Chapter 8: Coldwater Fish in Wadeable Streams

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8.1 INTRODUCTION

8.1.1 Definition of Water Body

Small, wadeable streams comprise the majority of habitats available to fishes in fluvial networks. Wadeable streams are generally less than 1 m deep, and fish can be sampled without the use of water craft. Cold waters are defined as having mean 7-d summer maximum water temperatures of less than 20°C and providing habitat for coldwater fishes.

8.1.2 Targeted Fishes

Fish fauna of small coldwater streams of North America typically include trout and salmon, sculpins, minnow, sticklebacks, suckers, or lampreys (Hocutt and Wiley 1986). Standard sampling protocols provided herein apply primarily for trout and salmon because of their sport and commercial values (Johnson et al. 2007). As interest in nonsalmonid species grows, further development of sampling methods for a broader diversity of species is expected. However, many of the following methods can be applied effectively to sampling fish other than trouts and salmon.

8.1.3 Standard Gears

The broad taxonomic diversity and size-specific morphological, physiological, and behavioral characteristics within and among salmonid species have led to a diverse array of sampling methods for cold, wadeable streams. While some level of standardization is possible, each method provides complementary information about different species and life stages. The primary elements of standardized sampling programs are based on three of the most common methods for quantitatively sampling fishes in wadeable streams: (1) electrofishing, (2) underwater observation by snorkeling, and (3) nest or redd counts. Methods described herein are intended to provide more specific directions than are found in general references (e.g., Murphy and Willis 1996), and as a complement to a recent volume providing detailed information on salmonid field protocols (Johnson et al. 2007). Our goal is to assist biologists who may be inexperienced in sampling small, wadeable streams, who may be beginning new sampling programs, or who may need information to fine-tune ongoing sampling programs.

8.1.3.1 Electrofishing

For cold, wadeable streams, electrofishing is effectively accomplished with a pulsed, DC backpack electrofisher (see Chapter 4). Alternating current is effective for capturing fish but is more likely to cause injury than DC (Snyder 2003). Unpulsed DC reduces the chance of fish injury but is often less effective in capturing fish (Dolan and Miranda 2003; Snyder 2003) and may not be feasible because of power requirements. In addition to the type of current, the effectiveness of electrofishing varies with size, physiology, coloration, behavior, and habitat use of target organisms, and protocols can be adjusted to account for those differences. Many additional aspects of electrofishing are covered in other chapters of this volume (see also Temple and Pearsons 2007).

8.1.3.2 Underwater observation by snorkeling

Snorkeling offers many advantages over other methods of sampling fishes, including conceptual simplicity, versatility, cost effectiveness, nonintrusiveness, and ability to obtain in situ behavioral information (O'Neil 2007; Thurow et al., in press). The relatively modest personnel and gear requirements for snorkeling can reduce costs and are well suited for sampling remote locations (Thurow 1994). Snorkeling methods are also useful for avoiding the risks of more potentially destructive methods when sampling rare species or species of special concern. Underwater observation may also be appropriate when effectiveness of other sampling methods, such as electrofishing, seining, or trapping, are compromised by environmental conditions such as extreme conductivity (low or high), habitat complexity, or water depths greater than 1 m, where effectiveness of backpack electrofishing is limited (Dolloff et al. 1996; Thurow et al., in press). It is easily adapted to a variety of applications, including broad-scale inventories of aquatic organism distribution and abundance (Hankin and Reeves 1988), highly specialized observations of behavior (Noakes and Baylis 1990), evaluations of habitat use (Fausch and White 1981), estimates of fish length structure (Griffith 1981), and assessments of gear performance (Thurow et al. 2006).

Despite compelling reasons for choosing snorkeling, investigators must first clearly define their survey goals, objectively assess the limitations of snorkeling, and determine whether the method is appropriate and feasible (Thurow et al., in press). Even when conditions are ideal (see below), snorkeling alone may be insufficient to address the objectives of a given sampling program. Unlike electrofishing, snorkeling typically does not allow fish capture; therefore, snorkeling alone may not be sufficient to address important ecological questions. For example, it may be infeasible to accurately determine length, sex, age, or reproductive status or to identify small individuals or cryptic species. Electrofishing may also be more advantageous in small, shallow stream segments that physically preclude snorkeling.

8.1.3.3 Nest or redd counts

Redd counts are a common method for monitoring reproduction of stream-dwelling trout and salmon (Gallagher et al. 2007). A basic understanding of redd construction is prerequisite to accurate identification and enumeration of redds. Only female trout and salmon are believed to participate in redd construction (Esteve 2005). After selecting a suitable spawning location, the female begins fanning the substrate, typically with a rapid sideways motion, creating a depression or pit where the eggs are deposited and fertilized by one or more males. The female then moves upstream and begins excavation of a new pit while simultaneously covering the fertilized eggs with the tailspill (Figure 8.1). This process may be repeated several times, resulting in the formation of several discrete redds or a single large structure (Figure 8.1; Esteve 2005). Redds are most frequently constructed in stream reaches containing gravel sorted by size. The most likely sites within wadeable streams include transitions

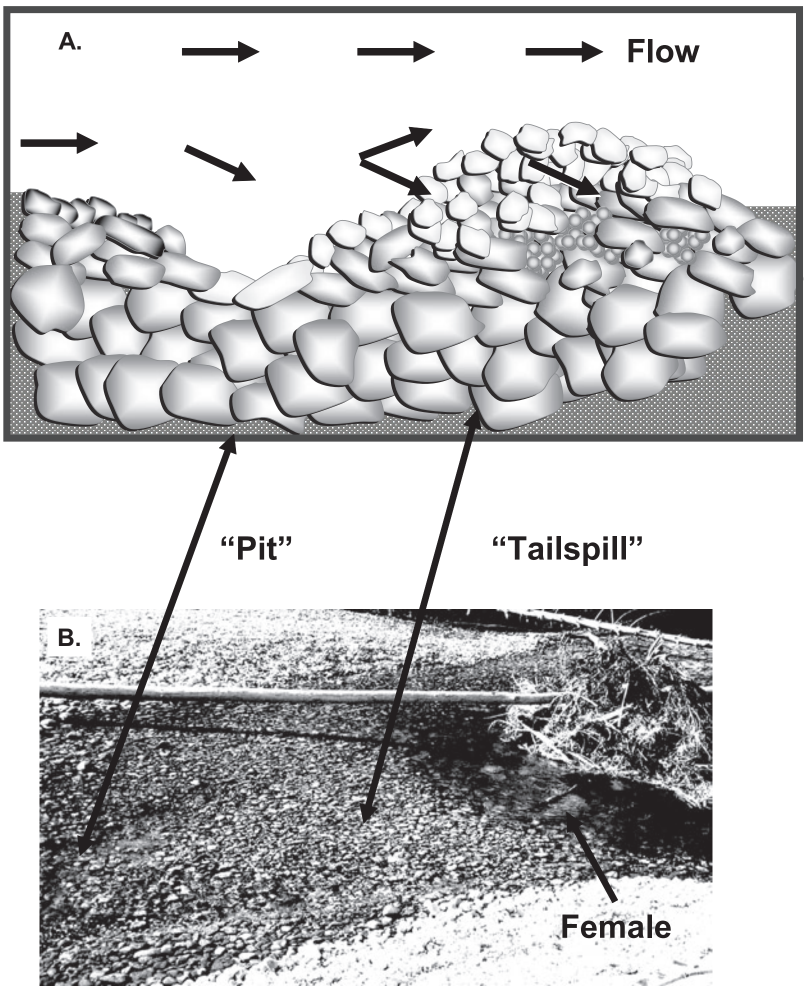

Figure 8.1 A. Depiction of a longitudinal profile of a redd within a stream (eggs shown as small spheres within the "egg pocket"). B. Photograph of a Chinook salmon redd in Nason Creek, Washington State (Wenatchee River basin). The excavated "pit" is visible as an obvious depression in the substrate, whereas the egg pocket is visible as an elevated mass of substrate deposited downstream of the pit. Arrows from A to B show locations of the redd pit and egg pocket in the photograph. The female attending this redd is visible downstream in the scour pool just upstream of the root wad and indicated by an arrow.

between pools and riffles, lower gradient channels, tributary and debris flow confluences, and sites adjacent to instream objects such as wood and boulders.

Although the "clean" surface of a recently constructed redd typically contrasts with the dull appearance of adjacent undisturbed substrate, which often is covered by periphyton or fine organic material, contrast tends to decrease over time. Contrasts in the appearance of streambed surfaces also can result from causes other than redd construction (e.g., stream hydraulics, disturbance by other animals). Other ways to recognize redds include (1) presence of adult trout and salmon near a suspected redd, (2) an elliptical area of disturbed gravel oriented directly into the current, (3) a three-dimensional streambed morphology with the pit and tailspill clearly visible (Figure 8.1), or (4) presence of the preceding streambed morphologies in locations where natural scour and deposition of substrate are unlikely to produce them. Redd size and substrate sizes used are influenced by adult size and life history of the species (Kondolf and Wolman 1993; Devries 1997) and should be considered when attempting to identify redds. For example, salmon redds can encompass more than 5 m^2, whereas redds constructed by resident trout often cover 1 m^2 or less.

Given the wide range of conditions under which redds are constructed, the utility of redd counts can be highly variable. In some situations, redds may be counted accurately (Muhlfeld et al. 2006), but in others, accuracy may be very low (Dunham et al. 2001). Even if redds are counted without error, there are many biological factors that influence how well counts actually reflect population abundance. For example, there is considerable variation within species both in how redds are constructed (Esteve 2005) and in the relationship between the number of redds and spawning population size (Dunham et al. 2001; Al-Chokhachy et al. 2005; Esteve 2005). In some cases, eggs could be broadcast directly into the substrate with no redd construction. To our knowledge, this behavior is not well documented for stream-living salmon and trout, but field observations (e.g., lack of observable redds in streams with high juvenile recruitment) indicate that this is a distinct possibility. Furthermore, individual females will sometimes construct multiple redds or excavate "test" depressions in which eggs are not deposited. Sex ratios will also vary among locations or times, thus influencing the association between redds and adults because only females construct redds. In cases where redds are closely clustered or superimposed, it can be difficult to discern individual redds. All of these potential influences should be factored into decisions about whether or not redd counts are useful for monitoring populations.

8.2 ELECTROFISHING

8.2.1 Targeted Species

Species targeted by electrofishing include all taxa that occur in cold, wadeable streams. We focus here on species that are more likely to remain in the water column or on the stream bottom during sampling, and less coverage is given to life stages and species that characteristically dwell in substrates, such as the larvae of lamprey (see Steeves et al. 2003; Moser 2007).

8.2.2 Specifications

Pulsed DC backpack electrofishers equipped with a circular probe anode and a rat-tail cathode are the standard gear for wadeable, coldwater streams (Table A.7). In theory, water conductivity is the most important factor influencing electrofishing efficiency and dictates appropriate settings. In practice, electrofishing efficiency is influenced by many factors such as fish species size and behavior, water depth, visibility, and temperature, and cover complexity. Accordingly, a mix of theory and experience is usually best in determining appropriate electrofishing settings (Cowx and Lamarque 1990; Reynolds 1996), which highlights the importance of consulting with local, knowledgeable fishery biologists concerning appropriate procedures before heading into the field (Reynolds 1996).

The principal backpack electrofisher settings for pulsed DC include voltage, frequency (Hz), and pulse width or duration (sometimes expressed as duty cycle). High frequency, particularly greater than 30 Hz, is the primary factor associated with increased injuries (Snyder 2003). The effects of pulse width and shape are less clear. Voltage may need to be increased with decreasing conductivity or increasing depth. In general, stress and mortality (and capture efficiency) are a function of the intensity and duration of the electrical field (Snyder 2003). As a rule, to reduce fish injury, begin with lower electrofisher settings and gradually increase them in a sequence of voltage, duty cycle, and frequency, if necessary, until they are effective for capturing fish. We recommend testing the settings in a stream section that is different from the sample reaches but has similar characteristics. Testing should not occur in sites used for formal sampling or influence them in any way.

To begin test sampling, we recommend beginning with a 30-Hz DC pulse at around 12% duty cycle (4 ms) and 220–280 V (Reynolds 1996). If these settings are ineffective, first increase voltage incrementally at 100-V intervals up to 1,100 V. If this is still ineffective, decrease voltage to 300 V, increase the frequency 10–15 Hz, and repeat the process up to a maximum of 60 Hz (Snyder 2003). In streams with extremely low conductivities (less than 100 μS/cm), salt can be added upstream of the site to artificially enhance water conductivity (Zalewski and Cowx 1990). Livestock supply stores sell salt blocks that can be used for this purpose.

During electrofishing, captured fish should be examined for signs of injury (Snyder 2003), such as bent backs, dark bruises or bands along the body (i.e., "brands"), and hemorrhaging of the gills. Not all injured fish exhibit obvious signs of injury (Snyder 2003). In cases where injuries to individuals are a major concern (e.g., rare or endangered species), live wells or instream cages containing recently shocked fish should be monitored to ensure that captured fish quickly regain equilibrium and swim normally. To decrease the probability of injuries and time to recovery, maintain settings at the lowest level that still effectively capture fish.

8.2.3 Operation and Deployment

A closed population is often a primary assumption of fish abundance estimators (White et al. 1982). Disturbance from sampling can bring about bias in estimators by causing fish to move out of the sampling area unless a movement barrier prevents escape (Peterson et al. 2005). Accordingly, block nets should be set at both upstream and downstream site boundaries. We recommend seines with 5–7 mm bar mesh, "lead" bottom lines, and unweighted top lines as standard. Avoid locating nets where local conditions can cause nets to fail (e.g., swift currents). Secure the ends of all lines to each bank and weight the lead line to the stream bottom with rocks or sandbags. The top of the net should extend at least 30 cm above the water surface to minimize the possibility of fish jumping out of the sample site. Avoid placing nets in locations where strong currents may cause fish (especially small individuals) to become impinged. Maintaining block nets for long periods (>1 d) usually requires repeat visits to remove debris that may clog nets and cause them to breach. During fish sampling, the downstream net should be inspected for stunned fish and cleaned of debris.

For narrow (<5 m wide) streams, standard electrofishing is accomplished with a minimum of two nets (fisher and dip netter), but increase as needed to capture a majority of stunned fishes. All personnel should wear polarized eyeglasses to aid visibility and serve as protection during sampling. Other necessary personal protective equipment includes waterproof waders, sturdy wading boots with appropriate traction (e.g., sticky rubber soles for slippery rocks, studs for traction on large wood), rubber lineman's gloves, and a hard hat in situations where falling limbs or other overhead hazards are present (e.g., in recently burned forests). We refer readers to Reynolds (1996) for additional imperative safety considerations.

Sampling begins after the electrofisher operator checks with other crew members to ensure that they are ready to begin. To sample fish, the operator depresses the activation switch and sweeps the anode across the stream channel eventually exposing all areas of the stream to electricity. Fish can often be found in areas that defy preconceived notions of suitable habitat, so all available habitats should be sampled to avoid bias. Operators should alternate application of current with brief periods of no current to avoid pushing fish ahead of the electric field. As fish are immobilized, the operator may release the anode switch to minimize potential injury to fish. Netters should be clearly aware at all times, whether the electrofisher is on or off. The primary netter closely follows the electrofisher and actively captures stunned fish near the anode. Depending on habitat structure and stream size, optimal dip-net size varies. Smaller nets are usually more suited to smaller streams and tighter cover. The secondary netter typically follows the primary netter and carries a wider D-shaped or rectangular net that may be placed on the bottom in a high velocity area to capture stunned fish drifting downstream. Many of these drifting fish are not visible; therefore, frequent inspection of the dip net by the secondary netter or an additional netter stationed at the downstream block net is important. The secondary netter also carries a bucket for temporarily holding captured fish.

A single pass in the sampling site typically consists of electrofishing by moving upstream. Zigzagging back and forth is often necessary to sample all accessible areas of the channel, with particular emphasis on areas of complex structure (e.g., cover), including accumulations of woody debris or vegetation, turbulence, maximum depth, undercut banks, or large boulders. By depressing the switch after inserting the anode into complex cover, operators often can draw fish toward the anode, thereby taking advantage of galvanotactic response associated with pulsed DC. Many investigators sample to the upstream block net, reverse direction, and continue sampling to the downstream end of the site in a single pass. Sampling downstream can be advantageous in streams with higher velocity as stunned fish may be captured in the downstream block net. In these cases, the downstream block net should be monitored during sampling so that impinged fish can be removed immediately and included as part of the catch in each pass. The choice of a single upstream or an up/downstream circuit will be dictated by local conditions. Wider streams (>5 m wide) may require multiple electrofishers conducting a single pass with operators in a line perpendicular to the flow, followed by enough dip netters to cover the width of the stream. Operators work together to intercept fish between electrodes and along the edge of the stream. Good communication between electrofisher operators and netters is essential for safety and sampling efficiency.

To reduce the chances of fish injury, crews should move continuously and deliberately upstream. Quick capture and transfer to live wells will avoid subjecting fish to undue stress. Netters should frequently transfer fish to live wells and use freshwater in holding buckets. Onshore live wells may be equipped with battery-powered aerators, and instream containers typically have small mesh or drilled holes (e.g., plastic buckets) to allow exchange of fresh, oxygenated water. Observers should carefully monitor fishes held in live wells for signs of stress (e.g., surface breathing), avoid potential for predation (e.g., holding large fish with small fish), and keep instream live wells away from the electrofishing activity so fish are not re-shocked.

Electrofishing sampling efficiency, the proportion of true numbers of fish captured (Snyder 2003), is influenced by a host of site-specific features, including water conductivity, habitat size and complexity, and fish species and size (e.g., Steeves et al. 2003; Peterson et al. 2004; Rosenberger and Dunham 2005). Consequently, implementation of standardized electrofishing protocols and uniform sampling effort is not sufficient to ensure that data will be comparable among sites. We recommend establishing a baseline or true number of individuals in multiple sites and sampling those sites to determine sampling efficiencies. These estimates of efficiency can then be used to adjust catch data from sites where the true number of fish is unknown (Chapter 12). We anticipate that the most common method of determining sampling efficiencies will involve establishment of a valid baseline using marked fish and comparison to a single-pass catch or removal estimate (Peterson et al. 2004; Rosenberger and Dunham 2005).

To establish a baseline number of fish, crews select one or more sites with characteristics that are similar to sample sites. They next enclose the upstream and downstream ends with block nets, as described above, and complete a single pass with an electrofisher. All captured individuals of the target species are then anesthetized (Kelsch and Shields 1996), measured, and marked (Guy et al. 1996). In general, marked fish should exceed 60 mm total length. Smaller individuals are inefficiently captured by electrofishing and can pass through dip or block nets. However, crews should collect and transfer undersized or non-target fish to live wells to minimize injury and stress. After recovery, uninjured marked individuals are then redistributed throughout the length of the site. A minimum number of marked individuals is required for reliable population estimates (see White et al. 1982 for guidelines), and mark–recapture may, therefore, not work for sites with low fish numbers (e.g., Rosenberger and Dunham 2005). After returning the fish to the site, we recommend a recovery period of 12–24 h before resampling, keeping in mind that the longer the recovery period, the greater the likelihood that fish will escape (Peterson et al. 2004; but see Temple and Pearsons 2006). For single- or multiple-pass electrofishing, the number and lengths of recaptured (marked) fish are recorded and compared to the number of marked and released fish to estimate sampling efficiency for the site (Chapter 12).

8.2.4 Time of Sampling

Standard backpack electrofishing is conducted during the day during summer base flows, unless sampling objectives dictate otherwise. For some species (e.g., some minnows, sculpins) or conditions (e.g., temperatures < 9°C), sampling at night can be much more effective if safety concerns are satisfied. Common sense dictates avoidance of electrofishing when conditions may be particularly stressful to aquatic taxa, such as when water temperatures are very high, when dissolved oxygen levels are low, or during times or in locations where fish are spawning or eggs are incubating. Such conditions tend to be predictable and often are outlined in permits issued by regulatory agencies along with specific times when electrofishing is not allowed.

8.2.5 Computation of Abundance

Depending on study objectives, information on abundance of fish sampled by electrofishing has been calculated as the number or biomass of fish per linear (e.g., fish/m), area (e.g., fish/m^2), or volume (e.g., fish/m^3) dimension of channel (Grant et al. 1998). For the purposes of standard reporting of results, we recommend expressing standing crop as fish/100 m^2 wherever possible. However, information should be provided that allows other users to convert from one form to another if possible. For example, to allow conversions, both the numbers and sizes of fish and the dimensions of sample sites (length, width, and depth) should be reported in publications or the data archived in a readily accessible location. Increasingly, scientific journals and online databases are providing this capability.

8.3 UNDERWATER OBSERVATION BY SNORKELING

8.3.1 Target Species

Snorkeling can be readily adapted to any species of fish, and most life stages beyond age 0+ that remain in the water column or are visible on the stream bottom. Observers need to be aware that different species and life stages will have different probabilities of being detected and that detection probability is likely influenced by a variety of environmental and biological factors.

8.3.2 Specifications

Essential snorkeling equipment includes a mask and snorkel and some form of thermal protection. In water temperatures less than 20°C, typical of coldwater streams, a neutrally buoyant Lycra suit will keep a diver comfortable to about 15°C; neoprene wetsuits 2–7 mm thick are useful to about 7°C; and in colder water, a dry suit is necessary (Dolloff et al. 1996; Thurow et al., in press). These are general guidelines, and the appropriate type of suit for a given situation will vary considerably. Most dry suits have attached socks or boots and snug latex wrist and neck seals. Neoprene hoods and gloves, and fleece undergarments to wear under the dry suit, complete the thermal protection. Hand-held halogen lights are required for nighttime surveys and are also useful for inspecting crevices during daytime surveys. Data may be recorded directly by the diver or communicated to a nearby assistant, who also provides information on survey boundaries and hazards. Many divers prefer to record data on a waterproof cuff, which leaves both hands free. Cuffs may be manufactured from materials commonly available at hardware stores (see Dolloff et al. 1993). A handheld counter may be useful if large numbers of fish are encountered. Equipment to measure underwater visibility and water temperature is also essential. If estimates of fish sizes are desired, various methods are discussed by Thurow (1994) and described below.

Snorkeling can pose serious hazards if basic safety considerations are not properly addressed. Potential site hazards should be assessed and a safety checklist developed (see Thurow 1994). An initial investment in crew training will help ensure observer safety and collection of accurate information (Figure 8.2). Training should address safety, equipment, observation techniques, and data collection and recording protocols (Dolloff et al. 1993, 1996; Thurow 1994; Thurow et al., in press).

8.3.3 Operation and Deployment

Snorkeling techniques vary depending on study objectives and environments to be surveyed. Thurow et al. (in press) described four approaches for measuring fish abundance via snorkeling: direct enumeration, expansion estimates, basinwide estimates, and mark–recapture estimates.

In wadeable streams, divers typically enter the water downstream from the area to be sampled. After entering the stream, the observer pauses to acclimate and to allow fish disturbed by the initial approach to resume normal behavior. Observers moving upstream are less likely to startle fish and cause them to flee or change their behavior because most stream-dwelling fish orient into the current. Moving slowly upstream, avoiding sudden movements and minimal disturbance of the substrate allows a snorkeler to approach fish closely. When it is impractical to move upstream as a result of current or depth, observers may float downstream with the current, remaining as motionless as possible.

Figure 8.2 Snorkeling crews being trained in sampling techniques. Crew training is an important part of any standard sampling program.

In small streams, where an observer can see from bank to bank from a single point underwater, fish can be counted using the following techniques. Depending on the characteristics of the habitat, the observer proceeds in a zigzag pattern between banks, taking care to thoroughly search stream margins and all cover, such as undercut banks, substrate interstices, and woody debris. Target fish are counted and recorded by species and length-class. Where water depth, turbulence, or clarity limit the ability to see and accurately identify fish, first move up one bank and count all fish to the limits of visibility and then repeat the procedure for the opposite bank. Snorkelers should survey areas of high visibility before moving into areas with lower visibility.

Even if water clarity can allow one observer to see the entire channel width, additional observers may be needed to count concealed or less conspicuous fish. Shallow water habitats, such as riffles, typically require more observers than deeper habitats. For relatively homogeneous habitats, sampling units can be divided into lanes of equal width and observers move slowly upstream, counting all fish within an assigned lane. Lane boundaries should be discussed and agreed upon in advance of sampling. Delineation of boundaries will depend on characteristics of the site (e.g., visibility, cover) as well as abilities of individual observers. If conditions are too turbulent or complex, natural features such as a line of boulders can be used to delineate sampling lanes. In all situations where multiple observers are used, the distance between them should always be no greater than the maximum underwater visibility. Observers must start and stop at the same time, remain in their assigned lanes, and move at the same speed. Observers must be careful to avoid counting fish that move among lanes to prevent counting the same fish twice.

Several methods have been developed to estimate fish length. One method is to estimate length relative to fixed points on the substrate (i.e., features near the fish's head and tail). The diver then swims to the reference points and measures the distance with a measuring device (Cunjak and Power 1986; Baltz et al. 1987). Alternatively, a scale such as a ruler or marked dive slate may be placed within the field of view of the diver near where fish are to be observed (e.g., Steinhart et al. 2004). Divers can carry a scale to use as a reference to compare with fish length (Mueller 2003). Swenson et al. (1988) developed a calibrated bar that attaches to the diver's mask. The diver observes length on the bar and measures distance to the organism to estimate its length. Divers can also practice estimating fish length by viewing wooden dowels or fish silhouettes of known lengths underwater. Accuracy of length estimate improves with training. Marked improvements in precision of diver estimates of fish length were achieved with an underwater stereovideo system (Harvey et al. 2002).

Observers may fail to detect or incorrectly identify target organisms, count them more than once, or incorrectly estimate length (Griffith et al. 1984). Counting organisms accurately in a dense population can be difficult (Heggenes et al. 1990), and some species and sizes of fish are harder to see than others, especially those that remain near the substrate (Hillman et al. 1992) or are concealed by cover (Rodgers et al. 1992). Just as differences in fish behavior during different times of the day or year are known to affect detectability, differences in ability, training, and experience among observers will influence the data collected.

We recommend, for cases where a high level of accuracy is needed, that snorkelers attempt to validate their methods. This involves routine evaluation of potential violations of estimator assumptions and comparison of counts to less biased estimates (Chapter 12). Although snorkeling is versatile and has many advantages over other sampling methods, use of raw snorkel counts, unadjusted for the effects of these biases, will result in biased conclusions (Thurow et al. 2006). It cannot be assumed that sampling efficiencies are equal among observers or sites, even if exactly the same protocol is followed. Methods for estimating efficiency to validate assumptions of snorkeling should parallel those outlined for electrofishing described above (for additional details, see Thurow et al. 2006).

8.3.4 Time of Sampling

For snorkeling to be effective, local environmental conditions must meet minimum criteria for water clarity, water depth, light conditions, and water temperature. Underwater visibility is affected by water clarity, which is influenced by turbidity (suspended material), turbulence (so-called "bubble curtains"), and color (e.g., tannins and other dissolved materials). Water clarity can severely limit an observer's ability to complete reliable counts of fish (Thurow 1994). Water clarity must be sufficient to enable observers to see the stream bottom in the deepest habitats, identify fish by species, and detect avoidance of the observer by fish. Within most wadeable streams, a visibility of 2–3 m meets these criteria (R. F. Thurow, personal observations). Observers should routinely measure and record the visibility of a known object prior to sampling. A suitable object is a fish silhouette with distinguishing markings. Estimate visibility with a Secchi-disk like approach that averages three measurements of the maximum distance at which the marks on the silhouette are visible. Use of consistent colors and patterns are important for comparable measurements of visibility.

In addition to clarity, water depth is a basic consideration in snorkeling. The area to be sampled must have sufficient depth to enable the observers to submerge a mask (Thurow 1994). Shallower water will limit the observer's ability to view fish hiding beneath and behind obstructions in the stream channel. Observers may count fish in water that is deep enough to submerge a mask but too shallow to float the observer, provided the observer is able to crawl through the stream.

Quality of data collected by snorkeling is highly dependent on light conditions and the time of day (Spyker and Van Den Berghe 1995). Investigators usually establish protocols that specify certain hours with optimum light conditions (e.g., 1000–1700 hours for day-time sampling). Observations conducted at night or during twilight hours require handheld or fixed-position underwater lights. Disturbance or displacement can be minimized by not shining light directly on the fish, by directing lights to the underside of water surface (Contor 1989), or by using color filters (Riehle and Griffith 1993).

Water temperature influences fish behavior and may result in biased underwater counts. For example, as temperatures decline, stream-living trout and salmon typically become more cryptic, and the efficiency of counts declines at temperatures less than 9°C (Thurow 1994). Accordingly, if higher sampling efficiencies are desired, sample when water temperatures are warmer, or if sampling must be focused on colder seasons, we recommend consideration of sampling at night. Furthermore, species such as sculpins may be more active at night, regardless of water temperatures. In general, however, snorkel surveys are conducted in the daytime during low streamflows in summer.

8.3.5 Computation of Abundance

For standard reporting, snorkel data should be expressed as number of fish per 100 m^2. As with electrofishing (see above), information should be reported to allow conversion to other common measures (e.g., fish per meter) whenever possible. We further suggest that observers also record data on factors potentially influencing the efficiency of snorkel sampling (see Thurow 1994; Thurow et al., in press). This can assist greatly in the interpretation of snorkeling results (e.g., to identify situations where sampling efficiency may have been seriously biased) and provide opportunities for calibration of snorkel counts (Chapter 12).

8.4 NEST OR REDD COUNTS

8.4.1 Target Species

Many fishes construct nests by excavating stream substrates, which are often called redds. Here, we focus on counting redds of trout and salmon in coldwater streams. Other fishes construct nests in gravel, but redd counts are most consistently used for estimating abundance of salmonid spawning populations. Furthermore, redd counts for some species, such as lamprey, are not considered reliable for estimating abundance, and alternative methods have been applied more effectively (Harvey and Cowx 2003; Moser 2007). However, our recommended methods have the potential to be modified to apply to other species that move sediment or construct nests for reproduction (e.g., chub *Nocomis* spp. mounds).

8.4.2 Specifications

Most redd counts in wadeable streams are conducted from streambanks or by careful wading. One advantage of such counts compared to aerial surveys is that gear requirements and costs are relatively minimal. Basic gear for redd counts includes waders, wading/measuring staff, polarized glasses, flagging and waterproof markers for marking redd locations, and forms or devices for recording information (Dunham et al. 2001).

8.4.3 Operation and Deployment

Redd counts are most effective if observers become familiar with sampled reaches, either by working with experienced observers during redd counts or by conducting a site visit before fish begin spawning. Familiarity with local stream habitats, including natural features that could be confused with redds, can be critical. Furthermore, redds of species spawning prior to the target species or still-visible redds from prior years can be identified and eliminated as a source of bias.

Following the prespawning survey, we recommend multiple passes through sample sites over the duration of spawning. In each pass through the site, observers should number and flag redd locations, usually on an overhanging tree branch or similar site on shore. The total number of these locations by the end of the spawning season is the redd count, and at the end of the season, all flagging should be removed. The number and frequency of survey passes depend on the extent of the spawning period and how long redds are visible. More passes may be necessary if the spawning season is protracted or if conditions affecting detectability of redds (e.g., variable streamflows) are common. Multiple counts have the advantage of accounting for variation in spawning timing and allowing observers to see new redds as they are constructed.

Redd counts can be conducted by walking upstream or downstream, depending on the sun's location. It is advantageous to have the sun at your back and complete counts during peak daylight hours. In some cases, spawners near redds can more easily be seen by observers walking upstream, which may make some redds more apparent. Observers should take care to avoid disturbance of stream substrates that could obscure visibility and disturb fish. Some redds can be difficult to identify, and observers walking through spawning areas risk trampling unidentified redds. Accordingly, care should be taken to carefully inspect all locations of the stream, regardless of a preconceived notion of where fish should spawn. Redds sometimes do not have the classic morphology (e.g., Figure 8.1) and can be constructed in locations that violate conventional notions of spawning habitat.

When multiple species construct redds in similar locations and at similar times, redd counts can be more complicated. In such cases, it may be critical to conduct very frequent (e.g., 2–4 d) surveys to closely track redd accumulations and to increase the chances that adults of a given species can be identified on redds. Through this process, characteristic redd shapes and dimensions for different species can be identified to help in discriminating redds. Similarly, frequent redd counts may be needed in locations where there may be a high probability of redd superimposition due to large numbers of spawners or limited availability of spawning habitat. Superimposition occurs when portions of an existing redd are reexcavated by the spawning activities of another female, which may or may not be conspecific. In locations where spawning habitat is limited or fish populations are very large, high levels of superimposition may make it impossible to distinguish individual redds.

Redd counts can be prone to high levels of measurement error and biological limitations to how accurately they represent actual numbers of spawning adults. Accordingly, it is important to validate methods by conducting evaluations of measurement error and common causes of errors, rigorous training for novice field observers, and periodic evaluations of bias and precision of experienced redd observers (e.g., Dunham et al. 2001; Muhlfeld et al. 2006). Additional study of the relationship between numbers of spawning adults and redd counts (e.g., estimating females per redd) may also be needed if greater accuracy in estimating numbers of adults is desired.

8.4.4 Timing of Sampling

Optimal timing of sampling depends on the species in question. Most species have a spawning season that can extend over several weeks to months, which may be conditioned on seasonal variability in stream discharge, temperature, or other factors that influence adult movements and migration or suitability of conditions at spawning sites. This is a primary reason for our emphasis on conducting prespawning surveys and multiple counts, in addition to providing important insights into the timing and extent of spawning or other activities related to reproduction.

8.4.5 Computation of Abundance

The abundance and trend of redd counts in a particular system can be estimated where a complete census of spawning habitat is conducted or with an unbiased sample (e.g., Larsen et al. 2001). In some cases, linear (e.g., redds/km) or area-based densities of redds may also be calculated when evaluations of spawning habitat use are of interest. To allow comparison of populations among reaches and streams or among years, we recommend that observers report numbers of redds observed by survey date as well as the length and average wetted width, water depth, and other pertinent features of the surveyed reaches. For the purposes of standard reporting of results, we recommend expressing counts as redds per 100 m^2.

8.5 FINAL CONSIDERATIONS

Recommendations so far have been based primarily on what to do after a sample site or reach has been selected with emphasis on the importance of method validation and obtaining unbiased measurements and estimations within sampling units. Additional information on selection of sampling sites is provided because failure to consider the spatial context of the sampling program may result in biased inferences about fishes and their habitats (e.g., bias due to errors in measurements or estimates at sites versus those resulting from sampling design). Although logistical and safety considerations are of obvious importance, site selection for sampling in stream networks first and foremost depends on the question to be addressed as well as the spatial or temporal context within which inferences are to be made (e.g., Larsen et al. 2001; Fausch et al. 2002). Estimates of abundance, presence, or other characteristics of fishes within a sampling site, no matter how accurate, are of limited (e.g., site-specific) value in the absence of an appropriate question and spatial context. Samples must be distributed in space or time to represent the frame of interest. Stratified, simple random, fixed, and other types of sampling designs have all been used to estimate fish densities over large areas. Because this topic is of critical importance but outside of the scope of this chapter, we urge readers to consult with sample design experts and texts such as Thompson et al. (1998).

In addition to sampling design, selection of a basic unit to be sampled is also important. Sampling can occur in smaller quadrats (e.g., lamprey; Steeves et al. 2003; Moser 2007) or transects (e.g., benthic riffle dwellers, Ensign et al. 1995) of small fixed-stream areas for electrofishing capture or underwater observation of species that require intensive benthic sampling (compared to water column dwellers such as trout and salmon, suckers, and minnows), where sampling can occur in the water column over a greater stream length. Sampling sites may extend anywhere from several meters to kilometers of stream reach or over entire stream networks (particularly for redd counts), and sites may include a variety of habitat units, including pools, cascades, riffles, runs, and side-channel habitats (Frissell et al. 1986). The length of site to be sampled varies depending on whether the sampler wishes to estimate species abundance or richness or both (e.g., Kaufmann et al. 1999). Lengths of stream to be sampled can be fixed (e.g., 100-m lengths of stream), scaled according to stream size (e.g., multiples of stream width; Kaufmann et al. 1999), or based on habitat unit boundaries (Hankin and Reeves 1988).

In closing, the protocols outlined in this chapter are considered to be common to many, but not all, situations when sampling cold, wadeable streams. Although use of a common method is an important step in standardization, adherence to a method without validation will not produce standardized or comparable results. For example, during the authors' experiences electrofishing in cold, wadeable streams, we found that sampling efficiencies vary from about 10–100% among sites, despite our use of identical protocols and personnel (e.g., Peterson et al. 2004; Rosenberger and Dunham 2005; Thurow et al. 2006). Clearly such a range in results argues for more rigorous understanding of the causes of measurement error and the need for approaches to adjust estimates for error (Chapter 12). In the broader view, standardization should be seen as the process of providing estimates of fish population parameters accompanied by clear understanding of the precision and accuracy of measurements relative to the spatially and temporally distributed true population parameters of interest. Pleas to validate methods are met with resistance because assessments of measurement error are viewed as excessively expensive and time-consuming. The reader is reminded that the cost is small compared to the benefit of increased utility, confidence, and defensibility of conclusions drawn from validated samples. For example, it is standard practice in analysis of water quality samples to include comparisons with standards of known concentration so that analytical methods are shown to be valid. While these practices are commonly accepted in analysis of water quality and other fields (Taylor and Kuyatt 1994), the same has not always been true of fisheries data. We argue that investments in validation of sampling methods are essential to ensure trust and confidence in the ability of biologists to manage public fisheries.

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