FISHERY RESEARCH

PROTOCOLS FOR TRAPPING ANADROMOUS EMIGRANTS IN IDAHO

Photo: Bruce Barnett

Prepared by:
Timothy Copeland, Fisheries Program Coordinator
Bruce Barnett, Fisheries Data Coordinator
William C. Schrader, Principal Fisheries Research Biologist
Kimberly A. Apperson, Fisheries Biologist
Laurie Janssen, Fisheries Technician 2
Ronald V. Roberts, Fisheries Technician 2

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By
Timothy Copeland
Bruce Barnett
William C. Schrader
Kimberly A. Apperson
Laurie Janssen
Ronald V. Roberts

Idaho Department of Fish and Game
600 South Walnut Street
P.O. Box 25
Boise, ID 83707

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SUMMARY

Trapping juveniles emigrating from spawning and rearing habitats has long been a part of anadromous salmonid management. The goal of this report is to recount the history of trapping juvenile anadromous salmonids by Idaho Department of Fish and Game (IDFG), to describe the current status of the protocols, and to discuss how the protocols should move forward. IDFG has historically devoted a large amount of effort and resources to trapping juvenile salmon and steelhead emigrants. Trapping started in the 1950s and changed character as management needs and technologies evolved. In general, there have been four major goals to these historic trapping activities: 1) collect fish to move them; 2) mark or tag fish for downstream studies; 3) describe characteristics of migrating fish; and 4) estimate population parameters such as abundance, survival, and productivity. This history has led to how IDFG traps juvenile salmon and steelhead today. Rotary screw traps (RSTs) have been the primary tool used by IDFG since the early 1990s: 1) to sample populations or spawning aggregates of interest to estimate juvenile emigrant abundance, and 2) to tag emigrants with passive integrated transponder (PIT) tags to estimate smolt survival to and through the hydrosystem. Over the last ten years, IDFG reviewed operation of its RST fleet and made significant changes to expand collection of wild steelhead and to improve efficiency. In this protocol document, we review installation and safe operation of RSTs, daily processing of the catch, data management and analysis, and reporting. A properly run RST generates a powerful data series that describes the dynamics of the target population. The worth of these excellent quantitative data sets increases as they build over time, especially as they have documented a great range in year class strengths. This information supports the evaluation of several types of management programs, such as the effects of hatchery supplementation and habitat restoration. Full development of data sets has greatly increased understanding of how life history variations can influence population dynamics of anadromous salmonids. To improve operation of RSTs and the usefulness of the resulting data, we have three programmatic recommendations: 1) incorporate testing of mark-recapture assumptions as a regular part of trap operations and data analysis; 2) periodically revisit tagging strategy for each trap and across the fleet of RSTs; and 3) develop new opportunities for data analysis.

Authors:

Timothy Copeland, Fisheries Program Coordinator

Bruce Barnett, Fisheries Data Coordinator

William C. Schrader, Principal Fisheries Research Biologist

Kimberly A. Apperson, Fisheries Biologist

Laurie Janssen, Fisheries Technician 2

Ronald V. Roberts, Fisheries Technician 2

INTRODUCTION

Trapping juvenile salmon and steelhead (*Oncorhynchus* spp.) emigrating from spawning and rearing habitats has long been a part of anadromous fisheries management. Data collected are often used to evaluate river management (e.g., dam operations); assess effects of habitat restoration; estimate life-stage survival; evaluate hatchery effectiveness; or monitor status of naturally-spawning populations (Volkhardt et al. 2007). Metrics of interest typically include abundance; size, age, and growth of migrants; life-stage survival; and relation of emigrants to parental escapement (e.g., Burgner 1962). However, there are a multitude of issues to consider when trapping downstream migrating fish (Mason 1966). Hence, a successful trapping operation requires training, learning from experience, and a clear protocol (Volkhardt et al. 2007).

The goals of this report are to recount the history of trapping juvenile anadromous salmonids by the Idaho Department of Fish and Game (IDFG), to describe the current status of the protocols, and to discuss how the protocols should move forward. We describe in detail the methods and protocols applied by IDFG to install and operate rotary screw traps (RSTs); to mark fish for emigrant estimation; to manage data collected; and to estimate abundances of emigrants leaving natal streams and smolts leaving Idaho. Trap operation methodology has not always been standardized by IDFG among locations or over time, and this is the first protocol to do so. This report focuses on RSTs, but many of the features of the protocols are transferable to other types of migrant traps.

Traps for emigrating anadromous salmonids in Idaho are deployed to intensively monitor selected populations to support life cycle modeling, leading to a mechanistic understanding of population dynamics (IDFG 2019), as well as to support Intensively Monitored Watershed projects (e.g., Uthe et al. 2017), integrated broodstock programs for Chinook Salmon hatcheries (e.g., Venditti et al. 2018), and to monitor key long-term sentinel populations (e.g., Byrne and Copeland 2007). All current locations are supported by adult steelhead and spring/summer-run Chinook Salmon escapement monitoring to obtain information through the full life cycle. This information is valuable to understand the current status of wild salmon and steelhead populations in Idaho relative to the Endangered Species Act (ESA) and how it is changing. The IDFG program is spatially complementary to the Nez Perce and Shoshone-Bannock tribal fisheries programs. General locations were selected for anadromous emigrant monitoring to accomplish one of two objectives: 1) to evaluate specific management activities within a portion of a watershed, or 2) to conduct population status monitoring to provide information relative to recovery of anadromous salmonid populations in the Columbia River basin (Crawford and Rumsey 2011).

HISTORY OF EMIGRANT TRAPPING BY IDAHO DEPARTMENT OF FISH AND GAME

The Idaho Department of Fish and Game has devoted a large amount of effort and resources over the years to trapping juvenile salmon and steelhead emigrants. Trapping started in the 1950s and changed character as management needs and technologies evolved. In the recent past, most trapping effort has used RSTs (e.g., Venditti et al. 2018). In general, there have been four major goals to these historic trapping activities: 1) collect fish to move them; 2) mark or tag fish for downstream studies; 3) describe characteristics of migrating fish; and 4) estimate population productivity. This history has led to how IDFG traps juvenile salmon and steelhead today. In this section we briefly recount this history.

The importance of assessing downstream emigrants to fisheries management was recognized early in the development of the anadromous management program (Hauck 1952).

However, the techniques to do so were yet to be developed. Several efforts were undertaken, for the need was urgent for information to assess and mitigate the effects of construction of dams within and outside of Idaho (Hauck 1952).

A major push to develop effective means to trap fish in Idaho and across the Pacific Northwest in the 1950s is evident in the literature. Traps for downstream migrating fish fell into two general types: those filtering the entire stream flow and those sampling part of the flow (Mason 1966). Both were tried in Idaho. In an example of the first type, Craddock (1958), working in collaboration with IDFG, constructed a two-way, weir-based trap on Redfish Lake Creek. This was a stable location where the trap was run from 1953 to 1966 (Bjornn et al. 1968) and persisted in much the same form until recently. There was a three-year project based in the Clearwater River drainage with the objective of developing techniques for measuring the size and distribution of downstream migrant populations (Murphy 1955, 1956; Corning 1957; Keating 1958). Work was conducted in a variety of locations from the headwaters of the Lochsa River to Lewiston Dam. During the study, emphasis shifted from weirs and trap boxes located on the streambed to floating inclined-plane traps (Figure 1). Handling fluctuating flows and size selection of catch by traps were major obstacles. Trap designs developed in Washington State (Kray-Meekin inclined-plane traps) were used in the last year of the study with mixed success (Corning 1957). Ultimately, the project failed to develop techniques suitable for abundance estimates except in the summer and fall when flows were low (Keating 1958). A subsequent effort with an experimental design (a cable-suspended inclined-plane trap) aimed at sampling the spring steelhead smolt migration also failed (Reingold 1964). A common finding of these studies was existence of a fall migration associated with rain events out of the tributaries into the main river.

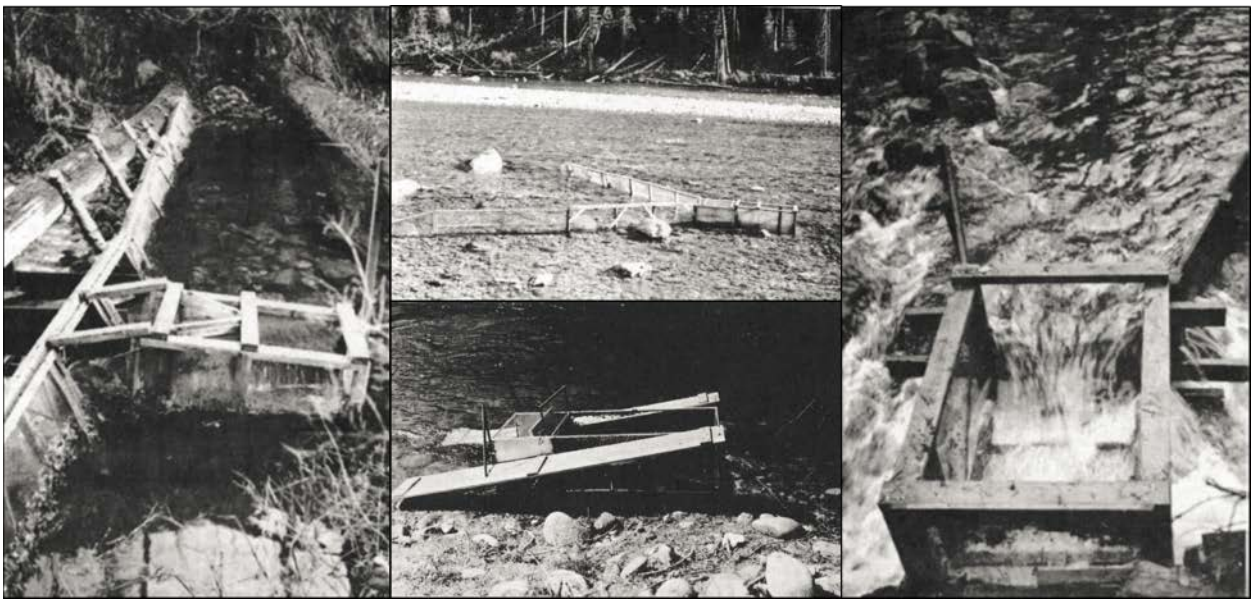


Figure 1. Examples of early emigrant trap designs used in Idaho streams (Keating, 1958). Left: weir trap on Johnny Creek. Top middle; V-trap on North Fork Clearwater River. Right: overflow-spill trap on Orogrande Creek. Bottom middle: floating inclined-plane trap on Middle Fork Clearwater River.

In the face of these methodological difficulties, collection of juveniles in the middle reaches of the Snake River proceeded for assessment of dam-related impacts. These collections were a

cooperative effort by Oregon, Idaho, the US Fish and Wildlife Service, and Idaho Power and used equipment designed to work in the main-stem Columbia River. Over time development of gear used in large rivers went from suspended fyke nets (Mains and Smith 1956) and batteries of inclined-plane traps (Schoeneman et al. 1961) to large barges with dippers, scoops, and travelling screens (Mason 1966; Krcma and Raleigh 1970). Bell (1957) assessed migration timing in Hells Canyon with Kray-Meekin inclined-plane traps during 1955 and 1956 but didn't capture any steelhead, noting many of the same difficulties found by Keating (1958). Emphasis shifted to large barge traps (Figure 2). This work found a large spring migration (April and May) by Chinook Salmon and steelhead smolts in the main-stem Snake River, but there were also movements out of tributaries during the fall and winter (Bell 1959). Findings were meant to guide operations of downstream migrant trap-and-transport operations at Brownlee Dam in order to perpetuate the anadromous runs upstream of the Hells Canyon dam complex. From 1958 to 1960, over 25 thousand downstream migrants were trapped at a weir in the Wildhorse River (between Oxbow and Brownlee dams) and re-located to the Weiser River drainage (upstream of Brownlee Dam; Bell 1961). A series of studies were undertaken by IDFG to evaluate the success of the Brownlee Dam downstream migrant collection facility (Bell 1960, 1961; Graban 1964). In the end, the facility was judged a failure as a method of collecting sufficient numbers of downstream migrating salmon and steelhead (Graban 1964), the anadromous runs upstream of Hells Canyon Dam were extirpated, and management turned to establishing hatcheries in the Salmon River drainage to compensate (Reingold 1966, 1967a).

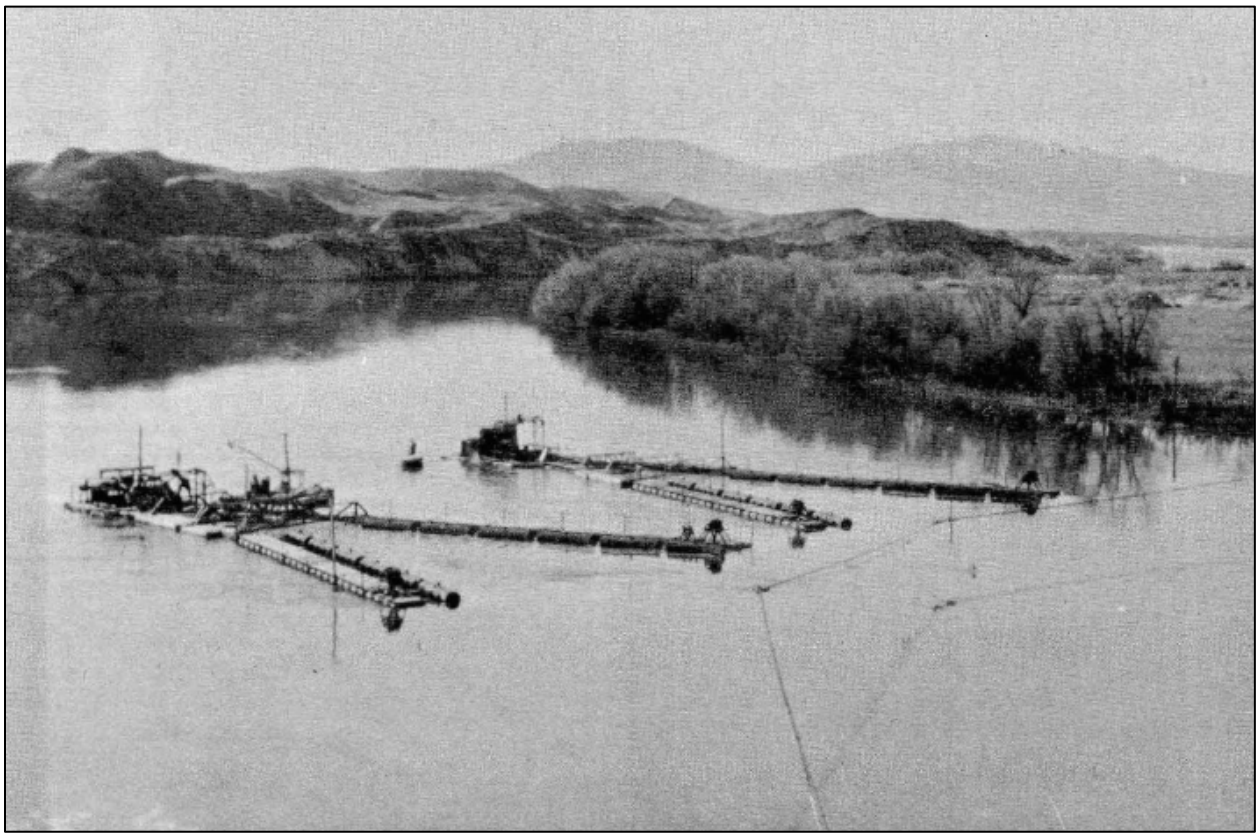


Figure 2. Barge traps (dippers) in the Snake River upstream of Brownlee Reservoir. From Krcma and Raleigh (1970).

Emigrant trapping was also used to evaluate another management program begun during this time, the screening of irrigation diversions in the upper Salmon River drainage. Trap boxes on diversion bypass pipes were used (Corley 1962). As part of this study, Kray-Meekin inclined-plane traps were operated in the Lemhi River at the mouth to describe fish movement patterns (Figure 3). Marks were applied to fish (tattoos or freeze brands) that were then released to estimate percentage of emigrants bypassed by each screen. This program at first focused on the Lemhi River but expanded to encompass much of the upper Salmon River drainage, including main-stem diversions (Schill 1983).

Figure 3. Kray-Meekin inclined-plane trap operating in the Lemhi River (Holubetz 1968). Note the trap is resting on the stream bed rather than floating on pontoons.

Observations of smolt mortality at Columbia River dams (e.g., Schoeneman et al. 1961) prompted marking of Idaho salmon and steelhead to evaluate effects of dams being constructed on the lower Snake River in the 1960s and 1970s. Marking was conducted in order to estimate survival and timing from the tributaries to the uppermost dam and between dams (Ebel and Raymond 1976; Raymond 1979). Fish were marked at hatcheries and in the wild. IDFG was contracted by the Bureau of Commercial Fisheries (today called NOAA Fisheries) to capture and mark migrating juveniles in the Salmon River and selected tributaries (Bjornn et al. 1965, Bjornn and Holubetz 1966; Reingold 1967b). The traps used included the Lemhi River louver trap (Figure 4), Marsh Creek weir and inclined-plane trap, and a floating scoop trap in the South Fork Salmon River from fall 1965 through winter 1967 (Ortmann 1968). Fish were also marked by the Bureau of Commercial Fisheries at scoop traps operated in the lower Salmon River near Riggins and Whitebird (Raymond 1979). IDFG operated the Whitebird trap during the fall of 1964 and noted the presence of emigrants in the main-stem Salmon River during the fall with catches peaking in November (Bjornn et al. 1965).

Quantitative assessment of anadromous population production in Idaho began in locations where traps could effectively sample from the entire stream. The first such study was directed at the Sockeye Salmon population in Redfish Lake (Bjornn et al. 1968). From 1955 through 1966, smolt production was measured as the sum of downstream weir catches expanded for missing days by interpolation. A series of studies employing emigrant traps ran during 1962-1975 in the Lemhi River drainage in cooperation with the University of Idaho (Bjornn 1978). Emigrants were collected with a louver trap (Bates and Vinsonhaler 1957) located on a weir in the upper Lemhi River (Figure 4) and with an inclined-plane trap on Big Springs Creek, tributary to the Lemhi River. Flows at these locations are dominated by groundwater (Holubetz 1967) and hence did not have the fluctuations typical of most streams in central Idaho. The study started in Big Springs Creek as part of a project to re-establish steelhead (Bjornn 1965, 1966) and expanded to include

steelhead and Chinook Salmon in the Lemhi River (Holubetz 1967, 1968). Fish were marked with brands and fin clips to make a mark-recapture estimate of abundance. These studies were very influential and were widely used to infer population dynamics and life history of anadromous salmonids in Idaho, because quantitative information on emigrants from other places in Idaho was so scarce.

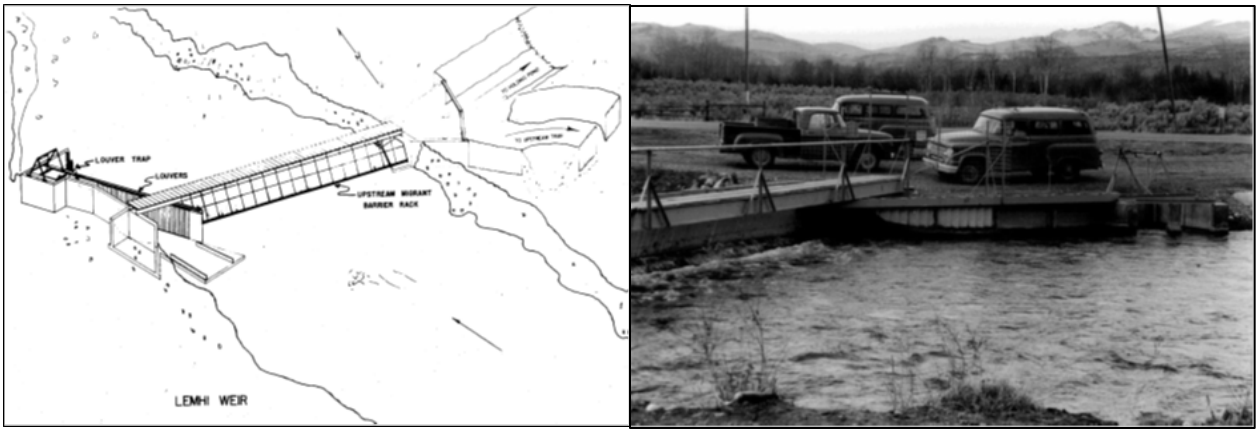


Figure 4. Schematic (left panel) and picture (right panel) of the weir and louver trap on Lemhi River operated 1962-1975. Images are from Holubetz (1968).

Migrant traps were used to evaluate hatchery programs such as the re-introduction of spring-run Chinook Salmon into the Clearwater River drainage and the initiation of mitigation hatcheries in the Salmon River drainage. A scoop trap was run near the mouth of the Selway River and two Kray-Meekin inclined-plane traps upstream near the mouth of Bear Creek during fall 1962 and spring 1963 to evaluate emergence from planted Chinook Salmon eggs (Welsh et al. 1964). A downstream inclined-plane trap was originally part of the Rapid River hatchery trap; it was intended to monitor emigration of hatchery smolts, although it also caught wild Chinook Salmon and steelhead (Reingold 1966). Movement of hatchery Chinook Salmon smolts out of Rapid River was swift and complete (Ortmann 1967). Releases of hatchery steelhead smolts into the Lemhi River were evaluated with the louver trap, a bypass trap at the L-5 irrigation diversion, and Kray-Meekin inclined-plane traps near the mouth; however, survival was very poor and the hatchery program was switched to the Pahsimeroi River (Reingold 1967a). Bypass traps with mark-recapture studies were used to evaluate emigration of hatchery steelhead in the Pahsimeroi River (Reingold 1967a, 1970). Eventually there was a transition from trapping downstream of hatcheries to marking before release and noting recaptures at the Whitebird scoop trap and lower Snake River dams to evaluate hatchery emigration (e.g., Reingold 1971; Irizarry 1971).

In the early 1980s, IDFG began a main-stem smolt monitoring project to collect information on smolt movements, characteristics, and survival in order to inform flow management in the Columbia River hydrosystem (Scully et al. 1983). Two scoop traps with travelling screens and a dipper trap started operation on main-stem rivers in 1983 (Mason 1966; Krcma and Raleigh 1970). The Red Wolf trap operated on the Snake River just downstream of the Clearwater River confluence and the Whitebird trap operated on the Salmon River 1 km downstream of Whitebird Creek. In 1984, the Red Wolf trap was moved upstream of the Clearwater River confluence to the bridge between Lewiston and Clarkston on the Snake River and a scoop trap was installed on Clearwater River at rkm 10 (Scully and Buettner 1986). In 1996, the Whitebird trap on the Salmon

River was relocated upstream to Twin Bridges (rkm 103, Buettner and Brimmer 1996). The intent of the program was to monitor smolts through June but high flows in late spring usually precluded that. Trap efficiencies were generally low (~1%) for yearling Chinook Salmon and even lower for steelhead. The first use of passive integrated transponder (PIT) tags in Idaho was noted in 1987 (Buettner and Nelson 1987). Subsequent development of PIT detection systems in the hydrosystem made smolt monitoring more informative as precise survival estimates could be derived (Burnham et al. 1987). The smolt monitoring project provided an evaluation point for hatchery releases but became more important as a means to place PIT tags to evaluate hydrosystem operations. This emphasis became much greater as Idaho's anadromous salmonids were listed under the ESA in the 1990s.

Renewed efforts were made in the late 1980s to monitor emigrants in tributary streams as numbers of returning adults continued to decline. Intensive trapping efforts began on two sites (Crooked River in the Clearwater River drainage and the upper Salmon River) in 1987 to evaluate population smolt production (Kiefer and Apperson 1987). The intent of the study was to determine the relationships between spawning escapement and juvenile production (Scully et al. 1983) in a manner similar to Bjornn's (1978) work in the Lemhi River. Fish were marked with PIT tags. Emigrant abundance was estimated to derive egg-to-parr survival from redd counts and average fecundity. Survival and smolt production were also estimated at Lower Granite Dam (LGR), the upstream-most dam in the migration corridor. Floating scoop traps, based on a design from Oregon, were used in this study to collect emigrants. The "Humphreys" trap, a floating scoop trap with a self-propelled traveling screen, was developed by the Oregon Department of Fish and Wildlife in 1966 for sampling downstream migrant salmonids (McLemore et al 1989, Figure 5). In this trap type, a paddle wheel or motor turned an inclined-plane travelling screen with baffles to carry the fish back to the trap box. The trap box was kept free of floating debris by a rotating drum screen at the back of the holding compartment. Motorized traps required direct access to power or a series of 12v batteries. Unfortunately, motors and belts had a tendency to freeze and break down in extremely cold weather, limiting their use in much of Idaho.

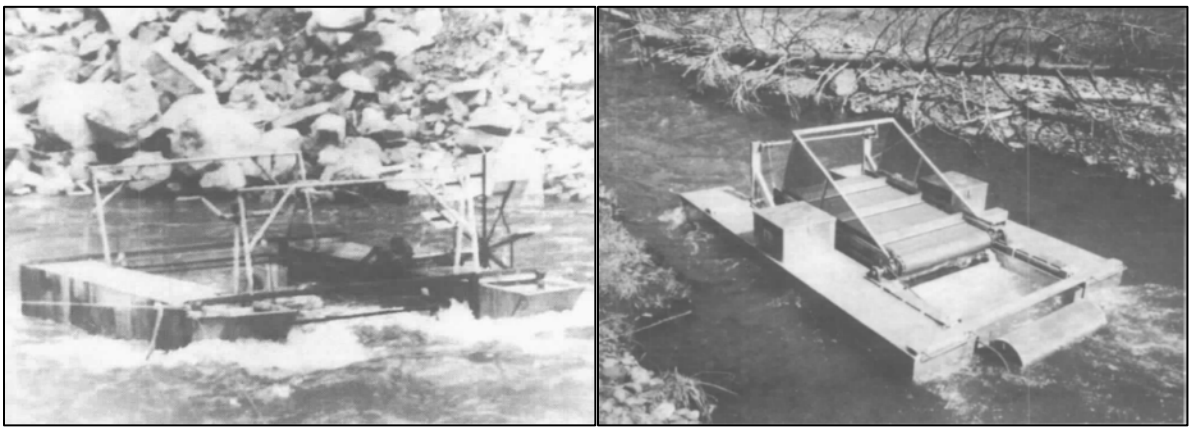


Figure 5. Development of floating scoop traps in Oregon (McLemore et al. 1989). Left: the first self-cleaning scoop trap in Grande Ronde River in 1966 powered by a paddle wheel. Right: a motorized Humphreys scoop trap at Meadow Creek in 1987.

The life-cycle approach to monitoring was greatly expanded with the advent of the RST in the 1990s (Bowles and Leitzinger 1991; Kennen et al. 1994; Thedinga et al. 1994). Rotary screw traps were developed in Oregon and patented in 1990 (Kennen et al. 1994). They were first

deployed in Idaho during fall 1992 as part of a large-scale hatchery supplementation study (Idaho Supplementation Study [ISS], Leitzinger et al. 1993). The purpose of trapping was to collect juvenile Chinook Salmon emigrating downstream to estimate cohort abundance and survival to LGR (Venditti et al. 2015). The ISS project was influential to IDFG trapping operations because of the wide variety of river conditions in which emigrant traps were operated. The study also experimented with fin clips and Bismarck brown stain as marks for small fish (Venditti et al. 2007). Initially, among the study's cooperating agencies, there were eleven emigrant traps (Walters et al. 1999), of which IDFG ran six (Red River, Crooked Fork Creek, South Fork Salmon River, Marsh Creek, Lemhi River, Pahsimeroi River). The scoop traps in Crooked River and the upper Salmon River were included in the study and were eventually converted to RSTs. Traps ran from early March to mid-June, were removed for two months, then re-installed and operated from mid-August to ice up until 1996, after which traps were run through the entire ice-free season, river conditions permitting (Byrne 2000). Traps were added in Colt Killed Creek and American River during 1998. By the end of the study in 2014, IDFG was operating ten screw traps for ISS (Venditti et al. 2015a).

Other IDFG projects used emigrant trapping methods developed by ISS to address their objectives. A new era of Sockeye Salmon smolt trapping was initiated at the Bjornn et al. (1968) weir in 1991 to establish brood stock and estimate smolt production (Kline 1994). Steelhead monitoring studies collected and tagged emigrants in several spots using RSTs: Fish Creek (Lochsa River tributary, 1994-present), Boulder Creek (Lochsa River tributary, 2001-2002), Lick Creek (Secesh River tributary, 2004-2006), lower South Fork Salmon River (1997-2002), Rapid River (lower Salmon River tributary, 2004-present), lower Secesh River (2005-2008), and Big Creek (Middle Fork Salmon River tributary, 2007-present). The objectives were to implant PIT tags and assess life history characteristics (Byrne 2000, 2003). The many trap locations in the PIT tag era are documented in History of Smolt Traps in Idaho on the Wild Salmon and Steelhead (WSS) Collaboration site (WSS Collaboration/WSS Manuals/Screw Trap Manual Supporting Material). Most recently, intensively monitored watershed projects developed other RST locations to address questions regarding habitat restoration and its effect on production of anadromous emigrants (Uthe et al. 2017): Big Bear Creek (2004-present), Hayden Creek (2006-present), and East Fork Potlatch River (2007-present, Figure 1, Figure 2).

Estimation of abundance from emigrant trap data proved difficult and analysis evolved over time. Bjornn et al (1968) simply expanded trap catches for days not sampled because the Redfish Lake Creek weir covered the entire stream width and was assumed to be a complete barrier. In other places, marked fish were released upstream of traps to estimate trap efficiency from the ratio of marked to unmarked fish captured. Early marks included tattoos, fin clips, and freeze branding. Murphy (1955) used a tandem trap design in Orogrande Creek to estimate steelhead emigration past the upper trap. The two traps were 4 miles apart, operated from July 29-November 30, 1954 (Keating 1958). The early enumeration studies were discontinued because of inefficiency of the traps and the effort required to get enough recaptures (Keating 1958). An alternate approach used on large river traps was to expand trap catch by volume of water sampled and time fished (Bell 1958). Mark-recapture studies in the Lemhi River calculated efficiency based on recapture rates (Holubetz 1967; Bjornn 1978). Hatchery studies were more successful because the directed, short-term movements of large numbers of fish facilitated estimates even at low efficiencies (e.g., 2.63%, Reingold 1967a). Scully et al (1983) released marked fish upstream of the Salmon River trap at certain intervals to estimate trap efficiency and then related it to flow.

Analysis became more sophisticated as trap data quantity and quality increased. A statistical model suitable for a single trap was developed by MacDonald and Smith (1980). By the

early 1990s, emigration abundance was estimated from continuous release of marked fish from traps but estimates were made on a seasonal basis (Kiefer and Forster 1991; Leitzinger et al. 1993). When possible, shorter time intervals were used (Nemeth et al. 1996). Bootstrap confidence intervals were first used by Walters et al. (1999; see Thedinga et al. 1994). Abundance estimation at IDFG traps stabilized after the study by Steinhorst et al. (2004). Steinhorst et al. (2004) proposed a time-stratified method that was statistically valid for migrating fish (Sockeye Salmon and steelhead) with recommendations for defining time periods. There are alternative ways of doing time-stratified emigrant estimates (e.g., Bjorkstedt 2000 in California). Volkhardt et al. (2007) give a short summary of alternate approaches.

Survival of emigrants from the tributary traps to the hydrosystem is also important. Several early studies established the importance of the fall movements; hence, winter mortality must be accounted for in order to estimate true production from anadromous rearing habitat in Idaho. The basic methods to estimate survival within the hydrosystem were derived by Burnham et al. (1987) and enabled by the development of the PIT tag. Survival from tributary to the hydrosystem was a logical extension of these methods. Operations in the 1990s included release of PIT-tagged fish for estimation of production of smolts to LGR (Kiefer and Forster 1991; Leitzinger et al. 1993). Initially, Bowles and Leitzinger (1991) recommended 300-500 tags be placed for these investigations.

Marking and tagging at traps in Idaho have played an important part in evaluation of hydrosystem management. The Comparative Survival Study (CSS) was developed and initiated in 1996 to evaluate smolt transport and hydrosystem management (upstream/downstream comparison). The CSS started with a focus on hatchery spring/summer-run Chinook Salmon (Berggren and Basham 2000). Soon the focus expanded to wild yearling Chinook Salmon and emphasized tagging upstream of the hydrosystem, rather than at dam bypass traps (Bouwes et al. 2002). Steelhead were added as a study species in 2005, as were fall Chinook Salmon and Sockeye Salmon later. Initially the CSS relied on tagging at hatcheries and at large main-stem river traps operated by IDFG (Snake River, Salmon River, and Clearwater River) and by Oregon Department of Fish and Wildlife (Grande Ronde River). The CSS supplemented tagging efforts by providing PIT tags to the projects operating the traps. The distribution of tags by CSS expanded over time to include RSTs collecting wild Chinook Salmon (starting in 2002) and steelhead (starting 2010). Recent efforts rely more on RSTs located in the tributaries to allow marking wild fish at the major population group level (McCann et al. 2018). The CSS project maintains series of hydrosystem survival and smolt-to-adult return (SAR) estimates, including effects of hydrosystem management (in-river passage and transportation), as well as life cycle models and analyses of subsidiary questions (e.g., comparison of PIT-tag-based versus run-reconstruction SARs). Thus, RSTs operated by IDFG contribute to downstream management studies that extend to adult return.

RECENT STATUS

The current goal of RST operations by IDFG is to provide long-term, continuous research, monitoring, and evaluation of the status of the state's populations of anadromous salmon and steelhead. Recommendations for monitoring to address population status assessments across the Columbia River basin relevant to juvenile trapping include: 1) annual estimation of juvenile emigrant abundance across major populations, and 2) estimation of the adult-to-juvenile productivity of both tributary emigrants and smolts through the Columbia River basin hydrosystem (Crawford and Rumsey 2011). These are two of several critical metrics necessary to assess overall trends in abundance and productivity within freshwater habitat. Standardized sampling

through time and across locations can allow long-term evaluations of population trends and comparisons, if traps are located appropriately downstream of important spawning and rearing habitats. Rotary screw traps have been the primary tool used by IDFG since the early 1990s: 1) to sample populations or spawning aggregates of interest to estimate juvenile emigrant abundance, and 2) to tag emigrants with PIT tags to estimate smolt survival to and through the hydrosystem (Venditti et al. 2015b; Copeland et al. 2015; Apperson et al. 2016, 2017).

Over the last ten years, IDFG reviewed operation of its RST fleet and made significant changes to expand collection of wild steelhead across the Clearwater and Salmon major population groups and to improve efficiency. In 2010, IDFG operated sixteen RSTs, ten of which were operated to support ISS and focused on Chinook Salmon. An additional three were operated to monitor the steelhead populations in Fish Creek, Rapid River, and Big Creek. Another three traps were operated to support Intensively Monitored Watershed projects (IMWs) in Big Bear Creek, East Fork Potlatch River, and Hayden Creek. Beginning in 2010, a new site was started in lower Marsh Creek to sample from the entire Marsh Creek Chinook Salmon population. In 2013, Quantitative Consultants Inc. started the lower Lemhi River trap to support the Lemhi River IMW project and the Integrated Status and Effectiveness Monitoring Program (ISEMP), as well as to improve sampling for the entire Lemhi Chinook Salmon population. As ISS was concluded in 2015, many changes were made. Operations ceased at five locations: Crooked Fork Creek, Colt Killed Creek, Red River, American River, and upper Marsh Creek. The trap located at Knox Bridge on the South Fork Salmon River trap was moved downstream to a location near the confluence with Krassel Creek. Traps were also deployed to two new locations (North Fork Salmon River and the Lochsa River). The upper Lemhi River trap operations were transferred to the Lemhi River IMW project and operations of all other ISS traps (Upper Salmon River, Pahsimeroi River, and Crooked River) were transferred to Idaho Natural Production Monitoring Project or Idaho Steelhead Monitoring and Evaluation Studies to maintain critical data series. In 2020, IDFG assumed operation of the lower Lemhi River trap and the Idaho Natural Production Monitoring Project Idaho Steelhead Monitoring and Evaluation Studies were merged. As of this writing, IDFG operates 15 RSTs to trap juvenile anadromous salmonids (Figure 6, Appendix A).

Three non-RST juvenile traps have been operated by IDFG for over 20 years. Two are operated on the main-stem Salmon and Snake rivers to place tags for the Smolt Monitoring Project. These are large scoop and dipper traps, respectively. The Clearwater River scoop trap proved ineffective and ceased operations in 2014; it was replaced by operation of the Lochsa RST and a RST operated by the Nez Perce Tribe on the South Fork Clearwater River. The last current IDFG trap is a weir used to sample juvenile Sockeye Salmon emigrants, as well as adults, from Redfish Lake Creek for the Sockeye Captive Propagation project. Each also places tags used by the Comparative Survival Study (CSS, project 1996-020-00). Because these traps are so different from RSTs in their purposes, they are mentioned here for completeness and will not be discussed further.

The IDFG uses its current RST fleet for several purposes. Traps are used to quantify emigrant abundance, gather basic biological data, and place PIT tags in steelhead and spring/summer-run Chinook Salmon. This information is used in multiple contexts: estimation of freshwater production and productivity for key populations, evaluation of hatchery supplementation and habitat restoration, and estimation of hydrosystem survival and SARs. Traps are deployed in locations in accordance with the Anadromous Salmonid Monitoring Strategy (CBCAMW 2010) as well as to support IMW projects, integrated broodstock programs for Chinook Salmon hatcheries, and to monitor key long-term sentinel populations. All current locations are supported by adult monitoring. All IDFG traps place tags used by CSS to conduct analysis of hydrosystem and ocean effects on survival of wild salmon and steelhead. The information

generated is used to assess freshwater production and productivity for key populations, evaluate hatchery supplementation and habitat restoration, and estimate hydrosystem survival and SARs. Hence, data generated by each trap is used in multiple contexts from local watersheds to the Columbia River basin.

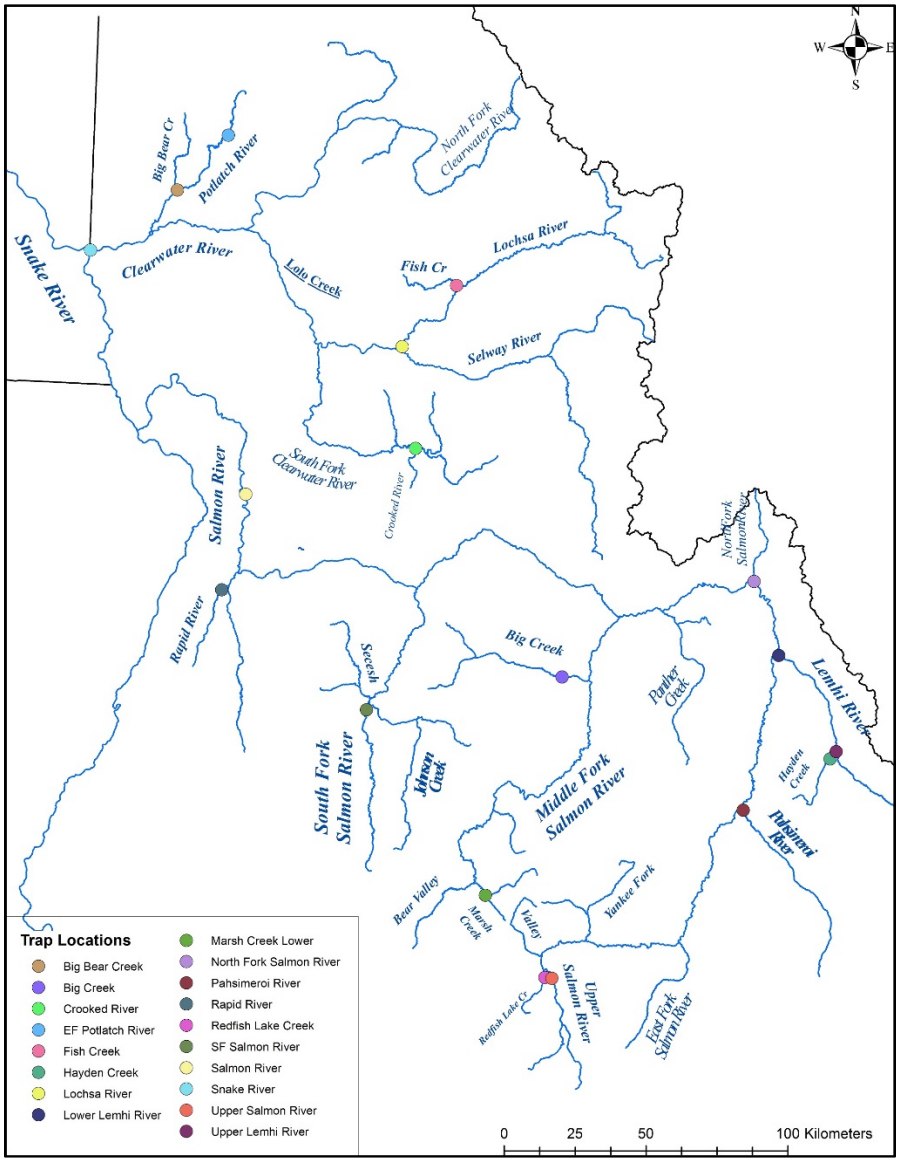


Figure 6. Locations of 18 emigrant traps operated to collect anadromous salmonids by Idaho Department of Fish and Game during 2020. All are RSTs except Redfish Lake Creek (weir trap), lower Salmon River (scoop trap) and Snake River (dipper trap).

We now turn to how the information to support population monitoring is developed. This is a complex topic extending from field work to data analysis. Several main sections follow.

METHODS TO INSTALL AND OPERATE ROTARY SCREW TRAPS

In this section, we review how to put traps in the water and operate them safely. Processing the catch and dealing with the data will be covered in subsequent sections. Getting RSTs into a specific location on the stream involves planning and permitting, so we start with those considerations.

Locating and Permitting Traps

Emigrant traps are strategically placed because good trapping locations don't exist everywhere. Crawford and Rumsey (2011) recommended working at locations low enough in major drainages to cover a maximum of habitat while still in stream sizes that are small enough to achieve abundance estimates with coefficients of variation ≤15% for salmon and ≤30% for steelhead. Trapping duration should be sufficient to encompass at least 90% of the emigration period. Therefore, boundaries of the target population or major spawning group should be considered, as well as tributaries to be included or excluded from sampling.

Selection of a monitoring location involves several logistical considerations. Permission should be obtained from the landowner or land manager before considering any specific sites. Stream channel morphology must have adequate depth to allow trap operation during all seasons of interest. An ideal location has a stream cross section with a well-defined thalweg, yet has a variety of velocities and depths, and relatively simple substrate with few or no large boulders near where the trap will float. Such a stream configuration will allow operation to be extended through a wider range of discharge by providing the ability to operate the trap at various locations across the stream. An eddy on an accessible side of the stream will provide for a safe place to assemble the trap, and a safe harbor in which to anchor the trap during times of high discharge when safe operation is not possible. Other considerations include accessibility of the site, safety for trap install, an area for a work station, proper anchoring support, and upstream access to release fish.

Trapping locations can have a variety of constraints. A site visit with the property owner or land manager early in the process can help establish and maintain relations, understand each entity's goals and constraints, and expedite permitting. The individual or group proposing a new trapping operation should have the proposal for need and location (with any alternatives), installation methods, and general operation plan internally vetted with IDFG regional and Fisheries Bureau leadership before pursuing permitting with land managers. Agreements with private property owners or special use permits from federal agencies may be necessary. Ideally, sites located on private land should be established with written agreements specifying the department's access stipulations, including any maintenance, costs, and liabilities.

Proper procedures must be followed before installing a RST at a site for the first time. Installations on public land may require public land managers to conduct National Environmental Policy Act of 1970 (NEPA) compliance. The purpose of NEPA is to assure that all branches of government give proper consideration to the environment prior to undertaking any major federal action that significantly affects the environment. Each year, Bonneville Power Administration (BPA) issues a Categorical Exclusion providing for environmental compliance for operation of RSTs in Idaho to monitor juvenile anadromous emigrants for projects funded by BPA. Environmental compliance is a specific work element in the BPA statement of work. The lead IDFG biologist obtains ESA authorization for handling listed species at the proposed traps and shares them with the BPA Environmental Compliance lead who reviews the statement of work before the corresponding contract is issued. The BPA Environmental Compliance lead updates and maintains information about the current status of environmental compliance requirements (https://www.bpa.gov/efw/FishWildlife/InformationforContractors/Pages/Environmental-

Compliance-Overview.aspx). Hence, some permitting activities also occur annually before operations can commence.

An archeological survey may be required prior to ground disturbance at a new trap site. If any digging will be required to install anchors or other infrastructure then reviews must be completed to satisfy the National Historic Preservation Act of 1966. The review process is explained and conducted through the Idaho State Historic Preservation Office (https://history.idaho.gov/section-106/) as part of a Section 106 Review.

Permits to collect and handle fish species listed under the ESA must be obtained before trapping. For anadromous salmonids listed as 'threatened' under the ESA, permits for take under ESA Section 4(d) from NOAA Fisheries are renewed each year through application by IDFG permit holders, principle investigators, or primary contacts: https://apps.nmfs.noaa.gov/. Existing permits are renewed by going into the portfolio, opening the application, and clicking on the "Renew Permit/Authorization" link. The window for permit applications is September through mid-October. Tagging and handling information is coordinated among multiple locations. All coordinating project permits are posted online, along with the Terms and Conditions, and Authorization Letter. The current permit for Bull Trout *Salvelinus confluentus* is an ESA Section 6 Cooperative Agreement that is not renewed annually but remains in force until terminated by either party. There is also an ESA Section 10 permit for tagging, handling, and transport of endangered Sockeye Salmon. Current copies of all permits must be kept at all trap sites.

An ESA Take Tracking spreadsheet is used to keep track of take covered under the 4d permit during the season to ensure that permits are not exceeded. Trap tenders keep track of the number of Chinook Salmon and steelhead captured and tagged and submit those numbers to their supervisors weekly. Trap supervisors fill out the Take Tracking spreadsheet online https://collaboration.idfg.idaho.gov/ScrewTrapTake/Shared%20Documents/Forms/AllItems.asp each week during the trapping season. The Take to Date and Mortality to Date are cumulative. Explanations of take types are noted in the column headers. This spreadsheet is monitored by NOAA Fisheries and the WSS Data Coordinator at Nampa Research to 1) make sure that take is not exceeded, 2) that modifications can be made as necessary, and to 3) assist with the 4(d) Take Report at the end of the year.

All handling of ESA listed species is reported annually. A detailed take report is submitted to NMFS at the end of each year. The report summarizes measures to minimize stress or harm to fish, according to the terms and conditions. The annual 4(d) reports are due by January 31. The report is filed on the NOAA Apps website by clicking on Start Report when logged in to the portfolio. Reporting for non-anadromous fishes is handled by request from the Fisheries Bureau.

Installation of Rotary Screw Traps

Installation of RSTs involves several steps that are covered in this section. The rigging that holds the trap and allows movement across the stream is important, as is the connection of the trap to those cables. Both are described separately. Lastly, we describe how RSTs themselves are assembled and disassembled. Trap installation can be difficult, so having someone with experience on-site is ideal. An engineer should be consulted before initial install.

Trap Rigging

The main cable (static line) that will hold the trap must be securely anchored to each bank approximately ten meters upstream from where the trap is to be operated. The cable is strung a

minimum of two meters higher than the stream surface at high discharge to allow large debris and recreational boaters to pass under safely. The options for anchors used largely depend on the location and what is available (Figure 7).

A) Eye bolt in boulder

B) Webstrap around tree

C) Rock gabion

D) Wood pole

E) Cement posts

F) Heavy duty winch

Figure 7. Six types of anchors for the main static cable.

Anchoring into solid bedrock or existing large boulder clusters is ideal, and is typically readily accepted by land owners or managers because anchor installation causes no ground disturbance and is visually benign. A hole is drilled vertically into the boulder with a hammer drill and carbide masonry bit to accommodate a 5/8” by 8” eye bolt. Injectable mortar epoxy adhesive is used to cement eye bolts in place. Trees are also natural anchors but require, at minimum, a 20 inch diameter and well rooted trunk. Smaller trees can break, pull out, or snap if there is too much force on the trap, e.g., from high water or catching a log. Artificial anchors can be wooden, steel or cement posts buried six feet in the ground, stacked rock gabions, bridges, or heavy-duty winch systems. Heavy-duty winch systems are used in larger streams, such as the Salmon River or Lochsa River, where the water pressure makes it too difficult to pull the trap over by hand.

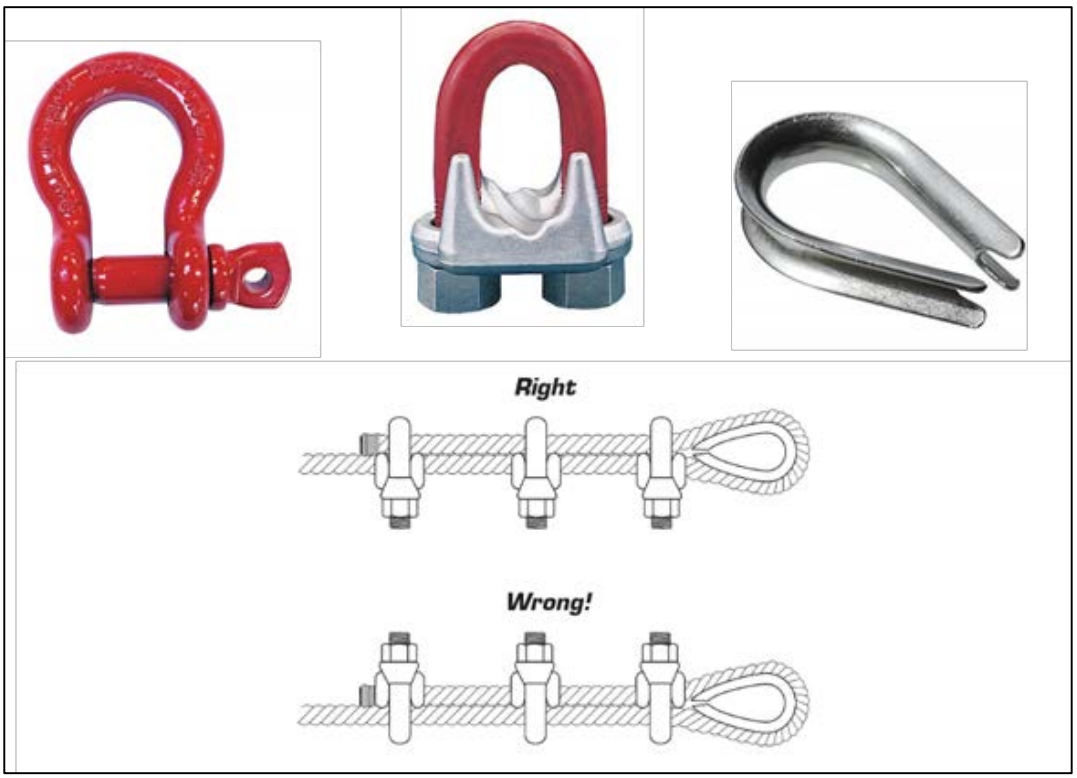


Figure 8. Hardware and schematic for rigging cable ends. Top row shows a shackle bolt, Crosby wire rope clip, and thimble (left to right). Bottom two illustrations show the proper and wrong way to rig a cable end. Never saddle a dead horse.

In most situations the static line is secured to the anchor via webstraps or eye bolts. The cable end is attached to the webstrap/eyebolt with a heavy duty anchor shackle bolt, at least ½” or larger in bigger water (Figure 7, panels a-d). The cable is attached to the far bank first. A Crosby wire rope clip (cable clip) is used to fix the loose end of wire rope back to the cable body to form a thimble eye or loop (Figure 8). It consists of a U-shaped bolt which fixes the wire rope cable against the saddle when the two nuts are tightened. The saddle is always against the live portion of the wire rope and not the dead or termination end of the wire rope; this leads to the expression “Never Saddle a Dead Horse” (Figure 8, bottom). Two or more cable clips are required for each eye or loop, in case one slips. The cable clip is sized the same as the cable. A 3/8” cable

takes 3/8” cable clips. When securing the end of a cable to anything other than a snatch block a thimble is used to prevent fraying of the cable.

A come-along with cable grippers attached to each end is used to tighten the cable through a snatch block (fastened to the anchor) on the near bank (Figure 9). One cable gripper is attached to the “live’’ cable that is stretched across the river and the other cable gripper is attached to the cable end has been run through the snatch block. Once the cable is tight the cable clips are tightened down to secure the cable end. It is important to move back and forth between nuts when tightening cable clips as one loosens as the other is tightened.

Figure 9. Gear for setting the static line. Come-along hand winch with cable gripper attached to the static line.

A larger snatch block is hooked to the static line and supports the cable and snatch block system that is attached to the trap, referred to as the yoke. Two snatch blocks with shackles can be fastened to the cable, one near each bank, and a rope looped through and tied to the main block (Figure 10). This allows the trap to be moved back and forth across the river. This type of rigging varies depending on the anchor type and locations. Winch systems may not loop back (bottom right, Figure 10). Either way, a cable or rope will need to be attached to the main snatch block in order to move the trap side to side.

Figure 10. Snatch block rigging. Top: looped rope attached to the static line pulley for lateral movement. Bottom left: Yoke attached to static line pulley with looped rope. Bottom right: non-looped lateral cable and yoke attached to static line pulley.

Yoke Rigging

Moving the trap upstream or downstream is accomplished by hand or by winch. Moving the trap upstream can be done by one person if the yoke rigging has a three-way snatch block reduction (Figure 11). Starting at the main snatch block on the static line, thread the cable down to a yoke cable snatch block, back up to another snatch block attached to the main, and back down again to the pontoon. Pull the cable from the end in the upstream direction to get the reduction of the last snatch block on the pontoon. The cone is always raised before attempting to move the trap up- or downstream. Two cable clips are placed on the extra cable behind the pontoon snatch block to prevent the trap from moving downstream on its own; they block the cable from sliding through the snatch block. They are loosened and slid to move the trap. At least one of these clips is tight when moving the trap and both of them tight when not moving the trap.

Always make sure both cable clips are tight before lowering the cone after it is in position. If a winch is used then the three-way reduction is not needed and the cable can go from the yoke to a snatch block attached to the main block and back down to the winch, which is typically attached to the front of a pontoon (Figure 12). If there isn't a need to move the trap upstream or downstream, then no snatch blocks are required for the yoke.

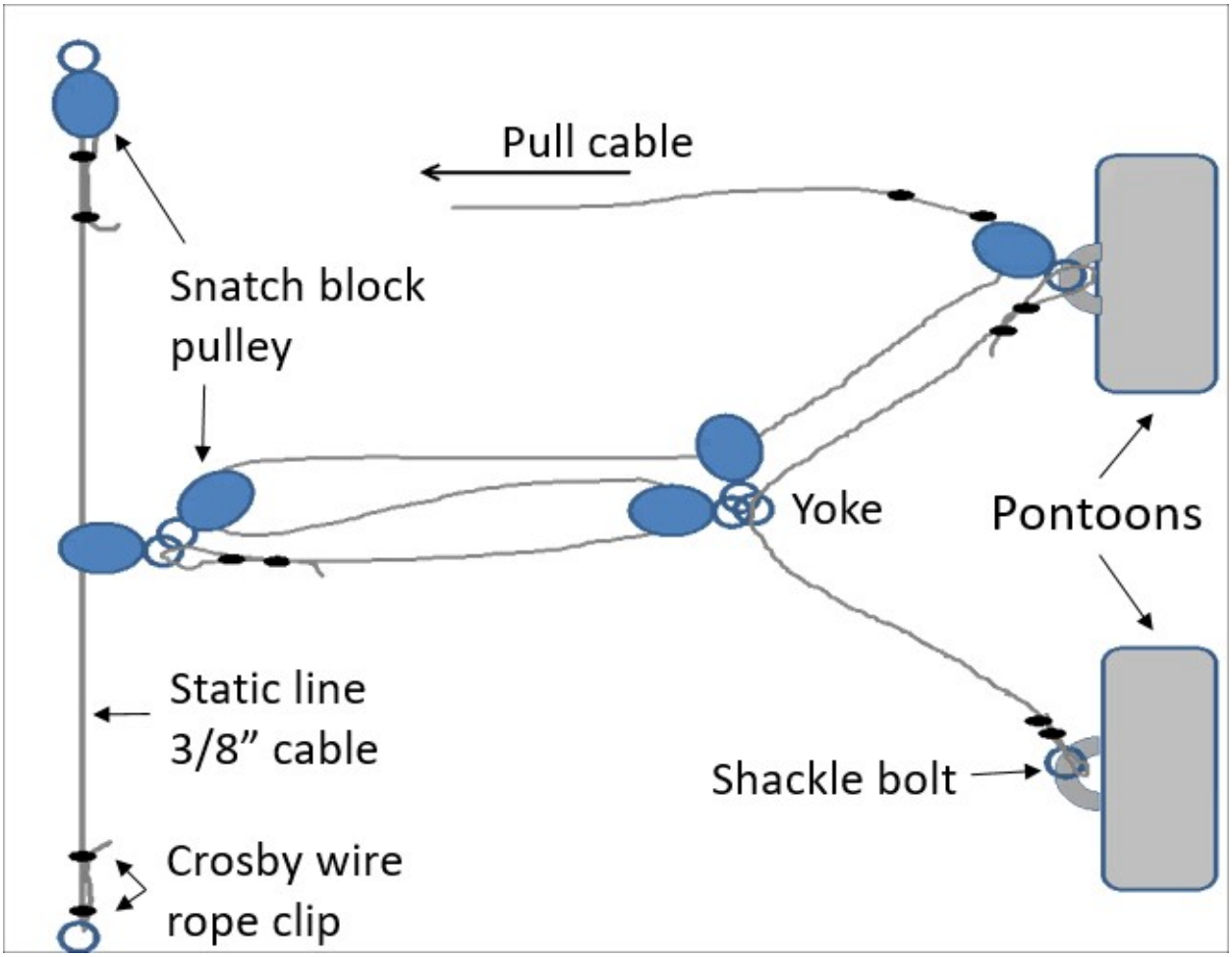


Figure 11. Rigging the yoke to the pontoons with a three way reduction for one person operation. Pulling the end of the cable in the direction of the arrow moves the trap upstream.

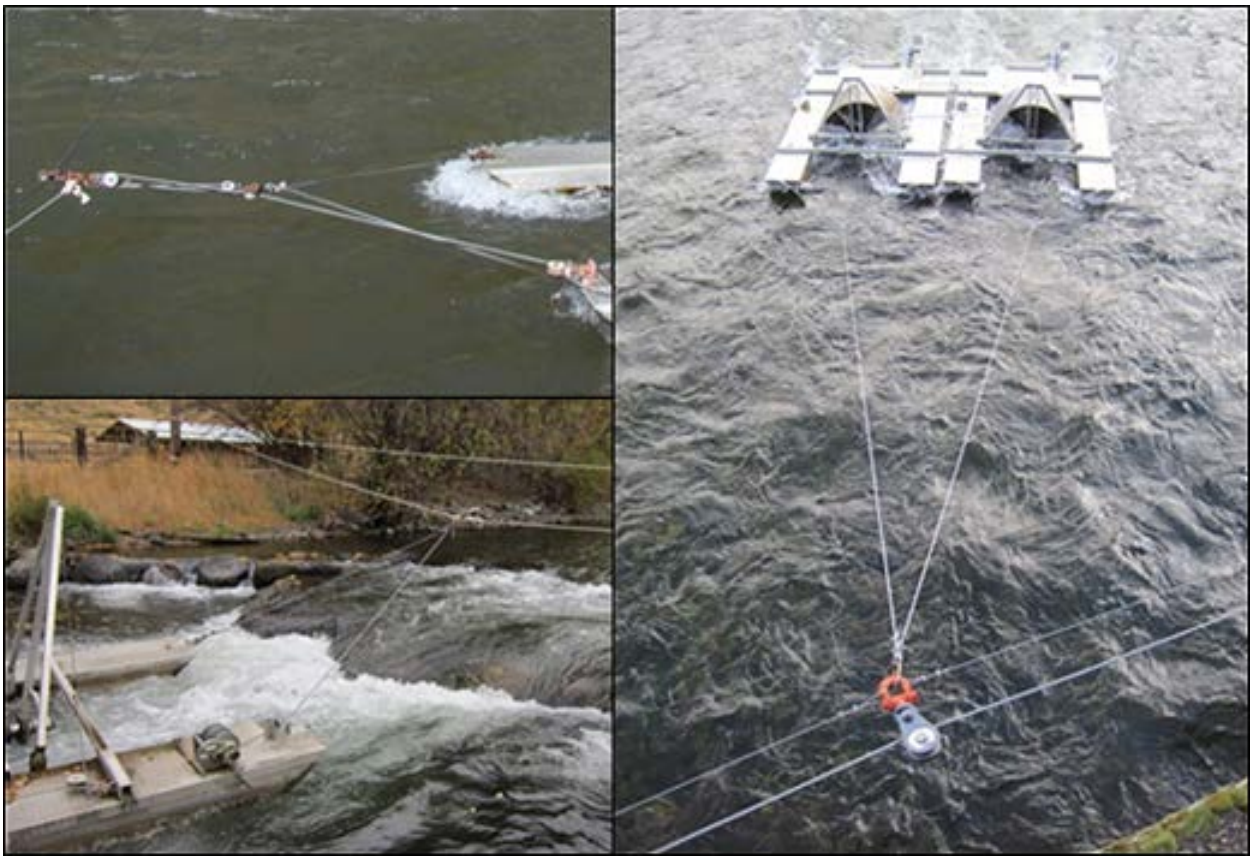


Figure 12. Options for rigging the trap to the static line. A three way reduction manual yoke (left top), a winch yoke (left bottom), and a double yoke without snatch blocks or winches (right).

Trap Assembly and Removal

Rotary screw traps can be broken down to seven components for installation, removal, and transport. These components are: 1) port pontoon, 2) starboard pontoon, 3) #1 crossbeam, 4) cone with #2 crossbeam attached, 5) trap box with #3 and #4 cross beams, 6) bi-pod, and 7) pontoon handrails. All crossbeams are secured with 9/16” stainless steel bolts, flat washers, and nylon lock nuts. The bipod and shaft collars are secured with 3/4” bolts, flat washers, and nylon lock nuts. Ideally four people are needed for installation, as parts are heavy and awkward to carry. One-inch weir pickets or piping are used to carry the pontoons, inserting them where the crossbeams would go. Complete assembly instructions are in Appendix B.

Assembly of the trap begins with connecting the two pontoons with the #1 crossbeam (Figure 13). The pontoons are connected to the rigging or tied to the bank with a rope to prevent them from drifting away. Next, install the trap box with #3 and #4 cross beams. It may be necessary to lightly pry the beam channels open with a crow bar to loosen the fit, being careful not to break the welds. Tie a heavy rope across both pontoons at the cleats to rest the cone on until it is secured to the trap box and the bipod. The rope prevents the cone from falling through to the substrate. Ensure the cone has front and rear wear sleeves over the main shaft with four

collars total. Remove the #4 collar from the downstream end of the cone shaft so the shaft will fit through the flange bearing of the trap box. Temporarily rest the downstream end of the cone shaft on the #3 crossbeam of the trap box. Position two people on the #2 crossbeam on the front of the cone and one person to direct the shaft through the bearing. Carefully lift and move the cone forward so the rear shaft clears and slowly back it into the flange bearing. Bolt the #4 collar back on once the shaft is all the way through. Attach the bipod while the cone is still resting on the rope with the cross bar positioned on the same side as the crank (starboard) winch on the pontoon used for raising the cone (Appendix B). If the cross bar is on the port side, then the stress can break the bipod. The bipod is marked S on the starboard side and P on the port side. The bolts that attach the bipod to the pontoons are fastened just loose enough to allow it to pivot freely but snug enough that it stands straight up. Ensure the crossbeam #2 is over the front wear sleeve with the lip of the plastic bushing facing forward. If the bushing is backwards, it could wear through the flange bearing. The black sliding block fits onto the front cone shaft and the last collar is secured on the front sleeve. With the bipod in place, attach the trap winch cable using a series of three cable pulleys attached to three points on the trap with stainless steel quick links. Attached the end of the cable to the eyelet on the crossbeam #2 with a stainless steel quick link (Appendix B). Crank up the slack in the cone cable and remove the cone-supporting rope. Add the handrails to the pontoons. If they are not already, connect the trap and yoke to the main cable. A safety cable or heavy rope is attached to the pontoon and tied to the bank in case the static line, yoke, or anchors fail.

Removal of the trap reverses the assembly process described above. Lift the cone and move the trap close to shore. It may be easier to keep the trap attached to the main cable until the cone is removed. Replace the bolts after removing each part so they won't be lost. Drain the water from both pontoons before attempting to carry them. Inspect all parts for cracks and damage and rigging cable for fraying. Clearly label ropes and rigging before storage.

The trap is now ready for transport to place of storage. In the absence of a specifically manufactured screw trap trailer, a single or double axel 16' x 8' foot flatbed trailer is needed to haul a RST (Figure 14). Traps typically weigh about 1300 lbs (590 kg) so the trailer must be capable of handling that weight. Load pontoons facing forward, starboard side first. Load the cone on the front half of the trailer with the back shaft facing forward. The port pontoon goes on the other side of the cone. Place the trap box between the pontoons and behind the cone at the back of the trailer. It may be necessary to block the trap box with 4x4 posts to clear the pontoons. Finally, the bi-pod and railing lay flat on the trap box lid. Use four heavy ratchet straps to secure the trap. Place one strap over the back cone shaft that is facing forward. The shaft must be supported by a t-post. A second strap over the center rib of the cone. Another strap over the trap box and a final strap across the front pontoons. Scrap plywood is wedged between pontoons and the cone to prevent damage to the screen. A manufacturer-supplied setup guide is provided in Appendix B.

Figure 13. Installation and assembly of a RST in the water. Top left: Attach the #1 crossbar to the pontoons. Top right: attach trap box to the pontoons. Middle left: Secure a cone support rope to the pontoons. Middle right and bottom left: rest the cone on the rope and slide the shaft into the trap box flange bearing. Bottom right: attach the bipod and winch cable to the cone.

Figure 14. Screw trap loaded on a flatbed trailer. Note locations of straps and tie-downs.

Trap Operations

Operation of RSTs is determined by fish behavior as modified by logistical factors. Juvenile steelhead and Chinook Salmon move throughout the year with pulses in the fall and spring at most locations in Idaho (e.g., Figure 15; also see Lutch et al. 2003). Traps are deployed typically in late winter (March) and removed when streams begin to ice up (November). The main exceptions are the RSTs in the low-elevation areas of the Potlatch River watershed, where summer flows are too low to operate RSTs. During full operation, RSTs are fished round-the-clock, seven days a week, conditions permitting (e.g., unsafe flows, releases of hatchery fish). For example, deployment of the Rapid River RST is delayed for volitional hatchery smolt releases but operates round-the-clock as soon as possible.

The actual fishing position of the trap in the stream is determined by a tradeoff between efficiency at catching fish and the risk of damaging the trap. Efficiency is tracked by the number of recaptures. The goal is to fish the trap at the highest efficiency possible because the confidence interval of the abundance estimate decreases as recaptures increase. Ideally, the trap is fished in the thalweg (region of the stream that has most of the flow by volume) and in line with the current to maximize efficiency. To prevent excessive wear and damage, the cone should not exceed 12 RPM. An RPM higher than 12 is an indication that the water is high and swift enough to catch large debris, logs, and/or fill the trap box with silt, all of which can result in damage or sinking to the bottom. These guidelines were developed for older traps with small diameter shafts in smaller tributaries. On main-stem sites like the Lochsa River, traps may be run up to 15 RPMs, but that requires replacing bushings as needed and added extra pontoons to decrease the chance of sinking. To reduce the RPM, the trap can be moved a few feet downstream or closer to shore. Moving a trap out of the thalweg will lower trap efficiency but should be done when necessary. Assuming conditions safely allow, trap tenders can find the most efficient fishing position at a particular flow by trial and error, i.e., fishing it just a few feet in either direction for a few days. In low water, the highest efficiencies will usually be attained directly in the thalweg.

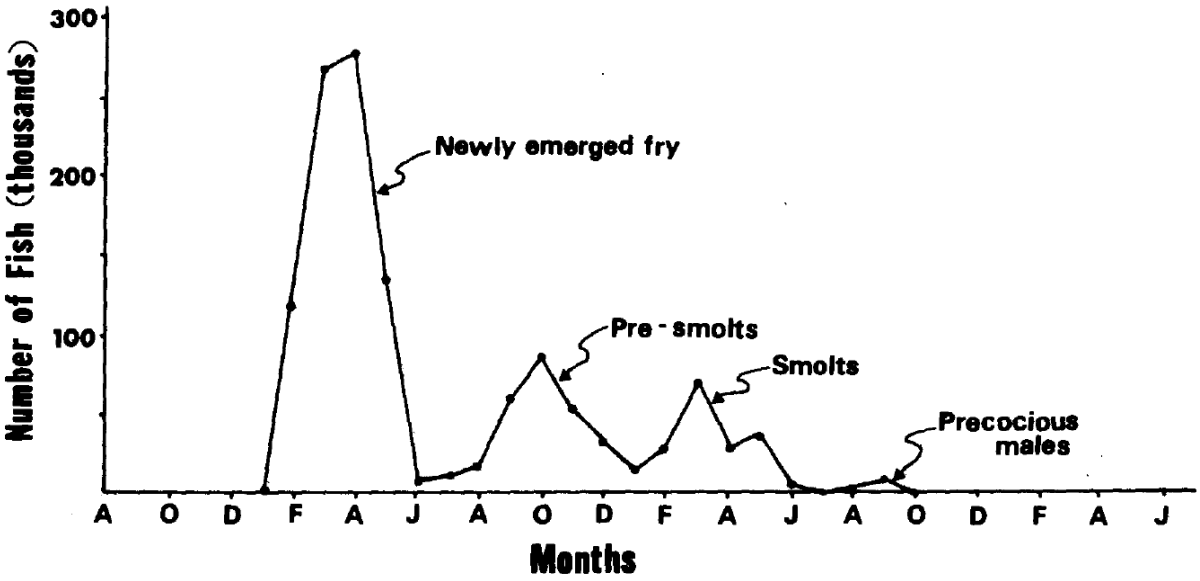


Figure 15. Movement of the 1964 brood year of Chinook Salmon past the Lemhi River weir. From Bjornn (1978). Letters on the X axis correspond to every other month (February, April, June, August, October, December). Numbers were estimated based on recaptures at the louver trap (Figure 4).

During low flows, experienced in March or late summer, it may not be possible to run the trap with the cone lowered all the way down. The cone may be raised a few turns but if it is not supported the front shaft will wear through the bushing and flange bearing of the #2 crossbar. Place blocks on the pontoons under the ends of the crossbar (Figure 16) and lower the cone until there is slack in the cable. This lifted position allows the cone to turn without stressing the cable and bushings. If the water current is so slow that the cone has difficulty turning, then try removing the cleaning drum tire to decrease drag. Generally, during flows this low, automated cleaning doesn't work well and cleaning must be done manually. Trap tenders can also increase flow into the cone building rock fykes upstream of the trap or installing flow diverters made of plywood and T-posts. In some cases, larger boulders may need to be moved with rock bars out of the thalweg at the trap location. Ice, sand, and debris can weigh the trap down enough to make it rub on the substrate, which will tear rivets and mesh. A good rule of thumb is to lower the cone until it touches bottom and then raise it two turns of the winch to prevent it from rubbing on the substrate. Always lock the winch once the trap and cone are in place.

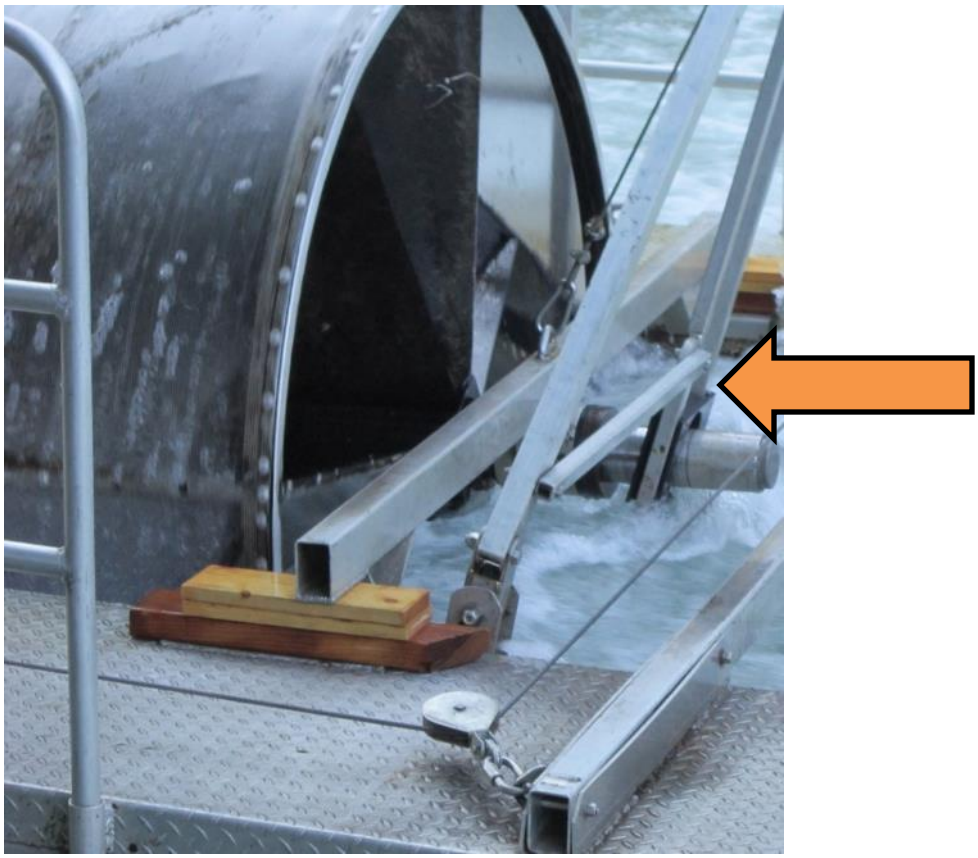


Figure 16. Wood blocks can be placed under the #2 crossbeam to keep the cone raised off the substrate while relieving tension on the cable. Note that the cross bar (indicated by the arrow) is on the same side as the cone winch (not visible in picture).

During high flows, the trap is best operated between the thalweg and the bank. If the water rises slowly, work the trap closer to the bank if possible. The trap can fish right off of the bank if depth allows. The recapture rate (efficiency) will be lower during higher water because there are fewer fish per volume of water in the river. If water rises rapidly, cease operations.

Program personnel must check traps and process fish at least once daily during daylight hours. Traps can be accessed by wading, a gangplank or walkway, or boat. Move the trap to a safe spot to access if the water is too high to wade. Make sure that depth is sufficient for easy wading. Flows may increase during the day, making it difficult to get to and from the trap as fish are removed for processing. If the trap is pulled to shore for access, then secure it to the bank so it does not drift back out while collecting fish out of the trap box. High water flows, debris, and ice prevent trap operation on some days. When such problems are anticipated or when unusually high numbers of hatchery juveniles are passing following releases, traps may be checked several times throughout the day and night as necessary to avoid harm to fish and avoid damage or loss of the RST. There will be times when it is necessary to raise the cone and park the trap for a week or more due to hatchery releases or high water. Guidance for safety, maintenance, and fish handling follows below.

Trap Safety

Safety is paramount when working on an RST. Water exerts powerful force on the trap and personnel, if they are in the water. Traps operate in cold temperatures such that hypothermia becomes a risk. Further, trap tenders are often at the site alone. It is imperative that trap tenders never compromise safety procedures. Personnel should be in uniform and not wearing any loose clothing that can catch on the cone. In cold weather, neoprene waders are worn to prevent hypothermia. A wading belt will prevent the waders from filling with water in the case of falling. Wading boots should have felt soles to help traction. If there is a thin layer of ice on the pontoons, then rubber will not grip and personnel may slip into the cone or water. Open toed sandals or flip-flops are not allowed while working on a trap. A dry set of clothes should be kept nearby at all times (i.e., in the tagging station, truck, or wader bag). A personal flotation device is required at all times (e.g., Figure 17).

Figure 17. Safety features for working on an RST. Note the railings, personal flotation device, safety line, and waders.

Safety features are built into the trap design. Several are mandated by the Occupational Safety and Health Administration (OSHA; Figure 18). A railing on the trap prevents personnel from accidentally getting impinged on the side or back of the cone. There should be a solid plate along the port side rail near the pinch point at the bottom. OSHA also requires shaft and cone guards, a throw rope, and life ring be present on the trap. These items are for the safety of the trap tender while they are on the trap.

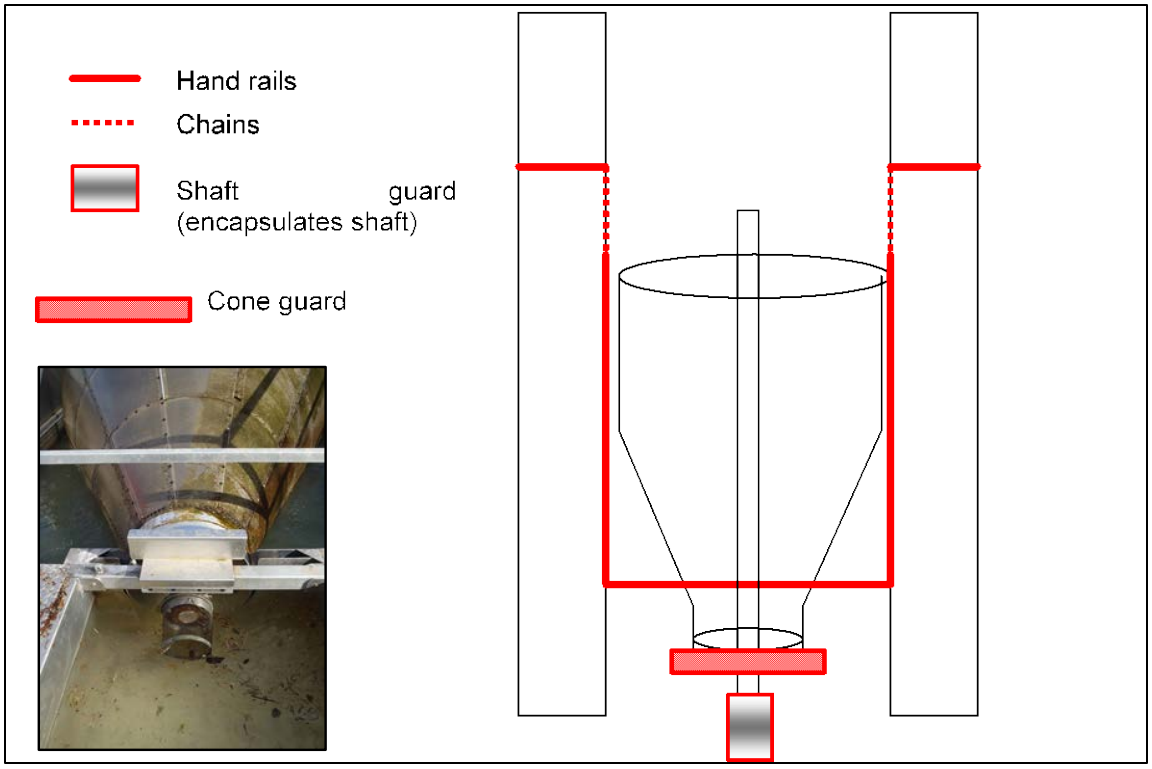


Figure 18. Schematic of OSHA-required safety features for screw traps. Inset at lower left shows a detail of the cone and shaft guards.

Signage and flagging are required for public safety. Wires and cables are marked with bright colored flagging. Static cables must be high enough for boaters to fit underneath during high water. Caution signs must be placed upstream of screw traps for boaters and at or on the trap to inform the public and prevent them from getting on the trap and possibly injuring themselves (Figure 19).

Because of the isolated nature of the work, trap tenders must be able to respond to emergencies. In remote locations, vehicle failure or accident, serious injury, or need for assistance can be solved with the use of proper communication devices such as a truck radio. If a truck radio is not available, then an emergency transponder such as Garmin Spot, Inreach, or Satellite phone are supplied. Vehicles and/or tagging stations are equipped with first aid kits stocked with all necessary, up-to-date supplies. The kit should be suitable for the treatment of a variety of injuries such that the patient can be stabilized until they are safely transported or until more qualified help arrives.

Figure 19. Signage at a trap. Left panel shows a warning sign for boaters. Right panel shows a sign for informing the public of trap operations.

Situational awareness is the key to accident prevention. The trap itself presents several hazards. Personnel should always be aware of moving parts: turning cone, spinning shaft, tire, and cleaning drum. Stay clear of the upstream opening of the cone while it is in operation. Do not stand on the pontoons in front of the #1 crossbeam. Never wade upstream of the trap while it is in operation. Be cautious when raising and lowering the cone; if the winch handle is let go it will "free wheel" and can break a wrist or cause other injury. Never attempt to remove slush or debris from inside the cone while it is lowered and turning. We recommend installing an access door on the cone to provide a safe option for removing debris. Never reach into the back of the cone from the trap box while it is turning or it will break an arm or worse – pinch the arm in place with no means to get free.

Weather is the main ingredient of adverse conditions. Personnel should always be aware of freezing temperatures, heavy precipitation, and water levels in early spring and late fall when icy conditions may occur. If temperatures are below freezing, then the cone and trap box may be full of slush. The pontoons could have a layer of ice on them that is not always visible (Figure 20). Surface ice may build up on the trap and stop the cone from rotating. Ice and slush can also harm fish in the trap box. Break up ice on the surface of the pontoons with a mallet. Carefully remove slush from the trap box and remove fish while taking care not to slip on the icy surface. If the cone is frozen in place, remove fish from the trap box first, then raise the cone and turn it by hand while tapping lightly on the mesh to release the slush and ice inside. Fish are removed first because slush and ice can migrate into the box and harm fish. If the cone can't be raised, then warming daytime temperatures will thaw the trap. Ice can also collect on boulders on the bottom of the river. Personnel should move slowly and carefully while walking to the trap. While on the trap, maintain hand contact with the safety line and rails to avoid slipping. If freezing conditions persist during the fall period, the trap supervisor may decide to remove the trap for the season. This event is usually predictable for a given location.

Figure 20. A trap with the box full of slush and icy pontoons.

When water levels rise, conditions can quickly become dangerous. Streams will rise gradually in the spring as snow melts. Typically, runoff from warming daytime temperatures will peak in the evening and early morning. However, a sudden increase in temperatures or rain-on-snow events can cause the water to rise as much as 20 cm overnight. Trap tenders should watch the forecasts and, in these situations, check the trap and river conditions two or three times per day. While the water is coming up, the trap can be fished to the side of the thalweg or near the bank. Move the trap to the bank to recover the fish during periods of rising water. When the water becomes turbid and large amounts of debris are present, then the cone should be raised and the trap secured to the bank until it is deemed safe to fish the trap again. After it is apparent that flows are declining, start by fishing 1.5 meters to 3.0 meters off the bank, then slowly work the trap out to the thalweg over the next week or two as water levels drop.

High flows usually move debris that will accumulate on or in the trap (Figure 21, bottom panels). Impinged debris can damage or sink a trap, putting the trap tender at risk through damage control in high water. Logs and trees or large amounts of smaller debris can jam the cone up. The trap box can become full of silt and sink. Too much force on anchors can cause them to fail, which will cause the trap to hang up and sink or float away. These can be avoided by being aware of the weather and hydrograph and securing the trap to the bank until the peak has passed (Figure 21, top panels).

If high water damages or sinks a trap, personnel must proceed safely to move and repair the trap. Large logs can be removed from the cone with the use of a come-along attached to the bank via a tree or truck. Hand saws or chainsaws may be required, taking all precautions necessary. The trap may react unexpectedly to the release of a log and should be secured to the bank with another rope or cable. If the trap is sunk in the middle of the stream and cannot be pulled to the side by hand, truck, or boom, then it will have to remain there until the water level drops to safe conditions. Repairing damage to traps caused by high water should not be attempted unless the trap can be moved to calm water and secured to the bank. Under no

circumstances should personnel attempt to wade out in high water. Help should be sought as it is unsafe to attempt these actions alone.

Figure 21. Examples of RSTs in high water. If traps aren't parked during high water (top), then bad things will happen (bottom).

Trap Maintenance

Maintenance is a key part of routine trap operations. Traps are maintained on a daily, weekly, and annual basis, depending on need. Repairs in the offseason can be costly and time consuming. Many post-season repairs can be avoided by proper attention to the traps when they are operational.

Several maintenance tasks should be completed on a daily basis. Clean debris out of the cone and trap box. Scrub algae and dirt off of the cone and pontoons with a deck brush or spray it off with a pressure washer. Brush the cleaning drum to clear the small holes of material that doesn't come off with water. Check for worn bushings and seals, and missing rivets or bolts.

Aluminum rubbing on aluminum causes the most damage to traps. Check the shaft in the flange bearing of the trap box and in front of the cone at the bipod and tighten any bolts as necessary, especially on the collars. Collar bolts are known for backing out over time, causing the cone to slip and wear into the trap box. Make sure the cleaning drum is operating correctly and the tire is properly inflated. Check that the sides of the drum are not rubbing on the trap box. Make sure the cone and pontoons are not hitting or rubbing on rocks. Check that ropes are not rubbing on cables or the trap.

Some tasks should be completed on a weekly basis. Grease the grease fittings, also known as zerks. If the u-joints are not greased regularly, they will probably need to be replaced in the off-season. Use biodegradable grease. Tighten nuts and bolts on the collars and bipod and check for rust conditions that can seize up bolts. Replace rusted bolts or spray WD-40 or other lubricant to keep threads in good condition. Inspect for worn rivets and replace as needed. Worn rivets can be drilled out using a 7/32 drill bit and replaced using a rivet gun.

Trap tenders should keep an eye for occasional repairs to be made as needed, as well as annually after the trapping season. Inspect rigging pulleys, cable clips, quick links, ropes, web straps, and shackle bolts. Replace or repair them as necessary. Pontoons should be inspected for holes on the bottom from rubbing on rocks, for worn spots from crossbeams, for broken welds, and for saturated foam inside the pontoons. Foam will need to be replaced about every five or six years. Make sure the snatch blocks are operational. Unwind the cable all the way and check for frays or worn sections. Make sure the winch works properly. Worn rivets in the cone should be replaced. Check the shaft and shaft sleeves for excess wear. Look for cracks where the cone is attached to shaft. Repair any torn screen on the cone. Check the rear assembly of the trap for a warped or dented drum, damaged tire/wheel, worn u-joints, and worn flange bearing/bushing. Worn bushings can be cut out or pounded out with a mallet and wood blocks, being careful not to break the weld on the flange. Check for tears in the trap box front side gasket. Replace the oil in the gear box. Make sure the cross beams and bipod are not bent. Check the bushings on the #2 crossbeam and replaced them if they are more than half worn. The #2 crossbeam has a tendency to wear where it rests on the pontoons, so weld more aluminum on as necessary.

Fish Processing Station

Trap operations include processing and tagging the catch, which requires a stable work station. Processing stations are best when they are a semi-permanent structure that can be left set up on location throughout the trapping season. A covered workstation is beneficial to keep fish, equipment, and trap tenders out of the weather. Work stations can be set up in a wall tent, utility trailer, camp trailer, or removable shed (Figure 22). Trap tender safety and fish health are important when setting up a work station. Select a location that is easy to access from the river and preferably in the shade. All work should take place out of direct sunlight with care to provide cool water temperatures. It is important to consider a power source in case the internal batteries of the computer and PIT tag reader are depleted. Power sources in a remote location can be an AC invertor plugged into a truck, a generator, or solar panels that charge 12-volt batteries.

Figure 22. Two examples of tagging stations. The left panel shows a wall tent with a sign for public information and a solar panel. The right panel shows a removable shed.

Work stations should be arranged to process fish efficiently (Figure 23). The proper order is the bucket containing the fish, the tub with the anesthesia, tag reader and antennae, a 300 mm rigid chrome ruler, tags and tag injectors, a balance, a container for weighing the fish, data sheets, a laptop, and preferably an external keyboard. A half section of PVC pipe epoxied to a rectangle block helps contain fish while measuring them on the ruler. Baby bread loaf pans work well for balance containers. The external keyboard keeps the laptop from getting wet. The upstream and downstream release buckets are last and must be well aerated with battery operated aerators or an air pump hooked to a power source.

Figure 23. Example of work station setup. Work flows from left to right; anesthetic tub, 300 mm rigid chrome ruler in a PVC half-pipe, PIT-tags, tag antennae and reader, balance, DNA vials, scale envelopes, laptop with external keyboard, and release buckets with aerators

PROCESSING FISH

In this section, we discuss the daily processing of the trap catch. We start with some general procedures that apply to all fish. However, target and non-target species are processed differently, so we review pertinent information on these species. A large part of the processing the target species involves fish marking or tagging and those procedures are explained. Lastly, we discuss collection of biological samples.

Sufficiently cool water temperature is a key factor in safe handling of fish. The terms and conditions of the ESA 4(d) take permit do not allow sampling or tagging of ESA-listed fish at temperatures exceeding 21°C; they may only be identified and counted under these conditions. According to the 2014 PIT Tag Marking Procedures Manual, "tagging juvenile salmonids in water temperatures >17°C is not recommended and handling juvenile salmonids in water temperatures >20°C must not occur." Therefore, to be conservative and ensure fish health, IDFG policy is to not sample or tag when water temperature is >17°C. Fish may be counted and scanned before release when temperatures are 17°C-20°C. Trap tenders should always measure water temperature before retrieving fish from the trap and adjust operations to stay within these guidelines. In the summer, it is good practice to process fish early in the morning. Further, water temperature should be taken frequently whenever fish are being processed. Methods of keeping water temperatures cool in the work station include circulating water from the river with a small pump, adding frozen water bottles to the five gallon buckets, holding the majority of the fish in live wells near the river bank, and using coolers instead of buckets.

All necessary equipment should be prepared prior to netting fish out of the trap box. The goal is to process fish efficiently and return them as soon as possible to the river. Set up the computer and open the tagging P4 software and turn on the HPR PIT tag reader before collecting fish. Recommended settings for the HPR reader are in Appendix C. Plug into the power source if needed. Scan a test PIT tag to make sure everything is working correctly. Calibrate the balance and set up the measuring ruler. Fill two five gallon buckets with fresh water for recovery and identify an upstream-release bucket and a downstream-release bucket. Five-gallon buckets with holes drilled into the top third of the bucket are helpful so that fresh water can be added and circulated through. Have aerators on buckets ready to turn on to maintain proper oxygen levels. Fill the anesthetic tub two-thirds full or to the previously marked line, so that each time the tub is filled, there will be the same amount of water. Use enough water to keep fish submerged with sufficient oxygen, but not so much that they can jump out. Cover the tub with a bucket lid or net, if necessary. Setting up the work station prior to collecting the fish from the trap minimizes time the fish are out of water.

Once the work station is prepared to receive fish, it is time to net fish from the trap box and transport them for processing. Every effort is taken to avoid injuring fish when removing them from the trap. Fish often try to hide in debris in the trap box; carefully sort through debris while netting fish. If predators (e.g., Bull Trout or Smallmouth Bass *Micropterus dolomieu*) are observed in the trap box, they should be removed first and processed separately. Separate small and large fish in different bucket. Larger fish (<200 mm) can thrash about and stress or injure smaller fish. Fish are taken from the trap in small batches of no more than 50-75 small fish (<75 mm) per five-gallon bucket. As average length increases above 75 mm, fewer fish should be put into each bucket. All fish are held in dark buckets and kept in the shade with lids. Use a lid or net over the bucket to prevent fish from jumping out while on the trap. Always put a lid on each bucket before moving fish off the trap. Aerators are used to ensure proper oxygen levels during holding and transport. If there are more fish in the trap box than will fit in two buckets, then return after the initial fish are processed. When large numbers of fish are captured, set up live wells (e.g., large

totes with holes for water flow) in the stream at a convenient, shaded location, and stage fish there until they can be transferred to the work station.

The number one priority when processing fish is keeping stress to a minimum; not only stress on the fish but also on the trap tenders. The terms and conditions of the ESA 4(d) take permit say listed fish must be processed first to minimize handling stress. Also follow the recommendations in the PIT Tag Steering Committee's 2014 PIT Tag Marking Procedures Manual, which include: monitoring water temperatures, supplying plenty of oxygen, reducing handling time, and avoiding crowding. Frequent jumping in the buckets or lethargy are signs of stress. These signs usually mean the fish are overcrowded, temperature is too high, or there is not enough oxygen in the water. When personnel detect signs of stress, corrective action must be made immediately. The fish can be put back into the trap box until the issue is diagnosed. If the issue cannot be resolved, then the fish are enumerated without further handling and released downstream of the trap.

After processing the catch, the fish must be safely transported to their respective release location. Upstream release sites are approximately 0.5 km or at least two riffles and a pool upstream of the trap; far enough to allow the marked fish to mix with unmarked fish before being recaptured. This distance will vary by location, dependent primarily on stream size. Fish should not return to the trap before dusk of the same day. If they are, then the trap supervisors should move the release location farther upstream. Only newly tagged fish intended for estimating trap efficiency are released upstream of the trap. All other fish are released downstream. Downstream release sites should be far enough from the trap so that released fish are unlikely to be caught in the trap again. Each trap has designated release sites and these need to be consistent throughout the year. All release locations should have adequate holding habitat to reduce immediate predation risk. Predatory fish may need to be transported farther downstream for release. Fish are released as soon as they are recovered from anesthetic. If large numbers of fish are being processed, trap tenders may set up live wells at the release sites. In some cases, fish may be held until dark before release.

Counting and Measuring Fish

Fish are usually anesthetized before processing to reduce stress. The Food and Drug Administration does not approve of the use of clove oil as an anesthetic for fish due to the carcinogenic nature of eugenol and methyleugenol https://www.fda.gov/media/69954/download. Tricaine Methanesulfonate (MS or MS-222) is approved by the FDA and is recommended by PIT tag Steering Committee in the 2014 PIT tag Marking Procedures Manual. Fish should not be consumed until 21 days after exposure to MS-222; therefore, it is recommended not to expose larger game fish to the anesthetizing agent.

Trap tenders should prepare an adequate supply of buffered MS-222 solution. Powdered MS-222 is diluted into a stock solution of 15 g/l with water. Sodium bicarbonate is diluted in a stock solution of 30 g/l with water. Stock solutions should be stored in dark containers to reduce exposure to light. Add approximately 15 ml of MS-222 solution and 15 ml sodium bicarbonate solution to a tub containing about 7.5 liters of water. Many trap tenders put a line in their processing tub to ensure the proper volume of water is in the tub. MS-222 and sodium bicarbonate should always be added in equal amounts. The age of the MS-222 stock solution and the amount of time it has been exposed to sunlight will change its effectiveness so the amount needed may have to be adjusted. Also note that MS-222 is more effective at higher water temperatures. Monitor the water temperature in the anesthetic tub throughout the tagging session and change

it out if necessary. In colder water (<12°C), use about 17 ml of each stock solution. In warmer water (>15°C), use about 12 ml of each solution.

To start, only put one or two fish in the tub until confident that the mixture is adequate. Anesthetize only 5-20 fish at a time, depending on size (2-5 fish for beginners). Be careful not to over anesthetize the fish. Chinook Salmon are more susceptible to the effects of MS-222 than steelhead. Fish are ready to be measured, marked, or sampled when they cannot right themselves. Add fresh water if they turn over too quickly (less than 1 min). Add a small amount (2ml-5ml) of MS-222 and sodium bicarbonate if fish do not roll over within 4-5 minutes. Under no circumstances should fish be allowed to stay in an anesthetic bath for more than ten minutes. It is important to remember that water temperature, anesthetic concentration, fish density, and size can all increase the stress load on the fish. If fish are overdosed and have a difficult time reviving, do not tag them. Instead, transfer them to fresh water, enumerate them, and release them downstream of the trap once revived.

The level of processing depends on whether the fish is one of the target species or not. Members of the target species are processed before non-target fish to minimize their time out of the river, with the exception of large predators. Depending on location, the target species is Chinook Salmon, steelhead, or both. Individuals of the target species are usually measured (lengths and weights), then marked or tagged (PIT-tagged, stained, or fin-clipped). Lengths are measured from the tip of the snout to the fork of the caudal fin to the nearest whole millimeter (fork length; Figure 24). Weights should be measured to the nearest tenth of a gram. The balance should be frequently tared to make sure weights are accurate. The water on small fish may bias their weights. If time is a consideration, weights may be omitted. All Chinook Salmon and steelhead must be enumerated in order to calculate abundance. Any information taken on target and non-target species should be recorded as described in the Fish Data Collection subsection.

Figure 24. Measuring the fork length of a juvenile steelhead in a modified PVC pipe measuring board.

Chinook Salmon and steelhead should be processed prior to non-target species. Lengths are taken on all steelhead captured as long as fish health and temperatures are acceptable. At some locations in some years, there is a large pulse of emigrants in the fall after rain events, which will strain the capacity to do full processing on every fish. There will normally only be one person working the trap but if possible supervisors should schedule two people at a trap if conditions are forecasted that will greatly boost catches. One person works up the fish needing

data taken while the other works to tally the rest and get fish out of the trap box efficiently. Tagging should be prioritized according to time and stress on the fish. It is up to the trap supervisors to determine what their limit on tagging in a day should be. Trap tenders should scan all target fish to identify recaptures, tag up to the set limit, and enumerate the rest. Tagging protocols are described in more detail in the Fish Marking and Tagging subsection.

Treatment of Chinook Salmon depends on season (Box 1). From March to May, usually few Chinook Salmon are trapped in RSTs (<200/day) but they can include two age classes: yearlings, which are smolting, and young of the year, which are newly emerged fry or young parr. These must be enumerated separately because they are from two different brood years (e.g., Figure 25, top panel). Lengths are taken on all Chinook Salmon smolts during the spring. Lengths are also required on all recaptures because trap efficiency can vary between groups and is correlated with length for Chinook Salmon <50 mm (Venditti et al. 2007). Fry can be systematically subsampled for lengths at every n^{th} fish to reduce handling. In the Pahsimeroi and Lemhi rivers, there are age-0 smolts that migrate out in May and June (Figure 25, bottom panel). As many lengths and weights as possible are to be taken on these smolts. Discriminant analyses based on fork length and date are usually necessary to differentiate among age-1 smolts, age-0 smolts, and parr. From June through August, Chinook Salmon catch increases at many traps, with a wide variation in size. During this period, lengths are taken on all Chinook Salmon large enough to be tagged and subsampled from the first 20 sub-taggable Chinook Salmon per day. Besides a few yearling Chinook Salmon, only one age class is trapped from September into December (Figure 25, top panel).

A list of non-target fish species likely to be caught at an RST is given in Table 1. All non-target fishes are enumerated. Lengths and weights are measured on all Bull Trout. Lengths and weights are measured for other salmonids up to twenty per day. Lengths only are measured on non-salmonids up to ten per day. All non-target fish are released downstream. Trap tenders should become familiar with the local fish community and document the occurrence of rare or unusual species by submitting their observations to the Idaho Fish and Wildlife Information System https://idfg.idaho.gov/species/observations.

Bull Trout require extra consideration because they are a large predatory salmonid that is protected as threatened under the ESA. All Bull Trout are scanned for PIT-tags because they may eat tagged Chinook Salmon and steelhead. If a PIT tag is detected in a Bull Trout that was implanted in a Chinook Salmon or steelhead, then enter it in the tagging file as an 11W or 32W, with a conditional comment of M RE and a text comment of RC or RE and note in the text comments or notes that it was consumed by a Bull Trout. According to IDFG's Section 6 ESA take permit, we are required to notify the USFWS about any Bull Trout mortalities on a timely basis. In the event of a Bull Trout mortality, notify the supervising biologist immediately. The supervising biologist should then notify the Regional Fisheries Manager or Nampa Research supervising staff to ensure adequate mortality take remains on the IDFG/FWS Section 6 Annual Agreement for that area. If reasonable, keep the carcass. If it is unreasonable to keep the carcass, take a fin clip (genetic sample) and record length and sex. If Bull Trout are being tagged as part of trap operations, tagging procedures described in the next section of this report should be used. However, all tagged Bull Trout should be released downstream from the trap at a separate location from juvenile salmonids.

Box 1. Life stage terminology for juvenile anadromous salmonids collected at Idaho RSTs.

The terminology used at RSTs in Idaho is somewhat different from that used in standard ichthyological and ecological texts. Biologists and trap tenders should familiarize themselves with the terminology and life stages presented here. The juvenile freshwater stages of salmon and steelhead, in sequence, are egg, alevin, fry, parr, and smolt (Moyle and Cech 1988; Webb et al. 2007; Quinn 2018). Alevins are post-hatch larvae that have a yolk sac and live inside the redd until the yolk is absorbed. The next stage are fry, which have lost or are re-absorbing the yolk sac and have left the redd. Once fry have developed vertical barring on their sides (parr marks), they are called parr. There is no set stage when fry become parr (Quinn 2018). Eventually a parr loses its parr marks as it prepares for seaward migration and becomes a smolt. The smolt transformation is a series of behavioral, morphological, and physiological alterations that change a stream-dwelling parr into a silvery smolt ready for life in the ocean. These terms are standard for anadromous salmonids in English-speaking countries.

Additional definitions and terms were developed for Chinook Salmon collected at RSTs in Idaho by the Idaho Supplementation Studies for hatchery supplementation research. These life stage terms have specific date ranges and criteria and are tabulated as follows:

Life Stage	Date Range	Brood Year	Criteria
Fry	February 1 – June 30	Current year -1	Heavy parr marks
Parr	July 1 – August 30	Current year -1	Heavy parr marks
Presmolt	September 1 – December 30	Current year -1	Parr marks
Smolt	February 1 – June 30	Current year -3 Current year -2 Current year -1	Parr marks have faded.
Yearling	July 1 – August 30	Current year -2	Larger juveniles that do not express milt.
Precocial	August – October	Current year -2 Current year -1	Young of year or yearlings that express milt during spawning period.

Brood year criteria help assign Chinook juveniles to the spawning cohort that produced them. The first four terms (fry, parr, presmolt, smolt) correspond to hatchery release types, linking natural production to hatchery supplementation. The date ranges depart from the standard terminology. For example, by the time the youngest Chinook are caught in RSTs, they have been out of the redd for several months and have parr marks. Most Chinook smolts collected in RSTs in Idaho are yearlings (brood year = current year-2) but age-0 and age-2 smolts are possible. Yearling Chinook collected after the spring exhibit two tactics; juveniles that rear in the natal stream and leave during their third spring in fresh water, and those that migrate from the natal stream and rear downstream (Kinzer et al 2010). They may be found in most locations but they seem more prevalent at higher elevations (Ryan Kinzer, Nez Perce Tribe, personal communication).The terms in this table may not transfer to other species or locations. Steelhead are much more variable in the time spent at each life stage and there is a resident form similar to precocial Chinook but older; hence, RST terminology for steelhead is similar to the standard. In western Washington, life stages are not assigned using date criteria, smolts just beginning their transformation are distinguished from advanced smolts, and fry are defined as ≤45 mm FL (Dan Rawding, Washington Department of Fish and Wildlife, personal communication). Biologists should understand how this terminology is useful or counter-productive to data analysis and interpretation.

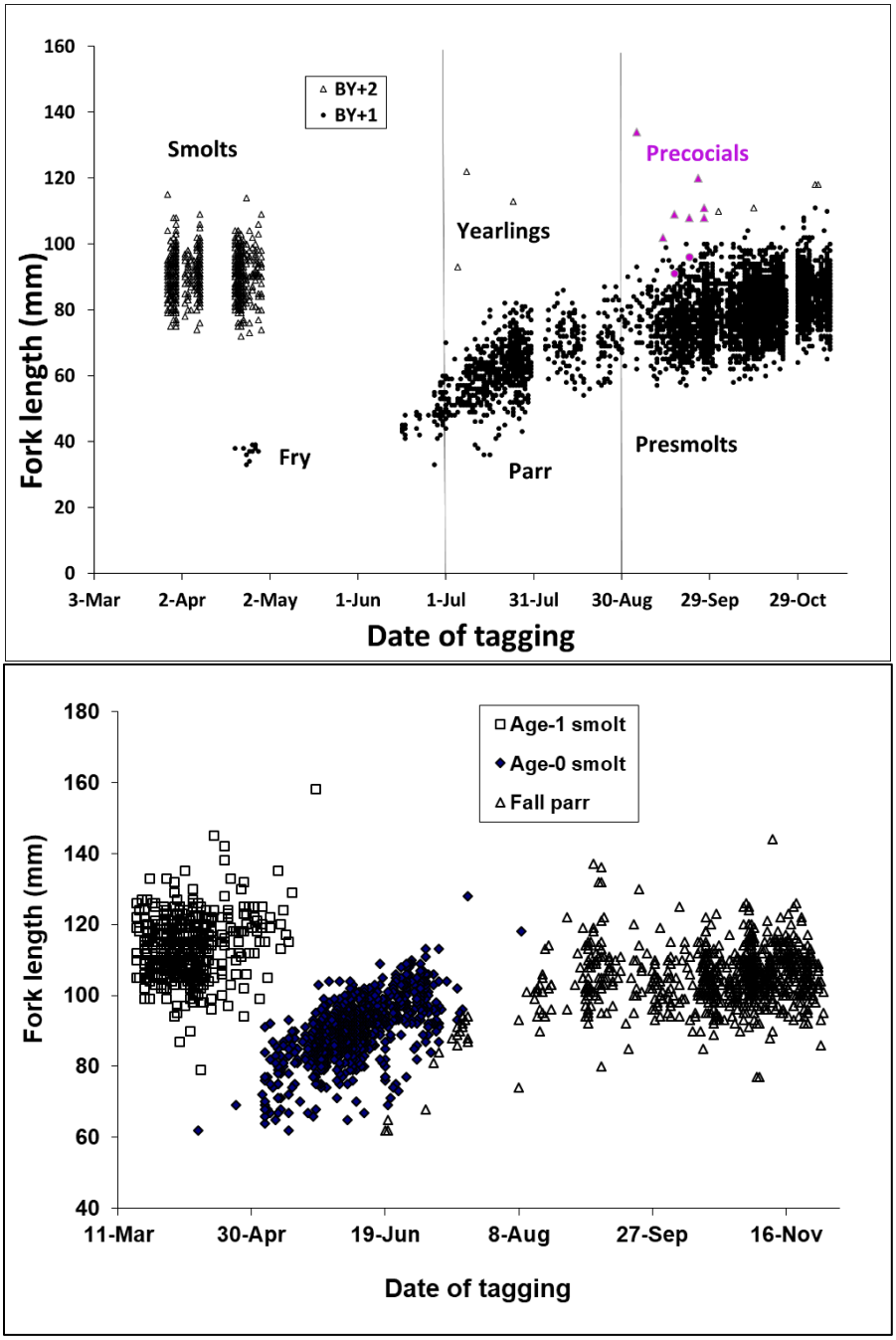


Figure 25. Patterns in Chinook Salmon emigration from the South Fork Salmon River in 2020 (top panel) and the Pahsimeroi River during 1999 (bottom panel, from Copeland and Venditti 2009). Vertical lines in top panel show date ranges for used to distinguish smolts/fry from parr and presmolts. Precocial males (pink filled symbols) are expressing milt. Bottom panel shows the difference between age-1 and age-0 smolts. Both groups arrive at Lower Granite Dam the year they are tagged.

Table 1. Common and scientific names of non-target fish species likely to be captured at one or more Idaho RSTs.

Common Name	Scientific Name
Pacific Lamprey	*Entosphenus tridentata*
Bull Trout	*Salvelinus confluentus*
Brook Trout	*Salvelinus fontinalis*
Westslope Cutthroat Trout	*Oncorhynchus clarkii lewisi,*
Mountain Whitefish	*Prosopium williamsoni*
Chiselmouth	*Acrocheilus alutaceus*
Peamouth	*Mylocheilus caurinus*
Northern Pikeminnow	*Ptychocheilus oregonensis*
Longnose Dace	*Rhinichthys cataractae*
Speckled Dace	*Rhinichthys osculus*
Redside Shiner	*Richardsonius balteatus*
Bridgelip Sucker	*Catostomus columbianus*
Largescale Sucker	*Catostomus macrocheilus*
Mottled Sculpin	*Cottus bairdii*
Paiute Sculpin	*Cottus beldingii*
Shorthead Sculpin	*Cottus confusus*
Torrent Sculpin	*Cottus rhotheus*
Smallmouth Bass	*Micropterus dolomieu*

Pacific Lamprey are also a non-target species of concern. Pacific Lamprey are identified by life stage as ammocoete (no eye) or macrophthalmia (eye present). The respective conditional comment codes in P4 are AM and MP. Lamprey take longer to anesthetize than salmonids. Immerse them in MS-222 right away and observe their behavior while working up other fish. If necessary, once the target species are removed, add 5-10 ml more MS-222 solution until they stop swimming. Placing AM and MPs in clear straws or tubes helps with taking measurements and tissue samples which minimizing handling time. Lamprey also take longer to fully recuperate from anesthesia. Give them additional time in an aerated recovery bucket and make sure they are actively swimming before releasing back into the waterbody. If possible, release them away from other fishes over a sandy bottom.

Fish Marking and Tagging

The target species are marked or tagged and released upstream on a daily basis to estimate trap efficiency. There are three types of marks or tags IDFG uses for this purpose: PIT tags, stain, or fin clips. The marked or tagged fish are released approximately 0.5 km or at least two riffles and a pool upstream of the trap to maximize the probability marked fish mix randomly with the population. We also PIT-tag fish to estimate survival to downstream locations. Fish in excess of those needed for trap efficiency trials should be released downstream of the trap.

Size is a primary consideration on which mark or tag is used (e.g., Skalski et al. 2009). Internal tags require that the animals be large enough to minimize effects on growth or survival (Pine et al. 2012). However, few studies have explicitly evaluated the minimum size for PIT tagging (Acolas et al. 2007). Current practice at most IDFG RSTs is that Chinook Salmon ≥60mm FL and steelhead ≥80 mm FL are implanted with PIT tags. Brown Trout *Salmo trutta* 41-70 mm FL that were tagged with PIT tags had 99% survival for 4 weeks and 80% tag retention with

negligible growth effects; however, survival for fish 52 mm FL was 95% and survival dropped steeply for shorter lengths (Acolas et al. 2007). Similarly, Richard et al. (2013) found variations in tag retention, survival, and growth in Brown Trout tagged by different taggers, but only if the fish were <55 mm total length. Smaller Chinook Salmon and steelhead may represent a large fraction of the total emigrants from some streams, especially those at higher elevations. In these locations, Bismarck Brown Y stain can be used to mark subsamples for mark-recapture abundance estimates (e.g., Venditti et al. 2015b). Sub-taggable steelhead and Chinook Salmon can also be marked with a caudal fin clip. Do not choose injured or compromised fish for marking or tagging because their performance will not be representative of the population. While all three methods can be applied to abundance estimates at the screw traps, only PIT-tags can be used for survival estimates to LGR and adult return (SARs). Because most of the target species are usually implanted with PIT tags and that takes most of the processing time, we begin there.

PIT tags

The first step prior to placing a PIT tag in a fish is to make sure there isn't one already implanted in the fish. All target species of taggable length should be scanned with the PIT tag reader for the presence of a previously implanted PIT tag by passing the fish through the antennae of the reader. If a tag is detected, check the fish's belly to see if the implantation scar is fresh or healed. A fresh scar indicates a recent recapture (trap efficiency recapture) and a healed or non-visible scar indicates an older non-efficiency recapture. Note that these fish are still measured but no other procedures are done. The proper coding in P4 is described in the Screw Trap Data Management document. All previously-tagged fish are released downstream of the trap.

If no tag is detected, healthy members of the target species are eligible to be tagged. Injured or unhealthy fish should be measured, their status recorded, and then released downstream. The total number of Chinook Salmon and steelhead tagged each day is determined by the supervising biologist considering water temperature, processing time, personnel, and the ratio of the number of tags available or in storage to the number of trapping days remaining in the year. The number tagged at a RST in a year is not to exceed the take given in the ESA Section 4(d) permit (see Locating and Permitting Traps subsection above).

Details of the tagging procedures can be found in the 2014 PIT Tag Procedures Manual. https://www.ptagis.org/resources/document-library/ptagis-program-documents. In general, the steps taken are as follows. Tags are inserted into the body cavity slightly offset from the midline of the belly and anterior to the pelvic fins (Figure 26). Tags are loaded into hypodermic needles, which are gently pushed into the fish until the skin is penetrated. Care needs to be taken to avoid internal organs. Do not push the needle any deeper than necessary. The tag is then injected into the body cavity. The tag site should be carefully inspected to make sure the tag does not pop back out, especially for smaller fish in which tag retention is a concern (Acolas et al. 2007). Tag retention may depend on the skill of the tagger (Skalski et al. 2009). The new tag is scanned to input the tag number into the P4 tag file, either in the needle before tagging the fish or by scanning the entire fish after the tag is implanted. Length and weight are measured as previously described. If required, a tissue or scale sample is taken (see Biological Samples subsection below). Tagged fish are then placed in the proper recovery bucket. Once the fish is in the recovery bucket, then the length, weight, and sample numbers are recorded in P4, on the paper data sheet, and on any sample envelopes. Depending on the tagger, these steps can take a few minutes. Remember that fish can't breathe out of water. It is good practice to hold the fish in the anesthetic tub for a few seconds between steps while measuring, tagging, and sampling. Fish tagged for efficiency trials are placed in the upstream recovery bucket and released in the river upstream of the trap. Recaptures, tagged fish in excess of efficiency trial needs, and untagged fish are placed in the

downstream recovery bucket and released downstream of the trap far enough to deter fish from swimming back upstream. After the tagging session and prior to release, fish are allowed to recover in large, lidded plastic boxes with sufficient free flow of water or in buckets of water with aeration and temperature control for at least ½ hour. Examples of data sheets and specific data are shown in the Fish Data Collection subsection below.

Figure 26. Proper PIT tagging procedures. Left panel shows tag implantation using a single-use injector. Right panel shows a tagged juvenile steelhead with the arrow pointing at the thin black line of a fresh tagging scar.

Several PIT tag marking procedure documents have been referred to by IDFG juvenile tagging projects over the past few decades. Kiefer and Forster (1991) provide a detailed description of the procedure. The PIT Tag Steering Committee has put out two important documents on tagging protocols. The most current documentation is the 2014 PIT Tag Marking Procedures Manual (PIT Tag Steering Committee 2014; https://ptagis.org/resources/document-library), which gives illustrated guidance on proper technique for hand tagging and tagging operations in general. The current PIT tagging codes, interrogation site configurations, naming standards, and other data specifications at https://www.ptagis.org/data/data-specification.

All PIT tags placed at RSTs are obtained by preseason request made through the PIT Tag Information System (PTAGIS). Around the middle of August each year, project leaders (point of contacts) are notified with an invitation to request tags. The email contains a current price list and instructions for ordering. Currently, the Tag Distribution Inventory web application http://www.ptagis.org/services/tag-distribution-inventory is used to provide information and make the request for tags. There is a video tutorial and instructions on how to complete requests. Additional tags are supplied by CSS for the purpose of evaluating passage in the hydrosystem. Most PIT tags are provided via coordination through the IDFG Nampa Fisheries Research office. However, projects may acquire additional tags directly. The WSS Fisheries Data Coordinator at Nampa Research maintains a PIT-tag inventory and distribution spreadsheet on the WSS intra-web collaboration site http://nr/NPM/PIT_Tags. Each year, before trapping begins, the Data Coordinator works with cooperating biologists and agencies to determine tagging needs and later to distribute the tags. Tags can be preloaded in single-use hypodermic needle injectors or kept in their shipping vials for use in multi-use needles. Single-use injectors are used at most traps instead of the traditional multi-use injectors because they significantly speed processing time

without compromising survival estimates (Venditti et al. 2013). Single-use injectors can be purchased from Biomark.

Marking Small Fish

Batch marks are most useful for marking numbers of small fish (Skalski et al. 2009; Pine et al. 2012). Staining by immersion is a commonly used technique to mark batches of fish (McFarlane et al. 1990). Fin clipping is one of the oldest methods of marking fish (Pine et al. 2012) and can also be useful for small fish. However, these mark types have limited capacity to distinguish different groups and limited durability (Skalski et al. 2009). In past years, IDFG RST operators have used Bismarck Brown 'Y' as a stain or fin clips to mark small fish. Here we discuss those methods. Note that neither mark should be used on fish <35 mm FL (Venditti et al. 2005).

If significant numbers of small fish are collected, staining is a good option. When properly stained, the mark fades after 4 days (Gaines and Martin 2004). Retention time could be adjusted by changing dye concentration and exposure time. A subsample of the total trap catch is selected for staining at intervals at least 4 days apart The process takes a little over an hour (instructions are in Appendix D). No more than 100 fish should be stained at a time per the instructions. Recaptured fish that are stained are identified visually. When stained fish have been released upstream from a trap, technicians should check all small fish in a shallow, white tub of water where stained fish are readily identifiable (Figure 27). To better detect stained fish, personnel should not remove more than twenty fish in any one net load from the bucket to check in the tub.

Figure 27. Bismarck Brown stained fry are distinguished from the other fry by their orange appearance.

If few small fish are collected, fin clips may be a better option. Fin clipping is more intrusive than staining and may affect swimming ability, so fish health should be carefully considered. However, clipped fins are detectable (with care and training) for considerably more than a month (Johnsen and Ugedal 1988; Dietrich and Cunjak 2006). Caudal and ventral fin clips have been used at IDFG RSTs in the past. Only a small portion of the caudal fin should be removed (about the size of a pencil eraser). A square notch leaves a distinct, distinguishable mark and can be applied to either the lower or upper caudal lobe (e.g., Figure 28). The left or right ventral fin may be partially clipped but the amount removed should not be so much that the fish could later be confused with hatchery fish that may be marked by complete removal of the fin. Adipose fin clips are used to identify hatchery fish and should not be used to mark at a RST. Dorsal and pectoral fins are important to stability and should not be clipped. When clipped fish have been released upstream from a trap, technicians should check the relevant fins of all small fish through the next week. Because of the durability of fin clips, marks should be rotated between release periods.

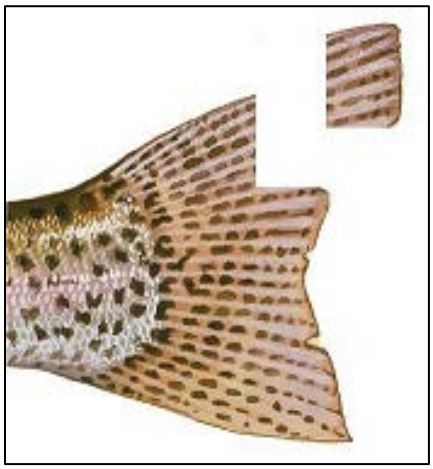


Figure 28. Example of an upper caudal fin with a square notch removed. From Apperson et al. (2015).

Biological Samples

Biological samples are often collected from Chinook Salmon, steelhead, and occasionally other species. These are usually scale samples for age analysis or tissue samples for genetic analysis. In all cases, the storage container should be properly labeled with location, species, date, and length, at a minimum. In general, samples are taken from a subsample of individuals selected for tagging.

Genetic analysis on juveniles is not standard at every RST. Directions on how many and which fish to sample should be provided by trap supervisors as needed. To collect tissue samples, use clean scissors. Remove a small portion of tissue from a fin, approximately 3 x 3 mm. Lamprey need to be fairly immobile to collect a tissue sample, otherwise there is a risk of snipping off a large part of their tail. To make handling easier, gently wrap the lamprey in a wet strip of paper towel or insert them into a clear tube, leaving the tail exposed to take the sample. Rinse and clean the scissors with water or alcohol between samples to avoid cross-contamination. Tissue samples can be stored in vials filled with pure ethanol or on Whatman sheets. If using Whatman sheets, place the tissue in the center of a grid cell printed on the sheet. For optimal adhesion, Whatman paper should be dry and the tissue should be damp. Press the tissue against the paper with

enough force to ensure contact across the whole sample. Record the fork length in the space provided in the filter paper grid cell. Record the genetic sample number in the data entry form of the P4 tag session. Keep the filter paper in the binder provided and allow it to dry. The faster the tissue dries, the better the DNA is preserved. It may be a good idea to bring the samples back to the office to dry if they are not getting dry at the trap site.

A systematic sample of scales are taken from steelhead emigrants to estimate population productivity because juveniles of different ages emigrate together. In general, scale samples are taken from PIT-tagged fish. Sample sizes needed are 150 for the spring period (until May 31st) and 150 for the summer and fall combined. Sampling is systematic (every nth fish). Trap supervisors should estimate what the trap catch will be during the year and calculate a sample rate to get at least 150 samples per season. At traps that catch <200 steelhead per season, take scales from all steelhead captured. Scale samples should be representative of the lengths of the juveniles used for abundance estimates. Trap tenders at most RSTs take scales from steelhead ≥80 mm because that is the standard lower size limit for PIT tagging. Steelhead 60-79 mm are PIT-tagged at the lower Lemhi River, Lemhi Weir, Hayden Creek, East Fork Potlatch River, and Big Bear Creek RSTs, and therefore scale sample range at those RSTs includes these fish in order to effectively estimate productivity. Trap supervisors may decide to take scales from steelhead too small to tag, in which case sample needs should be discussed with program leaders and the ageing laboratory supervisor before the trapping season.

Scale samples are collected as described in Wright et al. (2015). Take scale samples from the target area on the fish (Figure 29). Try to get 10-15 scales and spread them out as evenly as possible on a piece of Rite in the Rain paper. Put the scales on one half of the paper and fold the other half over the top of the scales (avoid putting scales in the crease of the paper). Place the Rite in the Rain paper in a labeled coin envelope. In most cases, coin envelopes will be provided with printed labels to record stream, sample number, date, species, length, and initials of the person who collected the sample.

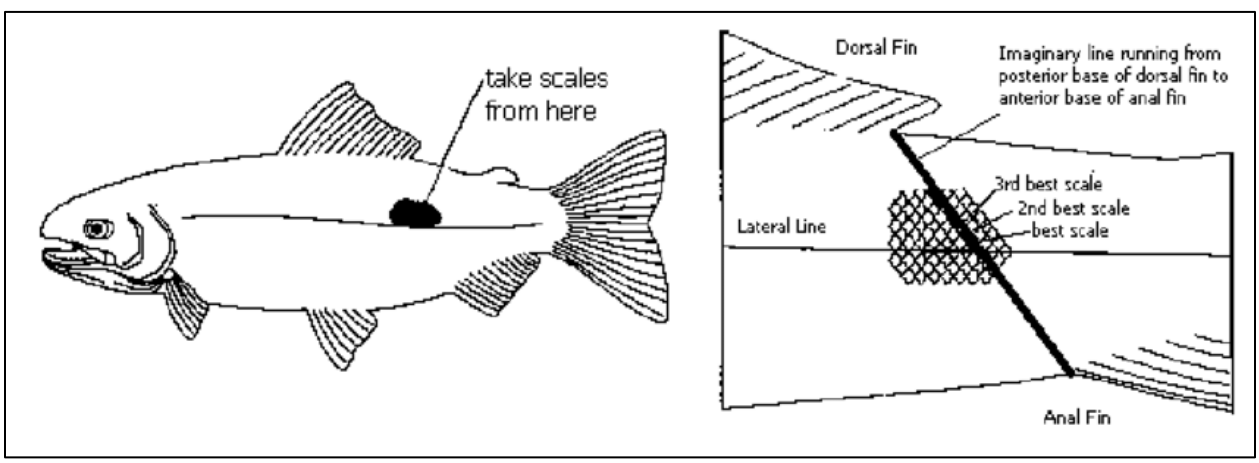
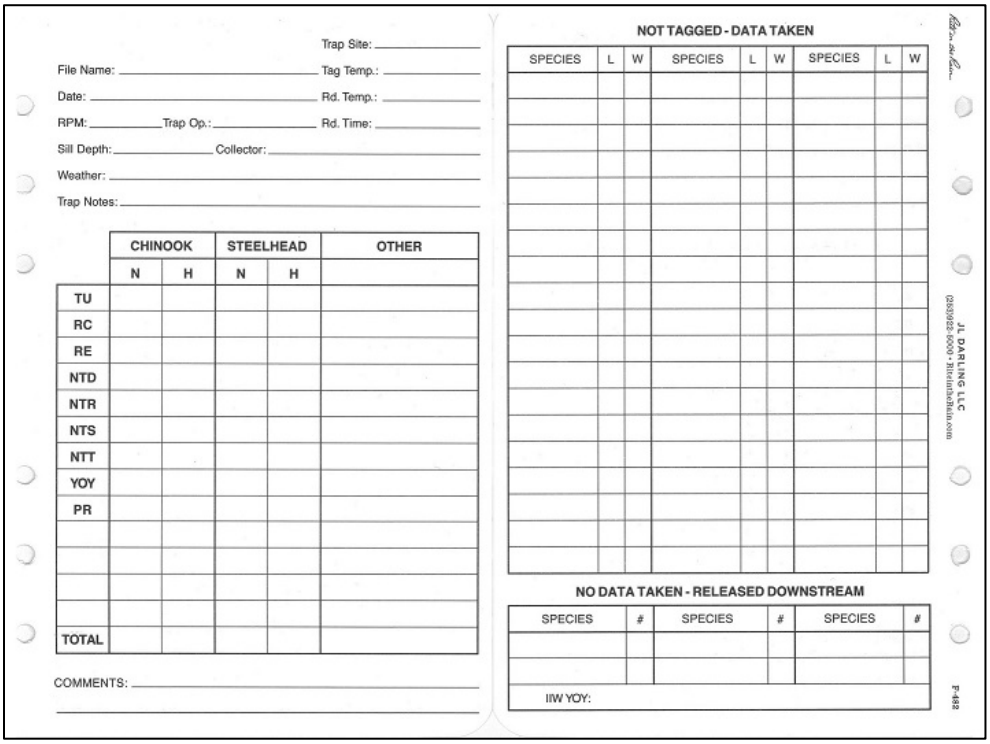


Figure 29. Preferred scale collection location for salmonids. Scales should be taken within six scales on either side of an imaginary line running from the posterior base of the dorsal fin, to the anterior base of the anal fin, and within two to three scale rows above the lateral line. Picture source: https://www2.gov.bc.ca/assets/gov/environment/natural-resource-stewardship/nr-laws-policy/risc/fishml04.pdf.

Trap Data Collection

Relevant data are recorded every day of the trapping season whether the trap was fishing or not. Recorded data include general information relative to the trap and water conditions and fish specific data (Figure 30). Trap Operation is a 0 if the cone was not turning the night before (trap pulled or parked on the bank), a 0.5 is the cone stopped turning during the night for some reason, and a 1 if the cone was turning upon arrival to the trap. The trap and water data is referred to as header information. The location is the trap code from PTAGIS as there can be more than one trap on a waterbody. The file name is the P4 session name. Date is always required. Water temperature is in degrees Celsius. The RPM (revolutions per minute) of the cone indicates how fast the water is flowing through the trap. Most locations record a water/sill depth. Tagger name (collector) is also required. Weather is optional but can be useful in stratification of the migration estimates.


Trap Site:
File Name: Tag Temp.:
Date: Rd. Temp.:
RPM: Trap Op.: Rd. Time:
Sill Depth: Collector:
Weather:
Trap Notes:

	CHINOOK		STEELHEAD		OTHER
	N	H	N	H	
TU					
RC					
RE					
NTD					
NTR					
NTS					
NTT					
YOY					
PR					
TOTAL					

COMMENTS:

NOT TAGGED - DATA TAKEN

SPECIES	L	W	SPECIES	L	W	SPECIES	L	W

NO DATA TAKEN - RELEASED DOWNSTREAM

SPECIES	#	SPECIES	#	SPECIES	#

IIW YOY:

JL DARLING LLC

F-482

Figure 30. Example of a printed daily data sheet for a rotary screw trap. Header information is on the top left. Target species are tallied in the left-hand box as natural (N) or hatchery (H) by PTAGIS disposition codes (left-hand column). Other species are tallied in the right-hand boxes by individual if length (L) and weight (W) are taken or enumerated if not (bottom).

Fish data is organized by disposition and species. With the advent of P4 and the ability to enter non-tagged fish into PTAGIS, less data needs to be recorded on paper data sheets. Each biologist determines which data should be recorded on sheets in addition to P4 summary numbers (i.e., lengths of non-tagged fish). As data is entered into P4, it is automatically saved into a database. This data is then uploaded to PTAGIS and the IDFG Idaho Fish and Wildlife Information

Systems (IFWIS). Session files and the P4 database are backed up every day to another drive. Data entry and management in P4 are covered in the next section in more detail.

ROTARY SCREW TRAP DATA MANAGEMENT

Data generated from all RST operations are stored in two databases. All data associated with trap operations, PIT-tagged fish, and non-tagged fish, are archived in the IFWIS Juvenile Fish Trapping database (JTrap, https://idfg.idaho.gov/data/fisheries/jtrap). All PIT tag data are uploaded to the PTAGIS database (https://www.ptagis.org/home). Additionally, steelhead age data are archived in the IDFG BioSamples database housed at the IDFG Nampa Fisheries Research office.

Data entry, quality control/assurance, and flow of RST data involve several steps. During trapping, data is entered on data sheets and into P4 tagging software simultaneously. From P4, data are uploaded to the PTAGIS database. Only PIT-tag data is archived in PTAGIS. Any non-tagged fish and extra operational data in P4 are filtered out of the upload to PTAGIS. This feature necessitates an IDFG database to incorporate all trapping data. Data is uploaded to IFWIS through the JTrap Uploader application. Once data is uploaded to IFWIS it can be accessed by going to the IFWIS report viewer and running a simple query.

This data management section is a brief synopsis of P4, the JTrap upload program, and instructions to access data from JTrap. An in-depth manual has been created that will walk users through each step in configuring and operating these programs as well as quality control. This Screw Trap Data Management document is posted on the WSS Collaboration website in the Screw Trapping section or on the IFWIS JTrap page.

P4 Tagging Software

The software for entering PIT-tag data to be uploaded to PTAGIS is a SQL Server database called P4. The P4 program and P4 Help file is downloaded from https://www.PTAGIS.org/software/p4. There is also a P4 Training Webinar: https://www.PTAGIS.org/support/tutorials. The P4 Help file has been placed on the WSS Collaboration website in the Screw Trapping folder. P4 allows users to collect and manage data locally before upload. P4 is flexible in that users can: 1) add non-tagged species to tag files, 2) add user-defined fields to the session and the detail records, and 3) customize the data entry layout. Disposition and species codes are standardized across IDFG RSTs using a WSS P4 configuration file (xml), and a Validation Codes file (xml) have been placed in the Screw Trapping folder. These two files can be imported into local P4 databases. P4 Session Files are to be backed up daily to another drive besides the hard drive of the field laptop, even if they have already been uploaded.

Juvenile Data Upload Program

The primary function of the JTrap Uploader program is to upload P4 data to JTrap on the IFWIS server. Secondary functions include checking recapture dates, updating/checking the Brood Year field, and providing a summary of fish caught within a specified time period. There is no data entry component to JTrap Uploader, but brood year and recapture information can be edited in it if the automated systems are inadequate. The program can be installed by clicking this link: https://fishandgame.idaho.gov/ifwis/JTrap4Installer/publish.htm

The JTrap Uploader interface is an easy way to view P4 sessions. When the program is opened, it examines the P4 database on your computer, retrieves all the session names, and displays them in a list box. All subsequent actions start by selecting one or more sessions from this list. For convenience, there are buttons to select all or none of the sessions, but in practice selecting all the sessions will likely become tedious. Once selections are made, users can choose any of the tools in the program. Uploading to IFWIS and checking recaptures requires an internet connection. Managing Brood Year and Viewing Summary do not require a connection.

Data can be accessed by going to the IFWIS report viewer. This requires a login and password from IFWIS. On the IFWIS main page, scroll down to Fisheries Management then to Juvenile Fish Trapping. User manuals are available at the bottom of the page. In the column on the right side of the page are two queries, JTrap Fish Detail and JTrap Daily Summary. The JTrap Fish Detail query will return a report view of fish along with all their values based on the criteria you choose. Users can query by date range or by brood year, by trap or by species. Any field that is left blank will return all the values for that field. After selecting the desired criteria click the Search button. The results can be exported to a csv file. Some operations data is returned with this query as well such as speed (RPMs) and sill depth. The Pivot button is non-functional for these queries. Search will get the desired results. The JTrap Daily Sum query will provide summaries of the data by date and species and disposition along with Operations data. Lengths do not return from this query. These query results can also be exported to a csv.

Quality Control

All data are checked for accuracy and completeness at several stages (e.g., trap tender prior to initial uploading, trap supervisors, IDFG database coordinators, PTAGIS database managers). Much of the data quality control is P4 specific and is covered in the Screw Trap Data Management document. Brood years and recaptures can be checked with the JTrap Uploader Program. Quality control begins when the taggers in the field check the data they just entered in P4 against the raw data sheets. At the second level, the trap supervisors check the data again (via export to a spreadsheet or in P4) and notify trap tenders of errors. Users then make any corrections and re-upload to PTAGIS and IFWIS. Uploading to PTAGIS should be expedient (as soon as possible or at the latest 3 days after collection) and in most cases will be before the trap supervisor can conduct a quality control check. Remember that only tagged fish are uploaded to PTAGIS, and if there are errors then PTAGIS will reject the session and notify the tagger. There is not an option in P4 to “Upload Correction.” The original session must be uploaded again and data overwrites in PTAGIS based on the PIT-tag code. Note that sessions will not be rejected based on mistakes with IDFG dispositions, lengths, etc. Uploading to JTrap is not as time sensitive, so data should be thoroughly checked for errors beforehand. Supervisors should check JTrap data during and at the end of the trapping season. A data tracking sheet is helpful in keeping track of which sessions have been transferred, checked for quality, uploaded to PTAGIS, uploaded to IFWIS, and backed up. A final check is made by the WSS Data Coordinator at the end of the year by querying JTrap on IFWIS and checking for logical inconsistencies in brood years, lengths, dispositions, etc.

Trap supervisors should double-check that scale sample numbers are entered correctly into P4 periodically throughout the year. Scale sample numbers are predetermined by using a naming convention in the format YY-XXXXX, two digits for the year and five for the sample number (e.g., 19-00011). The sample number must be entered into P4 precisely as it is on the envelope in order for the ages to be shown in the juvenile database. A stored procedure takes apart the ScaleFinalAge field in the Biosamples database, which is a formatted string, to get out the numeric parts and turn them into an age. The age is reflected in the JTrap Report Viewer

https://idfg.idaho.gov/data. Until the scale data has been entered into Biosamples, the report query will return an empty age. Once the final quality control checks have been made, the data are ready for analysis.

DATA ANALYSIS

Operation of a RST generates a large amount of data that can be analyzed in several different ways. In this section we describe the current practices, most of which have been stable for over a decade. Readers should note that these are the basic ways to view RST data and can be elaborated upon for more detailed investigations.

Primarily, RST data are used in a mark-recapture framework to estimate abundance and survival. Other metrics may be derived from ancillary information. Data from RSTs are used in annual reports to summarize annual trap operations, to develop length and age frequencies of the target species, and to estimate three important population metrics (abundance, survival, and productivity). Abundance is the number of fish passing the trap by species, summarized by season, calendar year, or brood year. Tagging and tag interrogation data from PTAGIS enable estimation of survival through the end of freshwater residency and exit from Idaho's rearing habitats towards the ocean. The product of survival to LGR and abundance at the trap is the number of smolts at LGR, which provides an accounting that includes rearing downstream of the RST. Productivity is the rate at which the population produces juvenile emigrants and is estimated at the RST and at LGR.

Abundance

Abundance of the target species moving past the trap is an important piece of information. The current estimation method used by IDFG is a single-trap design first pioneered by MacDonald and Smith (1980) and later modified by Steinhorst et al. (2004) to incorporate time strata for extended trapping periods. Because of differences in life history and behavior, abundance is estimated differently between the target species.

Age-specific abundances of Chinook Salmon emigrants are estimated by brood year within three seasons (see Box 1). Season designations for analysis of Chinook Salmon data follow standard calendar periods used by Venditti et al. (2015b). Spring is defined as trap deployment through June 30, a period of time dominated by catch of age-1 fish that are smolting and will be emigrating past LGR the same year. Smolts are distinguishable by their fading parr marks and deciduous scales. Age-0 Chinook Salmon are also captured in the spring; these fish are often termed 'fry' although this group includes sub-taggable parr as well as true fry (larvae with yolk sacs). Body size and overall appearance are used to distinguish cohorts (age-0 from age-1 fish) as two ages are often captured simultaneously in the spring (Figure 25, bottom panel). Note that some age-0 Chinook Salmon in the Pahsimeroi and Lemhi populations are actively smolting in the spring and are large enough to tag. Summer is defined as July 1 through August 31, a period of time when age-0 fish grow large enough to be marked with PIT tags at all locations. Chinook Salmon are often termed 'parr' during this summer period. Fall is September 1 through the end of the trapping year, a period of time when age-0 fish appear to actively emigrate out of upper tributary rearing reaches (Venditti et al. 2015b). Chinook Salmon emigrating past the trap in the fall period are traditionally termed 'presmolts'. We found that emigrants of a given cohort PIT tagged within each of these time periods generally display distinct differences in overall survival rates to LGR (Venditti et al. 2015b). Note that the terms traditionally used by IDFG for spring/summer-run Chinook Salmon at RSTs don't apply elsewhere.

Abundances of steelhead emigrants passing RSTs are estimated by two seasons. Spring is the period of time from trap installation until May 31, a period of time when most steelhead emigrants past RST are smolting and moving directly toward the ocean. We observed that age composition abruptly shifts to a younger composition at the end of the spring period (Poole et al. 2019). The summer/fall period is from June 1 to the trap pull date around November, a time period that emigrants continue to rear in freshwater for at least one more winter. Emigration typically slows in the summer then increases during the fall, but summer and fall age compositions appear to be similar. Further, summer is usually combined with fall in order to obtain sufficient recaptures for analysis. However, at low elevation locations (e.g., in the Potlatch River drainage) flows during summer and fall are insufficient to support emigration, so there may only be a spring pulse (Poole et al. 2019).

Abundance is calculated by time strata within the groupings described above. The basic procedure uses the stratified Lincoln-Petersen estimator with Bailey's modification:

$$N = \sum_{i-1}^{k} m_i \, (c_i + 1)/(r_i + 1),$$

where N is abundance of juveniles of a given life stage for Chinook Salmon or season for steelhead emigrating in a given year, i is strata (defined below for each species), c_i is the number of all unique fish captured in strata i, m_i is the number of tagged fish released in strata i, r_i is number of recaptures in strata i, and k is the total number of strata. Estimates of each life stage or season are summed for the year to estimate the total number of migrants. Assumptions are that fish are captured independently with probability p (equivalent to trap efficiency) and tagged fish mix thoroughly with untagged fish. The 95% confidence intervals are calculated using nonparametric bootstrapping and the 0.025 and the 0.975 quantiles of the bootstrap distribution (Steinhorst et al. 2004).

The time strata are established to reflect changes in trap efficiency within a season, which increases the precision of the estimate. The proportion of recaptured fish in the daily catch (R/C ratio) should be checked to look for changes signally a change in efficiency. Environmental conditions (e.g., flow and temperature) also have important effects on trap efficiency. However, strata are constructed to contain a minimum of seven recaptures to reduce the effects of small sample size. If a stratum does not contain a sufficient number of recaptures, it can be included with the previous or subsequent stratum depending on stream and trap conditions and based on the professional judgment of the biologist responsible for the RST. Estimates can still be computed for strata with less than seven recaptures, but it does need to be noted when they are with caution regarding accuracy as the confidence intervals widen with fewer recaptures.

Computation is conducted in R using an iterative maximization of the log likelihood. Estimation can be done using automated routines from the http://ifwisshiny.idfg.state.id.us:3838/JLM/IDFGStatApps/ web page or by running the R script on a personal computer. Step-by-step instructions of stratifying data to prepare them for input into the online application are posted on the WSS collaboration page. The R script is given in Appendix E. Complete cohort abundance for Chinook Salmon at the RST is calculated by bootstrapping all the strata for a brood year. Complete cohort abundance of smolts at LGR is calculated by multiplying the seasonal emigration estimates by the corresponding survival proportion estimates (RST to LGR) before summation. Details of survival estimation follow in the next section.

Survival

Survival from RST to points downstream can be estimated using the basic approach first derived by Burnham et al. (1987). These procedures require a durable mark that can be detected at multiple locations (i.e., PIT tags). Multiple recapture opportunities are necessary to statistically separate the probability of detecting a tagged individual at a location, given it was present, from the probability of surviving from tagging to that location. These data are analyzed using a Cormack-Jolly-Seber open-population mark-recapture model based on individual detection histories (Lebreton et al. 1992). The following is a brief synopsis of survival software and calculation. Step by step instructions of querying and preparing the data for input are posted on the WSS collaboration page.

The primary survival rate of concern is from RST to LGR. Estimates are made separately for the groups described in the Abundance methods because of their inherent differences in survival. Tagged fish are assumed to represent untagged fish in each group. Potential interrogation sites downstream of LGR (Little Goose, Lower Monumental, McNary, John Day, Bonneville dams and the estuary towed array) are lumped together for this analysis. The data used to calculate survival are queried from the PTAGIS database in Advanced Reporting (https://www.ptagis.org/). Tagging detail data (i.e., the tagged fish from the screw traps) and Interrogation data (i.e., the PIT tag detections at the dams on the Snake and Columbia rivers) are required. The software program PitPro (Westhagen and Skalski 2009) is used to translate raw PTAGIS PIT-tag data into usable capture histories. The PitPro program and manual can be downloaded from Columbia Basin Research (http://www.cbr.washington.edu/analysis/apps/pitpro). This program also outputs the basic point estimates of survival and detection probabilities.

A more complete treatment of the Cormack-Jolly-Seber model is implemented by the Survival Under Proportional Hazards (SURPH) program (Lady et al. 2001). The output file from PitPro, which already contains the tag identification codes and detections at each dam is loaded into SURPH. This program estimates confidence intervals about the survival and detection probabilities and provides the ability to incorporate the effects of individual and group covariates on the probabilities. Output can include diagnostic plots, survival plots, and models. The SURPH program and manual can be downloaded from http://www.cbr.washington.edu/analysis/apps/surph. Copies of the PitPro and SURPH manuals are archived on the WSS website.

Application of this model to Chinook Salmon is straightforward because of their juvenile life history and behavior. Juveniles are either tagged as yearlings or sub-yearlings and it is assumed that migration pathways to LGR by age group are consistent among individuals. Estimates are made by the seasonal and age groups described above, subject to the constraint that a sufficient number of tags in a group are detected in the hydrosystem. Current practice is to estimate 95% confidence intervals around the survival and detection probabilities using the profile likelihood option.

Steelhead exhibit two characteristics that complicate survival estimation. First, juvenile Idaho steelhead may spend more than one winter rearing downstream of the RST before becoming a smolt. Second, steelhead smolts can vary in age from age-1 up to age-7 (Peven et al. 1994) with overlapping length ranges by age making a CJS model not feasible for estimating survival. The Lowther-Skalski model is a multistate release-recapture model that allows flexibility for delayed migration and multiple tributary releases (i.e., years tagged at a RST) for a given cohort (i.e., brood year; Buchanan et al. 2015). The Basin TribPIT program and instructions

manual can be downloaded from http://www.cbr.washington.edu/analysis/apps/BasinTribPit. Two inputs are needed for the model: 1) main-stem observation history and 2) age data. For the main-stem observation history, a list of all known PIT tags implanted in juveniles at a RST across brood years is generated from PTAGIS. The PIT tag list should be uploaded to http://www.cbr.washington.edu/dart/query/pit_tagids using the Basin TribPIT "Observation File" option to generate the observation history for all PIT-tagged juveniles. For the age data, all PIT-tagged juvenile steelhead are paired with year they were tagged and age determined either from scales or assigned from the age-length-key method if scales were not sampled. The observation and age data files are loaded into the Basin TribPIT program. An example of the complete procedure is found in Chapter 2 of Feeken et al. (2020).

We need to briefly mention smolt-to-adult return rate (SAR) as a survival estimate. Survival from rearing habitats to return as a spawning adult is important metric but the previous survival procedures only cover a portion (i.e., RST to LGR). The SAR is the proportion of smolts that have returned from the ocean, often expressed as a percentage, and hence covers the subsequent portion of the life cycle. Sandford and Smith (2002) and Ryding (2006) give details on the complicated ways estimates are done for salmon and steelhead from the Snake River basin to assess the effects of the hydrosystem. One does not have to use these complicated procedures if the intent is to measure population performance. Two common approaches are (a) dividing abundance of returning adult salmon abundance by abundance of smolts from the same cohort, or (b) tagging a portion of the migrating smolts and estimating the return rate of tagged adults (Cochran et al. 2019). For the purposes of this document, a wild smolt is a juvenile that has passed LGR and is heading to the ocean. Adults could be measured at ocean exit (e.g., at Bonneville Dam), at re-entry to rearing habitats (e.g., at LGR), or even at the spawning reach (e.g., with spawning ground surveys or a PIT-detector array in the stream). Adult measurement point should depend on the question.

Productivity

Productivity estimates are derived from the primary metrics described above. Requirements include abundance by brood year and number of parents that produced each brood year. Productivity is used with abundance to evaluate population performance of ESA-listed anadromous salmonids (McElhany et al. 2000).

Adult-to-juvenile productivity for spring/summer-run Chinook Salmon is estimated at two points on the landscape. Productivity at a RST is estimated by dividing the seasonal sum of emigrants by the number of adult female spawners that produced them (i.e., by brood year). Productivity at LGR is estimated by dividing the estimate of smolts at LGR by the number of adult female spawners. The number of adult female spawners is obtained by either redd counts upstream from RST or weir counts at locations with both an RST and a weir (Stiefel et al. 2015). One female per redd is usually assumed when using redd counts (Murdoch et al. 2009).

The adult-to-juvenile productivity of steelhead at a RST is estimated by dividing the seasonal sum of cohort abundances by the number of adult female spawners that produced them. The number of adult female spawners is obtained by either PIT tag arrays or weir counts at locations with both a RST and an array or weir. Because the large majority of tagged spring emigrants are detected at LGR in the same year, age composition for spring samples is calculated separately from combined summer and fall age compositions. Scale sample age proportions are directly applied to the seasonal emigrant abundance estimates. Brood tables are constructed by summing emigrant abundances by cohort, then dividing by the number of female spawners upstream from the RST to calculate brood year productivity.

The relationship of juvenile and adult abundance in a population is quantified with a stock-recruitment analysis. Estimates of the number of redds (estimated from single or multiple pass surveys) or number of females (estimated from weir passage) above screw traps are taken as a measure of "stock" and estimated number of smolts at LGR are taken as a measure of "recruits." The stock-recruit relationship is modeled using a $\log_e$ transformed Beverton-Holt (Beverton and Holt 1957) model:

$$log_{\mathrm{e}}[R] = \log(\frac{(\log(\alpha * S))}{(1 + \beta * S)}),$$

where recruits (*R*) is a function of stock (*S*), α is the maximum recruitment rate at low spawner abundance, and β is the level of density dependence. A Bayesian hierarchical approach is used to estimate global and trap-level parameter estimates. This framework assumes parameters for groups (e.g., populations) are distributed around global or shared parameters (Gelman and Hill 2007). Analysis is conducted using the R2jags package (Su and Yajima 2015) in Program R (R Development Core Team 2017), which executes code in Program JAGS (Plummer 2003) from the program R interface.

ARCHIVING AND REPORTING

All information from RSTs must be securely stored and clearly reported. Information transfer happens in several modes to reach internal and external audiences. As information technology evolves, reporting formats have expanded. These changes have also included stable databases that can be accessed from remote locations as well as common electronic work spaces. Abundance, survival to LGR, smolt abundance at LGR, and productivity estimates are archived in the Juvenile Data spreadsheet on the WSS website. These products, along with seasonal age composition of steelhead, life stage of Lamprey captured, and RST operations are reported in annual Anadromous Emigrant Monitoring reports, which are publicly available in the IDFG technical report library. We currently report the most current estimates of adult-to-juvenile freshwater productivity for Chinook Salmon using the Beverton-Holt stock-recruit relationship and steelhead using brood tables. In addition to the results of stock-recruit analysis, we report the relationship between Chinook Salmon juvenile productivity and adult spawner abundance. Abundance at the traps and smolt equivalent abundance at LGR are uploaded to the Coordinated Assessments Data Exchange.

DISCUSSION

A properly run RST generates a powerful data series that describes the dynamics of the target population. The worth of these excellent quantitative data sets increases as they build over time, especially as they document a range in year-class strengths. This information supports several types of management programs. For example, data from RSTs showed how hatchery supplementation increased juvenile production but that the effect faded through the life cycle and did not persist after supplementation ceased (Venditti et al 2015a, 2018). Similarly, RST data enabled IDFG biologists to detect the effects of habitat restoration (Uthe et al. 2017; Copeland et al. 2021). Full development of RST data sets has greatly increased understanding of how life history variations influence population dynamics (Copeland et al. 2014; Dobos et al. 2020). The adult portion of the anadromous life cycle has long been a focus, but it has been relatively recently that corresponding details on the juvenile stages have been elucidated.

The annual patterns observed in RST data are influenced by how young salmon and steelhead move within and exit their natal reaches. Fish movement starts with initial dispersal from the redd (late winter to early spring for Chinook Salmon, Figure 15; summer for steelhead). This initial movement is usually short distance but could be much farther (Bradford and Taylor 1997). Because requirements for feeding territories in salmonids are related to length, competition will cause fish to move as they grow (Steingrimsson and Grant 2003). This phenomenon leads to self-thinning at the population scale once a critical biomass is achieved (Elliott 1993). Long, cold winters in Idaho's high country leads to a re-distribution by juvenile salmon and steelhead in anticipation of the harsh conditions (Bjornn 1971). Survival estimates must account for extensive downstream rearing and data for steelhead is usually sparse. Most spring movements are related to smoltification, the timing of which influences the likelihood of surviving in the ocean (Scheuerell et al. 2009; Haeseker et al. 2012). Steelhead in the Snake River basin may rear up to six years before smolting (Camacho et al. 2017), hence accounting for extended periods between trap and LGR is vital.

Freshwater rearing of anadromous salmonids in Idaho is spatially extensive and emigration is temporally protracted, especially for steelhead. Chinook Salmon and steelhead may rear from headwater spawning areas to the lower Snake River throughout the year. Cohorts of Chinook Salmon are relatively easy to distinguish by length and time, with a few exceptions (e.g., Pahsimeroi River, where a significant proportion of age-0 emigrants smolt; Copeland and Venditti 2009; Figure 25, bottom panel). However, extensive ageing of steelhead emigrants is necessary to estimate population productivity because several cohorts emigrate together (Dobos et al. 2020). These characteristics mean that, to collect reliable information, RSTs must be run through the year as long as is feasible.

Trapping wild juvenile anadromous salmonids in Idaho and collecting reliable data has always been challenging. Traps are often in remote locations and must operate for a long field season that includes snow melt and leaf fall. Keating's (1958) lament should sound familiar to anyone supervising a trap operation today: "Ice filled and plugged the traps during late fall, winter, and spring. Ice and debris caused the V-traps to wash out, the floating traps to sink, and the spill traps to overflow." However, now we have greater logistical support to do the field work; a durable, individually-identifiable tag; more sophisticated data storage and manipulation capabilities; and increasingly complicated statistical models to deal with the imperfections in trapping data.

Conditions at IDFG RSTs can vary tremendously, given the large and diverse area in which they are situated. Trap elevations range from less than 400 m above sea level in Big Bear Creek to almost 2000 m at sites in Marsh Creek and the headwaters of the Salmon River. Similarly, distances from trap to LGR range from 99 rkm in Big Bear Creek to 1442 rkm in the Salmon River headwaters. Streams at low elevations may not have summer or fall flows to support significant fish movements. The Pahsimeroi and Lemhi rivers have significant groundwater inputs modified by irrigation withdrawals that lead to relatively moderate flow variations and water temperatures. Stream hydrology at other locations are driven mostly by snowmelt with spates in the fall from rainfall events. These varying conditions lead to differences among locations in the life histories and movements of the target species, such as steelhead age structure or large fall emigration pulses at higher elevations. Even geographically proximate RSTs may have operational differences. For example, in Big Bear Creek, flows in the spring can be very flashy and then quickly drop to limited flow by June; whereas at the trap on the East Fork Potlatch River, the surrounding forests moderate and extend spring flows. Similarly, the traps on Hayden Creek and the upper Lemhi River are less than 1 km apart but the latter sits downstream from a groundwater-driven valley reach, whereas the other is on a stream that drains the east side of the

Lemhi Range and therefore gets most of its flow from snowmelt. These points illustrate how trap management will differ to some extent depending on the hydrograph at each location.

Trap operations also vary from year to year. Stream flows fluctuate annually, with high peak flows often associated with large snow packs. Rains may mobilize debris and fall precipitation often keys pulses of fish movement. On the other hand, drought reduces stream flow, potentially to the point where trap cones do not turn. Numbers of young salmon and steelhead available for trapping depends on spawner abundance. The interaction of cohort density and habitat volume leads to changing patterns of trap catches among years. These points illustrate how effective trap management needs to adjust for changing conditions.

Variations among traps and among years can present challenges for data analysis and interpretation. The basic analytical tool for RST data is a mark-recapture model structured to output abundance or survival. These models are made possible by a series of simplifying assumptions that can be sensitive to changing conditions. We discuss these assumptions next.

Rotary Screw Traps and Assumptions of Mark-Recapture Models

All mark-recapture studies have simplifying assumptions. The following section is chiefly derived from the treatises by Otis et al. (1978) and Pollock et al. (1990). This discussion focuses on mark-recapture analysis as modified to fit RST trapping but also applies to survival estimates. There are three assumptions for deriving abundance: 1) the population is closed; 2) all individuals are equally vulnerable to capture; 3) all marked animals are identified. For survival estimates, there is a fourth assumption: 4) all marked animals have equal probability of survival. Biologists must understand how these assumptions affect interpretation in order to make sound decisions about trap operations and data analysis. Probability of assumption violations can be minimized by training and study design. Standardized methods will help reduce assumption violations (e.g., tagging protocols and choosing only healthy fish for marking). Departures can be detected by appropriate investigations. If necessary, ancillary information can be used to statistically adjust estimates (e.g., covariates, stratification). We discuss how mark-recapture assumptions apply to Idaho RSTs and what relevant work has been done by IDFG in the past.

Population Closure

The most basic assumption for most abundance estimates is that there are no additions or losses to the population between the capture and recapture efforts. If there are additions, then the estimate applies to the population at the second sample. Losses will result in bias unless they are equally likely between marked and unmarked animals. If marked animals die at a higher rate, a serious upward bias in $\widehat{N}$ could result. For RSTs, we assume individuals move only downstream and that marked fish don't die before passing the trap site again. This assumption implies that the abundance estimate does not include any fish upstream from the trap (i.e., it is a measure of the number passing the trap location). It also implies that recapture is instantaneous relative to the trapping time strata (i.e., marked fish pass the trap again within five days). If movement of marked fish is delayed relative to the length of a time stratum, the trap efficiency estimate will be biased low because not all marked fish are available for recapture during the stratum.

The closure assumption can be shown to be satisfied by a number of factors or addressed by several actions. Traps operate over most of the ice-free portion of the year and hence sample from the vast majority of emigrants. We assume a negligible amount of winter movement or alternatively that fish moving in winter likely don't survive. Porter et al. (2019) showed most Chinook Salmon juveniles settle for winter by late November when water temperatures drop to

4°C. However, Bjornn (1978, Figure 16) and Achord et al. (2012) demonstrated that some fish move in the winter and can survive to emigration. Similarly, we assume that few fish move when flows are too low to turn RST cones. We account for some level of predation on marked fish in trap boxes by scanning captured predators. At some RSTs, brush is added to the trap box to give smaller fish shelter. Scanning predators for ingested PIT tags means these tags can be censored from survival estimates. Predation on marked fish can happen after release as well; therefore, release sites need to be at sheltered locations and should be observed for predation potential. Marked fish need to be fully recovered from the anesthetic before release. Release sites should be located to ensure adequate mixing by tagged fish but not so far upstream that mortality can happen. All trap supervisors should evaluate their release sites at regular intervals. Missing trap days results in a downward bias, which can be removed analytically (e.g., Bonner and Schwarz 2011; Oldemeyer et al 2018). The magnitude of delayed movements (e.g., presence of PIT-tagged fish with healed scars) can be easily evaluated. If necessary, a movement parameter can be added to the models (e.g., Mäntyniemi and Romakkaniemi 2002; Bonner and Schwarz 2011) to adjust for the proportion of fish delaying.

The closure assumption is relaxed for open-population mark-recapture models. These models are capable of estimating additions and losses between samples as well as abundance. The Cormack-Jolly-Seber model simplifies this situation by focusing on survival (i.e., losses) with probability of capture estimated as a nuisance parameter. In the case of survival from RST to LGR, the closure assumption is that all tagged fish are attempting to emigrate past LGR. This could be violated by *O. mykiss* adopting a fluvial life history or precocial male Chinook Salmon that will return to their natal reach. These fish may mature and spawn but will be accounted as losses to the population.

Equal Capture Probability

Captured and marked fish must represent the population of interest in terms of vulnerability to the capture gear for proper statistical inference. This assumption has several implications. Caught fish should have the same capture probability as uncaught individuals. Marked fish should be recaptured with same probability as their initial capture. As a corollary, we assume marked fish recover from handling and mix with the rest of the population before being available for recapture. Further, we assume that the behavior of marked fish is not different than unmarked fish (e.g., trap shyness or higher propensity to get trapped a second time). Violations can be related to heterogeneity in capture probability caused by differences in size, age, and sex, or by changes in behavior related to being caught and tagged. Consistent capture heterogeneity will cause bias. For example, if smaller fish are more likely than larger fish to be caught and marked, then they will have a higher likelihood of being recaptured, resulting in a higher estimated efficiency and a downwardly-biased abundance estimate. Survival estimates tend to be robust to violations of the equal capture assumption; abundance estimates are likely to be biased if the differences are persistent (Pollock et al. 1990).

The equal capture assumption can be shown to be satisfied by a number of factors or addressed by several actions. For RSTs, we assume that changes in capture probability are coincident with time strata breaks but should be similar within strata. We know trap efficiency changes with season and flow, so we stratify by time to capture this variability, as explained in Data Analysis. Stratification also helps as fish behavior changes with ontogeny. Fish behavior should not be greatly affected if they are released after a sufficient recovery time. Length frequencies of tagged fish should be comparable to that of the total catch and of recaptures to all tagged fish. For survival estimates, steelhead passing the trap may reach LGR in different years

with different flow characteristics and spill regimes, changing the probability of being detected at LGR. The latter is not an issue for Chinook Salmon.

Mark Identification

All marked-recapture models assume that marks are never lost or misreported. Lost or overlooked marks will bias trap efficiency downwards, inflating the abundance estimate. Loss or malfunction of PIT tags will obviously reduce estimated survival. Likelihood of violations is reduced if samples are instantaneous (i.e., daily) and release is immediate. Trap tenders should check transport buckets and holding cages for lost tags. Tag shedding usually occurs within 24 h and is affected by tagger experience (Dare 2003; Meyer et al. 2011). Training personnel in marking and tagging techniques is already standard practice to assure data quality. Tag loss can be estimated by recording the presence of tagging scars to corresponding PIT detections. This approach is a double marking study in the sense that the scar is the primary mark and the PIT tag is secondary. Abundance and survival estimates can then be adjusted if loss rate is known.

Equal Survival Probability

This assumption is similar to the equal capture assumption but applies only to survival estimates. All marked animals must have same probability to reach downstream detectors. This assumption is violated frequently by steelhead behavior, which can include multiple winters of rearing downstream of the trap (Dobos et al. 2020). The effects of assumption violations could also be complicated by size selective mortality in Chinook Salmon if tagging is not representative of the population length distribution (e.g., many fish are too small to PIT tag). Effects of assumption violations can be reduced by stratifying survival by seasons or life stages or incorporating covariates into the analysis. This issue could be explored by checking if survival is affected by day of tagging within season (survival thresholds).

Assumption Testing at Idaho Rotary Screw Traps

Mark-recapture assumptions need to be regularly assessed as part of trap operation and data analysis. These assessments do not always need to be a formal experiment or statistical test. Nonetheless, tests of assumptions should be made at regular intervals to evaluate tagging and analytical strategy. In the past, IDFG investigators conducted several tests with Chinook Salmon released at RSTs; however, no formal tests have been completed on steelhead in Idaho. Here we review some of the relevant findings.

A significant number of Chinook Salmon too small to PIT tag are captured at some RSTs, sparking concerns that PIT-tagged fish might not represent the population. Venditti et al. (2007) compared capture efficiencies of stained Chinook Salmon <60 mm and ≥60 mm to PIT-tagged fish ≥60 mm in three streams. Differences depended on season and stream. Venditti et al. (2007) concluded the disparity between recapture rates for the two groups indicated that the PIT-tagged groups were not representative of the entire emigrant population moving past the traps. However, they also observed that when most of the sub-taggable sized fish were between 50 and 60 mm FL, the PIT-tagged group was representative of the entire population moving past the trap. This topic bears further investigation, especially for steelhead.

There are several considerations concerning small fish relevant to the assumptions of a mark-recapture model (Skalski et al. 2009). Early in the summer, lower trap efficiencies and small sizes are confounded (at least for Chinook Salmon); hence, it may be difficult to collect enough small fish to generate enough recaptures needed for precise and unbiased abundance estimates.

Very small fish (i.e., swim-up fry) may be searching for shelter and not exhibiting sustained downstream movements. If they settle into a habitat before passing the trap again, that would generate a violation of the closure assumption. If stained fish move slowly, Bismarck brown stains may fade before they pass the trap, violating the mark identification assumption. To investigate the relationship of trap efficiency and length, trap operators should test combinations of marks and staining. With the fin clips described under Marking Small Fish, there are four mark types; adding a stain-fin clip combination increases this to nine (Table 2). Avoid clipping multiple fins, which may compromise survival (Johnsen and Ugedal 1988). To investigate the possibility that small fish are not making sustained downstream movements, compare recapture rates between two release locations at different distances upstream.

Table 2. Marks and combination marks for fish too small to PIT tag. All fin clips are partial, see Figure 28 for an example.

Single Marks	Mark Combinations
Left ventral clip	Stain and left ventral clip
Right ventral clip	Stain and right ventral clip
Upper caudal clip	Stain and upper caudal clip
Lower caudal clip	Stain and lower caudal clip
Stain only	

Tag burden is an important consideration. The use of 12 mm X 2.05 mm PIT tags has been standard since screw trapping began. In recent years, smaller tags have become available (8.5 mm X 2.05 mm and 10 mm X 1.41 mm). Recently, IDFG biologists tested the efficacy of implanting Chinook Salmon juveniles with smaller PIT tags in order to determine differences in detection and survival rates for Chinook Salmon with FL 60 mm-70 mm (Poole et al. 2019). Overall, the detection probability and survival of Chinook Salmon was not greatly affected by the size of PIT tag used. We do not believe that tagging smaller Chinook Salmon with either type of smaller PIT tag will have a population level effect, and thus, recommend continuing use of the standard 12 mm tag for all Chinook Salmon over 60 mm.

Release timing and release site distance from the RST has also been examined. Venditti et al. (2008) observed that, at some locations, PIT-tagged fish were recaptured at different times of the day than the run-at-large. In response, they evaluated the effect of release time (day or night) and two upstream distances on the trapping efficiency of juvenile Chinook Salmon at several traps. Additionally, trap operators checked trap boxes several times daily during daylight hours to determine if there were obvious differences in the time of capture for the two groups of fish. Release distance was varied above the South Fork Salmon River trap at Knox Bridge (150 and 500 m). Releases occurred at mid-day versus dusk at the upper Salmon River, Pahsimeroi River, and Crooked Fork Creek traps. Differences in time of release were not significant for any comparison. Release distances should be tested for Chinook Salmon and steelhead at traps that have new locations or traps that have not done so in the past.

Guidelines for Tagging at Traps

An important facet of trap management is placement of the marks and tags needed to estimate the primary metrics. Three proportions of interest can be directly estimated based on marks and tags placed at RSTs: trap efficiency, survival from trap to LGR, and SAR. The ability to estimate the three proportions is controlled by the number of recaptures. Number of recaptures is determined by three things: the probability of recapture/detection given the fish survive, the number of tags released, and random chance, the effects of which can be reduced by releasing more tags. For a given number of detections, precision of a proportion will decrease as the estimate nears 0.5 and increase as an estimate approaches 0 or 1 (Zar 1999). Readers should be aware of this property; however, we focus on number of recaptured fish and assume that the minimum goals will generate estimates precise enough for most applications. Desired levels of tagging goals for Chinook Salmon in Idaho were originally developed by Bowles and Leitzinger (1991). Here we review and update their process.

The desired number of tags placed to estimate trap efficiency is controlled by the number needed for statistical estimation of abundance with desired precision and the concurrent efficiency of the trap. The recommended minimum number of recaptures per stratum is seven (Roper and Scarnecchia 2000; Steinhorst et al. 2004). Dividing this number by the expected range of trap efficiencies gives guidance for the target for daily release of tags upstream (Box 2). For example, to estimate a daily efficiency when the expected rate is 2%, at least 350 tags should be released upstream. However, it is not usually desirable or possible to have daily strata due to low sample sizes. If the desired length of a time stratum is a week, then at least 50 tags should be released upstream each day when expected efficiency is 2%. Note this process applies to any of the three mark types discussed above (stain, fin clip, PIT tag). If efficiency is low but daily catch is highly variable, then increasing the number of tagged fish released upstream above the minimum (e.g., to 100) accomplishes a few things: 1) it will ensure enough recaptures within a strata, 2) it will provide enough recaptures to combine with surrounding strata with fewer recaptures, and 3) it will slightly narrow the intervals around the abundance estimate.

Box 2. Calculating the minimum number of tags to release for estimating trap efficiency. To get the minimum number of tags to release per day, divide desired number of recaptures by the expected trap efficiency during the stratum. The equation is

$$R = n_{min}/(p * d),$$

where *R* is the daily number of tags to release upstream, n_{min} is the desired minimum number of recaptures per stratum, *p* is the expected trap efficiency during the stratum, and *d* is the number of days in the stratum. In this example, we assume two levels of stratification: daily and weekly. In practice, strata lengths will be weekly or greater. Lutch et al. (2003) reported trap efficiencies for Chinook Salmon ranging from 4% to 35%. For simplicity's sake, we will start with a lower bound of 1%. The range of tags to release daily is tabulated as follows:

	Number of tags to release each day to get 7 recaptures	
Trap Efficiency	**Daily stratum**	**Weekly stratum**
1%	700	100
2%	350	50
5%	140	20
10%	70	10
25%	28	4
35%	20	3

If trap efficiency is 1%, 7 recaptures/0.01=700 tags. That would be a lot of fish to tag in a day but consider that the stratum is likely a week long at least. In that case, 700 tags/7 days= 100 tags released per day.

The number of tags that can be released is, of course, controlled by how many fish are caught. Hence, low catches typical of certain times of the year may be limiting. Usually, trap catches of Chinook Salmon are low in the spring but increase through the summer; therefore, all Chinook Salmon yearlings collected in the spring are tagged. Catches of steelhead are often low throughout the year; therefore, all steelhead caught usually are tagged. In the fall, the number of fish tagged per day should not exceed the ratio of the number of tags on hand to the number of days left in the trapping year. A balance needs to be met between minimizing handling time of listed species while also putting out enough tags to estimate the primary metrics. Supervisors of traps that catch large numbers of fish and have high enough efficiencies may reduce the number of tagged fish released upstream, with the remainder released downstream, to minimize handling of recaptures.

Survival from trap to LGR is typically estimated on a seasonal basis (spring, summer, and fall). Regression analysts commonly use a rule of thumb that there should be at least 10 observations for every parameter to be estimated (e.g., Harrell et al. 1996). In this case the CJS model is simultaneously estimating survival and detection probabilities, i.e., at least 20 detections are necessary per seasonal release group. Bowles and Leitzinger (1991) recommended tagging targets to obtain reliable estimates of survival for Chinook Salmon. Their goal was to achieve at least 35 subsequent detections, given a range of season-specific survivals. The recommended targets were 500 for summer parr tagged upstream of the trap, 300 for fall fish caught at the trap, and 100 for spring fish caught at the trap. We made calculations based on more recent data and aiming for 20 or 35 detections (Box 2). The results were similar to Bowles and Leitzinger's (1991) recommendations. Note we focused on worst case scenarios (low survival and low detection probability). These are minimums for a group survival rate; if there are subdivisions within season (e.g., steelhead by cohort), more tags will be required to support estimation of the additional parameters. However, increased detection capability from the new spillway detector at LGR will shrink the necessary minimum sample sizes or, alternatively, increase estimated precision given the same number of tags.

Box 3. Calculating the minimum number of tags needed to estimate survival from trap to Lower Granite Dam by season of release.

Estimating survival to points downstream of the trap is similar to the previous calculation with two changes. First, we are estimating survival by season, therefore the desired quantity is the number of tags to release by season. Second, one of the assumptions of the efficiency calculation is that there is no mortality prior to tagged fish passing the detection site; therefore, we need to specify the expected loss rate. The equation is then

$$R = n_{min}/(p_{total} * s),$$

where R is the number of tags to release per season, n_{min} is the desired minimum number of recaptures per stratum, p_{total} is the expected probability of detection in the hydrosystem, and s is the expected survival to LGR. There are multiple locations where fish could be detected; most detections are at three dams in the lower Snake River (Lower Granite, Little Goose, and Lower Monumental dams). The probability of being detected in at least one of the three dams is 1 minus the product of the probability of being missed at each. We will assume detection probabilities are similar at each dam, so $p_{total} = 1 - (1 - p_{lgr})^3$. A high-flow year (2010) had average by-pass efficiency of 25% at LGR for steelhead and yearling Chinook smolts (Copeland et al. 2013). In this scenario 58% of the tags in the hydrosystem should be detected at least once $(1-(1-0.25)^3)$. Recent minimum survivals by season (Feeken et al. 2020) for Chinook Salmon were 14%, 14%, and 41% for the summer, fall, and spring periods, respectively. To achieve 20 detections under these conditions, there should be 84 tags released in the spring and 148 tags released in the each of the other seasons. To achieve 35 detections, the calculations yield 247 spring tags and 432 tags in the other seasons.

Smolt-to-adult return is estimated on a migratory year basis. Copeland et al. (2014) computed the number of PIT tags placed in steelhead at the Fish Creek screw trap necessary to get a reliable SAR, which they defined as 10 adult detections. Regression analysts commonly use a rule of thumb that there should be at least 10 observations for every parameter to be estimated (e.g., Harrell et al. 1996). Detection efficiencies in adult ladders at Columbia and Snake River dams approaches 100% (Tenney et al. 2017), so a target of 10 detections is reasonable. Bowles and Leitzinger (1991) estimated that, to achieve 30 adult detections with survivals ranging 0.2%-0.4% from emigration to adult return to the stream, 7,500 to 15,000 tags would need to be placed. We calculated the number of tags based on four scenarios with a range of expected SARs (Box 3). The results are highly influenced by SAR level but are achievable in the more optimistic scenarios, especially if the LGR spillway detector is effective at generating higher detection rates in the hydrosystem. In some cases, it may be justifiable to group tags placed at different traps to compute SARs, for example, traps within the same population (e.g., Big Bear Creek and East Fork Potlatch River traps). Precision in SAR estimates increases with good ocean survival. Although precision is low at low SAR rates, we can be confident that the true survival after passing LGR is poor.

Box 4. Tags needed per migratory year to estimate SAR.

The principles for getting the desired number of tags for estimating SAR are the same with an additional survival component. The calculation hinges on the probability of tags from a trap being detected as smolts in the hydrosystem and the expected range of SAR values. The equation is then

$$R = \frac{n_{min}}{p_{total} * s * SAR}.$$

For this example, we used four juvenile detection and survival scenarios at three SAR levels to estimate the number of tagged juveniles necessary to get 10 or 30 adult detections. Low detection probability was from the previous example (25% bypass efficiency). We also assumed the spillway detector would increase LGR detection to 66%, while detection rates at the other dams remained 25%. Juvenile survival was either 0.14 or 0.63; the latter is the median survival for juvenile Chinook tagged in the spring from Feeken et al (2020). The results are tabulated as follows:

Juvenile detection and survival scenario	SAR levels to get 10 or 30 adult detections		
	0.5%	**1%**	**2%**
Low detection, low survival	24,710 / 74,131	12,355 / 37,066	6,178 / 18,533
LGR spillway detector, low survival	17,542 / 52,626	8,771 / 26,313	4,385 / 13,156
Low detection, high survival	5,491 / 16,474	2,746 / 8,237	1,373 / 4,118
LGR spillway detector, high survival	3,898 / 11,695	1,949 / 5,847	975 / 2,924

Expected juvenile survival and SAR had more important effects than increased detection capability at LGR.

Recommen dations for the Future

Changes to analytical methods for anadromous emigrants over time have been enabled by the flexible trapping platform, the durable primary mark (PIT tag), increased detection capability, and greater computing power. Newer statistical tools allow biases to be countered and more information to be distilled from the data. With the advent of main-stem spillway detectors and instream antenna arrays, there is an opportunity to provide greater understanding throughout the life cycle (e.g., location-specific SARs and adult return to spawning reaches). The elements of life history diversity can then be linked to population dynamics such that the effects of management actions can be evaluated directly at the population level. We have three programmatic recommendations to improve operation of RSTs and analysis of their data. These are: 1) incorporate testing of mark-recapture assumptions as a regular part of trap operations and data analyses; 2) periodically revisit tagging methodology strategy for each trap and across the fleet of RSTs; and 3) develop new opportunities for data analysis. These programmatic recommendations imply several specific recommendations which we explain further.

Consideration of mark-recapture assumptions should be a regular part of trap operation and data analysis (Roper and Scarnecchia 2000). Some assumptions may need annual checking but all should be re-visited at regular intervals. To date, no assumptions have been tested for steelhead at IDFG's RSTs and it has been many years since assumptions have been checked for Chinook Salmon. Size selectivity by RSTs is likely a problem for steelhead (Roper and Scarnecchia 2000; Tattam et al. 2013). Retention of PIT tags is a concern for smaller fish. Distance of releases upstream of the RST location should be evaluated, especially for newer traps. Trap supervisors should examine survival period breaks (i.e., between spring and summer and between summer and fall) by trap and species to investigate the suitability of temporal strata for survival estimates. If violations are suspected or detected, assumptions can be relaxed or addressed analytically by the incorporation of covariates. However, adding covariates to the analysis must be accompanied by enough observations to support the estimation of the extra parameters.

Tagging goals to estimate trap efficiency, survival to LGR, and SAR should be assessed for each RST. In the previous subsection, we walked through some hypothetical examples with summary numbers pulled from recent reports. The biologists supervising RSTs need to do those computations for themselves and manage their traps accordingly. Developing tagging goals to support SAR estimation will take creativity and a careful examination of the existing record of adult detections. On a broader level, it is not clear how representative the tags placed annually at RSTs are of Idaho's wild salmon and steelhead populations outside of the study watersheds. There should be an assessment of tags placed and detected relative to spatial distribution of abundance (e.g., adults spawning or smolts by stock).

Lastly, future analyses of RST data should use a more synthetic analytical framework than the current single-year approach. Conceptually, the emigration of juveniles is a process that repeats every year and is similar among populations in Idaho. Therefore, the annual emigration of juveniles is treated as a universally recurring process and each annual data set is a single realization of this process. In other words, compared to more distant populations, there are more similarities than differences in the emigration of anadromous salmonids in Idaho. Given a flexible modelling framework (e.g., Bayesian hierarchical method; Royle and Dorazio 2008), RST data can be aggregated to harness the information where data are strong to increase knowledge where the data are sparse. Recent analytical advances provide the basis for addressing estimation problems using existing data sets. Smoothing models allow for estimation across discontinuities in trap operations but with the untestable assumption that patterns in the observed data continue

over the missing strata (Bonner and Schwarz 2011). This assumption can be avoided by aggregating data to improve reliability of model selection and increase accuracy of abundance estimates for each original sampling unit (Litt and Steidl 2009). Oldemeyer et al. (2018) provided an example for aggregating across years within a site. Data can also be aggregated among sites or between species at a site. Analyses could be extended through later life stages using PIT detections. This framework also allows for explicit model-based inferences to support habitat restoration (McHugh et al 2017), integrated hatchery evaluations (Venditti et al. 2018), and harvest impacts (McCormick et al. 2021), to name a few examples.

In closing, IDFG has extensive experience in monitoring emigrating anadromous salmon and steelhead. This history has yielded valuable knowledge for assessment of population status in a life-history framework and for management evaluations. In this report, we described the current methodology and made recommendations to improve field operations and data analysis. Adopting these recommendations will increase the flow of useful information and allow IDFG's anadromous fisheries management program to meet new challenges in the future.

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Appendix A. Photographs of emigrant traps currently operated by IDFG for anadromous salmonids. Note that all but three are rotary screw traps.

Big Bear Creek

East Fork Potlatch River

Crooked River

Fish Creek

Lochsa River

Snake River

Appendix A. Continued

Salmon River

Rapid River

South Fork Salmon River

Big Creek

Marsh Creek

North Fork Salmon River

Appendix A. Continued

Upper Lemhi River

Lower Lemhi River

Hayden Creek

Pahsimeroi River

Upper Salmon River

Redfish Lake Creek

Appendix B. Screw trap assembly instructions.

Screw Trap Assembly Instructions

E.G. Solutions, Inc. - www.screwtraps.com

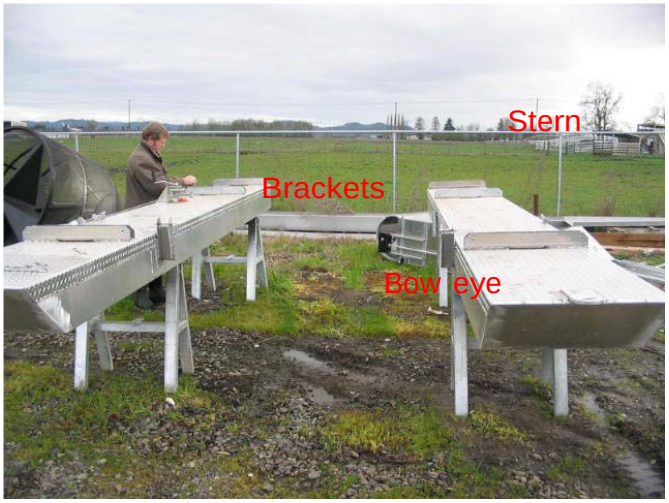
egsolutions@hotmail.com

Tools needed: 7/16 inch, 9/16 inch and 3/4 inch sockets (or the metric equivalent) and a ratchet. 7/16 inch, 9/16 inch and 3/4 inch open end or box wrenches (or the metric equivalent), or a crescent wrench or adjustable spanner.

These instructions show assembly on land. You should assemble the trap in the water unless you have a crane to lift it.

Place the pontoons 3/4 into the water, stern first. They should be about 5 ½ feet apart for a 5-ft trap and 8 ½ feet apart for an 8-ft trap. Port (left) and starboard (right) pontoons are marked with a "P" or "S" at the bow eye.

Note the bipod and handrail brackets are on the inboard side of the pontoons.

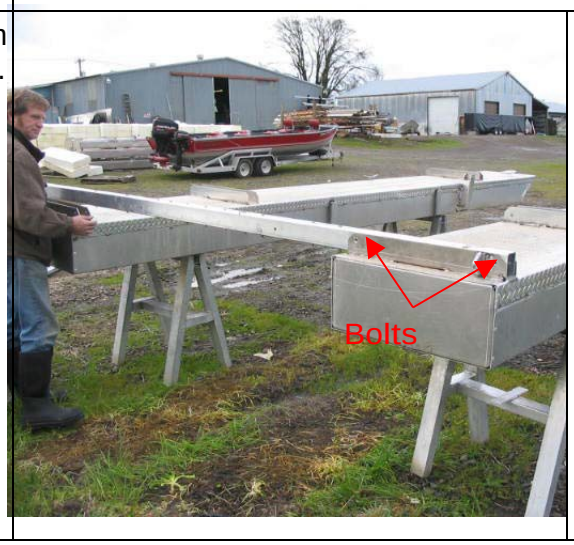


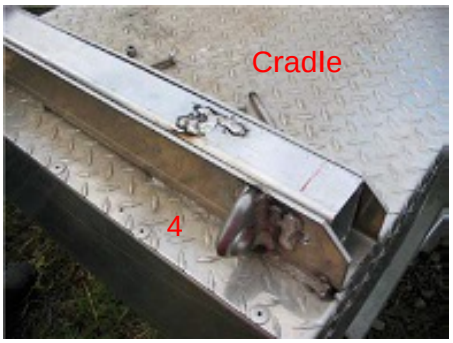
It is often easier to place the cone in shallow water and assemble the trap around it. The cone is not shown in this picture so that we could label the parts

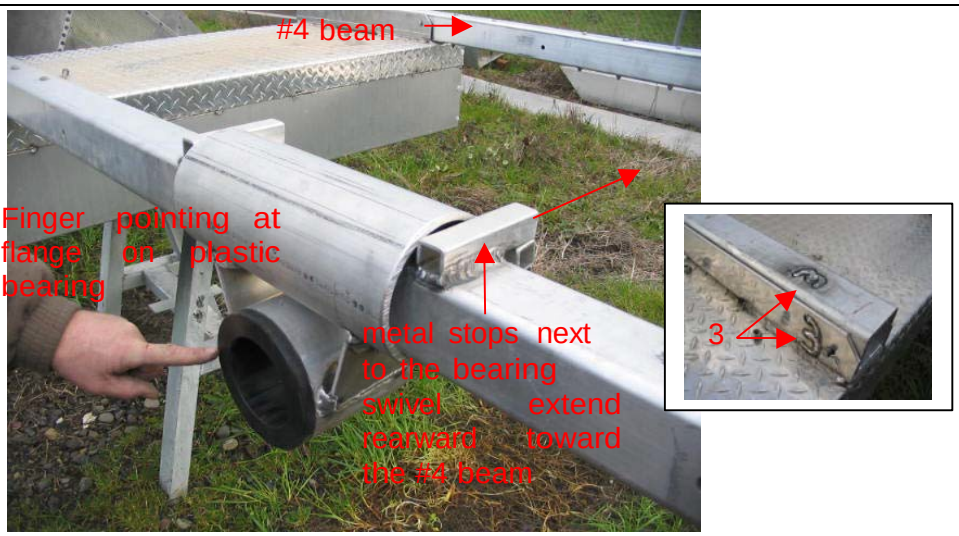
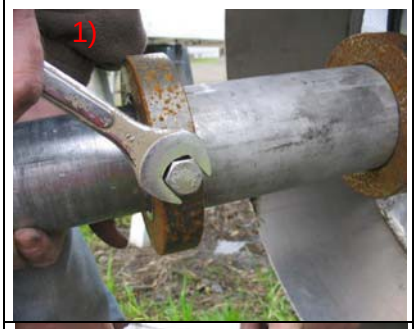
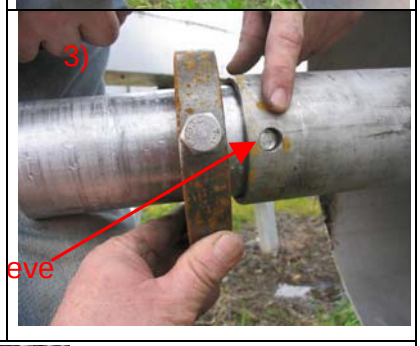
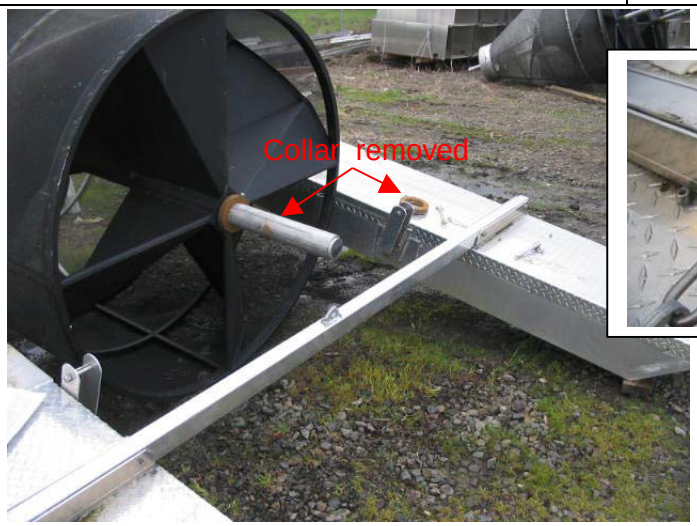
Port (left)

Starboard (right)

Match the #4 on the beam with the #4 on the pontoon. Insert and loosely bolt the #4 beam into cradles on both pontoons.

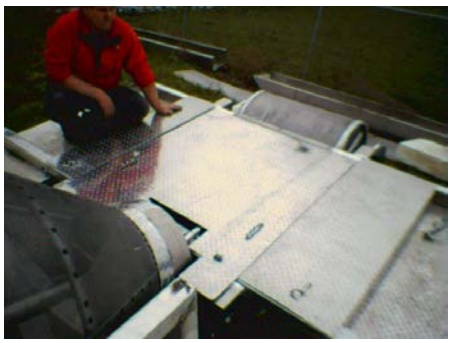




Insert and loosely bolt the #3 beam. The flange of the plastic bearing should face forward toward the bow and the metal stops next to the bearing should extend rearward toward the #4 beam. Match the #3 on the beam with the #3 on the pontoon.	
1) Remove one shaft collar from the small end of trapping cone. 2) Insert the shaft into the bearing on the #3 beam. 3) Replace the shaft collar making sure the bolt in the shaft collar goes through the hole in the wear sleeve and tightens on shaft.	  
Insert the #1 beam into the cradles and loosely bolt. Match the #1 on the beam with the #1 on the pontoon. Remove one collar from the large end of the trapping cone.	 

Slide the #2 beam onto the shaft with the flange of plastic bushing facing forward toward the bow. Replace and secure the shaft collar making sure the bolt in the shaft collar goes through the hole in the wear sleeve and tightens on shaft.	
1) Install the lifting bipod in the brackets between the #1 and #2 beams with the cross brace toward the starboard (right) side of the trap. 2) Center the vertical guides and sliding block around trapping cone shaft. 3) Secure the cross brace to the starboard bipod leg.	  

<u>5-ft trap:</u> String the cable and snatch blocks from the winch forward to the starboard end of the #1 beam, across to the center of the #1 beam, up to the front side of the apex of the bipod then down to the center of the #2 beam. <u>8-ft trap:</u> String the cable and snatch blocks from the winch forward to the starboard end of the #1 beam, across to the center of the #1 beam, up to the front side of the apex of the bipod, down to the center of the #2 beam, back up to the rear side of the apex of the bipod then down to the #1 beam.	 Stringing the cable for a 5-ft trap is illustrated in these pictures.	
		
Hoist the trapping cone to the up position with the winch.		

Install the handrail in the pockets near the center of the inboard side of the port (left) pontoon.	
Slide the livebox under #3 and #4 beams, lift into position and secure with bolts. This step could be done later if the trap is to be floated through shallow riffles or towed long distances to the trapping site,	
Install the decks (marked with a P or S) and livebox lid between the #3 and #4 beams. It is easier to insert the bolts up from the bottom.	  

Attach the arm between the live box and the lid.	
Move the trap to trapping site and secure in position using the eyes on the front of pontoons. Attach a safety line from eye on the rear of the pontoon to shore.	
Lower the cone until the #2 beam rests on the pontoons. Tighten the hoisting system without lifting the #2 beam and so that the bipod and cable do not contact the rotating trapping cone.	
Check that the bicycle tire contacts the trapping cone on the diamond plate portion of the cylinder at the small end of the cone and the debris drum is turning. If you are trying to trap salmonid fry or other very small fish you may disable the debris drum by removing the tire because small fish may be impinged and ride over the drum and out of the livebox. You may also want to remove the tire in very slow water to reduce the friction on the trapping cone.	

Appendix C. Recommended settings for HPR readers.

Menu – Settings - Device -

Timezone	-7.0
Backlight	35%
Beep Volume	100%
Power save	●
Auto Dim Time	0 min
Auto Sleep Time	10 min
Auto Shut Down	1 hour
Time Stamp Format	MDY
Tag Format	HEX
Tag Record Format	Tag only
Baud rate	9600
Bluetooth Enable	○
Bluetooth Password	na
External Power Cutoff	10.5 volts

Menu - Settings - Memory

Tag Type	FDX
Store Tags	●
File Number	Can customize
Send Tags	USB-PC, Drive, Serial
Read Indicator	Audible, visual
Unique Mode timeout	1 minute
Geotag Enable	○
Biotherm Enable	○
Fast Tag Enable	○
Single Tag Read	○
Read Error Rejection	●
Search Mode	Disabled
Search File number	0
Search Indicator	Audible, Visual
Send Search Results	na

Menu – Settings - Diagnostics

Send Communications	USB-PC, Drive, Serial
Alarm Indicator	Audible, Visual
Status Report Interval	0 minutes
Noise Report Interval	0 minutes
Background Noise Level	80%
Memory Low	10%
Antenna Current Low	0.1 amps
External Power Alarm	11.0 volts

Radio Frequency Settings & Diagnostics – Contains detailed information and settings related to the Radio Frequency (RF) functions of the device.

Menu- RF

Tuning	Set to auto or 25
Power	60%
Voltage	Voltage display
Current	Current display
FDX-B Noise	Higher the noise the lower the read range
HDX Noise	Higher the noise the lower the read range
BKG	Amount of background noise that will affect read range of both tags

Appendix C. Instructions for staining with Bismarck Brown Y.

1. Fill bucket with 16.0 liters of water.

2. Count out the number of fish to be stained with Bismarck Brown 'Y' and place them in the bucket containing 16.0 liters of water.

3. Put 2 to 3 aerators in bucket. Keep water aerated very well. Do not anesthetize fish prior to emersion in staining solution.

4. Measure out 0.4 grams of Bismarck Brown 'Y'.

5. Mix Bismarck Brown 'Y' in bucket containing fish and 16.0 liters of water. The solution is now 1:40,000 conversion by weight of Bismarck Brown 'Y' and 1:78,400 conversion by the true dye content of Bismarck Brown 'Y'.

6. Put thermometer in bucket.

7. Set lid on bucket to prevent fish from leaping out.

8. Check time or set timer for 1 hour.

9. Observe water temperature and fish every 5 to 10 minutes. If fish are reacting very abnormal, remove them immediately and place them in well-aerated fresh water.

10. If water temperature is warming up, add bottles of ice as needed to keep water temperature constant.

11. Stir water and observe fish often (fish will appear to be somewhat sluggish).

12. Remove fish after 1 hour has expired. If mark on fish appears too light or is unidentifiable, leave fish in solution a while longer. Do not exceed 1.5 hours of emersion time in solution.

13. Put fish in recovery live well. Observe them for a while. Fish will be very sluggish, so be careful to ensure that they do not get impinged on the sides if there is too much current flowing through the live well.

14. Release fish after full recovery for trap efficiency test.

Appendix E. R script for an iterative maximization of the log likelihood for a time-stratified Lincoln-Peterson estimator with Bailey's modification.

Emigration abundance estimates are calculated from trap operations with the Lincoln-Petersen estimator with Bailey's modification:

$$N = M(C + 1)/(R + 1)$$

where *N* is abundance of juveniles emigrating in a given year, *C* is the number of all unique fish captured, *M* is the number of tagged fish released upstream, and *R* is number of recaptures (Bailey 1951). The estimator is computed using an iterative maximization of the log likelihood, using R on the shinyapps webpage: http://ifwisshiny.idfg.state.id.us:3838/JLM/IDFGStatApps/. This weblink is only available from the network. Assumptions are that fish are captured independently with probability *p* (equivalent to trap efficiency) and tagged fish mix thoroughly with untagged fish. The 95% confidence intervals are computed with the bootstrap option (10,000 iterations).

```
LSBoot<-eventReactive(input$RunBoot,{
    data<-TrapData()
    colnames(data)<-c('Trap', 'Brood.Year', 'Life.Stage', 'Strata', 'C', 'M', 'R')
    UCITrap<-list()
    LCITrap<-list()
    UCITrapAll<-list()
    LCITrapAll<-list()
    CapProb4<-list()
    UpCapProb4<-list()
    LwCapProb4<-list()
    traps<-sort(unique(data$Trap))
    for(i in 1:length(traps)){
      trap.dat<-subset(data, Trap==traps[i])
      BY<-sort(unique(trap.dat$Brood.Year))
      UCIBYTrap<-list()
      LCIBYTrap<-list()
      UCIBYTRapall<-c()
      LCIBYTRapall<-c()
      CapProb3<-list()
      UpCapProb3<-list()
      LwCapProb3<-list()
      for(j in 1:length(BY)){
        BYdat<-subset(trap.dat, Brood.Year==BY[j])
        LS<-sort(unique(BYdat$Life.Stage))
        UCITrapBYLS<-c()
        LCITrapBYLS<-c()
        BootEstTrapBYLS2<-matrix(data=NA, nrow=length(LS), ncol=reps)
        CapProb2<-list()
```

```
    UpCapProb2<-list()
    LwCapProb2<-list()
    for(k in 1:length(LS))
    {LSdat<-subset(BYdat, Life.Stage==LS[k])
    strat<-sort(unique(LSdat$Strata))
    BootEstTrapBYLSStr<-matrix(data=NA,nrow=input$SelectIter2,
    ncol=length(strat))
    CapProb<-c()
    UpCapProb1<-c()
    LwCapProb1<-c()
    for(p in 1:length(strat)){
        stratdat<-subset(LSdat, Strata==strat[p])
        C<-stratdat$C
        R<-stratdat$R
        M<-stratdat$M
        Rboot <- rbinom(input$SelectIter2,size=C,prob=R/C)
        CapProb[p]<-R/M
        BootEstTrapBYLSStr[,p]<- (M)*(C+1)/(Rboot+1)
        UpCapProb1[p]<-quantile(Rboot/M, 0.975)
        LwCapProb1[p]<-quantile(Rboot/M, 0.025)}
    BootEstTrapBYLS<-apply(BootEstTrapBYLSStr, 1, sum)
    UCITrapBYLS[k]<-as.vector(quantile(BootEstTrapBYLS, 0.975))
    LCITrapBYLS[k]<-as.vector(quantile(BootEstTrapBYLS, 0.025))
    BootEstTrapBYLS2[k,]<-apply(BootEstTrapBYLSStr, 1, sum)
    CapProb2[[k]]<-CapProb
    UpCapProb2[[k]]<-UpCapProb1
    LwCapProb2[[k]]<-LwCapProb1}#LifeStage

    UCIBYTrap[[j]]<-UCITrapBYLS
    LCIBYTrap[[j]]<-LCITrapBYLS
    BootEstTrapBY<-apply(BootEstTrapBYLS2, 2, sum) #####
    UCIBYTRapall[j]<-as.vector(quantile(BootEstTrapBY, 0.975))
    LCIBYTRapall[j]<-as.vector(quantile(BootEstTrapBY, 0.025))
    CapProb3[[j]]<-unlist(CapProb2)
    UpCapProb3[[j]]<-unlist(UpCapProb2)
    LwCapProb3[[j]]<-unlist(LwCapProb2)
    #change order}

    #BroodYear
UCITrap[[i]]<-UCIBYTrap
LCITrap[[i]]<-LCIBYTrap
UCITrapAll[[i]]<-UCIBYTRapall
LCITrapAll[[i]]<-LCIBYTRapall
CapProb4[[i]]<-CapProb3
UpCapProb4[[i]]<-UpCapProb3
LwCapProb4[[i]]<-LwCapProb3}
```

```
  #Trap
UCITrapOut<-round(unlist(UCITrap),1)
LCITrapOut<-round(unlist(LCITrap), 1)
UCITrapAllOut<-round(unlist(UCITrapAll),1)
LCITrapAllOut<-round(unlist(LCITrapAll),1)
CapProbOut<-round(unlist(CapProb4), 4)
UpCapProbOut<-round(unlist(UpCapProb4), 4)
LwCapProbOut<-round(unlist(LwCapProb4), 4)
CILS<-list()
CILS[[1]]<-UCITrapOut
CILS[[2]]<-LCITrapOut
CILS[[3]]<-UCITrapAllOut
CILS[[4]]<-LCITrapAllOut
CILS[[5]]<-data.frame(CapProbOut, LwCapProbOut, UpCapProbOut)
CILS})
```

Prepared by:

Timothy Copeland
Fisheries Program Coordinator

Bruce Barnett
Fisheries Data Coordinator

William C. Schrader
Principal Fisheries Research Biologist

Kimberley A. Apperson
Fisheries Biologist

Laurie Janssen
Fisheries Technician 2

Ronald V. Roberts
Fisheries Technician 2

Approved by:

IDAHO DEPARTMENT OF FISH AND GAME

_____

J. Lance Hebdon, Chief
Bureau of Fisheries