Electrofishing: Backpack and Drift Boat

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Background and Rationale

Electrofishing is one of the most widely used methods for sampling salmonid fishes because it is relatively inexpensive and easy to carry out in a variety of conditions and has relatively low impacts to fish and other animals. Essentially electrofishing reflects the use of electricity to stun and capture fish that come within the electrical fields produced by two electrodes. The technique has been used for a variety of objectives and has generated a rich literature. This literature includes the theory and practice of electrofishing (Taylor et al. 1957; Vibert 1963; Hartley 1980; Bohlin et al. 1989; Sharber and Black 1999), the application to abundance estimation (Vincent 1971; Peterson and Cederholm 1984; Rosenberger and Dunham 2005), species richness or community structure sampling (Simonson and Lyons 1995; Reynolds et al. 2003), and estimation of the size structure of fish populations (Thurow and Schill 1996; Vokoun et al. 2001; Bonar 2002). Despite the popularity of electrofishing as a monitoring technique, recent studies have revealed that historical electrofishing practices and commonly made assumptions should be reconsidered and in some cases abandoned (Bohlin and Sundstrom 1977; Riley and Fausch 1992; Peterson et al. 2004; Rosenberger and Dunham 2005).

This paper will help fisheries biologists maximize the utility of the data produced by applying appropriate sampling designs, good planning, and optimal electrofishing techniques. In our experience, the utility of electrofishing data is frequently limited because of insufficient planning prior to field work. We have found that the most serious errors in estimates produced using electrofishing are not caused by the technique of capturing and netting fish (e.g., electrofisher settings, movement of the anode, netting); rather, errors are more likely the result of the type of estimation technique used, the validity of the assumptions, and the representativeness of the sample sites used for extrapolation. In short, it is the sampling design and analysis phases that offer the greatest potential for error reduction.

Approach and Sampling Design

We have outlined several questions that every practitioner should ask prior to conducting an electrofishing survey (Table 1); the answers can significantly improve the utility of the data. Setting specific quantitative objectives for what the practitioner hopes to achieve is the foundation upon which all other tasks must be built. Failure to articulate specific objectives hampers the proper allocation of resources and sometimes results in the production of data of limited use.

TABLE 1. — Checklist of questions to answer before field sampling is initiated

1.	Do I have a specific quantitative objective (e.g., estimating abundance, spatial distribution, species richness, size distribution, some combination of variables)?
2.	Am I interested in learning about the status or trend in one or a number of the aforementioned variables?
3.	Do I have a strategy to accomplish my objective?
4.	Is the strategy the best approach to address the objective?
5.	Can I implement the strategy?

6.	What is the magnitude of change am I interested in detecting?
7.	How soon do I need an answer?
8.	Do I know how I will analyze the data and do I have the necessary tools?
9.	Am I collecting the necessary data to improve my technique and evaluate assumptions?
10.	How precise should my estimates be?
11.	Over what area and time do I hope to apply the data?
12.	Do I know what information I will need to report and when?

The process of objective setting often involves competing values (e.g., time versus precision, status versus trends). Once objectives are set, the critical task of designing an approach to meet objectives can begin. Examples of quantitative objectives for electrofishing studies include

- detection of 20% change in harvestable (>254 cm) rainbow trout *Oncorhynchus mykiss* abundance in central Washington within 5 years;
- abundance estimates for chinook salmon *O. tshawytscha* parr in Rock Creek, Washington, in 2005 with a coefficient of variation of less than 10%; and
- distribution of bull trout *Salvelinus confluentus* abundance less than 0.001/m^2 with 90% certainty.

Designing a sampling plan involves evaluating a series of trade-offs and finding the optimal suite of benefits relative to cost; some practitioners describe the process of creating a sampling design as part art, part science, part economics, and part experience. Some of the competing factors cannot be compared in the same currency, so human judgment must play a large role. Some of the factors that should be considered are cost of sampling and analysis, precision within versus between sites, sampling bias, biological effects of sampling, human safety, public perception, and area and time of inference. A robust sampling design for an endangered species may look great on paper but may not be permitted because of the biological effect on a population. Effort expended to reduce variance within a sample site might be better spent by sampling more sites with less precision (Hankin and Reeves 1988). Reducing estimator bias may be less important than reducing variance if it is a small component of the mean square error (MSE) (Cochran 1977). If block nets cannot be practically installed and maintained because of factors such as high discharge, depth, or debris, then selection of a few long sites (which are less likely to be mischaracterized due to fish movement) may be preferable to the selection of many small ones. Sites to be sampled should be selected in ways that allow for maximum extrapolation of the data. This means selecting samples that are representative of some larger area or time period. Selecting a representative sample can be random, systematic, stratified random, or stratified systematic, and should be spatially balanced. The overarching questions that must be asked of an experimental design are (1) is it technically feasible and defensible? and (2) does it meet estimate quality and statistical power considerations? After a sampling design has been developed, it is desirable to ask other professionals to review it.

The MSE and prospective power analysis can be used to improve experimental designs by identifying the best allocation of resources to achieve the objective. These tools may reveal that the likelihood of success in achieving the stated objective with the amount of resources that are available is low. In other cases, a more optimal balance of sample size and bias reduction may be identified. Regardless, use of these tools will help improve sampling design.

The first of the tools that can be used to allocate effort is the MSE (Cochran 1977). The MSE can be calculated with the following equation:

$$\text{MSE} = \text{bias}^2 + \text{variance} \quad \text{(eq 1)}$$

Bias can be calculated as the difference between the true values and the estimated values. Variance can be calculated as the variance of means between sites or within a site. The MSE can be reduced by reducing the bias^2 and the variance. Reducing the MSE will improve the quality of the estimate. If bias is constant or varies from known factors, then it can be corrected; however, these corrections may require additional effort. Alternatively, switching to less biased methods can also be done. Variance can be reduced by increasing the number of sites sampled or by decreasing sampling errors such as data entry errors (Cummings and Masten 1994; Johnson et al. 2007). We recommend the following actions given different amounts of bias and variance (see Table 2). Options provided in Table 3 can be used to reduce bias and variance. Cochran suggested a working rule to identify when bias has a significant effect on the accuracy of an estimate: "The effect of bias on the accuracy of an estimate is negligible if the bias is less than one-tenth of the standard deviation of the estimate." (Cochran 1977)

TABLE 2. — General recommendations to reduce the mean square error if operating with a fixed budget

	High variance	**Low variance**
High bias^2	Reduce bias or decrease variance, whichever is less expensive	Reduce bias using most cost-effective means
Low bias^2	Decrease variance using most cost-effective means	No changes necessary

TABLE 3. — Options to reduce bias and variance

Methods to reduce bias	Use less biased estimator (e.g., choose mark–recapture over multiple removal)
	Correct known bias (e.g., calibrate with unbiased estimate)
Methods to reduce variance	Increase sample size
	Use more precise methods (e.g., more effort into increasing recaptures)
	Reduce human error (e.g., poor netting, transcription error, data entry error)
	Maintain sampling consistency (e.g., sample same sites at the same time with the same equipment and people) (for trend monitoring)

Prospective power analysis is a very useful way to evaluate whether a sampling program will be able to achieve the necessary level of rigor to meet objectives. It is also useful in helping determine the number of samples and levels of precision that are necessary to achieve objectives (Green 1989). Frequently, plans for monitoring fish populations are designed with insufficient statistical power (Peterman and Bradford 1987; Peterman 1990; Ham and Pearsons 2000). This can result in failure to reject a false null hypothesis (e.g., a change or difference is undetected), which can be very serious when impact detection is critical to decision making (Peterman 1990; Ham and Pearsons 2000). It is often difficult to detect changes in salmonid population abundance of less than 20% in 5 years because of the high interannual variation (Ham and Pearsons 2000). Other parameters such as size at age and distribution may be less variable, making the detection of slight differences more likely. Power can be improved by increasing sample size and by reducing the amount of unexplained variation. Reducing the amount of unexplained variation might be done by reducing sampling error or by explaining variation with a model (Ham and Pearsons 2000). Methods to calculate statistical power can be found in Zar (1999) and also in commercially available statistical packages for computers.

After the initial sampling design is completed, another set of questions can be asked to further refine the design (Table 4). After the designers feel confident that they have answered the questions and addressed the issues, then the necessary permits, approvals, personnel, and equipment can be obtained and the fieldwork can begin.

TABLE 4. — Questions that should be asked when designing an electrofishing study

Can I get the necessary permits and site access to carry out the work?
What sampling technique (e.g., backpack electrofishing, drift boat electrofishing) is appropriate?
Can I implement the techniques (e.g., flow, depth, turbidity, water temperature) that are proposed across the range of conditions that are likely to be encountered currently and in the future?
Are the sites representative of the area that I want to apply the data and can I prove it?
Are the times that I am sampling representative of the times that I want to apply the data (e.g., fish movement) and can I prove it?
What assumptions do I have to make to calculate an estimate (e.g., equal catchability, no loss of marks)?
Are the assumptions testable and have they been tested in other areas?
What are the consequences if assumptions are not met?
Are other independent methods of estimation available that can be implemented to test the quality of the primary estimate?
What method or computer program do I plan to use to calculate an estimate?
What are the risks to human safety?
What will be the likely impacts on target and nontarget taxa?
Does the allocation of resources result in the best quality estimate or highest statistical power?
Is the quality of the estimate or statistical power sufficient to meet goals?
Are there approaches designed to contribute to an increase in knowledge that can be used to improve the future allocation of resources (e.g., decrease or reduce sampling effort)?
Has the sampling design been reviewed by professional biologists and statisticians?

The choice of backpack versus driftboat electrofishing gear depends on the physical features of the stream (see Table 5 and Figure 1). Stream size, temperature, conductivity, and discharge determine the effectiveness of electrofishing gear types and configurations (Novotony and Priegel 1974; Meador et al. 1993; Thompson et al. 1998). Several U.S. federal agency programs, including the U.S. Geological Survey's National Water Quality Assessment Program and the U.S. Environmental Protection Agency's Environmental Monitoring and Assessment Program (<www.epa.gov/nheerl/arm>), have presented guidelines to aid in determining appropriate sampling gear selection based on stream conditions. Generally, backpack electrofishing units are used in small streams and boat- or raft-mounted units can be used in larger streams and rivers. Two backpack-mounted units can be used simultaneously when the stream size becomes too large to be effectively sampled with a single unit. When stream size and/or velocities become too large to sample with backpack units, a boat (Novotony and Priegel 1974) or raft (Stangl 2001) equipped with electrofishing equipment may be used. Successful electrofishing can be applied across a variety of field conditions; however, its effectiveness may be hampered when stream temperatures fall below approximately 7°C, as often happens during the winter (Roni and Fayram 2000). Many salmonid species bury themselves in the interstitial spaces of the substrate or in woody debris during the day when the water is very cold; however, they often come out during the night. Effective winter daytime electrofishing depends on successfully removing fishes from concealment habitat (Roni and Fayram 2000).

TABLE 5. — General criteria and applicable electrofishing equipment appropriate for use in streams of different sizes

Equipment	Temperature (°Celsius)		Discharge (m^3/s)		Stream width (m)		Conductivity (mmhos)	
	Min.	Max.	Min.	Max.	Min.	Max.	Min.	Max.
1 backpack	6	18	0	0.3	0	7	40	250
2 backpacks	6	18	0.3	3.3	7	15	40	250
Boat or raft	6	18	11	200	15	>15	40	250

The timing of sampling should also be considered relative to the species being studied. For example, many salmonid species make spawning migrations and may be highly mobile during these periods. Abundance sampling during these time frames will provide a snapshot estimate of a moving population that should not be applied to other times or seasons. When estimating salmonid abundance, distribution, or size structure, sampling during low-movement periods may be beneficial (e.g., low summer base flow periods).

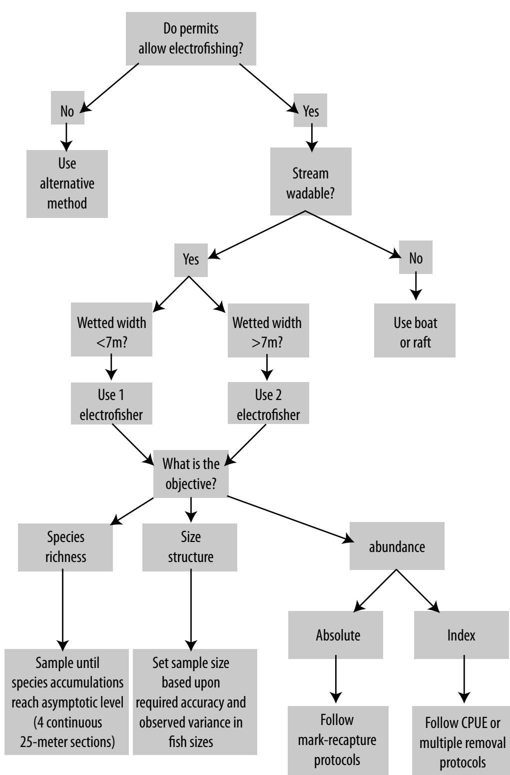


FIGURE 1. — Decision tree for electrofishing studies.

Abundance Estimation

Determining the absolute number of fish in a given area using electrofishing methods is not an easy task, and the most common field sampling protocols used in small streams (e.g., catch-per-unit-effort [CPUE], multiple-removal/depletion) generally lead to estimates that are negatively biased (Riley and Fausch 1992; Riley et al. 1993; Peterson et al. 2004). Estimator bias typically results from violations of critical estimator assumptions. The level of the bias is not usually evaluated in most studies. In situations where bias is not directly evaluated, abundance estimates should be treated as biased indices of abundance (Peterson et. al. 2004). Abundance indices generated from multiple-removal or CPUE sampling can be corrected or calibrated when an unbiased abundance estimate exists and can be used in an "index to unbiased estimate" comparison (Rosenberger and Dunham 2005). The key to generating accurate, unbiased estimates is to test the assumptions associated with the estimator employed and to validate resulting estimates by sampling known numbers of fish or by using an alternative unbiased method.

Important Assumptions

As previously mentioned, estimator bias typically results from failing to meet the assumptions of the estimator employed under typical field conditions. White et al. (1982) describe the assumptions associated with common closed-model estimators used for estimating stream fish abundance (e.g., multiple removal and mark–recapture). In mark–recapture studies, general assumptions include

1. the population is static during the course of sampling (i.e., there is no net movement of fish into or out of the study site where movement can arise from births, deaths, immigration, or emigration);
2. fish do not lose their marks during the course of sampling; and
3. the mark history of each fish is noted correctly on each sampling occasion (i.e., captured fish are reported correctly as marked or unmarked).

In multiple removal/depletion studies, two corresponding general assumptions include

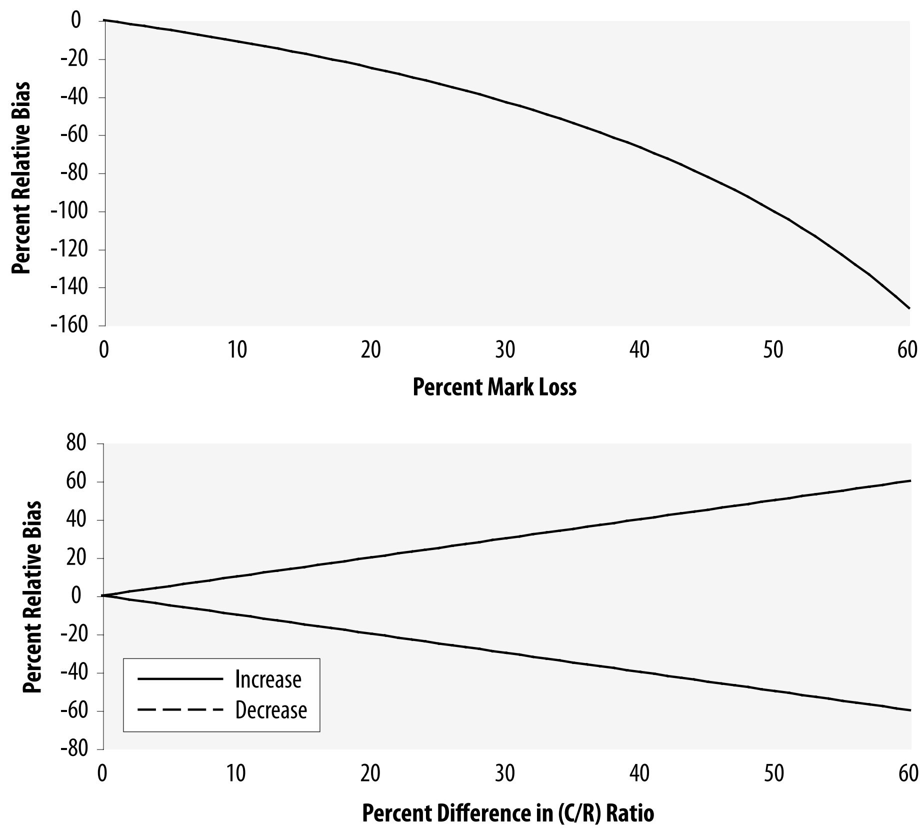
1. the population is static during the course of sampling (i.e., there is no net movement of fish into or out of the study site where movement can arise from births, deaths, immigration or emigration); and
2. the number of fish captured during each removal pass is reported correctly.

Movement

The importance of population closure during sampling was discussed in detail by White et al. (1982). Essentially, closed-model estimators, such as those commonly used to sample stream fish, make the assumption that N is constant in the study site during sampling. Net barriers, or block nets, are often installed at the upstream and downstream ends of the sampling site and are generally assumed to prevent movement (Peterson et al. 2005), although this assumption is rarely tested in practice. It has been our experience that salmonids can bypass block nets and that population closure should not be assumed and should be tested during field studies (Temple and Pearsons 2006). Violations of the movement assumption will result in biased abundance estimates (see Figure 2a). When movement assumptions cannot be met under field conditions (i.e., the population is not closed), alternative open models may be applied. Discussion of open model estimators is beyond the scope of this protocol and we refer readers to the review of alternative models provided by Schwarz and Seber (1999).

Capture Efficiency

Both removal and mark–recapture estimators have additional assumptions regarding capture efficiencies for target species. In removal studies, it is assumed that the probability of capture for every fish is equal and that it does not change between removal passes (Zippin 1956). It is common to group fish based on size and to calculate separate estimates for each group to satisfy the assumption of equal capture efficiency of individual fish (Anderson 1995). Satisfying the assumption of equal capture efficiency of fish during each removal pass is more difficult. By performing at least three removal passes, this assumption can be tested statistically (White et al. 1982). The problem with this technique is that these tests are based on nonparametric statistics that have been shown to have poor power to detect true differences in electrofishing studies (Riley and Fausch 1992). Thus, removal estimates may be biased due to undetected differences in capture efficiencies across electrofishing passes. In mark–recapture sampling, it is assumed that marked and unmarked fish are equally catchable. Unequal capture efficiencies of marked and unmarked fish can severely bias abundance estimates (see Figure 2B). Most evaluations of capture efficiencies of marked and unmarked fish are based on observations of behavior and physiology (Schreck et al. 1976; Mesa and Schreck 1989). It is difficult and time consuming to test capture efficiency assumptions directly in practice, and some authors have considered it to be untestable (referenced in Gatz and Loar 1988; however, overall estimator bias can be evaluated directly with careful planning.



FIGURES 2a and 2b. — Percent relative bias (PRB) of mark–recapture estimates when (2a, top) movement or (2b, bottom) catchability (or capture efficiency) between marked and unmarked fish assumptions are violated to different degrees.

There are many factors that potentially influence electrofishing capture efficiency (Zalewski and Cowx 1990) (Table 6). Variation in these factors can cause differences in response variables such as catch-per-unit-effort, assemblage composition, and size assessment. Practitioners should attempt to control or account for variation in these factors. Some have developed estimators that use factors correlated with capture efficiency (Peterson and Zhu 2004). The most common correction is to account for differences in capture efficiency associated with variation in fish size.

Electrofishing is known to produce biased estimates of fish size (Anderson 1995). In other words, the capture efficiency of different sizes of fish is unequal. In general, if electrofisher settings are set to have the highest efficiency on the average size of fish, then the smallest fish will be captured with the lowest efficiency, average-sized fish with the highest efficiency, and largest fish with an intermediate efficiency. Different methods can be used to compensate for size-based bias (Vadas and Orth 1993). Population estimates can be made for each size class of fish and then the estimates summed. The maximum-likelihood method and other methods can be used to estimate capture efficiencies of different sizes of fish, and then differential capture efficiencies can be factored into the calculation of the abundance estimate. Fish-size data can be weighted with differences in capture efficiency.

TABLE 6. — Some factors that affect electrofishing capture efficiency

Factor	Relationship	Reference
Fish species	Variable—some species are more vulnerable than others	Buttiker 1992
Fish abundance	More fish decreases efficiency	Simpson 1978; Riley et al. 1993
Fish size	Depends on initial settings; generally positive capture efficiency with increasing fish size	Buttiker 1992; Anderson 1995
Fish behavior	Fish may be more or less susceptible after exposure to previous electrofishing; territorial fish may be more susceptible than schooling fish	Mesa and Schreck 1989
Percentage of pool volume taken up by rootwads	Volume of rootwads negatively related to efficiency	Rodgers et al. 1992
Amount of undercut banks	Positively related to bias (more undercut bank = more bias)	Peterson et al. 2004
Amount of cobble substrate	Negatively related to efficiency	Peterson et al. 2004
Cross-sectional stream area electrofished	Decreasing efficiency with increasing stream area	Kennedy and Strange 1981; Rodgers et al. 1992; Riley et al. 1993; Peterson et al. 2004
Water temperature	Efficiency is normally distributed with temperature	Bohlin et al. 1989
Water discharge (Q)	Negatively related to increasing Q	Funk 1947; WDFW (unpublished data)
Water conductivity	Generally positive at low conductivities, negative at high conductivities (>500 mmhos)	Reynolds 1983; Hill and Willis 1994
Water transparency	Dependent upon how fish respond to water transparency (i.e., more able to avoid electroshock)	Reynolds 1983; Dumont and Dennis 1997
Water surface disruption (e.g., rain, wind)	Efficiency decreases with increasing surface disruption	Reynolds 1983
Time of day/night	Variable	Paragamian 1989; Dumont and Dennis 1997
Time of year	Variable	Roni and Fayram 2000
Human effort and proficiency	Higher effort and proficiency increases efficiency	Reynolds 1983

Evaluating Bias

Three sampling methods that provide mechanisms to evaluate estimator bias include dual gear procedures (Mahon 1980), stocking fish (Rodgers et al. 1992), or sampling known numbers of marked fish (Peterson et al. 2004). Dual gear procedures assume that one gear produces accurate and precise estimates. Employing a second gear type that is 100% efficient would be ideal for this comparison. In reality, however, very few sampling gears can achieve 100% efficiency. Examples of common applications have included the use of toxicants as the second gear type to generate accurate and precise estimates for comparison with electrofishing estimates (Mahon 1980). Sampling with toxicants is lethal to the sample fish and may not be acceptable for sampling rare or listed species, not appropriate for long-term monitoring, and unlikely to be 100% efficient (Bayley and Austen 1990). Stocking known numbers of fish into selected areas and subsequently sampling them has also been proposed as a means to evaluate gear bias (Rodgers et al. 1992); however, it may be counterproductive to artificially stock fish into areas because it is not known if parameter estimates generated from artificial stocking will be representative of the wild population, and stocked fish may not exhibit normal behaviors. Capture–recapture approaches may be the most appropriate methods for sampling wild fish in wadable, coldwater streams. With the considerations of bias, we will focus our discussion on mark–recapture sampling to generate estimates from known numbers of marked fish. We do this because the commonly used alternative, multiple-removal/depletion sampling, has been documented to produce biased estimates under typical field conditions (see Table 7) (Cross and Stott 1975; Riley and Fausch 1992; Peterson et al. 2004); however, recall from discussions of the MSE, that when estimator bias is small relative to sampling variance, multiple-removal sampling may still produce useful information because the error associated with estimator bias is only a small portion of the total MSE. Thus, using estimators or expansions based on multiple-removal/depletion based efficiencies, such as single-pass (Jones and Stockwell 1995) or CPUE (Simonson and Lyons 1995) sampling, might be adequate to index abundance in many situations. For studies that require accurate estimates or that do not have prior knowledge of sampling variance and estimator bias under typical field conditions, we recommend following mark–recapture protocols to generate abundance estimates.

TABLE 7. — Species (Spp) abbreviations include coho salmon *Oncorhynchus kisutch* (COH), brook trout *Salvelinus fontinalis* (EBT), brown trout *Salmo trutta* (BT), rainbow trout *O. mykiss* (RBT), bull trout (BULL), and cutthroat trout *O. clarkii* (CUT).

Reference	Location	Spp	Bias
Cross and Stott 1975	Rectangular ponds	Gudgeon roach	34%
Petersen and Cederholm 1984	Small Washington streams	COH	9%
Riley and Fausch 1992	Small Colorado streams	EBT BT RBT	9%
Rodgers et al. 1992	Small Oregon streams	COH	33%
Riley et al. 1993	Newfoundland streams	Atlantic Salmon parr	23%
Peterson et al. 2004	Small Idaho and Montana streams	BULL CUT	88%
Temple and Pearsons 2005	Small Washington streams	RBT	18%

Catch-per-unit-effort (CPUE)

Generally, increasing and/or validating the accuracy and precision of abundance estimates requires increased time, effort, and money (Bohlin et al. 1989). In some instances, a simple index of abundance is acceptable when study objectives do not require accurate and precise estimates. In such cases, simple CPUE indices may be appropriate. CPUE indices assume that the rate of catch in a sample is proportional to stock size (Thompson et al. 1998). Examples of backpack electrofishing CPUE indices have included single-electrofishing pass sampling to index abundance (Jones and Stockwell 1995; Mitro and Zale 2000). In some instances CPUE sampling may provide abundance estimates that are as reliable as those from traditional removal sampling (Jones and Stockwell 1995; Kruse et al. 1998), particularly when calibrated with unbiased estimates; however, caution should be used when interpreting CPUE indices because the level of bias in absolute terms usually will not be known (Bohlin et al. 1989).

CPUE sampling is perhaps the least labor-intensive electrofishing method available for indexing abundance. Sampling generally consists of selecting the study site(s), establishing the metric of effort, and sampling fish. Effort is often recorded as the amount of time (seconds or minutes) the electrofishing unit supplies electricity to the water and abundance indices are presented as fish/time (e.g., fish/minute). Other measurements of effort include establishing and sampling a specific length of stream or establishing a particular number of fish to sample. CPUE sampling can be performed following the mark–recapture electrofishing protocols presented in Table 9 in the following sequence: Perform steps 1–3, 6–10, 12–13, 15–16, and 23. Calculate the abundance index as the number of fish captured per unit effort.

One problem associated with CPUE sampling is that capture efficiency is assumed to be independent of field conditions (e.g., turbidity, discharge, temperature, depth, habitat complexity). This may be an appropriate assumption if conditions are very similar across comparisons. If it is not, corrections may be necessary. Correcting CPUE indices can be performed by calibrating them with known estimates (Fritts and Pearsons 2004).

Multiple Removal/Depletion

A common method used for enumerating salmonids in small streams is based on the multiple removal/depletion technique proposed by Moran (1951) and by Zippin (1958). Under the removal model, the declining catch of fish between multiple electrofishing passes is used to calculate capture efficiencies and abundance estimates. A common protocol requires a section of a stream to be isolated with block nets and a minimum of two removal electrofishing passes to be performed, although three or more removal passes are generally recommended so that catchability assumptions can be statistically tested (White et al. 1982). Practitioners should be aware that the tests used to evaluate equal catchability assumptions generally have low power to detect true differences (Riley and Fausch 1992). The Zippin removal estimator appears sensitive to violations of catchability assumptions (Bohlin and Sundstrom 1977). To circumvent this, Otis et al. (1978) proposed the generalized removal estimator that allows for unequal catchability of fish between removal passes. This estimator requires conducting four or more removal passes; a protocol that could be costly and time consuming. In addition, completion of four removal passes is no guarantee that the resulting removal estimate will be unbiased (Riley and Fausch 1992). The most efficient removal electrofishing protocol will minimize the number of removal passes that must be performed and will maximize the utility of the data. Conducting only two removal passes may satisfy effort requirements but likely produces biased estimates with wide confidence intervals (Riley and Fausch 1992). Additionally, two pass estimates fail when the number of fish captured on the second pass is greater than the number captured on the first electrofishing removal pass. To correct for this, some studies have recommended pooling catch data from multiple sites (Heimbuch et al. 1997). To satisfy effort and utility requirements, Connolly (1996) developed charts for field use based on stringent removal guidelines that produced the most reliable estimates with a minimum number of removal passes. In situations where accurate estimates are not necessary, removal sampling can be a useful method to index abundance. When accurate estimates are necessary, removal estimates should be interpreted with caution unless calibrated with an unbiased estimate, or critical assumptions are tested and validated under field conditions.

Multiple removal sampling protocols are similar to the mark–recapture protocols presented in Table 9, with the exception that fish do not need to be marked and fish captured after each electrofishing pass are enumerated and held in live wells (preferably outside the sampling section) until at least two but preferably three or more electrofishing passes are performed. Tables such as those presented by Connolly (1996) should be used as an aid to determine the number of removal passes to perform based upon the removal patterns observed in the field and the precision required by the study. Multiple removal sampling can be performed following the mark–recapture electrofishing protocols presented in Table 9 in the following sequence: Perform steps 1–13, then repeat steps 6–13, holding fish captured in the first electrofishing pass live wells. Consult removal tables presented by Connolly (1996) to determine if another electrofishing pass is required. If so, repeat steps 6–13. If not, consider step 20 and perform steps 21–23. Calculate the abundance index following the equations presented in Seber and LeCren (1967) or Zippin (1956) or consult Table 13 for online sources for computer programs to aid in computations.

Mark–Recapture

Mark–recapture (capture–recapture) electrofishing protocols have been shown to be useful means to measure the sampling efficiency of known numbers of marked fish (Rodgers et al. 1992; Peterson et al. 2004) and to generate population estimates (White et al. 1982); however, one drawback of this technique is logistical constraints that are commonly assumed to be associated with the recovery period—the time delay between the marking and recapture sampling. Recovery periods lasting longer than a single workday limit the utility of the mark–recapture method for short sites (i.e., 100 m) in tributaries because fish movement is difficult to control during longer periods, thereby violating the closed population assumption of the estimator. In some streams with high flows or debris loads, installing and maintaining block nets for long periods is extremely difficult. Thus, small stream electrofishing protocols extending beyond one working day are impractical for fisheries practitioners in many locations.

Most authors have recommended a minimum 24-hour recovery period between mark and recapture sampling. Peterson et al. (2004) suggested that sampling protocols utilized for electrofishing in coldwater areas should allow a recovery time of between 24 and 48 hours. This recommendation was assumed to provide a balance between the violations of movement and equal catchability assumptions. Similarly, Mesa and Schreck (1989) observed that wild cutthroat trout resumed normal behaviors only after a 24-hour recovery period between marking and recovery. Schreck et al. (1976) questioned the validity of mark–recapture estimates when the recovery period was shorter than a working day. These studies based their recovery periods on behavioral observations (Mesa and Schreck 1989) or physiological response to electrofishing (Schreck et al. 1976). Regardless of the behavioral or physiological differences between marked and unmarked fish, the appropriate recovery period should satisfy the critical assumption that marked and unmarked fish are equally catchable.

The appropriate recovery period to provide between mark and recapture sampling can be evaluated by comparing the catchability of marked fish that have recovered for at least 24 hours versus those that have recovered for shorter periods. The length of the shorter recovery period should allow both mark and recapture sampling to be completed in a single day. In a case study in the Yakima River, Washington, there was no significant difference in the catchability of rainbow trout that had recovered for 24 h versus a 3-h period. Thus, a 3-h recovery between marking and recapture proved to be a sufficient recovery period to allow marked rainbow trout to recover from handling (Temple and Pearsons 2006). This finding should be tested in other areas under different conditions before it is widely applied.

Similar to multiple removal sampling, mark–recapture sampling can produce biased estimates when the assumptions of the estimator are not met under field conditions (Table 8). The severity of the bias is generally lower for mark–recapture estimates of stream fish than reported for removal sampling (Peterson and Cederholm 1984; Rodgers et al. 1992). In contrast to removal estimates, mark–recapture estimates have the benefit of being fairly robust to potential bias introduced from poor crew experience and variable environmental conditions (e.g., channel type, habitat complexity, stream size). Nevertheless, the assumptions associated with mark–recapture estimates should be tested under field conditions.

TABLE 8. — Examples of published literature documenting negative bias in mark–recapture estimates of roach and coho (COH) salmon abundance.

Reference	Location	Spp	Bias
Cross and Stott 1975	Rectangular ponds	Roach	12%
Petersen and Cederholm 1984	Small Washington streams	COH	5%–6%
Rodgers et al. 1992	Small Oregon streams	COH	15%

Mark–Recapture Backpack Electrofishing Protocol

The step-by-step protocol is presented in Table 9 for quick reference. We will provide more information in the text for each step. Sequential numbers in Table 9 correspond to the following paragraphs for easy reference.

1. Select sampling site. Sampling sites should be representative with respect to habitat parameters and fish abundance if the sample will be extrapolated over any spatial scale greater than the site.
2. Identify appropriate length of stream to sample. See Establishing Linear Site Length—Backpack Electrofishing section, p. 112.
3. Measure site length along the contours of the stream channel. Minimize walking in the stream channel to prevent spooking fish.
4. Install blocking nets at the upstream and downstream boundaries of the site perpendicular to the flow of the water. Ensure that the size of the net mesh is small enough to prevent movement of the smallest fish that will be estimated. When determining net placement, nets may be moved upstream or downstream if the site boundary is unsuitable for net placement. For example, deep pools, swift water, or large debris piles may be difficult areas to secure nets. Stretch net across the stream channel. Small rocks may be used to secure the bottom of the net to the streambed. Once the net is secured along the stream channel, the ends may be secured to standing timber using ropes or string or may be anchored to the stream bank using large boulders. Branches or sticks may be used to prop the net up across the stream channel (see Figure 3).

5. We recommend installing an additional block net in the middle of the site to provide a means to evaluate movement rates and potential violations of movement assumptions. If evaluating movement rates, follow procedures 6–16 and then consider step 17.
6. Place live well buckets along the stream margin to hold captured fish. Conventional 5-gal buckets make convenient live wells that are cheap, durable, and easy to carry. We suggest using buckets colored similarly to the stream substrate to prevent substantial pigment changes in captured fish. The number of buckets needed may depend upon site length and fish density. We recommend separating sites into 10–25 m intervals. This allows replacement of fish close to their point of capture. Avoid overcrowding fish in live wells (see Figure 5).
7. Measure and record stream temperature and water conductivity prior to electrofishing. Conductivity, or the water's potential to carry electricity, is temperature-dependent and can vary throughout a single day. Electrofisher settings should be adjusted to the manufacturer's recommended guidelines based on water conductivity and temperature. In our experience, straight, unpulsed DC produces minimal injury to salmonids (McMichael et al. 1998) and efficient capture of salmonids. Generally, straight DC outputs ranging between 200 and 400 Volts are effective for capturing salmonids when water conductivities range between 150 mmhos and 40 mmhos.
8. Conduct safety check prior to electrofishing. Ensure electrofishing equipment is functioning properly. All crew members who will participate in electrofishing should be outfitted with waterproof waders and rubber or neoprene gloves to insulate against electric shock. Polarized field glasses should be worn by all field personnel to minimize glare and to protect the eyes from bright conditions. This also facilitates fish capture.
9. Clear or reset seconds timer on electrofishing unit.
10. Begin electrofishing. When sampling salmonids, we recommend electrofishing in an upstream direction (see Figure 6). When stream size requires two electrofishing units, both should operate in tandem (side by side), moving upstream at the same pace. Begin electrofishing at the bottom block net, thoroughly checking for fish that may be impinged on the net. Systematically progress upstream, taking care to electrofish all habitat in the stream channel. Complex habitat such as debris jams and deepwater areas will require more effort than homogenous habitats. One or two crew members should be outfitted with dip nets to facilitate the capture of stunned fish. It is often effective for netters to remain downstream from the operator and to keep dip nets within 1 m of the anode. All fish observed should be netted and swiftly removed from the water to prevent injury. Captured fish should be immediately placed into the appropriate live well held at the stream margin. It may be convenient for one crew member to carry a live well bucket during sampling to facilitate transfer of netted fish to the bucket.
    a. In homogenous habitats it is often effective for the electrofisher operator to move the anode in a W-shaped pattern across the stream

channel while wading upstream. Netters should be prepared at all times to net stunned fish.

b. In complex habitats such as debris jams and undercut banks, it is often effective for the operator to insert the uncharged anode into the debris, depress the electrofisher switch, and slowly move the anode into open water areas. Fish will often be "pulled" from the debris into the open water where netters can capture them. Complex areas such as these often conceal several fish and should be thoroughly sampled until no additional fish are captured.

c. In deepwater areas such as pools or deep runs, it may be difficult to capture fish. One effective technique may be to "chase" fish into shallow water areas where they can be easily captured. The operator can keep the electrofisher charged while moving it back and forth across the channel and up and down in the water column. Netters should attempt to capture fish that become stunned but should remain conscious of the water depth to avoid submersion of hands or arms in the water. Systematically electrofish the entire pool area and slowly move upstream. Fish will often flee the deep water moving upstream.

d. In fast water areas, it is often effective for the operator to insert the anode into the water an arm's reach upstream, depress the electrofisher switch, and move the anode downstream at approximately the same velocity the water is traveling. Netters should have their nets pinned against the substrate in the fast water areas. Constriction points in the streamflow, such as between two large rocks, make good areas for dip-net placement. The operator can move the anode downstream into the dip net and then release the electrofisher switch to discontinue shocking. The net should then be immediately removed from the water and inspected for fish. Often, shocked fish will be pushed into the net by the stream flow or drawn into the net by the anode. Netters should check their nets frequently in these areas because fish will often become impinged in them without the crew's knowledge.

11. Continue electrofishing upstream until the upstream block net is reached. Thoroughly sample the substrate along the bottom of the block net for fish that may have moved upstream during sampling. Check downstream net for fish that may be impinged.
12. Record the amount of time electricity was supplied to the water (from seconds timer on unit).
13. Live wells can now be retrieved and fish can be anesthetized, identified, enumerated, and measured. Fish length is reported in a variety of ways and commonly measured as the maximum standard length, maximum total length, or fork length (Anderson and Gutreuter 1983). We recommend fork length for its ease of use. There are numerous conversions presented in the literature, to convert between length measurements (Carlander 1969; Ramseyer 1995).
14. For short-term sampling experiments, we recommend clipping or notching a small portion of one of the fins to identify marked fish. There

are several fish marking techniques that can be employed to mark fish (Parker et al. 1990), but for simple batch marking, fin clipping is a simple, cost-effective mark that is easy to apply and to identify in field settings.

15. Allow fish to recover from anesthesia and handling. If anesthetics are used to reduce handling stress, follow the manufacturer's guidelines for dosage information and appropriate recovery time. Fish will generally regain their equilibrium and begin to swim upright when recovering from anesthesia. Avoid releasing fish that have not fully recovered from the anesthetic. Do not release marked fish that are injured until sampling is complete because if they do not swim or behave normally, they may have a different catchability than their unmarked counterparts. Injured fish that are not marked and released should be accounted for in the final estimate by simple addition.
16. Release marked fish. Note the release time. We recommend releasing marked fish close to their point of capture. Subdividing the site into 10–25 m sections facilitates returning fish close to their capture point.
17. Allow fish to recover for a predetermined length of time. Avoid disturbing the stream section during the recovery period. Some authors recommend at least 24 hours for fish to recover from electrofishing, handling, and marking (Mesa and Schreck 1989); however, there are few investigations that evaluate appropriate recovery periods in terms of catchability. In the Yakima River Basin, we found 3 hours to be a sufficient amount of time for fish to recover and exhibit catchabilities similar to their unmarked counterparts. Appropriate recovery periods should be tested for individual studies.
18. Electrofish the sampling site again and capture all observed fish following steps 6–13. It is best to attempt to apply an effort similar to what was employed during the first electrofishing pass, but this is not critical in a Petersen-type mark–recapture protocol.
19. Captured fish can now be anesthetized, identified, enumerated, and measured. Pay particular attention to identify and enumerate marked fish and unmarked fish on the field data sheet. Fish can be released near their point of capture after fully recovering from handling. An example data sheet is presented in Appendix A.
20. If evaluating movement rates, repeat steps 6–16 in the adjacent stream section established by installing a block net in the middle of the site (step 5). This adjacent section can be sampled during the recovery period established for the section sampled first. Apply a different fin clip than that used in the adjacent section. Differentially marking fish between sections will allow the identification of fish to their upstream or downstream origin after taking the recapture sample. Release fish as in step 16. Perform steps 18–19 in the first section sampled after the minimum recovery period established in step 17 has been satisfied. Finally, perform steps 18–19 in the adjacent stream section after the minimum recovery period established in step 17 has been satisfied.
21. Release all fish near their point of capture at the end of sampling.
22. Remove block nets.

23. Sampling is complete. Return to office.
24. See Data Analysis section.

FIGURE 3. — Block nets used to isolate stream section.

TABLE 9. — Step-by-step backpack electrofishing mark–recapture protocol

1.	Select site location.
2.	Identify appropriate section length to sample.
3.	Measure site along stream contours.
4.	Install block nets at upstream and downstream boundaries.
5.	If evaluating movement, install additional block net in the middle of the site.
6.	Distribute live well buckets at 10–25 m intervals along the stream margin.
7.	Record stream conductivity and temperature.
8.	Perform safety check.
9.	Reset seconds timer on electrofishing unit.
10.	Electrofish in an upstream direction and net all fish observed within the site.
11.	Thoroughly check all nets for fish.
12.	Record electrofishing seconds.
13.	Retrieve live wells, anesthetize, identify, enumerate, and measure fish. Record data on waterproof data sheets.
14.	Mark fish (e.g., fin clip). Record marking data on waterproof data sheets.
15.	Allow fish to recover from handling until they swim and behave normally (generally 5–10 min.).
16.	Distribute and release marked fish back into site.
17.	Allow appropriate recovery period (3–48 hours; avoid disturbing site during this period).
18.	Perform recapture electrofishing pass (following steps 6–12).
19.	Retrieve live wells, anesthetize, identify, enumerate, measure, and identify all fish. Record mark history of all fish (marked or unmarked). Record data on waterproof datasheets.
20	If evaluating movement, perform steps 6–19 in adjacent netted stream section.
21	Redistribute and release all fish at the end of sampling.
22	Remove block nets.
23	Sampling is complete. Return to office.
24	Perform analysis to generate abundance estimate.

Species Richness/Community Structure/Size Structure Sampling

Electrofishing techniques have commonly been used to capture fish for describing species richness/community structure (Lyons 1992; Angermeier and Smogor 1995; Patton et al. 2000; Reynolds et al. 2003) and the size structure of fish populations (Paragamian 1989; Thurow and Schill 1996). Species richness studies often utilize single-pass electrofishing, or CPUE sampling, to estimate species presence or absence and relative abundance. When using presence/absence sampling to identify species richness, rare fish distributions, or simple presence or absence of a species at a particular geographical locale, considerations of sampling efficiency should be taken into account (Bonar et al. 1997; Bayley and Peterson 2001). Failure to identify an individual species at a location does not demonstrate that it does not exist there and may be the result of poor sampling efficiency. Failure to identify an individual species will demonstrate that it is not present only when the probability of detecting it when it is present is 100%—a highly improbable scenario (Bayley and Peterson 2001). Thus, there is some level of uncertainty when concluding that a species is not present because it was not captured, and that is related to sampling efficiency.

A common assumption that is made when estimating relative abundance is that capture efficiencies are the same for different species and/or for different age-classes of fish. This assumption is unlikely to be true, particularly for species of different sizes that use different habitats. Some have tried to rectify this by generating capture efficiencies for each species observed (Bayley et al. 1989; Angermeier and Smogor 1995; Bayley and Austen 2002).

Establishing Linear Site Length—Backpack Electrofishing

One important consideration should be the amount of stream to sample when using backpack electrofishing to capture stream fish. Sampling too long of a stream section may become labor intensive and could be expensive. Sampling too short of a stream section may not provide a true representation of the population parameter under study and could lead to poor data quality, particularly if the sample is intended to be extrapolated to estimate some population level parameter. We acknowledge that there are many different ways in which electrofishing may be used to study fish and their communities, and sampling methods may be very different based on study objectives. Commonly, stream sections of equal length are selected (based on statistically valid techniques) and sampled to estimate abundance or density. Hankin (1984) advised against selecting and sampling stream sections of equal length and proposed sampling in stream sections of varying length based upon breaks in natural habitat units. This technique can increase the precision of estimates, particularly when the objective is to estimate abundance (Hankin 1984); however, in very small streams, selecting stream sections based on habitat units that are extremely small may not be appropriate for estimating abundance because common abundance estimators (e.g., removal or mark–recapture) are based on a large sample theory that may not apply if there are very few fish in each small habitat unit sampled. Thus, the minimum amount of stream to sample will be dependent on the type of stream, habitat characteristics, and the population density in the units (Bohlin et al. 1989).

In a case study in the Yakima basin, Washington, estimates of rainbow trout density and of the size structure of the population were independent of stream width, suggesting that sites based on set linear stream distances were appropriate for monitoring these parameters. Our observations of cumulative abundance and size structure estimates of rainbow trout and the temporal variability associated with 25 m incremental increases in sampling distances suggested that longer sampling distances generally increased the accuracy of the estimates; however, abundance estimates were more variable than size estimates with increasing stream area. We found that sampling 200 m sites provided an acceptable balance between effort requirements and estimate precision for long-term monitoring of these variables. Of course, practitioners will need to balance their required level of accuracy with the costs associated with the sampling required to achieve that level of accuracy.

We found that species richness estimates were not independent from stream size. Similar to Cao et al. (2001), our data indicates that species accumulations will reach an asymptotic level at shorter linear stream distances in smaller streams than in large streams. Thus, large streams will require longer sampling sections than small streams when the primary objective is to assess community structure. Sampling effort requirements have been typically reported as the number of channel widths that must be sampled to collect a large proportion of the species present in a given stream reach at some predetermined level of accuracy (Lyons 1992; Angermeier and Smogor 1995; Patton et al. 2000; Reynolds et al. 2003). We found that 27–31 channel widths was the minimum sampling distance required to detect 90% of the species present in our streams. For generating accurate species richness estimates, Lyons (1992) recommended sampling stream lengths comprised of a minimum of 35 channel widths. Patton et al. (2000) found that stream lengths of 12–50 times the mean wetted stream width should be sampled to capture 90% of species when electrofishing small streams in the Great Plains region of the United States. In western Oregon streams, Reynolds et al. (2003) found that electrofishing linear stream distances of 40 times the mean channel width captured 90% of the species present. It may be more efficient and accurate to construct species accumulation curves to help determine the amount of stream to sample to collect a given percentage of species present (see Figure 4). For instance, study sections can be sampled in 25-m increments and cumulative species totals determined for each 25 m sampled. Hypothetically, we will assume research goals require 90% of species in the site be detected. Using Figure 4, we sample contiguous 25 m increments until no new species are captured for four contiguous 25 m sections. In this case, a 250-m stream section will be the minimum acceptable linear sampling distance required to capture 90% of the species present in the stream section (judged from the mean line in Figure 4). In some streams, 90% of species present will be captured at short linear stream distances and in others, long sections must be sampled. Initially, a long stream section must be thoroughly sampled to determine the true numbers of species present and to serve as the benchmark for calculating the percentage of species detected in each 25 m increment.

Since abundance and size variables can be effectively monitored using standard linear distances and species richness is more effectively monitored based upon variable site lengths, monitoring programs may need to adopt different sampling effort strategies for monitoring different response variables. When trout abundance or size variables are of interest, sampling effort requirements may be based on predetermined sampling distances. When species richness is the primary variable of interest, linear sampling distances should be based upon multiples of the mean wetted width. When all variables are of interest, a hybrid approach may be used in which set linear distances are sampled for abundance information and additional stream area sampled for species richness estimates. The appropriate amount of effort will be a balance between the program objectives and a fixed budget. When stream size was less than 40 channel widths, Reynolds et al. (2003) suggested that 150 m was necessary to ensure that sufficient numbers of individual fish were captured for estimating species richness in Oregon streams. We attempted to maximize our sampling to collect abundance, size, species richness, and rare fish distribution and found 200 m to be an appropriate site length for our needs.

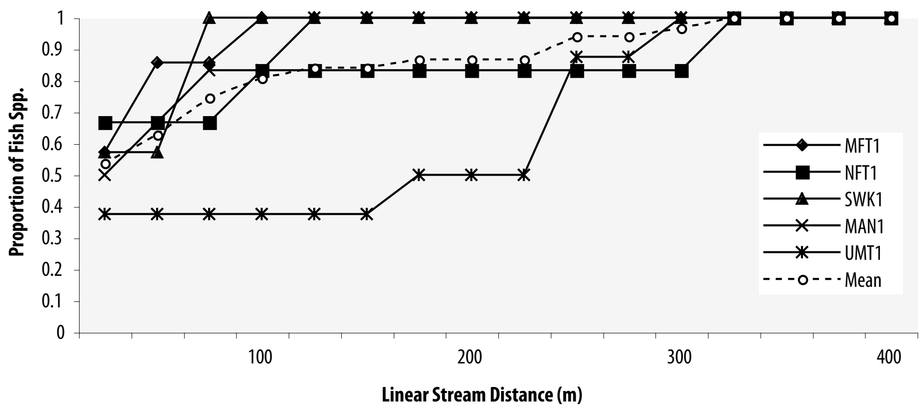


FIGURE 4. — Species accumulation curves for five sampling sites and the mean of the five sites.

Permitting and Equipment

Prior to initiating electrofishing, appropriate research and/or sampling permits should be obtained from the appropriate authority. For waters containing sensitive or listed species in the United States, research permits may be obtained from NOAA Fisheries (formerly National Marine Fisheries Service) for anadromous salmonids or from the U.S. Fish and Wildlife Service for resident salmonids; additional sampling permits may be required from state governments. Practitioners will need to facilitate good relations with private landowners to maintain access to sites located on private lands. Often landowners are willing to grant access to sites via their property when field personnel take the time to communicate the purpose of the study and maintain a professional and open dialogue with them. With the appropriate permits in hand and after all sampling design issues have been considered, practitioners will be ready to begin sampling fish. We recommend that equipment checklists be developed to aid in preparing field gear for sampling activities. An example equipment list for a typical mark–recapture backpack electrofishing sample is presented in Table 10. Examples of mark–recapture data sheets are provided in Appendices A and B and should be printed on waterproof paper. When all field gear has been assembled for a stream survey, crews can travel to the sampling sites and begin sampling.

TABLE 10. — Sample equipment checklist for typical backpack electrofishing mark–recapture survey

Sampling permits
Backpack electrofisher
Spare electrofisher batteries (or fuel for gas-powered units)
Pole mounted anode (and spare)
Dip net(s)
Block nets
Fish sampling gear (balance, measuring board, anesthetic)
Sharp scissors
Buckets or live wells
Thermometer and conductivity meter
Data sheets and pencils (examples provided in appendices A and B)
Waterproof gloves and waders
Polarized field glasses
Other gear:
Global positioning system (GPS) unit
Rain gear

Boat Electrofishing

When environmental conditions such as stream size or discharge (see Table 5) prevent effective electrofishing with backpack-mounted electrofishers, larger boat- or raft-mounted electrofishing units may be more effective for capturing fish for estimating abundance. Most of our experience includes using a drift boat-mounted electrofishing unit; we will discuss mark–recapture protocols in the context of drift electrofishing. However, much of this discussion will apply to other boat electrofishers in streams as well. There are several anode and cathode arrangements utilized on electrofishing boats.

There are other electrofishing boat designs, including motor-powered boats, that may require different specific protocols; however, the underlying principles in mark–recapture estimation remain the same—with the primary objective being to capture, mark, release, and recapture fish. The Petersen-type estimator is commonly used in large rivers because multiple removal techniques are impractical in them. The assumptions of the Petersen-type mark–recapture estimate are the same for both boat and backpack electrofishing techniques, although there may be additional considerations for practitioners to consider when sampling large streams and rivers with boat- or raft-mounted units.

FIGURE 5. — Overcrowded fish in livewell.

FIGURE 6. — Electrofishing a wadable stream.

Movement

Sampling in large streams and rivers becomes more problematic due to their large size. It is often not practical or feasible to isolate the study site with block nets to prevent movement of fish, and thus movement may be difficult to control during sampling; however, it is practical to sample longer linear stream sections, and as the sampling reach length is increased and the number of fish sampled is sufficiently large, the proportion of fish that potentially move in or out of the site at the margins becomes a small proportion of the population and will have a minimal effect upon the resulting estimate. Although mark–recapture estimates assume there is no movement during sampling, note that equal immigration or emigration of unmarked fish at the reach margins will not have any negative effect upon a mark–recapture estimate. Violations of movement assumptions will not negatively affect mark–recapture estimates as long as the effective ratio of marked/unmarked fish in the sample is not altered during the course of sampling. Thus the basic Petersen-type mark–recapture estimate is sensitive only to emigration of marked fish or unequal immigration or emigration rates of unmarked fish. Violations of movement assumptions that may bias the estimate can be tested directly using traps or weirs, or by some other direct estimate (Gatz and Loar 1988). If movement assumptions cannot be met, practitioners may consider using a mark–recapture model that does not assume population closure such as those discussed by Schwarz and Seber (1999).

Capture Efficiency

It is not generally recommended for both mark and recapture sampling to be completed in a single day when drift electrofishing. Captured fish are put in live wells kept on board the boat and transported downstream as the boat drifts downstream and are therefore relocated. Fish that are relocated will need time to redistribute and randomly mix with the unmarked fish. Releasing marked fish at several points within the site will facilitate redistribution. We recommend subdividing the sampling site into smaller reaches, and captured fish should be identified, measured, marked, and released at those points within the site. Vincent (1971) recommends that sampling sites should be at least 1,000 feet (305 meters) in length—or long enough to ensure a minimum sample size of 150 fish. It is best to release fish in shallow-water areas that are not in close proximity to deep pools, side channels, or river mouths to ensure that marked fish redistribute themselves and stay in the sampled river channel. If fish are released in deepwater areas such as deep pools, they have the potential to move to the bottom of the pool and effectively become uncatchable during the recapture sample. Releasing fish in several shallow-water areas and allowing at least 2 days between mark and recapture sampling (Vincent 1971) may help satisfy the assumption of equal catchability of marked and unmarked fish. In some instances, night electrofishing can increase sampling efficiency (Paragamian 1989), and boats or rafts should be outfitted with halogen lamps to aid navigation if sampling at night (see Figure 8).

Drift Boat Electrofishing Protocol

While actively electrofishing for bank-oriented species, close proximity to bank structures should be maintained while keeping the boat or raft at an angle of about 45° to the direction of the flow. In this position, the cathode and anode are in an effective fishing position while giving the rower the greatest ability to respond to changes in bank structures. It may not be necessary to avoid overhanging trees if the netter can duck down, brace him/herself, or otherwise be prepared and the boat can pass underneath without making contact. Avoid close proximity to sweepers or low-hanging structures in fast water and alert the net person of low-hanging branches or potential collisions. Keep the anode ring and droppers just far enough off the shore to minimize its contact with the bank or stream bottom. In areas with gradual sloping and/or uniform bottoms, try to keep the anode suspended about 0.7 m from the stream bottom.

Finally the rower should maintain a speed equal to that of the water. If drifting too fast, fish will be missed as the boat passes over them before the netter can net them. If moving too slowly, fish will be pulled downstream ahead of the boat before the netter can catch them. The rower can often adjust the boat speed and position ahead of time to present the gear where the fish are most likely to be. The step-by-step protocol is presented in Table 11 for quick reference. We will provide more information in the following text for each step. Sequential numbers in Table 11 correspond to the following paragraphs for easy reference.

1. Determine site location, boundaries, and site length.
2. Launch drift electrofishing boat at the upstream access point.
3. Start generator and turn on electrofishing equipment while floating downstream with the current.
4. Check that voltage and amperage output settings do not exceed 7 A and 450 V (McMichael et al. 1998).
5. Rower maintains boat at a 45° angle towards the bank.
6. Netter nets all visible target fish and places in on board live well.
7. Drift to predetermined workup station, disengage electrofishing equipment, and anchor or beach the boat.
8. Identify, measure, weigh, and mark target fish. For short-term sampling experiments, we recommend clipping or notching a small portion of one of the fins (caudal, ventral, or anal) to identify marked fish. There are several fish marking techniques that can be employed to mark fish (Parker et al. 1990), but for simple batch marking, fin clipping is a simple, cost-effective mark that is easy to apply and to identify in field settings. We recommend using fish anesthetic to facilitate fish handling. An example data sheet for recording fish data is provided in Appendix A.
9. Allow a minimum of 15 min for fish recovery from handling. If anesthetic is used, allow the appropriate recovery period per manufacturer's recommendation.
10. Release marked fish in shallow water 15 m upstream from boat to facilitate redistribution and prevent reshocking marked fish. Do not release injured fish or fish that have not recovered from anesthesia. Account for fish that are not released during analysis.
11. Quickly return to boat, pull anchor, and resume electrofishing.
12. Repeat electrofishing procedures 3–11 until the end of sampling site is reached. Drift to boat launch.
13. Electrofishing run is complete. In large rivers, it may be necessary to perform two marking runs to successfully capture fish on both riverbanks.
14. Allow fish to recover and redistribute from capture and marking before resampling the same stretch of stream/river.
15. Return to site. Repeat steps 2–13. Substitute fish marking with examining for marks and recording marked and unmarked fish in the sample. In large rivers, it may be necessary to perform two recapture runs to capture fish successfully on both riverbanks.
16. Data can be analyzed considering the information presented under the Data Analysis section.

TABLE 11. — Drift boat electrofishing mark–recapture protocol

1.	Select site.
2.	Launch drift boat.
3.	Turn on electrofishing equipment.
4.	Check that voltage and amperage output do not exceed 7 A and 450 V.
5.	Rower maintains boat at a 45° angle towards bank.

6. Netter nets all target fish and place in on board live well.
7. Navigate to station workup, disengage electrofishing equipment, and anchor boat.
8. Identify, measure, weigh, and mark target fish.
9. Allow fish minimum of 15 min to recover from handling.
10. Release marked fish 15 m upstream from anchored boat.
11. Quickly return to boat, pull anchor, turn on electrofisher, and resume electrofishing.
12. Repeat procedures 3–11 until end of sampling site is reached and float to boat launch.
13. Remove boat from river. Electrofishing run is complete.
14. Allow fish to recover and redistribute between sampling runs.
15. Repeat steps 2–13 for recapture run. Substitute fish marking with enumerating marked and unmarked fish in the sample.
16. Analyze data.

FIGURE 7. — Navigation through challenging obstacles should be carefully planned.

FIGURE 8. — Night drift boat electrofishing crew.

Personnel Requirements and Training

The required crew size for effective backpack electrofishing depends on the size of the stream sampled. Effective capture of salmonids can be obtained in most instances with a three-person crew when sampling with a single backpack-mounted unit. This allows one person to be dedicated to operating the electrofisher and at least one person dedicated to netting fish, while a second person carries a portable live well and provides additional back up netting. When two backpack units are used in tandem, an additional operator and net person should be used (i.e., a total of five persons). In many instances, budgetary restraints may limit crew sizes; in these situations, volunteer help may be a valuable resource (Leslie et al. 2004). Under no circumstance should electrofishing be performed alone.

Drift boat and raft electrofishing requires a minimum of two crew members; on larger rivers, three or four are appropriate. Under most conditions, two are sufficient. One person is dedicated to maneuvering and navigating the boat or raft while the other nets fish from the bow of the craft. Crew members may alternate rowing and netting to avoid fatigue. Both crew members should be familiar with the stream section that will be sampled to avoid encountering any unforeseen obstacles (see Figure 7). Drift electrofishing should not be performed by personnel inexperienced in navigating drift boats or rafts in the riverine environment. In most cases, the river will be large enough to be dangerous and crews should carefully read the section on safety before venturing out.

All personnel who work in the general vicinity of an electrofishing operation must be thoroughly trained about electrofishing theory, application, and safety concerns before electrofishing begins. Every electrofishing crew should have at least one principal operator or crew leader who is experienced or certified in electrofishing in charge during the electrofishing operation (Reynolds 1983). The U.S. Fish and Wildlife Service's National Conservation Training Center provides formal electrofishing training and certification courses; course information and scheduling is available online at <http://training.fws.gov/BART/courses.html>. In addition, some equipment manufacturers also provide training to familiarize practitioners with their equipment.

Safety

Backpack Electrofishing Safety

The safety of the crew is always the top priority. All crew members should reserve the right to refuse electrofishing without fear of reprimand if conditions appear unsafe. Perform a safety check prior to electrofishing to ensure that the equipment is functioning properly following the equipment manufacturer's guidelines. Do not electrofish with equipment that is not functioning properly. All crew members should be outfitted with waterproof waders (e.g., neoprene) to insulate against electric shock. Waterproof rubber lineman-style gloves should be worn by all crew members anytime electrofishing is underway. Electrofishing should not proceed when environmental conditions are unsafe (e.g., deep water, high velocities, excessive rainfall).

Drift Electrofishing Safety

As previously stated, crew safety is paramount. Unfortunately, due to the variability between river characteristics, experience of the crew rather than specific environmental guidelines is ultimately the deciding factor on whether or not a survey should be undertaken. For example, high flow has the potential of pushing fish towards the riverbanks, thereby increasing capture efficiency; however, choosing to perform a drift boat or raft electrofishing survey under high-flow conditions and in an area where stream width is not sufficient to allow maneuverability, or where there may not be adequate time to maneuver around obstacles, would exhibit very poor judgment.

The application of electricity into water to induce the capture of fish is inherently dangerous work. The voltage and amperage produced by most boat or raft mounted units is significant enough to cause injury to personnel, and safety precautions should be employed to minimize the risk of electrocution. Crew members should wear waterproof waders and dry, rubber, lineman-style gloves to insulate against electric shock. The dip net used should be constructed with a nonconductive fiberglass handle. The use of a personal flotation device (PFD) is mandatory for all crew members anytime electrofishing is underway. All crew members should familiarize themselves with the location of the power shutoff switches associated with the electrofishing unit. The netter stationed on the bow should always utilize a dead-man switch and should attach it to his/her PFD. This switch is designed to shut off the electrical current should the netter fall out of the boat or raft. The electricity should be shut off anytime the boat or raft is beached or anchored and whenever a crew member exits the craft.

Crews should expect to hit submerged objects while drift electrofishing. The rower should use good judgment while navigating and always maintain control. If it appears that a collision is imminent, the best tactic is to point the bow at the object and try to slow down as much as possible to minimize the force of impact. The netter can also use the net handle to help push off the obstacle. Constant communication among the crew members is critical. The netter should alert the person rowing of hidden dangers and the person rowing should alert the netter of dangers as well. Always steer clear of potentially hazardous areas where the safety of the crew and boat may be jeopardized. Safe operation of the boat or raft and crew should be the top priority.

There may be occasions when the crew is faced with working in inclement weather or under adverse conditions. Mild drizzle, light rain, or snow usually poses little threat to the crew. Electrofishing can proceed as long as the vital electrical components are protected from the rain; however, should heavy rains form a continuous sheet of water on the boat or raft, crew, and electrical components, electrofishing should be discontinued until more favorable conditions develop. Electrofishing trips should be aborted anytime a thunderstorm produces lightning in the general vicinity of the crew.

Safe towing of the boat or raft to and from work locations should also be a concern of the crew. Make sure the trailer is in good operating condition and that it is properly hitched to the tow vehicle with the ball, hitch, light wiring, and safety chains. The trailer wheel bearings should be checked and greased frequently. The tread wear and tire pressure on the trailer tires should be carefully monitored while traveling. The operation of the trailer lights should be periodically checked. Finally, remember that a trailer extends the length of the tow vehicle and requires a greater stopping distance, wider cornering radius, and generally more cautious driving.

Fish Handling

Fish handling procedures are a critical element of sampling, especially in systems with threatened or endangered species. As researchers and managers, we are obliged to handle the fish we are sampling responsibly (Nickum et al. 2004). This is an important topic that should be considered by all practitioners sampling fish. Detailed fish handing guidelines are available from Nickum et al. 2004, the Canadian Council on Animal Care (CCAC) 2005, and Stickney (1983).

When mark–recapture sampling, it is critical that handled and marked fish return to normal activities before the recapture sample begins to help satisfy the equal catchability assumption associated with mark–recapture estimators. There are several examples of things we can do to reduce the handling stress of sampled fish (see Table 12). For instance, the zone 0.5 m from an electrofishing anode has been termed the potential zone of injury; thus fish in close proximity to the anode should be quickly removed from it while electrofishing (NMFS 2000). After removal from the water, captured fish should be held for the shortest time possible in live wells with recirculated or aerated water that has approximately the same temperature as the ambient water temperature they were removed from. An innovative live well design used for holding captured stream fish was proposed by Isaak and Hubert (1997) for stream sampling and by Sharber and Carothers (1987) for boat or raft units. Perhaps the most important consideration is to minimize handling of fish during any research or sampling activity.

TABLE 12. — Examples of activities that help minimize stress in sampled fish

Use approved anesthetic when handling.
Use aerated live wells or frequently change water.
Use live well colors that mimic the natural environment and include cover in them (e.g. rocks, leaves).
Minimize handling time.
Minimize sampling at water temperatures above 18°C.
Avoid overcrowding fish in live wells.
Avoid drastic differences in water temperature between live wells and ambient stream water.
Segregate large predatory fish from small fish to prevent predation in live wells.

Electrofishing Injury

Concerns over the potential for electrofishing to harm fish populations, particularly in areas with endangered species, have been presented in the literature (Snyder 1995; Nielsen 1998). It is clear that electrofishing can harm individual fish, particularly large individuals or susceptible life stages or species (McMichael et al. 1998). Injuries can range from bruising to broken backs; however, in most cases, the proportion of fish within populations that are actually sampled is small, and even large injury rates at the sample scale may be acceptable for monitoring and conservation-oriented studies (McMichael et al. 1998). Furthermore, electrofishing-induced injury rates evaluated from the population level may be negligible when considering natural mortality rates of stream fish (Schill and Beland 1995). Repeated electrofishing from long-term monitoring did not appear to have detrimental population level effects on salmonids in small Colorado streams (Kocovsky et al. 1997). As with any sampling technique, the value gained by sampling must be weighed against the ecological impact to the population.

Data Analysis

There has been much literature published on the theoretical and mathematical properties of both open- and closed-model mark–recapture estimators, including the classic works of Ricker (1975), Otis et al. (1978), and Seber (1986, 1992). Schwarz and Seber (1999), building on the work of Seber (1982, 1986, 1992), provide a comprehensive review of the literature related to estimation of animal population parameters. There have been many statistical advances in evaluations of mark–recapture data through the years. Many recent advances include variations on the basic mark–recapture abundance estimator to allow for various violations of underlying estimator assumptions. Discussing the statistical developments associated with evaluating mark–recapture data is beyond the scope of this protocol. We refer readers to the detailed review and references in Schwarz and Seber (1999).

We will provide the formula for the basic Petersen abundance estimator so that field practitioners without access to the cited references or computer software can calculate an abundance estimate based on simple mark–recapture field data. We direct readers to Ricker (1975) for more detailed discussion of the Petersen estimator. We would also like to make readers aware that the Petersen estimator is a simple case (approximation) of what are known as maximum likelihood estimates (White et al. 1982). When more than a single marking and a single recapture sampling occasion are performed, abundance estimates cannot be computed with the simple form of the Petersen estimator presented and iterative calculus techniques and likelihood functions should be used to calculate abundance. We refer readers to Otis et al. (1978) for detailed discussions regarding likelihood functions and maximum likelihood estimation. Researchers from Montana Fish, Wildlife and Parks have generated easy-to-use software that will calculate likelihood estimates quickly and efficiently (see Table 13).

The basic premise of mark–recapture estimates is that the ratio of marked/unmarked fish collected in a sample where M fish are marked is the same as the ratio of marked fish in the total population (M/N). An estimate of abundance from mark–recapture data can therefore be calculated from equation 2,

$$N = MC/R \tag{eq 2}$$

where N = the population estimate, M = number of fish marked during the mark run(s), C is the total number of fish in the recapture sample(s), and R is the number of marked fish captured in the recapture sample.

Bailey (1951) and Chapman (1951) presented mathematical corrections to the Petersen estimate when it was recognized that it may be biased when sample sizes are low. Chapman's modification of the Petersen estimate is provided in equation 3:

$$N = \frac{(M+1)(C+1)}{(R+1)} - 1 \tag{eq 3}$$

Robson and Reiger (1964) suggest that an unbiased estimate of *N* can be generated from the Chapman modification of the Petersen estimate when one or both of the following conditions are met:

1. The number of marked fish *M* plus the number of fish captured during the recapture sample *C* must be greater than or equal to the estimated population *N*.
2. The number of marked fish *M* multiplied by the number of fish taken during the recapture sample *C* must be greater than four times the estimated population *N*.

Calculations providing approximate 95% confidence intervals in the population estimate (*N*) are summarized by Vincent (1971) and can be calculated using equations 4 and 5:

$$Estimate \pm 2\sqrt{Variance} \qquad \text{(eq 4)}$$

where

$$\text{Variance} = \frac{(Population\ Estimate)^2(C - R)}{(C + 1)(R + 2)} \qquad \text{(eq 5)}$$

Equations 2–5 allow for hand calculation of population abundance estimates and confidence limits when all assumptions are met during sampling. There are many more complex estimators that can be used to estimate population abundance when assumptions cannot be tested in field settings due to budget or time constraints. Many of these estimators are available through Internet resources and are often free or inexpensive (see Table 13). These resources contain information as well as Internet links to computer software programs for estimating various population and community parameters beyond simple abundance calculations. Many computer programs contain complex procedures that will select appropriate population estimators based on the observed field data. Other programs use iterative calculus techniques to produce maximum likelihood estimates based on observed mark–recapture data.

TABLE 13. — Population analysis online information and software links available on the Internet

Web address	Affiliation
www.mbr-pwrc.usgs.gov/software	U.S. Geological Survey
www.warnercnr.colostate.edu/~gwhite/software.html	Colorado State University
www.fisheries.org/units/cus	American Fisheries Society computer user section
www.cs.umanitoba.ca/~popan/	University of Manitoba
http://fwp.state.mt.us/default.html	Montana Fish, Wildlife and Parks

Closing Remarks

Electrofishing will likely continue to be one of the dominant methods of sampling freshwater fishes until improved sampling techniques are developed. Although the practice of electrofishing is relatively easy, designing the right approach to achieve objectives is quite complicated. Many trade-offs and assumptions must be evaluated to ensure that the best design can be implemented. In addition, flexibility to make improvements in long-term monitoring should be anticipated as new information, techniques, and equipment become available. This may involve implementing new methods and conventional methods at the same time so that historical data sets can be calibrated to new ones.

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Appendix A: Field data sheet for backpack electrofishing

pass	time	segment	spp	age: YJA	##	length	weight	mark	M	BR	OS	BS	G	R	S	ECU	ECL	IHK	IEF	IO	HS	SCB	SCO	EIL	EIR	EPF	EPG	comments

Notes:

Minnows:	CH M	LND	SPD
	RSS	NPM	
Sculpins:	MSC	PSC	SSC
	TSC		
Suckers:	BLS	LSS	MNS
Salmonids:	RBT	CUT	BUL
	EBT	SPC	COH
	MW F		

M	-	mortality	IEF	-	injury / electrofishing
BR	-	bruise	IO	-	injury / other
OS	-	orange slash	HS	-	hooking scar
BS	-	black spot disease	SCB	-	scar / bird
G	-	green	SCO	-	scar / other
R	-	ripe	EIL	-	eye injury / left
S	-	spent	EIR	-	eye injury / right
ECU	-	eroded caudal / upper	EPF	-	external parasites / fins
ECL	-	eroded caudal / lower	EPG	-	external parasites / gills
IHK	-	injury / hooking			

INITIALS:

H2O TEMP:_______C@____:____(start) ______C@____:____(noon) ______mmhosmhos Page___ of ___

______C@____:____(end)

Appendix B: Electrofishing mark–recapture datasheet

Mark - Recapture Electrofishing Form

WDFW- Ecological Interactions Team (YSIS, rev. 9/99)

STREAM: ______________________________ SECTION: ________________

DATE: ______/______/______ FLOW: *L / M / H* INITIALS: __________ PAGE: _______

TRIP TYPE: *MARK / RECAP , LEFT / RIGHT* WATER TEMP: _______ @ _____:______ _________ mmhos

	SPP	FL mm	WT g	MARK CODE*	MARK TYPE**	SCALES	TAG NUMBER	REMARK CODES	COMMENT
1									
2									
3									
4									
5									
6									
7									
8									
9									
10									
11									
12									
13									
14									
15									
16									
17									
18									
19									
20									
21									
22									
23									
24									
25									

* MARK CODE:
- 0 = UNMARKED
- 1 = MARKED

** MARK TYPE:
- LC = LOWER CAUDAL
- UC = UPPER CAUDAL
- AN = ANAL
- LV = LEFT VENTRAL
- RV = RIGHT VENTRAL

REMARK CODES >>
- M = mortality
- BR = bruise
- OS = orange slash
- I_ = injury + EF(=electrofishing) / O(ther)
- SC_ = scar + B(ird) / O(ther)
- EI_ = eye injury + L(eft) / R(ight)
- EP_ = external parasites + F(ins) / G(ills)
- EC_ = eroded caudal + U(pper) / L(ower)
- HS_ = hooking scar + L(eft) / R(ight)
- G_ = green + M(ale) / F(emale)
- R_ = ripe + M(ale) / F(emale)
- S_ = spent + M(ale) / F(emale)

// AT WORK-UP STATION,
// RECORD THE STATION
// NUMBER AND E.F. TIME
// IN COMMENTS AS:
// **"sta # (####)"**