Protocols for Monitoring the Response of Anadromous Salmon and Steelhead to Watershed Restoration in California

February 13, 2006

Prepared for:

California Department of Fish and Game
Salmon and Steelhead Trout Restoration Account
Agreement No. P0210565

Walter G. Duffy
California Cooperative Fish Research Unit
Humboldt State University
Arcata, CA 95521

TABLE OF CONTENTS

TABLE OF CONTENTS (Continued) **Page**

TABLE OF CONTENTS (Concluded) **Page**

LIST OF TABLES

List of Figures

INTRODUCTION

The goal of California Department of Fish and Game, Fisheries Restoration Grant Program (FRGP) is to assist in the recovery of salmon and steelhead trout populations. The desire to restore salmon and steelhead trout populations in California watersheds represents a sweeping societal challenge. Achieving this goal will require a process to identify and guide restoration actions and the resources needed to carry them out. Ideally, the process of identifying and guiding restoration actions would; 1) identify the types of restoration actions needed to improve stream habitat for salmon and steelhead trout, 2) identify where restoration actions may be most effective, 3) determine how much restoration is required to bring about a population response within watersheds or sub-watersheds, and 4) implement procedures for evaluating the success of restoration actions in meeting objectives.

Watershed restoration may be defined as any action that starts or accelerates the recovery of a watershed toward its pre-disturbance trajectory (SER 20002). Trajectory here implies some trend in biological and physical composition, processes and functions. Although the ideal pre-disturbance trajectory may be interpreted as the historical condition, resetting watershed functions to the historical condition is often unrealistic and goals are more commonly defined in the context of existing reference conditions.

A restored watershed has been defined as one that "…contains sufficient biotic and abiotic resources to continue its development without further assistance of subsidy" (SER 2002). This definition does not mean the watershed has been returned to a pristine condition. Rather, it means the watershed has recovered enough to be resilient to periodic disturbances such as floods or fires and that it interacts with the surrounding ecosystem. Some characteristics of functioning or restored watersheds are that they, contain species assemblages similar to a reference watershed, have primarily indigenous species, contain all the functional groups needed for continued functioning and development, and have physical habitat adequate for sustaining naturally reproducing populations.

Goals or end points for watershed restoration cannot be successfully established using intuition or seat-of-the-pants reasoning. Restoration goals should be developed with careful consideration of societal wishes and economic realities. They should be guided by science, that is, restoration goals should be the product of a watershed assessment or characterization that provides a basis for comparing current condition with the desired condition (Bohn and Kershner 2002, Palmer et al. 2005). Finally, goals set should be realistic and attainable within a defined time period.

In this report we describe protocols for validation monitoring to evaluate the outcome of restoration actions in meeting California coastal watershed restoration program objectives. Three monitoring activities are commonly recognized (ONRC 2000). Implementation monitoring is monitoring to document the fulfillment of contract obligations or compliance with regulations or laws. Effectiveness monitoring is monitoring to document trends in resource condition following a management action. Effectiveness monitoring is most often associated with physical or chemical processes and habitats. Validation monitoring is monitoring to document the response of biota to restoration actions. Validation monitoring, ideally, establishes cause-and-effect relationships between restoration actions and biota (ONRC 2000). However, lack of pre-project monitoring data or inadequate replication limits the ability of validation monitoring in establishing cause-and-effect relationships. Validation monitoring differs from implementation and effectiveness monitoring in that it is primarily concerned with the response of biota, as opposed to physical habitats or physical processes. The time required to document pre-restoration condition or change after restoration varies with the species being monitored, the biological measure being used and number of replicate samples. In general terms, documenting pre-restoration condition for most fish response measures will require one or more years of sampling while documenting post-restoration change will require multiple years.

This report presents recommendations for validation monitoring protocols intended to detect responses of salmon and steelhead trout to watershed restoration actions. The question guiding selection of protocols was: what measurements are both practical and sensitive enough to detect a response by salmon and steelhead trout to restoration actions? The assumption inherent in this question is that salmon and steelhead trout will respond to watershed restoration actions.

Protocols recommended in this report are not comprehensive. Rather, our protocol selection was guided by the watershed restoration program goal of restoring salmon and steelhead trout, with consideration of the varied types of restoration actions.

The freshwater fish community of coastal California is more diverse than is often recognized. Monitoring programs instituted by the California Department of Fish and Game provide an opportunity to gather information on native species, as well as alien species. Accurate records on the distribution of fish species would better inform managers about a range of topics, for example; recovery of listed species, the introduction or range expansion of alien species. To be most useful, these species records will require quality-assurance and quality-control procedures, including taxonomic verification of some specimens.

3. Taxonomic Quality Control and Quality Assurance

We recommend that independent verification of taxonomic determinations be sought for some fishes collected in the course of monitoring (Walsh and Meador 1998). Independent determinations should be sought when:

1. Field personnel lack experience in fish taxonomy.
2. Species are collected that cannot be reliably identified in the field.
3. Species that represent new distribution records for a watershed.

Photographic documentation of fish species should be included as part of the monitoring QA/QC program and can sometimes provide for independent taxonomic determinations. Problematic species, particularly some alien fishes, should be preserved in a 10% formalin solution after the photographic record is made. Independent taxonomic determination of these preserved specimens can then be sought from one of the ichthyological curation facilities listed below.

California Academy of Sciences
Department of Ichthyology
Golden Gate Park
San Francisco, California 94118

Humboldt State University
Fisheries Biology Department
Ichthyology Collection
Arcata, California 95521

Natural History Museum of Los Angeles County
Ichthyology Section
900 Exposition Boulevard
Los Angeles, California 90007

2. Collecting Permit Requirements

Collecting fishes in California for any purpose requires a Department of Fish and Game Scientific Collectors Permit. For collecting certain species, a memorandum of understanding or other written permission may also be required . Permit application procedures, costs and reporting requirements may be obtained from:

California Department of Fish and Game
License and Revenue Branch
3211 S Street
Sacramento, California 95816-7088

Additional federal permits are required if sampling is targeting species listed as threatened or endangered under the Endangered Species Act or if there is a likelihood of capturing federally listed species during sampling and if sampling is to be conducted on state or federal lands. Federal collecting permits for anadromous species are administered by the National Marine Fisheries Service. Questions regarding permits may be obtained from:

Chief, Protected Resources Division
National Marine Fisheries Service - F/SWR3
777 Sonoma Avenue, Room 325
Santa Rosa, CA 95404
Phone: (707) 575-6050
http://www.nmfs.noaa.gov/pr/permits/esa_permits.htm

Federal collecting permits for freshwater and terrestrial species are administered by the U. S. Fish and Wildlife Service (USFWS). Questions regarding permits may be obtained from:

U.S. Fish and Wildlife Service, Endangered Species Permit Office, 911 N.E. 11th Ave., Portland, OR 97232-4181

Web: http://pacific.fws.gov/ecoservices/
Phone: 503-231-2071
email: permitsR1ES@fws.gov

Local offices of the California Department of Fish and Game can often provide contact information for requesting collecting permits under these conditions. Access agreements are often necessary when sampling on private lands and must be sought from property owners.

JUVENILE SALMON AND STEELHEAD ABUNDANCE AND POPULATION SIZE

1. Rational

Abundance and population size are terms used, in fisheries biology, to express two similar but different measures. Abundance refers to the number of fish sampled in an area. When expressed as the number of fish observed or captured per unit area, abundance may also be referred to as density. Abundance is also expressed as the catch given some standardized unit of effort (CPUE), for example the catch per hour of electrofishing.

Population size refers to the number of fish of a particular species occupying a geographic area. The geographic area occupied by a population is usually an entire stream or watershed, although large watersheds may have more than one population. Estimates of population size could be obtained from sampling the entire area of interest, but this is not practical. Population size is instead estimated by sampling a statistically selected sub-sample from those habitats available, then extrapolating density to the total area of habitat.

The number of juvenile salmon or steelhead present in a stream or stream reach often requires less effort than estimating abundance of other life history stages, such as adults, smolts or eggs. For example: all field sampling to estimate juvenile coho salmon population size in a six km reach of Prairie Creek in Humboldt County required about 530 person hours, while weekly sampling to estimate adult escapement required about 900 person hours and daily smolt trapping during February to June required about 8,400 person hours. Measurements of the number of juvenile salmon or steelhead present in a stream also provides several types of information useful to monitoring:

- When measured over multiple years, trends juvenile salmon or steelhead abundance provide information on the response of juvenile salmonids to habitat change and environmental conditions.

- When combined with estimates of the number of adults spawning the previous season, abundance of juvenile salmon and steelhead can provide information on survival from the egg to juvenile period.

- When combined with estimates of the number of smolts migrating from a stream, data on abundance of juvenile salmon and steelhead can provide information on survival during the entire juvenile period.

Methods described here are intended to provide information on juvenile coho salmon or steelhead abundance within streams or stream reaches. These abundance estimates can be expanded to the watershed scale to provide population estimates. Most Chinook salmon in California streams migrate to the estuary soon after hatching and do not occupy stream habitats for an extended period. Abundance estimates require less rigorous sampling and are usually better suited to monitoring population trends or the response of a watershed to management actions. For example, measuring change in the abundance of juvenile salmonids over time. More rigorous sampling for population estimates is required when comparisons of survival at distinct life stages is desirable.

2. Assumptions

The method described here employs both diver observation and electro-fishing techniques. The primary assumption inherent in this method is that fish are susceptible to the gear. For divers, susceptibility means that fish are visible to divers and that divers can accurately identify and count species. In electro-fishing, susceptibility means both that the gear is efficient in temporarily stunning fish and that field personnel are efficient in capturing fish stunned by electrical current. Furthermore, the method assumes that diver observations and electro-fishing estimates are correlated. These assumptions are not always met (Peterson et al. 2004). Environmental conditions such as turbidity, specific conduction, water temperature, complexity of the habitat, light and other factors can influence efficiency of both diver observations and electro-fishing capture.

3. Limitations

Methods described here are intended for small – medium size streams in which most pools (>75%) are <1.1m in deep and the stream has a wetted perimeter of $\leq$ 10 m. Water in streams must also allow divers to see fish clearly at 3-5 m if visual counts of juvenile salmonids are to be considered reliable.

These conditions are necessary for two divers to effectively sample a stream. Streams that are too large to be sampled with snorkeling should be sampled with electro-fishing equipment. Similarly, streams too small to dive or in which the visibility is limited should be sampled with electro-fishing equipment.

In California, we recommend sampling during August – October. Sampling during late summer through early fall will increase the likelihood that assumptions and limitations involved with methods are met. During late summer – early fall, water clarity in California streams is greatest and juvenile coho salmon and steelhead are large enough to be visually located and distinguished.

4. Sampling Design

The design of a sampling program to estimate fish abundance should incorporate random selection of sampling sites. The design recommended here incorporates systematic random sample selection, stratified by habitat type. Systematic random sample selection is relatively simple and the calculations required to estimate either abundance or population size are not cumbersome. This sampling design may be applied to stream reaches, sub-watersheds or smaller watersheds.

Sampling designs for large watershed, regional or state wide monitoring programs often employ techniques other than systematic random sampling for selecting a statistically valid random sample. The chief reason for these more elegant approaches is that, in sampling over large areas, it is impractical to define all the possible habitat units that could be sampled. Instead, these techniques are usually designed to randomly select sampling points from information in geographic information databases.

5. Methods

A. Estimating Abundance

Estimating the abundance of fish in an area requires information on the habitat and fish. This information is gathered in two steps. First, the habitat available to be occupied is classified and measured. Second, the fish using those habitats are sampled.

Measuring habitats

A minimum of two people are needed to classify and measure habitat units. Habitat measurements should be completed one – two weeks before fish sampling. A longer interval between measuring habitats and sampling fish may result in habitat depths and areas changing before fish are sampled. In measuring habitats, one person carries a hip-chain to measure linear distance from the starting point and a stadia rod to measure width of the habitat units and their depth, if desired. A second person records data. All habitat units within the stream or stream reach in which abundance estimates are to be made must be classified and measured.

Individual habitat units are classified as either runs, riffles, pools, deep pools or other habitats, equivalent to level II habitat typing in Flosie et al. (2002). Each habitat unit must be longer than its average width. It should be separated from neighboring habitat units by a distinct hydraulic break so that movement of fish between units during the dive survey is limited. Habitat units that appear to be comprised of two habitat types should be classified to reflect the majority of the unit. General definitions of habitat types for fish sampling adopted from Flosi et al. (2002) are:

Pool (P) - a scoured habitat unit with slow currents, little surface turbulence, and maximum depth $\leq$ 1.1 m. Pools > 1.1 m deep are considered deep pools.

Riffle (R) – habitats with fast-flowing water and substrate breaking the surface, causing surface turbulence. Riffle habitats are too shallow to dive.

Figure 1. Example of pool and riffle habitat.

Run (N) – quickly flowing water having little surface agitation and few occurrences of substrate breaking the surface. In defining habitat for fish sampling, we recommend combining glide and run habitats as defined by Flosi et al. (2000). Run habitats have a minimum of 60% of their area in water ≥ 40 cm deep.

Figure 2. Example of run habitat.

Cascade (C) – habitat units having sharp gradient changes that create turbulent water. Cascades typically produce bubble curtains that important habitat for juvenile steelhead.

Figure 3. Example of cascade habitat

Other (O) – other habitats are those that present features that make either snorkel observations or electro-fishing difficult. For example; side channel habitats may be small and shallow relative to the main channel, or habitats having complex structures that present obstacles to visual recording or netting fish.

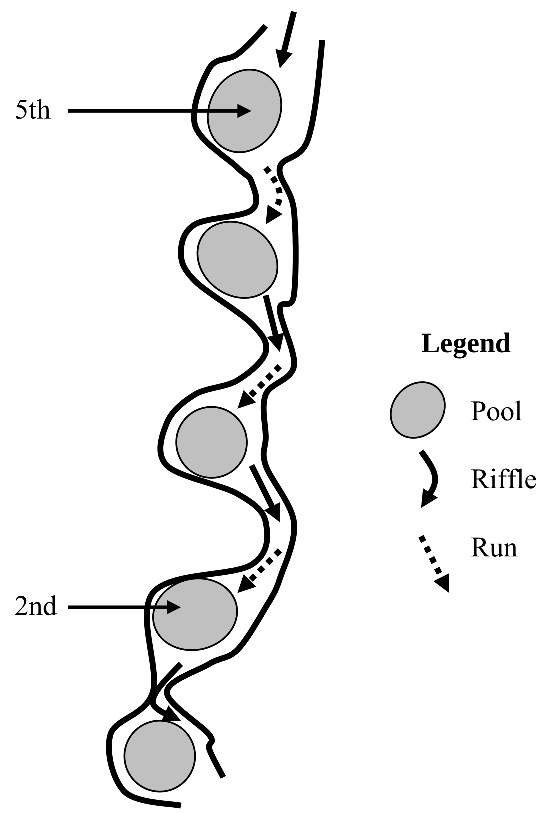
This classification of habitats represents a minimum in complexity. Other habitat types can be included if necessary to meet objectives of specific monitoring projects.

Habitat unit length, width, and depth are recorded on the data sheet (Appendix table 1) in numerical sequential order (NSO) from the downstream starting point. Each NSO number can then be associated with a specific habitat unit.

Time and effort of measuring habitats can be reduced by visually estimating surface area of the habitat. If visual estimation is used, accurate measurements should be recorded on subset of the total of each habitat type. This can be accomplished by systematic random sampling:

Steps in systematic sampling if 33% of the total habitat units are selected for accurate measurement.

Step 1. For each habitat type, first draw a random starting number between 1 and 3. For example, assume the starting random number for pool habitats was 2. From the illustration below then, accurate measurements should be recorded on the 2nd and 5th pools corresponding to NSO 3 and 11. This process would be repeated until the survey was completed. A separate random starting number would be drawn for riffles and runs, and the process repeated. .



~~Step 1~~Step 2 Visually estimate and record the area of all habitat units.

~~Step 1~~Step 3 Physically measure and record the area and depth of those habitat units selected at random.

Step 4. Calculate a calibration ratio ($\hat{Q}$) using at least 10 habitat units:

$$\hat{Q} = \frac{\sum_{i=1}^{n} m_i}{\sum_{i=1}^{n} x_i}$$

Where m_i = the accurate measurement of habitat area and x_i = the visual estimate of habitat area.

Step 5. The total area of each habitat type ($\hat{M}$) may then be estimated from:

$\hat{M} = T_x \hat{Q}$, where the sum of all visual habitat estimates is $T_x = \sum_{i=1}^{N} x_i$ and N = the total number of units of a particular habitat type.

Step 6. The variance ($\hat{V}(\hat{M})$, a measure of uncertainty) of the estimated total habitat type can then be calculated from:

$$\hat{V}(\hat{M}) \cong \frac{N(N-n)}{n(n-1)} \sum_{i=1}^{n} (m_i - \hat{Q} x_i)^2$$

Where n = sample size or number of accurately measured habitats.

Accurate measurement of habitat units should follow standardized procedures. We recommend measuring width at 3 or 4 intervals on simple habitats. Measurement interval may require adjustment on irregularly shaped habitat units. Use multiple width measurements to calculate average width, and multiply average width by habitat length to obtain surface area.

Conducting the Fish Census

The primary sampling method recommended for counting juvenile coho salmon and steelhead is visual observation using snorkel gear. This method is less costly and intrusive than electrofishing. However, visual observation techniques are not possible in all types of habitats, nor are they applicable in some streams. Electro-fishing is recommended in situations where visual observation is either not possible or would provide inaccurate results. Methods for electro-fishing are described later in this section.

Visual observation may be used to sample run, pool, and deep pool habitats. A systematic random sample of each habitat type should be drawn from the total of habitat units measured (Hankin and Reeves 1988). The proportion of units selected for sampling can differ among habitat types and may be adjusted to reflect project objectives (Doloff et al. 1993). For example, sampling that targets juvenile coho salmon could include 25% of pool and run habitats, but only 10% of riffle habitats since few coho salmon are found in riffles. Increasing the percentage of any habitat type sample will obviously improve precision of the estimated population size, but at the cost of greater labor. Selection of fish sampling units may be carried out using the methods described in box 5.1.

The proportion of habitat units to be sampled should be determined before habitats are surveyed. Then the upper and lower boundaries of habitat units selected for later fish sampling can be marked with flagging during habitat surveys. Having habitat surveyors delineate those habitats to be sampled for fish minimizes uncertainty in later locating specific habitat units and delineating their boundaries.

Two pool or run habitat units outside the area to be sampled should be identified for practice. Snorkel divers should survey these habitats before starting the fish survey. These practice habitats allow the divers' to familiarize themselves with the species and size classes of salmonids they will likely encounter in subsequent habitats. Age and size classes of salmonids can very among streams during any season because of differences in time of emergence and growth.

Identification of all species can be problematic within the range of coastal cutthroat trout. Juvenile steelhead and cutthroat trout cannot be consistently distinguished until the reach a length of around 80 mm fork length. The distribution of coastal cutthroat trout in California extends from the Eel River in Humboldt County, north to the Oregon border. Their distribution extends inland about 10 km at the southern end of this range to about 40 km inland along the border with Oregon (Gerstung 1997). Thus, from the Eel River northward, small trout should be counted as age 0+ trout species. Steelhead and cutthroat trout > 80 mm FL can usually be assigned to age 1+ of their species. However, these species should be recorded as age 1+ trout if divers are not confident in their ability to separate these species.

The fish census is conducted primarily by visual observation using snorkeling, with limited electro-fishing. Visual observations of pre-selected pool, deep pool and run habitats are conducted, progressing from downstream to upstream. Divers should enter the downstream end the habitat unit to be surveyed. They should move upstream, parallel to one another, through the habitat unit using deliberate movements so as to minimize disturbance to fish. It is important for divers to observe both the edges of the stream where cover may be present and the middle of the stream. If two divers cannot observe both the steam edges and center, an additional diver may be required. In some habitats, water velocity may be too great for divers to swim upstream. Where this occurs, divers should count fish while swimming downstream. Fish are counted as divers move through the habitat and recorded using either a hand counter or underwater record slate. Using a recording device is especially important where fish are abundant and where multiple species occur. After completing the census for a specific habitat unit, data are recorded in small "Write-in-the-Rain" or plastic paper notebooks than can be carried in a dive pouch.

Visual observation methods are not possible in riffle habitats and may not be effective for entire reaches of some shallow streams. Furthermore, cobble and other obstructions in riffle and other shallow habitats also make seine netting inefficient. These habitats must be sampled using electro-fishing techniques.

Sampling with electro-fishing techniques requires a minimum of two or three people. One person carries the backpack electrofishing unit, while others net fish that are stunned by the electrical current. Specific conductance and temperature of the water should be measured and recorded before sampling (see Box 5.2 for guidelines on water temperature and specific conductance). Specific conductance provides information on how well water will conduct an electrical current and should be used in selecting electrofisher settings. Before sampling, a fine mesh net should be stretched across the downstream end of the habitat unit. This net serves to block stunned fish that may float downstream so that they may be captured and properly revived before release.

As with visual observations, the electrofishing crew enters a habitat unit at the downstream end and proceeds upstream. The area of the habitat unit should be electrofished thoroughly, but excessive time should not be spent in small areas due to potential harm of exposing fish to the electrical field for extended periods (NOAA 2000). Fish that are stunned should be removed from the electrical field as quickly as possible and placed in a bucket containing fresh stream water. This sampling process is considered one pass and is repeated a second time after any turbidity created during the first electrofishing has cleared and visibility is restored. If the number of target species (juvenile coho salmon or steelhead) caught in this second pass is 20% or less than was caught in the first pass, sampling the habitat unit is complete. If, however, the number of the target species caught during the second pass is 21% or more than was caught on the first pass, a third pass of electrofishing is conducted. Sampling is terminated after three electrofishing passes.

After the habitat unit has been completely sampled, all fish collected are identified, counted and biological data are recorded. Biological data that should be recorded includes fork length (mm) and live weight (g) should from 100 or more specimens of each species from each sub-watershed. In addition, scale samples should be collected from 100 or more juvenile steelhead. After recording these data from the fish captured, they should be allowed to recover, then released in the vicinity of their capture. These biological data will allow for further assessment of either condition or age structure of the population (described later in this report). Although this and other protocols described are intended to document the response of salmon and steelhead to watershed restoration actions, recording data on all fish species captured is also recommended. California's coastal rivers support a variety of species (Appendix B). Information on their presence, abundance and basic biological information such as size contribute to better management of California fisheries. Moyle (2002) is a useful guide to identifying freshwater fishes found in California waters.

Numbers of juvenile salmonids observed during visual surveys and captured during multiple-pass electrofishing can be used to provide and index of abundance. When divided by the area of habitat sampled, this index of abundance can be expressed as a density estimate (number/m^2). However, neither is equivalent with a population estimate.

B. Estimating Population Size

Estimating the size of a juvenile salmonid population requires additional sampling and analysis. The additional sampling is essentially devoted to validating assumptions about the efficiency of visual observations (Hankin and Reeves 1988). Added analyses are needed to extrapolate estimates from a sub-sample of habitats to the entire area represented by that type of habitat.

While the calculations presented seem tedious, they are needed to produce a statistically valid population estimate and satisfy the assumptions of sampling theory. Variance estimators for the mean density of fish in a habitat and the total number of fish per habitat type are provided in Hankin and Reeves (1988) or Dolloff et al. (1993). These same variance estimators are included in a spreadsheet for calculating abundance estimates using the approach presented here, the spreadsheet is available at the following California Department of Fish and Game website,
http://www.dfg.ca.gov/nafwb/pubs.html.

Calculating estimated abundance or population size from systematic random samples.

Step 1. Calculate a ratio for calibrating the diver visual observations with the more accurate electrofishing samples results:

$$\hat{R} = \frac{\sum_{i=1}^{n'} y_i}{\sum_{i=1}^{n'} x_i}$$

where the sum applies to i = 1 – n’ and n’ = the number of habitat units in which both diver counts and electrofishing estimates are made. The values for y_i are the number of fish collected with electrofishing and x'_i are the mean of counts by two divers in habitat units sampled by both electrofishing and diving.

Step 2. The density of fish in a habitat unit ($x'_{i,HA}$) is calculated as the number of fish observed in the habitat unit i (x'_i) during snorkeling divided by the are of habitat unit i (H_A) in m^2:

$$x'_{i,HA} = \frac{x_i}{H_{Ai}}$$

Step 3. The estimated mean density of fish per habitat is calculated as the product of the calibration ratio and mean density of fish from diver counts:

$$\bar{y}_{dr} = R(\bar{x})$$

where $\bar{x} = \frac{\sum_{i=1}^{n} x'_i}{n}$, and n = the total number of units sampled by the divers.

Step 4. The total number of fish in a habitat type (Ŷ) is estimated as the product of the mean density per habitat total area of the habitat type:

$$\hat{Y} = N(\bar{y}_{dr})$$

6. Data Analysis

Example:

Table 1. Example of calculating the adjusted habitat area for a sample 30 pools in which habitat area was visually estimated (x_i) and a random sample of 33% were physically measured (m_i).

NSO	Estimated Area	Estimated Area x_i	Measured Area m_i	NSO	Estimated Area	Estimated Area x_i	Measured Area m_i
1	1.9			16	11.8		
2	4.4			17	14.4		
3	6.4	6.4	7.1	18	20.7	20.7	22.1
4	4.8			19	14.7		
5	5.0			20	16.4		
6	11.8	11.8	13.2	21	21.4	21.4	22.2
7	8.6			22	16.5		
8	8.9			23	16.7		
9	13.8	13.8	15.0	24	25.2	25.2	26.6
10	9.0			25	17.7		
11	10.8			26	18.4		
12	14.7	14.7	16.3	27	25.0	25.0	26.7
13	10.9			28	21.8		
14	11.3			29	24.1		
15	16.6	16.6	18.4	30	26.7	26.7	28.7

Using the area of habitats those that were both estimated and measured, divide the sum of the estimated area into the measured area to calculate a calibration ratio:

Estimated area of habitats measured = 182.3 m^2
Actual area of habitats measured = 196.3 m^2
$\hat{Q}$ = 182.3 m^2/196.3 m^2 = 0.93

Next, sum the estimated area each of the 30 pools yields the estimated area in pools, (T_x);

T_x = 430.5.

Multiply (T_x)($\hat{Q}$) to calculate the total area in pools ($\hat{M}_{pools}$),

$\hat{M}_{pools}$ = (430.4 m^2)(Q), = (430.5 m^2)(1.08), = 463.6 m^2.

An approximate variance for the area in pools can be calculated as;

$$\text{Variance} = \hat{V}\left(\hat{M}\right) \approx \frac{N(N-n)}{n(n-1)} \sum_{i=1}^{n} (m_i - \hat{Q}x_i)^2$$

$$= \hat{V}\left(\hat{M}\right) \approx \frac{30(30-10)}{10(10-1)} (2.1)$$

$$= 14.1 \text{ m}^2$$

where $\left(m_i - \hat{Q}x_i\right)^2$ is the difference in measured area $\left(m_i\right)$ and predicted area $\left(\hat{Q}x_i\right)$ of habitat units.

The variance term and Students-t value for n-1 habitat units measured can then be used to calculate confidence intervals about the area of habitat.

$$\text{Area} \pm 95\% \text{ Confidence intervals} = \hat{M} \pm t_{0.05;n-1} \sqrt{\hat{V}\left(\hat{M}\right)}$$

$$= 463.6 \pm 182.9 \text{ m}^2$$

Table 2. Example showing adjusted habitat area and hypothetical fish abundance data for 30 pool habitats from Table 1.

Adjusted Habitat Area $\hat{Q}x_i$	∑E-fish count y'	Average dive count x'	Adjusted Habitat Area $\hat{Q}x_i$	∑E-fish count y'	Average dive count x'
2.1	1	1	12.7		
4.7			15.6		
6.9			22.4		
5.2			15.9		
5.4	6	5	17.7	22	21
12.8			23.1		
9.2		7	17.8		
9.6		12	18.0	17	16
14.9			27.2		
9.7			19.1		
11.7			19.9		
15.8			27.0	25	24
11.8	20	19	23.5		24
12.2			26.0		
17.9			28.8		24

Table 3. Calculation of fish abundance parameters from data presented in Table 2.

		Pools
Calibration ratio	$\hat{R}$	0.805
Number of units sampled by both divers and efishing	$n_i^{'}$	6
Total number of fish collected by efishing in units n'	$y_i^{'}$	91
Total number of fish observed by divers in units n'	$x_i^{'}$	113
Number of units sampled by divers	n	10
Average density of fish observed by divers in units n	$\overline{x}$	0.98
Total area of units	N	464.6
Estimated average density of fish per habitat	$\overline{y}_{dr}$	0.8
Variance of density of fish in habitat type	$\hat{V}\left(\overline{y}_{dr}\right)$	0.041
Variance of number of fish in habitat type	$\hat{V}\left(\hat{Y}\right)$	8886
Student's t-value for number of units sampled by divers	$t_{0.05;\, n-1}$	2.262
95% Confidence intervals for total fish in habitat type	$\hat{Y} \pm t_{0.05;n-1}\sqrt{\hat{V}\left(\hat{Y}\right)}$	213
Estimated total number of fish in habitat type	$\hat{Y}$	368
Lower confidence interval	$\hat{Y}$ - 95% C.I.	155
Upper confidence interval	$\hat{Y}$ + 95% C.I.	581

7. Quality Control and Quality Assurance

Quality assurance and quality control procedures should be established before juvenile salmon and steelhead sampling. These procedures should include elements of the following:

Training that addresses,
1) safety practices in both stream snorkeling and electrofishing,
2) identification of fish species likely to be encountered,
3) 8 hours training in identifying fish when diving, and
4) proper handling of fish and

The quality assurance plan for data entry and management should include,
1) data entry
2) data management, including editing for errors
3) data analysis
4) chain of custody for data
5) backing up data in central repository

The assurance for fish sampling should include independent assessment of efficiency. This might include;

1) independent divers sampling a percentage of habitats previously sampled and
2) independent observers participating in electrofishing (we hesitate to recommend added electrofishing due to the potential for added stress on fish).

Data entry and management elements of QA/QC procedures should include the use of metric units of measure, proper use of measuring boards and balances, data coding of field sheets and data entry. Procedures to verify the accuracy of recorded field data and data entry into an electronic format should be developed. These typically involve an independent observer check 5 – 10% of the original entries. The entire data base should be checked for errors if this sample of original entries reveals a rate of error of more than 5%.

8. Personnel and Equipment Needed

For Snorkel Observations	
Personnel	Materials (continued)
Divers (2)	Rubber gloves
	Backpack
Materials	Dive slate or plastic sheets
Dry or wet suits	Underwater flash lights
Masks	Spare batteries
Snorkels	Small write-in-the-rain notebook
Hoods	Pencils
Booties	First aid kit
Wading boots	

For Electrofishing	
Personnel	Materials
Electro-fisher operators 1-2	Pencils
Netters 2-3	Data sheets
Data recorder 1	Chest waders
	Eye glasses with UV filter
Materials	Field clothing
1-2 Backpack electro-fishers	Anesthetic (Alka Seltzer, clove oil, or MS-222)
2-3 Dip nets	Metric measuring board
3-4 18 L (5 gal) plastic pails	Portable electronic balance (0.1 g resolution)
2 Block nets	First aid kit
Specific conductance meter	Rubber gloves
Thermometer	

9. Stream electrofishing guidelines (from NOA, 2000).

Initial Site Surveys and Equipment Settings

1. In order to avoid contact with spawning adults or active redds, researchers must conduct a careful visual survey of the area to be sampled before beginning electrofishing.

2. Prior to the start of sampling at a new location, water temperature and conductivity measurements should be taken to evaluate electroshocker settings and adjustments.

3. No electrofishing should occur when water temperatures are above 18°C or are expected to rise above this temperature prior to concluding the electrofishing survey. In addition, studies by NMFS scientists indicate that no electrofishing should occur in California coastal basins when conductivity is above 350 µS/cm.

4. Whenever possible, a block net should be placed below the area being sampled to capture stunned fish that may drift downstream.

5. Equipment must be in good working condition and operators should go through the manufacturer's preseason checks, adhere to all provisions, and record major maintenance work in a logbook.

6. Each electrofishing session must start with all settings (voltage, pulse width, and pulse rate) set to the minimums needed to capture fish. These settings should be gradually increased only to the point where fish are immobilized and captured, and generally not allowed to exceed conductivity-based maxima (Table 5.1). Only direct current (DC) or pulsed direct current (PDC) should be used.

Guidelines for initial and maximum settings for backpack electrofishing.

	Initial settings	Maximum	settings
Voltage	100 V	Conductivity (uS/cm)	Max. Voltage[2]
		<100	1100
		100–300	800
		>300	400
Pulse width	500 us		5 ms
Pulse rate[1]	30 Hz		70 Hz

[1] In general, pulse rates > 40 Hz will injure more fish than rates < 40 Hz.
[2] In California coastal streams, settings should never exceed 400 volts and electrofishing should not occur if conductivity is greater than 350 µS/cm.

~~1.~~7. Sampling should begin using straight DC. Remember that the power needs to remain on until the fish is netted when using straight DC. If fish capture is unsuccessful with initial low voltage, gradually increase voltage settings with straight DC.

Electrofishing guidelines (concluded).

Electrofishing Technique

~~1.~~8. If fish capture is not successful with the use of straight DC, then set the electrofisher to lower voltages with PDC. If fish capture is unsuccessful with low voltages, increase pulse width, voltage, and pulse frequency (duration, amplitude, and frequency).

~~2.~~9. Electrofishing should be performed in a manner that minimizes harm to the fish. Stream segments should be sampled systematically, moving the anode continuously in a herringbone pattern (where feasible) through the water. Voltage gradients may be high when electrodes are in shallow water where boundary layers (water surface and substrate) tend to intensify the electrical field.

~~3.~~10. Do not electrofish in one location for an extended period (e.g., undercut banks) and regularly check block nets for immobilized fish.

~~4.~~11. Fish should not make contact with the anode. Remember that the zone of potential injury for fish is 0.5 m from the anode.

~~5.~~12. Electrofishing crews should be generally observant of the condition of the fish and change or terminate sampling when experiencing problems with fish recovery time, banding, injury, mortality, or other indications of fish stress.

~~6.~~13. Netters should net fish quickly and not allow the fish to remain in the electrical field any longer than necessary.

Sample Processing and Recordkeeping

~~7.~~14. Fish should be processed as soon as possible after capture to minimize stress. This may require a larger crew size.

~~8.~~15. All sampling procedures must have a protocol for protecting held fish. Samplers must be aware of the conditions in the containers holding fish; air pumps, water transfers, etc., should be used as necessary to maintain safe conditions. Also, large fish should be kept separate from smaller prey-sized fish to avoid predation during containment.

~~9.~~16. Use of an approved anesthetic can reduce fish stress and is recommended, particularly if additional handling of fish is required (e.g., length and weight measurements, scale samples, fin clips, tagging).

~~10.~~17. Fish should be handled properly (e.g., wetting measuring boards, not overcrowding fish in buckets, etc.).

~~11.~~18. Fish should be observed for general condition and injuries (e.g., increased recovery time, dark bands, apparent spinal injuries) and be completely revived before releasing at the location of capture. Every attempt should be made to process and release ESA-listed specimens first. Record any mortalities.

~~12.~~19. Pertinent water quality (e.g., conductivity and temperature) and sampling notes (e.g., shocker settings, fish condition/injuries/mortalities) should be recorded in a logbook to improve technique and help train new operators.

10. Data sheets and metadata for fish abundance sampling

Electrofishing data sheet #1 (front page)

Date:	Page ___ of ___
Time:	Stream name:
Stream condition:	County:
Water clarity:	Site: (Lat/Long or UTM)
Specific conductance:	Personnel:
Water Temp:	
Air Temp:	

NSO number	Habitat type	Volts	Frequency (Hz)	Time (sec)	Species	Pass 1	Pass 2	Pass 3	No. Mortality

Electrofishing data sheet #2 (back of page).

NSO number	Species	Length (mm)	Weight (g)	NSO number	Species	Length (mm)	Weight (g)

Diver visual observation data sheet.

Date:	Page ___ of ___
Time:	Stream name:
Stream condition:	County:
Water clarity:	Site: (Lat/Long or UTM)
Water Temp:	Personnel:

Diver visual observation samples

NSO number	Habitat type	Species	Pass 1	Pass 2	Pass 3

Metadata for electrofishing and diver observations of juvenile fish.

Item	Description
Date	Calendar date (MM/DD/YY)
Time	Military time (HHMM)
Stream name	Stream name on USGS 1:24,000 Quad. Map
County	California county name
Location	Coordinates of trap site in either latitude and longitude or UTM
Stream condition	Includes discharge or stage height if available, amount of debris visible, turbidity.
Water clarity	Estimated visibility in meters.
Water temp	Water temperature in ^{o}C
Page	Number pages consecutively
Personnel	Name of field personnel recording data
Species code	(Convention is first letter of genus name and first letter of species name. When species names within a genus begin with the same letter, include the first and second letter of the species name.
OC	Cutthroat trout
OK	Coho salmon
OM	Steelhead
OT	Chinook salmon
ON	Sockeye salmon
OKE	Chum salmon
TR	Trout too small (< 80 mm) to accurately identify
CO	Sacramento sucker
LT	Pacific lamprey
LA	River lamprey
LR	Western brook lamprey
AM	Unidentified lamprey ammocete
CA	Coast range sculpin
CA	Prickley sculpin
	Add other species as encountered
NSO	Number of habitat unit in "numerical sequential order".
Habitat type	Pool, deep pool, run, riffle or other
Voltage	Voltage setting of electrofishing unit used.
Frequency	Frequency setting of electrofishing unit used.
Time	Time electrofishing was applied, in seconds.
Pass 1, 2, or 3 number	Number of the species collected on that electrofishing pass.
Length	Fork length in mm
Weight	Wet weight in g
Mortality	Record if fish died during collection

Habitat Survey for Fish Sampling Data Sheet

Date:	Page ___ of ___
Time:	Stream name:
Stream condition:	County:
Water clarity:	Site: (Lat/Long or UTM)
Water Temp:	Personnel:

NSO	Habitat type	Dist-ance	Length	Width 1	Width 2	Area	Depth 1	Depth 2	Max Depth	Pool tail depth (m)	Vol
		(m)	(m)	(m)	(m)	(m^2)	(m)	(m)	(m)	(m)	(m^3)

Metadata for habitat survey before sampling juvenile fish.

NSO	Habitat unit number, beginning with 1 at the downstream end and numbered sequentially upstream.
Hab Type	P = pool, N = run or glide, R = riffle
Distance	Cumulative distance (in meters) to the lower end of the habitat unit from the beginning (downstream end) of the habitat survey.
Length	Length (m) of the habitat unit
Width 1	Width (m) of the habitat unit.
Width 2	Width (m) of the habitat unit.
Area	Area (m^2) of habitat unit calculated from average width and length.
Depth 1	Depth (m) of the habitat unit.
Depth 2	Depth (m) of the habitat unit.
MaxDepth	Maximum depth (m) of the habitat unit.
Pool tail depth	Pool tail depth (m). Used to calculated residual pool volume if desired.
Area	Volume (m^3) of habitat unit calculated from average width and average depth.

RELATIVE WEIGHT OF JUVENILE COHO SALMON AND STEELHEAD

1. Rational

Length and weight of fish is commonly used as a management tool in inland fisheries to express population size structure and condition. Relationships between length and weight in fish have been mathematically expressed as condition factors (Blackwell et al. 2000). Condition factors express the predicted weight or plumpness of a fish at a given length. Until recently, however, limitations imposed by the statistical properties of length and weight relationships prevented their use in comparisons of populations. The development of a “relative weight” (Wr) index (Murphy et al. 1990) appears to have overcome these statistical limitations and presents potential for comparing condition among different populations. Condition has been used as a surrogate for fish body composition, as a measure of fish health and to assess productivity or prey available (Blackwell et al. 2000).

The concept of relative weight was first introduced by Wege and Anderson (1978) for measuring the condition of largemouth bass (*Micropterus salmoides*). Advantages Wege and Anderson saw in relative weight over other condition indices were that: 1) it is easy to calculate, 2) it does not change with different measurement units, 3) standard weights (Ws) compensate for inherent changes in body form, 4) variation in Wr may be due to ecological changes and 5) Wr values can compared between fish of different lengths from different populations (Blackwell et al. 2000). Relative weight is calculated as:

$$Wr = \left(\frac{W}{Ws}\right) \bullet 100$$

where W = weight (g) of fish being compared and Ws = the standard weight for the same species of fish at the same length. Multiplying by 100 expresses the equation as a percentage of Ws and makes the result conceptually easier to grasp.

Weight of juvenile Pacific salmon and steelhead has not been routinely recorded in the past. Accurately recording weight of small live fish in the field was difficult with earlier technology, and many saw limited use in these data. These limits in balance technology produced variable results of questionable value. Consequently, condition indices for these Pacific salmon and steelhead have not been calculated. However, improvements in portable electronic balances now offer the opportunity to collect precise measurements to $1/100^{th}$ of a gram in the field.

2. Assumptions

The use of relative weight as a measure in monitoring the response of coho salmon to watershed restoration is based on the hypothesis that *growth and physiological performance of fish can be correlated with habitat condition*. Furthermore, since coho salmon and steelhead use freshwater habitats for a year or more before migrating to the Pacific Ocean, the condition of these species should reflect habitat condition. Mechanisms describing the response of coho salmon and steelhead condition relative weight to changing habitat condition may not be simple or linear. For example, improving habitat could increase prey biomass available, prey visibility and habitat available to fish. Ultimately, the usefulness of relative weight as a monitoring response measure will rest on identifying mechanisms such as those, as well as identifying any biases in the method.

3. Limitations

The standard weight equations described below was developed for juvenile coho salmon ranging from 45-119 mm FL and for juvenile steelhead ranging from 50-200 mm FL. Thus, they cannot be applied reliably to fish smaller or larger than these ranges. Reasons for this limitation are twofold. Errors associated with measuring the weight of live fish increase as size decreases and Wr values become highly variable in juvenile coho salmon < 45 mm FL or steelhead < 50 mm FL. Although the Wr for juvenile coho salmon my be expected to apply to fish > 119 mm FL, we presently lack data to test this relationship. Variation around the average steelhead Wr increases in fish > 200 mm FL. This variation may be related to change in body form associated with smolting or with sample size. It is recommended that the equation be applied to steelhead no longer than 200 mm FL, because of this uncertainty.

4. Sampling Design

Sampling designs to collect length and weight data for calculating juvenile coho salmon and steelhead Wr should consider three sources of variation. First, the number of fish measured should be adequate to describe the mean with confidence intervals that are not so large that the data are meaningless. Second, the spatial distribution of fish sampled should be representative of the area of interest. Third, temporal variation will be important, particularly if the sample includes fish undergoing smoltification. Therefore, sampling during late summer through fall is recommended.

Confidence intervals associated with mean Wr values vary with number of fish measured. Regressing log of 95% confidence widths of the mean against log of sample size for 76 populations of juvenile coho salmon (Figure 2) yielded the equation:

$$\log_{10}(95\%CIW) = 1.7675 - 0.4625\log_{10} N$$

where CIW = 95% confidence interval width and N = sample size, $R^2 = 0.524$ and $P < 0.0001$.

It is possible to calculate the number of individual fish (N_i) required in a sample to give a predetermined confidence interval width by solving the by solving the regression of $\log_{10}$(CIW) on $\log_{10}$(N) for N_i (Murphy et al. 1990). From this regression:

$$\begin{aligned} N_i &= \left(\frac{CIW}{10^{1.7675}}\right)^{\frac{1}{-0.4625}} \\ &= \left(\frac{CIW}{58.54}\right)^{-2.16} \end{aligned}$$

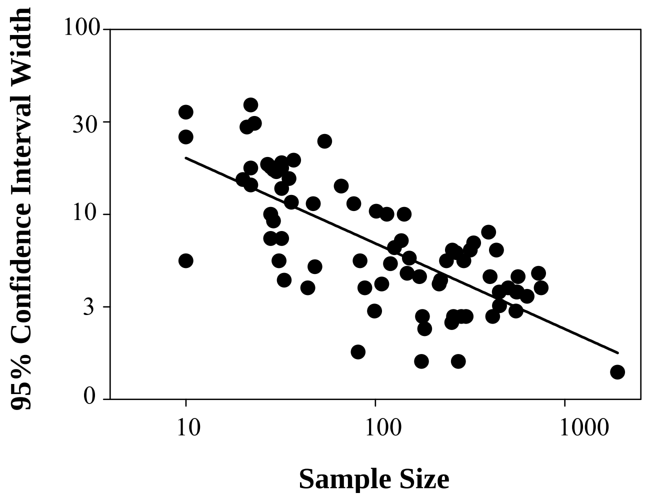


Figure 4. Scatterplot of the relationship between confidence interval width (CIW) for mean relative weight and sample size (N) in 76 populations of juvenile coho salmon.

Using this equation we can predict that measuring 74 juvenile coho salmon will yield a mean Wr with 95% confidence interval width of 8, or $\pm$ 4. Since the majority of mean Wr values fall between 90 and 110 (54 of 76 we examined), a confidence interval width of 8 will often be equivalent to $\pm$ 3.4-4.4% of the mean Wr value. Thus we recommend measure length and weight of 74 or more juvenile coho salmon to calculate Wr for each stream sampled.

Where samples are collected within a stream will influence how representative estimates of Wr are, and may also influence precision. In 2003, we calculated mean Wr and associated 95% confidence intervals from four streams. Fish were sampled from 1.5 km long stream reaches of Bond Creek, Hollow Tree Creek, and Huckleberry Creek in the Hollow Tree Creek watershed, Mendocino County and along a 6 km reach in Prairie Creek, Humboldt County. Position within each stream was then expressed as percentage from the starting distance or downstream end (Figure 3). Analysis of these data indicated no difference in Wr among streams ($P = 0.7889$, but differences among distances in streams ($P = 0.0006$) and a significant stream - distance interaction ($P < 0.0001$).

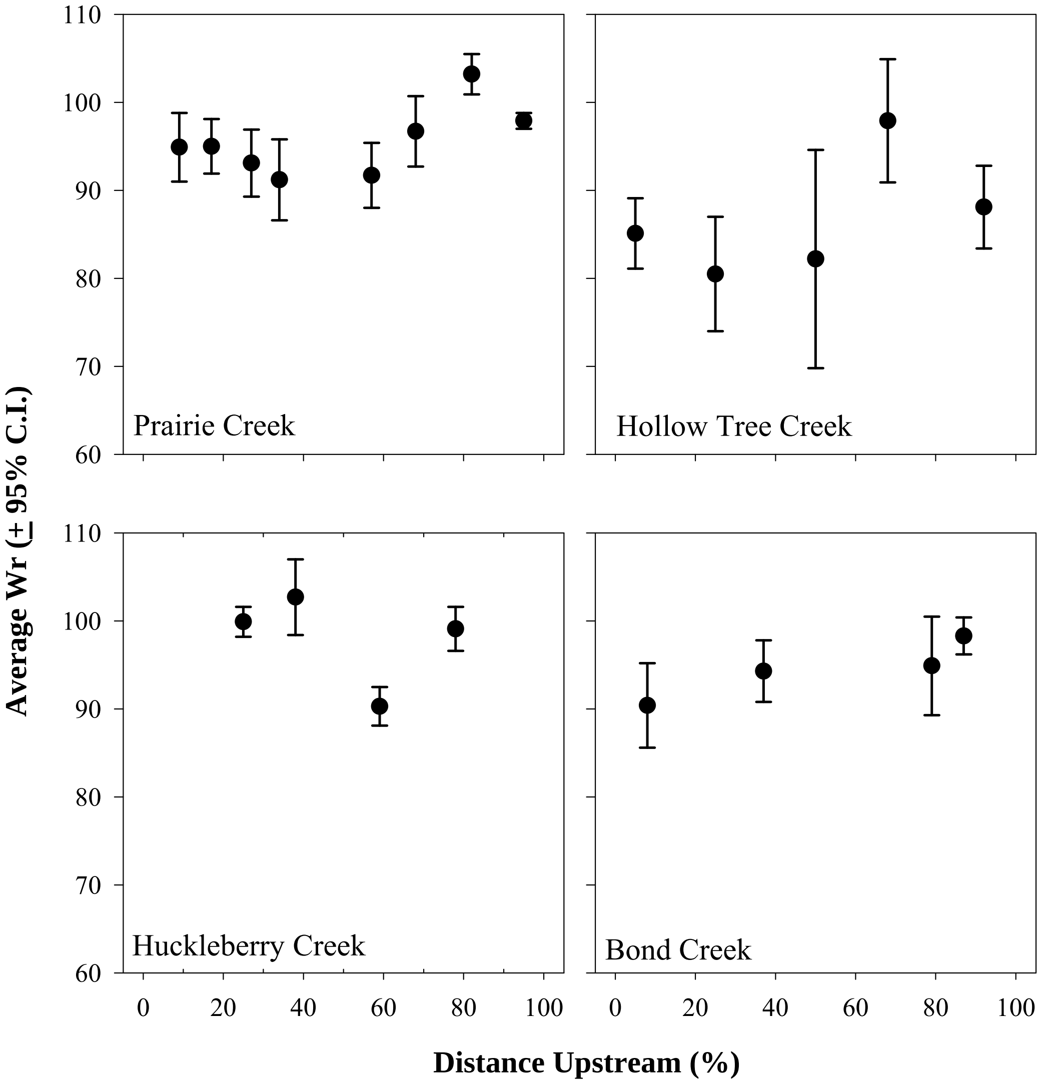


Figure 5. Mean and 95% confidence intervals for juvenile coho salmon Wr in four northern California streams. Data collected during October 2003.

We then examined distribution of Wr within each stream and found that only in Bond Creek were differences among sites not significant ($P = 0.0692$). Relative weight in Hollow Tree Creek ($P = 0.0142$), Prairie Creek ($P < 0.0001$) and Huckleberry Creek ($P < 0.0001$) all differed with distance. However, no consistent pattern was apparent (Figure 5). When possible, therefore, we recommend distributing samples over a stream reach of no less than 200-300 m.

5. Methods

Here we present a standard weight equation (Ws) for juvenile coho salmon and steelhead. For coho salmon, the equation was developed using data from southeastern Alaska to northern California. For steelhead, the data represent populations from southeastern Alaska through central California. Data for Alaska were provided by the USDA Forest Service, Pacific Northwest Field Station, Juneau, AK, data from Washington were provided by Brian Fransen, Weyerhaeuser Company, Federal Way, WA, and coho salmon data from California were gathered by the California Cooperative Fish Research Unit, Humboldt State University, Arcata, CA. Central California steelhead data were provided by Tommy Williams, National Marine Fisheries Service, Santa Cruz, CA.

We analyzed length – weight relationships for 80 populations of coho salmon. Data for four populations was removed because they had low coefficients of determination (e.g. $R^2 < 0.80$). The equation was developed from the remaining 76 populations using the regression-line-percentile technique (Murphy et al. 1990). The regression-line-percentile technique is based on 75^{th}-percentile weights and uses $\log_{10}$ transformed data from a series of populations as the statistical population to be modeled (Blackwell et al. 2000). The resulting Ws equation for coho salmon was:

$$\log_{10} Ws = -4.949 + 3.017 \log_{10} FL(mm)$$

where Ws = standard weight (g) and FL = fork length (mm).

For steelhead, 121 populations were analyzed using the same approach. The resulting Ws equation for steelhead was:

$$\log_{10} Ws = -4.790 + 2.928 \log_{10} FL(mm)$$

Collecting and Measuring Fish

Gathering data needed calculate relative weight is not difficult and can be combined with other methods that produce a sample of juvenile coho salmon. Electrofishing, minnow trapping and seining all should produce reliable data. The objective in sampling should be to obtain measurements that reflect the current range in size of the species being sampled.

After capture, fish should be anesthetized using tricane methanesulfonate (MS222), clove oil or Alka Seltzer in cool oxygenated water. Human health concerns have been raised over chronic exposure to MS222, therefore any personnel using this agent should be familiar with cautions explained on the material safety data sheet accompanying the product and should take appropriate precautionary measures. Effectiveness of anesthetic agents varies with concentration of the agent, water temperature, and fish density. Those using anesthetics should be familiar with dosage recommendations. Oxygenated, cool water should be provided to fish being held before anesthesia and those recovering from anesthesia.

Measurements of fork length should be recorded to the nearest 1.0 mm (Figure 1) and measurements of weight should be recorded to the nearest 0.01 g wet weight. Portable electronic balances having 0.01 g accuracy are sensitive to wind and excess water on fish. To obtain accurate weights, we suggest constructing a simple wind shield from plastic sheeting, thin plywood, or fabric. We also recommend the operator tare the balance after each measurement.

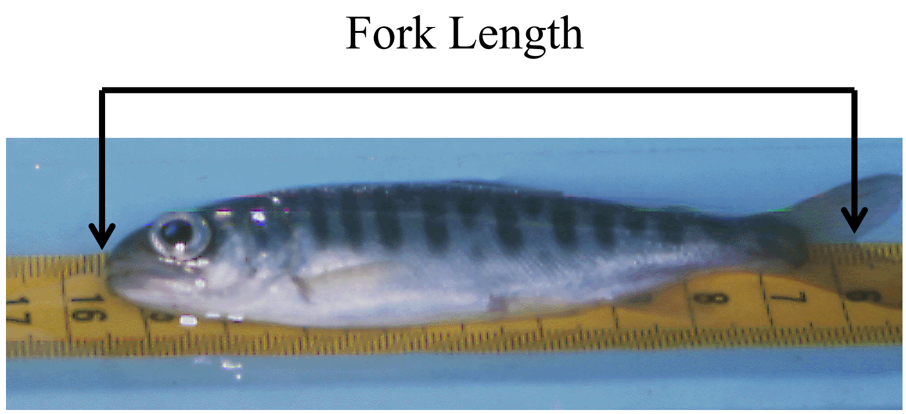


Figure 6. Juvenile coho salmon with fork length illustrated.

6. Data Analysis

The standard weight for juvenile coho salmon is used to calculate relative weight of individuals from the equation:

$$Wr = \left(\frac{W}{Ws}\right) \bullet 100$$

where W is the weight of an individual juvenile coho salmon and Ws is the standard weight for juvenile coho salmon. Note that Wr is obtained as the antilog of $\log_{10}$ Ws.

Relative weight values may be determined using a spreadsheet by first calculating $\log_{10}$ (Ws) for individual fish, then second taking the anti-log of these values (Ws), and using the Ws value in the above equation. These calculations are illustrated below using five hypothetical fish each 50 mm FL whose weights vary from 1.45-1.55 g. For example: fish number one in Table 7 is 50 mm FL and weighs 1.45 g, therefore

$$\begin{aligned} \log_{10}(Ws) &= -4.949 + 3.017 * \log_{10}(50) \\ &= -4.949 + 3.017 * 1.7 \\ &= 0.18 \end{aligned}$$

in an Microsoft Excel spreadsheet the antilog of 0.18 can be obtained by entering 10^0.18, yielding,

$$\text{antilog}(0.18) = 1.50$$

finally, Wr is calculated as the observed weight divided by Ws and expressed as a percentage,

$$Wr = \left(\frac{1.45}{1.50}\right) \bullet 100 = 97$$

Table4. Illustration of calculating Wr in a spreadsheet.

Fish No.	FL (mm)	Wt (g)	Log_{10} Ws (g)	Antilog [Log_{10} Ws (g)]	Wr
1	50	1.45	0.18	1.50	97
2	50	1.49	0.18	1.50	99
3	50	1.50	0.18	1.50	100
4	50	1.53	0.18	1.50	102
5	50	1.55	0.18	1.50	103

Note that Wr values are presented as whole numbers as suggested by Murphy et al. (1990). Alternatively, antilog [Log_{10} Ws (g)] values for juvenile coho salmon ranging from 45-119 mm FL are presented in Appendix B and can be entered directly into the Wr equation.

7. Quality Assurance and Quality Control

Quality assurance and quality control procedures should be established before recording length and weight of juvenile coho salmon. These procedures should include elements of the following:

Training that addresses,

5) identification of fish species likely to be encountered,
6) proper handling of fish and
7) use, care and calibration of balances

The quality assurance plan for data entry and management should include,

6) data entry
7) data management
8) data analysis
9) chain of custody for data

Data entry and management elements of QA/QC procedures should include the use of metric units of measure, proper use of measuring boards and balances, data coding of field sheets and data entry. Procedures to verify the accuracy of recorded field data and data entry into an electronic format should be developed. These typically involve an independent observer check 5 – 10% of the original entries. The entire data base should be checked for errors if this sample of original entries reveals a rate of error of more than 5%.

8. Personnel and Equipment Needed

Personnel	Materials
Two (2) persons	Portable balance with 0.01 g resolution
	Measuring board, 30 cm long with 1 mm increments
	Write-in-the-rain notebook or data sheets
	Pencils

9. Sample size, intercept, slope and significance values for regressions of weight (g) on fork length (mm) for 76 populations of juvenile coho salmon used in calculating Wr.

Location	Water Body	Year	N	Intercept	Slope	R^2	P
AK	108 Creek	1992	25	3.5333	0.1037	0.930	<0.0001
AK	25 Mile Pond	1995	487	-5.8840	0.1391	0.946	<0.0001
AK	Bambi Creek	1986	533	-4.3226	0.1184	0.896	<0.0001
AK	Bambi Creek	1987	571	-5.6582	0.1366	0.877	<0.0001
AK	Beach Creek	1986	738	-4.5978	0.1227	0.924	<0.0001
AK	Beach Creek	1987	642	-5.2967	0.1316	0.928	<0.0001
AK	Beaver Creek	1986	10	-11.9990	0.2258	0.950	<0.0001
AK	Bozo Creek	1986	29	-6.7038	0.1638	0.905	<0.0001
AK	Calder Creek	1992	28	-4.2500	0.1127	0.967	<0.0001
AK	Chuck River	1986	41	-5.7843	0.1419	0.899	<0.0001
AK	Deer Track Creek	1983	263	-10.0538	0.1995	0.907	<0.0001
AK	Deer Track Creek	1984	101	-13.6894	0.2266	0.793	<0.0001
AK	Deer Track Creek	1985	492	-5.0324	0.1236	0.911	<0.0001
AK	Devilfish Creek	1992	15	-2.9782	0.0871	0.910	<0.0001
AK	Dry Slough	1995	10	-21.4115	0.3477	0.959	<0.0001
AK	Ella Creek	1986	65	-2.8845	0.0910	0.898	<0.0001
AK	Fringe Creek	1992	23	-3.4048	0.0975	0.941	<0.0001
AK	Game Creek	1986	29	-4.1208	0.1188	0.862	<0.0001
AK	Hamilton River	1986	17	-5.7277	0.1402	0.940	<0.0001
AK	Haystack Pond	1995	30	-8.9857	0.1901	0.951	<0.0001
AK	Kake Bake Creek	1983	160	-18.4133	0.3007	0.935	<0.0001
AK	Kake Bake Creek	1984	67	-15.0452	0.2621	0.947	<0.0001
AK	Ken's Pond	1995	927	-4.8113	0.1283	0.899	<0.0001
AK	Lost Pond	1995	248	-7.7025	0.1748	0.879	<0.0001
AK	Maybeso Creek	1999	469	-6.4049	0.1615	0.934	<0.0001
AK	Maybeso Creek	2000	37	-8.4496	0.1728	0.897	<0.0001
AK	Meter Creek	1986	39	-4.7058	0.1244	0.919	<0.0001
AK	Painted Creek	1999	82	-3.6904	0.1055	0.966	<0.0001
AK	Red Bluff Creek	1986	48	-4.2497	0.1202	0.847	<0.0001
AK	Rio Beaver Creek	1986	12	-0.0438	0.1087	0.991	<0.0001
AK	Rio Roberts Creek	1986	26	-4.1810	0.1144	0.947	<0.0001
AK	Saginaw Creek	1989	182	-5.4120	0.1372	0.926	<0.0001
AK	Saginaw Creek	1994	116	-6.4120	0.1519	0.937	<0.0001
AK	Saginaw Creek	1995	170	-6.1583	0.1541	0.917	<0.0001
AK	Sal Creek	1997	310	-8.9906	0.1948	0.906	<0.0001
AK	Salamander Creek	1986	13	-4.4061	0.1229	0.900	<0.0001
AK	Skogs Creek	1986	34	-8.4476	0.1854	0.937	<0.0001
AK	Slippery Lake Creek	1988	722	-6.4412	0.1570	0.938	<0.0001
AK	Slippery Lake Creek	1989	906	-5.6762	0.1392	0.858	<0.0001
AK	Slippery Lake Creek	1990	261	-5.1303	0.1401	0.912	<0.0001
AK	Staney Creek	1996	398	-5.5079	0.1423	0.911	<0.0001
AK	Staney Creek	1997	35	-6.3513	0.1569	0.850	<0.0001
AK	Staney Creek	1998	29	-4.7818	0.1231	0.949	<0.0001
AK	Tonalite Creek	1998	257	-7.6902	0.1688	0.915	<0.0001

(Concluded)

Location	Water Body	Year	N	Intercept	Slope	R^2	P
AK	Tonalite Creek	1999	1988	-7.6673	0.1750	0.896	<0.0001
AK	Tonalite Creek	2000	576	-8.5643	0.1838	0.873	<0.0001
AK	Trap Bay Creek	1985	37	-4.6226	0.1213	0.855	<0.0001
AK	Turn Creek	1992	33	-3.4995	0.1001	0.816	<0.0001
AK	Wryno Creek	1986	24	-4.8719	0.1308	0.912	<0.0001
CA	Boyes Creek	2000	87	0.5117	0.1297	0.875	<0.0001
CA	Boyes Creek	2001	323	-4.8551	0.1309	0.875	<0.0001
CA	Caspar Creek	1998	42	-7.5604	0.1773	0.957	<0.0001
CA	Caspar Creek	1999	188	-4.6423	0.1244	0.911	<0.0001
CA	Freshwater Creek	1998	48	-6.7912	0.1606	0.942	<0.0001
CA	Freshwater Creek	1999	308	-3.8067	0.1056	0.927	<0.0001
CA	Hollow Tree Creek	1998	18	-3.9752	0.1131	0.937	<0.0001
CA	Lindsay Creek	1998	28	-7.4331	0.1806	0.952	<0.0001
CA	Lindsay Creek	1999	172	-9.3691	0.1999	0.941	<0.0001
CA	Pollack Creek	1999	96	-3.9140	0.1100	0.901	<0.0001
CA	Prairie Creek	2000	225	-6.8312	0.1627	0.855	<0.0001
CA	Prairie Creek	2001	277	-9.9059	0.2077	0.952	<0.0001
CA	SF Broken Kettle Creek	1999	88	-7.1758	0.1645	0.878	<0.0001
CA	Sharber Creek	1999	133	-5.9014	0.1454	0.906	<0.0001
CA	Streelow Creek	2000	161	-6.1630	0.1499	0.852	<0.0001
CA	WF Sproul Creek	1998	35	-6.4976	0.1624	0.959	<0.0001
CA	WF Sproul Creek	1999	271	-4.4851	0.1231	0.892	<0.0001
WA	Forks Creek	1995	310	-6.4500	0.1519	0.892	<0.0001
WA	Forks Creek	1996	288	-6.7798	0.1617	0.925	<0.0001
WA	Forks Creek	2001	189	-6.5943	0.1607	0.929	<0.0001
WA	Forks Creek	2002	169	-6.4428	0.1554	0.941	<0.0001
WA	Fowler Creek	1986	22	-6.8462	0.1579	0.930	<0.0001
WA	Herrington Creek	1997	100	-9.0987	0.1976	0.914	<0.0001
WA	Herrington Creek	1998	37	-9.2843	0.1966	0.809	<0.0001
WA	Herrington Creek	1999	139	-7.7610	0.1759	0.921	<0.0001
WA	Huckleberry Creek	2001	93	-6.0198	0.1474	0.912	<0.0001
WA	Huckleberry Creek	2002	75	-5.9711	0.1481	0.960	<0.0001

10. Juvenile coho salmon standard weight (Ws) values corresponding with fork length at 1 mm intervals.

FL (mm)	Ws	FL (mm)	Ws	FL (mm)	Ws	FL (mm)	Ws
		63	3.02	82	6.68	101	12.53
45	1.09	64	3.16	83	6.93	102	12.91
46	1.17	65	3.32	84	7.19	103	13.30
47	1.25	66	3.47	85	7.45	104	13.69
48	1.33	67	3.63	86	7.72	105	14.09
49	1.41	68	3.80	87	7.99	106	14.50
50	1.50	69	3.97	88	8.27	107	14.92
51	1.59	70	4.15	89	8.56	108	15.34
52	1.69	71	4.33	90	8.85	109	15.77
53	1.79	72	4.51	91	9.15	110	16.21
54	1.90	73	4.71	92	9.46	111	16.66
55	2.00	74	4.90	93	9.77	112	17.12
56	2.11	75	5.11	94	10.09	113	17.58
57	2.23	76	5.31	95	10.42	114	18.06
58	2.35	77	5.53	96	10.75	115	18.54
59	2.48	78	5.75	97	11.09	116	19.03
60	2.60	79	5.97	98	11.44	117	19.53
61	2.74	80	6.20	99	11.80	118	20.04
62	2.88	81	6.44	100	12.16	119	20.56

To calculate Wr using this table:

1. Find the fork length of the fish for which you wish to calculate Wr,
2. Divide the Ws value corresponding with that length into the weight of the fish for which you wish to calculate Wr,
3. Multiply the result by 100.

11. Steps in Calculating Relative Weight

1. Select habitat units to be sampled:
    a. Classify each habitat unit to be sampled following methods for either level II or III habitat typing (Harris 2004).
    b. Record the latitude and longitude, or UTM, of the most downstream and upstream habitats sampled.
    c. For any other habitats sampled, record the distance from the beginning (downstream) habitat to the habitat unit.

2. Collect juvenile coho salmon:
    a. Gear used may include electro-fishing gear, minnow traps, seines or other non-lethal gear.
    b. If possible, collect 65 or more juvenile coho salmon fish.
    c. Distribute the sample among 3 or more habitat units, preferably more, that are separated by $\geq$ 300 m.

3. Record the number of juvenile coho salmon and other species collected.
    a. Transfer coho salmon to cold, well oxygenated water.
    b. Release other species into habitats from which they were collected after counting.

4. Prepare an anesthetic solution in quantity sufficient to sedate the number of fish that will be measured. Recommended anesthetics include:
    a. Tricaine (MS-222) at 15-25 $mg \bullet l^{-1}$ (Sommerfelt and Smith 1990),
    b. Clove oil at 24-48 $mg \bullet l^{-1}$ (Cho and Heath 2000), or
    c. Alka-Seltzer ™ tablets.

5. Transfer small groups of 4-5 juvenile coho salmon to a small (~ 1 L) plastic container containing water with the anesthetic and allow them to reach a state of normal or light sedation (Sommerfelt and Smith 1990). This state of sedation should be reached in < 5 minutes and is characterized by:
    a. Reacting to visual and tactile stimuli,
    b. Having a normal or only slightly increased opercular rate, and
    c. Retaining equilibrium.

6. After sedation is achieved:
    a. Record the fork length (FL) to the nearest mm and wet weight to the nearest 0.01 g. Note: the balance should be tared after each measurement.

7. Placed sedated fish in a second container of cold, well oxygenated water and allow them to recover from sedation, then release them into the habitats from which they were collected.

8. Calculate Wr for each fish by using either data presented in appendix B or in a spreadsheet transforming individual fish weights to $\log_{10}$ values and calculating Ws the individual fish weights.

12. Data sheets and metadata for relative weight.

Data sheet for recording length and weight to be used in determining mean relative weight.

Date: ________________ Latitude Longitude

Stream: ____________________________ Lower: ____________________________

Basin: ____________________________ Upper: ____________________________

County: ____________________________ Crew: ____________________________

Habitat No.	Distance (m)	Fish/ Habitat	Fish No.	Fork Length (mm)	Weight (g)	Comments

Metadata for juvenile coho salmon relative weight.

Item	Description
Date	Calendar date (MM/DD/YY)
Time	Military time (HHMM)
Stream name	Stream name on USGS 1:24,000 Quad. Map
County	California county name
Location	Coordinates of sampling site in either latitude and longitude or UTM
Page	Number pages consecutively
Personnel	Name of field personnel recording data
NSO	Number of habitat unit in "numerical sequential order".
Habitat type	Pool, deep pool, run, riffle or other
Length	Fork length in mm
Weight	Wet weight in g

SALMON AND STEELHEAD SMOLT PRODUCTION

1. Rational

Smolt production is defined as the number of salmon or steelhead smolts migrating from a stream toward the ocean. Smolt production is the product of the number of adults that successfully spawn and survival of all life stages between spawning and emigration to the ocean. Thus smolt production may have use in assessing watershed restoration actions that attempt to improve habitat conditions for spawning, egg survival and/or juvenile survival.

All species of Pacific salmon, including steelhead and at least some proportion of coastal cutthroat trout, undergo smolt transformation and migrate to the ocean for some period of their life history. As a measure of the response by fish to most watershed restoration actions, production of coho salmon or steelhead smolts is most appropriate because these two species reside in freshwater habitats one or more years before undertaking ocean migration. Steelhead do, however, have a more variable life history than salmon and are know to smolt over a more extended period, making estimates of smolt production more difficult for steelhead than for salmon. Interior populations of Chinook salmon may also remain in fresh water for up to a year before beginning their migration to the ocean. Chinook salmon fry from coastal populations begin migrating downstream soon after hatching and make extensive use of estuaries before undergoing smoltification. Therefore, production of Chinook salmon fry may be an appropriate measure of adult spawning habitat or estuary conditions. Data on production of salmon or steelhead smolts leaving a stream can provide information on freshwater survival and, by inference, habitat quality. When combined with estimates of the numbers of adults returning to spawn, it can also be used to calculate ocean survival.

2. Assumptions

Smolt production is typically measured by capturing migrants using traps. Migrant smolt traps are not 100 percent efficient. Without an estimate of trap efficiency, the number of smolts captured in a trap represents an unknown portion of the total number migrating downstream. Trap efficiency, the proportion of the total migrant population captured by the trap, is influenced by stream flow, fish species, size and behavior. Most of these variables change during a sampling season. Because of this, trap efficiency tests must be conducted regularly to accurately estimate the number of downstream migrating smolts.

Trap efficiency tests are mark-recapture experiments in which a known number of smolts are tagged and released at a point where they may be recaptured. The assumptions inherent in mark-recapture experiments are:

1. The population is closed and smolts are not immigrating into the area (moving upstream above the trap),
2. Smolts do not lose their marks between the release site and the recapture site,
3. Handling and marking smolts does not affect the probability of their being captured at the trap,
4. Being caught, handled and marked has no effect on the probability that an individual will die or emigrate,

5. All marked and unmarked smolts have an equal probability of emigrating or dying,
6. All marked and unmarked smolts have an equal probability of being captured at the trap(s),
7. Trap samples represent a random sample, where each of the possible combinations of marked and unmarked smolts have an equal probability of occurring,
8. All marked smolts in the catch are identified and reported,

Several additional assumptions should be considered when two traps are operated, with smolts marked at the upstream trap and recaptures recorded at the downstream trap. These are:

9. Marked and unmarked smolts have similar movement patterns between the release site and recapture site,
10. Smolts can pass the trap site only once,
11. All marked smolts pass the lower trap site by the end of the study, and
12. There is no mortality and all smolts emigrating pass the lower trap.

3. Limitations

Several problems influence the use of smolt production data in assessing watershed response to restoration. The first of these is differences in life history among species of salmonids. For example: most coastal Chinook salmon migrate from their natal streams as fry, then rear and undergo smoltification in lower rivers or estuaries before entering the Pacific Ocean during summer or fall. Trapping juvenile Chinook salmon leaving coastal streams does not necessarily, therefore, provide information on smolt production. However, trapping Chinook salmon fry can still provide a measure of change in watershed condition. Smolt production is also not now applicable to coastal cutthroat trout since this species exhibits a variable and unknown period of freshwater residence before ocean migration, or may not use the ocean environment.

A second limitation arises from criteria necessary for operating a trap to capture migrating smolts. Sites selected for migrant smolt trap placement should be located near the lower end of the basin so as to provide an estimate of the number of smolts leaving. Alternatively, the trapping site may be located downstream from an area of focused restoration. In either case, the gradient at the trap site should be relatively low, and the equipment required dictates that the site be accessible. Most methods of smolt trapping are more efficient in small or medium sized streams. In larger rivers, rotary screw traps can be deployed, but efficiency of these traps is often difficult to estimate and can prevent estimating smolt production. At the watershed scale, these criteria reduce or eliminate the element of randomness that is desirable in sampling. At the scale of a larger geographic region, however, streams could be randomly selected for smolt trapping from multiple watersheds within the region.

4. Sampling Design

Sampling design considerations include the sampling location within a watershed, how many traps to use, and the type of trap to use.

Sampling location

Sampling locations should be selected on the basis of answering a question. In the context of monitoring watershed restoration actions, a reasonable question might be; *have restoration projects within a sub-watershed resulted in greater numbers of smolts migrating from the sub-watershed*? Locating a smolt trap as near as the sub-watershed outlet as is practical would provide the best opportunity to answer this question. General considerations in locating smolt traps are:

a. The stream being sampled should have spawning populations of steelhead, coho salmon or Chinook salmon.

b. The stream should be neither so large nor small that efficiency of the trap cannot be evaluated. Trapping sites should be located in streams as large as the gear will effectively sample since larger streams will usually yield more smolts. Size of streams in which various smolt trapping gear can effectively sample are generally second to fifth order and have an active channel width of no more than 30 m.

c. Stream gradient should not be too great, a gradient of 1 – 2% is best. High gradient sites can result in high water velocity that may injure fry and smolts during trapping. Conversely, velocity in wide unconstrained channels may not be adequate to operate some types of traps.

d. Depth of water is an important consideration in selecting sampling sites. Fyke net traps are limited to depths of 1 m or less. Rotary screw traps and inclined plane traps must be located at depths of 0.75 m or greater.

e. Water velocity or flow (m/s) must be sufficient to carry fish into fyke net or inclined-plane traps. For rotary screw traps, a flow of 0.8-2.0 m/s has been observed to be sufficient to rotate the screw. At some sites, panels can be installed to direct water into traps. Stream flow should enter the trap on a straight line. Placing traps in bend pools or near obstructions that create eddys may cause fry to be impinged on trap surfaces.

f. The stream substrate at the site should be relatively uniform. Presence of boulders and cobble will create turbulence that may limit trap efficiency or contribute to injury of fish.

g. Access is an important consideration, both physical and legal access. Trapping sites should be near roads, particularly if operating a rotary screw or inclined plane trap. The site should also be located where a land owner is willing to allow access for long periods, 10 or more years.

h. Finally, locating the trap where large trees can serve as suitable anchor sites is helpful, though not necessary.

Number of traps

Sampling a single stream may be accomplished with either one of two traps. If one trap is used, fish marked for efficiency testing are released upstream of the trap and required to pass the trapping site a second time. When two traps are used, fish a marked at the upper trap and recaptures at the lower trap provide information needed to calculate efficiency.

A primary concern in deciding to operate one or two traps is possible behavioral changes by trapped fish. When one trap is operated and juvenile salmonids are captured, marked, then released upstream, they often delay migration for one or more weeks, or indefinitely (W. Duffy, personal observation). This change in migratory behavior violates several important assumptions inherent in smolt trapping, and usually results in inflated estimates of production. Marking fish for efficiency testing at an upstream site and immediately releasing them, presumably minimizes these behavioral changes.

Type of trap

Salmon and steelhead smolts migrating downstream may be captured using traps of various designs. The most common traps used are pipe traps, fyke nets, inclined plane traps and rotary screw traps. All act on the principle of intercepting fish migrating downstream and allowing current to passively carry fish into a live box.

- Pipe traps – are constructed by using rock to create a damn which opens into a PVC pipe that leads to a live box. Pipe traps have been commonly used in small streams. Limitations of pipe traps are that rock dams used to shunt water into the pipe can easily be overtopped during fluctuating flows.

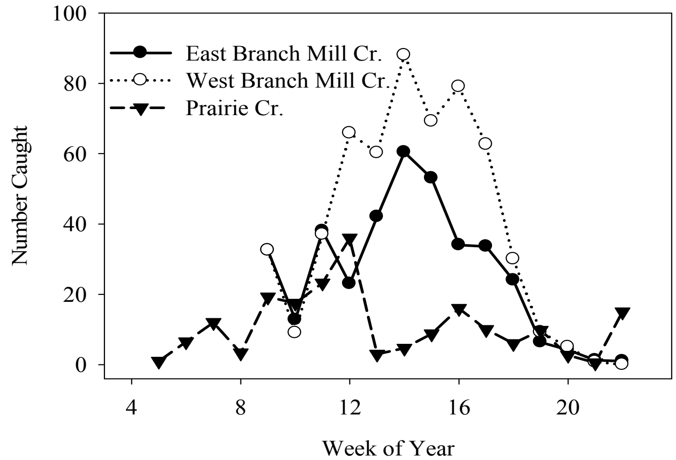
- Fyke net traps - consist of a fyke net having a live box attached to the cod end. In smaller streams, the fyke net can be fitted with wings and effectively cover all or most of the stream.

- Inclined plane traps - are constructed from rigid material and have a large rectangular opening that leads to a smaller opening at the live box. Inclined plane traps may be fished with the trap mouth resting on the stream bottom, or they can be fitted with pontoons and fish off bottom in larger streams (Todd 1994).

- Rotary screw traps - consist of a cone covered with screen and having an archemedes screw built into the cone. The trap is suspended on pontoons with the larger end of the cone facing upstream and adjusted so that the lower half of the cone is in the water. Water pressure forces the cone to turn on a central shaft and migrating smolts that enter the cone are trapped by the rotating screw and forced into a live box at the end of the trap. Rotary screw traps are better suited to larger streams and rivers having adequate flow to turn the cone and enough depth to float the trap.

None of these trap designs is appropriate for all streams or flow conditions. The type and size of trap used is both a function of the size and flow characteristics of the stream being sampled, and the size and species of the fish that are targeted for trapping. In general, the screw trap is more effective in larger streams, while the fyke net and inclined-plane traps are better suited to small or medium sized streams.

Care must be taken to minimize mortality to young fish when operating smolt traps. During high stream, smolt traps and live boxes can become clogged with debris contributing to mortality of young fish. Therefore, smolt traps must be carefully monitored during times when flow is high or when excessive debris might be carried in the stream. Another potential source of mortality is predation by larger fish on smaller fish in the trap live box. Fern fronds or fir boughs are often placed in the trap live box to provide hiding cover for smaller fish. A v-shaped water current deflector, intended to create a pocket of calm water for small fish, is often built into trap live boxes constructed of plywood or metal. Our research suggests that neither of these techniques is particularly effective in reducing mortality of fry. Instead, we recommend a live box designed to hydraulically separate salmon fry from larger fish (see personnel and equipment section below).



5. Methods

Sampling duration and frequency

Migration of coho salmon and steelhead smolts from California streams may occur from fall through summer, but peak migration for both species during most years is during March through May (W. Duffy unpublished, Shapovalov and Taft 1954, Ketcham et al. 2004, [Figure 7]). Sampling for migrating smolts should begin no later than February or early March and continue until the catch decreases, usually in June, or until water temperatures warm enough to present additional stress to fish. Timing of migration may vary among years, and would be expected to begin earlier in southern than northern streams.

Figure 7. Average number of steelhead captured in downstream migrant traps during 1999 – 2003 in Mill Creek, Del Norte County and Prairie Creek, Humboldt County. Data from Mill Creek were provided by Chris Howard, Green Diamond Resources, Blue Lake, CA.

To provide reliable estimates of smolt migration, smolt traps must be operated continuously, that is 24 hours per day seven days per week. Continuous sampling is necessary since salmon smolts continue to migrate during most periods of the day (Feola 2006) and methods for randomly selecting sampling dates, if possible, have not been developed. During continuous sampling, fish must be removed from live boxes and the trap cleaned daily.

Estimating trap efficiency

Trap efficiency is estimated by marking a known number of smolts caught in traps the previous 24 hours, then releasing them to be available to recapture. Ideally, some constant fraction of the smolts captured each day would be marked and released for recapture. If constant fractional marking is not possible, marking 50 or more smolts each week will usually provide recaptures adequate to estimate efficiency. However, capturing 50 or more steelhead smolts is not always possible (Figure 7) and fewer marked must be used. Although this number will vary with stream size and number of smolts passing the trap. The number of marked smolts recaptured is then recorded on subsequent dates.

Marking smolts for trap efficiency tests may be accomplished in several non-destructive ways. Some common on-destructive fish marking techniques that could be used in smolt trapping include fin clipping, freeze branding, PIT tagging and dye injection. The marking method selected should be flexible enough to provide a weekly batch mark. For example: different colors of acrylic dye can be injected under the skin using small hypodermic needle. Location or color of the dye can be rotated weekly so that fish marked on different weeks are able to be separated at recapture. By using only a few colors of dye in combination with several marking locations it is possible to have unique weekly marks over a 3-4 month period.

The release location for marked fish for trap efficiency estimates should located far enough upstream so the fish can evenly mix with unmarked fish moving downstream, yet not be so far upstream as to cause an extracted period of migration of marked fish over multiple days. Thus, when operating two traps the traps should be separated by 200-300 meters. When operating one trap, marked fish are typically released at least two pool/riffle units, but no more than 300 meters, above the trap.

Any smolts, or other fish, that are handled for marking or size measurements should be anesthetized to reduce stress. Recognized fish anesthetic agents include tricaine methanesulfonate (MS222), Alka Seltzer ™, and clove oil. Human health concerns have been raised over chronic exposure to MS222, therefore any personnel using this agent should be familiar with cautions explained on the material safety data sheet (MSDS) accompanying the product and should take appropriate precautionary measures. Effectiveness of anesthetic agents varies with concentration of the agent, water temperature, and fish density. Those using anesthetics should be familiar with dosage recommendations. Oxygenated, cool water should be provided to smolts being held before anesthesia and those recovering from anesthesia. Recovery can be accomplished by holding fish in buckets of cool oxygenated water for a brief period, typically 15-20 minutes.

Fish size

Smolt transformation is a physiological process that occurs over a period of days to weeks. Therefore, not all fish beginning smolt transformation will display the silvery sheen characteristic of a smolt. This is especially true when fish are trapped high in a watershed. For this reason, it is important to collect information on size of smolts. Size-frequency distributions can then be examined later to assign probably smolt status. Size measurements should include fork length (mm) and, when possible, wet weight (g).

6. Data Analysis

Analysis of smolt trapping data can be accomplished using DARR 2.0 (Bjorkstedt 2005) software developed for this purpose. This software, along with installation instructions, a users guide and documentation, can be downloaded from the NOAA-Fisheries, Santa Cruz Laboratory web site at http://santacruz.nmfs.noaa.gov/publications/software/439/. Analysis using DARR 2.0 requires xx steps:

- First the data must be stratified or grouped into increments matching the marking schedule. If, for example, marks used are changed weekly, daily catch records would be grouped into weekly catch records. The structure of the data needed for importing into DARR 2.0 is illustrated below.

			Marked Fish Recaptured			
Week	Captured (unmarked)	Marked and Released	Week 1	Week 2		Week t
1	u_1	m_1	r_{11}	r_{12}	…	r_{1t}
2	u_2	m_2	r_{21}	r_{22}		r_{2t}
3	u_3	m_3	r_{31}	r_{32}		r_{3t}
⋮	⋮	⋮	⋮		⋱	⋮
t	u_t	m_s	r_{st}		$(r_{s,\,t-1})$	r_{st}

- When importing data into DARR 2.0, the user is first presented with a screen summarizing the data they have entered into a spreadsheet. For the test data provided with the software, the four categories of summarized data are:

Unmarked captures by stratum (u)

20 19 33 56 60 106 119 59 39 117 88 94 91 64 46 22

Marks released by stratum (m)

20 19 33 50 58 70 70 57 39 70 69 70 70 57 46 22

Recaptures by stratum (R)

7	2	0	0	0	0	0	0	0	0	0	0	0	0	0	0
0	8	3	1	0	0	0	0	0	0	0	0	0	0	0	0
0	0	13	3	0	0	0	0	0	0	0	0	0	0	0	0

0	0	0	24	4	2	0	0	0	0	0	0	0	0	0	0
0	0	0	0	11	13	3	0	0	0	0	0	0	0	0	0
0	0	0	0	0	40	9	1	0	0	0	0	0	0	0	0
0	0	0	0	0	0	38	7	0	0	0	0	0	0	0	0
0	0	0	0	0	0	0	16	0	1	0	0	0	0	0	0
0	0	0	0	0	0	0	0	0	8	0	0	0	0	0	0
0	0	0	0	0	0	0	0	0	32	2	1	0	0	0	0
0	0	0	0	0	0	0	0	0	0	32	7	0	1	0	0
0	0	0	0	0	0	0	0	0	0	0	22	11	1	0	0
0	0	0	0	0	0	0	0	0	0	0	0	4	9	0	0
0	0	0	0	0	0	0	0	0	0	0	0	0	26	12	1
0	0	0	0	0	0	0	0	0	0	0	0	0	0	33	4
0	0	0	0	0	0	0	0	0	0	0	0	0	0	0	7

Summed recaptures by mark group

9	12	16	30	27	50	45	17	8	45	40	34	13	39	37	7

These data should be reviewed for consistency with the spreadsheet and to decide whether or not there are sufficient data in each stratified period to support calculations.

- After accepting these data, the user is asked if they wish to pool any strata. Sometimes the number of migrating smolts caught in the trap during a week is insufficient to obtain a weekly trap efficiency estimate. Low catches may result from a low number of migrants, low trap efficiency, or a combination of both. If trap efficiency estimates are not possible during some weeks, at this point DARR 2.0 allows for grouping weeks together. Please see Bjorkstedt (2005) for further explanation on grouping strata.

- Next, the used is prompted to select whether they used one trap or two. When the number of traps operated is selected, DARR 2.0 completes calculations and presents a summary analysis table and figure. These summary results can be printed or saved to a file.

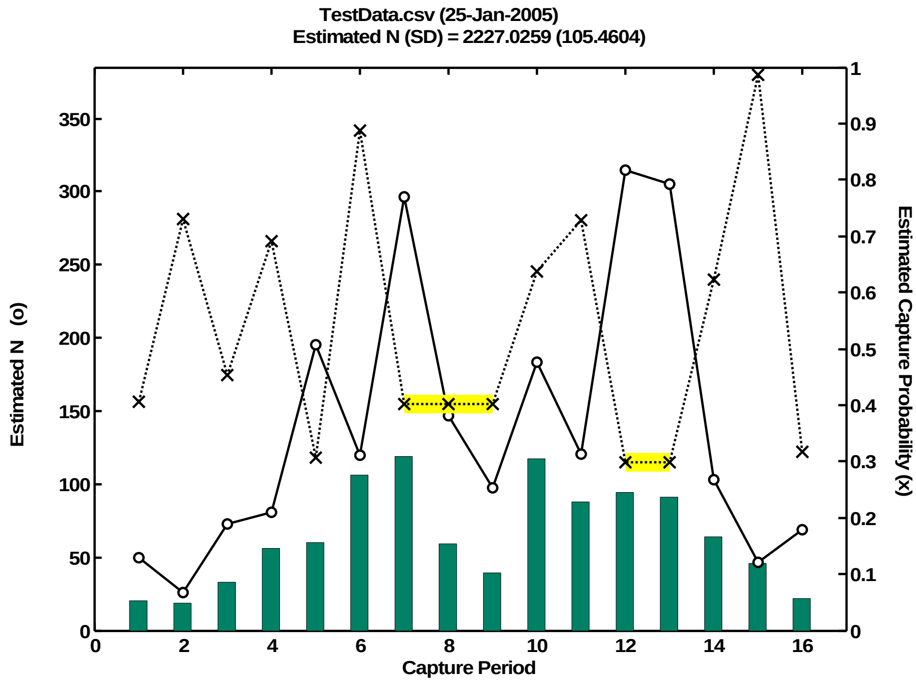


Figure 8. Summary results figure for test data from DARR 2.0.

7. Quality Assurance and Quality Control

Quality assurance and quality control procedures should be established for each salmon and steelhead smolt trapping program. These procedures should include elements of training, data entry and management, and independent assessment of methods.

The training program should address:

1) safety practices in the field,
2) identification of fish species likely to be encountered,
3) proper handling of fish and
4) data entry and management.

Data entry and management elements of QA/QC procedures should include the use of metric units of measure, proper use of measuring boards and balances, data coding of field sheets and data entry. Procedures to verify the accuracy of recorded field data and data entry into an electronic format should be developed. These typically involve an independent observer check 5 – 10% of the original entries. The entire data base should be checked for errors if this sample of original entries reveals a rate of error of more than 5%.

8. Personnel and Equipment Needed

Here we present personnel and equipment needs for installing a fyke trap. Materials needed for installing other types of traps are similar, but will vary.

Personnel
Installation and removal – 4 persons, installing or removing a trap requires 4 – 8 hours.
Operation – 2 persons, for safety reasons we recommend that two people be assigned to smolt trap sampling. A two person crew is also more efficient in recording size of smolts and, if desired, collecting scale samples for later aging,
Personal equipment should include clothing appropriate for weather conditions, chest waders and personal flotation devices.

Equipment for installation	Equipment for operation
Fyke net (see below for construction details)	4 Plastic pails (5 gal)
20’ – 8” dia PVC pipe	Portable air pump, tubing and air stone
Live box	Extra batteries for portable air pump
Sand bags	Aquarium dip nets for fish in buckets
100’ of 3/8” nylon line	Larger dip nets for fish in live box
Sturdy electrical zip-ties.	Measuring board
10-12 fence posts	Portable electronic balance (0.01 g resolution)
Post driver	Extra batteries for portable balance
3 – 4 Large hose clamps	Data sheets on write-in-the-rain paper
	Pencils
	Tarp for erecting temporary rain shelter
	Anesthetic
	Small plastic container for mixing anesthetic.

Commercially available fyke nets may be adapted to fyke traps by attaching a length of PVC pipe to the back of the cod end. Alternatively, a fyke trap can be constructed (Figure 9).

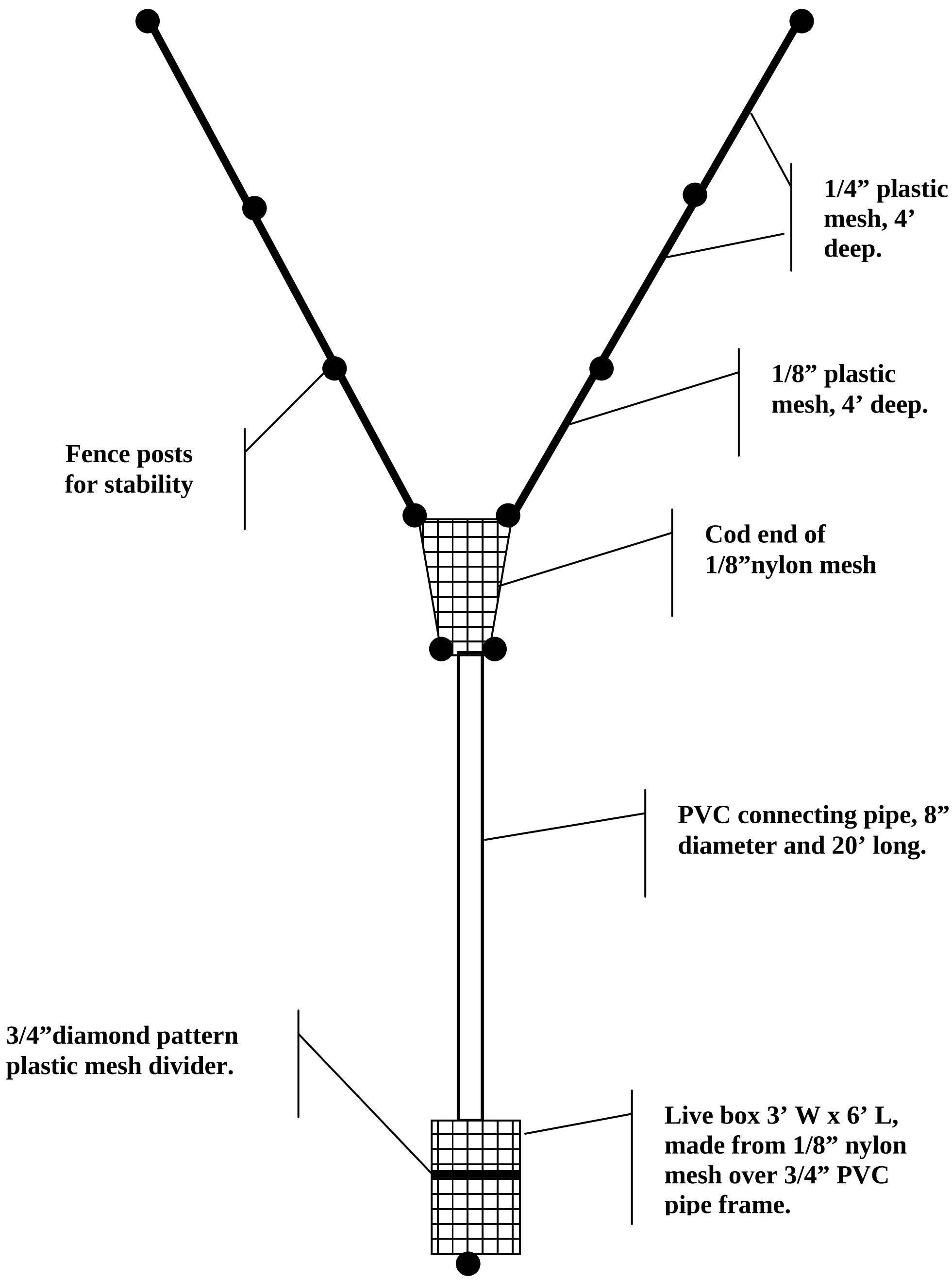


Figure 9. Diagram of fyke net smolt trap.

The fyke trap illustrated in figure xx consists of a set of wings, a cod end, tube and live box. Details on the construction of these parts of the trap are provided below.

Net wings are constructed from 4' by 8' plastic mesh panels sewn to a ¾" PVC pipe frame. The outer two panels on each side are constructed using ¼" mesh netting, while inner the panel on each side is made using 1/8" mesh.

Note that the mesh is extended 8" past the downstream end of each panel (Figure 10). This extension permits overlap

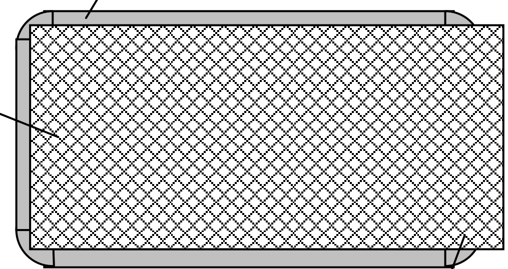


Figure 10. Diagram of fyke trap wing panel.

around fence posts driven in to support the trap and, when sewn into the adjacent panel, creates a seamless net panel.

Net wings are supported by eight fence posts. One post is placed on the upstream end, one of the downstream end and one at each panel juncture. Mesh panels are then secured to fence posts using electrical ties. Sand bags filled with stream gravel are stacked along the inner and outer side of the lower mesh panel for further stability.

The **cod-end** is made of 1/8 inch mesh nylon netting and is 3 ft high, 2 ft wide and 4 ft long. The mouth of the cod-end is kept open by sewing it to a 3 ft x 2 ft wide ¾ inch PVC rectangle frame. Behind the mouth, the cod-end is tapered down so that it fits snugly around an 8 inch diameter PVC pipe. A 6 inch wide collar of canvas or other heavy material is sewn to the netting to prevent chafing when the cod-end is attached to the PVC connecting pipe.

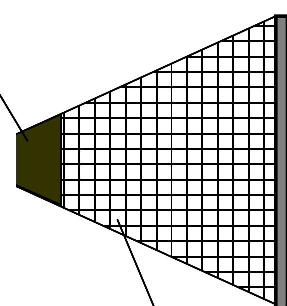


Figure 11. Diagram of fyke net cod-end.

The **PVC connecting pipe** is simply two sections of 8 inch diameter PVC pipe. In most streams, the length of pipe recommended is necessary to create a gradient differential between the cod-end and live box sufficient to carry fish into the live box.

The **live box** is 6 ft long, 3 ft wide and 3 ft tall and is constructed of ¼ inch square knotless nylon netting. It is divided into forward and rear compartments by ¾ inch diamond pattern knotless nylon netting.

The 8 inch PVC pipe is inserted through a fabric collar to within one foot of the mesh divider in the live box. In this position, water velocity is sufficient to carry smaller fish into the rear chamber of the live box. Larger fish may be impinged on this panel briefly, but are strong enough to overcome the water velocity.

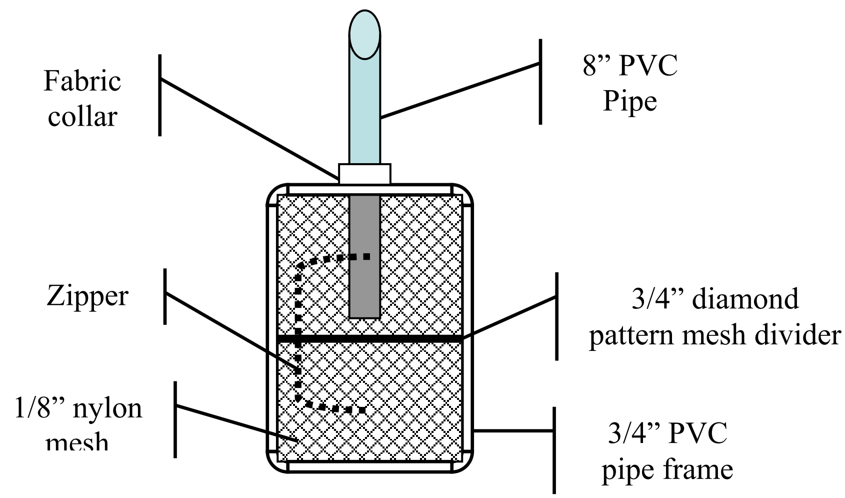


Figure 12. Diagram of smolt trap live box.

9. Migrant smolt trapping data sheet and metadata for smolt trapping.

Migrant smolt trapping data sheet.

Date:	Page ___ of ___
Time:	Site: (Lat/Long or UTM)
Stream name:	Personnel:
County:	Water temp:
	Stream stage height:
	Air temp:

Fry	**Total number**	**Smolts**	**Total number**
OC		**OC**	
OK		**OK**	
OM		**OM**	
OT		**OT**	
TR		**TR**	

Species	**Length (mm)**	**Weight (g)**	**Mark applied**	**Recapture mark**	**Mortality**	**Comment**

Metadata for migrant smolt trapping.

Item	Description
Date	Calendar date (MM/DD/YY)
Time	Military time (HHMM)
Stream name	Stream name on USGS 1:24,000 Quad. Map
County	California county name
Location	Coordinates of trap site in either latitude and longitude or UTM
Stream condition	Includes discharge or stage height if available, amount of debris visible, turbidity.
Page	Number pages consecutively
Personnel	Name of field personnel recording data
Species code	
OC	Cutthroat trout
OK	Coho salmon
OM	Steelhead
OT	Chinook salmon
TR	Trout too small (< 80 mm) to accurately identify
Total number	Total number of each species collected on that date
Length	Fork length in mm
Weight	Wet weight in g
Mark applied	Type and location of any mark applied to fish
Recapture mark	Type and location of mark on any recaptured fish
Mortality	Record if fish died during collection
Comment	Note any unusual conditions or circumstances.

AGE DISTRIBUTION OF JUVENILE STEELHEAD

1. Rational

Age of juvenile steelhead may be a useful measure for detecting a response to watershed restoration for several reasons. First, juvenile steelhead are widely distributed in coastal watersheds of California. Second, juvenile steelhead use fresh water habitat several years before smolting and migrating to the ocean. Third, smolt transformation in salmonids is regulated, in part, by size and will not occur if a fish has not reached some critical size (Groot and Margolis 1993).

The rational for using age of juvenile steelhead as a measure for detecting a response to watershed restoration is that growth should be slower under poor habitat conditions than under good conditions. With slower growth, more time will be required to reach the critical size for smolting, resulting in fish being older at the time of smolting. Extending this assumption, growth would hasten as restoration actions improve habitat conditions until age at smolting is eventually reduced.

This measure may be particularly suited to restoration actions that result in lower concentrations of turbidity, since turbidity is known to reduce feeding rates

2. Assumptions

Applying juvenile steelhead age distribution as a watershed response measure assumes that growth can be related to habitat condition. Several lines of evidence suggest this assumption may be valid. First, the average age of steelhead at smolting decreases with latitude (Figure 13). This relationship is likely a habitat response to water temperature and length of growing season. While not a reflection of habitat degradation, the relationship does indicate that age at smolting is sensitive to habitat conditions.

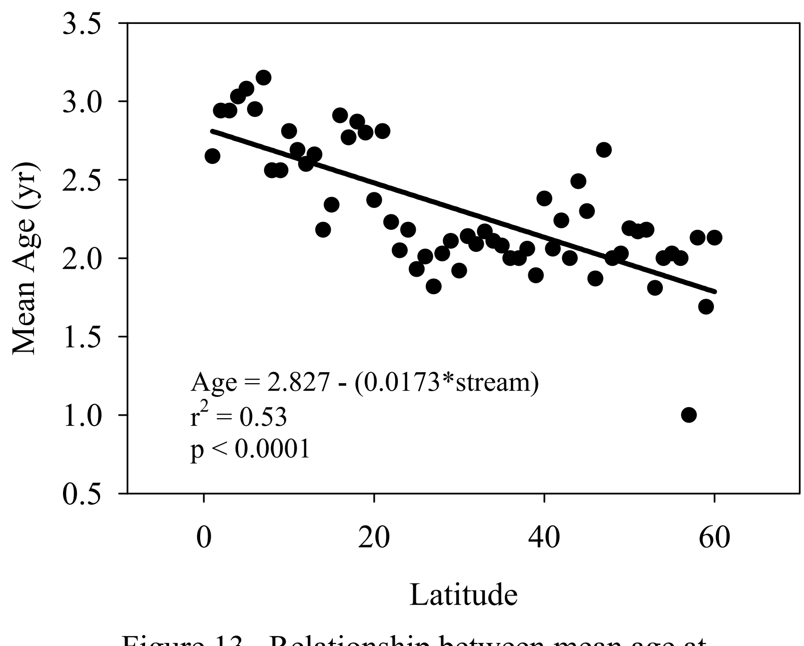


Figure 13. Relationship between mean age at smolting by steelhead and latitude for 60 Pacific Northwest streams. Data are from Busby et al 1996.

Second, we have found that maximum daily consumption (C_{max}), an important component in bioenergetics, in closely related cutthroat trout (*Oncorhynchus clarki*) is reduced as turbidity is increased (Figure 14). Moderate amounts of turbidity have also been shown to influence the bioenergetics of brook trout (*Salvalinus fontinalis*) (Swetka and Hartman 2001). These relationships suggest that turbidity negatively affects sight feeding by salmonids and could reduce growth.

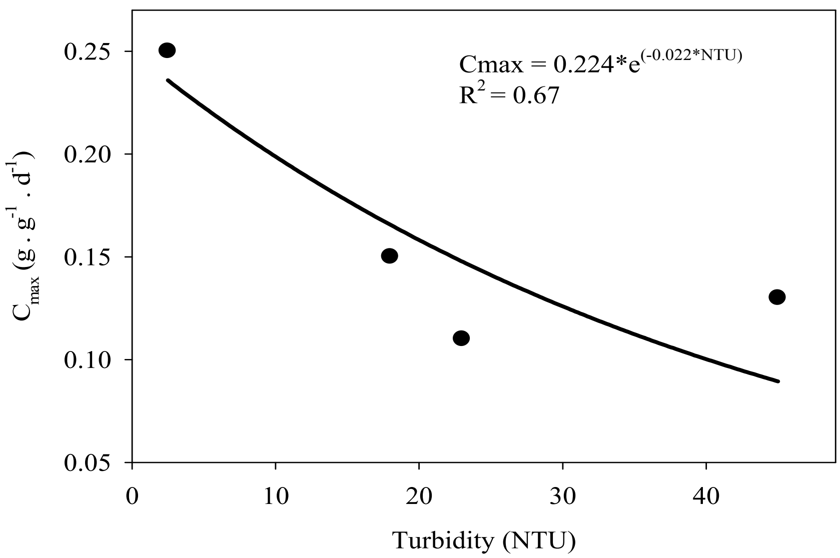


Figure 14. Relationship between Cmax and turbidity in coastal cutthroat trout.

3. Limitations

While intuitively appealing, the assumption that growth is related to habitat quality has not yet been rigorously tested. Multiple environmental factors such as water temperature, food available and density of juvenile salmonids influence growth and age at smolting. Testing this assumption is currently underway as part of the process of validating protocols for assessing watershed restoration.

4. Sampling Design

Juvenile steelhead for aging can be acquired from the distribution and abundance, presence sampling methods described elsewhere in this report, by smolt trapping or by other means. The sample of fish should be large enough and taken from multiple habitats so as to reflect conditions in the stream being samples. After a collection of fish has been obtained, two basic methods are available for age determination. First, one may use non-destructive sampling of scales to assign ages to individual fish (Frie 1982). Second, one may analyze the size distribution of populations in combination with aging from scales (Nielsen and Johnson 1983).

5. Methods

Methods for determining age structure involve recording the size distribution of juvenile steelhead collected and obtaining scales from a sub-sample of those fish for aging.

Step 1) Obtain a sample of fork lengths, in mm, from 100 or more juvenile steelhead, recording weight of these same fish is often useful as well.

Step 2) Collect a scale sample from 5 or more of the individual fish witin each 10 mm fork length category.

Step 3) Count the number of fish in each 10 mm fork length category and plot this length frequency distribution.

Step 4) Identify modes in the distribution and assign ages to each mode using known ages from scale samples.

Step 5) Determine the age of individual fish from scale samples and use these data to verify age modes as well as the uncertainty between ages.

a. Scales should be collected from mid-way between the back of the dorsal fin and the lateral line.
b. Collect scales from each fish by scraping a small knife blade from the back of the fish forward. Take a sample of 5-7 scales since some scales may present difficulties in aging due to false annuli, and other anomalies.
c. Using a 2 inch square of wax paper, wipe the scales from the knife blade, fold the wax paper and store the scale sample in a coin envelope.
d. Record the date, stream name, location within the stream, fish size and a unique number for the fish on the outside of the envelope.

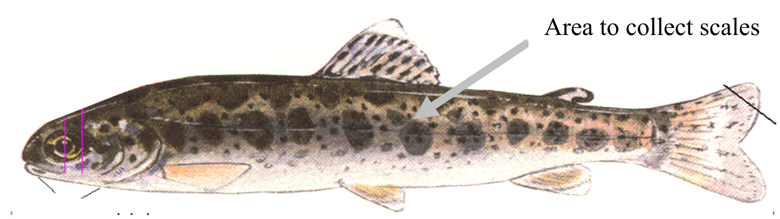


Figure 15. Location to collect scale samples from juvenile steelhead.

e. Later, mount the scales between two microscope slides and view them through a microscope or micro-fish reader to identify annuli.
f. Scales should be aged independently by two people, at least one having experience in aging fish scales.

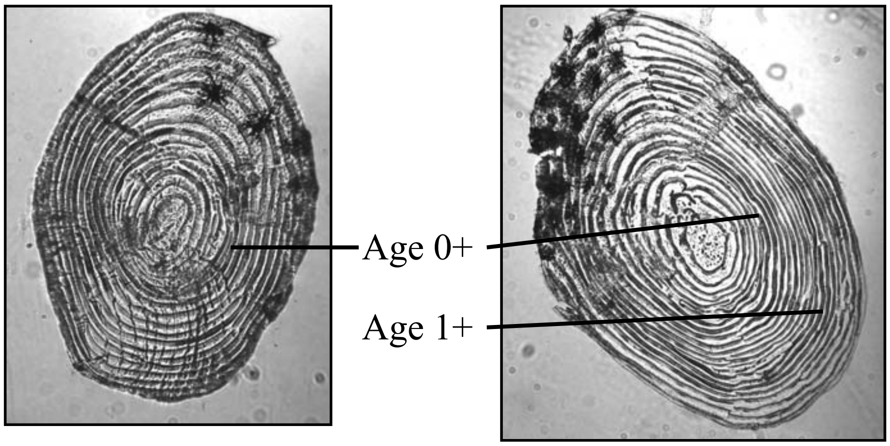


Figure 16. Example of juvenile steelhead scales showing an age 1+ fish on left and age 2+ fish on right.

6. Data Analysis

Age distributions can sometimes be easily distinguished from plotted data (Figure 16). However, modes in distribution that are well separated can be the result of too few samples being collected and not a clear size separation with age. With adequate sample sizes, all size ranges are typically represented and there is some overlap in size at age between modes (Figure 17). This overlap presents difficulty is assigning ages. Lack of a clearly discernable first annulus in some fish can also contribute to uncertainty in assigning age. Obtaining scales from fish whose age is known and using these known-age fish to calibrate age assignment can reduce uncertainty. This technique does, however, require marking fish and recapturing them after at least one winter season has elapsed.

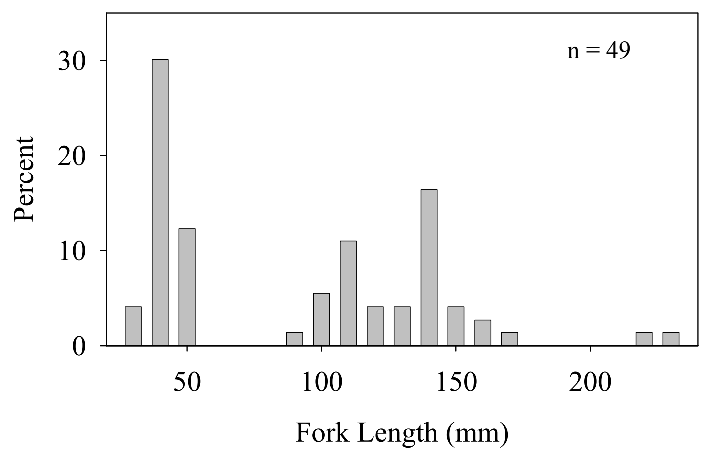


Figure 17. Length frequency distribution of juvenile steelhead from the South Fork Roach Creek, Humboldt County, California during July the 2002.

In general, we are less concerned with the total number of fish in each age category than we are with age at smolting. If the total number of fish in each age category is considered important, statistical methods may be employed to assign ages to individuals of size at age overlaps (Nielsen and Johnson 1983).

It should be noted, however, that accurate sample sizes combined with age determination allows for determining survival from one age to the next. Methods for determining survival from "catch curves" are described in Ricker (1975).

Clear guidelines for interpreting age at smolting are not presently available. However, most steelhead in California streams smolt at age 2. Using age 2 as the median, we can make qualitative inferences about age distributions that extend to ages 3 and 4, and conversely, those that are truncated toward age 1. We recommend the following interpretations of steelhead age distribution data:

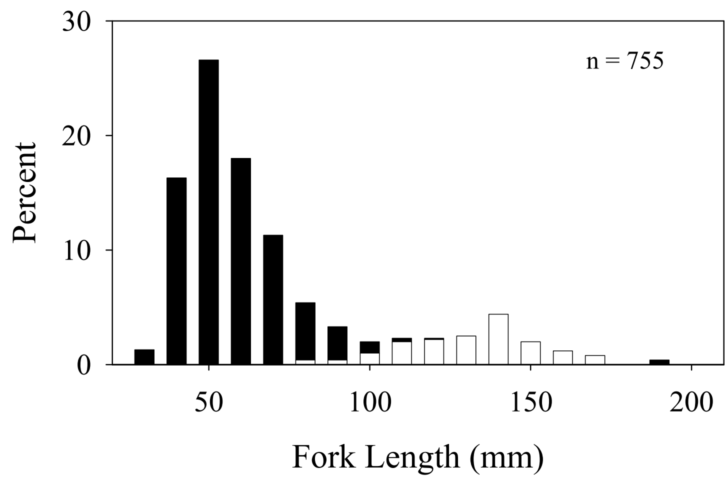


Figure 18. Length frequency distribution of juvenile steelhead from Bull Creek Humboldt County, California during August 2002.

1. For data sets spanning five or more years, age at smolting should be expected to decline as habitat improves. Thus, a trend of reduced age at smolting may be interpreted as a response to improved habitat.

2. For data sets covering less than five years, the proportion of juvenile steelhead in the population of age 3 or greater may indicate less than optimal habitat. This is not an absolute measure, but should be interpreted in the context of other California streams. The average age distribution of 16 populations of juvenile steelhead in streams extending from Santa Cruz County through Humboldt County illustrates that 85-90% of fish are probably age 1 and would be expected to smolt at age 2 (Figure). In contrast, a higher proportion of juvenile steelhead from Horse Mountain Creek, Humboldt County, were at age 2 or older in September and would be expected to smolt at age 3 or older.

Figure 18. Average age distribution of juvenile steelhead from 16 California streams and from Horse Mountain Creek, Humboldt County, California.

More rigorous methods for interpretation of these types of data will be dependent of gathering age distribution data from more streams representing different habitat conditions.

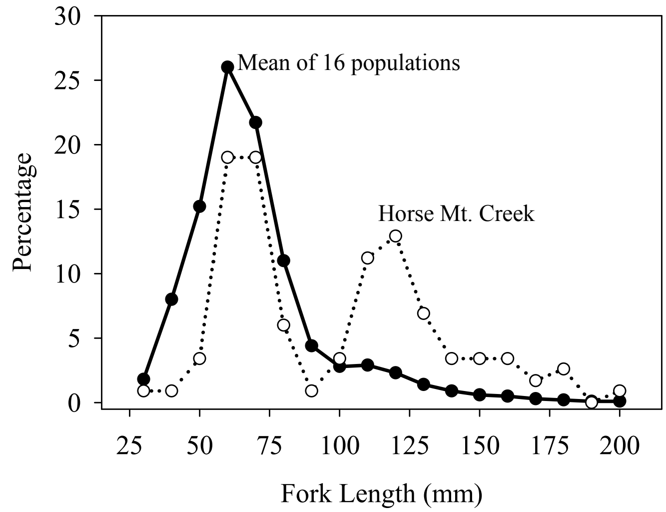


Figure 19. Average age distribution of juvenile steelhead from 16 California streams and from Horse Mountain Creek, Humboldt County, California.

7. Quality Assurance and Quality Control

Quality assurance and quality control procedures should be established for aging juvenile steelhead. These procedures should include elements of training in scale reading since experience is required to accurately assign ages from scales (Figure 15) Data entry and management, and independent assessment of methods should also be considered in QA/QC.

The training program should address:

1. identification of fish species likely to be encountered,
2. proper handling of fish,
3. scale sampling and
4. assigning ages to scales. .

QA/QC procedures in assigning ages to scales should include the verification of 100% of the original ages. That is, a second person without knowledge of ages assigned by the first person, reads scales previously aged and determines ages independently. Scale samples or photographs of scales taken with a microscope and imaging system should be archived for later review.

8. Personnel and Equipment Needed

Personnel	Materials
Two (2) persons	Portable balance with 0.01 g resolution
	Measuring board, 30 cm long with 1 mm increments
	Write-in-the-rain notebook or data sheets
	Pencils
	Coin envelops
	Wax paper
	Small knife
	Microscope or microfiche reader or optical analysis system

9. Data sheet for recording scale, length and weight data for juvenile steelhead and metadata for recording age data.

Field data sheet for recording scale collections, length and weight data.

Date:				Latitude	Longitude
Stream:			Lower:		
Basin:			Upper:		
County:			Crew:		

Fish species: steelhead

Fish No.	NSO	Habitat type	Distance (m)	Fork Length (mm)	Weight (g)	Age (1st reader initial)	Age (2nd reader initial)

Metadata for juvenile steelhead scale samples.

Item	Description
Date	Calendar date (MM/DD/YY)
Time	Military time (HHMM)
Stream name	Stream name on USGS 1:24,000 Quad. Map
Basin	Drainage basin that contains stream
County	California county name
Location	Coordinates of sampling site in either latitude and longitude or UTM
Page	Number pages consecutively
Personnel	Name of field personnel recording data
NSO	Number of habitat unit in “numerical sequential order”.
Habitat type	Pool, deep pool, run, riffle or other
Distance	Location of the sample in cumulative distance (m) from the start point.
Length	Fork length in mm
Weight	Wet weight in g

ADULT SALMON AND STEELHEAD ESCAPEMENT

1. Rational

The number of adult salmon or steelhead returning to a stream to spawn is defined as "escapement", meaning those adults that have escaped the fishery to reproduce. Estimates of escapement provide essential information on the size of populations. The number of adults escaping to spawn is influenced by mortality at all younger life history stages. Since habitat conditions in freshwater and the ocean influence survival, estimate
s of escapement are the often considered the ultimate measure of population response. These estimates of escapement are frequently used as an indicator of production for future generations of fish.

Escapement has been estimated using a variety of techniques. In larger rivers, aerial surveys or counts at dams are sometimes used estimate escapement. In smaller rivers and streams, carcass mark-recapture techniques, visual counts of live fish, and counts of redds constructed have all been used to estimate escapement or provide an index of the number of spawners. In addition to these methods, technological improvements in underwater video and hydroacoustic equipment are now being applied to estimating salmon escapement. These latter techniques offer promise, but their costs are currently beyond the scope considered for widespread use.

2. Assumptions

Here we describe methods for obtaining escapement estimates using carcass mark-recapture techniques, visual counts of live fish, and counts of redds constructed. Certain assumptions are inherent in each method.

The assumptions inherent in carcass mark-recapture techniques are:

1. The population is closed and carcasses are not immigrating into the area (drifting in from upstream),
2. Carcasses do not lose their marks between the time of release and recapture,
3. Marking carcasses does not affect the probability of their being re-sighted,
4. All marked and unmarked carcasses have an equal probability of emigrating, that is drifting out of the survey area or being removed by animals,
5. All marked and unmarked carcasses have an equal probability of being re-sighted,
6. Carcass surveys represent a random sample, where each of the possible combinations of marked and unmarked carcasses has an equal probability of occurring,
7. All marked carcasses re-sighted are identified and reported.

Assumptions in the technique using visual counts of live fish are:

1. Surveys begin before live fish enter the survey reach,
2. Surveys continue until live fish are no longer present in the survey reach,
3. Live fish in the survey reach are visible to observers,

4. Species of live fish can be distinguished by observers, and
5. Observer efficiency can be defined.

Assumptions in the technique using visual counts of redds are similar to those for live fish and are:

1. Surveys begin before fish construct redds in the survey reach,
2. Surveys continue until redds are no longer being constructed in the survey reach,
3. Redds in the survey reach are visible to observers,
4. Redds can be associated with the species constructing them, and
5. Observer efficiency in seeing redds can be defined.

3. Limitations

Estimating numbers of salmon or steelhead escaping may not be possible or may be difficult in some streams during some years. In streams with very small populations, estimating escapement using carcass mark-recapture methods may present statistical challenges if the number of re-sighted marked carcasses is small. Analysis of data from small populations may require consultation with a statistician familiar with mark-recapture experiments. Methods relying on visual observation of either live fish or redds may also be limited in streams that remain turbid for a substantial proportion of the spawning period. Finally, both carcass mark-recapture and visual observation methods require observers to regularly census survey reaches. This requires considerable labor and may not be possible during periods of high water.

4. Sampling Design

The objective for estimating escapement is often to estimate the number of adult fish returning to spawn in a tributary stream or some reach of importance. Sampling designs for reaches of streams that are not exceptionally long are typically to survey the entire reach. For visual observation methods, random subsampling can be employed if the objective is to estimate escapement for a steam or entire watershed that cannot be reasonably surveyed in its entirety. In the latter case, all the habitat in the survey area is first defined. Second, the survey area is divided into strata of similar size having similar physical attributes. Third, random reaches within each strata are selected to survey. Permission to access property may not be granted to some reaches. Because of this, it is advisable to select 20-30% more reaches than will be sampled. Having randomly selected a number of reaches in excess of the number desired will provide a valid process for selecting alternate reaches.

Sampling designs for larger rivers or watersheds can incorporate quantitative methods or a combination of quantitative methods and index sampling. Quantitative methods typically consist of intensive escapement estimates along survey reaches selected randomly from within the watershed. Alternatively, intensive surveys of selected reaches are sometimes combined with qualitative indices, such as single surveys during peak spawning activity, to provide information from a larger area.

5. Methods

It is helpful to define the amount of habitat within the survey reach before escapement estimates are initiated. This information can help define strata and determine sub-reaches that may be surveyed during a day. Distance upstream from the starting point may be measured by walking the survey reach using a hip chain. When measuring distances, affix plastic flagging to riparian vegetation at regular 50 – 100 m intervals and write the distance on the flagging with a waterproof marker. During later surveys, the distance location at which fish or redds are observed can then be estimated with relative accuracy. Recording information on the spatial distribution of fish and redds is valuable in identifying important spawning areas or habitats, as well as how areas used for spawning change with water flow or restoration actions.

Sampling should begin when the first adult salmon or steelhead enter the survey reach and continue until no adults are observed. Salmon usually complete spawning over a period of one- two months, steelhead populations may consist of two or more races that spawn during different periods.

Sampling frequency should be guided by the period of residence for individual adult fish. Gallagher (2003) reports average residence times of 11 days for coho salmon, 9.3 days for Chinook salmon and 12.6 days for steelhead in tributary streams. Ideally, surveys should be repeated at intervals that coincide with residence times. Thus, repeating surveys at 9-13 day intervals should be adequate if all three species are being considered. Decreasing the interval between surveys will improve estimates of escapement and, if possible, repeating surveys at 7-10 intervals is advisable if residence time is unknown.

Sampling during each of these periods involves two personnel walking the stream survey reach to record information.

For carcass capture-recapture estimates:

1. Record the number of carcasses found in the survey reach,
2. Tag each new carcass found with a uniquely number tag, an aluminum disc tag wired to the jaw bone is recommended since it is durable and biodegradable.
   Note: a variety of tags lend themselves to carcass tagging, numbered metal tags are only one option,
3. Record the location of the carcass, in meters from the starting point,
   Note: some surveys also include information on the habitat where carcasses are found (log jam, pool, etc.). These data can be used in calculating probabilities of re-sighting a carcass,
4. Record the gender of the carcass, size (fork length or standard length) and condition (Sykes and Botsford 1986; fresh, decayed or skeleton), and
5. On subsequent surveys, record the number of any tagged carcasses and mark newly sighted carcasses as above.

For estimates from live fish observations:

1. Record the number of each species of live fish observed in the survey reach,

2. Record the location of the fish, in meters from the beginning survey point, and
3. Record the gender of the fish and estimate its size.

For estimates from observations of redds:

1. Record the number of redds observed in the survey reach,
2. Record the species and gender of fish(es) on or associated with the redd,
3. Measure and record the length of the redd and its width at 2 or more locations for calculating area,
4. Record the location of the redd, in meters from the beginning survey point,
5. Using a sharpie permanent marker, record the date the redd was first observed, the species associated with the redd and a redd number on bright flagging tape. The redd number should be chronological, for either the survey season or survey date.
6. Tie the flagging tape to riparian vegetation near the redd where it is easily visible during subsequent surveys.

   Note: Surveyors should exercise care to avoid walking on redds, whether surveying for redds, carcasses or live fish.

The ability of observers to see fish and redds should be measured to provide an estimate of efficiency. Efficiency measurements are most easily accomplished by having separate observers sample random portions of a survey reach. Both observers record data separately and submit their results “blind”. Time elapsed between the actual survey and efficiency survey should be brief since adult fish may move. Efficiency tests should be conducted at least twice per season and preferably whenever flow conditions or numbers of fish change substantially.

6. Data Analysis

Analysis of escapement data involves developing an estimate of total population size using data from observations made at intervals during the period of spawning. Either carcasses or live fish may be used to estimate escapement. Estimating escapement from periodic counts of live fish has been accomplished using area-under-the-curve techniques (English et al. 1992). These methods are best suited to streams having a weir or other obstruction at which fish entering the stream may be counted. However, they can be employed on streams lacking a weir.

Capture-recapture methods are usually employed to estimate escapement from carcass data methods range from simple Lincoln type index to more rigorous statistical methods (e.g. Jolly-Seber model; Sykes and Botsford 1986, Schwarz et al. 1993). However, when working with low numbers of fish, assumptions of some of the more rigorous methods often cannot be met. We present the steps for calculating an estimate of escapement using the Lincoln type index in Box 6.1 and refer readers to the specialized literature on more rigorous methods.

Escapement estimates from observations of live fish

Escapement of adult salmon may be estimated from carcass capture – re-capture data as follows:

During sampling period 1 record:

1. n_1 - the total number of carcasses observed and
2. a_1 - the total number of carcasses marked.

During sampling period 2 record:

3. n_2 - the total number of carcasses observed,
4. r_2 - the total number of marked carcasses observed and
5. a_2 - the total number of new carcasses marked.

Calculate the estimated number of adults (N) in the area during the period as:

$$N = {a_1}^2 * \left[\frac{n_2 + 1}{r_2 + 1} \right]$$

The variance of this estimate is calculated as:

$$V = \frac{{a_1}^2 * (n_2 + 1) * (n_2 - r_2)}{(r_2 + 1)^2 * (r_2 + 2)}$$

During sampling period 3 record the same data recorded during period 2 and calculate N for the interval 2-3, continue this process until the period of sampling is covered.

Escapement estimates from observations of live fish

Escapement of adult salmon and steelhead may be estimated from live fish observations using an area-under-the-curve (AUC) techniques. This method is sensitive to residence time, but considered robust when observer efficiency can be validated (Perrin and Irvine 1990). The method consists of two components, an estimate of stream residence time and an estimate of aggregate residence time (total fish days). Escapement is then estimated from:

$$\hat{N}_{AUC,Spawner} = \frac{AUC_{Stpawner} * OE}{RT_{Spawner}}$$

where $\hat{N}_{AUC,Spawner}$ is the estimated escapement (population size), $RT_{Spawner}$ is the residence time of spawners in the stream, in days, OE is observer efficiency, and $AUC_{Spawner}$ is the total number of spawner days, calculated as:

$$AUC_{Spawner} = \sum t_i - t_{i-1} * \left[\frac{x_i - x_{i-1}}{2} \right]$$

where t_i is the ith day of the year and x_i is the number of spawners observed on the ith survey.

Escapement estimates from observations of redds

Early research in California suggests the number of redds found in a survey reach can be related to the number of adult female salmon escaping (Gallagher 2003). However, more research is needed to understand the sources and magnitude of variation around this relationship and to establish relationships between variables such as area of redd and number of females escaping. At present, using the mean number of redds per female reported by Gallagher (2003) to estimate adult escapement from redd counts is recommended. These are:

1. 1.25 redds per female coho salmon,
2. 1.00 redds per female Chinook salmon and
3. 1.93 redds per female steelhead

Assuming a 1:1 sex ratio, these estimates of the number of redds produced by each female must be multiplied by 2 to obtain an estimate of total adult escapement. When accurate observations of sex ratios are recorded during surveys, this multiplier can be adjusted. Previous studies of coho salmon have identified populations consisting of adult females, adult males and jack males in a ratio of 1:1:0.66. The number of redds observed must be multiplied by 2.66 to obtain an estimate of total fish, that is, females, adult males, and jack males. Similarly, male jack Chinook salmon have, on average, made up 16.8% of the fall run in the Klamath River during the period 1978-2005 (California Department of Fish and Game, unpublished data). Estimating total fish from redds in this Chinook salmon population would require using a multiplier of 2.40.

7. Quality Assurance and Quality Control

Quality assurance and quality control procedures should be established for all programs estimating salmon and steelhead escapement. These procedures should include:

.

Training that addresses,

1) safety practices in the field and hypothermia,
2) identification of adult salmonid species likely to be encountered,

The quality assurance plan for data entry and management should include,

1) data entry
2) data management
3) data analysis
4) chain of custody for data

The assurance for fish sampling should include independent assessment of efficiency as discussed above.

Data entry and management elements of QA/QC procedures should include the use of metric units of measure, proper data coding of field sheets and data entry. Procedures to verify the accuracy of recorded field data and data entry into an electronic format should be developed.

Spawning survey data sets are typically not large and it is recommended that an independent observer check 100% of the original entries.

8. Personnel and Equipment Needed

Personnel	Equipment and Materials
2 Persons	Pencils
	Flagging tape
	Sharpie permanent marker
Equipment and Materials	Number disc tags and wire
Waders	Hog rings or wire
Rain gear	Needle nose pliers
Polarized glasses	Hip chain
Measuring staff	
Write-in-the-rain notebook or data sheets	

9. Salmon and steelhead escapement data sheet and metadata for escapement.

Salmon and Steelhead escapement data sheet for live fish and carcasses.

Date:	Page ___ of ___
Time:	Site boundaries: (Lat/Long or UTM)
Stream name:	Personnel:
County:	Stream condition:

Distance	Species	Live No.	Estimated SL (mm)	Condition

Carcass No.	Carcass condition	Standard Length (mm)	Mark number	Recapture number

Salmon and Steelhead escapement data sheet for redds.

Date:	Page ___ of ___
Time:	Site boundaries: (Lat/Long or UTM)
Stream name:	Personnel:
County:	Stream condition:

Distance	Species Associated	Length (m)	Width 1 (m)	Width 2 (m)	Width 3 (m)

Metadata for salmon and steelhead escapement data sheet.

Item	**Description**
Date	Calendar date (MM/DD/YY)
Time	Military time (HHMM)
Stream name	Stream name on USGS 1:24,000 Quad. Map
County	California county name
Location	Coordinates of trap site in either latitude and longitude or UTM
Stream condition	Includes discharge or stage height if available, amount of debris visible, turbidity.
Page	Number pages consecutively
Personnel	Name of field personnel recording data
Distance	Distance in meters upstream from starting point.
Species code	
OK	Coho salmon
OM	Steelhead
OT	Chinook salmon
# Live	Total number of that species observed at that distance location.
# Carcass	Total number of that species carcasses observed at that distance location.
Carcass condition	
1	Recently died, eyes clear and flesh firm
2	Eyes are cloudy, but flesh still firm
3	Eyes are cloudy and flesh is soft
4	Eyes are cloudy and flesh is very soft, beginning to slough off
5	Only the head and part of the skeleton remain
Mark number	Number of mark applied to that carcass.
Recapture number	Number of mark existing on that re-sighted carcass.

LITERATURE CITED

Blackwell, B.G., M.L. Brown and D.W. Willis. 2000. Relative weight (Wr) status and current use in fisheries assessment and management. Reviews in Fisheries Science 8:1-44.

Busby, P.J., T.C. Wainwright, G.J. Bryant, L.J. Lierheimer, R.S. Waples, F.W. Waknitz and I.L. Lagomarsino. 1996. Status review of west coast steelhead from Washington, Oregon and California. National Marine Fisheries Service, Technical Memorandum NMFS-NMFSC-27. Seattle, WA.

Cho, G.K., and D.D. Heath. 2000. Comparison of tricain methanesulfonate (MS222) and clove oil anaesthesia effects on the physiology of juvenile Chinook salmon *Oncorhynchus tshawytscha*. Aquaculture Research 31:537-546.

Dolloff, C.A., D.G. Hankin and G.H Reeves. 1993. Basinwide estimation of habitat and fish populations in streams. General Technical Report SE-83, U.S. Department of Agriculture, Forest Service, Southeastern Forest Experiment Station, Asheville, NC. 25 pp.

English, K.K., R.C. Bocking and J.R. Irvine. 1992. A robust procedure or estimating salmon escapement based on the area-under-the-curve method. Canadian Journal of Fisheries and Aquatic Sciences 49:1982-1989.

Flosie, G., S. Downie, J. Hopelain, M. Bird, R. Coey and B. Collins. 2002. California salmonid stream habitat restoration manual, third edition. California Department of Fish and Game, Inland Fisheries Division, Sacramento, CA.

Frie, R.V. 1982. Measurement of fish scales and backcalculation of body lengths using a digitizing pad and microcomputer. Fisheries 7:5-8.

Gallagher, S. P. 2003. Discrimination of Chinook (Oncorhynchus tshawytscha) and coho (O. kisutch) salmon and steelhead (O. mykiss) redds and evaluation of the use of redd data for estimating escapement in several unregulated streams in northern California. California Deparment of Fish and Game, Fort Bragg, CA.

Gerstung, E. R. 1997. Status of coastal cutthroat trout in California. Pages 43-56 in J. D. Hall, P. A. Bisson and R. E. Gresswell, editors. Sea-run cutthroat trout: biology, management, and future conservation. Oregon Chapter, American Fisheries Society, Corvallis, Oregon.

Gibbs, J.P. 2003. Monitor: online software for estimating the power of population monitoring programs to detect trends in plant and animal abundance. http://www.mbr-pwrc.usgs.gov/software/monitor.html.

Groot, C., and L. Margolis. 1991. Pacific salmon life histories. University of British Columbia Press, Vancouver, British Columbia.

Hankin, D.G. 1984. Multistage sampling designs in fisheries research: applications in small streams. Canadian Journal of Fisheries and Aquatic Sciences 41: 1575-1591.

Hankin, D. G., and G. H. Reeves. 1988. Estimating total fish abundance and total habitat area in small streams based on visual estimation methods. Canadian Journal of Fisheries and Aquatic Sciences 45:834-844.

Keeley, E.R., and C.J. Walters. 1994. The British Columbia watershed restoration program: summary of the experimental design, monitoring and restoration techniques workshop. Ministry of the Environment, Lands and Parks and Ministry of Forests, Watershed Restoraton management Report No. 1. 34 pp.

Ketcham, B.J., G.G. Brown, D. Fong, and M.L. Reichmuth. 2004 Spring outmigrant smolt trapping program. Standard operating procedure 2. Prepared for San Francisco Area Network Inventory and Monitoring Program, National Park Service, Point Reyes Station, CA.

MacKenzie, D.I., J.D. Nichols, G.B. Lachman, S. Droege, J.A. Royle and C.A. Langtimmm. 2002. Estimating site occupancy rates when detection probabilities are less than one. Ecology 83:2248-2255.

Montgomery, D.C. 2000. Design analysis of experiments. John Wiley and Sons, New York. 672 pp.

Montgomery, D.R., and J.M. Buffington. 1998. Channel processes, classification and response. Pages 13-42 *In* R.J. Naiman and R.E. Bilby, Editors, River Ecology and Management: Lessons from the Pacific Coastal Ecoregion. Springer, New York.

Murphy, B.R., M.L. Brown and T.A. Springer. 1990. Evaluation of the relative weight (Wr) index, with new application to walleye. North American Journal of Fisheries Management 10:85-97.

Nielsen, L.A., and D.L. Johnson (Eds.). 1983. Fisheries Techniques. American Fisheries Society, Bethesda, MD.

NOAA. 2000. Guidelines for electrofishing waters containing salmonids listed under the endangered species act. National Marine Fisheries Service, Santa Rosa, CA. (http://www.noaa.gov/ESA-Salmon-Regulations-Permits/4d-Rules/upload/electro2000.pdf)

ONRC. 2000. The scientific basis for validation monitoring of salmon for conservation and restoration plans. Report of the validation Monitoring Panel to the Olympic Natural Resources Center, College of Forest Resources, University of Washington, Seattle.

Reeves, G.H., D.B. Hohler, B.E. Hansen, F.H. Everest, J.R. Sedell, T.L. Hichman and D. Shively. 1997. Fish habitat restoration in the Pacific Northwest: Fish Creek of Oregon. Pages 335-359 In: J.E. Williams, C.A. Wood and M.P. Dombeck (eds.) Watershed Restoration: Principles and Practices. American Fisheries Society, Bethesda, MD.

Reeves, G.H., F.H. Everest and J.R. Sedell. 1991. Responses of anadromous salmonids to habitat modification: how do we measure them? Symposium 10, American Fisheries Society, Bethesda, MD. Pp. 62-67.

Reisburger, B. 2003. Reduced juvenile coho salmon mortality in migrant smolt traps fitted with experimental live boxes. M.S. Thesis, Humboldt State University, Arcata, CA. *In preparation*.

Ricker, W. E. 1975. Computation and interpretation of biological statistics of fish populations. Bulletin 191, Fisheries Research Board of Canada, Department of the Environment, Fisheries and Marine Service, Ottawa.

Roni, P., M. Liermann and A. Steel. 2003. Monitoring and Evaluating Fish Response to Instream Restoration. Pages 318-339 *in* D.R. Montgomery, S. Bolton, D.B. Booth and L. Wall (eds.) Restoration of Puget Sound Rivers. University of Washington Press, Seattle.

Shapovalov, L. and A.C. Taft. 1954. The life histories of the steelhead rainbow trout (Salmon gairdneri gairdneri) and silver salmon (Oncorhynchus kisutch) with special reference to Waddell Creek, California, and recommendations regarding their management. Fish Bulletin No. 98, California Department of Fish and Game, Sacramento.

Schreck, C.B., and P.B. Moyle, editors. 1990. Methods for fish biology. American Fisheries Society, Bethesda, Maryland.

Schwarz, C.J., R.E. Bailey, J.R. Irvine and F.C. Dalziel. 1993. Estimating salmoon spawning escapement using capture-recapture methods. Canadian Journal of Fisheries and Aquatic Sciences 50:1181-1197.

SER. 2002. The SER primer on ecological restoration. Science and Policy Working Grooup, Society for Ecological Restoration. 9 pp.

Summerfelt, R.C., and L.S. Smith. 1990. Anesthesia, surgery, and related techniques. Pages 213-272 *In*: C.B. Schreck and P.B. Moyle, editors, Methods For Fish Biology. American Fisheries Society, Bethesda, MD.

Sykes SD, and LW Botsford. 1986. Chinook salmon, *Oncorhynchus tshawytscha*, spawning escapement based on multiple mark-recapture of carcasses. Fishery Bulletin: Vol. 84, No. 2: 261-270.

Swetka, J. A., and K. J. Hartman. 2001. Effects of turbidity on prey consumption and growth in brook trout and implications for bioenergetics modeling. Canadian Journal of Fisheries and Aquatic Sciences 58:386-393.

Todd, G.L. 1994. A lightweight, inclined-plane trap for sampling salmon smolts in rivers. Alaska Fishery Research Bulletin 12:168-175.

Walters, C.J., J.S. Collie and T. Webb. 1988. Experimental designs for estimating transient responses to management disturbances. Canadian Journal of Fisheries and Aquatic Sciences 45:530-538.

Walters, C.J., J.S. Collie and T. Webb. 1989. Experimental Designs for Estimating Transient Responses to Habitat Alteration: Is it Practical to Control for Environmental Interactions? Pages 13-20 *in* C.D. Levings, LB. Holtby and M.A. Henderson (eds.) Proceedings of the National Workshop on Effects of Habitat Alteration on Salmonid Stocks. Canadian Special Publication of Fisheries and Aquatic Sciences 105.

Wege, G.J., and R.O. Anderson. 19778. Relative weight (Wr): a new index of condition for largemouth bass. Pages 79-91 *In* G.D. Novinger and J.G. Dillard, editors. New approaches to the management of small impoundments. American Fisheries Society, North Central Division, Special Publication 5, Bethesda, MD.

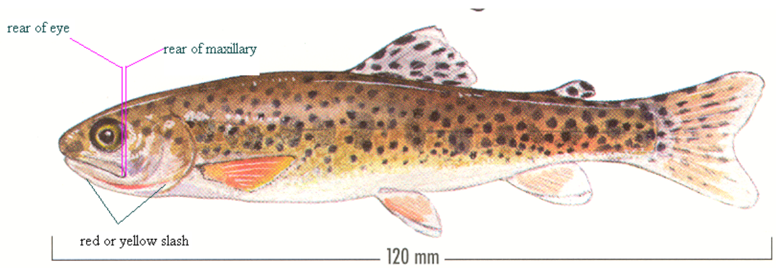
APPENDIX A

Key to Juvenile Salmonids Occurring in California

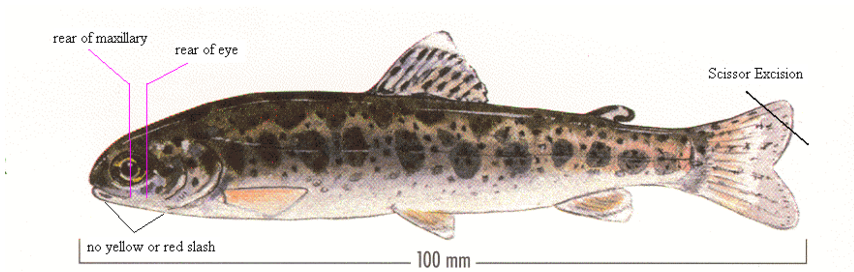
Key to Juvenile Salmonids Occurring in California

1) Anal fin higher than long with 8-12 rays, dorsal fin with large spots …………. 1a

   a) Maxillary extending past the posterior margin of the eye, red or yellow hyoid mark under jaw, small hyoid teeth at base of tongue, occurs only in Humboldt and Del Norte Counties
   ……………………………………………………. Cutthroat trout, *O. clarkii*

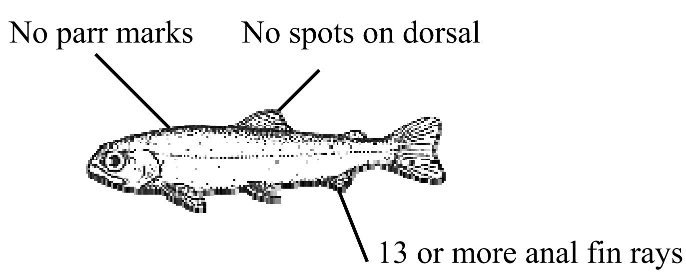


   b) Maxillary short, not extending past posterior margin of the eye, no red or yellow hyoid mark under jaw, parr marks nearly circular
   ………………………………………………………Steelhead, *O. mykiss*

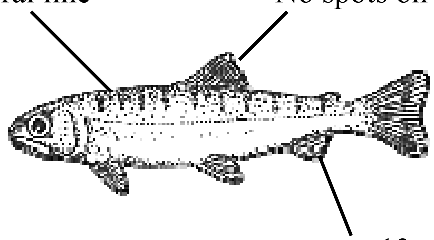


2) Anal fin longer than high with 13 or more rays, dorsal fin lacking large spots ……2a

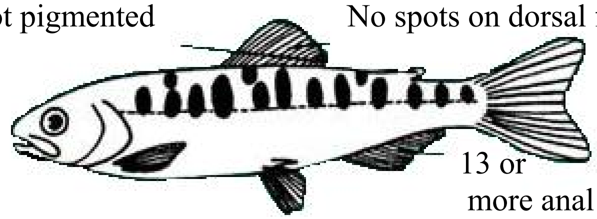
   a) Parr marks lacking, fry small, length about 45 mm, rare in California
   ……………………………………..……………. Pink salmon, *O. gorbuscha*



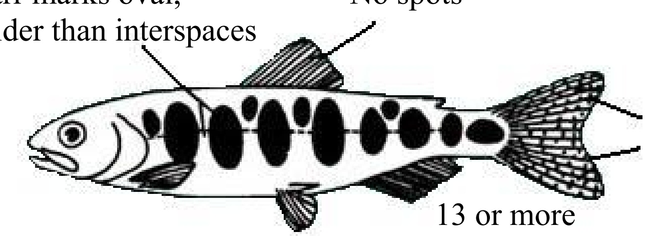
b) Parr marks present but faint and short, not extending below the lateral line, sides below lateral line iridescent green, length about 40 mm, rare in California
……………………………………………………..…….. Chum salmon, *O. keta*



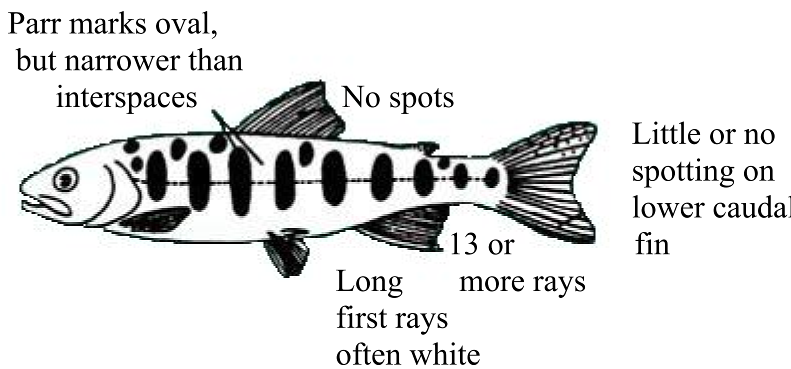
c) Parr marks present and sharp but short, not extending below the lateral line and faint, adipose fin clear, not pigmented, length 80-125 mm, occasionally present in California
……………………………………………………….. Sockeye salmon, *O. nerka*



d) Parr marks present and large, centered on the lateral line, and larger than interspaces between, anal fin not pigmented, spots on both upper and lower lobe of caudal fin, anterior rays of anal fin not elongated
………………………………………………….. Chinook salmon, *O. tshawytscha*



e) Parr marks present and large, centered on the lateral line, but narrower than interspaces between, anal fin pigmented between rays, spots only on upper lobe of caudal fin, anterior rays of anal fin elongated and often white, occurs from Santa Cruz northward
……………………………………………………….…….. Coho salmon, *O. kisutch*



APPENDIX B

Fish Community Diversity in Coastal Streams and Rivers

Fish Community Diversity in Coastal Streams and Rivers

The freshwater fish community of coastal California is more diverse than is often recognized. Total numbers of species that have been documented ranges from 49 in the North Coast Region (Table 5) to 34 in the South Coast Region (Moyle 2002). This diversity is, however, inflated by introduced alien species. Of the 63 total species of freshwater fish inhabiting coastal watersheds in California, 32 are alien (Table 6).

Freshwater fish communities of California's coastal watersheds are characterized by a high degree of anadromy. Two-thirds of the native species found in North Coast Region watersheds display anadromous of amphidromous life histories (Moyle 2002), and half or more of native species in other coastal regions display this life history. Furthermore, many of these anadromous species have or had distinct races that entered watersheds during different periods. The preponderance of this life history among fishes in coastal watersheds of California resulted in anadromous species being present in some rivers throughout the year.

The overwhelming number of alien species introduced to California's coastal watersheds not anadromous, the lone exception being American shad (Table 6). The impact of these introduced alien fish species on native fishes remains largely unknown, but alien species likely compete with native species for food and habitat space and act as predators on juvenile life stages of native fishes. Introductions of alien fishes have also clearly shifted the life history composition of coastal freshwater fish communities.

Table 5. Number of native, alien and anadromous[1] freshwater fish species occurring in coastal regions of California, Regions listed in headings are north coast (NC), San Francisco Bay (SFB), central coast (CC) and south coast (SC) of California.

Name	NC	SFB	CC	SC
Native species	24	20	11	7
Native species introduced from another California region	4	0	3	1
Alien species	21	25	20	26
Total species	49	45	34	34
Native anadromous species	16	10	7	4
Alien anadromous species	1	1	0	0
Total anadromous species	17	11	7	4

[1] Native anadromous species includes two amphidromous sculpin species.

Table 6. List of native (N) and introduced (I) freshwater fish species occupying coastal watersheds of the north coast (NC), San Francisco Bay (SFB), central coast (CC) and south coast (SC) of California. Data from Moyle 2002.

Name	NC[1]	SFB	CC	SC
Lampreys, Petromyzontidae				
Pacific lamprey, *Lampetra tridentate*	N	N	N	N
Klamath River lamprey, *Lampetra similes*	N			
River lamprey, Lampetra ayresi	N	N		
Western brook lamprey, *Lampetra richardsoni*	N	N		
Sturgeons, Acipenseridae				
White sturgeon, *Acipenser transmontanus*	N	N		
Green sturgeon, *Acipenser medirostris*	N	N		
Herrings, Clupeidae				
Threadfin shad, *Dorosoma petenense*		I	I	I
American shad, *Alosa sapidissima*	I	I		
Minnows, Cyprinidae				
Tui chub, *Siphateles bicolor*	I			
Arroyo chub, *Gila orcutti*			I	N
Hitch, *Lavinia exilicauda*	N	N	N	
California roach, *Lavinia symmetricus*	N	N	N	
Sacramento blackfish, *Orthodon microlepidotus*	I	N	I	I
Sacramento splittail, *Pogonichthys macreolepidotus*		N		
Hardhead, *Mylopharodon concephalus*	I	N		
Sacramento pikeminnow, *Ptychocheilus grandis*	I	N	I	
Speckled dace, *Rhinichthys osculus*	N		N	N
Golden shiner, *Notemigonus crysoleucas*	I	I	I	I
Fathead minnow, *Pimephales promelas*		I		I
Red shiner, *Cyprinella lutrensis*		I		I
Goldfish, *Carassius auratus*		I	I	I
Carp, *Cyprinus carpio*		I	I	I
Suckers, Catostomidae				
Santa Ana sucker, *Catostomus santaanae*				N
Sacramento sucker, *Catostomus occidentalis*	N	N	N	
Klamath smallscale sucker, *Catostomus rimiculus*	N			
Bullhead Catfishes, Ictaluridae				
Black bullhead, *Ameiurus melas*	I	I	I	I
Brown bullhead, *Ameiurus nebulosus*	I	I	I	I
Yellow bullhead, *Ameiurus natalis*				I
White catfish, *Ameiurus catus*	I	I	I	I
Channel catfish, *Ictalurus punctatus*	I	I	I	I
Blue catfish, *Ictalurus furcatus*		I	I	I
Smelts, Osmeridae				
Wakasagi, *Hypomesus nipponensis*	I	I		
Eulachon, *Thaleichthys pacificus*	N			

Continued

Table 6. (concluded).

Name	NC	SFB	CC	SC
Salmon and Trout, Salmonidae				
Coho salmon, *Onchorhynchus kisutch*	N	N	N	
Chinook salmon, *Onchorhynchus tshawytscha*	N	N	N	
Sockeye, *Onchorhynchus nerka*	N			
Pink salmn, *Onchorhynchus gorbuscha*	N			
Chum salmon, *Onchorhynchus keta*	N			
Steelhead, *Onchorhynchus mykiss*	N	N	N	N
Cutthroat trout, *Onchorhynchus clarki*	N			
Brown trout, *Salmo trutta*	I		I	I
Brook trout, *Salvelinus fontinalis*	I			
Silversides, Atherinopsidae				
Inland silverside, *Menidia beryllina*	I	I	I	I
Livebearers, Poeciliidae				
Western mosquitofish, *Gambusia affinis*	I	I	I	I
Sticklebacks, Gasterosteidae				
Threespine stickleback, *Gasterosteus aculeatus*	N	N	N	N
Brook stickleback, *Culea inconstans*	I			
Sculpins, Cottidae				
Prickly sculpin, *Cottus asper*	N	N	N	N
Coastrange sculpin, *Cottus aleuticus*	N	N	N	
Riffle sculpin, *Cottus gulosus*	N	N		
Marbled sculpin, *Cottus klamathensis*	N			
Pacific staghorn sculpin, *Leptocottus armatus*	N	N	N	N
Sunfishes, Centrarchidae				
Sacramento perch, *Archoplites interruptus*	I	N		
Bluegill, *Lepomis macrochirus*	I	I	I	I
Redear sunfish, *Lepomis microlophus*	I	I	I	I
Pumpkinseed, *Lepomis gibbosus*	I	I	I	I
Green sunfish, *Lepomis cyanellus*	I	I	I	I
White crappie, *Pomoxis annularis*		I	I	I
Black crappie, *Pomoxis nigromaculatus*	I	I	I	I
Largemouth bass, *Micropterus salmoides*	I	I	I	I
Smallmouth bass, *Micropterus dolomieu*	I	I	I	
Spotted bass, *Micropterus punctulatus*				I
Redeye bass, *Micropterus coosae*				I
Perches, Percidae				
Yellow perch, *Perca flavescens*	I	I		
Temperate bass, Moronidae				
Striped bass, *Morone saxatilis*		I		
Gobbies, Gobiidae				
Yellowfin goby, *Acanthogobius flavimanus*		I		I

[1]Notation as in Table 5.