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Susitna-Watana Hydroelectric Project (FERC No. 14241)

Draft Susitna River Productivity Study Implementation Plan

Prepared for

Alaska Energy Authority



Prepared by

R2 Resource Consultants, Inc.

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LIST OF ACRONYMS AND SCIENTIFIC LABELS

Abbreviation	Definition
ADF&G	Alaska Department of Fish and Game
ADNR	Alaska Department of Natural Resources
AEA	Alaska Energy Authority
Anadromous	Fishes that migrate as juveniles from freshwater to saltwater and then return as adults to spawn in freshwater.
APA	Alaska Power Authority
APA Project	APA Susitna Hydroelectric Project
Baseline	Baseline (or Environmental Baseline): the environmental conditions that are the starting point for analyzing the impacts of a proposed licensing action (such as approval of a license application) and any alternative.
Benthos (benthic)	Defining a habitat or organism found on the streambed or pertaining to the streambed (or bottom) of a water body.
Braided streams	Stream consisting of multiple small, shallow channels that divide and recombine numerous times. Associated with glaciers, the braiding is caused by excess sediment load.
Cascade	The steepest of riffle habitats. Unlike rapids, which have an even gradient, cascades consist of a series of small steps of alternating small waterfalls and shallow pools.
cfs	cubic feet per second
Channel	A natural or artificial watercourse that continuously or intermittently contains water, with definite bed and banks that confine all but overbank stream flows.
Cross-section	A plane across a river or stream channel perpendicular to the direction of water flow.
Depth	Water depth at the measuring point (station).
Devils Canyon	Located at approximately Susitna River Mile (RM) 150-161, Devils Canyon contains four sets of turbulent rapids rated collectively as Class VI. This feature is a partial fish barrier because of high water velocity.
Distribution (species)	The manner in which a biological taxon is spatially arranged.
et al.	"et alia"; and the rest
FERC	Federal Energy Regulatory Commission
Flood	Any flow that exceeds the bankfull capacity of a stream or channel and flows out on the floodplain.
Floodplain	1. The area along waterways that is subject to periodic inundation by out-of-bank flows. 2. The area adjoining a water body that becomes inundated during periods of over-bank flooding and that is given rigorous legal definition in regulatory programs. 3. Land beyond a stream channel that forms the perimeter for the maximum probability flood. 4. A relatively flat strip of land bordering a stream that is formed by sediment deposition. 5. A deposit of alluvium that covers a valley flat from lateral erosion of meandering streams and rivers.
Focus Area	Areas selected for intensive investigation by multiple disciplines as part of the AEA study program.
Geomorphic reach	Level two tier of the habitat classification system. Separates major hydraulic segments into unique reaches based on the channel's geomorphic characteristic.
Geomorphology	The scientific study of landforms and the processes that shape them.
GIS	Geographic Information System. An integrated collection of computer software and data used to view and manage information about geographic places, analyze

Abbreviation	Definition
	spatial relationships, and model spatial processes.
Glide	An area with generally uniform depth and flow with no surface turbulence. Low gradient; 0-1 % slope.
GPS	global positioning system. A system of radio-emitting and -receiving satellites used for determining positions on the earth.
Groundwater (GW)	In the broadest sense, all subsurface water; more commonly that part of the subsurface water in the saturated zone.
Habitat	The environment in which the fish live, including everything that surrounds and affects its life, e.g. water quality, bottom, vegetation, associated species (including food supplies). The locality, site and particular type of local environment occupied by an organism.
ILP	Integrated Licensing Process
Instream flow	The rate of flow in a river or stream channel at any time of year.
Juvenile	A young fish or animal that has not reached sexual maturity.
licensing participants; Participants	Agencies, ANSCA corporations, Alaska Native entities and other licensing participants
Lower segment Susitna	The Susitna River from Cook Inlet (RM 0) to the confluence of the Chulitna River at RM 98.
Main channel	For habitat classification system: a single dominant main channel. Also, the primary downstream segment of a river, as contrasted to its tributaries.
Main channel habitat	Level four tier of the habitat classification system. Separates main channel habitat types including: tributary mouth, main channel, split main channel, multiple split main channel and side channel into mesohabitat types. Mesohabitat types include pool, glide, run, riffle, and rapid.
Mainstem	Mainstem refers to the primary river corridor, as contrasted to its tributaries. Mainstem habitats include the main channel, split main channels, side channels, tributary mouths, and off-channel habitats.
Mainstem habitat	Level three tier of the habitat classification systems. Separates mainstem habitat into main channel, off-channel, and tributary habitat types. Main channel habitat types include: tributary mouth, main channel, split main channel, multiple split main channel and side channel. Off-channel habitat types include: side slough, upland slough, backwater, and beaver complex. Tributary habitat is not further categorized.
Major hydraulic segment	Level one tier of the habitat classification system. Separates the River into three segments: Lower River (RM 0-98), Middle River (RM 98-184), and Upper River (RM 184-233).
Mesh size	The size of holes in a fishing net.
Mesohabitat	A discrete area of stream exhibiting relatively similar characteristics of depth, velocity, slope, substrate, and cover, and variances thereof (e.g., pools with maximum depth <5 ft, high gradient rimes, side channel backwaters).
Middle segment Susitna	The Susitna River from the confluence of the Chulitna River at RM 98 to the proposed Watana Dam Site at RM 184.
Migrant (life history type)	Some species exhibit a migratory life history type and undergo a migration to from rivers/lakes/ocean.
Migration	Systematic (as opposed to random) movement of individuals of a stock from one place to another, often related to season.
Minnow trap	Normally composed of small steel mesh with 2-piece torpedo shape design, this trap is disconnected in the middle for easy baiting and fish removal.
N/A	not applicable or not available
Non-native	Not indigenous to or naturally occurring in a given area.

Abbreviation	Definition
°C	degrees Celsius
°F	degrees Fahrenheit
Off-channel	Those bodies of water adjacent to the main channel that have surface water connections to the main river at some discharge levels.
Off-channel habitat	Habitat within those bodies of water adjacent to the main channel that have surface water connections to the main river at some discharge levels.
Outmigrant trap	Several types of trapping equipment that can be used to estimate the abundance of downstream migrating anadromous salmonid smolts.
Overwintering	Freshwater habitat used by salmonids during the winter for incubation of eggs and alevin in the gravel and for rearing of juveniles overwintering in the stream system before migrating to saltwater the following spring.
pH	A measure of the acidity or basicity of a solution.
PIT	Passive Integrated Transponder tags used to individually identify animals and monitor their movements.
PM&E	protection, mitigation and enhancement
Pool	Slow water habitat with minimal turbulence and deeper due to a strong hydraulic control.
PRM	Project River Mile(s) based on the digitized wetted width centerline of the main channel from 2012 Matanuska-Susitna Borough digital orthophotos. PRM 0.0 is established as mean lower low water of the Susitna River confluence at Cook Inlet.
Project	Susitna-Watana Hydroelectric Project
Radiotelemetry	Involves the capture and placement of radio-tags in adult fish that allow for the remote tracking of movements of individual fish.
Rapid	Swift, turbulent flow including small chutes and some hydraulic jumps swirling around boulders. Exposed substrate composed of individual boulders, boulder clusters, and partial bars. Lower gradient and less dense concentration of boulders and white water than Cascade. Moderate gradient; usually 2.0-4.0% slope.
Resident	Resident fish as opposed to anadromous remain in the freshwater environment year-round
Riffle	A fast water habitat with turbulent, shallow flow over submerged or partially submerged gravel and cobble substrates. Generally broad, uniform cross-section. Low gradient; usually 0.5-2.0% slope.
Riparian	Pertaining to anything connected with or adjacent to the bank of a stream or other body of water.
River	A large stream that serves as the natural drainage channel for a relatively large catchment or drainage basin.
River corridor	A perennial, intermittent, or ephemeral stream and adjacent vegetative fringe. The corridor is the area occupied during high water and the land immediately adjacent, including riparian vegetation that shades the stream, provides input of organic debris, and protects banks from excessive erosion.
River mile	The distance of a point on a river measured in miles from the river's mouth along the low-water channel.
RM	River Mile(s) referencing those of the APA Project.
RSP	Revised Study Plan
Run (habitat)	A habitat area with minimal surface turbulence over or around protruding boulders with generally uniform depth that is generally greater than the maximum substrate size. Velocities are on border of fast and slow water. Gradients are approximately 0.5 % to less than 2%. Generally deeper than riffles with few major flow obstructions and low habitat complexity.

Abbreviation	Definition
Run (migration)	Seasonal migration undertaken by fish, usually as part of their life history; for example, spawning run of salmon, upstream migration of shad. Fishers may refer to increased catches as a "run" of fish, a usage often independent of their migratory behavior.
Side channel	Lateral channel with an axis of flow roughly parallel to the mainstem, which is fed by water from the mainstem; a braid of a river with flow appreciably lower than the main channel. Side channel habitat may exist either in well-defined secondary (overflow) channels, or in poorly-defined watercourses flowing through partially submerged gravel bars and islands along the margins of the mainstem.
Side slough	Off-channel habitat characterization of an Overflow channel contained in the floodplain, but disconnected from the main channel. Has clear water.
Slope	The inclination or gradient from the horizontal of a line or surface.
Slough	A widely used term for wetland environment in a channel or series of shallow lakes where water is stagnant or may flow slowly on a seasonal basis. Also known as a stream distributary or anabranch.
Spawning	The depositing and fertilizing of eggs by fish and other aquatic life.
Split main channel	Main channel habitat characterization where three or fewer distributed dominant channels.
Stratified sampling	A method of sampling from a population. In statistical surveys, when subpopulations within an overall population vary, it is advantageous to sample each subpopulation (stratum) independently. Stratification is the process of dividing members of the population into homogeneous subgroups before sampling.
Three Rivers Confluence	The confluence of the Susitna, Chulitna, and Talkeetna rivers at Susitna River Mile (RM) 98.5 represents the downstream end of the Middle River and the upstream end of the Upper River.
Tributary	A stream feeding, joining, or flowing into a larger stream (at any point along its course or into a lake). Synonyms: feeder stream, side stream.
TWG	Technical Workgroup
Upland slough	Off-channel habitat characterization feature that is similar to a side slough, but contains a vegetated bar at the head that is rarely overtopped by mainstem flow. Has clear water.
Upper segment Susitna	The Susitna River upstream of the proposed Watana Dam Site at RM 184.
Watana Dam	The dam proposed by the Susitna-Watana Hydroelectric project. The approximately 750-foot-high Watana Dam (as measured from sound bedrock) would be located at river mile (RM) 184 on the Susitna River. The dam would block the upstream passage of Chinook salmon, possibly other salmon species, and resident fish that migrate through and otherwise use the proposed Watana Dam site and upstream habitat in the Susitna River and tributaries.

1. INTRODUCTION

The Alaska Energy Authority (AEA) is preparing a License Application that will be submitted to the Federal Energy Regulatory Commission (FERC) for the Susitna-Watana Hydroelectric Project (Project) using the Integrated Licensing Process (ILP). The Project is located on the Susitna River, an approximately 300-mile-long river in Southcentral Alaska. The Project's dam site will be located at river mile (RM) 184, corresponding to Project RM (PRM) 187.1 of the updated GIS-based hydrography.

AEA filed the Revised Study Plan with FERC on December 14, 2012, which included a River Productivity Study (RSP Section 9.8). The overarching goal of the River Productivity Study is to collect baseline data to assist in evaluating the effects of Project-induced changes in flow and the interrelated environmental factor upon the benthic macroinvertebrate and algal communities in the Middle and Upper Susitna River. This implementation plan fulfills portions of the River Productivity Plan.

As described in RSP Section 9.8.1, the production of freshwater fishes in a given habitat is constrained both by the suitability of the abiotic environment and by the availability of food resources (Wipfli and Baxter 2010). Algae are an important base component in the lotic food web, being responsible for the majority of photosynthesis in a river or stream and serving as an important food source to many benthic macroinvertebrates. In turn, benthic macroinvertebrates are an essential component in the processes of an aquatic ecosystem due to their position as consumers at the intermediate trophic level of lotic food webs (Hynes 1970; Wallace and Webster 1996; Hershey and Lamberti 2001). Macroinvertebrates are involved in the recycling of nutrients and the decomposition of terrestrial organic materials in the aquatic environment, serving as a conduit for the energy flow from organic matter resources to vertebrate populations, namely fish (Hershey and Lamberti 2001; Hauer and Resh 1996; Reice and Wohlenberg 1993; Klemm et al. 1990). In turn, nutrients and energy provided by spawning salmon have the potential to increase freshwater and terrestrial ecosystem productivity (Wipfli et al. 1998; Cederholm et al. 1999; Chaloner and Wipfli 2002; Bilby et al. 2003; Hicks et al. 2005) and may subsidize otherwise nutrient-poor ecosystems (Cederholm et al. 1999). Recent studies have demonstrated the important role benthic macroinvertebrates play in processing salmon carcasses of coastal streams (Cederholm et al. 1999, Chaloner and Wipfli 2002).

The significant functional roles that macroinvertebrates and algae play in food webs and energy flow in the freshwater ecosystem make these communities important elements in the study of a stream's ecology. The operations of the proposed Project would likely shift one or more environmental factors that can affect the abundance and distribution of benthic algae and benthic macroinvertebrate populations, with an upward food chain effect on fish growth and productivity in the ecosystem. The degree of impact on the benthic communities and fish resulting from hydropower operations will necessarily vary depending on the magnitude, frequency, duration, and timing of flows, as well as potential Project-related changes in geomorphology, ice processes, temperature, and turbidity. By investigating the current condition of algal populations, benthic macroinvertebrates, and fish in the Susitna River and the trophic relationships between them, this study will provide a better understanding how changes in environmental factors might affect the availability and utilization of food resources at each trophic level in the system. In addition, by applying what is known about the effects of river

regulation and hydropower operation on these populations in riverine ecosystems, AEA can begin to assess the potential impacts of Project operations on river productivity in the Susitna River, as well as provide information to inform development of any necessary protection, mitigation, and enhancement (PM&E) measures, as appropriate.

The River Productivity Study (RSP Section 9.8) includes the description of the sampling scheme. However, decisions regarding specific site locations, timing, sampling devices, processing, and analyses were dependent upon recent results from 2012 data collection efforts.

A limited review of these 2012 results was used as a guide for a more detailed plan. This River Productivity Implementation Plan includes specific details for methods that will be used to conduct elements of this River Productivity Study.

Consistent with the RSP Section 9.8.4, this implementation plan includes: (1) a summary of relevant macroinvertebrate and algal studies in the Susitna River (Section 1.3.1), (2) an overview of the life-histories of the target fish species in the Susitna River that are selected for the trophic analysis (Section 1.3.2), (3) a review of the preliminary results of habitat characterization and mapping efforts and “Focus Areas” (Section 1.3.3), (4) a description of site selection protocols (Section 2.1), (5) a description of sampling protocols (Sections 2.2 through 2.11), (6) a description of sample processing protocols (Sections 2.2 through 2.11, and Appendix 1), (7) a discussion of data analysis methods (Sections 2.2 through 2.11), (8) development of field data collection forms (Appendix 2), and (9) development of database templates that comply with 2012 AEA QA/QC procedures (Section 2.12, and Appendices 3 and 4).

This implementation plan includes a level of detail sufficient to instruct field crews in data collection efforts. In addition, the plan includes protocols that will be used in the field, specific sampling locations, details about the choice and use of sampling techniques and equipment. The implementation plan will ensure that field collection efforts are consistent and repeatable among field crews and between river segments.

1.1. Study Goals and Objectives

To review, the overarching goal of the River Productivity study is to collect baseline data to assist in evaluating the potential Project-induced changes in flow and effects on environmental factors (e.g., temperature, substrate, water quality), and benthic macroinvertebrate and algal communities in the Middle and Upper Susitna River. Individual objectives that will accomplish this are as follows:

1. Synthesize existing literature on the impacts of hydropower development and operations (including temperature and turbidity) on benthic macroinvertebrate and algal communities.
2. Characterize the pre-Project benthic macroinvertebrate and algal communities with regard to species composition and abundance in the Middle and Upper Susitna River.
3. Estimate drift of benthic macroinvertebrates in selected habitats within the Middle and Upper Susitna River to assess food availability to juvenile and resident fishes.
4. Conduct a feasibility study in 2013 to evaluate the suitability of using reference sites on the Talkeetna River to monitor long-term Project-related change in benthic productivity.

5. Conduct a trophic analysis to describe the food web relationships within the current riverine community within the Middle and Upper Susitna River.
6. Develop habitat suitability criteria for Susitna benthic macroinvertebrate and algal habitats to predict potential change in these habitats downstream of the proposed dam site.
7. Characterize the invertebrate compositions in the diets of representative fish species in relationship to their source (benthic or drift component).
8. Characterize organic matter resources (e.g., available for macroinvertebrate consumers) including coarse particulate organic matter, fine particulate organic matter, and suspended organic matter in the Middle and Upper Susitna River.
9. Estimate benthic macroinvertebrate colonization rates in the Middle Susitna Segment under pre-Project baseline conditions to assist in evaluating future post-Project changes to productivity in the Middle Susitna River.

1.2. Study Area

The Study Area for this implementation is described in RSP Section 9.8.3. The River Productivity Study will entail field sampling throughout the Upper Segment and Middle Segment on the Susitna River (Table 1.2-1; Figures 1.2-1 and 1.2-2).

1.3. Background

1.3.1. Historic Data Collection Efforts

A number of evaluations of the benthic macroinvertebrate community were conducted on the Susitna River in the 1970s and in the 1980s for the original Alaska Power Authority (APA) Susitna Hydroelectric Project (Friese 1975; Riis 1975, 1977; ADF&G 1983a; Hansen and Richards 1985; Van Nieuwenhuyse 1985; Trihey and Associates 1986). ADF&G studies in the 1970s included sampling of macroinvertebrates using artificial substrates (i.e., rock baskets) deployed for a set period of time to allow for colonization. Friese (1975) set a total of eight rock baskets in Waterfall Creek and Indian River in various habitats (e.g., deep and shallow pool, deep and shallow riffle, quiet water) to determine species composition of the insect population in tributary streams. Rock baskets returned extremely low numbers of invertebrates, which were all aquatic insects, largely due to inadequate time allowed for colonization, as well as survey timing. The most common insects were *Isoperla* stoneflies (Plecoptera: Perlodidae) and "no-see-ums" (Diptera: Ceratopogonidae), with simuliid blackfly larvae (Diptera: Simuliidae), flat-headed mayfly nymphs (Ephemeroptera: Heptageniidae), and some predaceous caddisfly larvae also present (Friese 1975). Stomach content analysis of coho salmon fry was also collected at Sloughs 9, 11, and 15 on the Susitna River (August and September), and at Slough Number 2 on the Talkeetna River (June) to provide comparative data on food availability. Results demonstrated the importance of insect larvae, particularly Trichoptera and Diptera, in the diets of rearing fish. Salmon eggs were also an important food source. A larger variety of insects were present in the Talkeetna River stomach samples, probably due to the earlier time of year at which those fry were collected.

Riis (1975) used a total of eight rock baskets, colonized for 30 days during summer (July – September), to sample the mainstem Susitna River and Waterfall Creek in the Middle Susitna River segment. Mainstem Susitna River sites included points just upstream of the Deshka River and Willow Creek in the Lower Segment and above Gold Creek in the Middle Segment. Fourth of July Creek was also sampled using a kick screen. Numbers of organisms collected per site were low; the most common insects were stoneflies (Plecoptera: Perlodidae), mayfly nymphs (Ephemeroptera: Heptageniidae and Baetidae), and caddisfly larvae (Trichoptera: Rhyacophilidae and Sericostomatidae). Riis (1977) later deployed rock baskets in the Susitna River at several locations near the mouth of Gold Creek for a colonization period of 75 days; however, only two of seven baskets were retrieved. The two baskets collected a total of 118 organisms, comprised of 77 Plecoptera, 66 Ephemeroptera, and 55 Diptera.

Studies conducted in the 1980s for the original APA Susitna Hydroelectric Project focused on benthic macroinvertebrate communities in the sloughs, side channels, and tributaries of the Middle Segment of the Susitna River from RM 125 to RM 142 during the period from May through October. Efforts included direct benthic sampling with a kick screen or a Hess bottom sampler, and drift sampling. Alaska Department of Fish and Game (ADF&G) efforts in 1982 and 1984 also involved collection of juvenile salmon in these side channels and sloughs, and an analysis was conducted to compare gut contents with the drift and benthic sampling results (ADF&G 1983a; Hansen and Richards 1985). In addition, Hansen and Richards (1985) collected water velocity, depth, and substrate-type data to develop habitat suitability criteria (HSC), which were used to estimate weighted usable areas for different invertebrate community guilds, based on their behavioral type (swimmers, burrowers, clingers) in slough and side channel habitats. Efforts in 1985 (Trihey and Associates 1986) expanded to include sampling at nine sites in the Middle Susitna River Segment: three side channels, two sloughs, two tributaries, and two mainstem sites. Results presented are data in tabular format, by site and date, of samples processed to that point in time, and are not expanded or summarized, so limited conclusions can be made.

Algal communities were periodically sampled and analyzed for chlorophyll-*a* (*chl-a*) at Susitna Station from 1978 to 1980. In the 1980s, algae samples were collected as part of the APA Susitna Hydroelectric Project water quality studies, with sampling conducted at Denali, Cantwell (Vee Canyon), Gold Creek, Sunshine, and Susitna Station gage sites on the Susitna River, as well as on the Chulitna and Talkeetna rivers (Harza-Ebasco 1985 as cited in AEA 2011). Analysis showed low productivity (less than 1.25 mg/m³ *chl-a*) and indicated algal abundance was most likely limited by high concentrations of suspended sediment and turbidity (AEA 2011).

Baseline field data for estimating benthic primary and secondary production was also collected in 1985, as part of the Primary Production Monitoring Effort (Van Nieuwenhuyse 1985). Chlorophyll-*a* and macroinvertebrates were collected from early April to late October 1985 from a variety of off-channel and mainstem habitat sites. Early April sampling took place in an open-water lead in Slough 8A and revealed high macroinvertebrate densities (averaging 17,600 individuals/m²) comprised almost entirely of chironomid larvae and chlorophyll-*a* densities averaging 34.4 mg/m². Sampling in early May in Slough 8A revealed macroinvertebrate densities averaging 2,950 individuals/m², dominated by chironomids, with *chl-a* densities averaging 37mg/m². Results from five mainstem habitat sites showed similar macroinvertebrate numbers, with densities ranging from 393 to 8,820 individuals/m² in May 1985, but with considerably more diversity; chironomids accounted for an average of 53 percent of the density

and only 8 percent of the macroinvertebrate biomass. Algae samples beyond May 1985 had not been analyzed; therefore, no data were available for summer or fall. No sampling results were given for summer macroinvertebrate sampling (June and July). August and September 1985 sampling showed low average densities at mainstem sites (44 – 164 individuals/m²), with large increases occurring in October 1985 (1,729 – 7,109 individuals/m²). Average densities in Slough 8A in August 1985 remained similar to spring levels (2,851 individuals/m²), with a surge in September 1985 (13,964 individuals/m²); again, chironomids represented over 80 percent of the numbers. No further information or reports were available concerning the Primary Production Monitoring Effort task.

Benthic macroinvertebrate information from the 1980s is largely focused on a limited number of mainstem, side channel, and slough habitats located within a 17-mile reach of the Middle Susitna River. Additional information is needed on mainstem benthic communities, as well as those in side channel and slough habitats, within both the Middle and Upper Susitna River segments. Benthic algae information needs to be collected in conjunction with the macroinvertebrates, particularly with functional feeding groups, to define their relationship in the river's trophic system. To assess potential impacts of future hydropower operations on the benthic communities within the Susitna River, additional information must be collected through an increased sampling effort, including more sampling sites along the river in relation to the distance both downstream from the proposed dam site and upstream from the proposed Project reservoir area.

1.3.2. Life History Summary of Susitna Target Fish Species

1.3.2.1. Coho salmon (*Oncorhynchus kisutch*)

1.3.2.1.1. General Life History

Coho salmon are widely distributed throughout the North Pacific basin. Their distribution ranges from the Sea of Japan north to Point Hope, Alaska, and south to the Sacramento River in California (Sandercock 1991). Along the Pacific coast of Alaska, coho salmon are native to coastal rivers and streams in the Southeast, Southcentral and Southwestern regions of the state. Coho salmon have been documented in the mainstem and several Susitna River tributaries, including the Yentna, Talkeetna, and the Chulitna rivers (ADF&G 2012).

Like other Pacific salmon species, coho salmon are anadromous. North American coho salmon typically spawn from October to March, although entry into freshwater and spawning time varies among populations and with environmental conditions (Morrow 1980, Sandercock 1991). In northwestern Canada and Alaska, adult coho salmon may begin their upstream migrations as early as late June and July; however, most of the spawning in these areas occurs in November. In Southcentral Alaska, adult returns to freshwater peak in August and September (McPhail and Lindsey 1970) and spawning continues through the fall. Coho salmon adults die after spawning.

The duration of incubation for coho salmon ranges from 35 to 101 days (Laufle et al. 1986) and is temperature dependent. Specific to Alaska coho salmon, the incubation period ranges from 42 to 56 days (McPhail and Lindsey 1970). After hatching, larval fish typically spend 2 to 3 weeks in the gravel before emerging between early March and mid-May (Laufle et al. 1986, McMahon 1983). Juvenile coho salmon rearing time in freshwater is typically about 15 months, although some juveniles will remain in freshwater for up to 2 years (Sandercock 1991). Smolt outmigration begins in February and may continue into June; however, in more northern

populations, outmigration is likely to occur later and extend into July or August. While the majority of coho salmon reach maturity and return from the sea to reproduce in their natal tributaries as 3-year olds, precocious males that reach maturity during their first (referred to as “jacks”) or second year are a natural component of many Alaska coho salmon populations (Sandercock 1991).

1.3.2.1.2. *Periodicity*

During studies conducted in the 1980s, adult coho salmon migration timing in the main channel areas of the Lower Segment of the Susitna River occurred from early July through early October, with peak passage in late July and early August (Roth and Stratton 1985, Roth et al. 1986). Migration into Lower Segment spawning tributaries was estimated to start in mid- or late-July and peak during the month of August (Roth and Stratton 1985, Roth et al. 1986). Upstream spawning migration of adult coho salmon into the Middle Segment of the Susitna River typically began in late July and continued through early October based on studies conducted during the 1980s, with peak movement during early and mid-August (Jennings 1985, Thompson et al. 1986). Adult coho salmon primarily used main channel areas for migration to access tributary spawning sites (Jennings 1985). Upstream migration into Middle River spawning tributaries was delayed due to holding and milling behavior in the lower extent of the Middle Segment and in areas proximal to spawning tributaries (ADFG 1981-APA324, ADFG 1982-APA588). Based on observed milling and/or delay between date of radio-tagging and tributary entry, the timing of tributary entry and upstream migration was estimated to occur from early August through early October, with peak movement in late August and early September.

Coho salmon spawning in the Middle Susitna River occurred from mid-August through early October and peaked during mid- and late September (Jennings 1985). The timing of main channel spawning was assumed to be the same as tributary spawning due to sparse main channel spawning data. Primary spawning tributaries in the Middle Segment were Indian River, Gash Creek, Chase Creek, and Whiskers Creek (Jennings 1985, Thompson et al. 1986). Spawn timing in Lower Segment tributaries was slightly earlier relative to Middle Segment streams and occurred from early or mid-August through early October, with peak spawning in late August and early September (Roth et al. 1986). Coho salmon spawning in the Lower River Segment occurred almost entirely in tributary habitats during the 1980s studies, though approximately 13 percent of adult coho salmon tagged in a 2009 study utilized Lower River Segment mainstem areas for spawning (Roth and Stratton 1985, Roth et al. 1986, Merizon et al. 2010).

The timing and duration of coho salmon egg incubation and fry emergency is not well defined in the Susitna River due to sparse winter data. The incubation period begins with the start of spawning in mid-August and continues through fry emergence in the following spring. Coho salmon fry emergence began prior to the start of outmigrant trap operation in mid-May 1983 and 1985, though ice cover precluded trap operation prior to this point (Schmidt et al. 1983, Roth et al. 1986). Salmon egg incubation time depends on water temperature and the duration necessary for coho salmon egg development from the point of fertilization to fry emergence can range from 228 days at water temperatures of 2° C to 139 days at 5° C (Murray and McPhail 1988 cited in Quinn 2005). Based on these data and approximate timing of coho salmon emergence in similar areas, coho salmon fry emergence in the Susitna River is thought to begin in early March (Scott and Crossman 1973). Among age-0 coho salmon captured in June and July of 1981, 1982, and

1983, the lower extent of the length range was less than 35 mm, which suggests that emergence may continue through May or beyond (Jennings 1985).

Age-0+ coho salmon utilized natal tributaries for nursery habitats immediately following emergence, but many emigrated from tributaries soon after emergence to mainstem habitats between early May through October (Jennings 1985). Within the Susitna River mainstem, age-0+ salmon primarily used upland sloughs and side sloughs during the open water season. Juveniles also moved downstream to the Lower River Segment based on outmigrant trap catch data. Downstream movement of age-0 coho salmon to the Lower River Segment appeared to begin in early May, prior to outmigrant trap operation each year, and continued through October, with peak movement from late June to late August (Jennings 1985, Roth et al. 1986). Movement by age-0+ coho salmon observed in September and October may have been dispersal into suitable winter nursery habitats, which were side sloughs and upland sloughs in the Middle Segment (Jennings 1985, Roth et al. 1986). Within the Lower Segment mainstem, age-0+ coho salmon primarily used tributary mouths as nursery habitats, with comparatively little use of side channel or side slough habitats (Suchanek et al. 1985). A portion of age-0+ coho salmon may have emigrated to marine or estuarine areas during September and October based on capture data at the Flathorn Station (RM 22) outmigrant trap (Roth and Stratton 1985).

Ages-1+ and 2+ coho salmon primarily utilized natal tributaries, side sloughs, and upland sloughs as nursery habitat in the Middle River Segment (Dugan et al. 1984). Historic data indicates that juvenile coho salmon remained in the Susitna Basin as age-1+ parr but some portion of this age group dispersed from natal habitats in the Middle River, as suggested by few age-2+ coho salmon captures in the Middle River during the 1980s (Stratton 1986). These researchers surmised that these juvenile coho salmon had dispersed to the Lower River.

Dispersal from nursery habitats occurred during winter and early spring, although the timing and pattern of this movement is not well understood. Limited data collected during the winter of 1984-1985 suggested that juvenile coho salmon parr exhibit movements similar to juvenile Chinook salmon, with downstream migration between November and February (Stratton 1986). Age-1+ coho salmon in the Lower Segment redistributed to suitable habitats throughout the open water season, while a portion emigrated as smolts to estuarine areas (Roth et al. 1986). Based on limited data collected during winter in the Middle River Segment, age-1+ and age-2+ coho salmon were believed to have begun emigration from nursery habitats in early winter, and the peak of mainstem downstream movement likely occurred during the open water season (Stratton 1986, Roth et al. 1986). Age-2+ coho salmon emigration from the Lower Segment was estimated to have occurred between early January through mid-July, with movement in June (Roth et al. 1986).

1.3.2.1.3. Distribution

Coho salmon distribution in the Susitna River Basin extends from Portage Creek (RM 148.9) to Cook Inlet (RM 0.0; Jennings 1985, Thompson et al. 1986). Coho salmon counted at the Yentna Station represented 16 to 46 percent (average 35 percent) of the combined escapement estimated at the Yentna and Sunshine Stations (ADF&G 1981, ADF&G 1982C, ADF&G 1984, Barrett et al. 1985). Merizon et al. (2010) radio-tagged 300 coho salmon at Flathorn during 2009 and assigned a spawning location to 275 of the tagged fish based on tag detections and movement patterns. Coho salmon were strongly oriented toward the east or west banks. Consequently, fish

captured and tagged on the west side of the river primarily entered the Yentna River, while those captured on the east side tended to migrate up the Susitna River. Of the 275 coho salmon tagged at Flathorn and assigned a spawning location, four (1.5 percent) spawned in the Middle Susitna River, and none entered associated tributaries (Merizon et al. 2010). For the Lower Susitna River, 130 coho salmon (47.3 percent of those assigned a spawning location) spawned in the Yentna drainage, 39 (14.2 percent) spawned in the Lower Susitna River, and 102 (37.1 percent) spawned in other tributaries to the Lower Susitna River, primarily the Talkeetna, Deshka, and Chulitna drainages. Caution is warranted when considering the results of Merizon et al. 2010 as these researches based spawning on movement patterns and tag locations determined from the air and did not confirm spawning activity or the presence of redds in presumed spawning locations.

Spawning surveys were conducted each year from 1981 to 1985, but the level of intensity varied from year to year. In contrast to the 2009 radio tracking, spawning surveys conducted at 811 sites in the Lower Susitna River in 1982 did not identify any coho salmon spawning locations in the mainstem river (Barrett et al 1983). However, Barrett et al. (1985) and Thompson et al. (1986) conducted intensive surveys in 1984 and 1985 and identified coho salmon in tributaries of the Middle Susitna River. During 1984, Barrett et al. (1985) identified two non-slough and one slough spawning areas in the mainstem of the Lower Susitna River. They also identified 11 of 17 tributary mouths that were used as holding habitat, but not for spawning. Based on these historic data, Whiskers Creek, Indian River, and Chase Creek (RM 106.9) accounted for the majority of the tributary spawning in the Middle Susitna River. Thompson et al. (1986) observed coho salmon milling in five sloughs of the Middle Susitna River during 1985, and Barrett et al. (1985) observed milling in three sloughs during 1984, but no spawning activity was observed in sloughs during either year. In 1984, Barrett et al. (1985) identified one non-slough spawning area with two coho salmon in the mainstem of the Middle Susitna River.

While there is some uncertainty regarding the precise proportional distribution of coho salmon among the different Susitna River spawning areas due to annual variability, the tributaries associated with the Lower Susitna River are the major coho salmon production areas. In addition, adult coho salmon appeared to use mainstem channels and sloughs; however, actual documentation of spawning in these habitats has been very rare. The Middle Susitna River tributaries account for a small portion of the total Susitna River coho salmon production.

1.3.2.1.4. Adult Escapement and Juvenile Relative Abundance

Coho salmon are the least abundant anadromous salmon returning to the Susitna River Basin yet are important components for commercial and sport fisheries. From 1966 to 2006, an annual average of 313,000 coho salmon were caught for the commercial fishery in the Upper Cook Inlet (UCI) Management Area (Merizon et al. 2010). Next to Chinook salmon, coho salmon are the second highest contributor to the sport fishery with an annual average of 40,767 fish captured from 1998 to 2007 (Merizon et al. 2010). Average combined escapement for coho salmon in the Yentna Basin and Susitna Basin upstream of RM 80 from 1981 to 1984 was 61,400 fish; annual escapement was not estimated for the Susitna Basin downstream of RM 80 from 1981 to 1983, except for in the Yentna Basin (Jennings 1985). During 1981-1984, average escapement at the Talkeetna Station (RM 103) fishwheel was 5,700 fish, while escapement estimates at the Sunshine Station (RM 80) and Yentna River Station (Susitna RM 28.0; Yentna RM 4.0) fishwheels were 43,900 and 19,600 fish, respectively (Jennings 1985). Total coho salmon escapement in the Susitna Basin was estimated to be 663,000 in 2002 (Willette 2003).

Based upon sonar counts of fish returning to the Yentna River and Peterson estimates of returns to the Sunshine Station, minimum coho salmon returns to the Susitna River averaged 61,986 fish annually from 1981 through 1985 and ranged from 24,038 to 112,874 fish (ADF&G 1981, ADF&G 1982c, ADF&G 1984, Barrett et al. 1985, Thompson et al. 1986). These values represent minimum estimates, because sonar counts at the Yentna River station underestimate the total returns to the Yentna River (Jennings 1985). The average annual return to Talkeetna Station from 1981 to 1984 was 5,666 coho salmon. However, this may be an overestimate because coho salmon adults may enter the Middle Susitna River, and then migrate back downstream to spawn in other areas, as suggested by previous tracking studies. The Talkeetna Station was not operated in 1985. Average returns to Curry Station were 1,613 fish and ranged from 761 to 2,438 fish from 1981 to 1985.

From June through September of 1982, a total of 1,857 juvenile coho salmon were captured by all gear types at Designated Fish Habitat (DFH) sites from Goose Creek 2 upstream to Slough 21 (Estes and Schmidt 1983). Total juvenile coho salmon catch from this effort is shown by gear type and site in Figure 1.3-1. Juvenile coho salmon were present for at least one of the eight sampling periods in roughly 90 percent of the 17 DFH sites sampled.

Sampling in 1983 at Juvenile Anadromous Habitat Study sites captured 2,023 juvenile coho salmon between the Chulitna River (RM 98.6) and Portage Creek (RM 148.8; Dugan et al. 1984). Relative abundance determined from this effort is shown in Figure 1.3-2, both seasonally and by site. Age composition consisted of 97 percent age 0+, 3 percent age 1+, and less than one percent age 2+ fish. In general, juvenile coho salmon were widely distributed in low densities at many sites in the Middle Segment of the Susitna River, although high tributary densities were observed in early July and August. Juvenile coho salmon CPUE estimates were frequently highest at sites located in the lower portion of the Middle Segment.

1.3.2.1.5. *Habitat Associations*

Adult coho salmon spawn almost exclusively in tributary habitats, although adults have been documented in main channel, side channel and side slough habitats during the 1980s and in 2009 (ADF&G 1984-APA 1450, Barrett et al. 1985, Merizon et al. 2010). During 1984, coho salmon were recorded spawning at one side channel location in the Middle River Segment and in two side channels and one side slough site in the Lower River Segment (Barrett et al. 1985). No spawning was observed by coho salmon in surveyed slough or tributary mouth habitats (Barrett et al. 1985, Jennings 1985). Radio tracking studies conducted in 2009 indicated that 14 percent of all tagged coho salmon ($n = 275$) spent time in mainstem (i.e., main channel and off-channel) habitats in the Middle and/or Lower Susitna River segments (Merizon et al. 2010). Primary spawning tributaries for coho salmon based on the 1980s and 2009 data are Indian River and Whiskers Creek in the Middle Segment and the Chulitna, Deshka, and Yentna rivers in the Lower River Segment (Jennings 1985, Thompson et al. 1986, Merizon et al. 2010).

Based on scale analysis of returning adults, most juvenile coho salmon in the Susitna Basin reside in nursery habitats for 1 or 2 years prior to emigrating as age-1+ and age-2+ smolts to marine areas (ADF&G 1984, Barrett et al. 1985). The proportions of coho salmon that emigrate as age-1+ and age-2+ varied among years during the 1980s, though approximately equal proportions of adults exhibited each life history; a small portion (i.e., < 5 percent) of juvenile coho salmon emigrated as age-3+ smolts (ADF&G 1984, Barrett et al. 1985). During the open-

water period, age-0 and age-1 juveniles in the Middle River Segment primarily utilized clear water habitats associated with natal tributaries and upland sloughs (Figure 1.3-3), whereas those in the Lower River Segment used clear water tributaries and tributary mouths more consistently than side slough or side channel habitats, which were often more turbid (Schmidt and Bingham 1983, Dugan et al. 1984, Suchanek et al. 1985). Catch of age-0 juvenile coho salmon fry at tributary mouths peaked in July and August (Delany et al. 1981). These authors suggest that juvenile coho salmon movement in late summer may have been in response to declining water temperature and relocation to overwintering habitats. Coho salmon overwintered in side sloughs and upland sloughs in the Middle River Segment and tributary mouths and side channels in the Lower River Segment, though the distribution and intensity of fish sampling was reduced by ice cover and weather conditions (Delaney et al. 1981, Stratton 1986). Age-2 coho salmon were believed to rear primarily in Lower River Segment habitats during winter, based on low capture rates of age-2 fish in the Middle River Segment during winter (Stratton 1986).

1.3.2.2. *Chinook salmon, (Oncorhynchus tshawytscha)*

1.3.2.2.1. *General Life History*

Chinook salmon are distributed from northern Hokkaido, Japan, to the Anadyr River in Siberia and from the San Joaquin River in Central California to the Coppermine River in the Canadian Arctic (Healey 1991). In Alaska, Chinook salmon occur in large coastal rivers from the southern tip of Alaska's panhandle northward to Point Hope (Mecklenburg et al. 2002). The Chinook salmon stock of the Susitna River is the fourth largest in Alaska (Ivey et al. 2009).

As with other Pacific salmon, Chinook salmon are anadromous. Chinook salmon mature and begin their spawning migration between 3 and 6 years of age, but most spawning adults are ages 4 and 5 (Healey 1991). In northwestern Canada and Alaska, adults migrate to freshwater spawning grounds between late May and July, although this period may extend from April to September in some locations (Healey 1991). While spawning generally takes place from July to November, spawning time varies regionally and depends on the distance and duration of river migration (Morrow 1980, Scott and Crossman 1973). Northern populations, such as those in Alaska, tend to spawn from July through September (Healey 1991). Adults die following reproduction and egg deposition into one or more gravel nests known as redds.

Chinook salmon egg incubation varies with temperature, with lower temperatures resulting in increased time to hatching (Healey 1991). After hatching in the spring, the young remain in the gravel for 2 to 3 weeks and then emerge as free-swimming, feeding fry (Morrow 1980). While some juvenile Chinook salmon may rapidly disperse to sea, this life history pattern tends to be absent in locations north of 56 degrees North latitude, such as Alaska (Quinn 2005). In these northern locations, most juvenile Chinook salmon remain in freshwater streams for 1 year before beginning their outmigration to sea, but some will remain in freshwater for 2 years (Morrow 1980, Quinn 2005).

Owing to their large body size, adult Chinook salmon require deep holding water and sufficient stream flow to successfully complete their upstream migration. Spawning depths vary widely, from 5 to 720 centimeters (cm), with average spawning depths starting at 30 cm (Healey 1991). The large body size of Chinook salmon also enables them to use large gravel and cobble substrates for spawning (Raleigh et al., 1986). Successful incubation requires clean water percolating through spawning gravels at temperatures less than 16 °C (Healey 1991).

Juvenile Chinook salmon occupy a variety of habitats during their stay in freshwater. Younger, smaller fry inhabit stream margins, eddies, backwaters, and side channels and are often associated with fallen trees, root wads, and areas with bank cover. As they increase in size, juvenile Chinook salmon move into stream and river habitats with increasing velocities (i.e., up to 1.2 meters per second). This movement is associated with a shift from predominantly sandy substrates to those with larger-sized gravel and boulders (Healey 1991).

1.3.2.2.2. *Periodicity*

In the Susitna River, adult Chinook salmon begin their upstream migration in late-May to early June (Jennings 1985, ADF&G 1984). Although a few Chinook salmon may pass Susitna Station (RM 26.7) as late as mid-August, nearly all Chinook salmon (95 percent) have passed the station by the first week of July (ADF&G 1981, Jennings 1985). Peak run timing is generally later at Talkeetna Station (RM 103) compared to Sunshine Station. However, peak run timing at Curry Station appears to be similar or earlier than at Talkeetna Station, suggesting that upriver fish (i.e., Chinook salmon bound primarily for Indian and Portage creeks) enter and migrate during the early portion of the overall Chinook salmon migration period in the Susitna River Basin. Spawning generally begins in mid-July and is finished by the end of August (Barrett 1985, Jennings 1985). Peak spawning is during the last week of July and first week of August (Jennings 1985). Run timing may be affected by high flow levels, as indicated by decreased fishwheel catch rates; however, this pattern was not consistent across all years (Jennings 1985).

The timing of Chinook salmon fry emergence in Susitna River tributaries is poorly understood due to the difficulty of early and mid-spring sampling in the Susitna River Basin. Sampling for outmigrating fish following ice-out can seldom occur prior to mid-May and frequently cannot begin until early June. Delaney et al. (1981) reported that Chinook salmon fry were collected in Indian River in April during 1981 as part of a winter sampling effort. In 1982, sampling did not begin until early June, and fry were already present by this time (Schmidt et al. 1983). During 1985, sampling in Portage Creek and the Indian River began on July 9, and Chinook salmon fry were captured at relatively high rates with lengths ranging from 36 to 64 mm (Roth et al. 1986), suggesting that emergence was primarily completed by that time. Schmidt and Bingham (1983) reported that Chinook salmon fry emerge in April and March, while Stratton (1986) reported that emergence occurs in April; however, neither of these authors provides any supporting field sampling data for these conclusions.

Nearly all Chinook salmon juveniles outmigrate to the ocean as age-1+ fish. The bulk of Chinook salmon fry outmigrate from the Indian River and Portage Creek by mid-August and redistribute into sloughs and side channels of the Middle Susitna River or migrate to the Lower River (Roth and Stratton 1985, Roth et al. 1986). Outmigrant trapping occurred at Talkeetna Station (RM 103) during open water periods from 1982 to 1985 and demonstrated that Chinook salmon fry were migrating to the Lower Susitna River throughout the time traps were operating (Schmidt et al. 1983, Roth et al. 1984, Roth and Stratton 1985, Roth et al. 1986). Peak catch often occurred during periods of high flows. Outmigrant traps were also fished at Flathorn Station (RM 22.4) in 1984 and 1985 and demonstrated peak periods of Chinook salmon fry movement during early July; however, many of these fry may have originated from the Deshka River (Roth and Stratton 1985, Roth et al. 1986). Roth and Stratton (1986) suggested that some Chinook salmon fry either overwinter in the Lower Susitna River between the mouth and Flathorn Station or outmigrate to the ocean as fry. They also suggested that outmigration as fry

is a relatively unsuccessful life history pattern for Chinook salmon in the Susitna River, because scale pattern analysis indicates that few adults return.

Based on the capture of a small number of age-1+ Chinook salmon juveniles in the Indian River during winter sampling (Stratton 1986), it is thought that some Chinook salmon fry remain in natal tributaries throughout their first year of life and overwinter in any available suitable habitat. In 1984, sampling in the Indian River to cold brand juvenile salmon failed to capture any Chinook salmon age-1+ fish during July, yet was successful during May and June, suggesting that age-1+ Chinook salmon juveniles emigrate from tributary streams shortly after ice-out (Roth and Stratton 1985). The cumulative frequency of age-1+ Chinook salmon captured in 1985 at Talkeetna and Flathorn stations reached 90 percent by early July and late-July, respectively (Roth et al. 1986). These data indicate that outmigrating age-1+ smolts are generally in estuarine or near-shore waters by mid-summer.

1.3.2.2.3. *Distribution*

Based upon observations of juveniles, Chinook salmon are distributed in the Susitna River up to at least the Oshetna River (RM 225) (Buckwalter 2011). During the 1980s two spawning Chinook salmon were observed in Fog Creek (RM 176.7) during 1984 (Barrett 1985). More recently Buckwalter (2011) observed adult Chinook salmon in Fog Creek (RM 176.7) and Tsusena Creek (RM181.3) during 2003 and in Kosina Creek (RM 201) during 2011. Juvenile Chinook salmon were also observed in Fog Creek, Kosina Creek, and Oshetna River during 2003 and a Fog Creek tributary during 2011. In addition, adult Chinook salmon were observed in Cheechako Creek (5), Chinook Creek (5), Devil Creek (7), Fog Creek (1), and Kosina Creek (16) during 2012, with evidence of spawning documented in Kosina Creek, as well (AEA unpublished data).

A series of three partial velocity barriers are present in Devils Canyon, restricting access to upstream habitat. Chinook salmon are the only known anadromous salmon that can pass all three barriers (AEA unpublished data). The lower two barriers appear to be passable by Chinook salmon at a relatively broad range of flows while the upper barrier, located downstream of Devil Creek, can only be passed under a narrow range of flows.

Chinook salmon spawn exclusively in tributary streams (Thompson et al. 1986, Barrett 1985, Barrett 1984, Barrett 1983). Consequently, the mainstem Susitna River primarily provides a migration corridor and holding habitat for adult Chinook salmon. Apportionment of Chinook salmon among the major Susitna River subbasins from peak spawning surveys is somewhat confounded by inconsistent surveys, in part because of poor visibility and partly due to annual differences in surveying priorities. Nevertheless, major patterns in the distribution of Chinook salmon spawning during the late 1970s and early 1980s are discernible based upon data summarized by Jennings (1985). Important spawning tributaries in the Lower River included the Deshka River and Alexander Creek, the Yentna, Talkeetna, and Chulitna Rivers. The Yentna River and Talkeetna R/Chulitna subbasins are big producers and typically accounted for about 20 percent and 15 percent, respectively, of the Chinook salmon spawning for the entire Susitna River. There was proportionally much less spawning in the Middle River tributaries, which typically accounted for about 5 percent of the total Chinook salmon spawning. When focusing in on the Middle River spawning habitats, Portage Creek and Indian River accounted for nearly all of the Chinook salmon spawning at approximately 90 percent or greater. Other tributaries, such

as Fourth of July Creek and Whiskers Creek, accounted for minor amounts of spawning, generally with no more than about 2.5 percent of the spawning in the Middle River.

1.3.2.2.4. *Adult Escapement and Juvenile Relative Abundance*

Of the five salmon species returning to the Susitna River, Chinook salmon have had the smallest run size, but have been the most important sport fish (Jennings 1985). Long-term escapement trend data from 1974 to 2009 was available for a number of index streams in the Susitna River Basin monitored by ADF&G, but comparisons among streams were unreliable because of different survey methods (weirs, foot, or aerial; Fair et al. 2010). Most index streams were tributaries to the mainstem in the Lower Susitna River or tributaries in the Chulitna and Talkeetna subbasins (Fair et al. 2010). The Deshka River (RM 40.6) had the highest escapement of all tributaries with a median of 35,548 fish. ADF&G installed a counting weir in the Deshka River prior to the 1995 season to improve the accuracy of salmon escapement counts (Fair et al. 2010). All other index streams generally had fewer than 5,000 fish spawning during peak surveys.

Total peak counts of Chinook salmon spawning in Middle River tributaries between 1981 and 1985 ranged from 1,121 to 7,180 fish, with a median of 4,179 fish; Jennings 1985, Thompson et al. 1986). Generally, over 90 percent of the Chinook salmon that returned to the Middle River spawned in Indian River or Portage Creek. Peak spawner counts from 1976 to 1984 ranged from 114 to 1,456 fish (median 479.5 fish) in Indian River and 140 to 5,446 fish (median 680.5 fish) in Portage Creek (Jennings 1985).

ADF&G used mark-recapture techniques to estimate escapement to various fishwheel stations. Total escapement, as estimated from point estimates, to Sunshine Station ranged from 52,900 to 185,700 fish, with a median 103,614 fish, from 1982 to 1985 (Barrett 1984, Barrett 1985, Thompson et al. 1986). Escapement to Talkeetna Station ranged from 10,900 to 24,591 fish (median 14,400 fish). However, this has been considered an overestimate, because many Chinook salmon tagged at the Talkeetna Station were found to have spawned in tributaries downstream of Talkeetna Station (Jennings 1985). The large difference between these two stations, especially considering the overestimate at Talkeetna Station, reflects the large number of fish that return to the Deshka River.

Declines in returns of Chinook salmon have prompted the Alaska Board of Fisheries to list some Susitna River tributary stocks as Stocks of Concern. These include the Alexander Creek stock, which was listed as a “Management Concern” in 2011, and the Willow Creek and Goose Creek stocks, where were listed as “Yield Concern” in 2011. Low returns to the Deshka River in 2007 through 2009 have also prompted concern, and in 2012, low returns resulted in an early closure to the sport fishery.

From June through September of 1982, a total of 963 juvenile Chinook salmon were captured by all gear types at DFH sites from Goose Creek 2 upstream to Slough 21 (Estes and Schmidt 1983). Total juvenile Chinook salmon catch from this effort is shown by gear type and site in Figure 1.3-4.

Sampling from May 1 to November 15, 1983 at Juvenile Anadromous Habitat Study sites resulted in the capture of 4,443 juvenile Chinook salmon between the Chulitna River (RM 98.6) and Portage Creek (RM 148.8; Dugan et al. 1984). Relative abundance by season and site determined from this effort is shown in Figure 1.3-5. Juvenile Chinook salmon were captured at

all study sites that were surveyed at least four times. Peak densities of 26.4 fish per cell were recorded at tributary sites.

1.3.2.2.5. *Habitat Associations*

Adult Chinook salmon in the Upper, Middle and Lower Segments were observed to spawn almost exclusively in tributaries during the 1980s, with some occasional use of tributary mouths (Barrett et al. 1983, Jennings 1985, Thompson et al. 1986). Chinook salmon spawning was not documented in main channel habitats from 1981 to 1985, although this may be due to the fact that surveys conducted from 1983 to 1985 did not specifically target Chinook salmon (Barrett et al. 1983, ADF&G 1984, Jennings 1985, Thompson et al. 1986). In 1981, mainstem surveys were performed from July 15 to August 15 and covered 37 and 280 sites in the Middle and Lower River segments, respectively (Barrett et al. 1983). In 1982, mainstem spawning was monitored at 397 sites in the Middle River Segment and at 811 sites in the Lower River Segment from August 1 to October 7, which was later than most observed spawning in tributaries (Barrett et al. 1983). Chinook salmon spawning was observed at tributary mouths in 1982 in the Middle Susitna at Cheechako Creek (RM 152.4) and Chinook Creek (RM 157) but was not documented at similar habitats elsewhere in the Upper, Middle, or Lower Segments (Barrett et al. 1983, Barrett et al. 1985, Thompson et al. 1986).

Most juvenile Chinook salmon in the Susitna River typically exhibit either of two freshwater life history patterns. One group of Chinook salmon fry rear in their natal tributary for nearly one year prior to emigrating to the ocean as age-1+ smolts, while a second group of Chinook salmon disperse from natal tributaries throughout the spring and summer to the Susitna River's main channel, side channel, and slough habitats in the Middle and Lower River segments (Roth and Stratton 1985, Stratton 1986). Winter studies during the 1980s suggest that most Chinook salmon fry utilize the Lower River Susitna as winter nursery habitat (Stratton 1986). A third freshwater life history pattern, in which juvenile Chinook salmon emigrate to the ocean as age-0+ smolts, was exhibited by very few juvenile Chinook salmon during the 1980s studies and was associated with high ocean mortality rates based on adult scale analyses (Barrett et al. 1985, Roth and Stratton 1985, Suchanek et al. 1985). Age analysis of adult Chinook salmon scales in 1985 indicated that 5 percent of the fish sampled had emigrated as age-0+ smolts (Thompson et al. 1986).

Primary nursery habitats in the Middle Susitna River for juvenile Chinook salmon during the open water season were tributaries, tributary mouths, side channels, and side sloughs (Dugan et al. 1984). Clearwater side channels and sloughs influenced by groundwater sources provided juvenile overwintering habitat (Roth and Stratton 1985). Middle Susitna River sites with high juvenile Chinook salmon use were: Portage Creek (RM 148.8), Indian River (RM 138.6), side channels 10 (RM 133.8) and 10A (RM 132.1), and Whiskers Creek Slough (RM 101.2; Figure 1.3-6; Dugan et al. 1984). In the Lower Susitna River, tributary mouths and side channels were the primary nursery habitats used by juvenile Chinook salmon, and there appeared to be a preference for low-turbidity (i.e., 10-20 NTU) sites (Suchanek et al. 1986).

1.3.2.3. *Rainbow Trout (*Oncorhynchus mykiss*)*

1.3.2.3.1. *General Life History*

Rainbow trout are native to both Asia and North America but have been widely introduced throughout the world. Their distribution in North America ranges from northwest Mexico to the Kuskokwim River in Alaska (Mecklenburg et al. 2002). In Alaska, native populations extend from the Alaska panhandle along the coastline north to the Kuskokwim River and west to the Point Moller region of the Alaska Peninsula (Mecklenburg et al. 2002). Rainbow trout have been introduced in several lakes located in the interior of Alaska near Fairbanks, including Big Delta and Summit Lake (Morrow 1980). Rainbow trout inhabiting the Susitna River represent one of the northernmost naturally-occurring populations of the species (Morrow 1980).

Resident rainbow trout are spring spawners. Spawning takes place between mid-April and late June when adults deposit eggs and milt into redds. Unlike other Pacific salmon species, rainbow trout are iteroparous (i.e., able to breed multiple times) and do not die shortly after spawning. Repeat spawning is common for resident rainbow trout (Quinn 2005), and annual spawning may occur for up to 5 consecutive years for some fish (Morrow 1980).

Incubation typically lasts from 4 to 7 weeks, depending on water temperature. Fry emergence occurs within 3 to 7 days, usually between mid-June and mid-August (Morrow 1980). After emergence, rainbow trout fry may quickly disperse to lake habitats or remain in natal streams for up to 3 years (McPhail and Lindsey 1970; Scott and Crossman 1973). Rainbow trout mature at an age of 3 to 5 years and may live for up to 9 years (Morrow 1980).

Rainbow trout can be either stream- or lake-resident fish. When in rivers and streams, rainbow trout are commonly found near lake outlets or below waterfalls and rapids (McPhail and Lindsey 1970). Tributary streams are used as spawning habitat by both stream- and lake-resident populations (Morrow 1980). Redds are often constructed in fine gravel substrates of riffles located adjacent to pools. Preferred water temperatures for spawning and incubation are between 10°C and 13°C, and groundwater upwelling and dissolved oxygen concentrations are important in determining egg survival rates (McPhail and Lindsey 1970; Morrow 1980). Juveniles from stream-resident populations occupy riffles during summer months and tend to shift into pools for autumn and winter months (McPhail and Lindsey 1970).

Rainbow trout are opportunistic predators that feed on a wide variety of prey items, including various insects (e.g., dipteran larvae and adults), plankton, crustaceans, snails, leeches, fish eggs, smaller fishes, and adult salmon carcasses (Morrow 1980, Quinn 2005, Scott and Crossman 1973).

1.3.2.3.2. *Periodicity*

Rainbow trout spawning migrations typically begin in March prior to ice breakup when adults move from main channel holding areas to spawning tributaries (Sundet 1986). Migration timing into clear, non-glacial tributaries used for spawning was observed in April and early May during the 1980s studies, while most spawning occurred during late May and early June (Schmidt et al. 1983; Suchanek et al. 1984; Sundet and Pechek 1985). Migration and spawn timing for rainbow trout appears to be similar between the Middle and Lower Susitna River segments, although timing of upstream migration into tributary habitats was noted to occur up to 10 days earlier in

the Lower Segment (Sundet and Pechek 1985). Rainbow trout located upstream of the Chulitna River confluence (RM 98.6) begin to migrate to tributary habitats to spawn in late May and early June (Schmidt et al 1984).

Adult rainbow trout reside primarily in tributary habitats during the open water season, but they may also use tributary mouths and clearwater side sloughs throughout the Middle River Segment for holding and feeding during summer (Schmidt et al. 1983). In 1983 and 1984, adult migration from tributary habitats occurred during late August and September, such that many individuals had moved to tributary mouths by mid-September, and few remained in tributaries by early October (Suchanek et al. 1984; Sundet and Wenger 1984; Sundet and Pechek 1985). Migration timing to overwintering areas in main-channel and side channel habitats occurred from mid-September through early February, with peak movement in October and late December (Schmidt and Estes 1983; Sundet 1986). October movement was in response to freeze-up as fish sought winter holding habitats in the main channel (Sundet 1986). By December, most adult rainbow trout were in main channel areas apart from spawning tributaries (Sundet and Wenger 1984).

There is minimal information related to rainbow trout incubation and emergence timing in the Susitna River; however, incubation is assumed to begin in May based on observed spawn timing (Schmidt et al. 1983; Suchanek et al. 1984; Sundet and Pachek 1985). Based on generalized incubation times for rainbow trout in cold water temperature regimes (e.g., 5-8° C), the start of rainbow trout fry emergence in the Susitna River's tributary habitats is estimated to occur in early July and continue through mid-August (Quinn 2005; Crisp 1988, 1991). After emergence, juvenile rainbow trout primarily reside in natal tributary habitats throughout the year, though occasional use of tributary mouths and clear sloughs has been documented (Schmidt et al. 1983).

1.3.2.3.3. *Distribution*

Within the Susitna River, rainbow trout populations are found up to and including Portage Creek at RM 148.8 (ADF&G 1983m). No rainbow trout have been identified upstream of Devils Canyon in the impoundment zone (FERC 1983). These results are consistent between the 1980s and 2012 studies. Rainbow trout in the Susitna River are distributed throughout tributary and mainstem areas downstream of Devils Canyon (RM 152; Schmidt et al. 1983). Upstream of the Chulitna River confluence (RM 98.6), Whiskers Creek (RM 104.4), Lane Creek (RM 113.6), and Fourth of July Creek (RM 131.1) are the major spawning areas, whereas the larger tributaries (e.g., Indian River and Portage Creek) are of lesser importance (Schmidt et al. 1984). Primary spawning tributaries in the 1980s were Fourth of July and Portage creeks in the Middle Susitna River Segment and the Talkeetna River (RM 97.2), Montana Creek (RM 77.0), and Kashwitna River (RM 61.0) in the Lower Susitna River Segment (Sundet and Pechek 1985). Primary holding and feeding locations for rainbow trout were the Fourth of July Creek (RM 131.1) and Indian River (RM 138.6) tributary mouths, Slough 8A (RM 125.1), and Whiskers Creek Slough (RM 101.2; Schmidt et al. 1983).

1.3.2.3.4. *Relative Abundance*

Data collected in the 1980s indicate that adult rainbow trout are more abundant in the Middle Segment of the Susitna River than in the Lower Segment (Schmidt et al. 1983). Based on a tag-recapture study conducted from 1981 to 1983, the estimated abundance of rainbow trout greater than 150 mm in FL in the Middle Segment was approximately 4,000 fish (Sundet and Wenger

1984). In the Lower River in 1984, a total of 155 rainbow trout were captured using multiple capture methods (Sundet and Wenger 1984). The highest number of rainbow trout captures (i.e., 62 fish) occurred in the Deshka River. Relatively high catches were made by boat electrofishing in the mainstem Susitna River between RM 30.0 and RM 98.5 in early September (31 fish captured) and at the mouth of Little Willow Creek (RM 50.3) in late September (14 fish captured). Only nine rainbow trout were captured in the upper reaches of east side tributaries during early September (Sundet and Pechek 1985).

Sampling at the DFH sites in 1982 resulted in the captured of 207 rainbow trout (Figure 1.3-7; Schmidt et al. 1983). The largest number of rainbow trout captured (n=43) was at the Fourth of July Creek site. Other DFH sites where more than 20 rainbow trout were captured included Whiskers Creek and Slough, Slough 8A, and Indian River. Whitefish Slough was the only DFH site sampled in 1982 at which no rainbow trout were caught.

From May to October 1983, sampling at 12 selected sites between the Chulitna River confluence and Devils Canyon captured 163 rainbow trout (Sundet and Wenger 1984). The highest catches were at Fourth of July Creek (RM 131.1) and Indian River (RM 138.6), where 46 and 45 fish were caught respectively. Other sites with relatively high catches included Whiskers Creek Slough (RM 101.2), Lane Creek (RM 113.6), and Portage Creek (RM 148.8). Sampling at locations other than the twelve selected DFH sites resulted in the capture of 228 rainbow trout, with 78 percent of these fish captured in the lower 1.5 miles of Fourth of July Creek. The highest catches of rainbow trout in tributary streams of the Susitna River were recorded in Fourth of July Creek, where significant spawning activity was documented (Sundet and Wenger 1984).

Rainbow trout were also documented in lakes within the Susitna River basin; a total of 390 fish were captured in six lakes surveyed in 1984, comprising 86 percent of the total fish catch (Sundet and Pechek 1985). Lakes in which rainbow trout were abundant in 1984 include those that flow into Fourth of July and Portage creeks (Sundet and Pechek 1985).

1.3.2.3.5. *Habitat Associations*

Rainbow trout in the Susitna River are distributed throughout tributary and mainstem areas downstream of Devils Canyon (RM 152; Schmidt et al. 1983). Upstream of the Talkeetna River, they mainly use tributaries for spawning and rearing, while overwintering occurs primarily in the mainstem (Schmidt et al. 1984). Upstream of the Chulitna River confluence (RM 98.6), the major spawning areas are Whiskers Creek (RM 104.4), Lane Creek (RM 113.6), and Fourth of July Creek (RM 131.1); larger tributaries (e.g., Indian River and Portage Creek) appear to be of less importance with regard to rainbow trout spawning (Schmidt et al. 1984).

Adult rainbow trout utilize clearwater tributary habitats to spawn following ice breakup each spring (Schmidt et al. 1983). After spawning, adults primarily hold and feed during the open water period in tributary and tributary mouth habitats, although some utilization of clearwater side slough habitat was observed during the 1980s (Schmidt et al. 1983). Holding and feeding areas during the open water period were closely associated with Chinook, chum and pink salmon spawning areas (Sundet and Pechek 1985). Primary holding and feeding locations for rainbow trout were the Fourth of July Creek (RM 131.1) and Indian River (RM 138.6) tributary mouths, Slough 8A (RM 125.1), and Whiskers Creek Slough (RM 101.2; Schmidt et al. 1983).

Prior to ice formation on the Susitna River, adult rainbow trout move from tributaries to main channel or side channel habitats to hold during winter (Schmidt and Estes 1983, Sundet and

Pechek 1985). In the Middle Segment, rainbow trout were found to utilize main channel areas, but in the Lower Segment, they typically used side channel habitat (Sundet and Pechek 1985). Movement from spawning or feeding tributaries to overwintering habitat is commonly in a downstream direction (Sundet and Pechek 1985). Many adults overwinter relatively close (i.e., <4 miles) to spawning tributaries, while others exhibit long-distance migrations that typically range from 10 to 20 miles downstream but can extend over 76 miles (Schmidt and Estes 1983, Sundet 1986). Winter holding areas include main channel and side channel habitat (Schmidt and Estes 1983, Sundet 1986). Specific habitat features of winter holding areas during the 1980s were difficult to ascertain, though upwelling and ice cover appeared to be common in fish habitat (Schmidt et al. 1983, Sundet and Pechek 1985, Sundet 1986). No tagged fish were observed in areas with anchor ice (Sundet 1986). Limited observations of tagged rainbow trout suggest the Susitna River between RM 78.0 and Talkeetna may also be an important overwintering area for Talkeetna River stocks (Sundet and Wenger 1984).

Juvenile rainbow trout generally utilize natal clearwater tributaries as nursery habitats (Schmidt et al. 1983). Some juveniles also rear in the mainstem and sloughs, but the use of these habitats appears to be limited (ADF&G 1983b, Schmidt et al. 1984). Fourth of July Creek (RM 131.1) is an important rearing area for juvenile rainbow trout (Schmidt et al. 1984). Capture of juvenile rainbow trout in main channel areas was low, though use of tributary mouths and clearwater sloughs was observed (Sundet and Pechek 1985). Lake systems associated with the Fourth of July and Portage creeks were believed to possibly supplement rainbow trout production in each basin based on analysis of juvenile scale patterns; however, no direct evidence of juvenile rearing in these lakes was recorded (Sundet and Pechek 1985). Winter rearing for juvenile rainbow trout occurred primarily in tributaries with occasional use of clear side slough habitats (Schmidt et al. 1983).

1.3.3. Middle River Mainstem Habitat Delineation Results

In winter 2012-2013, the frequency and proportion of habitat in the mainstem Middle River was delineated using geo-rectified aerial imagery in combination with available aerial videography. The objective of Middle River mainstem mapping was to characterize and classify river habitat in the Middle River mainstem from the Chulitna River confluence to the proposed Watana Dam site. These data were used to support the selection of representative focus areas for instream flow studies and the approach for fish distribution and abundance site selection.

A hierarchical and nested classification system developed specifically for the Susitna River with input from the Fish and Aquatics Technical Working Group was used to classify habitat to the mainstem habitat level. The geo-rectified imagery in combination with aerial videography was sufficient to map the Middle Susitna River mainstem habitat to the mesohabitat level. However, the imagery was not suitable for mapping off-channel or tributary habitats to this level. Thus, these habitats were delineated only to the level of mainstem habitat types in 2012(HDR 2013).

A summary of these results can be found in the Middle Susitna River Segment Remote Line Habitat Mapping Technical Memorandum (HDR 2013).

Main channel habitat varied by geomorphic reach within the Middle River Segment and generally increased in complexity from upstream to downstream locations. Mesohabitat in the main channel was generally dominated by a mixture of run and glide habitats. Glide and run habitats, which were not distinguished from each other at this level of classification, included

smooth-flowing, low-turbulence reaches as well as areas with some standing or wind waves and occasional solitary protruding boulders. Run-glide mesohabitat dominated all reaches except MR-4, where Devils Canyon is located. Riffle habitat was most prevalent in MR-4. Riffle habitat was lacking or found in very small amounts in the other Middle River geomorphic reaches.

Side channels were predominantly glide or run, with some riffle areas in the lower reaches. Many side channels were not completely inundated with flowing water and so identification of riffle or run habitat was not possible; these were classified as unidentified and were most prevalent in MR-6.

Cascade habitat was not found within any of the geomorphic reaches of the Middle River Segment. The geomorphic reach through Devils Canyon (i.e., MR-4) contained the only rapids in the Middle River, which accounted for 38 percent of the mainstem habitat in that reach. Only 3 pools were found in the Middle River, and all were located in MR-4 between rapids in Devils Canyon.

The habitat associated with the confluence of tributaries with the main channel river was documented as tributary mouth and clear water plume. Not all tributaries that entered the Middle River had tributary mouth habitat. Small tributaries where the vegetation line was close to the mainstem did not fan out and create the areas classified as tributary mouth habitat. In addition, small tributaries or tributaries that flowed into fast moving or turbulent sections of the mainstem did not produce clear water plume habitats. Clear water plume habitats were located in reaches MR-2, MR-3, MR-5, and MR-7, with the highest number in reach MR-2.

Off-channel habitat was assigned to one of three habitat types observed: upland sloughs, side sloughs, and backwaters. Upland and side sloughs were prevalent throughout the Middle River reaches outside of Devils Canyon and downstream of the uppermost reach at MR-1. Side sloughs were most abundant in MR-5, followed by MR-6. Upland sloughs were most abundant in MR-8, and generally increased in abundance towards the downstream reaches (Table 5). Backwater habitat was relatively rare and found in a few areas in the lower reaches from MR-6 through MR-8. A single backwater was also delineated in MR-2 and in MR-4, but each accounted for less than 1 percent of the linear habitat within their respective reaches. The greatest total area of backwater habitat was in MR-7, but the greatest frequency was found in MR-6.

Beaver complexes were consistently associated with slough habitats and as such were not categorized as a habitat type but were noted as a characteristic of that slough habitat unit. Beaver dams were rarely present in side slough habitat, and slightly more prevalent in upland sloughs. Beaver dams were only observed in reaches MR-6 and MR-7.

1.3.4. Documentation of TWG input to site selection protocol

This section will be provided in Final Implementation Plan, following February 2013 consultation.

2. METHODS

2.1. Sampling Site Selection Protocols

Sampling for the River Productivity Study will be stratified by river segment and mainstem habitat type, as defined in the Project-specific habitat classification scheme (e.g., main channel, tributary mouth, side channel, side sloughs). Sampling will occur at six stations on the Susitna River, and one station on the Talkeetna River, each station with three sites (one main channel site and two different macrohabitat sites associated with the main channel site), for a total of 18 sites. Two stations will be located in the Upper Segment, above the proposed dam and reservoir area (upstream of RM 223) (Table 1.2-1; Figure 1.2-1). In the Middle River Segment, two stations will be located between the dam site and the upper end of Devils Canyon, and two stations will be located between Devils Canyon and Talkeetna (Table 1.2-1; Figure 1.2-2). All stations established within the Middle River Segment will be located at Focus Areas established by the Instream Flow Study (RSP Section 8.5.4.2.1.2), in an attempt to correlate macroinvertebrate data with additional environmental data (flow, substrates, temperature, water quality, riparian habitat, etc.) collected by other studies (e.g., RSP Section 5.5, Baseline Water Quality), for uses in statistical analyses, and HSC/HSI development. Many of these Focus Areas are also highly utilized by the target fish species selected for this study's trophic analysis (RSP Section 9.8.4.5.1). Station and site locations are discussed below.

2.1.1. Middle River Segment Stations / Focus Areas

Within the Middle Segment of the Susitna River, the River Productivity Study will establish four study stations, each one located at a proposed Focus Area (Table 1.2-1; Figure 1.2-2 and Figures 2.1-1 through 2.1-4). For sampling between the proposed dam site and Devils Canyon, Focus Areas 184 and 173 have been selected for River Productivity Study activities. Between Devils Canyon and Talkeetna, Focus Areas 141 and 104 have been selected as stations for the River Productivity Study. In addition, two side sloughs are required for proposed storm event sampling. After review of historic data (ADF&G 1983c; Hale et al. 1984) regarding the mainstem discharge required to overtop various sloughs in the Middle Segment, sloughs in Focus Areas 104 and 144 have been selected for storm event sampling.

Focus Areas 184 and 173 have been selected due to their proximity to the location of the proposed dam site. Any effects on the benthic macroinvertebrate population due to Project operations would be most pronounced at these upper locations. Therefore, monitoring this area during pre-Project operations is critical for establishing baseline conditions. Focus Area 184 is located approximately 1.4 miles downstream of the proposed dam site and will provide a mainstem site and a side channel site within its 1-mile extent (Figure 2.1-1). In order to establish a third site with an additional habitat type, it will be necessary to move outside of the Focus Area to sample the mouth of Tsusena Creek. Focus Area 173 is located approximately 11.7 miles downstream from the proposed dam site and contains a complex of main channel and off-channel habitats within a wide floodplain, thus representing the greatest channel complexity within its geomorphic reach (MR-2; Figure 2.1-2). Focus Area 173 will provide a mainstem site, a side channel site, and a side slough site within its 1.8-mile extent.

Below Devils Canyon, Focus Areas 141 and 104 have been selected because of the diversity of main channel and off-channel habitats that they contain, and documented fish use in and nearby

these Focus Areas. Focus Area 141 includes the Indian River confluence, which is a primary Middle Susitna River tributary with documented high fish use. Focus Area 141 offers a range of main channel and off-channel habitat types and will thus provide a mainstem site, a tributary mouth site, and a side channel site within its 1.6-mile extent (Figure 2.1-3). Focus Area 104 is located approximately 3.3 miles upstream of the confluence of the Chulitna and Susitna rivers, making it the downstream-most station in the Middle Segment for the River Productivity Study. This Focus Area contains the confluence of Whiskers Creek, side channels, and side slough habitats that have been documented as supporting juvenile and adult fish use. Focus Area 104 will provide a mainstem site, a side slough site, and a side-channel site within its 1.2-mile extent (Figure 2.1-4).

For storm event sampling, Focus Area 104 was retained for study, and Focus Area 144 was additionally selected. Focus Area 144 is located approximately 2.3 miles upstream of the Indian River confluence, and features a side channel and a side slough complex (Figure 2.1-5). Both Focus Areas feature side sloughs that require similar levels of mainstem discharge for overtopping (ca. 22,000-25,000 cfs), and both side sloughs maintain at least some of wetted habitat during the summer months (ADF&G 1983c; Hale et al. 1984).

2.1.2. Upper Segment Stations

Two stations will be established in the Upper Segment of the Susitna River and above the upper limit of the proposed Project's maximum reservoir pool. After review of the results of 2012 data collection efforts, as well as information collected from ADF&G fish inventories in the Upper Susitna River (Buckwalter 2011), few details on habitat types present in the Upper Segment of the Susitna River are available. The data that is available is limited to habitats within the Oshetna River and downstream.

As a result, the two stations in the Upper Segment of the Susitna River have been selected using existing topographic maps and available orthographic imagery to review river features. A site reconnaissance trip will be used to confirm the selected stations (Table 1.2-1, Figures 2.1-6 and 2.1-7). The downstream-most station (tentatively designated RP-233) is a 1.5-mile reach containing the mouth of the Oshetna River, in which ADF&G has recently documented the presence of adult and juvenile Chinook salmon (Buckwalter 2011; ADF&G 2011). Station RP-233 will provide a mainstem site, a tributary mouth site, and a possible side channel or slough, although this will need to be confirmed during site reconnaissance (Figure 2.1-6). In the event that no additional habitat types are available, the study will set up two main channel sites, with one above the confluence with the Oshetna River, and another below. The second station established in the Upper Segment (RP-248) will be located within a 1.3-mile reach beginning approximately 1.25 miles upstream of the Tyone River confluence. Imagery and maps indicate this area is complex, with split channels, side channels, and possibly side sloughs (Figure 2.1-7). Station RP-248 will provide a mainstem site, side channel site, and a side slough site, and will be confirmed with site reconnaissance.

2.1.3. Talkeetna Station

One task within the River Productivity Study assesses the feasibility of the Talkeetna River as a reference site for post-Project monitoring activities at these stations. Because the Talkeetna River is outside of the Project area, results from 2012 study efforts and historic information from

the 1980s are limited. Review of the literature has revealed a single USGS study which reports on water quality and benthic macroinvertebrate data collected from the Talkeetna River, approximately 5 miles upstream from its mouth near a USGS gaging station (Frenzel and Dorava 1999). The USGS sampling reach was limited to the main channel, with benthic macroinvertebrate sampling taken off a cobble point bar.

The ideal station on the Talkeetna River for the feasibility study will be a match with physical conditions similar to one of the Focus Areas selected in the Middle Segment of the Susitna River. The Talkeetna station will feature both main channel and off-channel habitat types to allow for the establishment of a main channel site, a side channel site, and a side slough site. Habitat types have not been identified for the Talkeetna River, so station selection for the feasibility study will be limited to an initial review of topographic maps and available orthographic images. Final site selection will be made with a site reconnaissance trip.

2.2. Benthic Macroinvertebrate Sampling

Three sampling periods (also known as Index Periods) will occur from April through October in both study years (2013-2014) to capture seasonal variation in community structure and productivity. These seasonal periods are tentatively scheduled for April through early June for Spring, late June through August for Summer, and September through October for Autumn. In addition, benthic sampling will be conducted both before and after storm events that increase flows to levels similar to pulse flow increases from the proposed Project (ca., 5,000 cfs) at two side slough sites, located in Focus Areas 104 and 144. Specific details on timing of sampling are provided in Section 3 below.

2.2.1. Field Sampling Protocols

Benthic macroinvertebrate sampling will be conducted in fast-water mesohabitats (typically riffles/runs) within main channel (i.e., main channel, side channels, and tributary mouths) and off-channel macrohabitat types (i.e., side sloughs). Measurements of depth, mean water column velocity, mean boundary layer velocity (near bed), and substrate composition will be taken concurrently with benthic macroinvertebrate sampling at the sample location. While a benthic macroinvertebrate sample is being collected by one crew member, two other crew members will be collecting the associated benthic algae samples (Section 2.3) and associated habitat measurements (e.g., depths, velocities, etc.).

Water temperatures will be monitored using submerged temperature loggers at hourly intervals and deployed throughout the ice-free season. Temperature and flow monitoring will be coordinated with the Baseline Water Quality Study (RSP Section 5.5) and the Instream Flow Study (RSP Section 8.5), and supplemental temperature loggers will be deployed, if necessary, at all River Productivity Study sites.

Higher flows may inundate new shoreline substrates, which present a risk of sampling in areas that are not fully colonized. The shoreline bathymetry for each site will be evaluated such that changes in water level due to increasing or decreasing flows must remain constant enough that the substrates accessible for sampling will be continually inundated for a period of at least one month to facilitate colonization of those substrates. At each sampling location (see Section 2.2.1.1), a basic transect will be established, perpendicular to the shoreline. A stake or pin will be hammered into the riverbank at the high water mark. Measurements of depth and velocity

will be taken every meter, out to the sampling location, and up to 5 meter past it or up to a depth of 1 meter, whichever occurs closest to the shoreline. This localized transect information will be used in conjunction with multiple remote cameras and staff gages installed along the Susitna River, along with the USGS gage at Gold Creek, to closely monitor for conditions that indicate adequate inundation levels for sampling. Depth and velocity measurements will also be taken at 1-meter intervals both upstream and downstream of the Hess sample location, out to a distance of 5 meter or a depth of 1 meter. All depth and velocity information will be recorded on the field data sheet for the sample entry.

2.2.1.1. Hess Samples

Sampling will be conducted using a modified Hess sampler (0.086 m²-area) with a 243 micrometer (µm) mesh net (Canton and Chadwick 1984; Klemm *et al.* 1990). The modified Hess sampler is commonly used in benthic macroinvertebrate studies (Barbour *et al.* 1999; Klemm *et al.* 1990; Klemm *et al.* 2000; Carter and Resh 2001) including previous Susitna River studies from the 1980s (Hansen and Richards 1985, Trihey 1986). The modified Hess sampler is an enclosed cylinder 40 cm in height and 33 cm in diameter, with a screened opening in the front, and receiving mesh net bag opposite it (Figure 2.2-1). The cylinder is forcibly pushed and rotated into the substrate to depths of 7.5-15 cm, and all substrate within the enclosed 0.086 m² area is cleaned of macroinvertebrates. Water flows in through the upstream window and out the downstream window into the collecting net and bucket. The modified Hess sampler is easy to use, yet also prevents escape of organisms, which is an issue with other sampling devices, such as the Surber sampler and kick nets. The sampler also prevents drifting organisms and materials from entering the sample, which is an important factor in sampling the organic matter components for this study (Section 2.4). Replicate samples (n=5) will be collected to allow for statistical testing of results for short- and long-term monitoring. The following is the procedure for collecting Hess samples.

1. Walk the length of the sampling reach, identifying all locations that would be suitable for sampling. A suitable sampling area will be in fast-water habitat, offering coarse substrates at water depths of approximately 40 cm or less and which have been inundated for at least 30 days. The most ideal locations are likely to be shoreline reaches that offer larger areas of large gravel and cobble substrates. Select five of the suitable locations, spacing them as equidistantly as possible, to be representative of the site. If five unique and separate locations are not available, it will be necessary to collect more than one sample within the same location. If this is the case, space the sample locations out as far as possible. For example, if conditions require two samples in one riffle area, sample at the downstream end and then the upstream end. As a general rule, samples should not be taken within 10 m of each other. Selected locations at each site should be sampled in a downstream-to-upstream direction.
2. After selecting a sample's location, measure and record the depth, mean water column velocity (60 percent of depth), mean boundary layer velocity (near bed), and a visual estimate of substrate composition using the Wentworth scale, to the nearest 5 percent, where the sample is taken.
3. Position the Hess sampler securely on the river substrate at the chosen location, and slowly twist the bottom of the frame into the substrate. For streambeds comprised of larger cobbles, this may not be possible. As a general rule, larger substrates that are more

than 50 percent inside the sampling area should be lifted and moved into the sampler, and those less than 50 percent inside should be excluded. If complete containment of the sampling area cannot be accomplished, a neoprene skirt may be to be used along the bottom of the sampler.

4. Ensure the screened opening is facing into the current and the net portion is trailing downstream. Hold the sampler in position between your legs, applying pressure with your knees. Do not to disturb the substrate upstream from the sampler, as that area will supply the algal samples (Section 2.3) to associate with this Hess sample.
5. Reach into the cylinder and carefully turn over and lightly hand-scrub all large substrates, which will dislodge macroinvertebrates clinging to the stones and wash them into the net bag. Examine each rock for organisms, including larval or pupal cases that may be attached to it before removing it from the sampler.
6. Stir remaining finer substrate with your hands to a depth of 5 to 10 cm, to dislodge all remaining organisms, which will be collected in the mesh net.
7. To prevent the contents of the mesh net bag from washing out, slowly lift the sampler out of the substrate and the water, tilting the sampler so that the net bag's opening is oriented up.
8. Wash any debris and organisms clinging to the net bag down into the cod-end collection bucket by splashing river water on the outsides of the bag, or by lowering the bag into the stream and quickly removing it.
9. Return to shore and carefully remove the cod-end collection bucket from the sampling net over a 250- μ m sieve, and empty the contents into the sieve.
10. Carefully examine the mesh net bag of the Hess sampler for any clinging organisms and remove them with forceps, and place them with the rest of the sample in the sieve.
11. Examine the contents in the sieve. Closely inspect all large materials for attached invertebrates. Keep all organic matter; it will be needed for assessment of organic matter content (Section 2.4). Discard all larger inorganic materials.
12. Rinse the sample in the sieve, consolidating the material to one side of the sieve, and transfer the material into a storage container. Efforts should be taken to minimize the amount of water retained with the sample to prevent too much dilution of the ethanol used to preserve the sample. Next, scoop out the material with a spoon or spatula and place it in the sample container, and then rinse the sieve to consolidate the remaining material to one side of the sieve. Wash the remaining sample into the container with a wash bottle containing 95 percent ethanol.
13. A paper label (standard label hereafter) defining the station, site, sample number, date, collector, and unique sample identification code is added to the sample. Adhesive standard labels are also applied to the outside of the sample jars.
14. Preserve the sample with additional 95 percent ethanol, enough to completely cover the sample, and place the labeled lid on the container, making sure it is secured tightly and does not leak.
15. Rinse the Hess sampler net and cod-end collection bucket and reassemble.
16. Move the sampler upstream to the next riffle/run identified, and repeat this process. Continue until five replicate samples have been sampled, each upstream from the last.

2.2.1.2. Snag Samples

Due to the prevalence of large woody debris in the Susitna River, woody snags, if present at a sampling site, also will be sampled as a substrate stratum for benthic macroinvertebrates. Sampling methods for woody snags will be semi-quantitative, based upon protocols established by the USGS (Moulton et al. 2002). Moulton et al. (2002) defines wood snags as “submerged sections of wood (branch or log) having a minimum diameter of 1 cm and are colonized by aquatic organisms.” The following is the procedure for sampling wood snags.

1. Identify five (if present) woody snag locations throughout the site area. Suitable woody snags will have been submerged for an extended period of time so as to be clearly colonized.
2. At each wood snag location, identify a woody snag piece to remove from the water. Ideal snag pieces should be at least 5 cm in diameter. Sections of larger branches may need to be removed, using a hand saw. Ideally, each snag section will originate from a separate snag, and therefore count as a separate, replicate sample.
3. Measure and record: the depth of the snag piece, both from the water surface and from the stream bed; the current velocity at the snag piece and in the water column (60 percent of depth); and visually estimate the substrate composition using the Wentworth scale, to the nearest 5 percent, where the woody snag is positioned. This information is written on the field data collection worksheet for the site.
4. Place a collection net downstream of the woody snag piece, to capture and minimize loss of mobile or loosely attached invertebrates.
5. Remove the woody snag piece from the water, using a saw or lopping shears. Each woody snag piece removed from the river should be closely inspected for evidence of colonization and discarded if it appears to have recently fallen into the river.
6. Place the woody snag piece, lengthwise, over a plastic bin (e.g., a 10-Gal Rubbermaid™ storage container) to delineate the sampling area. Initially, loosely attached insects are rinsed from the surface of branches with a wash bottle or pump sprayer. The snags will be allowed to dry for a period of time (usually 1 hour), insects will begin moving from retreats and dropping into the container. Meanwhile, specimens should be picked from attached detritus, retreats, and cracks using fine-tipped forceps.
7. As desiccation begins to hinder insect movement, rather than encourage it, the woody snag piece should be lightly rinsed. The total amount of time spent collecting specimens from each sample is approximately 1-1.5 hours.
8. The invertebrates, associated debris, and water within the sample containers are agitated and poured through a 125- μ m sieve. The container should be rinsed, agitated, and sieved several times and any remaining specimens should be picked from the container by hand. The contents of the sieve are then washed into a 500-ml wide-mouth plastic jar with a wash bottle containing 95 percent ethyl alcohol. Any insects clinging to the sieve are transferred to the sample jar with forceps before filling the container with alcohol to completely cover the sample material.
9. A standard label defining the station, site, sample number, date, collector, and unique sample identification code is added to the sample. Adhesive standard labels are also applied to the outside of the sample jars. Samples from each site are numbered consecutively, 1-5.

10. After final inspection, snag sections sampled will be measured for length and average diameter to determine surface area sampled. The length of branch, or section of branch, within the containers is measured, as is the circumference at both ends, to estimate the surface area for each woody snag sample. This information is written on the field data collection worksheet for the site.

2.2.2. Sample Processing Protocols

Benthic macroinvertebrate samples from both Hess and snag samples will be sent to one or more accredited contract laboratories for subsampling, sorting, and taxonomic identification. Laboratories should have taxonomists on staff that are certified by the Society for Freshwater Science for taxonomic identifications of specific groups (EPT taxa, chironomids, etc.). Sample processing protocols should follow those established by the USEPA for the Rapid Bioassessment Protocols (Barbour et al. 1999) and modified for use in Alaska (Major and Barbour 2001; see Appendix 1).

A gridded subsampling tray (Caton 1991) will be used to acquire a 300-organism fixed-count (± 20 percent) subsample. All invertebrates are removed from debris with the aid of a dissecting microscope (7-45x) and sorted into major taxonomic groups. As specified in Major and Barbour (2001), benthic macroinvertebrates should be identified to the lowest practical level. For aquatic insects, identifications are to the genus level, with exceptions for damaged or immature specimens. Non-insect taxa are identified to family or order.

Sorted debris is retained in a labeled, 60-ml bottle and stored for later for QA/QC assessment and, for Hess samples, organic matter analysis (Section 2.4). At the conclusion of the subsampling effort, a large-rare organism sort is performed on the unsorted portion of the sample to identify taxa that were not accurately represented in the sorted grids. All large organic material removed from the tray prior to subsampling should also be retained for organic matter analysis (Section 2.4).

Biomass estimates will be taken for invertebrate taxa collected for benthic sampling. The fresh blotted wet mass of invertebrate taxa in samples will be recorded. The samples will be oven dried at 60°C until reaching constant mass, and the dry mass will be recorded.

2.2.3. Data Analysis Methods

The end result of benthic macroinvertebrate sampling after the collection, processing, sorting, and identification of the various taxa, is the creation of a matrix of abundances. Those abundances can then be transformed into a variety of quantitative measures, called metrics, which represent different attributes of the structure, composition, or function of the benthic macroinvertebrate assemblage (Wang and Lyons 2003). Results generated from the collections will include several descriptive measures commonly used in aquatic ecological studies (Table 2.2-1). Each measure will have the mean and variability (95-percent confidence intervals) calculated. Comparisons for these measures will be made among sites, to look for differences between habitat types, as well as spatial trends along the length of the river (upstream versus downstream sites). Comparisons will also be made over time, examining both the interannual (seasonal) and annual variability in the benthic macroinvertebrate community. Statistical tests (ANOVA, MANOVA) will be performed on each measure to look for an overall significant difference among sites, seasons, and years. If a difference is significant ($p \leq 0.05$) for the

measure, then a multiple comparison test will be used to describe the significant differences in the data. Assumptions of normality and equal variance will also be tested with each ANOVA. In the event data does not meet distribution assumptions, it may be transformed (i.e., log+1 or square root) prior to analysis. If the data does not pass a test for normality, then a non-parametric test, like the Kruskal-Wallis one-way ANOVA on ranks, followed by a Kruskal-Wallis multiple comparison z-value test will be used. If the data fails the modified-Levene-equal-variance test, a series of unequal-variance-two-sample t-tests can be utilized to test for significant differences.

In addition, multivariate ordination procedures, such as principle components analysis (PCA) and canonical correspondence analysis (CCA), may be utilized to explain the relative contribution of different measures, taxa, and environmental variables to observed grouping patterns that best explain variability in the data. The goal of ordination is to preserve differences between samples, to reduce the dimensionality of the data, and to create a set of independent covariates from a set of correlated variables. The general approach is to define a new set of axes that describe the majority of the variability in the multivariate data. The first axis is a vector fitted to the direction of maximum variability in the data. Successive axes are orthogonal (perpendicular) to the existing axes, with each additional axis explaining a smaller portion of the total variation in the data. PCA is probably the most widely known type of ordination. CCA is a procedure to analyze two (or more) data tables simultaneously and is the preferred method for analyzing species and environmental data, and is particularly well suited to ecosystem-species questions (ter Braak 1986). Cluster analyses using the Bray-Curtis measure of dissimilarity may be used to group sites or stations by the similarity of their community structure.

2.3. Benthic Algae Sampling

2.3.1. Field Sampling Protocols

Benthic algae (also referred to as periphyton) will be sampled in conjunction with benthic macroinvertebrate sampling with the Hess sampler (Section 2.2). Rock surfaces in fast-water habitats are sampled, based on the methods utilized by the USGS for the NAWQA program (Moulton et al. 2002), the USEPA for the Rapid Bioassessment Protocol (Barbour et al. 1999), and the USEPA for the Environmental Monitoring and Assessment Program (EMAP; Lazorchak et al. 2000; Peck et al 2006). Methods employ an area-delimiting sampling device that is pressed to a smooth rock or cobble substrate to consistently sample a defined surface area. For the purposes of this study, a PVC pipe area delimiter with a rubber collar at one end, as recommended by the EPA methods, will be adopted (Barbour et al. 1999; Lazorchak et al. 2000; Peck et al 2006). In the event that the PVC pipe method proves unsuitable for use, other sampling approaches may be adopted (Moulton et al. 2002; Fetscher et al. 2009). Moulton et al. (2002) describe the SG-92, a modified syringe-sampling device, which performs best on smooth cobble surfaces with moderate-to-dense coverage of microalgal periphyton. Other approaches are the plastic frame of a 35-mm or medium format slide and a rubber mat with an opening. The slide frame is preferred by some, because it is more flexible and form-fitting than a section of PVC pipe or the barrel of a syringe. The rubber mat is likewise flexible with the added feature of covering the area outside of that delineated and when rinsed, reduces the potential for sample contamination (Fetscher et al. 2009).

The following is the procedure for sampling benthic algae.

1. Randomly collect five rock substrates distributed in the undisturbed area located upstream of each Hess sample being collected. At each location where a cobble or rock substrate is collected, measurements of depth, mean water column velocity, mean boundary layer velocity, and area substrate composition will be taken. Light availability will be measured at each sample location with an underwater light sensor to measure the photosynthetically-active radiation (PAR) available to the algal community. PAR readings will be taken from just below the water surface to the stream bottom at regular 10-cm intervals. A turbidity measurement, using a portable turbidity meter, will also be taken at the sample site to determine water clarity. Record all measurements on the field data sheet for the sampling site.
2. Place substrates in a plastic dishpan and transport them to the shoreline, in a shaded area if possible, to collect algae from each cobble.
3. Place the area-delimiting sampler on a substrate surface. Press down on the gasket/o-ring and rotate slightly to create a tight seal. Maintain this seal while the collection is made.
4. Using a pipette, squirt approximately 5 ml of filtered stream water into the sampler on the cobble. If the water leaks from the barrel, select another place on the substrate and try again. If the water does not leak, insert a small brush into the barrel and scrub the enclosed area on the substrate to remove the algae.
5. Remove the algae and water mixture with a pipette and dispense it in a 100-ml graduated cylinder. *[Note: dispensing into a graduated cylinder instead of a 500-ml sample bottle is recommended in case the sampler seal fails while collecting the sample, thereby causing the collector to start over. If the seal fails, then only the contents of the graduated cylinder are discarded.]* Repeat this process several times until all of the visible periphyton is removed. Pour the contents of the graduated cylinder into a 500-ml sample bottle. Alternatively, an aspirator device (Bouchard and Anderson 2001) can be used to remove algae materials from the area delimiter.
6. Repeat the sampling procedure for a single area on each of the cobbles selected; the composited sample is composed of 5 discrete collections taken from 5 cobbles. Ensure that the sample volume does not exceed 475 ml. Place the bottle on ice inside a cooler and keep in the dark until the sample is processed.
7. Measure the diameter of the area sampled by the sampler at the beginning and end of sampling. Record these diameters on the field data sheet to establish an average diameter from which the sampling area can be calculated.
8. Calculate the total sampling area by using the following formula:
9. Total sampling area (cm^2) = $(n)(\pi)(d/2)^2$
10. Where, n = number of discrete collections, π = 3.1416, and d = average diameter of the sampled areas, in centimeters. *[note: if using the inside diameter of a 5-cm PVC pipe, then the total surface area sampled for 5 cobbles will be about 98 cm^2 .]*
11. Process the periphyton sample following the steps described in section 2.3.2. Processing (below).

2.3.2. Processing Protocols

Benthic algae samples will be initially prepared and processed for additional laboratory analyses by subsampling. Processing can take place immediately in the field, or after coming out of the field later that day. In either case, care should be taken to avoid sample exposure to direct

sunlight. Procedures for processing are taken directly from the Quantitative Microalgae processing procedures detailed in Moulton et al. (2002). Moulton et al. (2002) state:

“The goal of processing a composited algal sample in the field is to prepare subsamples for various laboratory analyses. Successful execution of the processing procedures described here to produce high-quality subsamples for analysis is dependent on measuring and tracking various volumes as the sample is processed.”

Two subsamples are taken from each benthic algae composite sample for the purpose of determining chlorophyll-*a* and ash free dry mass (AFDM) in the laboratory. The remaining volume of the sample component is preserved, and archived for additional analyses, if needed. The following is the procedure for processing benthic algae samples for *chl-a* and AFDM.

1. Measure the sample volume to the nearest milliliter. Record on field data sheet.
2. Calibrate the pipette. [Note: the calibration is important, especially if the tip has been trimmed to enlarge the opening for extracting dense algal material.]
3. Assemble the chlorophyll filtration apparatus by attaching the filter base with rubber stopper to the filtering flask. Join the flask and a hand-operated vacuum pump (with gage) using a section of tubing
4. Place a 47-mm glass fiber filter (e.g., Whatman™ GF/F) on the filter base and wet with deionized water. [Note: wetting the filter will help keep it in place in windy weather. Attach the filter funnel to the base.]
5. Homogenize the composite algae sample. Invert the sample bottle 10 times and use a battery powered stirrer to break up the clumps. Cut algal filaments, if present, into pieces about 2 mm in length.
6. Shake the sample component vigorously for about 30 seconds to ensure that it is well mixed before extracting subsamples.
7. Extract two 5-mL aliquots of homogenized sample using the pipette and dispense onto the wetted glass-fiber filter.
8. Filter the aliquots by using 7-10 psi to avoid rupturing the algal cells.
 - a) Examine the filter. An adequate amount of microalgal biomass for analysis is indicated by the green or brown color of material retained on the filter. Extract additional 5.0-mL aliquots and filter until the desired level of biomass is obtained.
 - b) Determine the number of 5.0-mL aliquots filtered, and record the subsample volume on the field data sheet (e.g., 2 aliquots x 5.0 mL/aliquot = 10 mL subsample volume).
 - c) Rinse the funnel sides with deionized water; allow the water to be vacuumed completely before releasing the vacuum from the filtering apparatus.
 - d) Remove the filter from the funnel base with forceps.
 - e) Rinse the filter funnel, filter holder, filter chamber, and graduated cylinder thoroughly with deionized water.
 - f) Repeat the filtering steps for each subsample (either Chl-*a* or ADFM) collected.
9. Prepare the filtered subsamples (Chl-*a* and ADFM) for storage and shipping
 - a) Fold each filter into quarters with filtered biomass inside. Wrap each filter in a small piece of aluminum foil and place in separate labeled resealable plastic bags.
 - b) Label the bag with the following required information: site, collection date, total sample area, sample volume, subsample volume, sample type (Chl-*a* or ADFM), and sample identification code.

- c) Place the labeled resealable plastic bags in a cooler containing dry ice. About 4.5 kg (10 pounds) of dry ice is needed for subsamples packed in a small cooler (< 2 gal). Insulate the cooler with newspaper to minimize sublimation of the dry ice.
10. Measure the volume of the remaining benthic algae sample component. This represents the subsample volume of the remaining subsample to be archived.
11. Preserve the archive subsample with a sufficient volume of buffered formaldehyde to obtain a final concentration of 3 to 5 percent buffered formalin. Record the preservative volume on the field data sheet.

Volume (mL)

Sample	25	50	100	125	200	250	300	350	400	450	475
Buffered formaldehyde	1	2	4	6	8	11	14	16	18	22	23

12. Place a completed sample label on the sample bottle.
13. Filtered subsamples should be stored in freezers (at -20°C) as soon as possible.
14. Samples should be shipped to the laboratory as soon as possible, because of a 25-day holding-time limit on the subsamples.
15. Complete a Chain of Custody form provided by the accredited contract laboratory for listed samples that indicates which laboratory analyses are to be performed. Contact the contracted laboratory to make them aware of plans to ship (via overnight shipping service) coolers containing dry ice and frozen subsample filters.

2.3.3. Data Analysis Methods

Results generated from the collections would include both AFDM and chlorophyll-*a*. Each measure will have the mean and variability (95-percent confidence intervals) calculated. Comparisons for AFDM and *chl-a* will be made among sites, to look for differences between habitat types, as well as spatial trends along the length of the river (upstream versus downstream sites). Comparisons will also be made over time, examining both the interannual (seasonal) and annual variability in algal biomass and *chl-a*. Statistical tests (ANOVA, ANCOVA, MANOVA) will be performed on each measure to look for an overall significant difference among sites, seasons, and years. If a difference is significant ($p \leq 0.05$) for the measure, then a multiple comparison test will be used to describe the significant differences in the data. Results may also be used as covariates in the analyses of benthic macroinvertebrate data, especially in regards to the functional feeding group compositions in the community. Multivariate ordination procedures, such as principle components analysis (PCA) and canonical correspondence analysis (CCA), may be utilized to explain the relative contribution of AFDM and chlorophyll-*a* to observed grouping patterns of sites, stations, times, and in relation to macroinvertebrate taxa, their distributions, and the functional feeding groups of the benthic macroinvertebrate community.

2.4. Organic Matter Sampling

Organic matter materials serve as an important food resource to benthic macroinvertebrates, serving as a conduit for the energy flow from organic matter resources to vertebrate populations,

such as fish (Hershey and Lamberti 2001; Hauer and Resh 1996; Reice and Wohlenberg 1993; Klemm et al. 1990). This organic matter exists as both fine particulate organic matter (FPOM) and coarse particulate organic matter (CPOM). FPOM includes particles ranging from 0.45 to 1000 μm in size and can occur in the water column as seston or can be deposited in lotic habitats as fine benthic organic matter (FBOM; Wallace and Grubaugh 1996). CPOM is defined as any organic particle larger than 1 mm in size (Cummins 1974).

Given the dominant characteristics of the Susitna River system (i.e., large, cold, and turbid during the growing season), secondary productivity is likely to be driven by allochthonous inputs of organic material from the terrestrial environment. Benthic organic material is one of the most important “interrelated environmental factors” influencing the macroinvertebrate community, and damming the river is likely to have significant consequences for the transport of organic matter from the upper watershed. Therefore, to address the importance of organic matter to productivity in this type of system, this study will quantify benthic organic matter as it is related to the benthic macroinvertebrates being collected.

2.4.1. Field Sampling Protocols

In order to quantify the amounts of organic matter available in the Susitna River for river productivity, CPOM and FPOM will be collected directly from all benthic macroinvertebrate sampling, in Hess samples and drift net samples. (RSP Objective 2, Section 9.8.4.2.1.; RSP Objective 3, Section 9.8.4.3.).

2.4.1.1. Benthic

In order to streamline the collection efforts, Hess sampling devices will possess a net mesh size of 250 μm in order to retain FBOM in the 250–1,000 μm size range for analysis, as well as CPOM particles. All organic debris collected within each Hess sample collected will be retained with the sample and preserved with the entire portion in 95 percent ethanol. Organic materials too large to fit within the sample jar (i.e., sticks) will be thoroughly examined for attached organisms and broken down enough to fit within the sample jar or in a large resealable plastic bag. A standard internal label will be placed within the bag, including the same information as the benthic sample jar. An additional note will be made on the field data sheet for that specific sample indicating that additional organic matter was collected and stored in the plastic bag.

2.4.1.2. Seston

Suspended FPOM (seston) will be collected from material in invertebrate drift samples, utilizing drift nets with a 250- μm mesh size in order to retain FBOM in the 250–1,000 μm size range for analysis, as well as CPOM particles (RSP Objective 3, Section 9.8.4.3). All organic debris collected within each drift sample collected will be retained with the sample and preserved with the entire portion in 95 percent ethanol.

2.4.2. Processing Protocols

Processing of benthic macroinvertebrates involves subsampling to acquire a 300-organism fixed-count (± 20 percent) subsample. All invertebrates are removed from debris with the aid of a dissecting microscope (7–45x), and sorted debris is retained in a labeled, 60-ml bottle and stored

for later for QA/QC assessment and, for Hess samples, organic matter analysis. Organic matter retained from subsampling after organism sorting and processing will be separated from inorganic material, rinsed through 1-mm and 250- μ m nested sieves, to separate CPOM and FPOM components of the detritus, oven-dried (60°C), and weighed. Results will be expanded according to the subsample factor, and calculated as amounts of CPOM and FPOM per unit area (g/m^2).

Processing of drift samples will require full sorting. After the detritus has been sorted and benthic invertebrates removed, the sample material should be rinsed through 1 mm and 250 μ m nested sieves to separate CPOM and FPOM components of the detritus. Components will then be oven-dried (60°C), and weighed. Results will be calculated as amounts of CPOM and FPOM per unit area (g/m^3).

2.4.3. Data Analysis Methods

Results generated from the collections would include amounts of CPOM and FPOM per unit area (g/m^2 for benthic samples and g/m^3 for drift samples). Each measure will have the mean and variability (95-percent confidence intervals) calculated. Comparisons for these measures will be made among sites, to look for differences in organic matter content between habitat types, as well as spatial trends along the length of the river (upstream versus downstream sites). Comparisons will also be made over time, examining both the interannual (seasonal) and annual variability in the amounts of organic matter within the sampled substrates. Statistical tests (ANOVA, ANCOVA, MANOVA) will be performed on each measure to look for an overall significant difference among sites, stations, seasons, and years. If an effect is significant ($p \leq 0.05$) for the measure, then a multiple comparison test will be used to describe the significant differences in the data. Results may also be used as covariates in the analyses of benthic macroinvertebrate data, especially in regards to the functional feeding group compositions in the community. Multivariate ordination procedures, such as principle components analysis (PCA) and canonical correspondence analysis (CCA), may be utilized to explain the relative contribution of organic matter to observed grouping patterns of sites, stations, times, and in relation to macroinvertebrate taxa and distributions, and the functional feeding groups in the benthic macroinvertebrate communities.

2.5. Invertebrate Drift Sampling

Stream dwelling invertebrates are often transported downstream in the water column, which is referred to as “drift”. Several categories have been used in the literature to describe drift: behavioral, constant, and catastrophic (Waters 1972). *Behavioral drift* occurs when organisms actively enter the water column, for example to escape predators or search for food. Behavioral drift has been found to show a diurnal pattern for many species. Many studies have reported increased drift densities during the night, peaking twice: one just after sunset, and a smaller peak just before sunrise (Brittain and Eikeland 1988). *Constant drift*, also called background drift, is drift that occurs in steady, low numbers, regardless of the time of day, because of accidental dislodgement from the substrate. *Catastrophic drift* is usually associated with flow-related disturbances but can also be due to disturbances involving pollution or changes in the temperature regime (Brittain and Eikeland 1988). Catastrophic drift can result from both flow

increases and decreases, either due to natural occurrences such as floods or spates and droughts or due to river regulation.

Regarding behavioral drift, it is unclear whether the benthic community in the Susitna River would exhibit the typical strong diel patterns. While many studies show that drift is characterized by a distinct diel periodicity, with greater drift in the night than during the daytime, such diel patterns are usually exhibited by Ephemeroptera, Plecoptera, Trichoptera, (EPT taxa) and Simuliidae taxa (Brittain and Eikeland 1988). Chironomidae are usually reported to be aperiodic, showing either no diel variation in drift densities, maximum drift during daylight hours, or a maximum drift at night (Brittain and Eikeland 1988). Measures of drift in a glacial river and its non-glacial tributary in Western Norway found that Chironomidae were the most abundant in drift and showed significant peaks in drift density at mid-day sampling (Saltveit et al. 2001). Light level serves as a signal for behavioral drift (Allan 1995). Müller (1973) found that the reaction of stream invertebrates to the long photoperiods of summers in higher latitudes is much different in that it extinguishes drift rhythm entirely.

In addition to aquatic invertebrates, terrestrial invertebrates often enter streamflows and drift from riparian vegetation, can comprise a significant proportion of drift, and may be an important food subsidy for salmonids (Elliott 1973; Cada et al. 1987; Wipfli 1997; Nakano et al. 1999; Kawaguchi and Nakano 2001; Allan et al. 2003) particularly in unproductive streams (Romaniszyn et al. 2007). This terrestrial component of drift does not display any diurnal patterns.

In Alaskan streams and rivers, the benthic community is dominated by Chironomidae, with EPT taxa together accounting for less than 25-percent of the fauna (Oswood 1989). Given that several studies have shown that chironomids do not necessarily adhere to the typical patterns of diurnal drift, and that such diel periodicity is disrupted by long photoperiods of summers in higher latitudes, invertebrate drift sampling conducted during daylight hours can be considered a valid approach under this study plan. Collecting drift samples concurrently with benthic macroinvertebrate sampling at all sites within the six established sampling stations will allow for comparisons between the drift component and the benthic macroinvertebrate community, as well as revealing the availability of terrestrial invertebrates to fish predation.

2.5.1. Field Sampling Protocols

Sampling will be conducted in fast-water habitats within the established mainstem sites, and associated off-channel habitat sites. Invertebrate drift sampling will be conducted based on the USEPA's EMAP drift net sampling protocols (Klemm et al. 2000). Drift sampling will be conducted during daylight hours, preferably beginning shortly after arrival at a site in the morning, and will involve collecting duplicate samples (Klemm et al 1990; Klemm et al. 2000). Drift nets with a 250-µm mesh size will be utilized (Figure 2.2-1). Water velocity will be recorded with an in-net flow meter. The following is the procedure for sampling invertebrate drift.

1. Locate the area to install the drift net pair for the site. Do not use drift nets in areas with currents less than 0.05 meters per second (m/s). Drift nets should always be deployed above the sampling reach to avoid the unintentional introduction of macroinvertebrates to the drift by disturbance of the stream substrate by the crew's other sampling efforts.

Ideally, the nets should be installed at the downstream end of a fast-water habitat (typically a riffle or run).

2. Install the net in an area of river that is receiving part of the main channel flow, but that can be safely accessed by wading. Depths of 1 m or less are preferred.
3. Drive steel rods or rebar into the substrate. Drift nets should be oriented perpendicular to and facing the stream flow and secured to the rods with cable clamps. The bottom of the net mouth should be suspended at least 2 cm above the stream bed to deter invertebrates from crawling into the net mouth. Position the net so that the top of the net is above the water surface at least 2 cm, such that drifting terrestrial invertebrates and debris are collected. Note on the field data sheet the distance from the bottom of the net (from the inside margin of the frame) to the water surface. This will be used to calculate the area of the net mouth receiving flow.
4. Install the in-net flow meter into one of the nets. Record the starting counter number and the start time of sampling on the field data sheet. In addition, measure the current velocity at the entrance of the net and at 60 percent of the depth using a flow meter, and record the measured velocity and depth, as well as a measure of turbidity and temperature, on the field data sheet.
5. Avoid walking upstream of the drift net during drift net deployment.
6. Leave the drift net assembly in the river for at least 1 hour, and as long as 3 hours, checking the nets often for signs of clogging. Drift nets can become clogged with suspended material, causing nets to back up water at the net mouth, and resulting in an inaccurate estimate of the total volume of water sampled by a net. If a net is filling rapidly and beginning to clog in less than one hour, sample for the shorter duration.
7. Before removal at the end of the allotted sampling time (1-3 hours), measure the current velocity at the entrance of the net and at 60 percent of the depth using a flow meter, and record the measured velocity and depth, as well as a measure of turbidity and temperature, on the field data sheet.
8. Record the end time counter number and the end time of sampling on the field data sheet.
9. Remove the nets from the water, holding the net vertically and taking care not to disturb the bottom upstream of the net. Concentrate the material in each net by swishing up and down in the stream or river.
10. Empty the contents of one drift net into a 250- μ m mesh sieve. Closely inspect all large materials for attached invertebrates. Keep all organic matter; it will be needed for assessment of organic matter content (Section 2.4). Discard all larger inorganic materials.
11. Rinse the sample in the sieve, consolidating the material to one side of the sieve, and transfer the material into a storage container. Efforts should be taken to minimize the amount of water retained with the sample to prevent too much dilution of the ethanol used to preserve the sample. Next, scoop out the material with a spoon or spatula and place it in the sample container, and then rinse the sieve to consolidate the remaining material to one side of the sieve. Wash the remaining sample into the container with a wash bottle containing 95 percent ethanol.
12. A standard label defining the station, site, sample number, date, collector, and unique sample identification code is added to the sample. Adhesive standard labels are also applied to the outside of the sample jars.

13. Preserve the sample with additional 95 percent ethanol, enough to completely cover the sample, and place the labeled lid on the container, making sure it is secured tightly and does not leak.
14. Repeat steps 10-13 for the 2nd drift net. Do not combine the contents of separate drift net samples.
15. Rinse the drift nets out completely.

2.5.2. Processing Protocols

Invertebrate drift samples will be processed in an accredited contract taxonomic laboratory, using methods similar to those used for benthic samples (Barbour et al. 1999; Major and Barbour 2001; see Appendix 1). Laboratories should have taxonomists on staff that are certified by the Society for Freshwater Science for taxonomic identifications of specific groups (EPT taxa, chironomids, etc.). Processing of drift samples will likely require full sorting; however, if a sample is too large (i.e., it will require greater than 3 hours to process), subsampling may be warranted, either by a sample splitter or a gridded tray. After the sample has been sorted and invertebrates removed, the organic debris must be retained for processing (Section 2.4.2).

Biomass estimates will be taken for invertebrate taxa collected for benthic sampling. The fresh blotted wet mass of invertebrate taxa in samples will be recorded. The samples will be oven dried at 60°C until reaching constant mass, and the dry mass will be recorded.

2.5.3. Analysis Protocols

Results generated from these collections will include drift density, drift rate, and drift composition. For a select subsample of the collection, energy density (J / g wet weight) will be estimated from the percent dry mass (dry mass / wet mass) of each sample (Ciancio et al. 2007; James et al. 2012). Energy density will be determined separately for the aquatic and terrestrial life stages of each primary invertebrate taxon for use in the trophic modeling efforts.

Data collected as part of this study will be compared to data from the benthic macroinvertebrate collections (RSP Section 9.8.4.2.1) and the fish dietary analysis (RSP Section 9.8.4.7). In addition, drift results will be compared to the results of 1980s drift studies (ADF&G 1983a; Hansen and Richards 1985; Trihey and Associates 1986) to evaluate any differences between the historic and current drift components of the macroinvertebrate communities.

2.6. Adult Insect Emergence Sampling

2.6.1. Field Sampling Protocols

To measure insect emergence, floating emergence trap samplers will be deployed, with one trap per site. The emergence traps will be based on previous designs of floating aquatic emergence traps (LeSage and Harrison 1979; Cushman 1983; Davies 1984; Walton et al. 1999). The emergence trap design will be a low-profile trap with a floating base (Figure 2.2-1), with a collection bottle or tray with alcohol preservation attached to the trap to collect adult specimens. The trap will be anchored to rebar stakes driven into the stream bed by a length of chain, cable, or rope. Ethanol (95 percent) with glycerol added will be placed into the trap collection bottle or tray. Samples will be collected from deployed traps approximately every 2 weeks by field crews. The collected adult insects will be removed from the trap, washed into a sample

container, preserved with ethanol with glycerol added, and labeled with information about the site, date collected, time collected, and the collectors' initials. This information will also be entered onto a field data sheet. The trap will be reassembled, with fresh ethanol added to the collection bottle or tray. The collected sample container will be sent to the contract laboratory for processing and analysis.

2.6.2. Processing Protocols

Adult aquatic insect samples will be sent to one or more contract taxonomic laboratories for identification. A dissecting microscope will be used to sort, identify, and enumerate collected specimens at the family level. If feasible, chironomid should be separated into 'morphospecies' with select reference organisms slide mounted and identified under a compound microscope to genus. If specimen counts are high, subsampling may be necessary, either by sample splitting, or a gridded tray approach. Biomass estimates will be taken for all identified taxa. The fresh blotted wet mass of each identified taxa in samples will be recorded, the samples will be oven dried at 60°C until reaching constant mass, and the dry mass will be recorded.

2.6.3. Data Analysis Methods

Results generated from these collections will include taxonomic composition, densities (numbers/m²/time period), and biomass (dry weight/m²/time period), which will be described qualitatively through graphical depictions for emergent insect families and chironomid genera. Adult aquatic insect emergence results will also be compared to data from the benthic macroinvertebrate collections (RSP Section 9.8.4.2.1), drift collections (RSP Section 9.8.4.3), and the fish dietary analysis (RSP Section 9.8.4.7). Comparisons of results from emergence traps with species composition of benthic community sampling will describe timing in the aquatic phase and time of emergence. This information will be useful in assessing how the timing of emergence may be altered due to Project operations.

For a select sub-sample of the collection, energy density (J / g wet weight) will be estimated from the percent dry mass (dry mass / wet mass) of each sample (Ciancio et al. 2007; James et al. 2012). Energy density will be determined separately for each primary invertebrate taxon for use in the trophic modeling efforts.

2.7. Fish Scale Sampling

2.7.1. Field Sampling Protocols

To support the fish stomach content analysis (Section 2.8) and trophic modeling (Section 2.10), fish scales for aging juvenile Chinook salmon, juvenile coho salmon, and juvenile and adult rainbow trout (DeVries and Frie 1996) will be collected in conjunction with fish abundance and distribution sampling in the Upper River (RSP Section 9.5.4.3.1) and Middle River (RSP Section 9.6.4.3.1). Scales will be collected from the first five fish of each species and age group captured at each sampling site, in conjunction with stomach content sampling (Section 2.8). For field sampling purposes during 2013, rainbow trout will be provisionally categorized as "juveniles" (ages 0 and 1) at less than or equal to 120 mm fork length, or "adults" (ages 2 and above) at greater than 120 mm fork length (ADF&G 1983b, pp. G-8, G-14; Sundet and Wenger 1984, part 5, pp. 69, 70). The length cutoff will be adjusted if necessary for the 2014 field season based on

the length-age relationship determined from the scale analysis. Scales will be collected after stomach contents are collected from anaesthetized fish and before fish are placed in the river water recovery tote (see Section 2.8). Scales will be collected from specific areas of each fish using forceps; those areas are below and posterior to the dorsal fin (Scarnecchia 1979). Multiple scales (approximately six) will be collected from each fish to increase the likelihood that at least one non-regenerated scale is available for aging. Scales will be stored dry in small paper envelopes individually labeled with a specimen number and transported to a laboratory for analysis.

2.7.2. Processing Protocols

The age and growth of juvenile Chinook salmon, juvenile coho salmon, and rainbow trout will be determined using scales and temporal length distribution data (DeVries and Frie 1996; Isely and Grabowski 2007). Seasonal length-frequency distributions will be examined for juvenile Chinook salmon, juvenile coho salmon, and juvenile rainbow trout, stratified by sampling season. If any species displays distinct length modes, suggesting that age-0 and age-1 fish are distinguishable from each other and from older fish based on length and sampling date alone (e.g., Daum and Flannery 2011), this method will be validated by aging scales from a random subset of 80 fish per size group. If seasonal length distributions do not contain distinct modes or if the length-frequency analysis fails to correctly assign at least 95 percent (76 of the 80 fish) to the correct age based on the scale analysis, then all scales from that species will be aged. Age classes of rainbow trout aged 2 years and older are expected to overlap in length, so all fish will be aged by scale analysis only.

Ages will be assigned to fish using scale annuli (DeVries and Frie 1996). Scales of juvenile fish will be removed from envelopes in the lab, soaked in water in a petri dish, and cleaned of any slime and foreign material. One suitable scale (neither regenerated nor damaged) from each fish will be identified under a dissecting microscope. Scales will be aged using one of two methods. First, scales will be mounted on microscope slides and directly examined under a dissecting microscope. Second, if direct microscopy does not yield consistent ages, scales will be aged from acetate impressions. Clean scales will be placed on a gummed card, sculptured side up. Gummed cards containing multiple labeled scales will be impressed into acetate slides, and the slides will be examined under a dissecting microscope. In each method, every scale will be aged independently by two readers, with the final age assigned by consensus. Images of a subset of scales will be captured and archived with a microscope-mounted digital camera interfaced with a desktop computer.

2.7.3. Data Analysis Methods

Growth rates of juvenile Chinook salmon, juvenile coho salmon, and rainbow trout will be characterized in terms of mean weight at age based on field data from all specimens sampled, stratified by sampling period and reach. To test whether size-selective mortality or migration introduces bias into these growth rate estimates, seasonal growth rates will also be estimated for individual fish that are recaptured during the PIT tag studies in the Upper River (RSP Section 9.5.4.3.2) and Middle River (RSP Section 9.6.4.3.2). The adopted growth relationships will serve as inputs for the bioenergetics models for each species (Section 2.10).

2.8. Fish Gut Content Sampling

2.8.1. Field Sampling Protocols

Fish stomach contents will be sampled by nonlethal gastric lavage in conjunction with fish abundance and distribution sampling in the Upper River (RSP Section 9.5.4.3.1) and Middle River (RSP Section 9.6.4.3.1). Stomach contents will be sampled from juvenile coho salmon, juvenile Chinook salmon, and juvenile and adult rainbow trout to provide input data for the trophic model (RSP Section 9.8.4.5.1). Stomach contents will be collected from the first eight fish per species and age class that are captured at each sampling site. A fish that is lavaged and found to have an empty stomach will be replaced by the next fish of that species and age class that is captured. Rainbow trout will be provisionally classified as juvenile (age 0 and 1) or adult (age 2 and older) for field purposes using the method described in the fish scale collection section (Section 2.7). Fish will be anesthetized with clove oil, measured for fork length (mm), weighed (g), and their stomach contents will be flushed with a 10-mL syringe assembly (Meehan and Miller 1978). Water will be gently pumped into fish stomachs to force stomach contents out of fishes' mouths. Stomach contents and associated water will be flushed into a small plastic bag and an equal volume of 95 percent ethanol will be added as a preservative for transport to the laboratory (Wipfli 1997). Fish will be held in a plastic tote filled with river water until they recover their ability to maintain an upright orientation. After recovery, fish will be returned to the river near the original place of capture.

This sampling effort will target a maximum of 1,728 stomach content samples over each of the two field seasons, allocated according to the detailed sampling protocol provided in Table 2.8-1. If this protocol cannot be achieved because not all target taxa are captured from all sites during all sampling periods, then a portion of the sampling effort may be reallocated to match the distribution of organisms encountered in the field, with the goal of achieving the study objectives most effectively.

2.8.2. Processing Protocols

Stomach content samples will be examined under a dissecting microscope in the laboratory (Bowen 1996). Invertebrate prey will be identified to life stage (i.e., aquatic or terrestrial) and family when possible, or otherwise the lowest possible taxonomic level. Fish prey will be identified to species when possible, or otherwise the lowest possible taxonomic level. The blotted wet weight of each prey category will be recorded to the nearest 0.1 g using an electronic balance. A representative subset of prey items in each category will be measured to the nearest millimeter and weighed to the nearest 0.01 g. All stomach contents will be archived in 95 percent ethanol for future verification.

2.8.3. Data Analysis Methods

The diet composition of each fish species and age class will be calculated in terms of diet proportions by weight (Chipps and Garvey 2007). Diet composition will be compared along an upstream-downstream gradient and among habitat types and seasons for each fish species and age class using multivariate statistics. In the event data does not meet distribution assumptions, it may be transformed (e.g., arcsine-transformed) prior to analysis. Multivariate analysis of variance of diet proportions, two-dimensional Kolmogorov-Smirnov tests, or alternative tests

equivalent to these statistical analyses will be used to make comparisons (Chipps and Garvey 2007). Potential ontogenetic shifts in diet will be identified graphically for each fish species by plotting the taxon and length of individual prey against the length of the associated fish predator (Beauchamp et al. 2007). The onset of piscivory in rainbow trout will be identified graphically by plotting the proportion of fish in each stomach content sample against the length of each rainbow trout. Data collected during this study will also be compared to the results of fish diet studies conducted on the Susitna River during the 1980s (ADF&G 1983a; Hansen and Richards 1985) to evaluate any differences between the historic and current fish diets.

2.9. Macroinvertebrate Colonization Sampling

In order to assess the influences of turbidity and temperature on the benthic community colonization rates, a field study will be conducted for both study years (i.e., 2013 and 2014) to estimate potential benthic macroinvertebrate colonization rates for four different habitat types that reflect these conditions in the Susitna River. Due to the difficulty of isolating each of these conditions under natural conditions, colonization will be examined under turbid/warm, clear/warm, turbid/cold, and clear/cold conditions. Locating and establishing appropriate sampling sites for colonization study will require an extensive review of all studies done in the segment, along with discussions with other research teams conducting field studies in the Middle Segment of the Susitna River to locate areas that display these conditions over an eight-week period. This effort will also require site reconnaissance trips to assess candidate sites.

Hester-Dendy multiplate samplers (Hester and Dendy 1962; Fullner 1971; Tsui and Breedlove 1978) will be used as the artificial substrates for the study of colonization rates in the Middle Segment of the Susitna River. The Hester-Dendy multiplate sampler consists of 14 tempered hardboard plates, either square or circular, which are spaced apart by smaller, circular hardboard spacers and mounted on a central eyebolt (Figure 2.2-1). The top nine plates are separated by one spacer, and the remaining plates are separated by 2, 3, and then 4 spacers, thus providing a simulated complexity of surfaces and interstitial spacing (Klemm et al. 1990). This version of the Hester-Dendy sampler offers 0.16 m² of surface area. Hester-Dendy multiplates are frequently suspended in the water column but can be deployed along the bottom with a heavy anchor weight (Klemm et al. 1990).

Multiplate samplers were selected, because each unit has the same surface area and the same microhabitat to offer potential colonizers. This permits standardized sampling in both the sampling effort, thus eliminating sampling error, and in surface area sampled. They also have a calculated surface area, which allows for more quantitative results to be collected. Multiplate samplers are also small and more easily transported when accessing remote areas, as opposed to rock baskets, which are bulky. Hester-Dendy samplers are also the most common type of artificial substrate samplers used today by several state and federal monitoring programs and have been successfully used on many large rivers, notably as part of standard programs in Florida, Wisconsin, and Ohio (Johnson et al. 2006).

2.9.1. Field Sampling Protocols

All Hester-Dendy samplers will be pre-conditioned prior to deployment by being placed for 4 weeks in the Susitna River (preferably at a project base camp) and then air-dried. Sets of three pre-conditioned artificial substrates will be deployed incrementally for set periods of

colonization time (e.g., 8, 6, 4, 2, and 1 week[s]) and then pulled simultaneously at the conclusion of the colonization period. Artificial substrates will be deployed at two depths at fixed sites along the channel bed. Depths at which to set the substrates should take into account the river stage fluctuations. Crew members deploying the multiplate samplers should attempt to set the shallower set of samplers during the lowest river stage and/or at locations that will ensure that the substrates are not dewatered during their time in the water (for up to 8 weeks). This will require consulting the real-time flow and stage gage data available to make an informed decision. Progressive sets for each new exposure period should be installed downstream of those previously installed, so as to minimize disturbances to the previous set(s). For example, the sets exposed for 4-weeks should be installed a short distance downstream from the 6-week sets, which, in turn, are located downstream from the 8-week sets.

For each set deployed, three multiplate samplers will be equidistantly attached to a 2-foot section of light chain. A two-foot rebar spike with an eyebolt will be driven into the riverbed at the appropriate location and depth, and the chain of multiplates then laid out upon the river bottom, in direct contact with the cobble/gravel substrates and attached to the spike with a quick link. The location, depth, velocity (both 60 percent of depth and near-bed measurements), PAR levels, and turbidity should be measured at the deployment of each set, and recorded on the field data sheet for the current deployment. If previous sets have been deployed, those sets should be checked, to see if they have been lost, vandalized, damaged, or exposed since the last deployment at the site.

The following is the procedure for the retrieval of multiplate sampler sets and collection of benthic macroinvertebrates.

1. Locate the shallow depth set of multiplate samplers deployed for the downstream-most set. At the beginning, this should be those sets deployed for 1 week.
2. Take measurements of depth, velocity (both 60 percent of depth and near-bed measurements), PAR levels, and turbidity, and record the data on the field data sheet, making sure to note which set of multiplate samplers is being retrieved (e.g., 1 week, shallow).
3. Place a D-net directly downstream of the multiplate set. Detach the chain from the rebar spike, and then remove the chain and multiplate samplers from the water.
4. Multiplates are quickly brought to shore, and placed in three separate wash-buckets. Each multiplate sampler is disassembled, and cleaned of colonizing macroinvertebrates in its wash-bucket. Cleaned parts are set aside for reassembly later.
5. Pour the contents of the wash bucket through a 250- μ m sieve. Rinse the bucket with water to ensure all material and invertebrates are washed into the sieve.
6. Rinse the sample in the sieve, consolidating the material to one side of the sieve, and transfer the material into a storage container. Efforts should be taken to minimize the amount of water retained with the sample to prevent too much dilution of the ethanol used to preserve the sample. Next, scoop out the material with a spoon or spatula and place it in the sample container, and then rinse the sieve to consolidate the remaining material to one side of the sieve. Wash the remaining sample into the container with a wash bottle containing 95 percent ethanol.
7. A standard label defining the station, site, sample number, date, collector, and unique sample identification code is added to the sample. Adhesive standard labels are also applied to the outside of the sample jars.

8. Preserve the sample with additional 95 percent ethanol, enough to completely cover the sample, and place the labeled lid on the container, making sure it is secured tightly and does not leak.
9. Reassemble the multiplate sampler, so as not to lose any parts.
10. Repeat steps 1-9 with the deep depth set of multiplate samplers deployed at that exposure time.
11. Continue upstream to the next set of multiplate samplers. Repeat steps 1-10.

2.9.2. Processing Protocols

Benthic macroinvertebrate processing protocols will be identical to those used for benthic macroinvertebrate sampling (Section 2.2).

2.9.3. Data Analysis Methods

Results generated from the collections will include a variety of descriptive metrics commonly used in aquatic ecological studies (Table 2.2-1). Comparisons will be made among exposure times, depths, and conditions to examine trends of benthic macroinvertebrate colonization. Colonization information will be compared with colonization results from other river systems and, in the future, with post-Project colonization results. In addition, results will be utilized in HSC/HSI development (RSP Section 9.8.4.6) and in the varial zone modeling task of the Instream Flow Study (RSP Section 8.5.4.6.1.6) to assist in determining potential Project effects of short-term flow fluctuations on benthic macroinvertebrates.

2.10. Trophic Modeling

2.10.1. Data Analysis Methods

To determine how water temperature, food availability, and food quality influence the growth performance of juvenile Chinook salmon, juvenile coho salmon, and juvenile and adult rainbow trout, field data from the Instream Flow Study (RSP Section 8.5), Fish Abundance and Distribution Studies in the Upper and Middle Rivers (RSP Sections 9.5 and 9.6), and the River Productivity Study will be analyzed using a bioenergetics approach. This analysis will allow comparisons of observed growth rates, estimated consumption rates, and estimated growth efficiency (i.e., the grams of growth achieved per gram of food consumed) among different habitats under the environmental conditions observed during 2013 and 2014. Consumption and growth efficiency will be estimated using Wisconsin bioenergetics models (Hanson et al. 1997) with species-specific physiological parameters for Chinook salmon (Stewart and Ibarra 1991; Madenjian et al. 2004), coho salmon (Stewart and Ibarra 1991), and rainbow trout/steelhead (Rand et al. 1993). Simulations for each species will encompass the full range of age classes for which sufficient field data are collected; at a minimum, these are expected to include ages 0-1 for Chinook salmon, 0-2 for coho salmon, and 0-8 for rainbow trout. Simulations will run on a daily time step from emergence from the gravel through smolting (or senescence for resident rainbow trout). Model inputs will include field data on growth rate, water temperature, diet composition, and the energy density of prey. Growth rates will be determined from seasonal mean weight at age data (Section 2.7.3). Water temperatures will be measured using temperature loggers (RSP Section 9.8.4.2.1). Diet composition will be determined from stomach contents (Section 2.8).

The energy density of prey will be estimated based on laboratory measurements of the percent dry matter of prey organisms (Ciancio et al. 2007; James et al. 2012) collected during sampling of macroinvertebrates, as described in Sections 2.1.2 and 2.1.5, and fishes (RSP Sections 9.5 and 9.6). Based on these inputs, the bioenergetics models will estimate consumption rates and growth efficiency on a daily basis. These metrics will be compared among habitats and seasons to determine whether growth is currently limited primarily by water temperature, food consumption, or food quality in the study area, and whether these limiting factors differ among habitats (McCarthy et al. 2009).

In addition to the descriptive bioenergetics analysis described above, a growth rate potential (GRP) analysis will be developed and evaluated as a potential prospective approach for predicting fish growth rates under changing environmental conditions. Detailed foraging parameters for juvenile coho salmon and juvenile rainbow trout have been published (e.g., Dunbrack and Dill 1984; Berg and Northcote 1985; Piccolo et al. 2007; Piccolo et al. 2008a, 2008b), enabling the development of well-supported drift foraging models for both species. The necessary bioenergetics model parameters are also available for these fishes (see above). Mechanistic drift foraging models are not available for juvenile Chinook salmon, so the growth rate potential approach will not be applied to this species.

Species-specific GRP models for juvenile coho salmon and juvenile rainbow trout will link a drift foraging model (Fausch 1984; Hughes and Grand 2000; Hayes et al. 2007) to a Wisconsin bioenergetics model (Kitchell et al. 1977; Hanson et al. 1997). The foraging models will estimate a consumption rate based on stream flow, turbidity, and prey density input data. The bioenergetics models will predict a growth rate from inputs of consumption, body size, water temperature, diet composition, and the energy density of prey.

Preliminary GRP models for each species will be developed using data from the 2013 field season as well as from prior Susitna Basin studies. Initial model predictions of the growth potential of particular sites will be tested by comparison with the observed growth and distribution of fish captured in those sites. A sensitivity analysis will be conducted to identify the most important parameters for further refinement (e.g., Beaudreau and Essington 2009). Field sampling during 2014 will focus on improving estimates for these parameters. Preliminary growth models will simulate GRP assuming that fish remain within a given habitat; however, final GRP models, developed after the 2014 field season, will allow simulated fish to move among habitats within a sampling location to enhance growth rates. Optimal simulated movement patterns will be estimated and compared with the observed movements documented by the biotelemetry components of the Fish Distribution and Abundance Studies of the Upper River (RSP Section 9.5.4.3.2) and Middle River (RSP Section 9.6.4.3.2). Final GRP models will also allow for inter- and intraspecific competition among juvenile coho salmon and rainbow trout (Hughes and Grand 2000).

The suitability of the GRP models for predicting the growth rates of each species will be tested using an information theoretic model selection approach (Burnham and Anderson 2002). For each species, a full model will be fit to the observed growth data using the observed water temperature, stream flow, turbidity, prey density, prey quality (energy density), and competitor density as explanatory variables. A set of simplified growth models will also be constructed using every possible subset of those variables. The full suite of candidate growth models will be fit to the data, and the most parsimonious models will be identified using AICc (Burnham and Anderson 2002). This analysis will evaluate whether the GRP approach or simpler approaches

may serve as useful tools for future predictive analyses of the effects of future environmental changes on fish growth in the Susitna River.

2.11. Stable Isotope Analysis

2.11.1. Field Sampling Protocols

Stable isotope samples will be collected from algae, OM, spawning salmon, aquatic and terrestrial macroinvertebrates, and focal salmonid fishes. Isotope samples will be collected from two of the River Productivity Study sampling stations in the Middle Susitna River, with three habitat-specific sampling sites per station, for a total of six sampling sites. To account for temporal variability in isotopic signatures (Post 2002), all sample types will be collected during three seasonal periods, with the exception of salmon carcasses, which will be collected only during the spawning run. Algae, OM collected by benthic sampling, and OM collected by drift sampling will each be collected in separate composite samples of approximately 10 g wet mass. Tissue from the carcasses of spawned out salmon will be collected during spawning runs in composite samples of approximately 10 g wet mass. Aquatic macroinvertebrates will be collected by benthic sampling in composite samples of approximately 20 g wet mass. Terrestrial macroinvertebrates will be collected by drift and emergence sampling in separate composite samples of approximately 5 g wet mass. In conjunction with fish abundance and distribution sampling in the Middle River (RSP Section 9.6.4.3.1), stable isotope samples will be collected non-lethally from fish by clipping a small portion of the caudal fin (Sanderson et al. 2009; Hanisch et al. 2010). Fin clips will be collected after stomach contents (Section 2.8) and scales (Section 2.7) are collected from anaesthetized fish and before fish are placed in the river water recovery tote (see Section 2.8). Fin clip sampling may cause a reduction in survival for fishes smaller than 50 mm in fork length (Sanderson et al. 2009), so any fish of this size that are selected for sampling will be sacrificed. These fish will be sacrificed with an overdose of buffered MS-222 and filleted to provide stable isotope samples.

This sampling effort will target a maximum of 1,246 total stable isotope samples over each of the two field seasons, allocated according to the detailed sampling protocol provided in Table 2.11-1. If this protocol cannot be achieved because not all target taxa are captured at all sites during all sampling periods, then a portion of the sampling effort may be reallocated to match the distribution of organisms encountered in the field, with the goal of achieving the study objectives most effectively. All samples will be stored in small plastic bags on ice in the field and subsequently frozen.

2.11.2. Processing Protocols

Stable isotope samples will be oven dried at 50-60°C to a constant weight and ground to a homogenous powder using a mortar and pestle. Aquatic macroinvertebrate samples will be separated into four subsamples by functional group (i.e., grazers, collectors, shredders, and predators), and caddisfly larvae will be removed from their cases before drying and grinding. Subsamples of approximately 3-4 mg for algae, 4-6 mg for OM, and 1 mg for animal tissue will be weighed to the nearest 0.001 mg on a micro-analytical balance and placed into tin capsules. Samples will be combusted and analyzed in an isotope-ratio mass spectrometer interfaced with an elemental analyzer. Data will be normalized using isotope reference standards, and analytical precision will be estimated by analyzing a subset of samples in duplicate.

2.11.3. Data Analysis Methods

The stable isotope analysis will be conducted to determine the relative contributions of freshwater, terrestrial, and marine nutrients to focal salmonid species along an upstream-downstream gradient and among habitat types in the river (Wipfli and Baxter 2010). Stable isotope signatures are conventionally reported in δ units, which indicate the ratio of heavy to light atoms in a sample, relative to a standard. Stable isotope signatures of C and N will be calculated as $\delta^{13}\text{C}$ or $\delta^{15}\text{N} = [(R \text{ sample} / R \text{ standard}) - 1]1000$, where R is $^{13}\text{C}/^{12}\text{C}$ or $^{15}\text{N}/^{14}\text{N}$ (Peterson and Fry 1987). Spawning salmon are expected to exhibit an enriched signature of $\delta^{15}\text{N}$ relative to freshwater or terrestrial energy sources (Bilby et al. 1996; Chaloner et al. 2002; Satterfield and Finney 2002), and the combination of the $\delta^{15}\text{N}$ and $\delta^{13}\text{C}$ signatures may additionally allow all three sources to be distinguished (Fry 2006).

To evaluate whether differences in lipid content among samples might influence isotopic signatures, variability in the elemental ratio of C:N will be examined after the samples collected during 2013 have been analyzed (Post et al. 2007). This ratio is measured and reported as part of the $\delta^{13}\text{C}$ and $\delta^{15}\text{N}$ analysis. If variability in C:N ratios is great enough to potentially influence the study conclusions (i.e., if the range of values exceeds approximately 4 percentage points), then mathematical lipid normalization approaches (Kiljunen et al. 2006; Post et al. 2007) will be evaluated to correct for this variability.

Variability in the diet composition of each focal salmonid species and age class will be evaluated with respect to sampling location (i.e., upriver vs. downriver) and habitat type. Broad patterns of energy flow within the riverine food web will be examined graphically by plotting $\delta^{13}\text{C}$ vs. $\delta^{15}\text{N}$ for all samples. For each focal salmonid species and age class, diet composition will be estimated and compared among locations and habitats using stable isotope mixing models, if the final dataset meets the assumptions of these techniques, including adequate sample sizes, contrast in isotopic signatures, and suitable geometry of the $\delta^{13}\text{C}$ vs. $\delta^{15}\text{N}$ plot (Moore and Semmens 2008; Semmens et al. 2009). Alternatively, if the mixing model approach is not well suited to the dataset, variability in the $\delta^{13}\text{C}$ and $\delta^{15}\text{N}$ signatures of the focal salmonids relative to other taxa may be evaluated directly using MANOVA, 2-dimensional Kolmogorov-Smirnov tests, or the most appropriate analytical approach given the distributional characteristics of the data.

2.12. Data Management

The goals of data management are to establish a data QA/QC protocol to be applied by study teams at logical stages of data collection and processing and to ultimately create a relational database including all finalized river productivity data collected for the Susitna-Watana Project.

2.12.1. Established QA/QC Protocol

- There will be 5 levels of data QC, named QC1 to QC5, each of which is tracked either within tabular datasets (as for Excel and database tables), or within file path names (as for raw field data files). This allows for quick determination of the QC status of all data.
- Details for the QC Protocol are found in Appendix 3: Susitna Field Data Standards.
- The QC levels, briefly, are as follows:

- QC1** – Field Review: Review of field forms before leaving the field, or the QC level of raw data collected via field equipment such as thermistors, cameras, GPS units, etc.
- QC2** – Data Entry: Data from paper forms are entered into an electronic format and verified.
- QC3** – Senior Review: Final review by senior professional before submitting field data to AEA, or the QC level of raw data cleaned up for delivery to AEA.
- QC4** – Database Validation: Tabular data files are verified to meet project database standards.
- QC5** – Technical Review: Data revision or qualification by senior professionals when analyzing data for reports.

2.12.2. Relational Database

A database template is being designed to store the river productivity data from all consultants and studies, providing a centralized data tool for users. The final database will be maintained in MS Access software and will include data collected in 2012, and new data from future studies in 2013 and 2014. It may include data submitted from other entities, such as ADF&G, UAF, and other contracted processing labs. The database will be available for querying and analysis by parties assigned by AEA.

A data dictionary describing the database entities and attributes will be compiled, to accompany the database and to provide an understanding of data elements and their use by anyone querying or analyzing the data.

See Appendix 4 for a template of the River Productivity database.

3. SCHEDULE

The preliminary schedule for the river productivity study elements is presented in Table 3.1-1. Field sampling at the Susitna River sites and the Talkeetna River test reference sites for benthic macroinvertebrates, algae, organic matter, drift, fish diet analysis, and stable isotopes will be conducted for three seasonal sampling periods from April through October in both study years (2013 and 2014). These seasonal periods are tentatively scheduled for April through early June for Spring, late June through August for Summer, and September through October for Autumn (Table 3.1-1), due to annual variability in the timing of seasons. In addition, seasonal sampling must be conducted within select flow ranges and stages. Higher flows may inundate new shoreline substrates, which present a risk of sampling in areas disturbed by periodic inundation and dewatering and will not be fully colonized. Therefore, changes in water level due to increasing or decreasing flows must remain constant enough that the substrates accessible for sampling will be continually inundated for a period of at least one month, to facilitate colonization of those substrates prior to sampling. Based on the criteria for sampling, the schedule will be determined within a window of several weeks. Multiple remote cameras and staff gages have been installed along the Susitna River, and these, along with the USGS gage at Gold Creek, will be closely monitored for target sampling conditions based on indicators such as flow and river stage conditions.

Two additional sampling events are planned for benthic macroinvertebrates, algae, and organic matter under storm conditions and will occur sometime during April through October. Specific dates are determined by criteria previously mentioned that will trigger storm event sampling.

Sample processing of organisms and materials collected in the 2013 field efforts will require extensive laboratory taxonomic analysis, and will continue throughout the remainder of 2013 and into the first quarter of 2014. Trophic analysis efforts will be initiated during the latter half of the first quarter of 2013 and continue throughout the rest of 2013 and into 2014.

Second-year field sampling efforts, adhering to the same tentative scheduling as in 2013, will resume in the latter half of the first quarter of 2014, with sample processing, data analysis, trophic analysis research continuing through the fourth quarter.

4. FIELD EQUIPMENT LIST

To be provided in Final Implementation Plan.

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6. TABLES

Table 1.2-1. Locations and descriptions of proposed Focus Areas selected for the River Productivity study in the Middle River Segment of the Susitna River. Focus Area identification numbers (e.g., Focus Area 184) represent the truncated Project River Mile (PRM) at the downstream end of each Focus Area.

Focus Area ID / RivProd ID	Common Name	River Productivity Study Use	Description	Geomorphic Reach	Location (PRM)		Area Length (mi)	Habitat Types Present							Fish use in 1980s	
					Upstream	Downstream		Main Channel, Single	Main Channel, Split	Side Channel	Tributary Mouth	Side Slough	Upland Slough	Beaver Complex	Spawning	Rearing
RP- 248	Complex Area 248	Study Station (3 sites)	Area with channel complex approximately 1.7 miles upstream of Tyone River	UR-1	250.1	248.8	1.3	X	X	X		X			N/A	N/A
RP-233	Oshetna River	Study Station (3 sites)	Area covering Oshetna River mouth and downstream	UR-2, 3	235.4	233.9	1.5	X			X				N/A	N/A
Focus Area-184	Watana Dam	Study Station (3 sites)	Area approximately 1.4 miles downstream of dam site	MR-1	185.7	184.7	1.0	X	X	X					N/A	N/A
Focus Area-173	Stephan Lake, Complex Channel	Study Station (3 sites)	Wide channel near Stephan Lake with complex of side channels	MR-2	175.4	173.6	1.8	X		X	X	X			N/A	N/A
Focus Area-144	Side Channel 21	Storm Event Site	Side channel and side slough complex approximately 2.3 miles upstream Indian River	MR-6	145.7	144.4	1.3	X	X	X	X	X		X	X	X
Focus Area-141	Indian River	Study Station (3 sites)	Area covering Indian River and upstream channel complex	MR-6	143.4	141.8	1.6	X	X	X	X		X	X	X	X
Focus Area-104	Whiskers Slough	Study Station (3 sites), Storm Event Site	Whiskers Slough Complex	MR-8	106.0	104.8	1.2	X	X	X	X	X	X		X	X

Table 2.2-1. Descriptive metrics commonly used in aquatic ecological studies to describe benthic macroinvertebrate (BMI) communities.

Biological Metrics	Description	Predicted Response to Impairment
Abundance Measures		
Density	The total number of individuals collected in a unit area (m ²)	variable
Richness Measures		
Taxa Richness	Total number of individual taxa	decrease
Ephemeroptera Taxa	Number of mayfly taxa	decrease
Plecoptera Taxa	Number of stonefly taxa	decrease
Trichoptera Taxa	Number of caddisfly taxa	decrease
Shannon-Weiner Diversity Index	Summary metric that combines taxa richness and abundances, calculated with the natural logarithm (ln)	decrease
Composition Measures		
Percent Composition: Major Taxa	Relative abundances of: Ephemeroptera, Plecoptera, Trichoptera, Coleoptera, Chironomidae, non-chironomid Diptera, other Insect taxa, and non-insect taxa	variable
Percent Dominant Taxa	Percent composition of the three most abundant taxa	increase
EPT:Chironomid Ratio	Ratio of EPT abundance to Chironomidae abundance, ranging from 0 to 1, with scores below 0.5 indicating more Chironomidae.	decrease
Tolerance/Intolerance Measures		
Biotic Index	Value between 0 and 10 weighted for abundance of individuals designated as pollution tolerant (higher values) and intolerant (lower values)	increase
Intolerant Taxa	Number of taxa in sample that are highly intolerant to impairment (tolerance value ≤ 4)	decrease
Percent Tolerant Organisms	Percent of organisms in sample that are highly tolerant to impairment (tolerance value ≥ 7)	increase
Functional Feeding Groups		
Percent Collector-Gatherers	Percent of macrobenthos that gather fine particulate matter	increase
Percent Collector-Filterers	Percent of macrobenthos that filter fine particulate matter	increase
Percent Scrapers (Grazers)	Percent of macrobenthos that graze upon periphyton	variable
Percent Predators	Percent of macrobenthos that feed on other organisms	variable
Percent Shredders	Percent of macrobenthos that shreds coarse particulate matter	decrease
Percent Other Groups	Percent of macrobenthos that are either omnivorous, macrophyte or piercer herbivores, or parasites	variable
Habits /Life History Measures		
Clinger Taxa	Number of taxa with physical adaptations that allow them to hold onto smooth substrates in fast water	decrease
Long-lived Taxa	Number of taxa that require more than 1 year to complete their life-cycles (semi-voltine)	decrease

Table 2.8-1. Itemized listing of the number of fish gut content samples to collect for the River Productivity Study in each study year.

Target Species / Lifestage	Sites	Seasons	Samples	Total
Chinook salmon - juveniles	18	3	8	432
Coho salmon - juveniles	18	3	8	432
Rainbow trout - juveniles	18	3	8	432
Rainbow trout - adults	18	3	8	432
Total				1,728

Table 2.11-1. Itemized listing of sample components to collect for Stable Isotope Analysis at the two sampling stations (6 sites total) in each study year in the Middle Segment of the Susitna River for the River Productivity Study.

Category	Taxon	Sites	Seasons	Samples	Total
Endmembers	Benthic Algae	6	3	5	90
	Organic Matter - benthic	6	3	5	90
	Organic Matter - drift	6	3	2	36
	Salmon carcass	2	1	20	40
Invertebrates	Benthic- grazers	6	3	5	90
	Benthic- collectors	6	3	5	90
	Benthic- shredders	6	3	5	90
	Benthic- predators	6	3	5	90
	Terrestrial Drift	6	3	2	36
	Emergents	6	3	1	18
Fish	Chinook salmon - juveniles	6	3	8	144
	Coho salmon - juveniles	6	3	8	144
	Rainbow trout - juveniles	6	3	8	144
	Rainbow trout - adults	6	3	8	144
Total					1,246

Table 3.1-1. Preliminary schedule for River Productivity Study.

Activity	2013				2014				2015
	1Q	2Q	3Q	4Q	1Q	2Q	3Q	4Q	1Q
Literature Review on Hydropower Impacts		—	—	—					
Sampling benthic macroinvertebrate communities, algal communities, and organic matter.		⊙	⊙	⊙		⊙	⊙	⊙	
Invertebrate drift sampling		⊙	⊙	⊙		⊙	⊙	⊙	
Sampling Talkeetna for Reference Site Feasibility Study		⊙	⊙	⊙					
Trophic analysis with bioenergetics and stable isotope analysis		⊙	⊙	⊙		⊙	⊙	⊙	
Generate habitat suitability criteria					—	—	—	—	
Conduct a fish gut analysis		⊙	⊙	⊙		⊙	⊙	⊙	
Establish baseline colonization rates		—	⊙	⊙			⊙	⊙	
Data Analysis and Reporting			—	—	—	—	—	—	
Initial Study Report				—	Δ				
Updated Study Report								—	▲

Legend:

- Planned Activity
- ⊙ Tentatively scheduled sampling event
- Δ Initial Study Report
- ▲ Updated Study Report

7. FIGURES

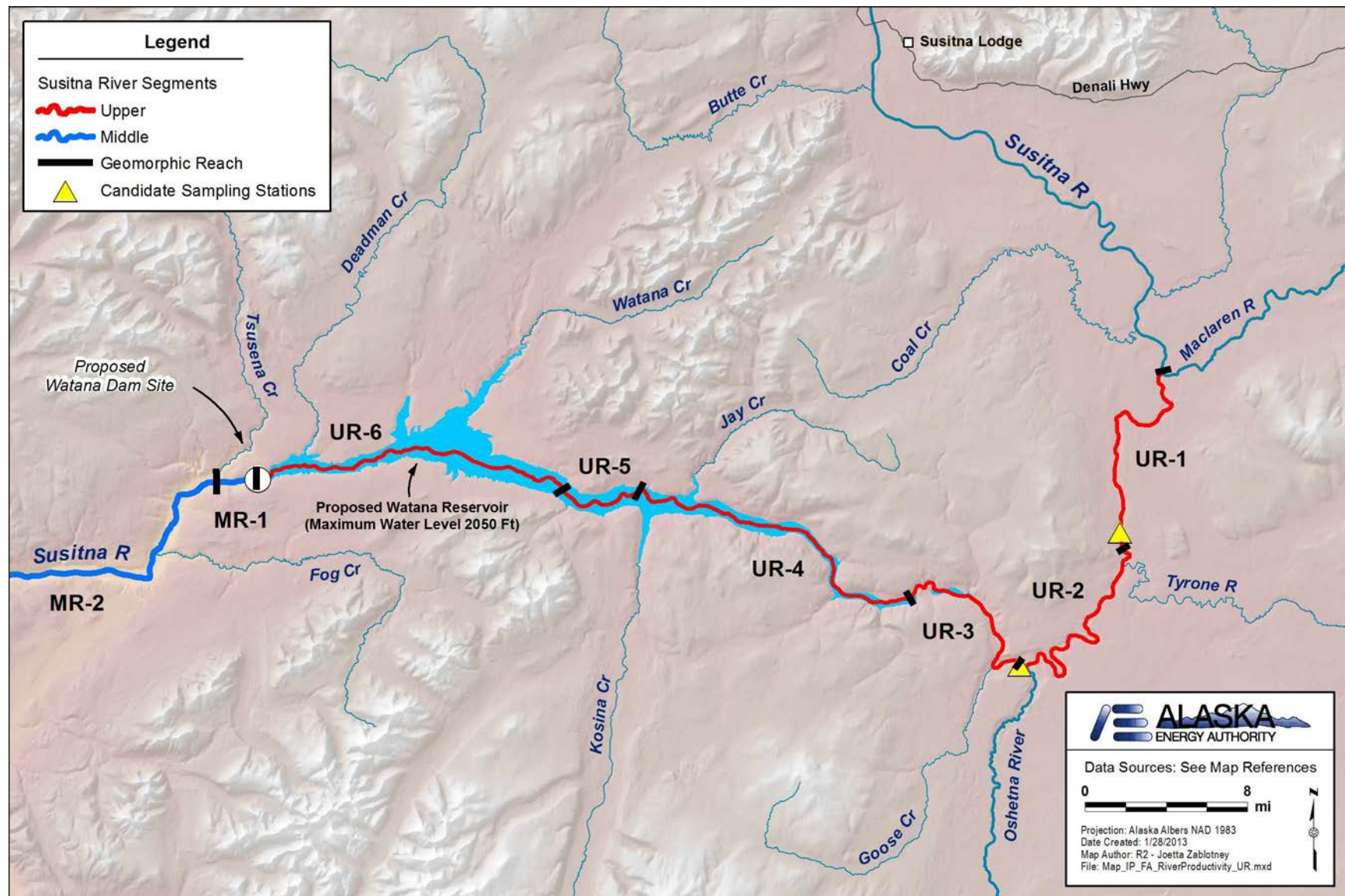


Figure 1.2-1. Upper Susitna River Segment, with the two proposed River Productivity sampling stations selected for the River Productivity Study.

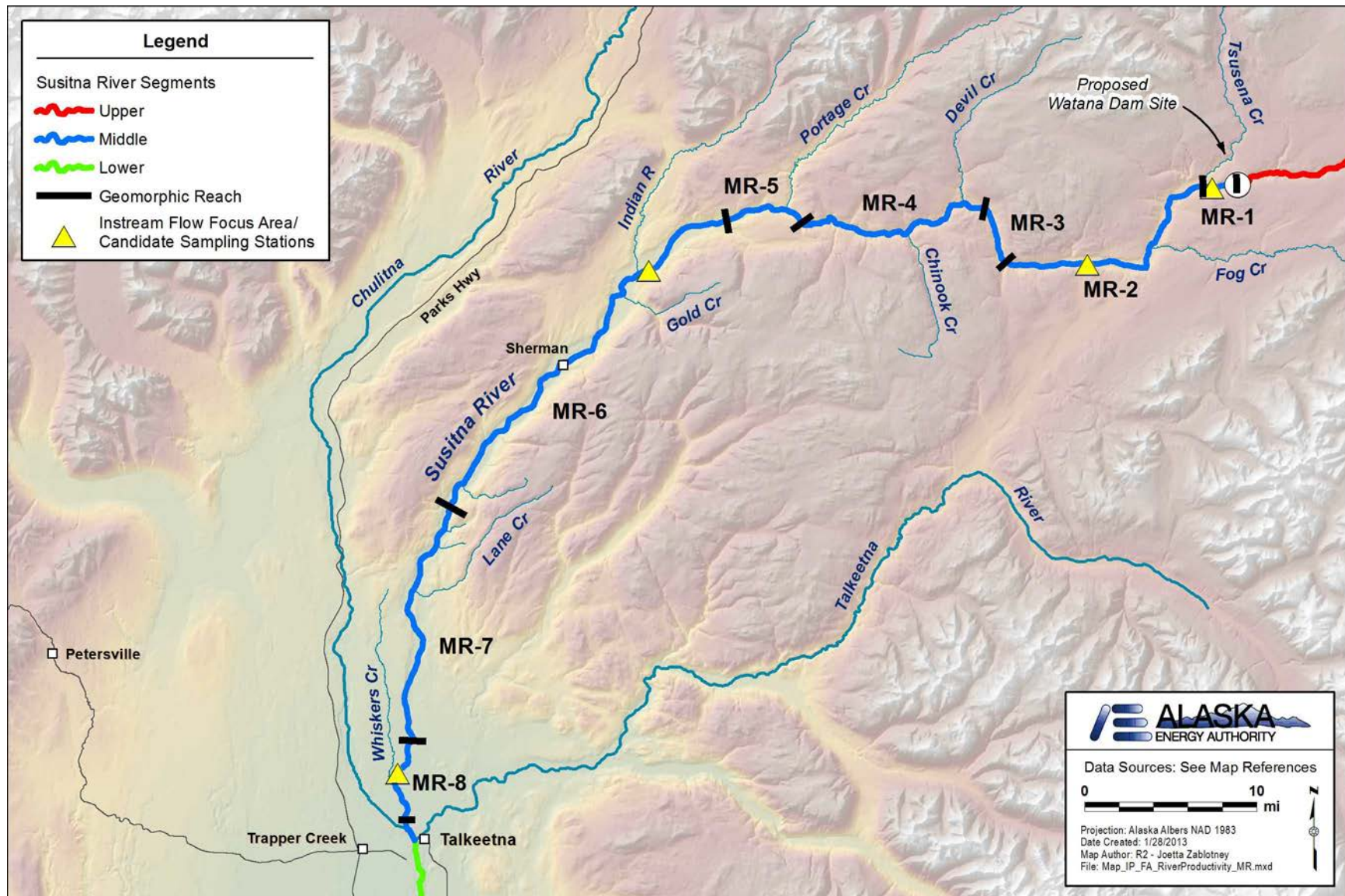


Figure 1.2-2. Middle Susitna River Segment, with the four proposed River Productivity sampling stations /Instream Flow Focus Areas selected for the River Productivity Study.

Total Catch of Coho Juvenile at DFH Sites From All Gear Types During 1982

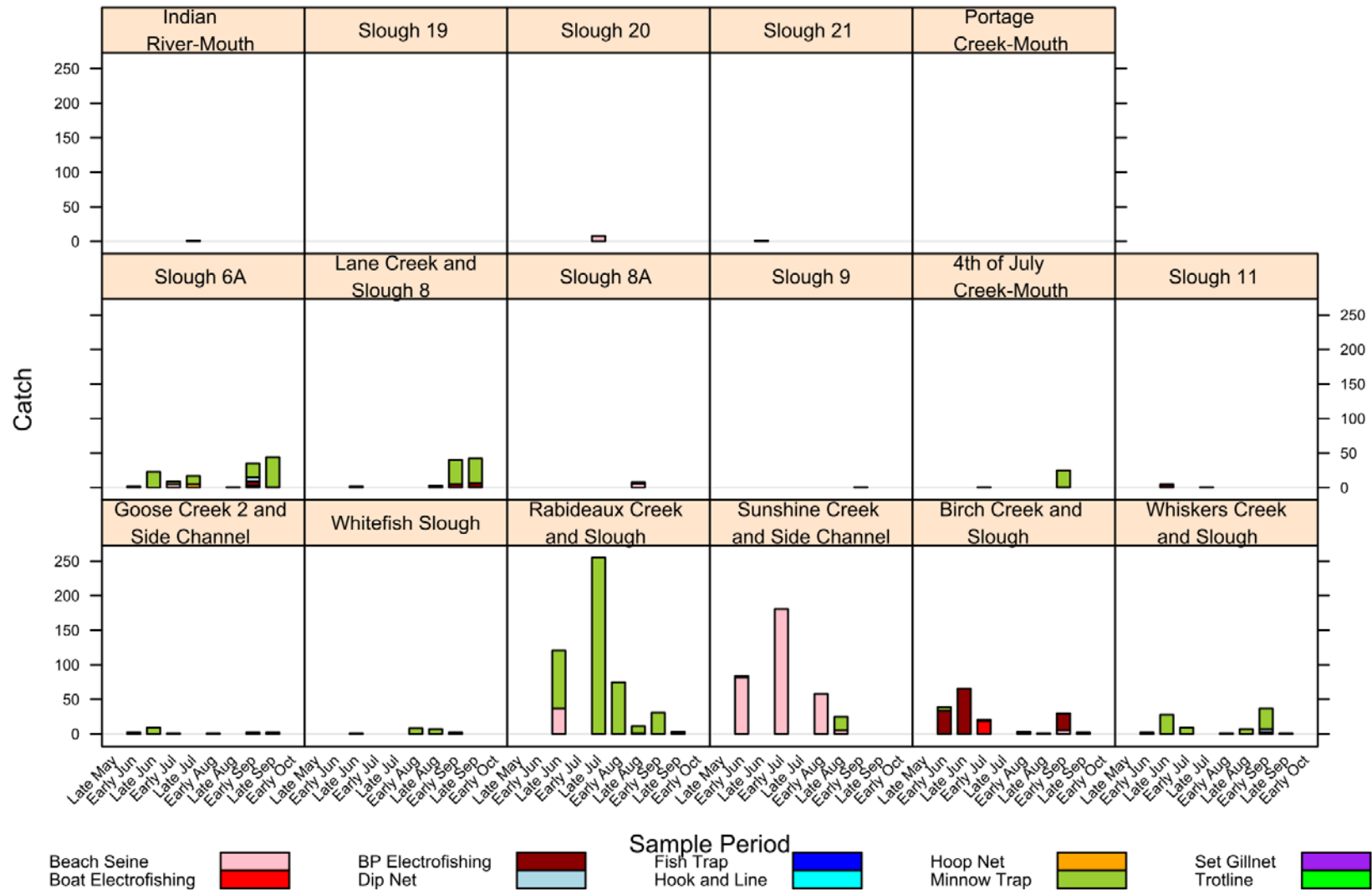


Figure 1.3-1. Total catch of juvenile coho salmon by sample period and gear type at DFH sites in 1982. Source: Estes and Schmidt 1983

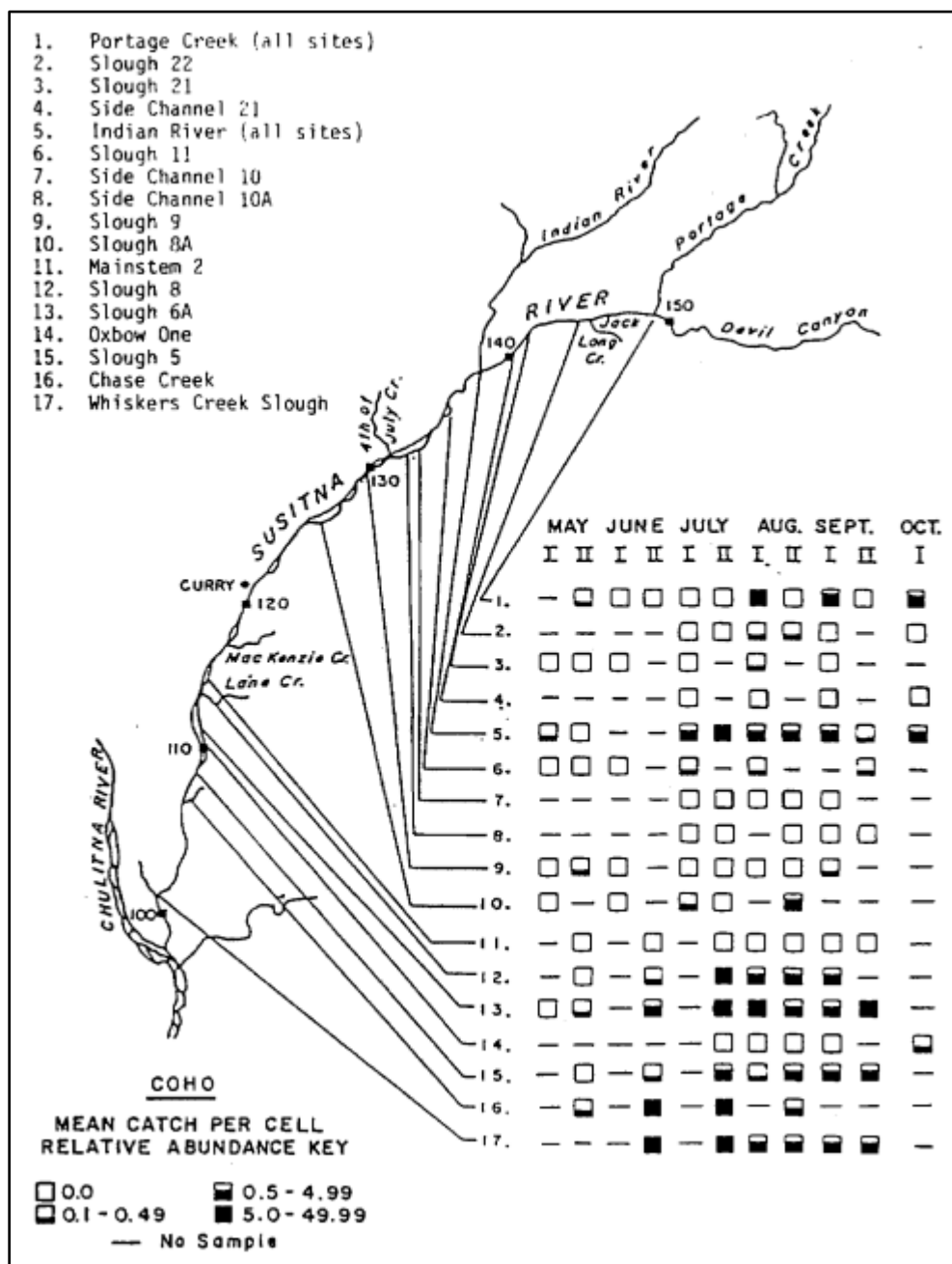


Figure 1.3-2. Seasonal distribution and relative abundance of juvenile coho salmon on the Susitna River between the Chulitna River confluence and Devil Canyon, May through November 1983. Source: Dugan et al. (1984).

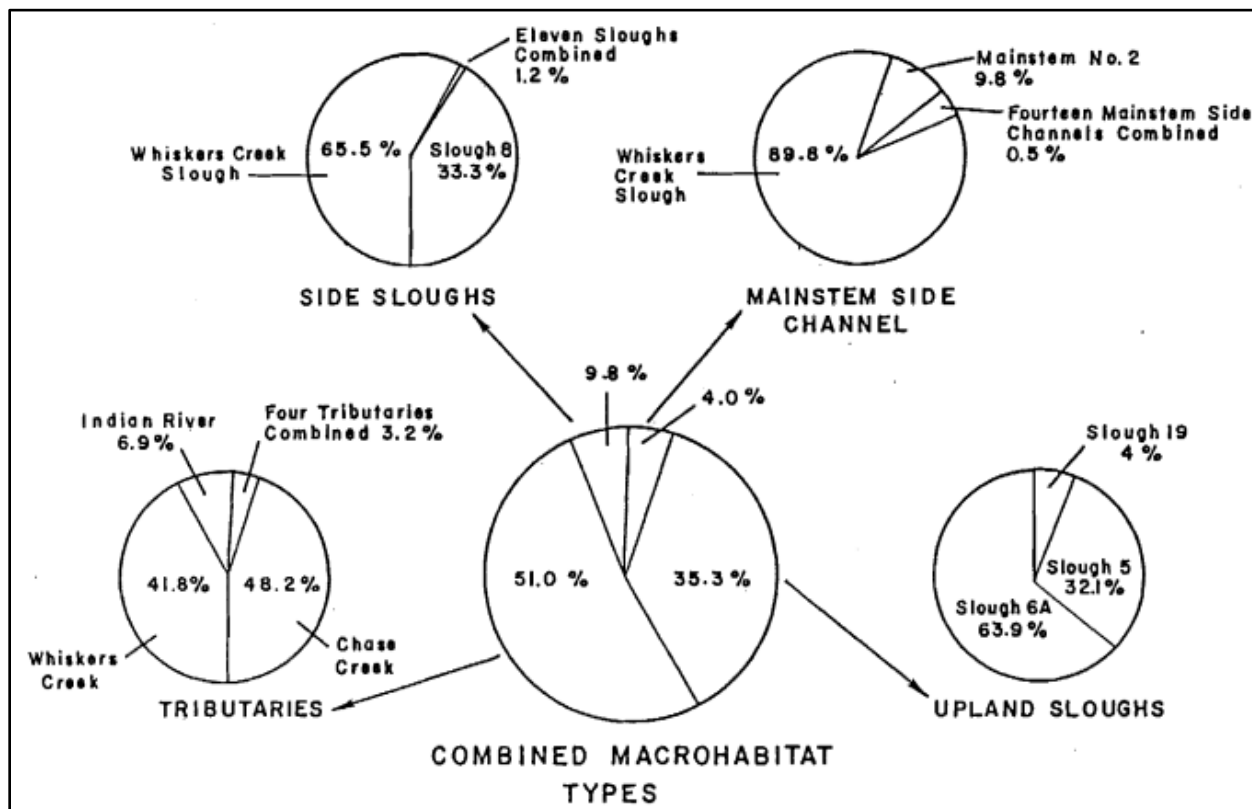


Figure 1.3.3. Density distribution and juvenile coho salmon by macrohabitat type on the Susitna River between the Chulitna River confluence and Devil Canyon, May through November 1983. Percentages are based on mean catch per cell. Source: Dugan et al. (1984).

Total Catch of Chinook Juvenile at DFH Sites From All Gear Types During 1982

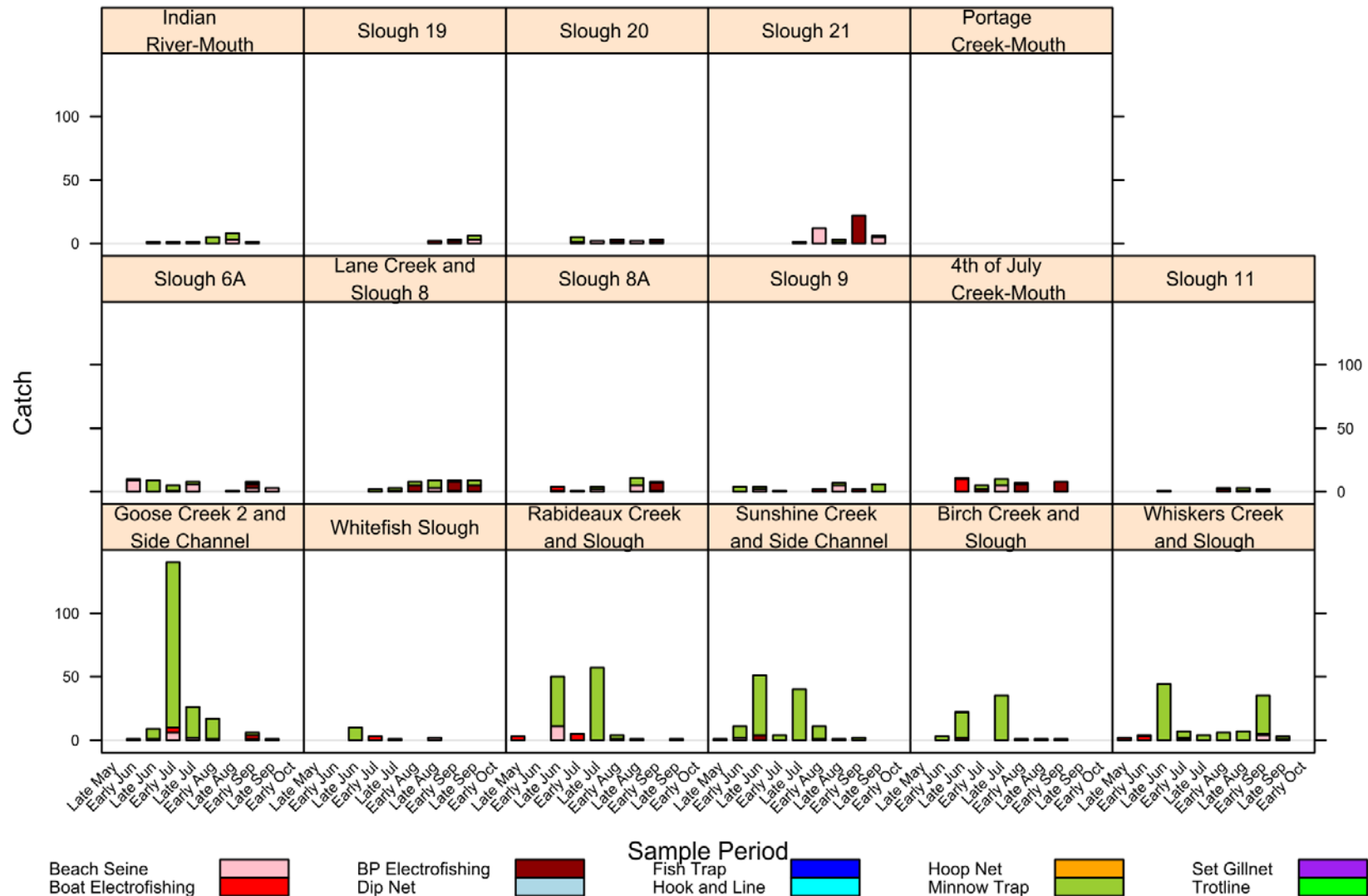


Figure 1.3-4. Total catch of juvenile Chinook salmon by sample period and gear type at DFH sites in 1982. Source: Estes and Schmidt 1983

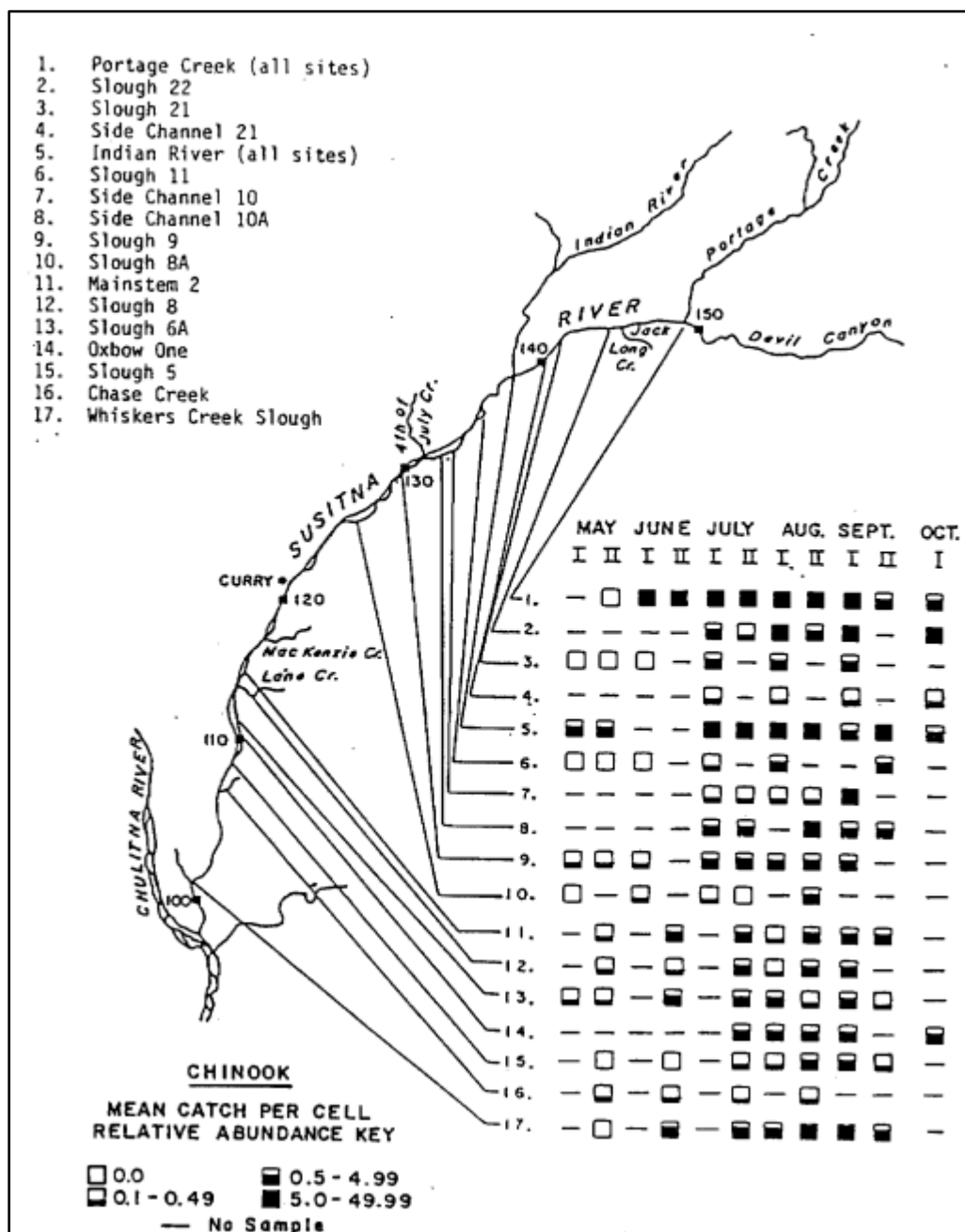


Figure 1.3-5. Seasonal distribution and relative abundance of juvenile Chinook salmon on the Susitna River between the Chulitna River confluence and Devil Canyon, May through November 1983. Source: Dugan et al. (1984).

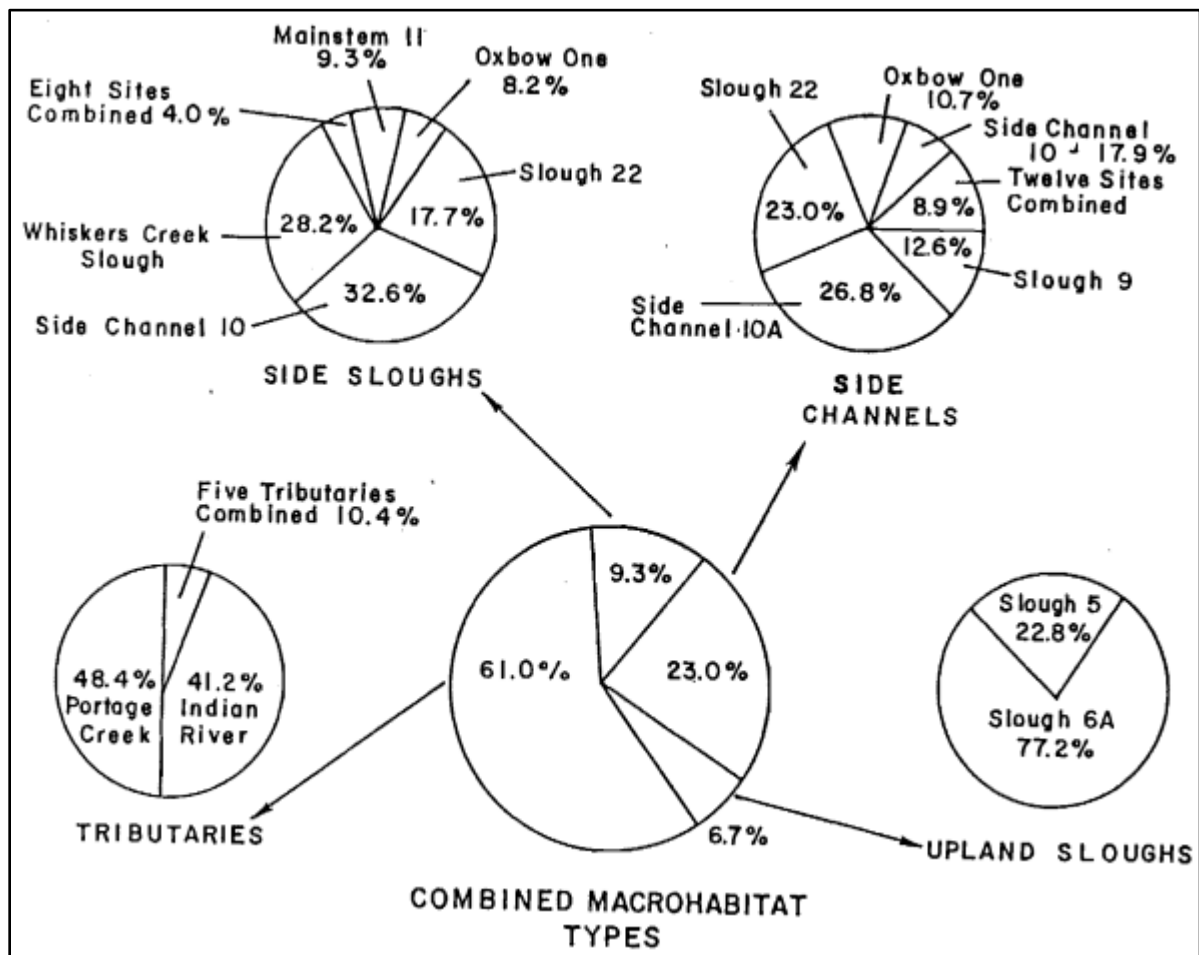


Figure 1.3-6. Density distribution and juvenile Chinook salmon by macrohabitat type on the Susitna River between the Chulitna River confluence and Devil Canyon, May through November 1983. Percentages are based on mean catch per cell. Source: Dugan et al. (1984).

Total Catch of Rainbow Trout at DFH Sites From All Gear Types During 1982

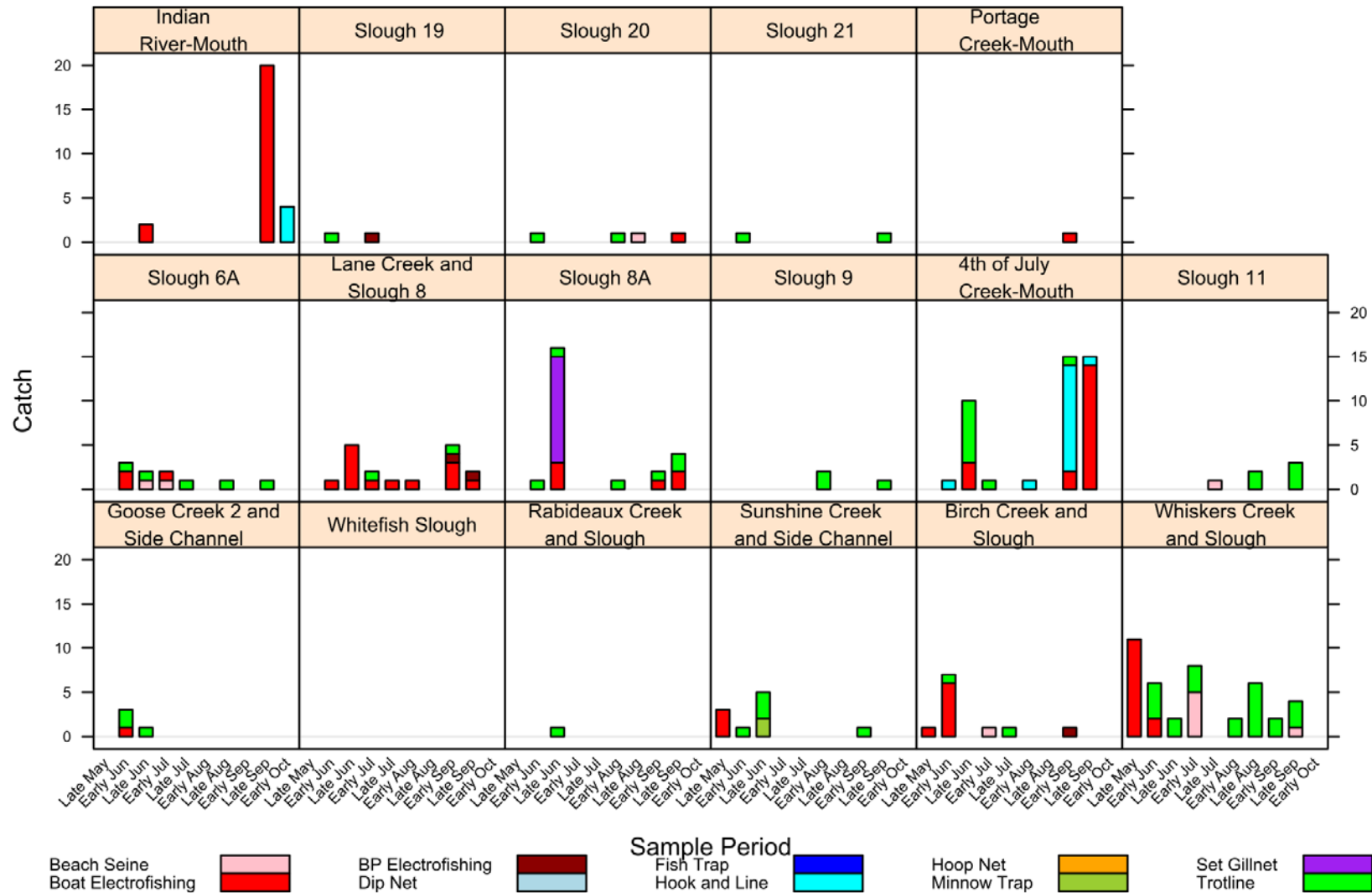


Figure 1.3-7. Total catch of rainbow trout by sample period and gear type at DFH sites in 1982. Source:

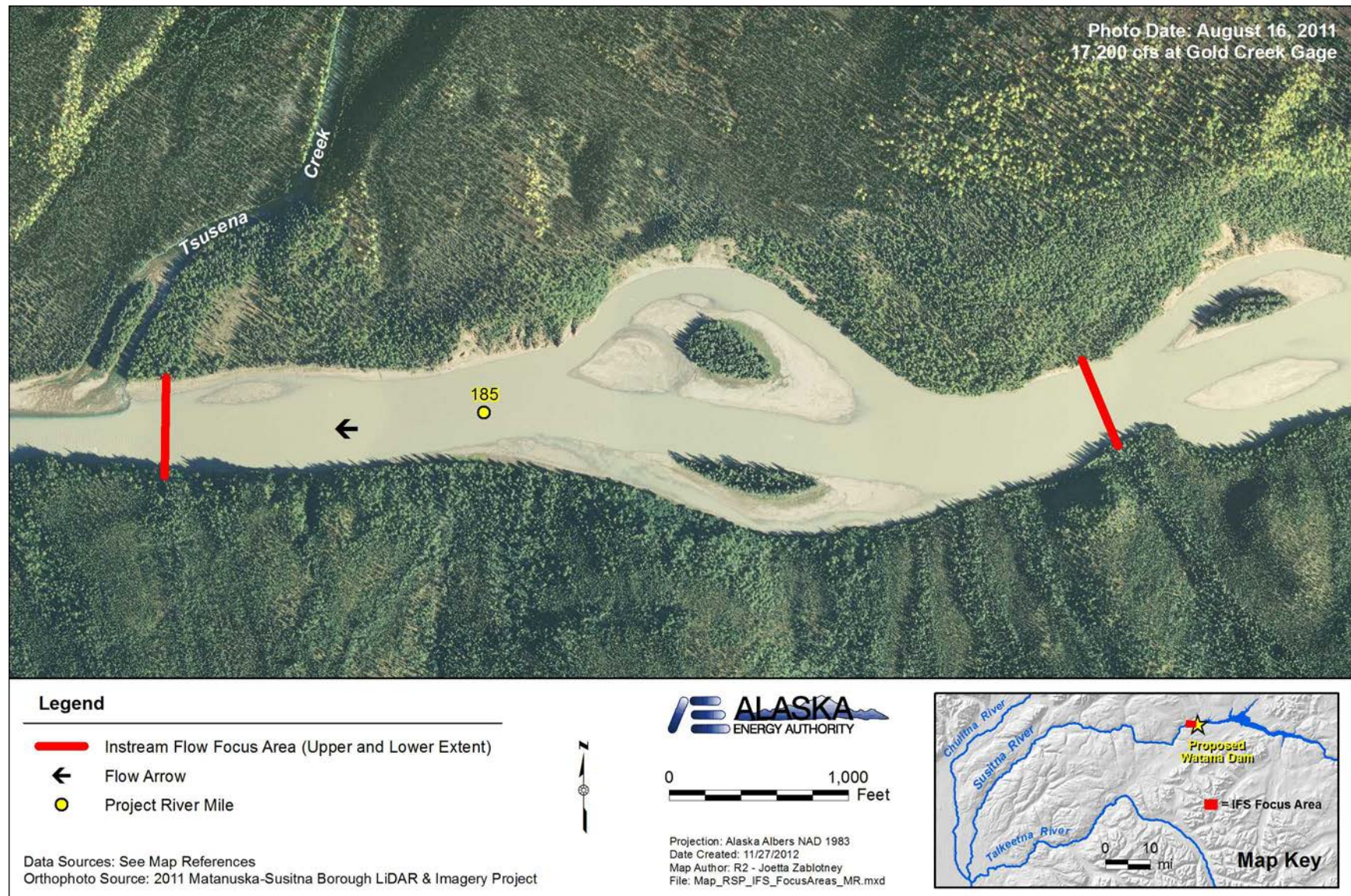


Figure 2.1-1. Map showing Focus Area 184 that begins at Project River Mile 184.7 and extending upstream to PRM 185.7. The Focus Area is located about 1.4 miles downstream of the proposed Watana Dam site near Tsusena Creek.

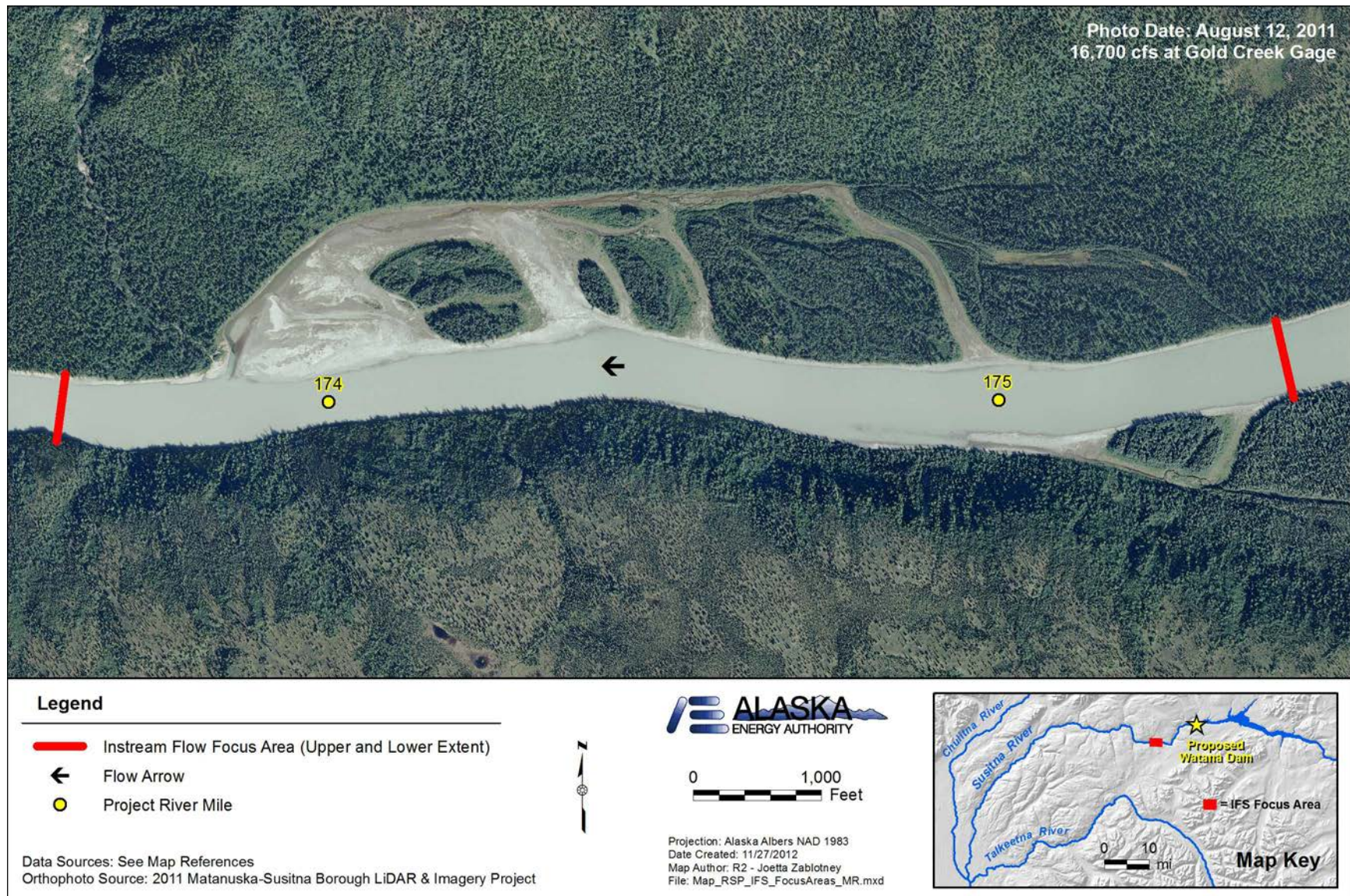


Figure 2.1-2. Map showing Focus Area 173 beginning at Project River Mile 173.6 and extending upstream to PRM 175.4. This Focus Area is near Stephan Lake and consists of main channel and a side channel complex.

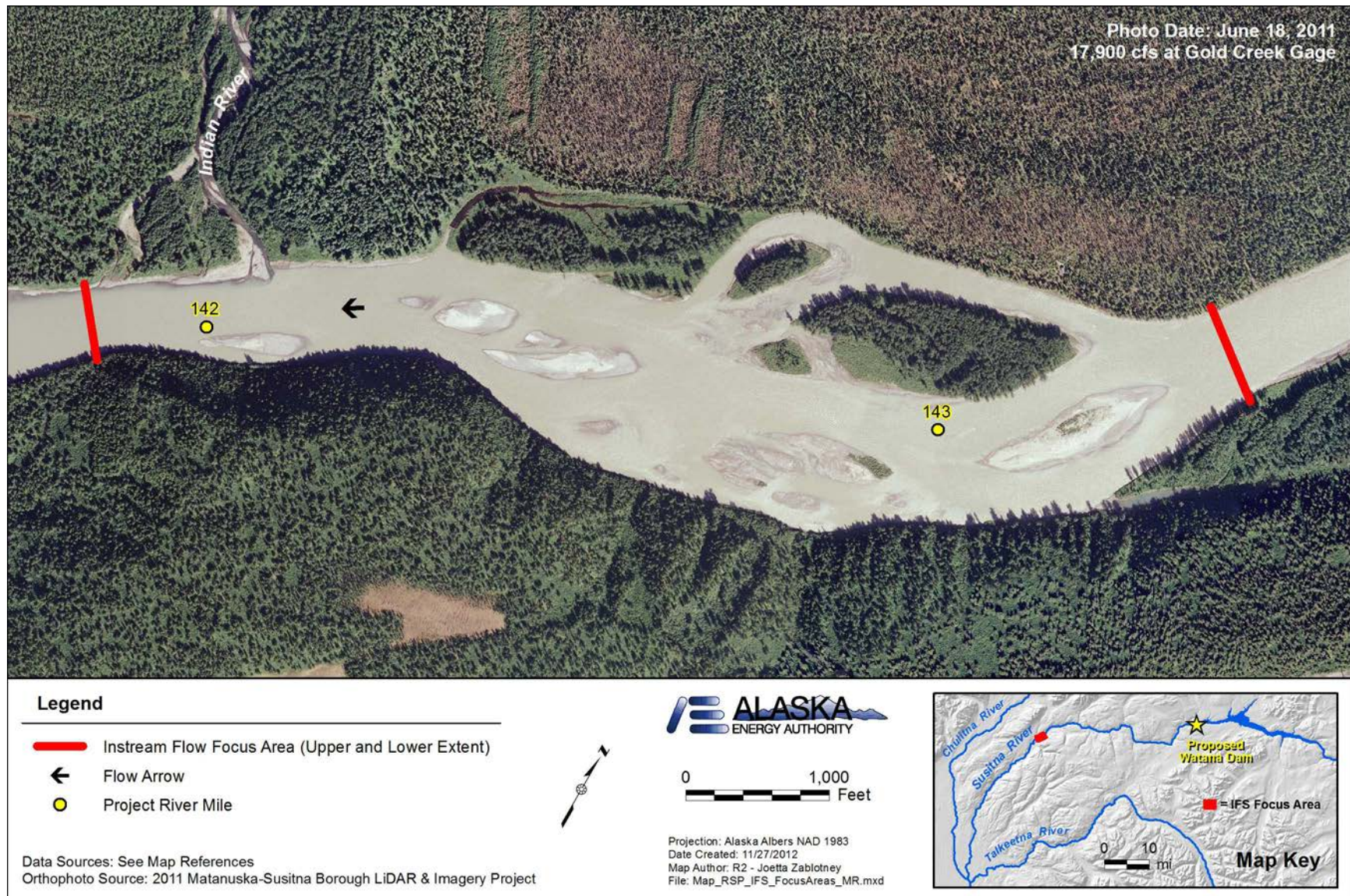


Figure 2.1-3. Map showing Focus Area 141 beginning at Project River Mile 141.8 and extending upstream to PRM 143.4. This Focus Area includes the Indian River confluence and a range of main channel and off-channel habitats.

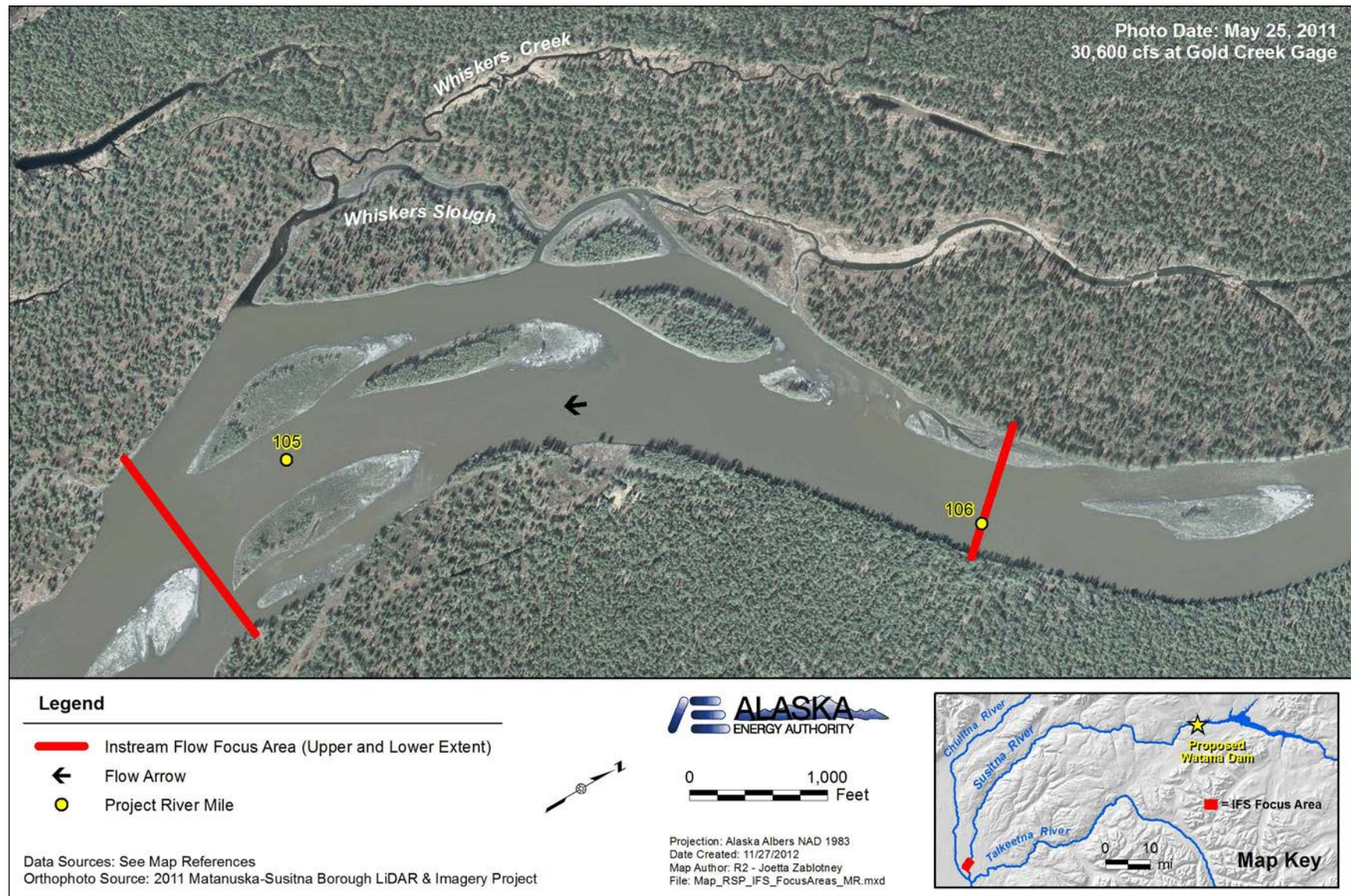


Figure 2.1-4. Map showing Focus Area 104 beginning at Project River Mile 104.8 and extending upstream to PRM 106. This Focus Area covers the diverse range of habitats in the Whiskers Slough complex.

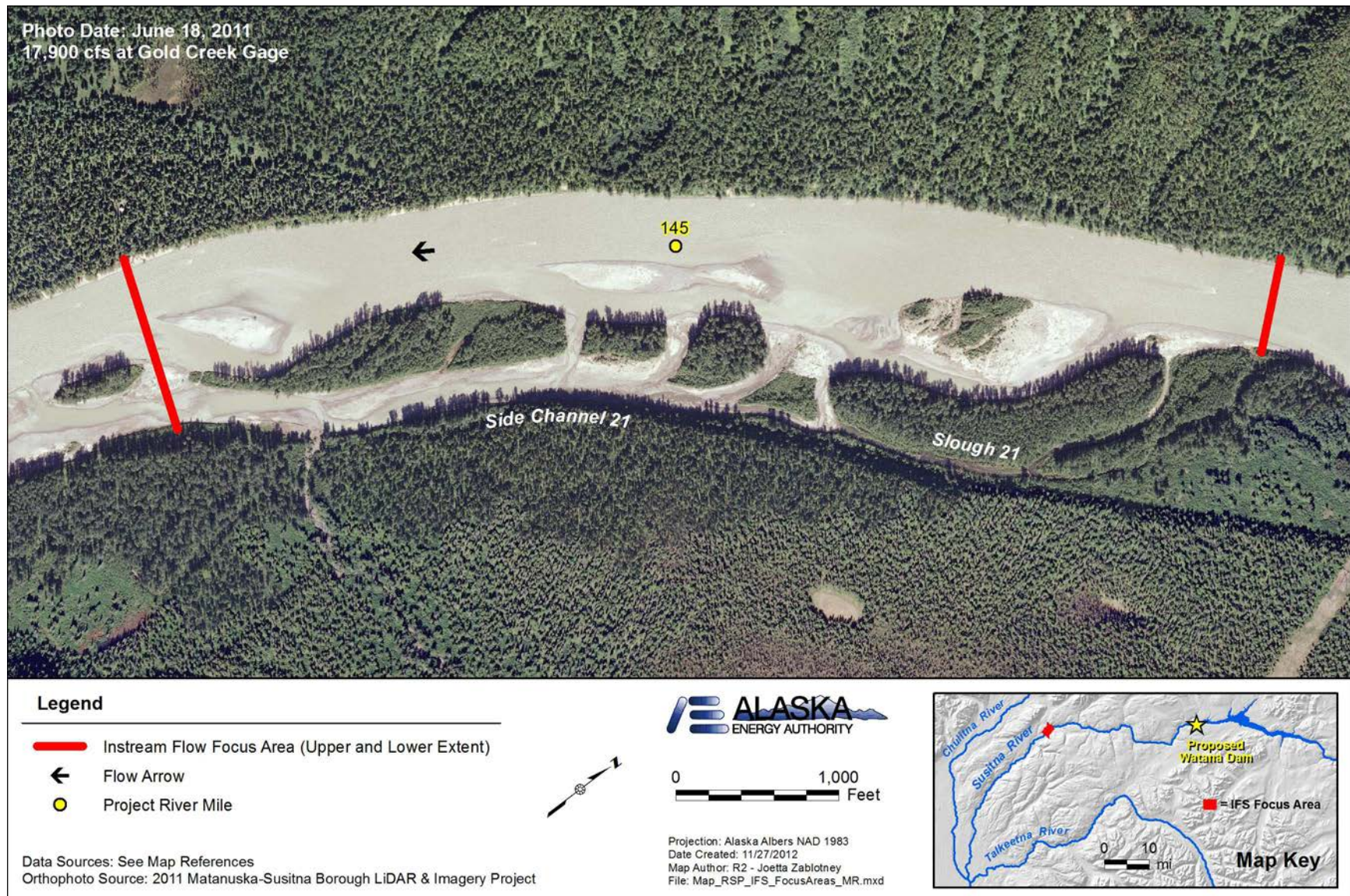


Figure 2.1-5. Map showing Focus Area 144 beginning at Project River Mile 144.4 and extending upstream to PRM 145.7. This Focus Area is located about 2.3 miles upstream of Indian River and includes Side Channel 21 and Slough 21.

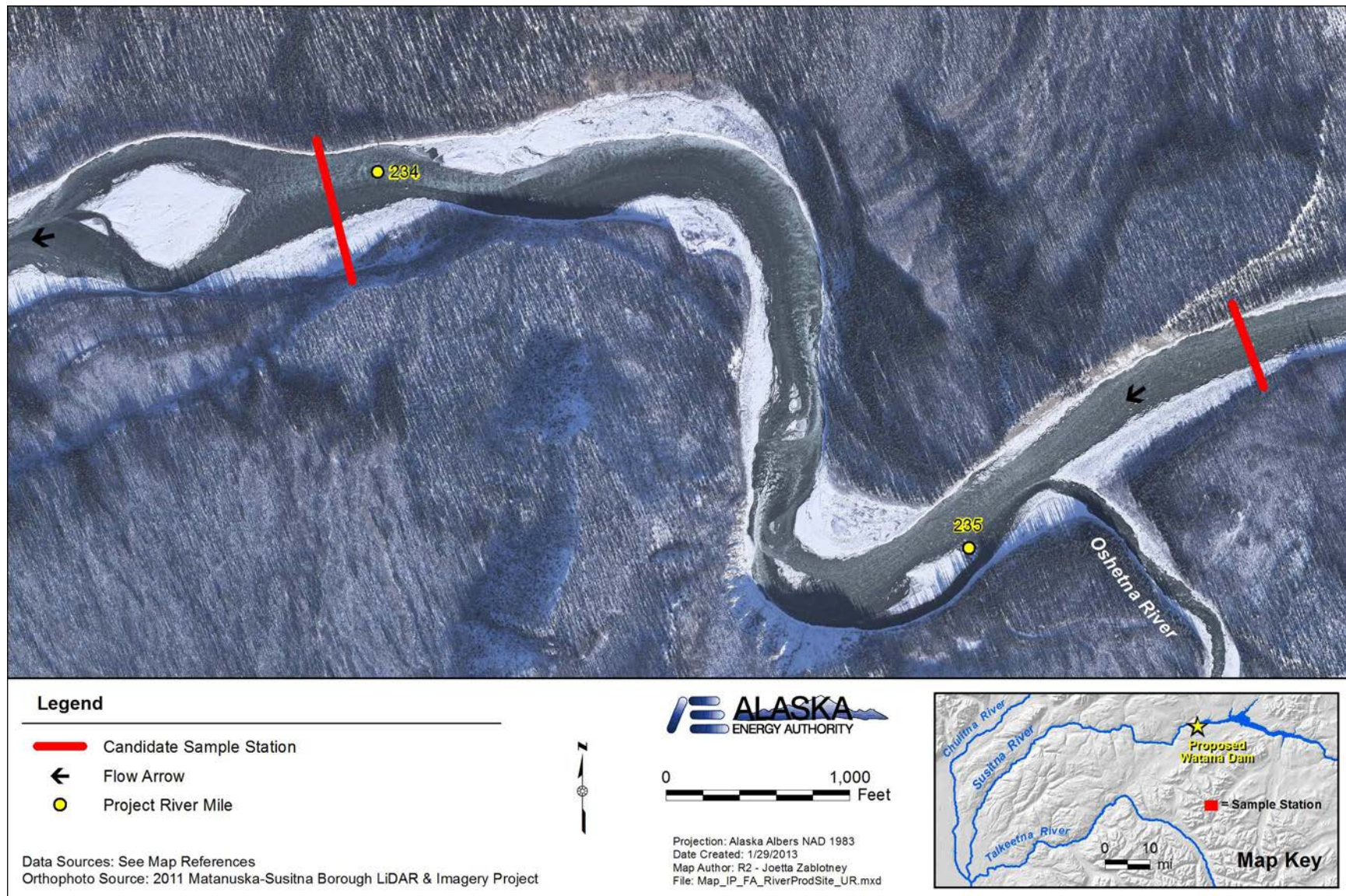


Figure 2.1-6. Map showing the River Productivity Upper Segment sampling station RP-233, located near the mouth of the Oshetna River beginning approximately at Project River Mile 233.9 and extending upstream to approximately PRM 235.4.

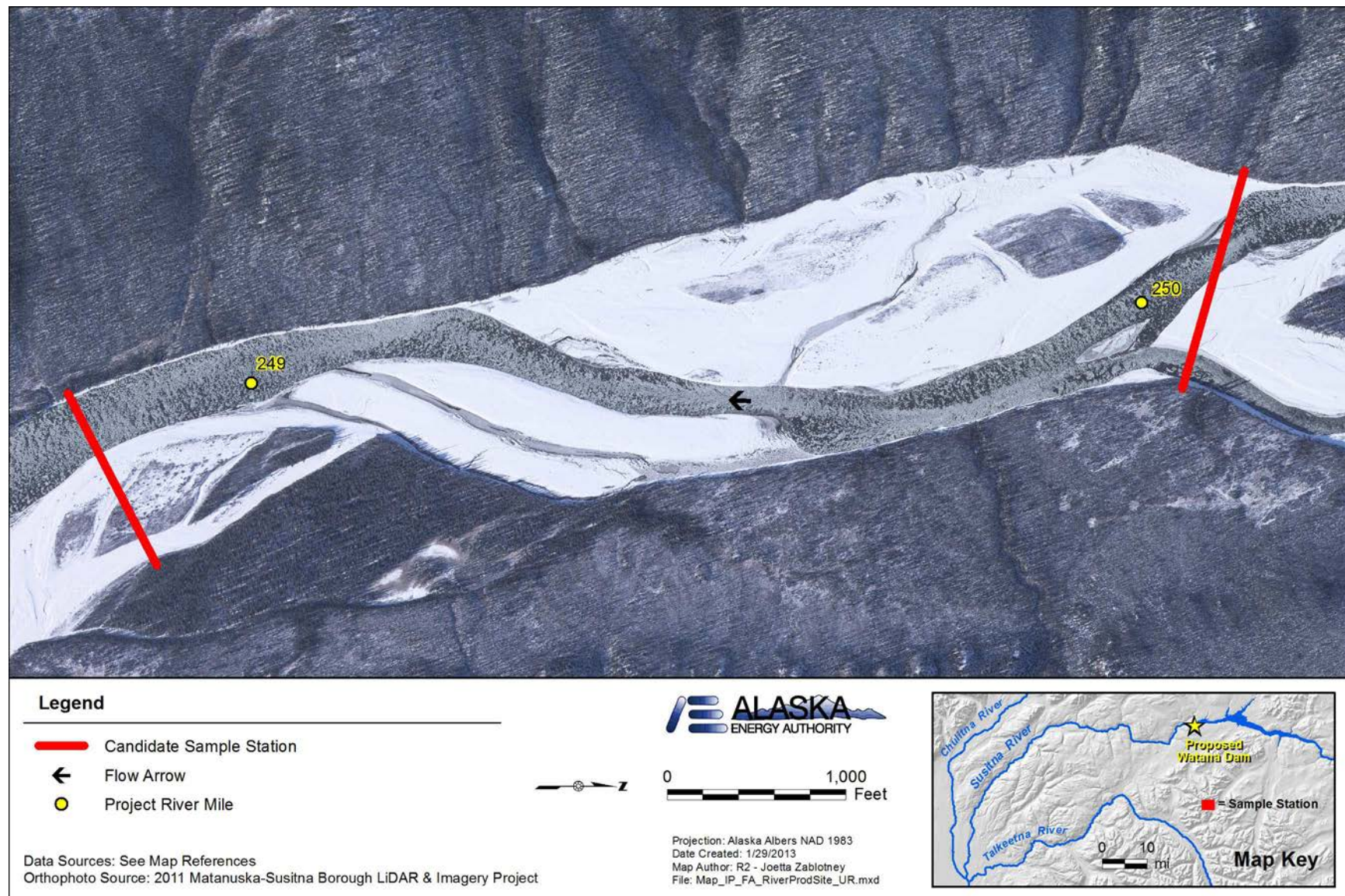


Figure 2.1-7. Map showing the River Productivity Upper Segment sampling station RP-248, located upstream of the Tyone River beginning approximately at Project River Mile 248.8 and extending upstream to approximately PRM 250.1.

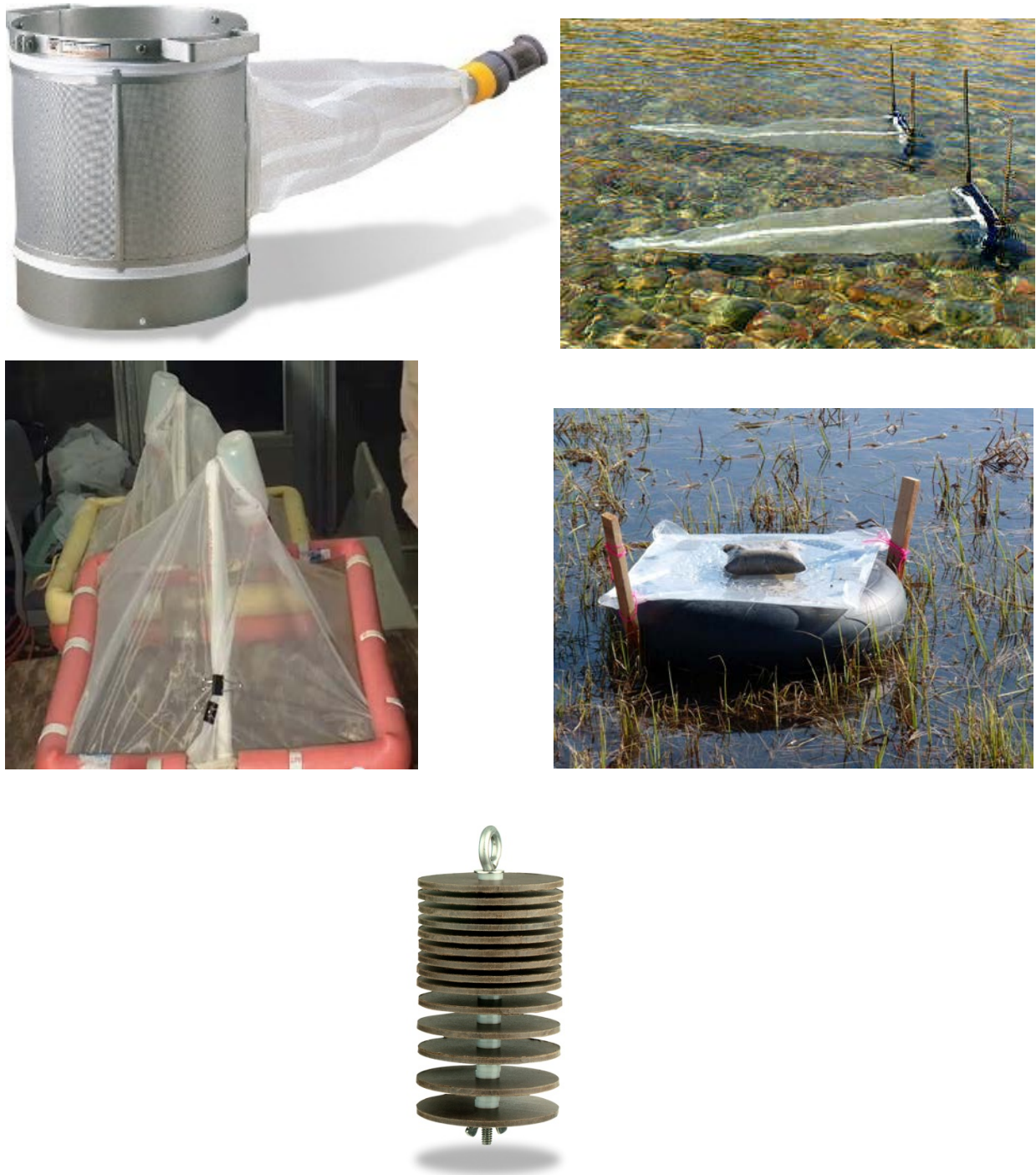


Figure 2.2-1. Sampling equipment used to collect benthic macroinvertebrates in streams and rivers. Top left: Hess stream sampler. Top right: drift net. Middle: examples of floating aquatic insect emergence traps. Bottom: Hester-Dendy multiplate sampler.

APPENDIX 1. MAJOR AND BARBOUR (2001). STANDARD OPERATING PROCEDURES FOR THE ALASKA STREAM CONDITION INDEX: METHOD 002 – LABORATORY PROCESSING

[Placeholder for Appendix 1]

APPENDIX 2. FIELD DATA FORMS

To be provided prior to initiation of field data collection

APPENDIX 3. SUSITNA DATA STANDARDS

[Placeholder for Appendix 3]

APPENDIX 4. DRAFT DATABASE TEMPLATES

[Placeholder for Appendix 4]