Field Manual

Okanogan Basin Monitoring and Evaluation Program

Rotary Screw Trap Protocols

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Prepared by:

The Colville Confederated Tribes

Michael D. Rayton
Paul Wagner(KWA)

SECTION 2. SMOLT TRAPPING

Protocol adapted from; *Seiler and Volkhardt, 2005, and Murdock et al. 2001.*

SUMMARY

In accordance with local, state, federal and tribal agency regulations, investigators will use floating, rotary screw traps to collect downstream-migrating smolts for estimating the total number (abundance) of smolts produced within the watershed or basin. Traps will operate at minimum the entire period of the smolt emigration. Trapping efficiency will be estimated throughout the trapping period by using a mark-recapture methodology. Methods for operating the trap, estimating trap efficiency, and determining the frequency at which efficiency tests will be conducted are described in Murdoch et al. (2000). Numbers of smolts will be reported for populations or subpopulations. The Fulton-type condition factor, a measure used to describe the well-being of smolts within a population or sub-population, will be estimated from length and weight measurements taken from captured fish. Genetic samples may be collected to characterize (via DNA microsatellites) the within- and between-population genetic variability of smolts.

PURPOSE

> Operating a downstream migrant trap allows investigators to sample salmonids produced in a watershed or tributary over time. The sample in itself is valuable as it documents the presence or absence of migrating juveniles. The sample can also determine the age, the condition, timing, species, rearing history and genetic characteristics at migration. Furthermore, if the location of the trap and hours of operation are held reasonably constant from year to year, catch of a given species or catch per unit effort can be used as an index of downstream migrant production (Seiler and Volkhardt, 2005).

Trapping information can be used to create estimates of the total freshwater production by using a simple mark-recapture population estimation methodology. The proportion of marked fish appearing in a random sample estimates the proportion of marked fish in the total population. The proportion captured, or trap efficiency, is estimated by conducting a series of trap efficiency experiments over the trapping season (Seiler and Volkhardt, 2005). Trap efficiencies can vary from day to day as discharge fluctuates, thus requiring frequent calibration.

This protocol describes methods used to achieve estimates of wild, downstream-migrant salmonid production using a rotary screw trap. Since a rotary trap samples only the upper portion of the water column, they are generally not very useful for capturing species that migrate along the bottom of the river (e.g., lamprey). Traps can be scaled to operate in various sized streams, but are most commonly used in streams that are too large or powerful to employ a fence weir (e.g., ~10 to 15-m or larger channels) (Seiler and Volkhardt, 2005).

The rotary screw trap is used in medium to large rivers. The screw trap consists of a cone covered in perforated plate that is mounted on a pontoon barge (Figure 1). Within the cone are two tapered flights that are wrapped 360-degrees around a center shaft. The trap cone is oriented with the wide end facing upstream and uses the force of the river acting on the tapered flights to rotate the cone about its axis. Downstream migrating fish are swept into the wide end of the cone (typically either 5 ft or 8 ft in diameter) and are gently augured into a live box at the rear of the trap. A winch is used to adjust the fore elevation of the cone (Seiler and Volkhardt, 2005).

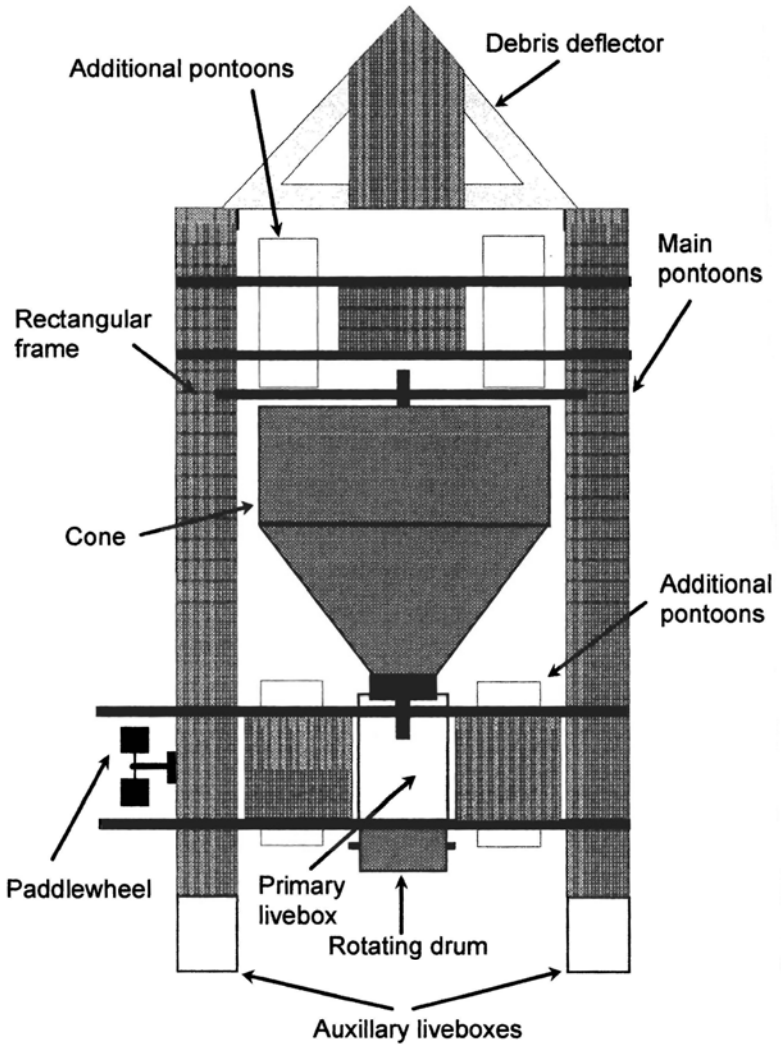

Figure 1. Rotary Screw Trap.

BACKGROUND

Traditionally, fishery managers have relied on escapement estimates to monitor anadromous salmonid population status and management effectiveness (Ames and Phinney 1977; Beidler and Nickelson 1980; Hilborn et al. 1999). In many salmon-bearing systems, population abundance is only monitored during the spawning stage. By estimating population abundances at earlier life stages researchers are able to partition survival among life-stages and develop hypotheses for restoration actions (Moussalli and Hilborn 1986, Mobrand et al. 1997).

Monitoring smolt abundance is particularly powerful since it enables the partitioning of mortality between the freshwater, egg-to-smolt life stages, and the marine life stages of smolt-to-adult (Seiler and Volkhardt, 2005). Juvenile fish traps have also been used to estimate the abundance, timing, size, survival, and behavior of downstream-migrant anadromous salmonids (Tsumura and Hume 1986; Baranski 1989; Orciari et al. 1994; Thedinga et al. 1994; Letcher et al. 2002; Wagner et al. 1963; Hartman et al. 1982; Orciari et al. 1994; Olson et al. 2001; Schoeneman et al. 1961; Wagner et al. 1963; Tsumura and Hume 1986; Olsson et al. 2001; Letcher et al. 2002; Brown and Hartman 1988; Roper and Scarnecchia 1996).

While estimating smolt abundance is the most common reason for operating a screw trap, the collection of downstream migrants also has wide utility. Traps can be used to monitor the effects of river management on wild stocks, such as the effectiveness of diversion, lock, and dam management. Traps can also be used to validate assumptions regarding the effect of watershed restoration programs and land-use policies on fish populations and to assess survival between life stages, such as egg-to-smolt survival or parr-to-smolt over-winter survival (Seiler and Volkhardt, 2005).

In addition to monitoring wild populations, traps are useful for evaluating hatchery programs and hatchery/wild fish interactions. These studies may include evaluating the instream survival of hatchery fish following release and evaluating treatments such as rearing strategy, release timing, release location, and flow manipulation on groups of hatchery fish. These later uses can evaluate hatchery supplementation strategies and avoidance of hatchery and wild fish interactions. In addition to abundance estimates, investigators use scoop and screw traps to collect samples of downstream migrants for such purposes as genetics sampling, fish disease research, predation (gut content) evaluations, and wild stock marking and tagging projects (Seiler and Volkhardt, 2005).

On the west coast of the United States and Canada, juvenile fish traps have primarily been used to estimate the natural production of juvenile coho (*Oncorhynchus kisutch*), sockeye (*O. nerka*), and steelhead (*O. mykiss*) from 5th order and smaller basins (Nickelson 1998). Nevertheless, with careful planning reasonably accurate production estimates have been obtained when 6th order and larger systems have been trapped (Schoeneman et al. 1961, Thedinga et al. 1994). For example, side-by-side scoop and screw traps have been used to successfully yield estimates of yearling coho and sub-yearling chinook migrants since 1990 in the Skagit River, a 7th order basin (Seiler et al. 2003).

SAFETY

When positioned in the river, screw traps (and the associated rigging) represent a hazard to boaters, float tubers, and swimmers. Wires and cables should be marked with bright colored flagging so as to be easily viewed by river users. Signs should be positioned both upstream and downstream of the trap to instruct boaters how to safely avoid the trap. Other protective measures may include flashing lights to improve the visibility of the trap and deflectors to help prevent water users and large woody debris from entering the trap (Seiler and Volkhardt, 2005).

A minimum of two persons shall operate the trap at any given time. Life jackets shall be worn at all times by personnel while traveling to and from or while operating the trap. Standard precautions should be taken by personnel to keep hands and loose clothing away from the cone and axle and other moving trap parts during trap operation.

EQUIPMENT

Trap/pontoon structure, anchor cables, boat (if necessary to reach trap), dip nets, fish anesthetic (MS 222), marking devices (scissors, dye, etc.), buckets (for collecting and working fish), brush/water pump for cleaning trap, flood lights (for night work), aeration equipment

SITE SELECTION

If the natural production of salmon is to be monitored, selection of trapping sites should be viewed from a variety of scales. At the watershed scale the river or stream should either be devoid of hatchery fish or all hatchery fish should be identifiable so that wild fish can be enumerated. Precision of the estimates increases with higher trap efficiency (i.e. proportion of migrants captured); therefore it is generally better to select sites where a higher proportion of the total flow can be screened through the trap. This becomes a trade-off, however, if the trap is placed below a hatchery release site since higher trap efficiencies can result in very large numbers of hatchery fish entering the trap following a fish release. Where this occurs, good communication between the trap operators and hatchery staff must be maintained to avoid a fish kill. In general, it is best to avoid these situations when choosing a trap site.

Another consideration when selecting watersheds is the hydrologic pattern of the basin. Flow is dependent on variables such as landform, geology, land cover, climate, and precipitation patterns, which of course, cannot be controlled. Since trap efficiency and migration rates often change dramatically with flow, rivers exhibiting a flashy hydrograph are very difficult to trap due to large fluctuations in flow conditions and debris loads. The effect of these variables on stream discharge needs to be considered when estimating total freshwater production (Seiler and Volkhardt, 2005).

Within a watershed, the trap should be placed as low in the watershed as practicable. Species exhibiting a stream-type life history pattern, such as coho salmon and steelhead often migrate within basin and rear away from their natal streams. Therefore, the smolt production measured from part of the basin may represent a variable proportion of the progeny from the adults that spawned upstream of the trap. Furthermore, species with an ocean-type life history pattern, such as pink salmon (*O. gorbuscha*), often spawn lower in the watershed. Estimating production for these species requires trap placement as low in the system as possible (Seiler and Volkhardt, 2005).

At the site scale, water velocity, depth, and proportion of the flow screened are also important considerations for trap placement. Velocity is an especially important consideration if trapping strong swimming species such as steelhead trout, and becomes less important when trapping newly emerged fry. For most species, water velocities of at least 1-mps (3-cfs) are desirable for scoop trap operation and over 2-mps (6-cfs) may be required to capture and retain most steelhead smolts. Similar velocities are recommended for screw trap operation. Screw traps should rotate at least 5-6 rotations per minute for retention of larger smolts. Care must be taken that the water depth under the trap and live well will be sufficient over all flow conditions expected during the outmigration period or damage to the equipment may result during low flow conditions. It is usually best to select a site where a relatively high proportion of the total flow can be screened through the trap in order to achieve the highest trap efficiency. The requirement for adequate velocity, depth, and trap efficiency usually argues for placing the trap in the thalweg of the channel. Consideration must be given, however, to the number of migrants captured. The investigator may opt to operate the trap in a slightly less advantageous position to avoid causing

stress or predation in the live well by capturing and holding too many migrants (Seiler and Volkhardt, 2005).

Screw traps are inherently noisy due to the rotation of the trap cone about its central axis. Migrants will avoid the trap if they are aware of its presence; therefore, it is best to select a site where the trap noise can be masked in order to maintain higher trap efficiency. Fortunately, higher velocity reaches are also noisy reaches. In smaller rivers, these conditions are encountered at the head-end of a pool or chute where water velocities over an elevation drop (e.g., riffles, cascades, or falls) can be directed into the trap. In larger rivers, channel constrictions may afford the best sites (Seiler and Volkhardt, 2005).

In addition to the above-mentioned criteria, consideration must be given to anchoring the trap in the stream. Scoop and screw traps can be anchored by cables to the base of stout trees on each bank, to anchors affixed to bridge abutments, retaining walls, or bedrock, or to a high lead suspended across the river. In the early 1960's, the mainstem Columbia River was trapped using a series of scoop traps cabled to large concrete blocks submerged in the river (Schoeneman et al. 1961 as cited in Seiler and Volkhardt, 2005).

Finally, investigators need to consider access and security when selecting trapping locations. Traps anchored in the river are a public curiosity and can undergo theft or vandalism when not attended. Ideally, the trap site would be located near a launch/recovery site to ease trap installation and removal (Seiler and Volkhardt, 2005).

PERMITTING

Before any thought to installation and active trapping can begin, all necessary permits must be obtained from appropriate local, state, federal and tribal agencies. Sufficient time must be allotted during the planning period to secure permits. The completion of the JARPA, or the Joint Aquatic Resources Permit Application, can be filled out once and used as the standard application for most permits. A list of the necessary permits required for installation of the rotary trap on the Okanogan River, and their issuing agencies, is listed below:

- Section 10 Incidental Take Permit — NOAA Fisheries
- Hydraulic Project Approval — Washington Department of Fish & Wildlife
- Scientific Collection Permit — Washington Department of Fish & Wildlife
- Bridge Attachment Permit — Washington State Department of Transportation
- Shoreline Exemption — City of Okanogan

Each permit carries with it various stipulations for trap deployment that must be rigidly adhered to. Several contain language requiring periodic reporting of operations and data while others need only be kept appraised of the continuation of trapping efforts from year to year.

PREPARATION AND INSTALLATION

Before trapping can begin, all equipment and supplies must be assembled to accomplish project objectives. At a minimum, this includes the trap/pontoon structure and appropriate anchoring cables, a means to get to the trap (e.g., boat or gangplank), dip nets for removing and handling fish, data forms, fish anesthetic, a marking device (e.g. scissors, dye, etc.), tanks or buckets for working up captured fish, a trap cleaning device (e.g. brooms, water pump and nozzle), and lights for night work (Seiler and Volkhardt, 2005).

The approach for trap installation depends on the size and weight of the trap used. Small traps that use lightweight aluminum pontoons can be transported disassembled in pickup beds and assembled on-site. Components of larger, heavier traps can be trucked to the site using a low-boy trailer. In this case, on-site assembly requires the use of a loader or other heavy equipment

to move the components into place. A third option is to truck an assembled trap to the site and position it at the water's edge using a boom truck or crane (Seiler and Volkhardt, 2005).

Once assembled at the water's edge, the trap is ready to be positioned in its fishing location. The approach used to accomplish this will depend on the size of the trap and stream, and the distance from the launch site to the fishing site. Small traps operating on small streams can be pushed and pulled into position by hand. Bow-mounted cables or ropes can be attached to trees or other anchoring structures on the banks. Movement of the trap into its final position can be accomplished by using hand winches or chainfalls. If the trap is anchored to trees, some method should be used to spread the load over the trunk and prevent girdling. Fabric straps make useful attachments (Seiler and Volkhardt, 2005).

Larger traps may use bow winches, mounted port and starboard, to store the attachment cable or rope. The most direct approach is to run the cabling out to the attachment points and pull the trap into position using the winches. Another approach is to attach the cabling directly from the trap to a highline that has been strung over the river. For larger traps (e.g., 8-foot diameter cone rotary screw trap), the trap should be secured in the river with 10 mm (⅜ in.) aircraft cable attached to a 13 mm (½ in.) aircraft cable and pulley system strung above the river between two large trees or bridge pilings on either bank (Murdoch et al. 2001). The position of the trap can be adjusted by the tension of the highline and length of the bow cables that are attached to it using a chainfall or similar device. The use of bow-mounted winches is the preferred approach since it makes repositioning the trap much easier (Seiler and Volkhardt, 2005).

In some cases, the launch point may be some distance from the fishing site. In this situation, the trap can be "walked" into position by alternating port and starboard attachment points either upstream or downstream and tightening or loosening the bow cables as necessary using winches. In navigable waters, a boat can be used to push the trap to a point near the trap site where one of the above methods can be used to secure the trap to its fishing position (Seiler and Volkhardt, 2005).

SAMPLING DURATION

The time frame for operation of the trap varies with the target species and trapping location. Table 1 provides general migration timing for Washington rivers. Downstream migration timing in specific watersheds can vary from these general guidelines. Timing may need to be investigated during the first year of monitoring where it is not well known (Seiler and Volkhardt, 2005).

Table 1. Generalized migration timing for anadromous salmonids in Washington State.

Species	Age	Migration Period
Chinook	0, 1	January – July/August
Coho	1	April – June
Sockeye	0	January – May
Chum	0	February – April
Pink	0	January - May
Steelhead	2	March – May
Cutthroat	0, 1, 2	January – December*
* Migration timing for cutthroat vary widely.		

In order to estimate production, traps should be operated throughout the migration period for the target species. Migration rates for most species are often highest at night; however, daytime migration rates can also be high on some streams, particularly where turbidity levels are high. At

a minimum, the investigator should stratify trapping periods to reflect different migration/capture rates. This often means checking the trap and processing the catch at dawn and at dusk to measure day and night catch rates. This doesn't infer that these are the only times to check the trap. Catch rates and debris loads determine the frequency of trap maintenance. Stratification facilitates sub-sampling and estimating catches during periods when trapping is suspended (Seiler and Volkhardt, 2005).

PROCEDURE

Trap Operation

The screw trap is lowered into its fishing position by cables attached to the forward and/or aft ends of the trap structure. Typically, a single hand-winch or chainfall is used to raise and lower each end. The forward end of the cone should be lowered until the axle is at the water's surface. The aft end is lowered so that fish can swim from the aft screw chamber into the live well, but not so low that they can ride the debris drum over the back of the trap (Seiler and Volkhardt, 2005).

Since the screw is constantly rotating, relatively little debris builds up on the screw's outer screen. As the debris drum removes much of the debris entering the trap, this gear requires less cleaning than a scoop trap. During each trap check, organic debris remaining in the live well is removed and returned to the river; man-made trash is collected and properly disposed of. The trap can usually remain in operation during this procedure. The date and time of the trap check is recorded. If the trap is outfitted with a counter to record rotations, the count is recorded. Rotations per minute are also recorded during the trap check. These later data are used to estimate the time fished if debris stops the screw between trap checks. Catch is enumerated by species and other data/samples are taken as required by the study (Seiler and Volkhardt, 2005).

Traps are checked as often as necessary to provide for the safe holding and handling of captured fish, and maintain the efficient operation of the gear. At a minimum, the trap should be checked at dawn and at dusk in order to evaluate day vs. night capture rates. When operated during period of high discharge, the trap will be checked and cleaned more frequently. Where sub-yearlings are captured, holding these in close proximity to larger piscivorous fish such as Northern Pikeminnow and sculpins increases the likelihood that catch counts on the sub-yearlings will be biased low due to live-box predation (Seiler and Volkhardt, 2005).

Some investigators have placed tree branches or other debris in the live well to provide refuge for small fish. Care must be taken when using this approach since the debris may cause de-scaling as turbulence in the live well increases. The safest approach for maintaining fish health and minimizing predation is to frequently check and remove fish from the trap (Seiler and Volkhardt, 2005).

Daily Capturing Procedure

Fish will be removed from the livebox with dipnets every morning and placed in an appropriate holding container. Fish will be identified to species, counted, scanned for a PIT tag and released off the back of the trap. Fish that are to be measured and weighed will be placed into a bath containing an anesthetic solution of MS-222 at a concentration of 50-60 mg/L (Please refer to Appendix 2 for more detailed information on MS-222). All fish placed into an anesthetic bath will be allowed to become mildly sedated before being measured and weighed, and completely recover before being transported in 5 gallon buckets to a release site. Fish should be fully recovered from the anesthetic prior to release (Seiler and Volkhardt, 2005). Note that all fish species react differently in their exposure to MS-222. Regardless, steelhead will be worked up first and released first to reduce the amount of time they spend out of the river (NOAA Section 10 permit). All anesthetized fish will be allowed to fully recover in fresh water prior to being released in an area of calm water downstream from the smolt trap. Juvenile target salmonid species will be held in separate live boxes attached to the end of the main pontoons for use during mark/recapture efficiency trials conducted in the evening.

Length and Weight
Fish that are to be used in the trap efficiency trial will not be anesthetized and thus will not be measured or weighed.

A random sub-sample of 10 fish per species per day will be weighed, measured and recorded if time permits; anesthetize these fish before working them up. Make sure to allow anesthetized fish adequate time to fully recovery before returning them to the river downstream of the trap. Enter data onto the data sheet and then to the spreadsheet.

Every steelhead handled out-of-water for the purpose of recording biological information must be anesthetized. Anesthetized fish must be allowed to fully recover in a recovery tank before being released. Steelhead that are simply counted must remain in water but do not need to be anesthetized.

Biometric measurements will be taken from fish that will not be marked so as to not expose marked fish to excessive handling. Fork length and weight to 0.1g will be recorded for the first 10 randomly selected fish of each species on each trapping day. Fish that are notably larger or notably smaller should also be measured and weighed with a notation of not being a random sample.
Length and weight measurements will be recorded for all target species, except on days when high numbers are captured, and then only target species used in mark/recapture efficiency trials are measured and weighed. Fork length to the nearest millimeter and weight to the nearest 0.1 g will be measured. A Fulton type condition factor ($W \times 10^5/FL^3$) will be calculated for all target species sampled. The degree of smoltification (parr, transitional, or smolt) will be determined by visual examination. Juvenile Chinook, sockeye, and steelhead *O. mykiss* will be classified as parr if parr marks are distinct, transitional if parr marks are not distinct, and smolts if parr marks are not visible and the fish exhibited a silvery appearance.

Condition
The Fulton-type condition factor describes the well-being of smolts within a population or subpopulation. Smolts collected with traps will be measured (fork length; mm) and weighed (to 0.1 g). Fulton-type condition will be estimated with methods described in Anderson and Neumann (1996).

Genetics
Genetic characterization (via DNA microsatellites) describes within- and between-population genetic variability of smolts. DNA samples from a systematic sample of smolts[1] will be collected and analyzed according to the WDFW protocols contained in Appendix 1.

Trap Efficiency Tests
Trap efficiency is measured by the rate that marked fish released above the trap are recaptured. Mark/recapture efficiency trials will be conducted throughout the trapping season when a minimum of 30 individual fish of a given target species are captured within a three day period. If less than 30 fish are captured within a three day period, all fish will be released unmarked. Bismark Brown 'Y' dye will be used at a concentration of 0.25 to 0.4 g of the powdered dye will be added to 5-gallons of water for marking the mucous layer of fish used in trap efficiency trials. Other marking methods, including applications of a fin clip, caudal punch, freeze brand or PIT tag, require handling and the application of MS-222 and will thus not be used.

The release point selected should be far enough upstream as to provide for a similar distribution across the channel compared to unmarked fish (at least 2 pool/riffles sequences), but not so far upstream that predation on marked fish is substantial. Murdoch et al. recommends that the

[1] The total number of smolts needed to characterize within and between-population genetic variability is presently unknown. Therefore, "**k**" (i.e., the k^{th} smolt sampled) remains undefined.

release point be located at least 1 km upstream of the trap. Try to release each group of marked fish evenly across the river to avoid biasing their lateral distribution and along approximately 100 m of the bank in pools or in calm pockets of water where possible. To reduce predation subsequent to recapture, marked fish should be released during the time strata that they migrate (Seiler and Volkhardt, 2005).

Mark groups can be comprised of hatchery fish or fish that have been previously captured in the trap. However, using hatchery fish complicates the study since one must assume their probability of capture is the same as for naturally reared fish. Groups of marked fish representing each targeted species are released upstream of the trap over the period of their migration.

While hatchery fish used for calibration may be of the same species and age as their wild counterparts, they may be larger, behave differently, and consequently, may be captured at higher or lower rates than wild fish. Rates of instream predation and residualism are likely higher for hatchery fish. For these reasons, trap efficiency estimates resulting from release groups using hatchery fish may be biased low (Seiler and Volkhardt, 2005).

Flow is the dominant factor affecting downstream migrant trapping operations in any system. It affects trapping efficiency and migration rates since high flows often stimulate fish to migrate. Therefore, minimal trap efficiencies may occur at the same time that peak flow events are causing migration rates to increase (Seiler and Volkhardt, 2005).

Visibility, fish size, and noise are other factors that affect trap efficiency. Larger downstream migrants, especially steelhead and coho, may be able to avoid capture when the trap is visible by swimming around the trap or back out of the mouth of the trap, especially where velocities are low. Some portion of ocean-type Chinook salmon may rear upstream for a short period of time and grow prior to migration; therefore, efficiency for a species may change over time. Fish behavior may also be important. Some species may primarily migrate down the thalweg of the channel whereas a higher proportion of others may use the channel margins. Noise created by the trap causes an avoidance response. This is mitigated through proper site selection as discussed above (Seiler and Volkhardt, 2005).

These factors indicate that efficiency tests should, if possible, be conducted over the entire migration period, over a range of flows and turbidity levels, and for each species whose production is to be estimated (Seiler and Volkhardt, 2005).

Emigration estimates can be calculated using estimated daily trap efficiency derived from the regression formula using trap efficiency (dependent variable) and discharge (independent variable) as described in Murdock et al. 2001.

A valid estimate requires the following assumptions to be true concerning the trap efficiency trials:

1) All marked fish passed the trap or were recaptured during time period *i*.

2) The probability of capturing a marked or unmarked fish is equal.

3) All marked fish recaptured were identified.

4) Marks were not lost between the time of release and recapture.

Incidental Species

When time permits, incidental species should be measured and weighed as described for target species. All incidental species will be released downstream of the trap.

DATA ANALYSIS

ESTIMATING TOTAL MIGRATION

Estimating migration for any period, whether a short time interval or an entire season, requires catching fish and estimating trap efficiency. Estimating abundance from a set of trapping data is not always straightforward. A variety of approaches have been used. In many cases the most appropriate approach will not become apparent until after all of the field work is completed and the data is analyzed. The biologist needs to always temper his/her decision on the approach with knowledge of the behavior of the targeted species. A plausible rationale should be developed to explain and support these decisions. Four general approaches are outlined in this section (Seiler and Volkhardt, 2005).

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1. Estimating discreet outmigration periods using individual trap efficiency estimates. This approach estimates migration for discreet time periods, typically a day or a week, using a single test to estimate trap efficiency or by pooling several efficiency trials to develop a mark-recapture based estimate of the migration for the time period.

Migration over the discreet period, N_i, is found using the simple equation;

$$\hat{N}_i = \frac{M_i C_i}{R_i} \quad (1)$$

Bias in this estimate can be reduced using the Peterson mark-recapture equation;

$$\hat{N}_i = \left[\frac{(M_i + 1)(C_i + 1)}{(R_i + 1)} \right] - 1 \quad (2)$$

Where

M_i	=	Number of fish marked and released during discreet period *i,*
C_i	=	Number of unmarked fish captured during discreet period *i*, and
R_i	=	Number of marked fish recaptured during discreet period *i*.

The variance, $V(N_i)$, of the Peterson estimate can be calculated using;

$$V(\hat{N}_i) = \hat{N}_i^2 \frac{(C_i - R_i)}{[(C_i + 1)(R_i + 2)]} \quad (3)$$

Total juvenile production is estimated by the sum of the estimated migrations over discreet periods and the variance of the total production is the sum of the variances. The 95% confidence interval (CI) is $\pm$ 1.96(sd).

This approach assumes each estimate of trap efficiency is an accurate measure of the proportion of downstream migrants caught in the trap. Since each test actually represents a single measure, it would be expected to include error. Assuming error is normally distributed, this approach argues for estimating discreet periods of short duration (e.g., 1 day) since cumulative error from many samples should approach zero. We cannot assume error is normally distributed where trap efficiencies are low, however. Estimates of efficiency that are lower than the true efficiency cannot offset those that are higher as the true value approaches zero (Seiler and Volkhardt, 2005).

A variation of this approach is to use another trap upstream to capture and mark migrants over the trapping season. The recapture of these migrants in the downstream trap over the season represents a single mark-recapture experiment. Since both marked and unmarked fish should have an equal chance of being captured over time, the timing distribution of marked releases should reflect the migration timing for the species. Therefore, a weir trap located in a tributary is the best choice for this second trap since it is designed to catch 100% of the passing migrants over the entire season. Total production is estimated using Equation 1, substituting the total migration (N), total catch of marked and unmarked fish (C), total marked releases (M), and total recaptures (R), for N_i, C_i, M_i, and R_i in the equations. Variance, V(N), is estimated by the variance of the trap efficiency estimate, R/M, which is a binomial multiplied by the C^2 over $(R/M)^4$. This reduces to:

$$V(N) = \frac{\frac{R}{M}\left(1-\frac{R}{M}\right)}{M} * \frac{C^2}{\left(\frac{R}{M}\right)^4} = \frac{C^2 M(M-R)}{R^3} \qquad (4)$$

2. Modeling Trap Efficiency. This approach estimates trap efficiency from an independent variable, typically stream flow. A series of trap efficiency tests are conducted over a range of flows and analyzed to determine if a significant relationship can be established (Figure 8). When using regression analysis, it has been suggested that the observed F should exceed the chosen test percentage point by a factor of four or more for the relationship to be considered of value for predictive purposes (Draper and Smith 1998).

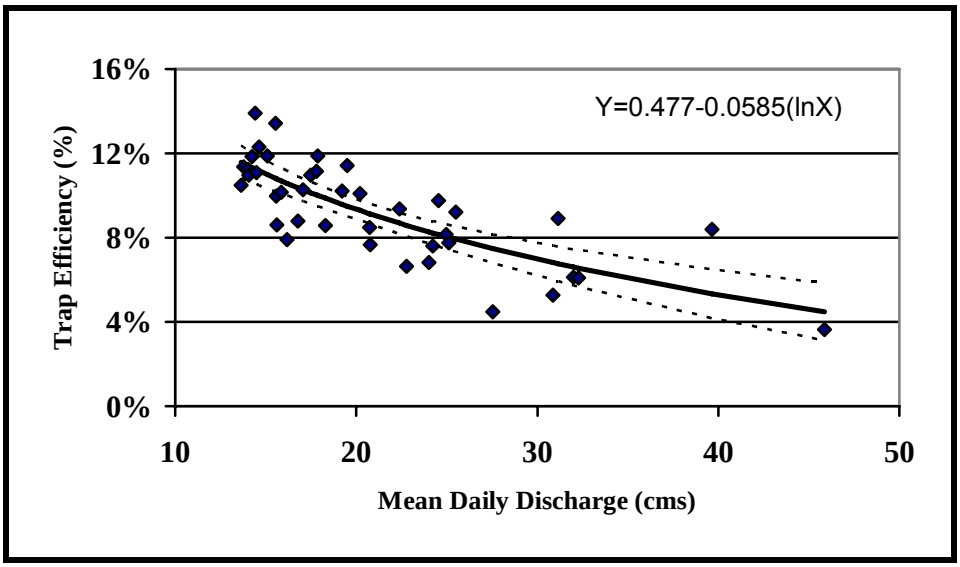

Figure 2. Age 0 sockeye trap efficiency and 95% confidence intervals as a function of stream discharge, Cedar River, Washington USA (Source: Seiler and Volkhardt, 2005).

Using this approach, migration on day *i*, N_i, and its variance, $V(N_i)$, are estimated by;

$$\hat{N}_i = \frac{C_i}{\hat{e}_i} \qquad (5)$$

$$V(\hat{N}_i) = V(\hat{e}_i)\frac{C_i^2}{\hat{e}_i^4} \qquad (6)$$

If linear regression is used to estimate trap efficiency, its variance is estimated by;

$$V(\hat{e}_i) = MSE\left(1 + \frac{1}{n} + \frac{(X_i - \overline{X})^2}{\sum_{i=1}^{n}(X_i - \overline{X})^2}\right) \quad (7)$$

where:

$\hat{e}_i$ = The trap efficiency predicted on day i by the regression equation, $f(X_i)$,

MSE = The mean square error of the regression,

n = The number of trap efficiency tests used in the regression, and

X_i = The independent variable on day i.

3. Stratifying Trap Efficiency. Like #2, this approach also predicts trap efficiency using an independent variable. In this case, efficiencies are fairly constant over some range of the independent variable or a condition class. Then as the independent variable passes some threshold or another condition class occurs, efficiencies change or "step" to a new level. For example, if the trap is placed in a "U"-shaped channel adjacent to a wide gravel bar, trap efficiencies may be at one level when flows are contained in the channel and another when higher discharge causes a substantial portion of the flow to spread out across the gravel bar. Fish size may change over the trapping season causing changes in trap efficiency by time strata. Turbidity levels may cause changes in efficiencies as well. In some locations, fish are better able to avoid traps during day fishing periods. In this case, efficiency data would be stratified by condition class (i.e., day and night periods) (Figure 3). Mean trap efficiency is calculated for each strata (Seiler and Volkhardt, 2005).

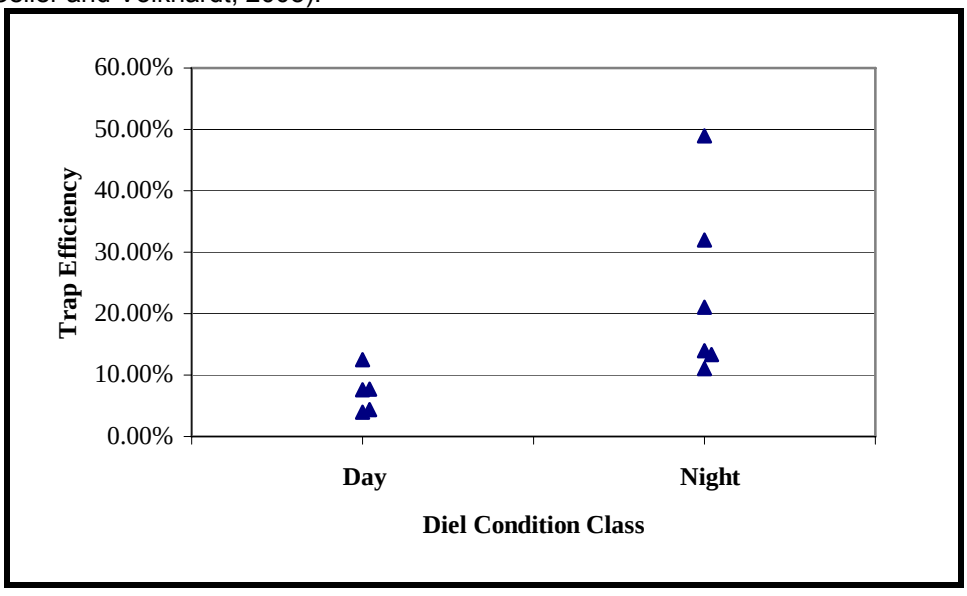

Figure 3. Range and mean trap efficiencies stratified by diel fishing periods, Issaquah Creek, Washington USA (Source: Seiler and Volkhardt, 2005).

Migration is estimated for discreet periods when the independent variable is within a defined stratum by dividing the sum of the catch by the mean trap efficiency for the stratum. The variance of the estimate is calculated using Equation 6, substituting the mean trap efficiency for the stratum, $\bar{e}_j$, for the predicted trap efficiency on day i. (Seiler and Volkhardt, 2005).

4. Back-Calculating Production. Using this approach, fish captured in the screw trap are marked or tagged and released downstream. Recapture occurs at another location and/or life stage and a Peterson estimate of production is made. Typically, recaptures occur when the returning adults are sampled in a fishery, upon the spawning grounds, or at another sampling location such as a trap. The term "back-calculating production" generally refers to calculating downstream migrant production from the recapture of adults marked as downstream migrants captured in the trap. However, production estimates could also be achieved using this method by sampling marked juveniles from the lower river or estuary (Seiler and Volkhardt, 2005).

Production is estimated using the same equation described for the variation of approach #1 above. The variance is estimated by Equation #4. This approach is most useful where trap efficiency estimates are difficult to make. If mark or tag sampling occurs while the juvenile fish are still on their seaward migration, then this approach could be used for all species. If sampling will not occur until the adults return, then this method is more easily applied where nearly the entire cohort returns in a single year (e.g. coho). Age sampling would be required for this approach to work for species that return to spawn in multiple year classes (Seiler and Volkhardt, 2005).

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APPENDIX 1

Guidelines for Non-Lethal Fry and Smolt DNA Sampling

The goal is to take a small enough piece of a non-critical tissue (e.g., fin) to have little or no impact on the subsequent survival of the fish but that is adequate to allow genetic analysis. DNA analysis is ideal for this for two reasons: 1) all living cells of an organism have essentially the same DNA composition (unlike the tissue specific expression characteristic of allozymes and other proteins), so that tissues such as fin and opercula can provide adequate samples, and 2) amplification of the resulting DNA from such samples via the PCR (polymerase chain reaction) provides the sensitivity of detection to enable working with very small pieces of tissue and small amounts of DNA. *[For mammals, this approach has been used successfully to characterize animals by analyzing DNA extracted from hair follicles, blot spatters, and scat samples.]*

The **minimum amount of tissue** that is needed is approximately the size of this circle: 
(a piece of tissue with the same surface area as a 1.5mm diameter disc). **Failure to take a large enough tissue sample can prevent successful DNA analysis.** The recommended sources of such a tissue sample are any of the following:

1) A distal portion of the dorsal lobe of the caudal fin
2) A distal portion of one of the pelvic fins
3) Smaller distal portions of both pelvic fins
4) One entire pelvic fin

By sampling only the distal portion of a fin, we expect that the fish will successfully regenerate the entire fin over time. In contrast, removing an entire fin often results in little or no fin regeneration, presumably leaving the fish at a selective disadvantage.

When sampling larger fish, a larger sample is preferred (e.g., a piece of tissue approximately the size of one of these circles:  [approx. 3mm diameter] or [approx. 4.5mm diameter]), because this will provide more material (DNA). The "extra" tissue provides a reserve that can be used to overcome some types of analytical problems in the lab by repeated analysis and/or it provides material that can be used for subsequent analyses (for example to examine additional loci at a future date) or can be shared with other laboratories/agencies.

Live fish should be handled appropriately before, during, and after sampling. This will probably involve: a) anesthetization prior to handling for tissue sampling (and taking of measurements or other biological samples such as scales), b) careful handling during sampling to avoid injury and scale/mucous loss, and c) holding fish in a recovery vessel after sampling (until the anesthetic has worn off) before releasing them in a way that minimizes immediate mortality due to predation of other effects.

Each tissue sample should be placed in a vial that contains DNA preservative solution (and an appropriate label -- preprinted by WDFW [preferred] or written in *pencil*) immediately after it is taken. We recommend using vials that are approximately 3/4 full of preservative solution and never adding more than 1/5 of this volume of tissue (to ensure adequate preservation). Please rinse forceps, scissors, etc. (with fresh water) and dry them between fish to minimize the chance of cross- contamination of samples. Such preserved samples should be stored at ambient temperatures (20-80^{o}F) until they are returned to the WDFW Genetics Laboratory in Olympia.

If you have questions or need additional information, please telephone Todd Kassler (360-902-2722), Sewall Young (360-902-2773), or the Genetics Lab at 360-902-2775).

APPENDIX 2

Guidelines for Finquel® MS-222 Anesthetic

MS-222 is very soluble (1:9) in water and it dissolves with equal readiness in spring water, tap water, or seawater. *Do not use distilled or deionized water, or water containing chlorine, copper, zinc, etc.), or other toxic contaminants.* The anesthetic solution will use fresh river water be well oxygenated, and its temperature should be similar to that of the water from which fish are taken. Make sure the aeration pumps are on for maximum oxygenation (Argent Chemical Laboratories).

Prior to use, MS-222 can be weighed out into convenient amounts:

- 1 gram of MS-222 (approximately ½ teaspoon) in 5 gallons of water yields a concentration of about 50 to 60mg/L.
- Published reference for pH buffering of MS-222 is 2 ppm of sodium bicarbonate for each 1 ppm MS-222.

To convert mg/liter into g/gal: multiply number of mg. by 0.00378 (e.g. 80 mg/L = 80 x 0.00378 = .0302 g/gal)
To convert mg/liter into a ratio of MS-222 to water: divide 1,000,000 by the number of mg (e.g. 80 mg/L = 1,000,000 / 80 = 1:12,500)

Excessive exposures are avoided by observing the following sensory and motor responses of the fish which characterize progressively deeper levels of anesthesia.

- Sedation - Decreased reactivity to visual and vibrational stimuli; gill activity reduced.
- Total loss of equilibrium - Fish turns over; locomotion increases; fish swims or extends fins in response to pressure on caudal fin or peduncle.
- Total loss of reflex - No response to pressure on caudal fin or peduncle; opercular rate slow and erratic.
- Medullary collapse - Gill activity ceases.

Laboratory and field investigations have shown that the action of MS-222 is readily reversed when the fish are transferred to fresh water before opercular activity ceases. *Additional exposure following medullary collapse may result in mortality.* A rough estimate of the safe total exposure can be made by multiplying the time required for anesthesia by a factor of 2 or 3.

Method of Application
General anesthesia: - Fish will be immersed in a bath of MS-222 anesthetic solution. Containers may be of glass, plastic, steel, aluminum, or other suitable material. However, do not use galvanized or brass containers unless treated or sealed to prevent dissolution of zinc. The fish should not be overcrowded. Discard anesthetic solutions when a loss in potency is noted, or when the solution becomes fouled with mucus or excrement.

Precautions for Using MS-222

1. Avoid inhaling or getting it into the eyes.
2. Always conduct preliminary tests to determine desired rates of anesthesia and optimal length of exposure.
3. Do not overexpose fish to lethal levels.
4. Do not anesthetize more fish than can be effectively handled.
5. Do note use water containing chlorine, or other toxic agents.
6. Insure adequate oxygen in anesthetic solution.
7. Discard anesthetic solutions when fouled with mucus or metabolic wastes.
8. Do not discard MS-222 solutions into water supplies of natural waters.
9. Store solutions and dry powder in a cool place away from light.*
10. Discard stock solutions of when they lose effectiveness.*

*The color of MS-222 solutions may change rapidly to yellow or brown when exposed to light. This does not affect activity in any significant way. However, for best results use freshly prepared solutions. A 10% solution stored at room temperature shows no significant loss of potency after three days, but after 10 days, a brownish color and an activity decrease of 5% is observed.

APPENDIX 3

Guidelines for Marking Fish with Bismark Brown 'Y' for Estimating Trap Efficiency

1. Fill a five gallon bucket with 16 liters of water (5 gallons = 18.93 L).
2. Measure out 0.25 to 0.4 grams of Bismark Brown 'Y'.
3. Mix Bismark Brown 'Y' into the 5 gallon bucket.
4. Place aerator in bucket. Keep water well oxygenated.
5. Count out the number of fish to be dyed with Bismark Brown 'Y' and place them in the bucket containing the dye bath. Record number of fish, time and temperature on data sheet. **Do not anesthetize the fish prior to immersion in dye solution**.
6. Place thermometer in bucket.
7. Set lid on bucket to prevent fish from escaping.
8. Check time or set timer for 1 hour.
9. Observe water temperature and fish every 5 to 10 minutes. Fish will probably appear sluggish.
10. If water temperature rises, add frozen water bottles as needed to keep water temperature constant.
11. Frequently and gently, stir water in bucket while observing fish. **If fish are reacting abnormally, remove them immediately and place them in a well-aerated bucket of fresh water.**
12. Remove fish from dye bath after one hour. Adjust time duration in, and amount of, powdered dye used in dye bath based upon color of stained fish. **Do not exceed 1½ hours**.
13. Allow fish to fully recover in fresh water. Observe them to make sure they are okay before releasing them.
14. Release fish at Salmon Creek Boat Launch (approx. ⅔ mile upstream from trap) at dusk or at night to avoid predation.

Adapted from guidelines used by Idaho Fish & Game

APPENDIX 4

Daily Operational Guidelines for Trap Tenders

Before any work is performed on site, make sure that:

1. At least two people are on site
2. All rigging equipment is sound
3. When on the trap, everyone wears a PFD
4. All safety equipment is in good working order
5. All needed sampling equipment is available and fully functional
   - PFD
   - Throw bags
   - Waders
   - Tools & hardware (wrenches, sockets, shackles, clips, etc.)
   - Fish measuring board and balance
   - Data sheets, notepad and pencil(s)
   - Brush for cone cleaning. Use trash pump when needed
   - Nets
   - Buckets
   - MS-222
   - Bismark Brown 'Y' Dye
   - Flashlight and fresh batteries
   - *Appropriate Clothing*!!
6. Conditions are within safe operational parameters
7. Date, time, trap rotation and temperature are recorded.

When fishing:

1. Check the screw trap a minimum of once daily. During periods of high flow, check trap repeatedly.
   a. things to look for:
      - debris in the cone and trap box
      - worn bushings and seals
      - missing rivets or screws
      - worn or broken parts
      - damage to straps, cables, blocks and other trap rigging
   b. make sure trash screens are clean
   c. look for means by which fish can escape the trap box
   d. make sure pontoons and cone are not rubbing on rocks
   e. make sure live boxes are secure
   f. check for the presence of predator fish in the trap box
   g. collect and dispose of man-made trash

2. Work up all fish in the AM
   a. fill out the trap notes in Rite-In-Rain notebook
      1. participants
      2. trap RPM
      3. discharge and staff gage reading from USGS website (Tonasket) http://waterdata.usgs.gov/wa/nwis/uv?dd_cd=05&dd_cd=19&dd_cd=20&format=html&period=2&site_no=12445000
      4. water temperature (°C)
      5. non-target fish
      6. trap operation (general notes)
   b. make sure to separate the large fish from the small fish
   c. work up steelhead first. Steelhead MAY NOT be handled if water temperature exceeds 21°C (69.8°F)
   d. avoid overloading five-gallon buckets with fish

3. Hold fish until fully recovered. Record water temperature (°C) and time (24 hour) at release.

4. Make sure equipment is kept neat and orderly.
    a. equipment is dried off and put away
    b. do not leave any equipment where the public can get to it.
    c. chemical bottles are to be kept full (use refill containers)

5. DON'T FORGET TO NOTE TIME AND TEMPERATURE WHEN RELEASING FISH.

6. Complete the LOG SHEET daily.