
401	B3	59.4	59.4	27.6	27.6	113.4	113.4	3.6	3.6
401	B4	34.4	59.4	26.2	24.0	154.7	132.6	4.5	3.1
401	B5	59.4	59.4	27.6	27.6	113.4	113.4	3.6	3.6
401	B6	59.4	59.4	34.4	34.4	138.8	138.8	4.5	4.5
401	B7	56.2	56.2	55.1	55.1	215.3	215.3	7.4	7.4

Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m^2/ha*-------		-------*No./ha*-------			
401	C1	56.2	56.2	43.8	43.8	132.6	132.6	5.8	5.8
401	C2	34.4	59.4	34.4	27.6	138.8	113.4	5.9	3.6
401	C3	34.6	56.2	55.1	48.2	215.3	189.8	9.4	6.4
401	C4	54.6	54.6	13.8	13.8	62.4	62.4	1.9	1.9
401	C5	31.1	54.6	67.3	9.1	329.8	42.4	12.1	1.2
401	C6	59.4	59.4	34.4	34.4	138.8	138.8	4.5	4.5
401	C7	34.6	56.2	73.8	67.9	287.4	243.2	12.5	9.1
401	D1	59.4	59.4	27.0	27.0	141.5	141.5	3.5	3.5
401	D2	34.6	56.2	55.1	41.3	215.3	164.3	9.4	5.5
401	D3	59.4	59.4	27.6	27.6	113.4	113.4	3.6	3.6
401	D4	34.6	56.2	68.9	48.2	266.3	189.8	11.7	6.4
401	D5	31.1	54.6	83.6	13.0	410.3	56.6	15.0	1.8
401	D6	34.6	56.2	48.2	41.3	189.8	164.3	8.2	5.5
401	D7	34.4	59.4	41.3	34.4	164.3	138.8	7.0	4.5
401	E1	56.2	56.2	62.0	62.0	240.8	240.8	8.3	8.3
401	E2	34.6	56.2	48.2	41.3	189.8	164.3	8.2	5.5
401	E3	59.4	59.4	27.6	27.6	113.4	113.4	3.6	3.6
401	E4	34.4	59.4	67.2	28.2	211.3	56.6	11.5	3.7
401	E5	34.6	56.2	72.1	57.5	221.0	176.8	12.3	7.7
401	E6	56.2	56.2	62.0	62.0	240.8	240.8	8.3	8.3
401	E7	31.1	54.6	41.9	10.1	183.0	28.3	7.5	1.4
401	F1	59.4	59.4	36.1	36.1	132.6	132.6	4.7	4.7
401	F2	56.2	56.2	48.2	48.2	189.8	189.8	6.4	6.4
401	F3	54.6	54.6	6.9	6.9	36.9	36.9	.9	.9
401	F4	31.1	54.6	68.9	11.5	307.7	42.4	12.4	1.6
401	F5	34.6	56.2	72.5	65.1	287.4	243.2	12.3	8.7
401	F6	34.6	56.2	68.9	62.0	266.3	240.8	11.7	8.3
401	F7	34.4	59.4	41.3	34.4	164.3	138.8	7.0	4.5

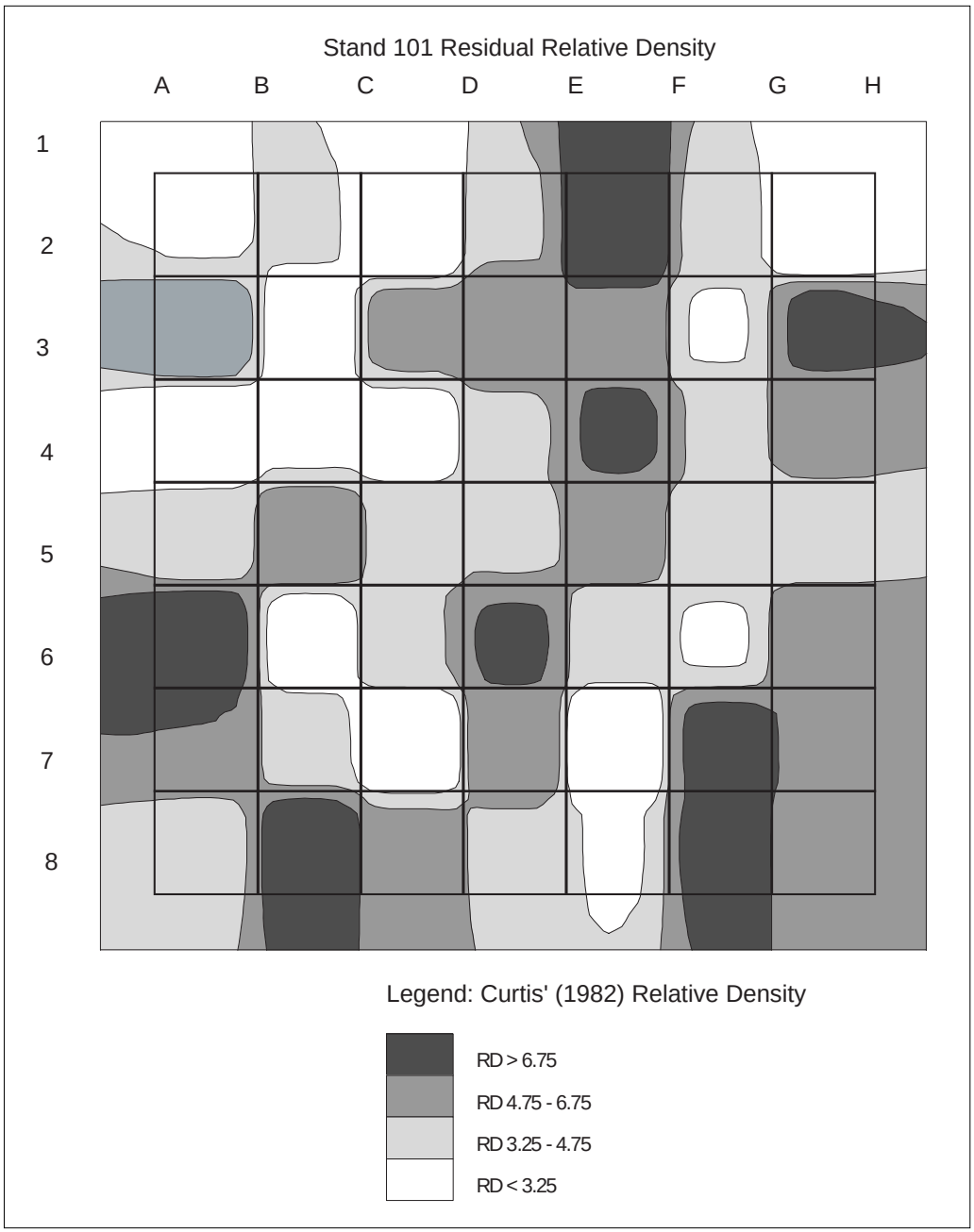
Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
401	G1	56.2	56.2	48.2	48.2	189.8	189.8	6.4	6.4
401	G2	59.4	59.4	27.6	27.6	113.4	113.4	3.6	3.6
401	G3	34.4	59.4	35.8	28.4	163.6	141.5	6.1	3.7
401	G4	34.4	59.4	41.3	34.4	164.3	138.8	7.0	4.5
401	G5	34.6	56.2	73.1	63.3	331.6	265.3	12.4	8.4
401	G6	56.2	56.2	48.2	48.2	189.8	189.8	6.4	6.4
401	G7	34.4	59.4	43.6	34.5	221.0	154.7	7.4	4.5
403	A1	72.6	72.6	48.2	48.2	189.8	189.8	5.7	5.7
403	A2	34.6	72.6	60.7	55.0	265.3	176.8	10.3	6.5
403	A3	34.6	72.6	68.9	48.2	266.3	189.8	11.7	5.7
403	A4	34.4	63.3	48.2	34.4	189.8	138.8	8.2	4.3
403	A5	63.3	63.3	27.0	27.0	117.9	117.9	3.4	3.4
403	A6	31.1	58.0	68.1	10.1	460.6	28.3	12.2	1.3
403	A7	34.6	72.6	62.0	48.2	240.8	189.8	10.5	5.7
403	B1	34.6	72.6	68.9	62.0	266.3	240.8	11.7	7.3
403	B2	72.6	72.6	41.3	41.3	164.3	164.3	4.9	4.9
403	B3	72.6	72.6	48.2	48.2	189.8	189.8	5.7	5.7
403	B4	34.6	72.6	54.2	46.8	243.2	198.9	9.2	5.5
403	B5	34.4	63.3	41.3	27.6	164.3	113.4	7.0	3.5
403	B6	34.6	72.6	75.8	62.0	291.8	240.8	12.9	7.3
403	B7	34.6	72.6	51.6	43.4	309.5	243.2	8.8	5.1
403	C1	34.6	72.6	62.0	55.1	240.8	215.3	10.5	6.5
403	C2	34.6	72.6	71.0	59.1	154.7	110.5	12.1	6.9
403	C3	34.6	72.6	68.9	41.3	266.3	164.3	11.7	4.9
403	C4	34.6	72.6	82.7	62.0	317.2	240.8	14.1	7.3
403	C5	63.3	63.3	31.1	31.1	66.3	66.3	3.9	3.9
403	C6	34.4	63.3	34.4	27.6	138.8	113.4	5.9	3.5
403	C7	34.4	63.3	42.8	30.5	198.9	132.6	7.3	3.8
403	D1	31.1	58.0	56.6	11.1	264.1	28.3	10.2	1.5

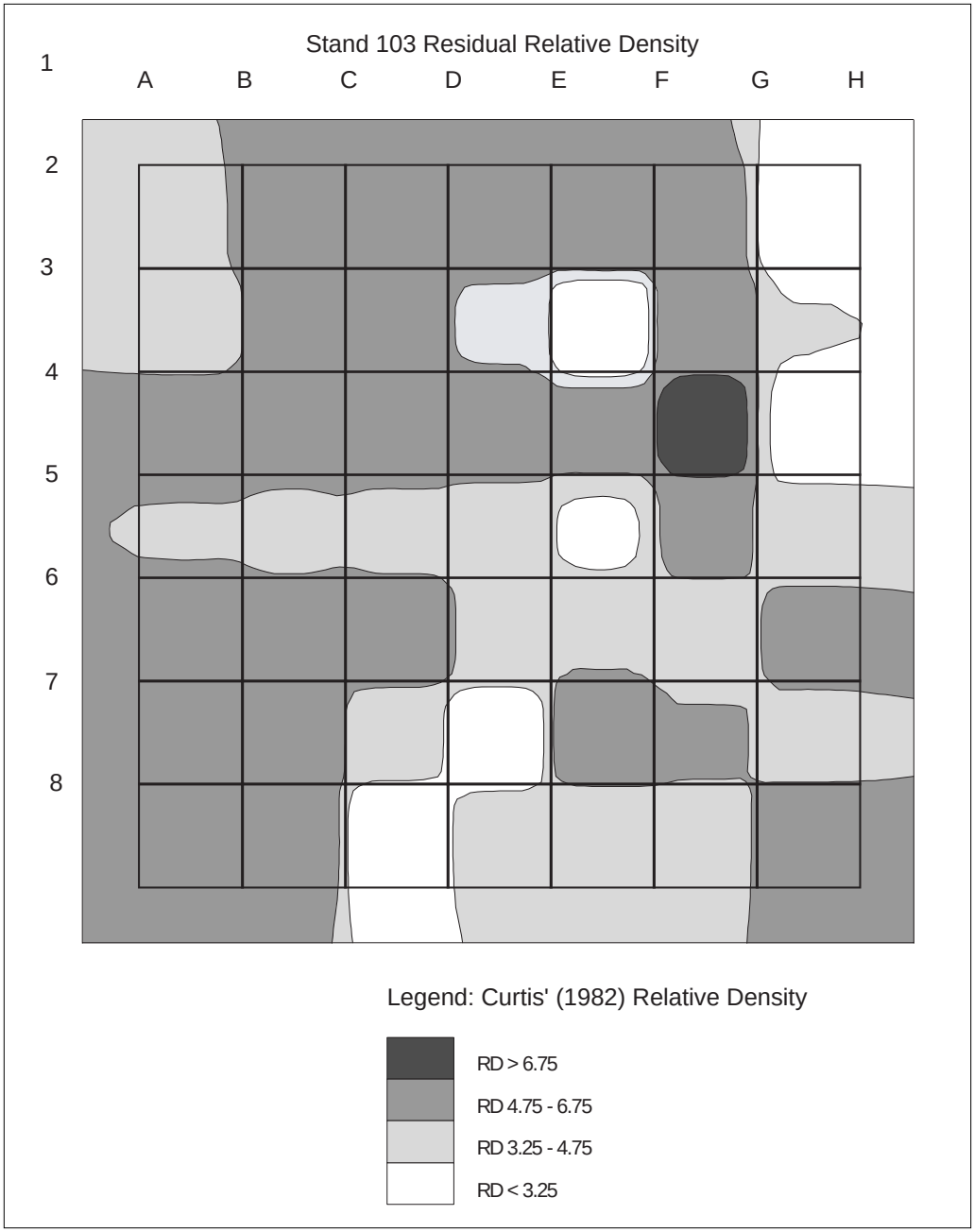
Stand	Grid cell (NW corner)	Prethin quadratic mean diameter	Residual quadratic mean diameter	Prethin basal area	Residual basal area	Prethin tree density	Residual tree density	Prethin relative density (metric)	Residual relative density (metric)
		----*Centimeters*----		-------*m²/ha*-------		-------*No./ha*-------			
403	D2	63.3	63.3	34.4	34.4	138.8	138.8	4.3	4.3
403	D3	34.6	72.6	68.9	55.1	266.3	215.3	11.7	6.5
403	D4	72.6	72.6	38.6	38.6	88.4	88.4	4.5	4.5
403	D5	34.6	72.6	48.2	41.3	189.8	164.3	8.2	4.9
403	D6	63.3	63.3	27.6	27.6	113.4	113.4	3.5	3.5
403	D7	31.1	58.0	58.0	19.3	278.2	42.4	10.4	2.5
403	E1	34.6	72.6	55.1	48.2	215.3	189.8	9.4	5.7
403	E2	31.1	58.0	28.3	5.4	92.7	14.1	5.1	.7
403	E3	31.1	58.0	67.0	17.1	160.3	42.4	12.0	2.3
403	E4	34.6	72.6	72.7	52.0	221.0	154.7	12.4	6.1
403	E5	63.3	63.3	34.4	34.4	138.8	138.8	4.3	4.3
403	E6	31.1	58.0	65.8	13.4	264.1	28.3	11.8	1.8
403	E7	34.4	63.3	47.8	31.7	309.5	176.8	8.2	4.0
403	F1	31.1	58.0	27.6	20.7	113.4	87.9	4.9	2.7
403	F2	72.6	72.6	48.2	48.2	189.8	189.8	5.7	5.7
403	F3	31.1	58.0	39.8	21.1	121.0	42.4	7.1	2.8
403	F4	34.6	72.6	47.6	46.0	176.8	154.7	8.1	5.4
403	F5	34.6	72.6	55.1	41.3	215.3	164.3	9.4	4.9
403	F6	34.6	72.6	62.0	55.1	240.8	215.3	10.5	6.5
403	F7	31.1	58.0	31.2	10.5	185.5	28.3	5.6	1.4
403	G1	34.6	72.6	48.9	41.1	198.9	154.7	8.3	4.8
403	G2	72.6	72.6	41.3	41.3	164.3	164.3	4.9	4.9
403	G3	72.6	72.6	41.3	41.3	164.3	164.3	4.9	4.9
403	G4	34.6	72.6	75.8	55.1	291.8	215.3	12.9	6.5
403	G5	34.6	72.6	48.2	41.3	189.8	164.3	8.2	4.9
403	G6	34.6	72.6	55.1	48.2	215.3	189.8	9.4	5.7
403	G7	34.4	63.3	27.4	23.1	132.6	88.4	4.7	2.9

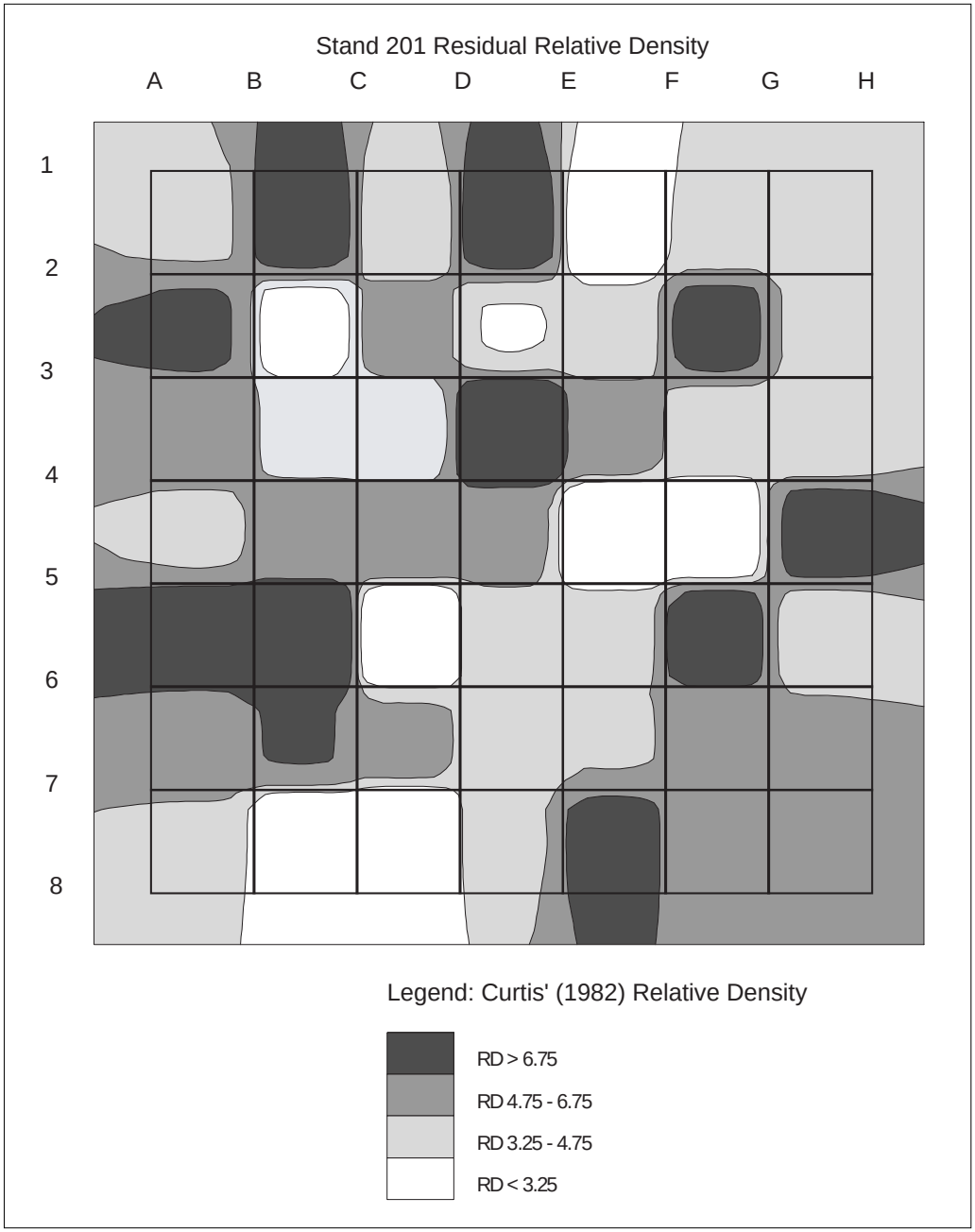
[1] First, 1 of 3 treatment codes was assigned to each cell based on its residual basal area: root rot thin (residual basal area < 22.5 m^2/ha); heavy thin (22.5-37.5 m^2/ha); or light thin (> 37.5 m^2/ha). Next, dbh was measured in a subsample of cells in each stand and a mean quadratic mean diameter was calculated by treatment code for each stand.

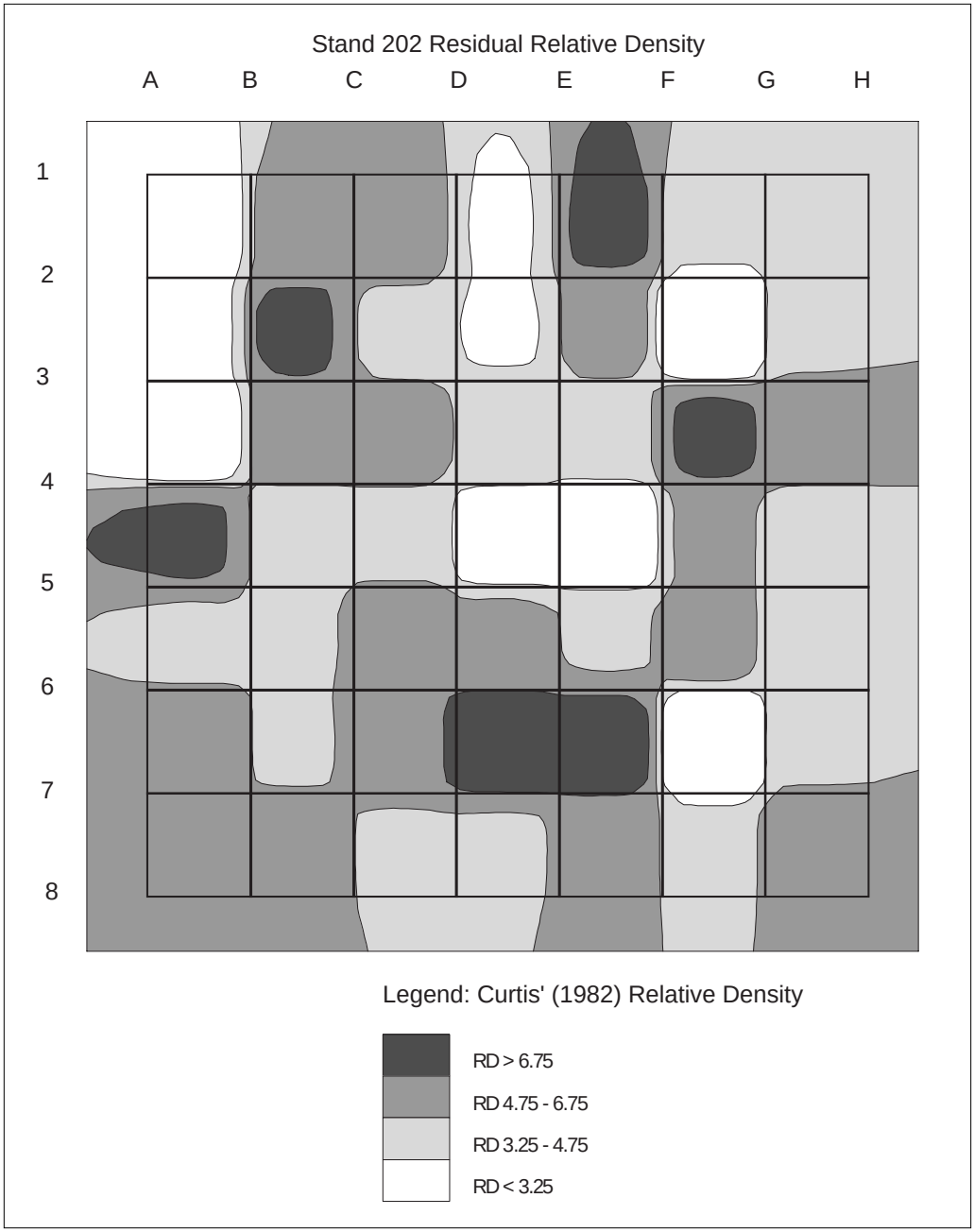
Appendix 5

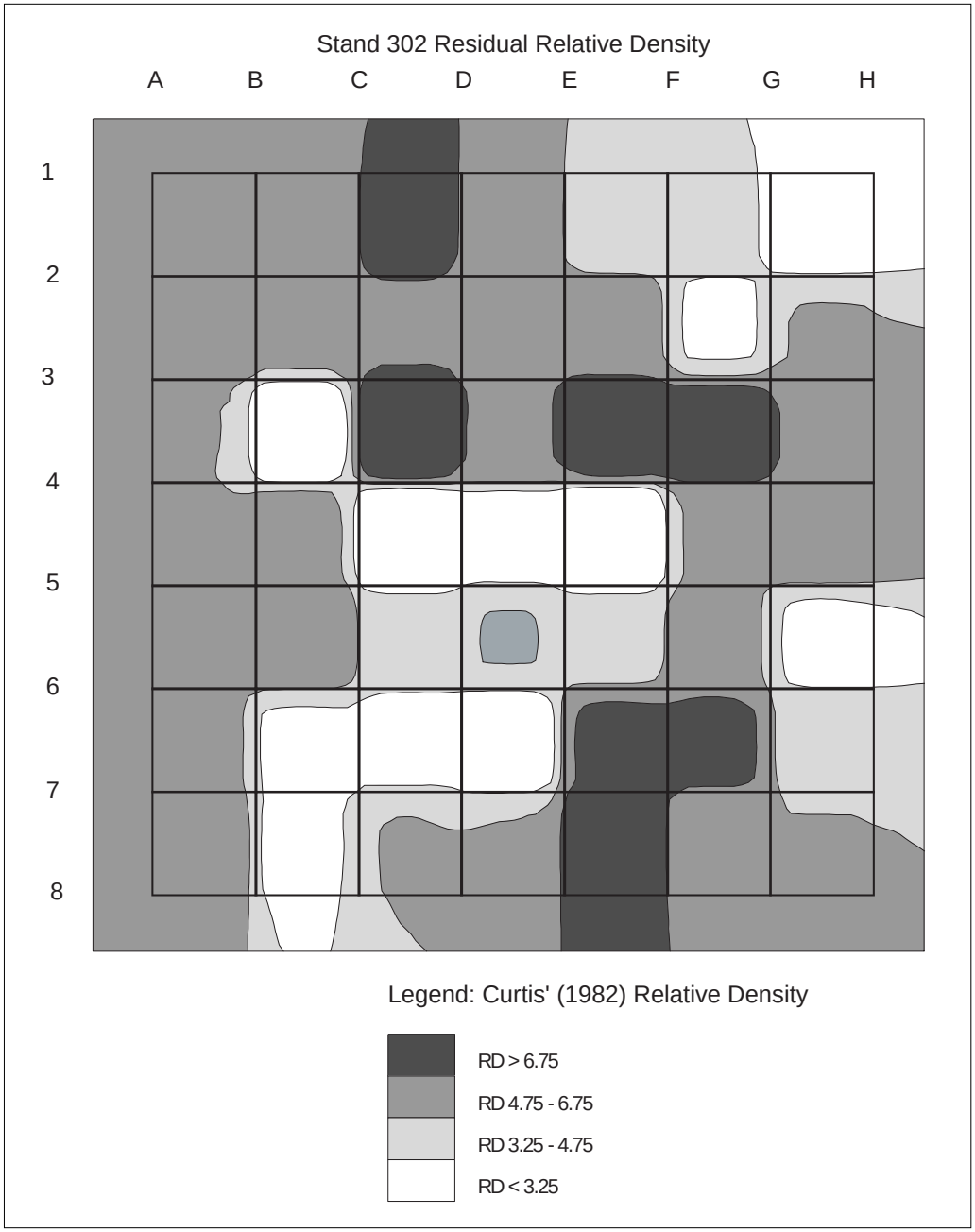
Postthinning Stand Maps

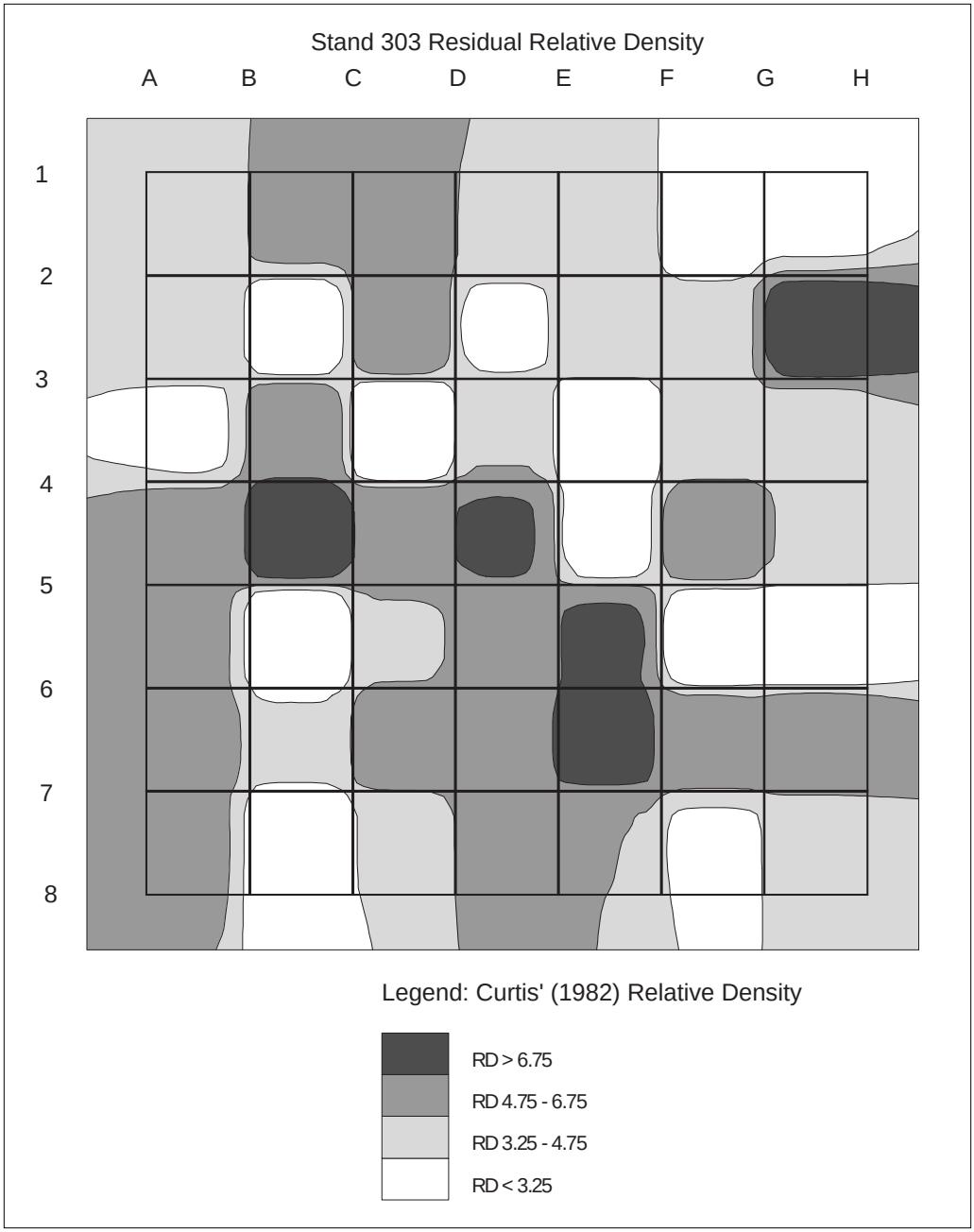


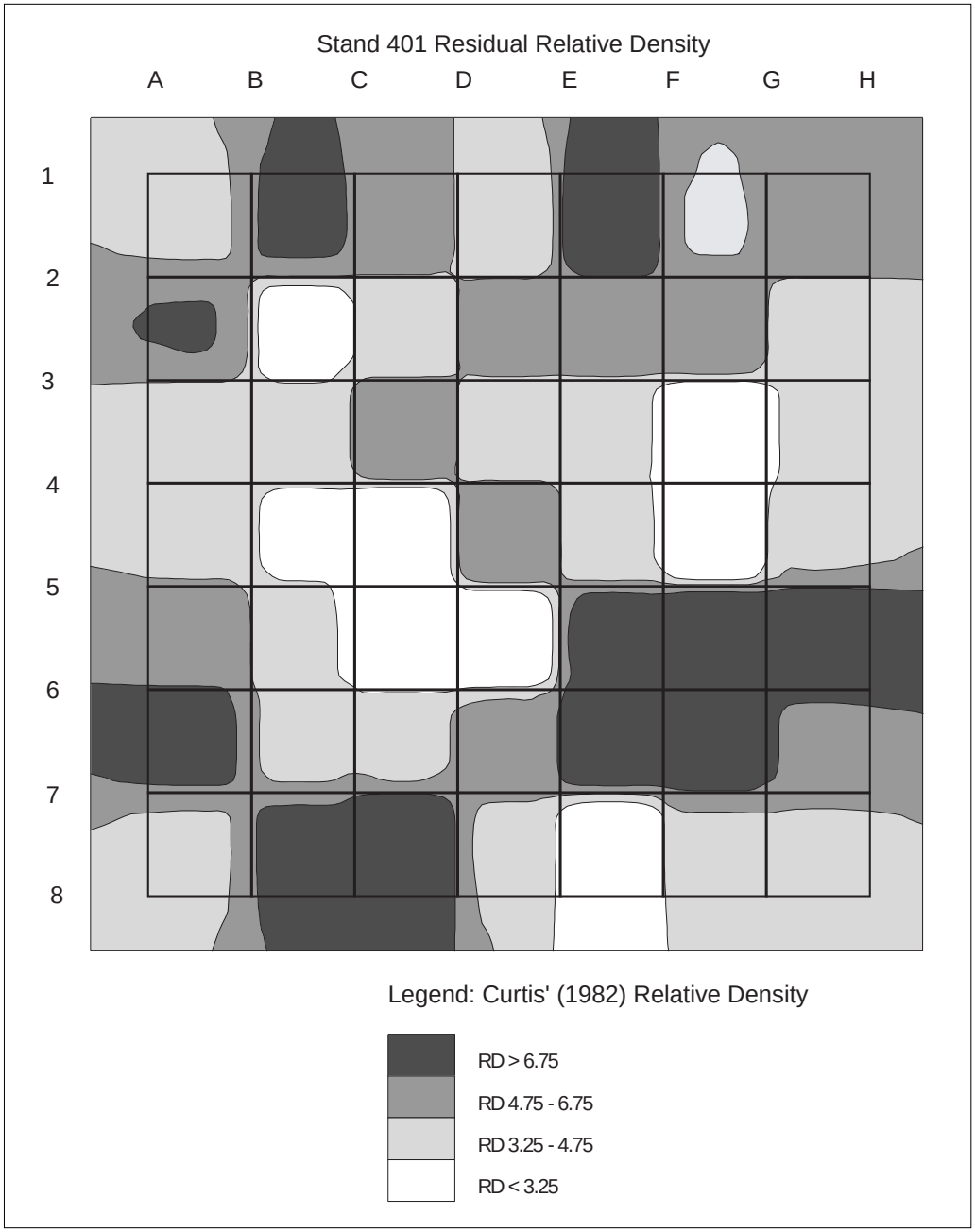

Stand 103 Residual Relative Density
1
A
B
C
D
E
F
G
H
2
3
4
5
6
7
8
Legend: Curtis' (1982) Relative Density
RD > 6.75
RD 4.75 - 6.75
RD 3.25 - 4.75
RD < 3.25

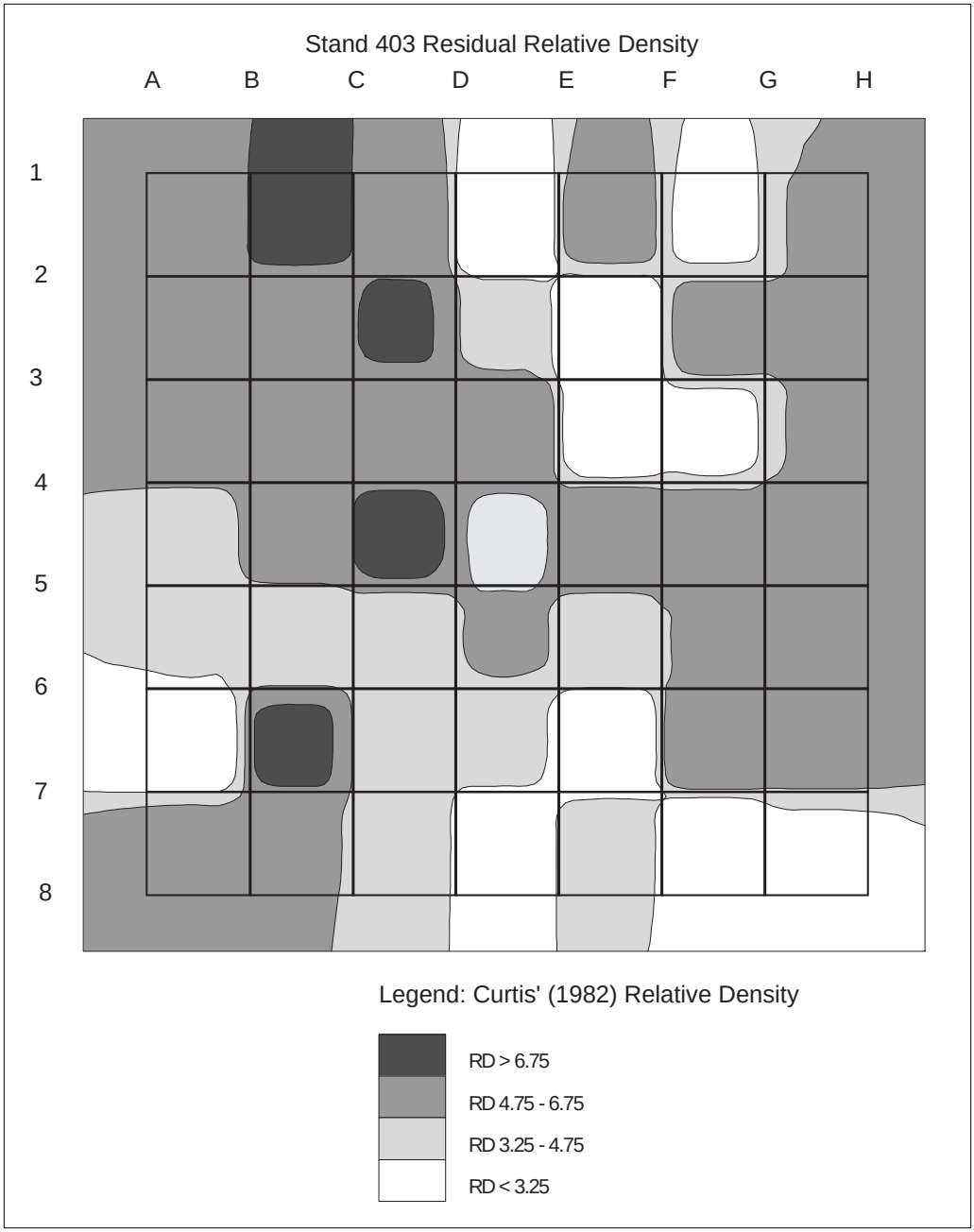

Stand 201 Residual Relative Density
A
B
C
D
E
F
G
H
1
2
3
4
5
6
7
8
Legend: Curtis' (1982) Relative Density
RD > 6.75
RD 4.75 - 6.75
RD 3.25 - 4.75
RD < 3.25


Stand 202 Residual Relative Density
A
B
C
D
E
F
G
H
1
2
3
4
5
6
7
8
Legend: Curtis' (1982) Relative Density
RD > 6.75
RD 4.75 - 6.75
RD 3.25 - 4.75
RD < 3.25


Stand 302 Residual Relative Density
A
B
C
D
E
F
G
H
1
2
3
4
5
6
7
8
Legend: Curtis' (1982) Relative Density
RD > 6.75
RD 4.75 - 6.75
RD 3.25 - 4.75
RD < 3.25


Stand 303 Residual Relative Density
A
B
C
D
E
F
G
H
1
2
3
4
5
6
7
8
Legend: Curtis' (1982) Relative Density
RD > 6.75
RD 4.75 - 6.75
RD 3.25 - 4.75
RD < 3.25


Stand 401 Residual Relative Density
A
B
C
D
E
F
G
H
1
2
3
4
5
6
7
8
Legend: Curtis' (1982) Relative Density
RD > 6.75
RD 4.75 - 6.75
RD 3.25 - 4.75
RD < 3.25


Stand 403 Residual Relative Density
A
B
C
D
E
F
G
H
1
2
3
4
5
6
7
8
Legend: Curtis' (1982) Relative Density
RD > 6.75
RD 4.75 - 6.75
RD 3.25 - 4.75
RD < 3.25

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Carey, Andrew B.; Thysell, David R.; Brodie, Angus W. 1999. The Forest Ecosystem Study: background, rationale, implementation, baseline conditions, and silvicultural assessment. Gen. Tech. Rep. PNW-GTR-457. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station. 129 p.

The Forest Ecosystem Study (FES) came about as an early response to the need for innovative silvicultural methods designed to stimulate development of late-successional attributes in managed forests—a need ensuing from the exceptional and long-standing controversies over old-growth forests and endangered species concerns in the Pacific Northwest. In 1991, scientists with the FES applied experimental, variable-density thinning to even-aged Douglas-fir (*Pseudotsuga menziesii* (Mirbel) Franco) forests on the Fort Lewis Military Reservation in western Washington after having first accumulated extensive baseline data on arboreal rodents, small mammals, trees and other vascular plants, and fungi. We present study background, rationale, baseline conditions, and selected preliminary responses, as well as a silvicultural assessment of the variable-density thinning.

Keywords: Variable-density thinning, Pacific Northwest, Douglas-fir, biodiversity, northern flying squirrel, truffle, Forest Ecosystem Study, experimental silviculture.

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