Susitna-Watana Hydroelectric Project (FERC No. 14241)

Susitna River Productivity Study Implementation Plan

Prepared for

Alaska Energy Authority

SUSITNA-WATANA HYDRO

Clean, reliable energy for the next 100 years.

Prepared by

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LIST OF ACRONYMS AND SCIENTIFIC LABELS

Abbreviation	Definition
ADF&G	Alaska Department of Fish and Game
AEA	Alaska Energy Authority
APA Project	Alaska Power Authority Project
CCA	canonical correspondence analysis
cfs	cubic feet per second
chl-a	chlorophyll-*a*
cm	centimeter
CPUE	catch per unit effort
DFH	Designated Fish Habitat
FERC	Federal Energy Regulatory Commission
GIS	Geographic Information System
HSC	habitat suitability criteria
ILP	Integrated Licensing Process
m^2	square meter
mg	milligram
MR	Middle River
NTU	nephelometric turbidity unit
PCA	principle components analysis
PM&E	protection, mitigation, and enhancement
PRM	Project River Mile
Project	Susitna-Watana Hydroelectric Project
QA/QC	quality assurance/quality control
RM	River Mile
RSP	Revised Study Plan
TWG	Technical Workgroup
UCI	Upper Cook Inlet

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 would be located at River Mile (RM) 184, corresponding to Project RM (PRM) 187.1 of the updated Geographic Information System (GIS) based hydrography.

AEA filed the Revised Study Plan with FERC on December 14, 2012, which included a River Productivity Study (AEA 2012, 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 Lower Susitna River. This implementation plan fulfills portions of the River Productivity Plan.

As described in the Revised Study Plan (RSP) Section 9.8.1 (AEA 2012), 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 (AEA 2012, 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 (AEA 2012), 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 quality assurance/quality control (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 Lower 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 Lower Susitna River.
3. Estimate drift of benthic macroinvertebrates in selected habitats within the Middle and Lower 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 Lower 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 Lower 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 plan is described in RSP Section 9.8.3 (AEA 2012). The River Productivity Study will entail field sampling throughout within the Middle River Segment and Lower River 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 Project (APA Project) (Friese 1975; Riis 1975, 1977; ADF&G 1983a; Hansen and Richards 1985; Van Nieuwenhuyse 1985; Trihey and Associates 1986). Alaska Department of Fish and Game (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* stonef1ies (P1ecoptera: 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 was 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 River Segment and above Gold Creek in the Middle River 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 stonef1ies (P1ecoptera: 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 Project focused on benthic macroinvertebrate communities in the sloughs, side channels, and tributaries of the Middle River 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. 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/m^3 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/m^2) comprised almost entirely of chironomid larvae and chlorophyll-*a* densities averaging 34.4 mg/m^2. Sampling in early May in Slough 8A revealed macroinvertebrate densities averaging 2,950 individuals/m^2, dominated by chironomids, with *chl-a* densities averaging 37mg/m^2. Results from five mainstem habitat sites showed similar macroinvertebrate numbers, with densities ranging from 393 to 8,820 individuals/m^2 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/m^2), with large increases occurring in October 1985 (1,729 – 7,109 individuals/m^2). Average densities in Slough 8A in August 1985 remained similar to spring levels (2,851 individuals/m^2), with a surge in September 1985 (13,964 individuals/m^2); 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 Lower 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 River 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 River 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 River 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 River Segment and in areas proximal to spawning tributaries (ADF&G 1981, ADF&G 1982). 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 River Segment were Indian River, Gash Creek, Chase Creek, and Whiskers Creek (Jennings 1985, Thompson et al. 1986). Spawn timing in Lower River Segment tributaries was slightly earlier relative to Middle River 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 are 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 River Segment (Jennings 1985, Roth et al. 1986). Within the Lower River 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 was 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 River 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 River 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 1982, 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 et al. 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 1982, 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 River Segment of the Susitna River, although high tributary densities were observed in early July and August. Juvenile coho salmon catch per unit effort (CPUE) estimates were frequently highest at sites located in the lower portion of the Middle River 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; 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 River 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 1984; 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 et al. 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 et al. 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 et al. 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 et al. 1984, Barrett et al. 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 River 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 River 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 River 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 Pechek 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 River Segment of the Susitna River than in the Lower River 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 River 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 River Segment, rainbow trout were found to utilize main channel areas, but in the Lower River 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

AEA presented the approach to River Productivity site selection at the February 15 Fish and Aquatics Technical Workgroup meeting. AEA reviewed the placement of six stations on the Susitna River, with two stations above the proposed reservoir pool in the Upper River Segment and four stations downstream of the proposed dam site in the Middle River Segment, and one station on the Talkeetna River. Middle River Segment station locations were selected at Focus

Areas proposed by the Instream Flow Study that feature a diversity of main channel and off-channel habitats with documented fish use. Agency representatives expressed concerns about limited number of stations within the Middle River Segment below Devils Canyon. Currently, stations are located at PRM 141 (near Indian River) and PRM 104 (Whiskers Slough). AEA has considered this comment, and maintains that the current design is appropriate for evaluating and monitoring the benthic community along the longitudinal gradient of the river continuum, both from the glacial source (Milner and Petts 1994; Brittain and Milner 2001; Milner et al. 2001), as well as from the downstream effects of a future dam (Ward and Stanford 1983, Stanford and Ward 2001). However, AEA has added one additional station that will be placed in the Lower River Segment. This station will expand the documentation of communities downstream of the Project and specifically will allow AEA to evaluate any influence the Chulitna and Talkeetna rivers may have on the mainstem Susitna River benthics and algal communities downstream of the Three Rivers Confluence.

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 slough, and upland slough). Sampling will occur at five stations on the Susitna River, and one station on the Talkeetna River, each station with three to five sites (establishing sites at all macrohabitat types present within the station), for a total of 24 sites. 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 1.2-1). All stations established within the Middle River Segment will be located at Focus Areas established by the Instream Flow Study (AEA 2012, 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., AEA 2012, 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 (AEA 2012, Section 9.8.4.5.1). The Lower Susitna River Segment is defined as the approximate 98-mile section of river between the Chulitna and Talkeetna rivers confluence and Cook Inlet. One station will be located in the upper portion of this segment (Figure 1.2-2) to determine to what extent, if any, the Project operations would affect benthic communities, as well as the influence the two tributaries may have on the mainstem Susitna River below the confluence of the three rivers. Station and site locations are discussed below.

2.1.1. Middle River Segment Stations / Focus Areas

Within the Middle River 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 1.2-1 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 River 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, a side slough site, an upland slough site, and a small tributary mouth 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, a side channel site, and an upland slough 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 River 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 main channel site, , a side-channel site, a side slough site, an upland slough site, and a tributary mouth 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. Lower River Segment Station

Within the Lower River Segment of the Susitna River, the River Productivity Study will establish one study station, with five sampling sites located in conjunction with individual sites proposed by the Instream Flow Study and Fish Distribution and Abundance sampling activities on the Lower Susitna River around the Trapper Creek area (Table 1.2.1, Figures 1.2-2 and 2.1-6). This lower river station (RP-92) will be located within a 4.5-mile reach beginning approximately 5 miles downstream of the confluence with the Chulitna and Talkeetna rivers.

Imagery and maps indicate this area is complex, with split channels, side channels, side sloughs, and tributary mouths (Figure 2.1-6). Station RP-92 will provide a mainstem site, side channel site, side slough site, upland slough site, and a tributary mouth site, which will be confirmed with site reconnaissance.

2.1.3. Talkeetna River 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 River 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 options 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, in consultation with the TWG.

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 meters or a depth of 1 meter, whichever is reached first. 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 m^2-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 m^2 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-µm 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 LWD 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." However, for the purposes of this study, we define woody snags as LWD, adopting the definition used from RSP Section 9.13: "LWD must be at least 0.1 m (4 inches) in diameter, and at least 1.0 meter (39 inches) of the LWD must be below the water's surface at bankfull flow." The following is the procedure for sampling LWD.

1. Identify five (if present) LWD locations throughout the site area. Suitable LWD will have been submerged for an extended period of time so as to be clearly colonized. Refer to information provided by multiple remote cameras and staff gages installed along the Susitna River, along with the USGS gage at Gold Creek, when determining whether recent stage conditions would provide adequate inundation levels for sampling. Also, look for evidence of algal growth and invertebrate cases or tubes as evidence of colonization of the LWD.
2. At each LWD location, identify a LWD piece to remove from the water. Per FERC's SPD, pieces should be at least 10 centimeters in diameter, Sections of larger branches may need to be removed, using a hand saw. Ideally, each LWD section will originate from a separate snag, and therefore count as a separate, replicate sample.
3. Measure and record: the depth of the LWD 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 LWD piece from the water, using a saw or lopping shears. Each 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. LWD pieces should be placed, 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 250-micrometer 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-milliliter 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, 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 LWD sample. This information is written on the field data collection worksheet for the site.

In addition to sampling these smaller, removable LWD pieces, crews will collect up to five replicates, if present, from large, immobile LWD at each site. Protocols will be similar to those for removable pieces, with the exception that sections of the LWD will be sampled *in situ* by positioning the collection net immediately downstream of the LWD section, and brushing its surface by hand or with a small hand brush, thus collecting dislodged organisms in the net as they enter the water column.

2.2.1.3. Grab Samples

Collection of macroinvertebrate samples in macrohabitats with fine substrate and low velocities (e.g., side sloughs and upland sloughs) is also necessary to evaluate food availability for resident fish species that feed in these habitats. Because it is not feasible to use the Hess sampler in habitats with deep water (> 40 cm), fine substrate or low velocities, benthic grab sampling will be employed to collect macroinvertebrates in those macrohabitats. Sampling will be conducted using a petite Ponar grab sampler. The following is the procedure for grab sampling.

1. .Identify locations throughout the reach where five discrete collections of fine sediment can be taken. Sampling may require a raft or small kayak in order to access substrates in deeper waters or in areas where it would be difficult to sample without disturbing the substrates.
2. Carefully lower the grab sampler into the water, towards the streambed, and then drop it to the bottom. The jaw release mechanism will disengage when the sampler hits the streambed.

3. Recover the grab sampler and inspect to ensure that the discrete collection was not lost because of debris catching in the jaws of the sampler. Place the grab sampler into a large plastic container (e.g., 5-gal. bucket, 10-gal. tub or bin) to avoid sample loss if the grab sampler should open.
4. Measure water depth and velocity for each discrete sample location. This information is written on the field data collection worksheet for the site.
5. Empty the grab sampler contents into the plastic container. Rinse all sediment remaining inside the sampler into the container. Record a visual estimate of substrate composition using the Wentworth scale, to the nearest 5 percent.
6. Pour all sampler contents through a sieve bucket with a 250-µm mesh screened bottom, and wash all fine sediments from the sample by quickly plunging the sieve bucket into the water, and then rapidly rotating the bucket back and forth. It may be necessary to rub the mesh screen in the bottom of the bucket to agitate and drain the water and finer sediments. Repeat this rinsing until the finer sediments are removed from the sample material.
7. Rinse the sample from the sieve bucket into a larger 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. Multiple containers may be necessary for samples with larger amounts of organic matter.
8. 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 container. If the sample requires more than one sample container, be sure to designate the container number out of the total number of containers needed for the sample (e.g., "1 of 2").
9. 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.
10. Rinse the sieve, sieve bucket, and grab sampler, and reset jaw release mechanism.
11. Repeat the collection procedure until all five discrete collections have been taken.

2.2.2. Sample Processing Protocols

Benthic macroinvertebrate samples from Hess, LWD, and Petite Ponar 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) may 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 would be used to describe the significant differences in the data. Assumptions of normality and equal variance would 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 would 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. Rock substrates should be evenly collected at multiple depths in one-foot depth categories (e.g., 0 – 1 foot, 1 – 2 feet, and 2 – 3 feet) to the extent feasible, given the limits of field safety. 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 sampling 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 AFDM) 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 AFDM), 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) may 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 allocthonous 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.

As the majority of benthic macroinvertebrates are closely associated with the substrate during at least part of their life cycle, it is logical that substrate characteristics and types should be a major determinant of the macroinvertebrate community's distribution and abundance (Ward 1992; Minshall 1984). The substrate provides habitat space, food, and protection (as flow refugia, mentioned above). Substrate characteristics that are of ecological importance to macroinvertebrates include particle size, organic content, stability, and heterogeneity (Ward 1992). Coarser bed materials generally provide more interstitial spaces for macroinvertebrates to use as refugia, as well as for the trapping of detritus for food (Hershey and Lamberti 2001; Rabeni and Minshall 1977). As a result, diversity and abundance generally increases with substrate stability and the presence of detritus (Minshall 1984). Therefore, to address the importance of organic matter to benthic productivity in this type of system, this study will quantify benthic organic matter as it is directly related to the benthic macroinvertebrates being collected – within the coarse substrates they reside in.

In addition, Project operations could affect turbidity downstream of the dam, with decreased turbidity potentially resulting in an increase in primary productivity in the Middle River Segment, and increased autocthonous imputs of organic matter. The Water Quality Modeling Study (RSP Section 5.6) will model water quality conditions in the Susitna River from the proposed site of the Susitna-Watana Dam downstream, including (but not necessarily limited to) temperature, suspended sediment, and turbidity. The reservoir model also being developed will be directly input into the downstream river model. This will enable downstream evaluation of potential impacts of the proposed Project on hydrodynamic, temperature, and water quality conditions. Calibration of the model(s) utilizing data collected in 2013 will be necessary, and preliminary results from the model will be available in early 2014. Model results will be reviewed for possible effects on organic matter in Susitna River downstream of the dam, and revisions to organic matter sampling, if necessary, will be made for the 2014 sampling season to address any additional issues revealed by the modeling results.

2.4.1. Field Sampling Protocols

In order to quantify the amounts of organic matter available in the Susitna River for benthic macroinvertebrate production, CPOM and FPOM will be collected directly from all benthic macroinvertebrate sampling, in Hess and Petite Ponar 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, and sieves used to rinse and retain sample contents from Hess and grab samplers, 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 and grab 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-µm nested sieves, to separate CPOM and FPOM components of the detritus, oven-dried (60°C), and weighed. Dried components will then be combusted in a furnace, and reweighed for ash free dry mass (AFDM) weights. Results will be expanded according to the subsample factor, and calculated as AFDM estimates of CPOM and FPOM per unit area (g/m^2).

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 detritus has been sorted and benthic invertebrates removed, the sample material should be rinsed through 1 mm and 250 µm nested sieves to separate CPOM and FPOM components of the detritus. Components will then be processed for AFDM.. Results will be calculated as AFDM estimates of CPOM and FPOM per unit area (g/m^3).

2.4.3. Data Analysis Methods

Results generated from the collections would include the AFDM 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) may 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 would 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 all established sites. In addition, at all tributary mouth sites, a drift net pair will also be deployed upstream of the site, to collect information on the relative contribution of tributaries to fish food resources in the mainstem Susitna River. 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-µm 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.

At sites with currents less than 0.05 meters per second (m/s), a plankton tow net will be used, taking a horizontal tow along a transect across the channel. Two calibrated tow lines should be attached to the tow net, with one going to a crew member on each side of the channel, allowing to collect tows along shore-to-shore transects across the channel without repetitive crossing, or attempts to toss the net to the other side of the channel. The following is the procedure for sampling horizontal plankton tows in still water areas.

1. Prior to each use, carefully clean and thoroughly rinse the interior of the plankton net and mesh cup with clean distilled water. Collections will be made using a 243-µm mesh plankton net with a 200 mm opening.
2. Carefully inspect the net and mesh cup for holes or tears. Make sure the clamp is on the tubing and closed securely.
3. Attach the metal ring of the plankton net to two calibrated ropes with meter markings.
4. Walk at least 3 m upstream from where macroinvertebrate collection took place (or any other activity disturbing bottom sediments).
5. Lower the 243-µm mesh plankton net into the open water out from the shoreline, in water approximately 30 cm deep. Care should be taken to place the net into water that has not been clouded up with the sediment you disturbed when walking. The crew member releasing the net should remain at this spot holding the 2nd calibrated line so a transect distance measurement can be made upon retrieval.
6. The crew member on the opposite shore should start pulling the net across the channel at a slow but steady speed, keeping the top edge of the net slightly submerged (i.e. let the net settle into the water but do not let it sink). Pull the net back at an upward angle so that the opening does not dip downwards towards the bottom. The goal is to sample the water column, not the bottom or surface.

7. The crew member retrieving the net should pick it up out of the water in shoreline water depths of approximately 30 cm, to prevent sampling the bottom or surface. The net should be pulled out of the water immediately to prevent backwash and loss of sample. If vegetation is present in the net when you retrieve it, pull it out gently and discard.
8. Note and record the transect distance of the tow on the field data sheet.
9. Empty the contents of the plankton net, and rinse the inside of the net into a 250-µm mesh sieve.
10. 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. Wash the remaining sample into the container with a wash bottle containing 95 percent ethanol.
11. 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.
12. 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.
13. Repeat steps 1-11 for the next plankton net tow along a new transect, for a total of five replicate plankton tows. Do not combine the contents of separate tow net samples.

2.5.2. Processing Protocols

Invertebrate drift and plankton tow 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 and plankton 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 drift 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

Adult aquatic insect emergence mass is a product of aquatic insect production from the stream, and is therefore a good surrogate for actual production (minus predation), and will be especially useful for relative comparisons between river sections and years (personal communication, M. Wipfli, University of Alaska-Fairbanks). To measure insect emergence, floating emergence trap samplers will be deployed, with one trap per site.

2.6.1. Field Sampling Protocols

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 (Cadmus and Pomeranz unpublished). 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, from the initial deployment following ice breakup until the last seasonal sampling event (September-October). 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.

In 2014, emergence traps will be first deployed in early April in off-channel ice-free areas, if available, at established study sites in the Middle and Lower river Segments. Traps will be sampled every 2 weeks, to be removed at the beginning of ice breakup. Traps will then be redeployed at all sites following ice breakup in late May or early June.

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/m^2/time period), and biomass (dry weight/m^2/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 Lower 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 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 Lower 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 Lower River Segment and Middle River Segment (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

River 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 River 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 m^2 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-µm 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), 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, and will be used to reassess benthic sampling protocols (Sections 2.2 and 2.3) which currently allow for at least 30 days of inundation for colonization.

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 Lower and Middle Rivers (RSP Section 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. Flow velocity and velocity-dependent capture probabilities will be incorporated into the GRP models for juvenile coho salmon and juvenile rainbow trout. 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 Lower 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. The two Focus Areas (stations), specific sample site locations, and number of adult salmon tissue samples to be collected will be determined through consultation with the TWG. 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 $\delta^{13}C$ or $\delta^{15}N = [(R \text{ sample } / R \text{ standard}) - 1]1000$, where R is $^{13}C/^{12}C$ or $^{15}N/^{14}N$ (Peterson and Fry 1987). Spawning salmon are expected to exhibit an enriched signature of $\delta^{15}N$ relative to freshwater or terrestrial energy sources (Bilby et al. 1996; Chaloner et al. 2002;

Satterfield and Finney 2002), and the combination of the $\delta^{15}N$ and $\delta^{13}C$ 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 $\delta^{13}C$ and $\delta^{15}N$ 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 $\delta^{13}C$ vs. $\delta^{15}N$ 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 $\delta^{13}C$ vs. $\delta^{15}N$ 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 $\delta^{13}C$ and $\delta^{15}N$ 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

A comprehensive list of sampling gear, water quality meters, field supplies, chemicals, and personal gear used to collect, and in some cases field process, samples for the River Productivity Study is provided in Table 4.1-1.

5. REFERENCES

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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
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 (4 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 (4 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 (5 sites), Storm Event Site	Whiskers Slough Complex	MR-8	106.0	104.8	1.2	X	X	X	X	X	X		X	X
RP-92	Trapper Creek Area Complex	Study Station (5 sites)	Area approximately 5 miles downstream of confluence	LR-1	97	92.5	4.5	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	21	3	8	504
Coho salmon - juveniles	21	3	8	504
Rainbow trout - juveniles	21	3	8	504
Rainbow trout - adults	21	3	8	504
			Total	2,016

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 River 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

Table 4.1-1. Suggested equipment list for River Productivity Study.

Sampling Gear

Macroinvertebrate / Organic Matter

- ❑ Modified Hess Sampler (250-µm mesh)
- ❑ D-frame kick net (250-µm mesh)
- ❑ Drift nets (pair) (250-µm mesh)
- ❑ Floating emergence traps
- ❑ Hester-Dendy multiplate samplers

Algae

- ❑ PVC pipe area delimiter, with brushes

Water Quality Meters

- ❑ Current velocity meter
- ❑ Mechanical flowmeter
- ❑ Portable turbidity meter
- ❑ Light meter with quantum sensor
- ❑ Thermometer
- ❑ Temperature Probes

Chemicals

- ❑ 95-percent ethanol
- ❑ 37-percent buffered formaldehyde
- ❑ 10-percent buffered formalin
- ❑ Fish aneshthetic (clove oil)
- ❑ Dry ice

Personal Gear

- ❑ Hip boots (per person)
- ❑ Chest waders (per person)
- ❑ Wader repair kit
- ❑ Rain gear
- ❑ Neoprene gloves
- ❑ Arm-length rubber gloves (per person)
- ❑ Personal flotation devices (per person)
- ❑ First aid kit
- ❑ Insect repellant
- ❑ Sun screen
- ❑ Polarized sunglasses
- ❑ Cellular phone
- ❑ Flashlight and lantern

Basic Field Supplies

- ❑ Wire mesh sieves (#60, 250 µm)
- ❑ Forceps, blunt and fine tipped
- ❑ Spatula, scoop or spoon
- ❑ Plastic dishpan
- ❑ Knives (pocket and putty), scalpels, scissors
- ❑ Oxford™ Macro-Set hand pipettor (1 mL–5 mL w/tips)
- ❑ Heavy-duty aluminum foil
- ❑ Hand-held electric stirrer (periphyton homogenizer)
- ❑ Batteries (various sizes including 12 volt)
- ❑ Whatman™ GF/F glass fiber filters (47-mm diameter)
- ❑ Hand-operated vacuum pump with pressure gauge
- ❑ Plastic Erlenmeyer flask (1 L)
- ❑ Filter funnel and base (for 25 mm and 47 mm filters)
- ❑ Hand saw or lopping shears
- ❑ Hand brush
- ❑ Syringes (10 mL)
- ❑ Surgical gloves
- ❑ Hand rake
- ❑ Squirt bottles
- ❑ Wide-mouth sample bottles (500 mL)
- ❑ Wide-mouth sample jars (500 mL, 1 L)
- ❑ Whirl paks (50 pk)
- ❑ Resealable plastic bags
- ❑ Plastic scintillation vials
- ❑ Alcohol/waterproof pens, black
- ❑ Pencils (lead, red wax)
- ❑ Measuring board
- ❑ Small paper/wax envelopes
- ❑ Ruler/meter stick
- ❑ Measuring Tapes
- ❑ Large plastic bins (10 gal) or buckets (5 gal)
- ❑ steel rods or rebar stakes (1 m in length)
- ❑ Large plastic insulated coolers
- ❑ Graduated cylinders (glass 10 mL, plastic 50–500 mL)
- ❑ Portable weighing balance
- ❑ Jerricans (3.5 gal)
- ❑ Digital camera (high-resolution)
- ❑ Handheld geographic positioning system unit

Forms

- ❑ Safety plan (with emergency phone numbers)
- ❑ Collecting permits
- ❑ Waterproof paper
- ❑ Field data sheets printed on waterproof paper
- ❑ Adhesive labels (blank and preprinted standard)
- ❑ Standard internal labels (preprinted)

7. FIGURES

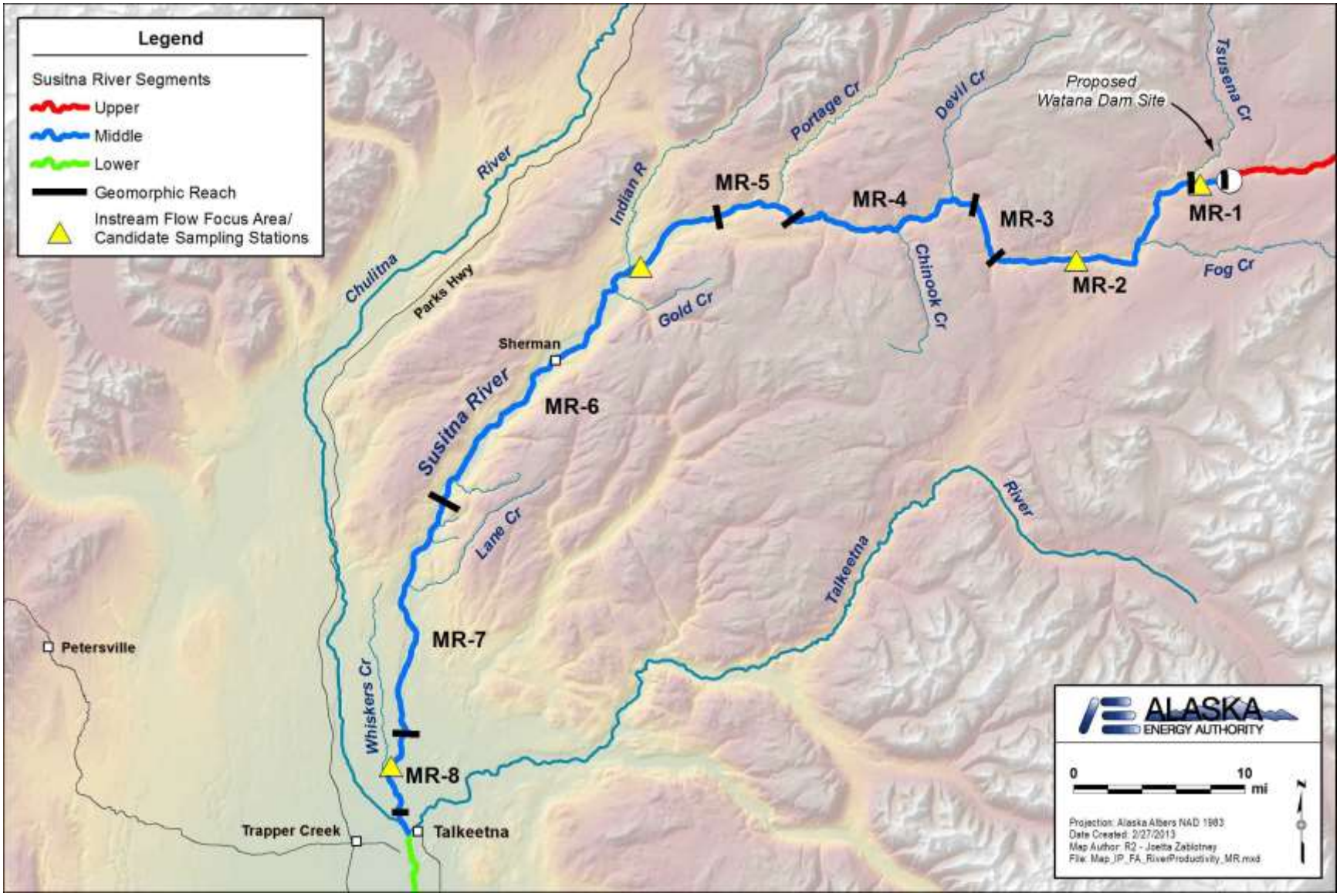


Figure 1.2-1. Middle Susitna River Segment, with the four proposed River Productivity sampling stations /Instream Flow Focus Areas selected for the River Productivity Study.

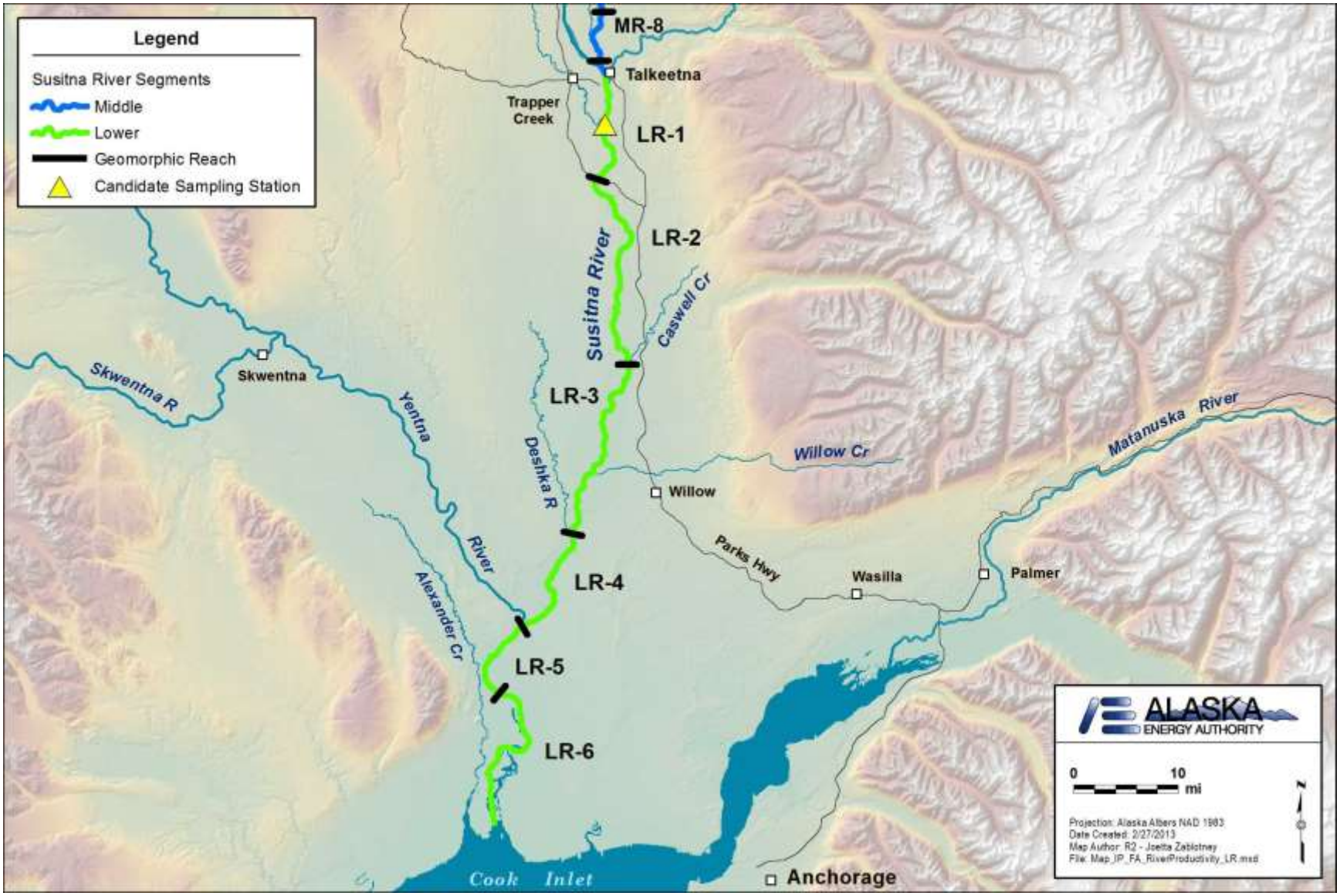


Figure 1.2-2. Lower Susitna River Segment, with one proposed River Productivity sampling station /Instream Flow study sites selected for the River Productivity Study.

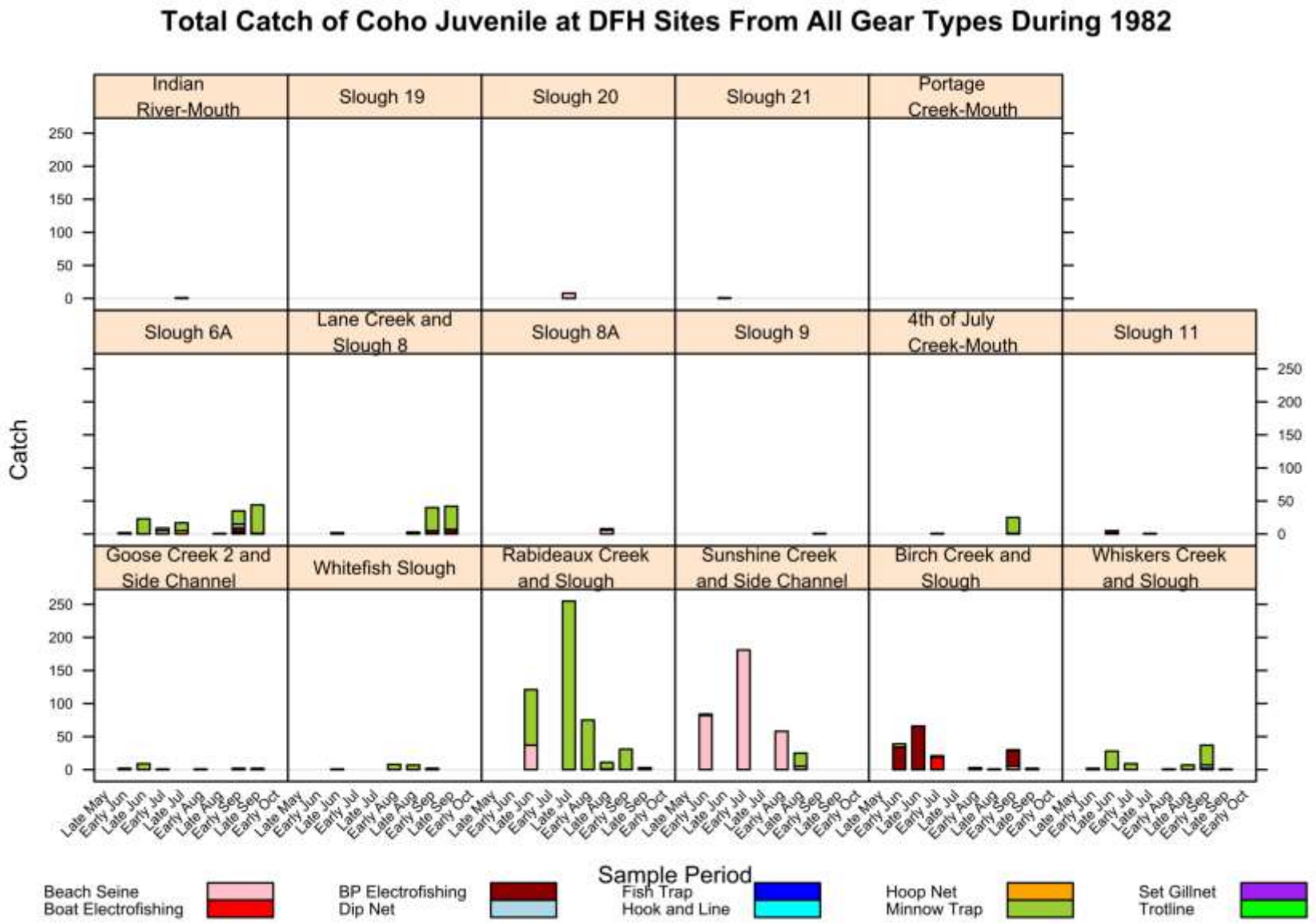


Figure 1.3-1. Total catch of juvenile coho salmon by sample period and gear type at DFH sites in 1982. Source: Estes and Schmidt 1983

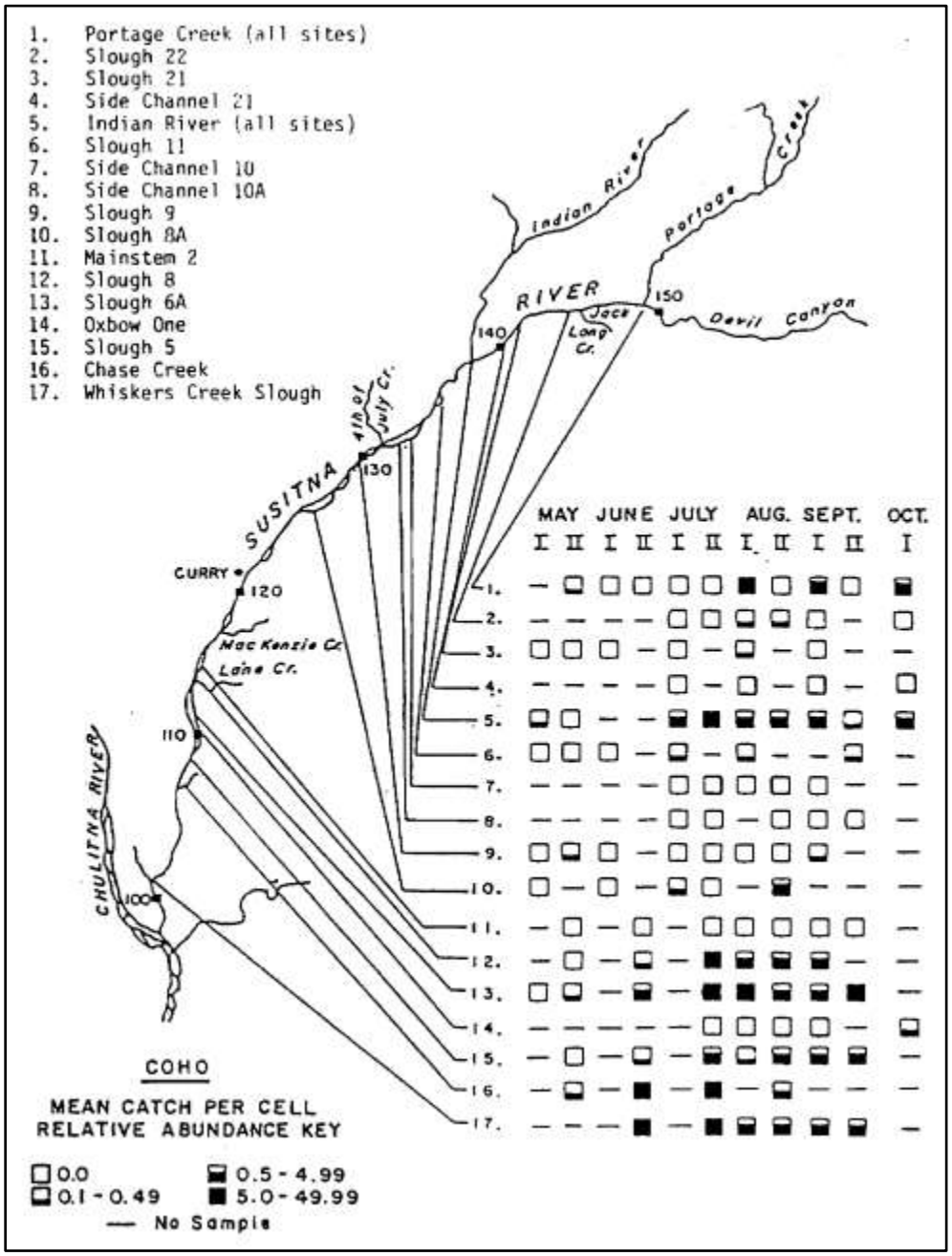


Figure 1.3-2. Seasonal distribution and relative abundance of juvenile coho salmon on the Susitna River between the Chulitna River confluence and Devil Canyon, May through November 1983. Source: Dugan et al. (1984).

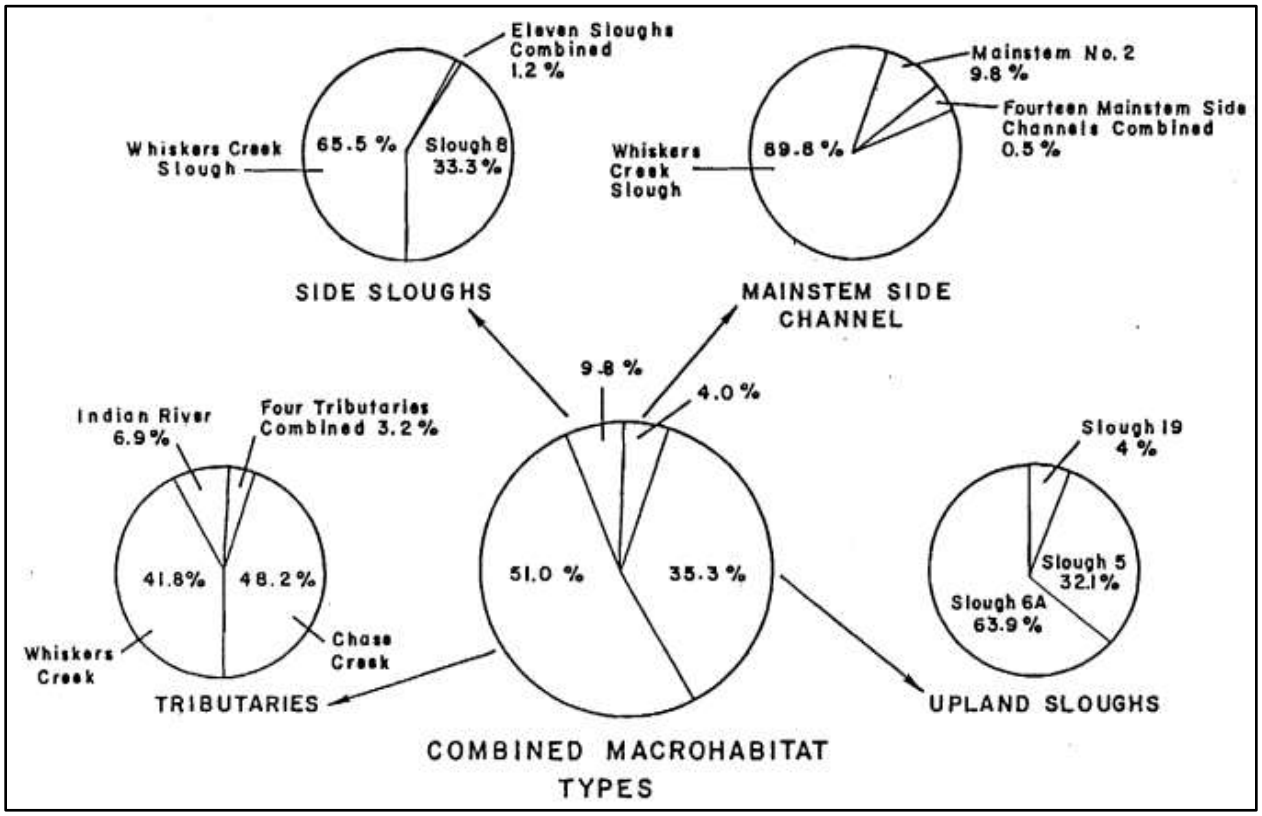


Figure 1.3.3. Density distribution and juvenile coho salmon by macrohabitat type on the Susitna River between the Chulitna River confluence and Devil Canyon, May through November 1983. Percentages are based on mean catch per cell. Source: Dugan et al. (1984).

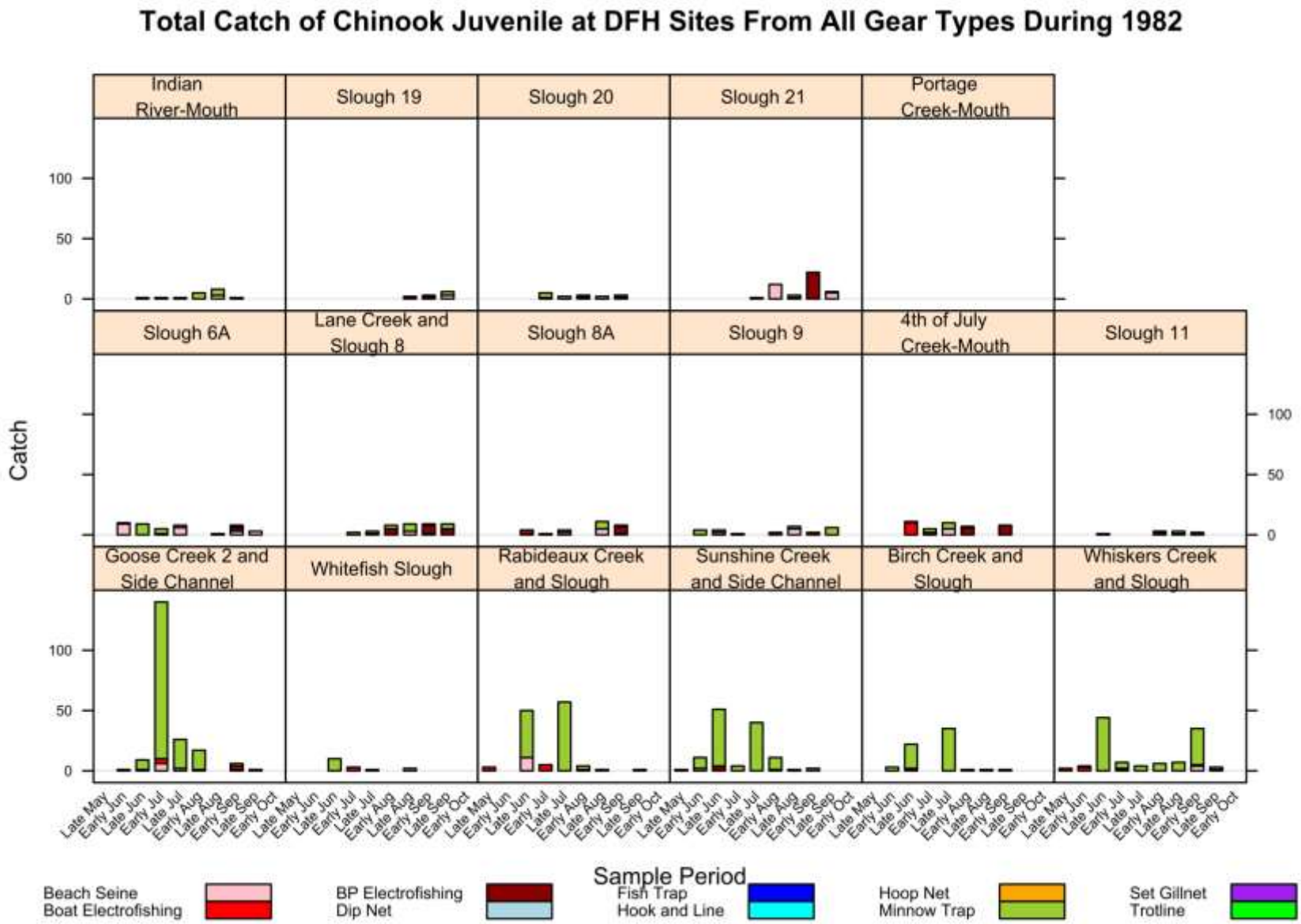


Figure 1.3-4. Total catch of juvenile Chinook salmon by sample period and gear type at DFH sites in 1982. Source: Estes and Schmidt 1983

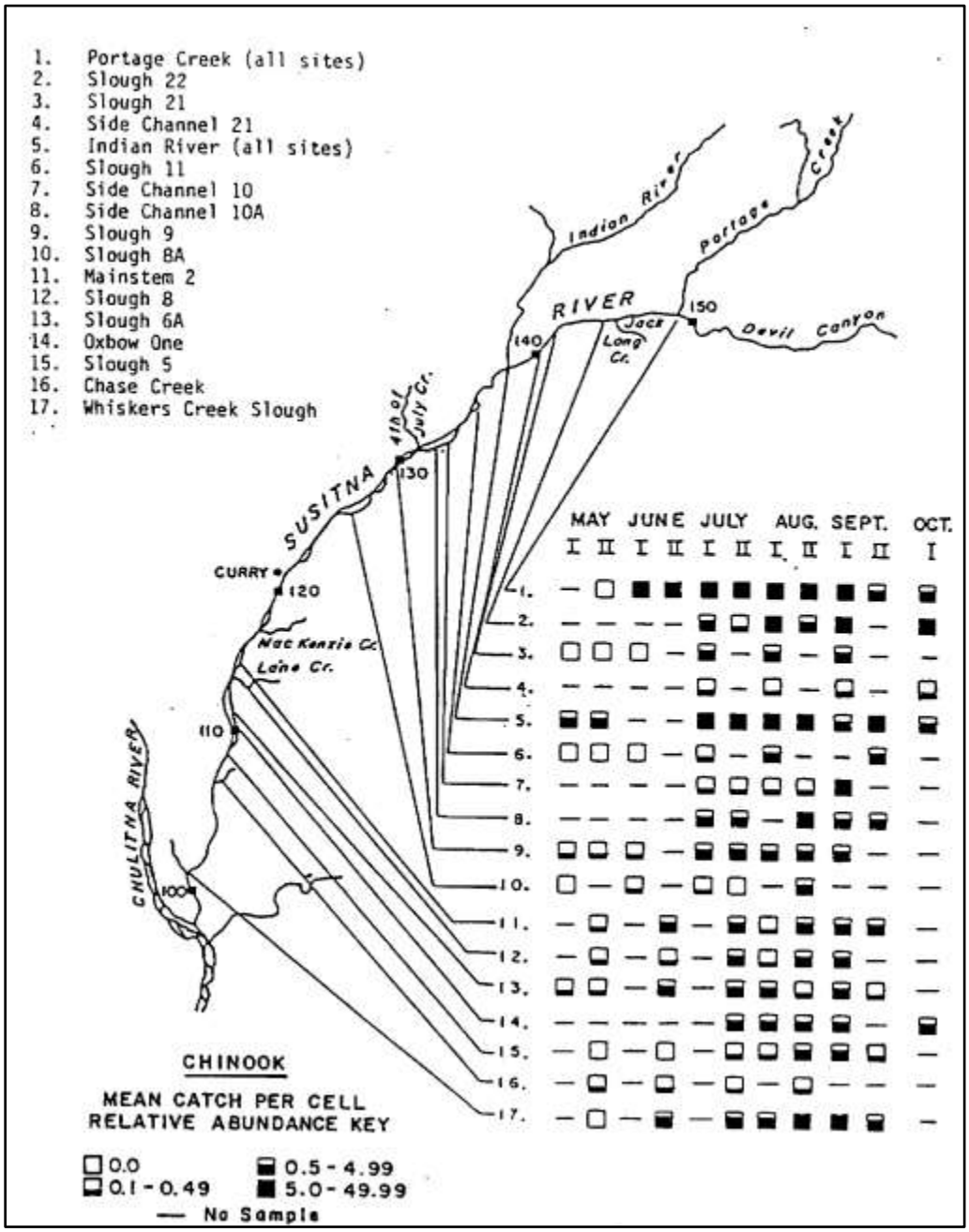


Figure 1.3-5. Seasonal distribution and relative abundance of juvenile Chinook salmon on the Susitna River between the Chulitna River confluence and Devil Canyon, May through November 1983. Source: Dugan et al. (1984).

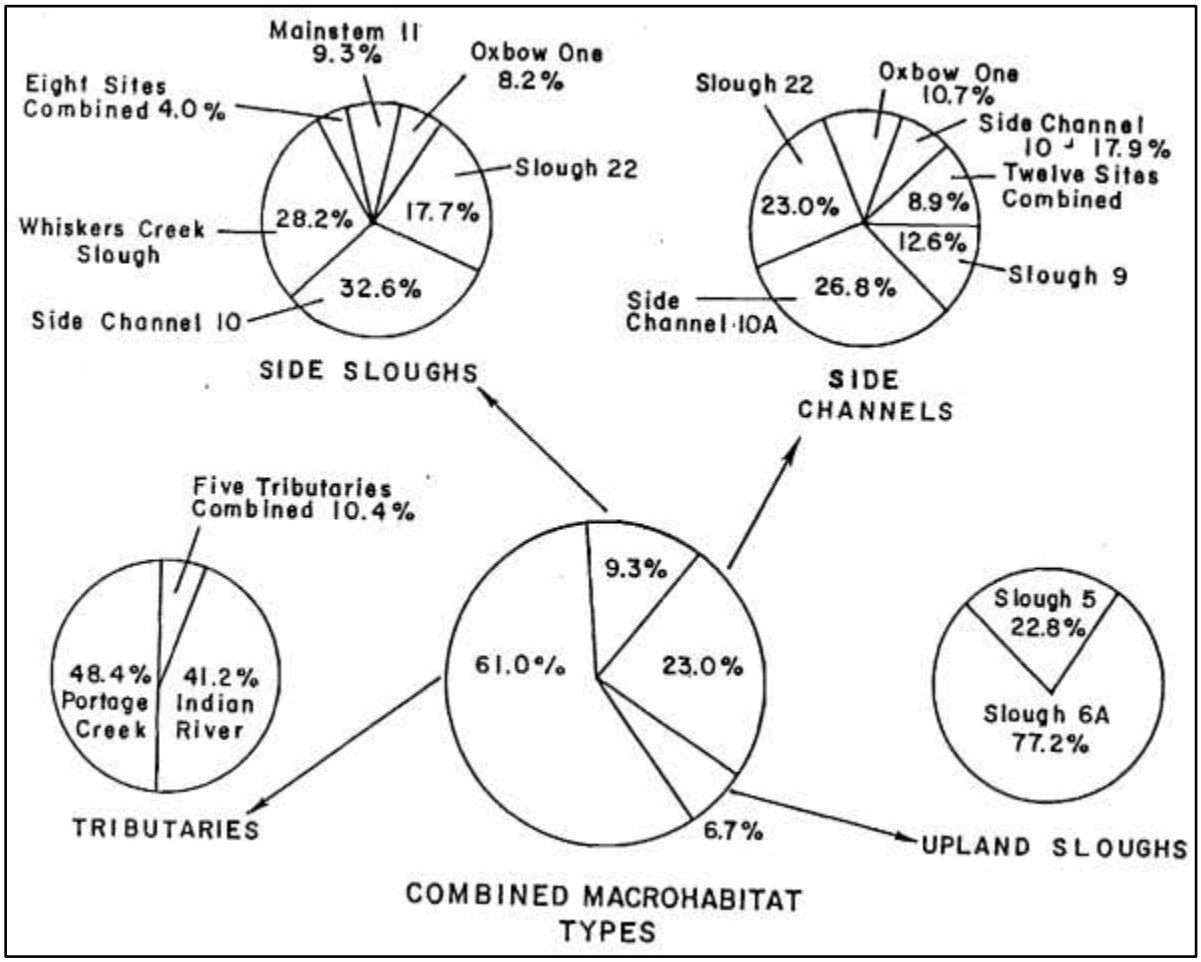


Figure 1.3-6. Density distribution and juvenile Chinook salmon by macrohabitat type on the Susitna River between the Chulitna River confluence and Devil Canyon, May through November 1983. Percentages are based on mean catch per cell. Source: Dugan et al. (1984).

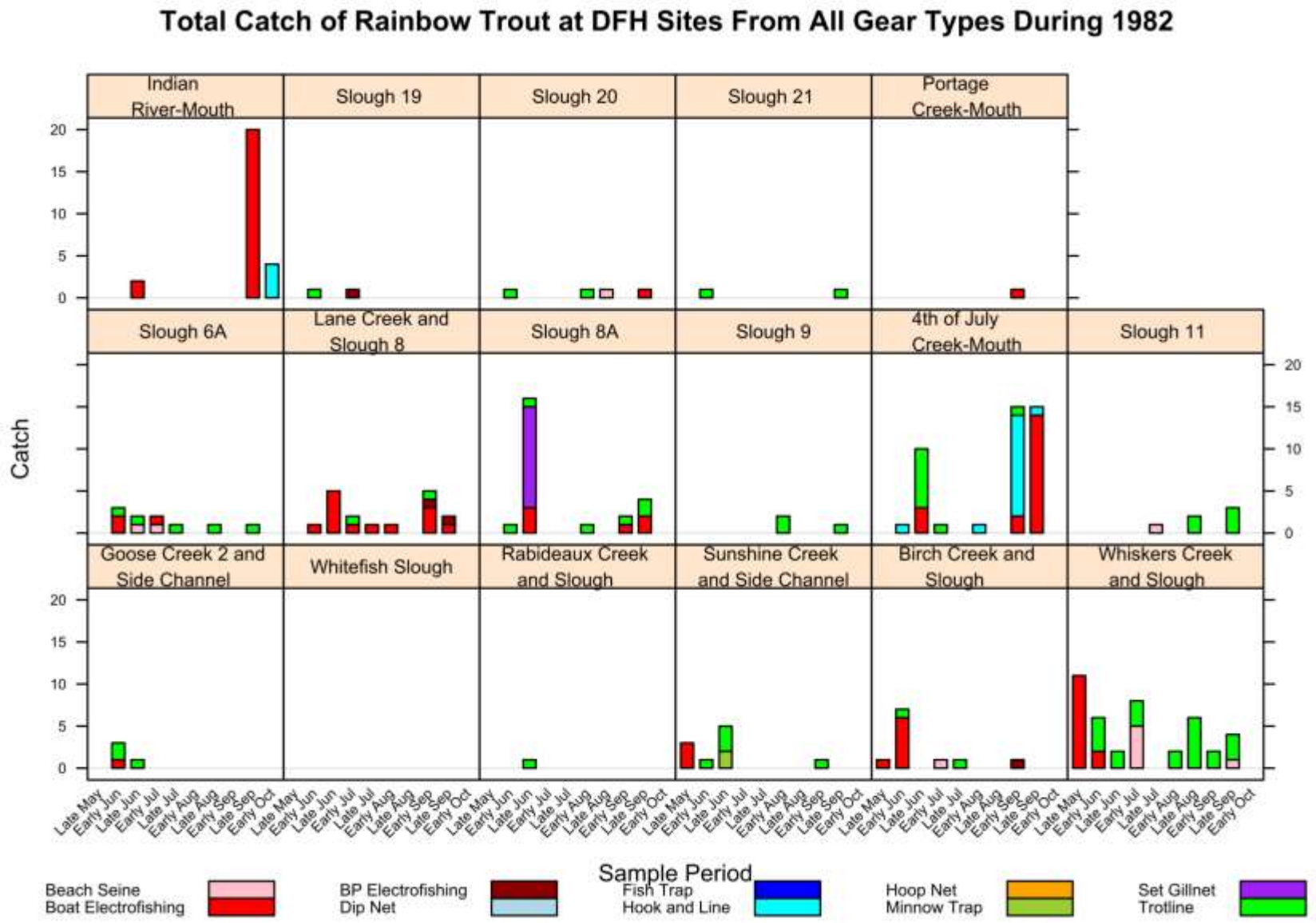


Figure 1.3-7. Total catch of rainbow trout by sample period and gear type at DFH sites in 1982. Source: Estes and Schmidt 1983

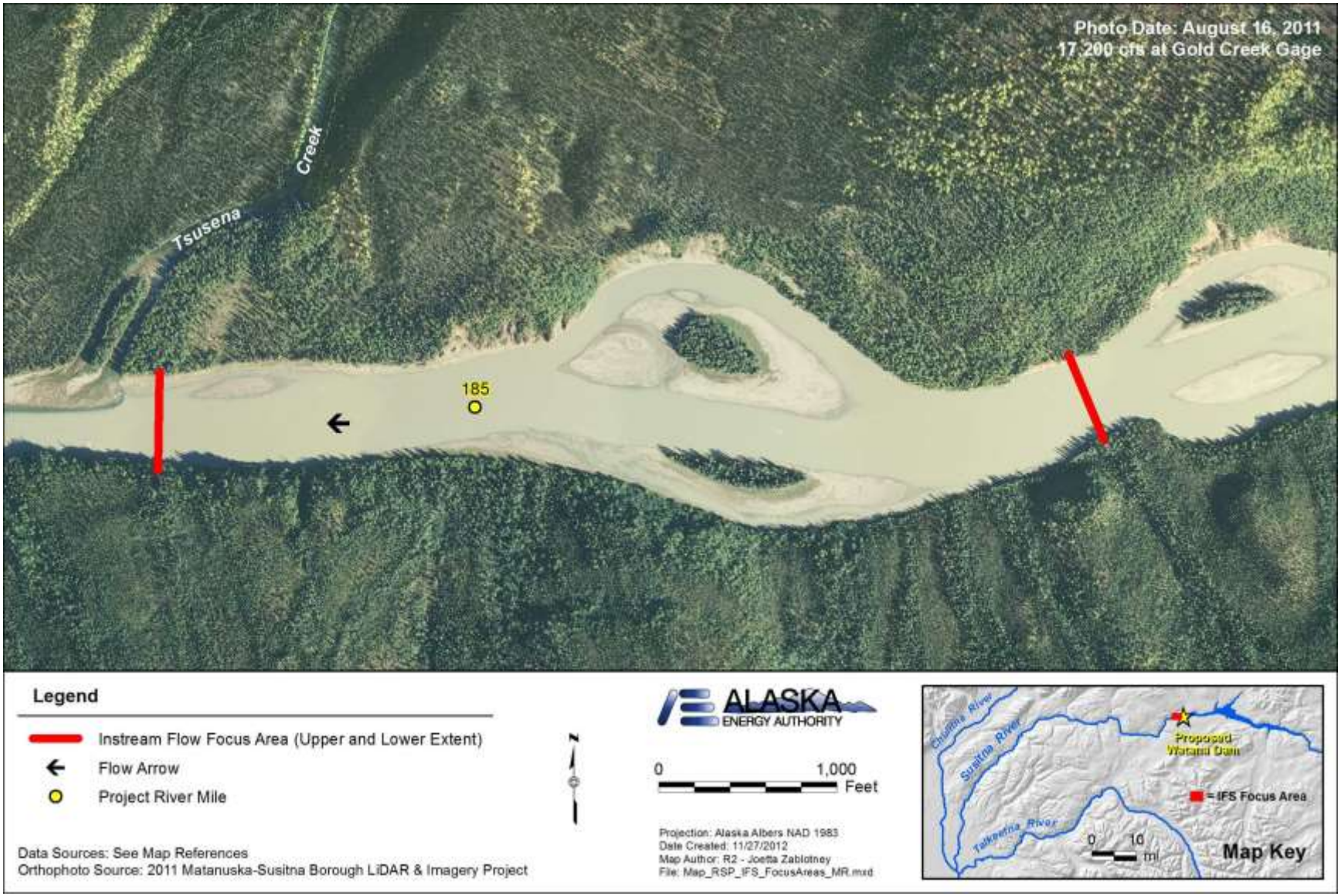


Figure 2.1-1. Map showing Focus Area 184 that begins at Project River Mile 184.7 and extending upstream to PRM 185.7. The Focus Area is located about 1.4 miles downstream of the proposed Watana Dam site near Tsusena Creek.

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.

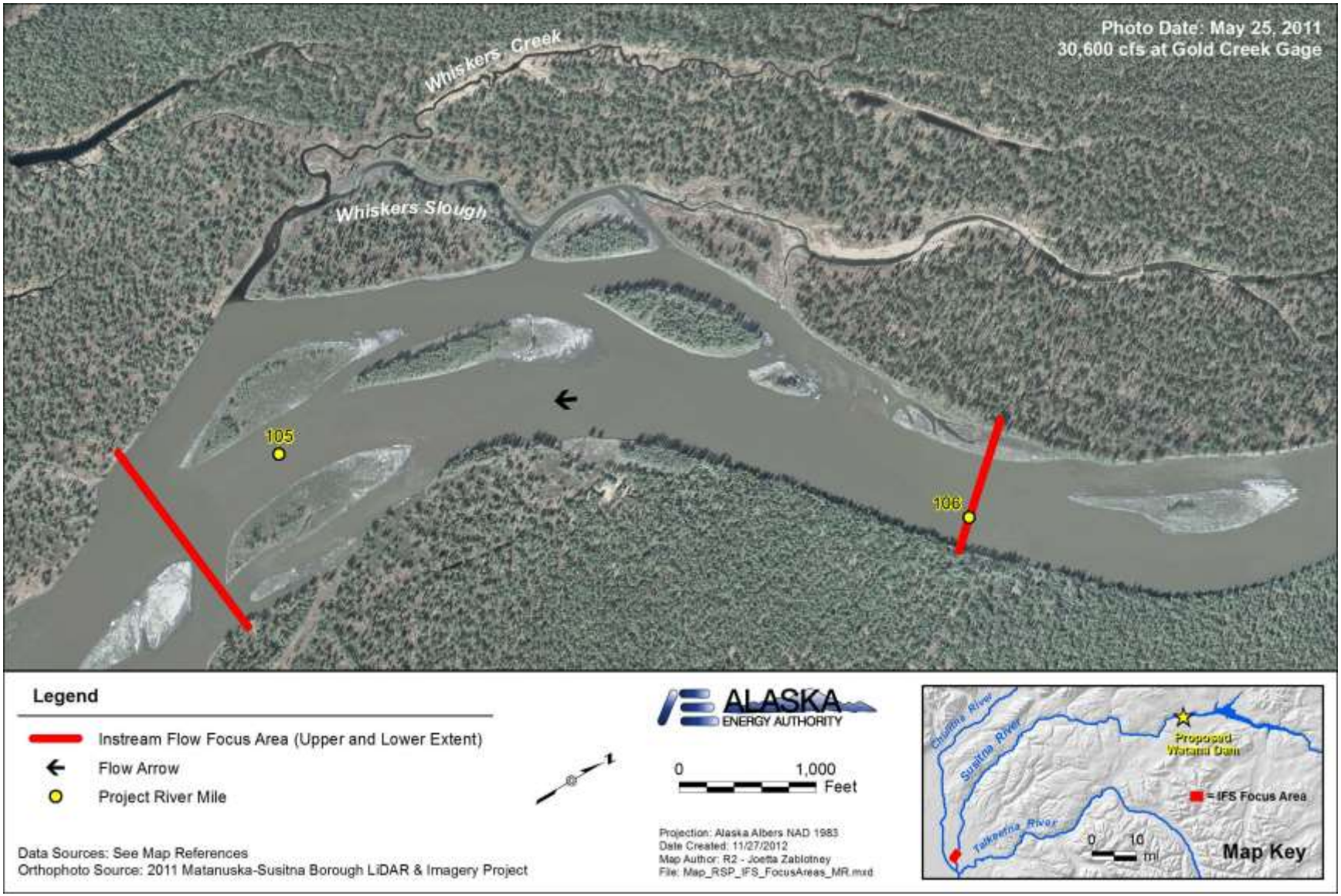


Figure 2.1-4. Map showing Focus Area 104 beginning at Project River Mile 104.8 and extending upstream to PRM 106. This Focus Area covers the diverse range of habitats in the Whiskers Slough complex.

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.

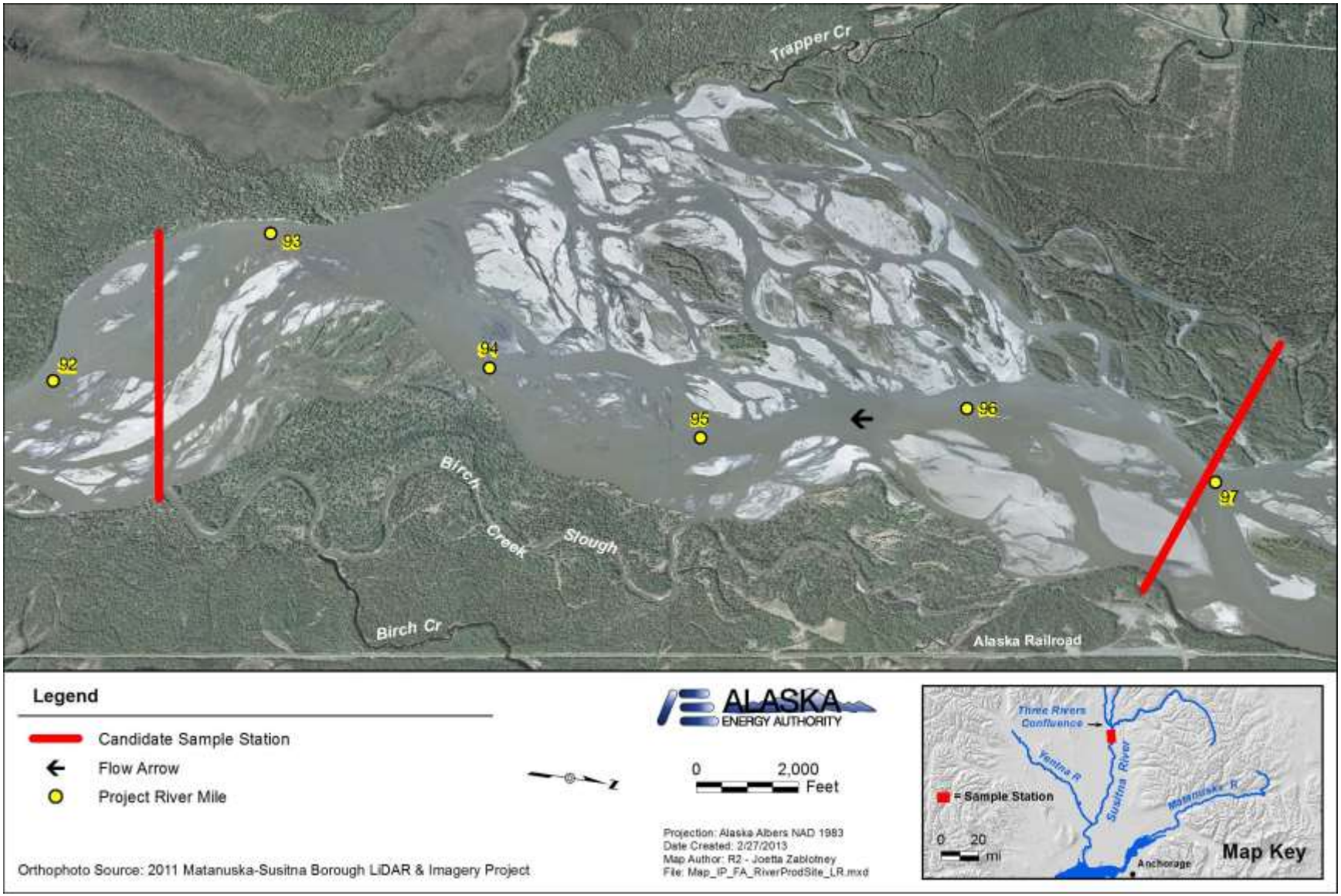


Figure 2.1-6. Map showing the River Productivity Lower River Segment sampling station RP-92, located downstream of the confluence with the Chulitna and Talkeetna rivers beginning approximately at Project River Mile 92.5 and extending upstream to approximately PRM 97.

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

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

Rapid Bioassessment Protocols: Technical Level
Method 002 – Laboratory Processing

Macroinvertebrate samples are subsampled, sorted, and identified in the laboratory under controlled conditions. All samples are recorded upon receipt by the laboratory. Information from the sample container label should be included on the login sheet. The number of containers should be indicated and equal those indicated on the label. All samples should be sorted in a single laboratory for quality control. Dates and types of sample processing should be recorded in the sample login sheet for each sample.

Laboratory Equipment and Supplies

- Two standardized 350 µ gridded subsampler trays (5.5 cm x 5.5 cm grids)
- 350 µ sieve
- Forceps
- White plastic or enamel sorting trays
- Specimen vials with rubber-lined caps or stoppers
- Sample labels
- Dissecting microscope
- Fiber optic light source
- Ethanol for storage of specimens
- Taxonomic keys
- Taxonomy validation notebook
- Compound microscope
- Microscope slides and cover slips
- Head mount medium (CMC-10)

Record Keeping *(See pages 6–9 of Method 002.)*

- Sample Login Sheet
- Laboratory Bench Sheet-Subsampling
- Laboratory Bench Sheet-Identification
- Laboratory Bench Sheet-Chironomidae Identification

Subsampling/Sorting Procedures

The protocol is used for a 300-organism subsample. The entire sample is processed and a 300-organism subsample (+/- 20%) is randomly selected, sorted, and preserved separate from the remaining sample.

SS-1. Note the total number of jars recorded on the login sheet and retrieve the jars. Pour the contents of all jars into the 350 µ mesh sieve or tray and thoroughly rinse the entire sample to remove preservative and fine sediment. Large organic material (whole leaves, twigs, algal or macrophyte mats, etc.) not removed in the field should be rinsed, visually inspected, and discarded. If the samples have been preserved in ethanol, soak the sample contents in water for about 15 minutes to hydrate the organisms. This will prevent them from floating on the water surface during sorting. Gently mix the sample by hand while rinsing to homogenize.

SS-2. Spread the sample evenly across a subsampling pan marked with 5.5 cm grids after washing. Put some water in the tray to distribute the contents evenly and then slowly pull the inside tray up to drain off the water to begin subsampling.

SS-3. Use random numbers representing the grids and select four numbers corresponding to squares within the gridded pan. Note the grids selected on the Laboratory Bench Sheet-Subsampling on the level 1 section (see page 7). Using a spatula, remove all material (organisms and debris) from the four grid squares and place the material into a shallow white sorting pan. Add water to facilitate sorting. If there appears to be 300 ± 20% organisms (cumulative of four grids) from a visual inspection, then subsampling is complete and sorting can begin. Consider any organism that is lying over a line separating two grids to be on the grid containing its head. In those instances where it is not possible to determine the location of the head (worms for instance), consider the organism to be in the grid containing most of its body.

If the density of organisms is high enough that many more than 300 organisms are contained in the first four grids, transfer the contents of the first four grids to a second gridded subsampling pan and go to the second level of subsampling. Randomly select grids for the second level of sorting as was done for the first, ***sorting grids one at a time*** until 300 ± 20% organisms are found. ***Mark the grids sampled on the table for level 2.*** If picking through the entire first grid of level two subsampling is likely to result in more than 300 ± 20% organisms, then that grid may be subsampled in the same manner as before. Continue to pick grids one at a time until the desired number is reached. Record the total number of grids sampled on the level 3 table.

Complete the laboratory bench sheet for the subsampling procedures. Record the date that the subsampling was completed and the sorters initials on the login sheet. All organisms should be picked from the subsample and the material from the sorting pans disposed of after quality control checks are complete.

SS-4. As the organisms are sorted, put them into glass vials and preserve in 70% ethanol. Label the vials inside with the sample identifier, sampling date, water body name, sample collectors, and initials. If more than one vial is needed, label and number each (e.g., 1 of 2, 2 of 2) to identify the total number of vials for the taxonomist. Insert the labels left-edge first so they can be easily read.

SS-5. Finally, inspect the entire sample for large and rare organisms that were not identified during the subsampling procedure. Pick out the types of organisms that were not collected in the original subsample and place them in a vial labeled “5-minute pick” with the sample identifier, sampling date, and stream name. This procedure is used to ensure that representative taxa are collected for identification. When the 5-minute pick procedure is completed, record the resulting data on the appropriate sections of the login sheet, subsampling sheet, and identification sheet. Record the total numbers of organisms to be identified by the taxonomist from the subsample on the identification sheet.

SS-6. After subsampling is complete, if it is necessary to save the unsorted debris residue, add the words “processed sample” to the outside label, put the contents of the subsampling tray back in the jar(s), and again preserve it in ethanol. The inside label should also indicate that the sample has been processed. Length of storage and archival time are determined by the program manager. If there is no reason to archive the material, discard the remainder of the sample.

SS-7. Turn the vials of organisms and subsampling and identification sheets over to the taxonomist for identification. Qualified taxonomists complete the identification process.

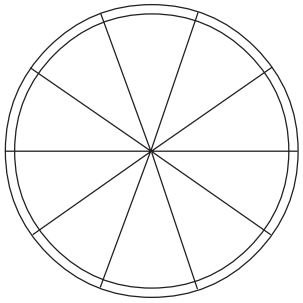
Identification

ID-1. Record on the Laboratory Bench Sheet-Identification (see pages 8 and 9) the station identification code, sample collector initials, collection date, stream location, sorter, date sorted, taxonomist initials, identification date, total number in subsample, and type of subsample. Begin the identification process and list the taxa identified and numbers of each found in the subsample. Insects are identified to genus level by a qualified taxonomist using a dissecting microscope.

Chironomidae (midges) tend to predominate biological communities in Alaska. Characterizing Chironomidae to genus level is a labor-intensive technical process but is important to accurately assess biological condition. Midge larvae are identified primarily by head capsule and mouthpart characteristics and generic identification requires specimens be slide-mounted for examination under a compound microscope (see ID-2 for details). Chironomidae are subsampled to identify them to genus level and yet gain generic-level information.

Non-insect taxa (e.g. Hydrachnidia, Oligochaeta, Amphipoda, etc.) are identified to family or order. These organisms are incorporated in the metrics based on the relative abundance to the total. Also, note in the Taxonomic Certainty Rating (TCR) column any suspect identifications (e.g., very small organisms or those with missing parts) and include the number from 1–5 that indicates the taxonomist's certainty that the identification is correct. Identification of organisms collected from the 5-minute pick procedure are used only to calculate taxa richness measures and are noted in a separate column on the laboratory bench sheet.

ID-2. Chironomidae Subsampling. Identify approximately 20% of the midges for each sample. (This number is derived from the number of Chironomidae recorded on the laboratory data sheet). Separate the midges out of the 300-organism subsample and spread them randomly in a petri dish divided into ten equal sections. (See figure at right.) Choose two sections of the dish to randomly collect approximately 20% of the organisms. Place them in a separate petri dish labeled "Chironomidae 20% subsample." If necessary, continue to remove midges from a third quadrant until you reach the 20% number.



Petri Dish Marked for 20% Chironomidae Subsampling

Sort the midge subsample, using a dissecting microscope, into distinct groups based on physical characteristics (morpho-taxa) including:

- General body appearance – shape and color, length, density, and placement of setae;
- Head capsule – shape, color or markings (stripes, spots, bars, or darkened posterior margin), shape of mentum;
- Antennae – shape, length, presence of elongated bases, presence and shape of Lauterborn organs, and ability to retract antennae.

Complete a separate visual pick using the dissecting microscope to ensure that all taxa are represented in the 20% random subsample by scanning the remaining 80% of the sample. Label a second vial with "Chironomidae visual pick" and place a representative of each taxon found in the visual pick.

Head Capsule Mounting. Slide-mount the organisms collected from both the 20% subsample and visual-pick to prepare for generic identification. Label the slides to clearly show whether it is from

the subsample or the visual pick. ***Those organisms identified for the visual pick are used for taxa richness calculations only.*** Label slides to include study designation, slide number, study site, sample type (i.e., a, b, or q), and subsample type (i.e., 20% subsample or visual pick). Slides with frosted glass labels are recommended.

Use small cover slips (10 ml) to allow mounting multiple midges on each slide and to minimize the number of slides and storage boxes needed for storage. This will accommodate mounting four midges on each slide. Place a drop or two of mounting medium to position each midge on the slide. Use a fast-drying, self-clearing medium such as CMC-10. Place one midge larva in each drop of medium. Use a dissecting microscope for the rest of the mounting process to position the larvae and cover slip. It is critical that the head capsule be positioned ventral-side-up, which may necessitate the decapitation of larger larvae. It is also helpful (but not necessary) to point the anterior of the head capsule toward the bottom of the slide. Positioned thus, the head capsule will appear right side up when viewed through a compound microscope for identification.

Add the cover slip after the midge is in position by holding it with forceps. Place one edge of the cover slip on the slide and then allow the opposite edge to fall. This will prevent air bubbles from being trapped beneath the cover slip. Gently press the tip of the forceps to the top of the cover slip in order to fine-tune the specimen and cover slip into position. Press the cover slip onto the head capsule with sufficient pressure to spread the mandibles. Mounting groups of taxa with similar morphological characteristics sequentially will speed the identification process.

Identification. A compound microscope, preferably with phase-contrast lighting and 4*x*, 10*x*, 40*x*, and 100*x* (oil immersion) objectives, is necessary for generic identification. Taxonomic keys used for midge larvae include Wiederholm (1983) or Merritt and Cummins (1996). The diagnoses and drawings in Wiederholm (1983) are very helpful to confirm identifications. Record final identifications on the Laboratory Bench Sheet-Chironomidae Identification (see page 9). Remember to multiply the abundance of each genus by five to extrapolate the number of Chironomidae in the entire sample.

ID-3. Refill the specimen vials with ethanol, label, and group by station and date for archival. Make sure the vials are tightly capped. Periodically examine the ethanol level in these jars and replenish as needed. Maintain archived samples in the laboratory.

ID-4. Refer to Merritt and Cummins (1996) for feeding group and habit designations.

Quality Control

QC-1. Sorting. Examine 10% of the sorted samples in each lot. This will be done by the person designated responsible for laboratory quality control. (A lot is defined as a special study, basin study, index period, or individual sorter.) Examine the material in the sorting tray to look for organisms missed by the sorter. Any organisms found are added to the vials for that sample. The sample passes if less than 30 organisms (10% of the subsample) are found; the sample fails if more than 30 (10%) are found. Check 100% of the samples sorted by new personnel until samples pass consistently. Complete random sort checks after this time.

QC-2. Identification. Maintain a voucher collection of all samples and subsamples. Label, preserve, and store these samples in the laboratory for future reference. Samples should be spot checked by a second taxonomist and differences in identification recorded in a taxonomy validation notebook. If no consensus can be reached as to the identification of the organism, send it out to a third taxonomist.

Add labels with specific taxa names to the specimen vials as they are identified. Extract individual specimens as necessary to develop a reference collection or for identification verification by a second taxonomist. The identifying taxonomist initials slides for Chironomidae identification. Keep these in a slide file.

Maintain a reference collection of each identified taxon that is verified by a second taxonomist. Add the word "validated" and the first initial and last name of the person validating the identification to the vial label. Record the date specimens are sent out for taxonomy validations in a taxonomy validation notebook and include the label information. Record the date received, the results, and the name of the person who performed the validation in the notebook upon return of the specimens.

QC-3. Tracking. Record information on laboratory progress of the samples (i.e., subsampling, sorting, and taxonomy) on the login sheet to track the progress of each sample within the sample lot.

QC-4. Cleaning Equipment. Rinse thoroughly all sieves, pans, trays, etc., that have come into contact with the sample after laboratory processing is complete. Examine the equipment carefully and pick off any organisms or debris; add any organisms found to the sample residue.

QC-5. Reference Resources. Maintain and update as necessary taxonomic literature (see list below). These resources are essential for identification of specimens. As possible, support taxonomists to participate in periodic training of specific taxonomic groups to ensure accurate identifications.

Taxonomic References

Clifford, H.F. 1991. Aquatic invertebrates of Alberta. The University of Alberta Press, Edmonton, Alberta, Canada. 538 pp.

Edmunds, G.F., Jr., S.L. Jensen, and L. Berner. The mayflies of North and Central America. University of Minnesota Press, Minneapolis, MN. 330 pp.

Merritt, R.W., and K.W. Cummins, eds. 1996. An introduction to the aquatic insects of North America. 3rd ed. Kendall/Hunt Publishing Company, Dubuque, IA. 862 pp.

Peckarsky, B.L., et al. 1990. Freshwater macroinvertebrates of northeastern North America. Cornell University Press, Ithaca, NY. 442 pp.

Pennak, R.W. 1989. Fresh-water invertebrates of the United States: Protozoa to mollusca. 3rd ed. John Wiley and Sons, NY. 628 pp.

Stewart, K.W., and B.P. Stark. 1993. Nymphs of North American stonefly genera (Plecoptera). University of North Texas Press, Denton, TX. 457 pp.

Wiederholm, T. 1983. Chironomidae of the Holarctic region: keys and diagnoses. Part 1-Larvae. Entomologica Scandinavica. Supplement 19.

Wiggins, G.B. 1996. Larvae of the North American caddisfly genera (Trichoptera). 2nd ed. University of Toronto Press, Toronto, Canada. 457 pp.

SAMPLE LOGIN SHEET

Sample ID	Stream Name	Collect Date	Samplers ID	Sample Processing						QC	
				# Jars	SS Y/N	5' Pick Y/N	Sorting Date/Init	ID Date/Init	Chiro Date/Init	Processing Date/Init	ID Date/Init

LABORATORY BENCH SHEET: Subsampling

Station ID_____Stream Name _____Sample Date _____

Processing Date _____ Sorter Init._____

Circle appropriate type of subsample processing: 300 5' Pick QC Other

Please check the appropriate subsampling levels used
and the number of grids picked in each:

Final SS: No. level 1 grids____ *x* No. level 2 grids _____ *x* No. level 3 grids _____

Level 1. Note random grids selected. **T No. Of Grids _____**

	1	2	3	4	5	6
A						
B						
C						
D						
E						

Level 2. Note random grids selected. **T No. Of Grids _____**

	1	2	3	4	5	6
A						
B						
C						
D						
E						

Level 3. Note random grids selected. **T No. Of Grids _____**

	1	2	3	4	5	6
A						
B						
C						
D						
E						

Total Number of organisms in final subsample _____

LABORATORY BENCH SHEET – Identification

Station ID _____Collected by _____Date _____ Stream_____

Sorted by_____ Date Sorted_____ Subsample *(underline)*: 300 (+/-20%) 5' Pick QC 5' Pick Other

Taxonomist Init. _____ Date ID _____

Organisms	No.	Found in 5' pick	TCR*
Ephemeroptera			
Ameletidae - *Ameletus*			
Baetidae - unid			
- *Acentrella*			
- *Baetis*			
Ephemerellidae - unid			
- *Drunella*			
- *Ephemerella*			
Heptageniidae - unid			
- *Cinygmula*			
- *Epeorus*			
- *Rithrogena*			
Leptophlebiidae - *Paraleptophlebia*			
Plecoptera			
Capniidae - unid			
- *Capnia*			
Capniidae/Leuctridae - unid			
Leuctridae - unid			
- *Despaxia*			
Chloroperlidae - unid			
- *Neaviperla*			
- *Plumiperla*			
- *Suwallia*			
- *Sweltsa*			
Nemouridae - *Nemoura*			
- *Zapada*			
Perlodidae - unid			
- *Isoperla*			
- *Diura*			
Pteronarcyidae - *Pteronarcella*			
Taeniopterygidae - *Taenionema*			
Turbellaria			
Gastropoda			
Lymnaeidae - Lymnaea			
Physidae - *Physa*			
Planorbidae - unid			
Valvatiidae - *Valvata*			
Bivalvia - Sphaeriidae			
Arachnoidea - Hydracarina			
Crustacea			
Amphipoda - unid			
Gammaridae - unid			
Ostracoda - unid			
Oligochaeta			
Nematoda			

Organisms	No.	Found in 5' pick	TCR*
Trichoptera			
Apataniidae - *Apatania*			
Brachycentridae - *Brachycentrus*			
Glossosmatidae - *Glossosoma*			
Hydroptilidae - *Ochrotrichia*			
- *Oxyethira*			
Hydrosychidae - *Hydropsyche*			
- *Arctopsyche*			
Lepidostomatidae - *Lepidostoma*			
Leptoceridae - *Ceraclea*			
Limnephilidae -unid			
- *Ecclisomyia*			
- *Eocosmoecus*			
- *Grensia*			
- *Hesperophylax*			
- *Limnephilus*			
- *Onocosmoecus*			
- *Psychoglypha*			
Polycentropodidae - *Polycentropus*			
Rhyacophilidae - *Rhyacophila*			
Uenoidae - *Neophylax*			
Diptera			
Ceratopogoniidae - unid			
- *Bezzia*			
- *Ceratopogon*			
- *Probezzia*			
Chironomidae - unid			
Empididae - unid			
- *Chelifera*			
- *Clinocera*			
- *Oreogeton*			
Psychodidae - *Pericoma*			
Sciomyzidae - unid			
Simuliidae - *Simullium*			
Tipulidae - unid			
- *Dicranota*			
- *Hesperoconopa*			
- *Hexatoma*			
- *Tipula*			
Coleoptera			
Other			
Other			

*Taxonomic certainty rating (TCR): 5 = most certain, 1 = least certain. If rating is 3–1, give reason (e.g., missing gills).

Total No. Organisms _____ **Total No. Taxa** _____

LABORATORY BENCH SHEET – Chironomidae Identification

Station ID _____ Collected by _____ Date _____ Stream_____

Sorted by_____ Date Sorted_____ Subsample *(underline)*: (20%) 5' Pick Other

Taxonomist Init. _____ Date ID _____ Total No. of Slides _____

Tribe-Genus	No.	Found in 5' pick	TCR*
Chironomidae – unid			
Chironomini – unid			
- Chironomus			
- Cryptochironomus			
- Dicrotendipes			
- Einfeldia			
- Glypotendipes			
- Kiefferulus			
- Omisus			
- Parachironomus			
- Paracladopelma			
- Paralauterborniella			
- Paratendipes			
- Phaenopsectra			
- Polypedilum			
- Stenochironomus			
- Stictochironomus			
Diamesinae – unid			
- Diamesa			
- Diamesinae			
- Lappodiamesa			
- Odontomesa			
- Pagastia			
- Potthastia			
- Prodiamesa			
- Sympotthastia			
Tanypodinae – unid			
- Brundiniella			
- Coelotanypus			
- Conchapelopia			
- Helopelopia			
- Krenopelopia			
- Larsia			
- Macropelopia			
- Natarsia			
- Nilotanypus			
- Oliveridia			
- Paramerina			
- Rheopelopia			
- Tanypodinae			
- Telmatopelopia			
- Thienemannimyia			
- Xenopelopia			
- Zavrelimyia			
Podonominae *- Boreochlus*			

*Taxonomic certainty rating (TCR): 5 = most certain, 1 = least certain. If rating is 3–1, give reason (e.g., young specimen).

Tribe-Genus	No.	Found in 5' pick	TCR*
Orthocladiinae – unid			
- Brillia			
- Cardiocladius			
- Chaetocladius			
- Corynoneura			
- Cricotopus			
- Diplocladius			
- Doithrix			
- Eudactylcladius			
- Eukiefferiella			
- Euorthocladius			
- Heterotrissocladius			
- Hydrobaenus			
- Krenosmittia			
- Limnophyes			
- Metriocnemus			
- Nanocladius			
- Oliveridia			
- Orthocladius			
- Paracricotopus			
- Parakiefferiella			
- Parametriocnemus			
- Paraphaenocladius			
- Paratrichocladius			
- Parorthocladius			
- Psilometriocnemus			
- Rheocricotopus			
- Rheosmittia			
- Smittia			
- Stilocladius			
- Symposiocladius			
- Synorthocladius			
- Thienemanniella			
- Tvetenia			
- Zalutschia			
Tanytarsini – unid			
- Cladotanytarsus			
- Constempellina			
- Micropsectra			
- Paratanytarsus			
- Radotanytarsus			
- Rheotanytarsus			
- Stempellina			
- Stempellinella			
- Tanytarsini			
- Tanytarsus			
- Zavrelia			

Total No. Chironomidae _____ **Total No. Taxa** _____

APPENDIX 2. FIELD DATA FORMS

To be provided prior to initiation of field data collection

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AEA SuWa FDA - River Productivity Study **Benthic/Drift Sampling Field Form** **Form A - Site Info**

Event & Site Information

Site ID:	Date(s):	Crew:
Weather:		Consultant/Organization:
Site Arrival Time:		Site Departure Time:
Event Type: Spring Summer Fall Pre-Storm Event (# ___) Post-Storm Event (# ___)		
Stream Name:	Stream Code:	PRM (if known):
Station ID:	Focus Area:	
DS Coords (WGS84): N W		DS Coord Description:
GPS Unit:	GPS Date:	GPS Wpt:
US Coords (WGS84): N W		US Coord Description:
GPS Unit:	GPS Date:	GPS Wpt:
Hydrologic Segment (L1): Upper Middle Lower	MC Hab Type (L3): MC Split MC Mult Split MC SC NA	
Geomorphic Reach (L2):	OCH Type (L3): Side Slough Upland Slough NA	
MS Hab Category (L3): MC OCH NA (i.e., TRIB)	MC/OCH Spcl Mesohab Type (L4): Trib Mouth Trib Plume None	

Site Comments & Sketches:

Data Logger Notes:

Camera ID	*Photo #*	*Photo Description*

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

R2 Resource Consultants, Inc
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AEA SuWa FDA - River Productivity Study **Benthic/Drift Sampling Field Form** **Form B - Benthic Macros & Algae**

Header Info

Site ID:	Date:	Crew:

Benthic Macroinvertebrate & Algae Sample Collection **Hess Sample Location #:**

Sample Start Time:		Sample End Time:	
Coords (WGS84): N	W	Coord Description:	
GPS Unit:	GPS Date:	GPS Wpt:	
MC/OCH Mesohab Type (L4): Rapid[1] Riffle Run Glide		MC/OCH Pool Subtype: Str Scour Lat Scour	
[1] Applies to MC only. [2] Applies to OCH only. Pool Beaver Complex[2] NA		Plunge Pool BW Pool NA	
Water Temperature (ºC):	Turbidity (NTU):	_____ (___):	_____ (___):
_____ (___):	_____ (___):	_____ (___):	_____ (___):
Water Depth (m):	Mean Velocity (60% of depth; fps):	Mean Boundary Layer Velocity (fps):	
Substrate: ____% Organic ____% Sand/Silt ____% Gravel ____% Cobble ____% Boulder ____% Bedrock			
Sample Location Comments:			
Hess (HS) Sample ID:	Sample Collected by:	Time Collected:	
Benthic Macroinvertebrate Sample Comments:			

Algal Substrate Piece	*Pre-Sample Diameter (cm)*	*Post-Sample Diameter (cm)*	*Average Diameter (cm)*	Combined Average Diameter (cm):
1				Surface Area Sampled (cm^2):
2				[calculated as $A = (n)(\pi)(d/2)^2$]
3				Sample Collected by:
4				Time Collected:
5				Total Samp Vol (ml):

Algal Samp Type	*Samp Vol (ml)*	*Subsamp Vol (ml)*	*Sample ID*	*Comment*
Chl-a (CH)				
AFDM (AF)				
Archived				

PAR Measurement Depth (m)	*PAR Measurement ($\mu mol\ s^{-1}\ m^{-2}$)*	*PAR Measurement Depth (m)*	*PAR Measurement ($\mu mol\ s^{-1}\ m^{-2}$)*	Algal Sample Comments:

Camera ID	*Photo #*	*Photo Description*

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

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AEA SuWa FDA - River Productivity Study **Benthic/Drift Sampling Field Form** **Form C - Transect Depth & Velocity**

Header Info

Site ID:	Date:	Crew:

Transect Depth & Velocity Data **Hess Sample Location #:**

Lateral Transect Description:

Longitudinal Transect Description:

Transect (Lat or Long)	*Tape Station (m)*	*Water Depth (m)*	*Mean Velocity (60% of depth; fps)*	*Mean Boundary Layer Velocity (fps)*	*Depth & Velocity Measurement Comments*

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

AEA SuWa FDA - River Productivity Study **Benthic/Drift Sampling Field Form** **Form D - Drift Samples**

Header Info

Site ID:	Date:	Crew:

Drift Sample Location

Sample Start Time:			Sample End Time:		
Coords (WGS84): N		W	Coord Description:		
GPS Unit:	GPS Date:	GPS Wpt:			
MC/OCH Mesohab Type (L4): Rapid[1] Riffle Run Glide			MC/OCH Pool Subtype:	Str Scour	Lat Scour
[1] Applies to MC only. [2] Applies to OCH only. Pool Beaver Complex[2] NA			Plunge Pool	BW Pool	NA
Sample Location Comments:					

Drift Net Sample 1 ***Drift Net Sample 2***

Drift Net Dimensions (width x height; m): _____ x _____			Drift Net Dimensions (width x height; m): _____ x _____		
Parameter	*Start*	*End*	*Parameter*	*Start*	*End*
Water Depth at Net Location (m)			Water Depth at Net Location (m)		
Dist from Bottom of Net to WSE (m)			Dist from Bottom of Net to WSE (m)		
Sample Time			Sample Time		
In-Net Flow Meter Counter Reading			In-Net Flow Meter Counter Reading		
Net Entrance Depth (m)			Net Entrance Depth (m)		
Velocity at Net Ent (fps)			Velocity at Net Ent (fps)		
Velocity at 60% Depth (fps)			Velocity at 60% Depth (fps)		
Turbidity (NTU)			Turbidity (NTU)		
_____ (____)			_____ (____)		
_____ (____)			_____ (____)		
Drift (DR) Sample ID:			Drift (DR) Sample ID:		
Sample Collected by:			Sample Collected by:		
Sample Collection Comments:			Sample Collection Comments:		

Camera ID	*Photo #*	*Photo Description*

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

R2 Resource Consultants, Inc
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AEA SuWa FDA - River Productivity Study **Benthic/Drift Sampling Field Form** **Form E - Snag Samples**

Header Info

Site ID:	Date:	Crew:

Snag Sample Collection **Snag Sample #:**

Sample Start Time:			Sample End Time:	
Coords (WGS84): N		W	Coord Description:	
GPS Unit:	GPS Date:	GPS Wpt:		
MC/OCH Mesohab Type (L4): Rapid[1] Riffle Run Glide			MC/OCH Pool Subtype: Str Scour Lat Scour	
[1] Applies to MC only. [2] Applies to OCH only. Pool Beaver Complex[2] NA			Plunge Pool BW Pool NA	
Water Depth at Snag (m):	Snag Depth (m):		Dist from Stream Bed to Snag (m):	
Velocity at 60% Depth (fps):			Velocity at Snag (fps):	
Substrate: ____% Organic ____% Sand/Silt ____% Gravel ____% Cobble ____% Boulder ____% Bedrock				
Snag (LW) Sample ID:		Sample Collected by:	Time Collected:	
Snag Length (cm):		Snag Diameter (cm):		
Circumference 1 (cm):	Circumference 2 (cm):	Circumference 3 (cm):	Avg Circumference (cm):	
Snag Sample Comments:				

Camera ID	*Photo #*	*Photo Description*

Snag Sample Collection **Snag Sample #:**

Sample Start Time:			Sample End Time:	
Coords (WGS84): N		W	Coord Description:	
GPS Unit:	GPS Date:	GPS Wpt:		
MC/OCH Mesohab Type (L4): Rapid[1] Riffle Run Glide			MC/OCH Pool Subtype: Str Scour Lat Scour	
[1] Applies to MC only. [2] Applies to OCH only. Pool Beaver Complex[2] NA			Plunge Pool BW Pool NA	
Water Depth at Snag (m):	Snag Depth (m):		Dist from Stream Bed to Snag (m):	
Velocity at 60% Depth (fps):			Velocity at Snag (fps):	
Substrate: ____% Organic ____% Sand/Silt ____% Gravel ____% Cobble ____% Boulder ____% Bedrock				
Snag (LW) Sample ID:		Sample Collected by:	Time Collected:	
Snag Length (cm):		Snag Diameter (cm):		
Circumference 1 (cm):	Circumference 2 (cm):	Circumference 3 (cm):	Avg Circumference (cm):	
Snag Sample Comments:				

Camera ID	*Photo #*	*Photo Description*

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

AEA SuWa FDA - River Productivity Study **Benthic/Drift Sampling Field Form** **Form F1 - Stable Isotope Samples**

Header Info

Site ID:	Date:	Crew:

Stable Isotope Sample Collection - Invertebrates

SI Sample Type	*Type ID*	*Taxon*	*# Samples*	*Sample ID(s)*	*Sample Collection Comments*
Bentic Macros	SIBM**G**	Grazers	3		
	SIBM**C**	Collectors	3		
	SIBM**S**	Shredders	3		
	SIBM**P**	Predators	3		
Macro Drift	SIDR__*		2		
	SIDR__		2		
	SIDR__		2		
	SIDR__		2		
	SIDR__		2		
	SIDR__		2		
Macro Emerg.	SIEM**G**	Grazers	1		
	SIEM**C**	Collectors	1		
	SIEM**S**	Shredders	1		
	SIEM**B**	Predators	1		

*** Note**: On the blank provided within the Type ID column, create a unique identifier letter that represents that taxon group sorted from drift samples. This taxon ID letter will be assigned to sorted and separate composite samples based on functional feeding group.

Time Zone _____

QC1 Init Date _____

Data Entry Init Date _____

QC2 Init Date _____

AEA SuWa FDA - River Productivity Study **Benthic/Drift Sampling Field Form** **Form F2 - Stable Isotope Samples**

Header Info

Pg ____ of ____

Site ID:	Date:	Crew:

Stable Isotope Sample Collection - Endmembers

SI Sample Type	*Type ID*	*Taxon*	*# Samples*	*Sample ID(s)*	*Sample Collection Comments*
Benthic Algae	SIAL	Composite	3		
Organic Matter (benthic)	SIBO	Composite	3		
Organic Matter (seston)	SISE	Composite	2		
Carcass (dorsal muscle tissue)	SICA*				

Carcass Taxon Types and IDs

SCK	Chinook salmon
SCO	coho salmon
SCH	chum salmon
SPI	pink salmon
SSE	sockeye salmon

* **Note**: A total of 20 carcass tissue samples will be taken if possible, one from each carcass found. Record each identified species using the taxon ID codes.

Time Zone _________

QC1 Init Date ____________

Data Entry Init Date ____________

QC2 Init Date ____________

AEA SuWa FDA - River Productivity Study **Benthic/Drift Sampling Field Form** **Form G - Grab Samples & Plankton Tows**

Header Info

Site ID:	Date:	Crew:

Grab Sample Collection

Sample Start Time:			Sample End Time:	
Coords (WGS84): N		W	Coord Description:	
GPS Unit:	GPS Date:	GPS Wpt:		
MC/OCH Mesohab Type (L4): Rapid[1] Riffle Run Glide			MC/OCH Pool Subtype: Str Scour Lat Scour	
[1] Applies to MC only. [2] Applies to OCH only. Pool Beaver Complex[2] NA			Plunge Pool BW Pool NA	
Water Temperature (°C):	Turbidity (NTU):		_____ (___):	_____ (___):
Water Depth (m):	Mean Velocity (60% of depth; fps):		Mean Boundary Layer Velocity (fps):	
Substrate: ____% Organic ____% Sand/Silt ____% Gravel ____% Cobble ____% Boulder ____% Bedrock				
Sample Location Comments:				
Grab Sample ID:			Sample Collected by:	Time Collected:
Grab Sample ID:			Sample Collected by:	Time Collected:
Grab Sample ID:			Sample Collected by:	Time Collected:
Grab Sample ID:			Sample Collected by:	Time Collected:
Grab Sample ID:			Sample Collected by:	Time Collected:
Grab Sample Comments:				

Plankton Tow

Sample Start Time:			Sample End Time:	
Coords (WGS84): N		W	Coord Description:	
GPS Unit:	GPS Date:	GPS Wpt:		
MC/OCH Mesohab Type (L4): Rapid[1] Riffle Run Glide			MC/OCH Pool Subtype: Str Scour Lat Scour	
[1] Applies to MC only. [2] Applies to OCH only. Pool Beaver Complex[2] NA			Plunge Pool BW Pool NA	
Water Temperature (°C):	Turbidity (NTU):	Tow Distance (m):		Tow Dir'n: US DS
Water Depth (m):	Mean Velocity (60% of depth; fps):		Mean Boundary Layer Velocity (fps):	
Substrate: ____% Organic ____% Sand/Silt ____% Gravel ____% Cobble ____% Boulder ____% Bedrock				
Sample Location Comments:				
Plankton Tow Sample ID:			Sample Collected by:	Time Collected:
Plankton Tow Sample ID:			Sample Collected by:	Time Collected:
Plankton Tow Sample ID:			Sample Collected by:	Time Collected:
Plankton Tow Sample ID:			Sample Collected by:	Time Collected:
Plankton Tow Sample ID:			Sample Collected by:	Time Collected:
Plankton Tow Comments:				

Camera ID	*Photo #*	*Photo Description*

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

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AEA SuWa FDA - River Productivity Study **Adult Emergence Sampling** **Form A - Site, Trap, & Sample Info**

Event & Site Information

Site ID:	Date:	Crew:	
Weather:		Consultant/Organization:	
Site Arrival Time:		Site Departure Time:	
Event Type:	Trap Installation	Sample Collection	
Stream Name:	Stream Code:	PRM (if known):	
Station ID:	Focus Area:		

Trap Location

Coords (WGS84): N		W	Coord Description:
GPS Unit:	GPS Date:	GPS Wpt:	
Trap Distance from Shoreline (wetted edge; m):			Wat Depth below Trap (m):
Trap Comments & Sketches:			

Sample Collection

Emerg (EM) Sample ID:	Sample Collected by:	Time Collected:
Sample Comments:		

Photos

Camera ID	*Photo #*	*Photo Description*

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

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AEA SuWa FDA - River Productivity Study **Colonization Sampling** **Form A - Site Info**

Event & Site Information

Site ID:	Date:	Crew:	
Weather:		Consultant/Organization:	
Site Arrival Time:		Site Departure Time:	
Event Type:	Substrate Deployment	Sample Collection	
Stream Name:	Stream Code:	PRM (if known):	
Station ID:	Focus Area:		
DS Coords (WGS84): N	W	DS Coord Description:	
GPS Unit:	GPS Date:	GPS Wpt:	
US Coords (WGS84): N	W	US Coord Description:	
GPS Unit:	GPS Date:	GPS Wpt:	

Hydrologic Segment (L1): Upper	Middle	Lower	MC Hab Type (L3): MC	Split MC	Mult Split MC	SC	NA
Geomorphic Reach (L2):			OCH Type (L3): Side Slough	Upland Slough			NA
MS Hab Category (L3): MC	OCH	NA (i.e., TRIB)	MC/OCH Spcl Mesohab Type (L4): Trib Mouth	Trib Plume			None
MC/OCH Mesohab Type (L4): Rapid[1]	Riffle	Run	Glide		MC/OCH Pool Subtype:	Str Scour	Lat Scour
[1] Applies to MC only. [2] Applies to OCH only. Pool	Beaver Complex[2]	NA			Plunge Pool	BW Pool	NA

Site Condition: Turbid/Warm Turbid/Cold Clear/Warm Clear/Cold

Site Comments & Sketches:

Camera ID	*Photo #*	*Photo Description*

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

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AEA SuWa FDA - River Productivity Study **Colonization Sampling** **Form B - Hester-Dendy Samplers**

Header Info

Site ID:	Date:	Crew:

Hester-Dendy Sampler Info, Conditions, & Sample Collection **Hester-Dendy Sampler Set ID:**

Coords (WGS84): N	W		Coord Description:
GPS Unit:	GPS Date:	GPS Wpt:	
Deployment Date:	Expected Deployment Duration (weeks):		Qualitative Deployment Depth: Shallow Deep

Sampler Depth (m): Veloc at 60% Depth (fps):	*PAR Measurement Depth (m)*	*PAR Measurement ($\mu mol\ s^{-1}\ m^{-2}$)*	*PAR Measurement Depth (m)*	*PAR Measurement ($\mu mol\ s^{-1}\ m^{-2}$)*
Mean Bound Layer Veloc (fps):				
Water Temperature (ºC):				
Turbidity (NTU):				
_____ (_____):				
_____ (_____):				

Sampler Comments:

Replic #	*Hester-Dendy (HD) Sample ID*	*Collected by*	*Time*	*Sample Comments*
1				
2				
3				

Hester-Dendy Sampler Info, Conditions, & Sample Collection **Hester-Dendy Sampler Set ID:**

Coords (WGS84): N	W		Coord Description:
GPS Unit:	GPS Date:	GPS Wpt:	
Deployment Date:	Expected Deployment Duration (weeks):		Qualitative Deployment Depth: Shallow Deep

Sampler Depth (m): Veloc at 60% Depth (fps):	*PAR Measurement Depth (m)*	*PAR Measurement ($\mu mol\ s^{-1}\ m^{-2}$)*	*PAR Measurement Depth (m)*	*PAR Measurement ($\mu mol\ s^{-1}\ m^{-2}$)*
Mean Bound Layer Veloc (fps):				
Water Temperature (ºC):				
Turbidity (NTU):				
_____ (_____):				
_____ (_____):				

Sampler Comments:

Replic #	*Hester-Dendy (HD) Sample ID*	*Collected by*	*Time*	*Sample Comments*
1				
2				
3				

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

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AEA SuWa FDA - River Productivity Study | **Fish Tissue, Gut, & Scale Sampling** | **Form A - Site Info & Sample Coll'n**

Event & Site Information

NOTE: FDA indicates that values used should be those obtained by the FDA team.

Site ID (FDA):	Date:	Gut Sample Tech(s):	Consultant/Organization:
Site Arrival Time:	Site Departure Time:	Fish Survey Crew:	Fish Survey Consultant:
Focus Area (if applic):	Stream Name (FDA):	Stream Code (FDA):	PRM (if known; FDA):
Corr RP Site ID:	Weather:		Camera ID:

DS Coords (non-FDA; WGS84): N		W	DS Coord Description:
GPS Unit:	GPS Date:	GPS Wpt:	
US Coords (non-FDA; WGS84): N		W	US Coord Description:
GPS Unit:	GPS Date:	GPS Wpt:	

Site & Event Comments:

Pit Reader Test: P F

Sample Collection *Target Species & Life Stages: SCK & SCO - JUV & CAR*; TRB - JUV & ADT; SCH, SPI, & SSE - CAR* (*CAR will be sampled for SICA only)*

Specimen ID (RP-A001)	*Species*	*Life Stg*	*Capt Meth*	*Coll'n Time*	*Pass #*	*FL (mm)*	*Weight (g)*	*Scales*	*Gut Contents*	*Tissue samples*	*PIT Scan*	*PIT Tag Code (recaptures only)*	*Comments (e.g., genetic sample type & bottle/vial ID, photo IDs)*

Species Codes		***Life Stage Codes***		***Scale Codes***		***Gut Content Codes***		***Tissue Sample Codes***		***Fish Capture Codes***					
SCK	Chinook salmon	ADT	adult	L	Scales collected from left side (preferred)	Y	Collected	FC	Fin clip (ADT + JUV > 50 mm	ANG	angling	GNF	gill net, floating	SNK	snorkel
SCH	chum salmon	CAR	carcass			N	Not collected	WB	Whole fish (JUV < 50 mm F	PEF	backpack electrofisher	GNS	gill net, sinking	STL	set line, unbaited
SCO	coho salmon	FRY	fry	R	Scales collected from right side			DM	Dorsal muscle (CAR)	BEF	boat-mounted electrofishe	HON	hoop net	STLB	set line, baited
SPI	pink salmon	JUV	juvenile					N	Not collected	DIP	dip net	MINB	minnow trap, baited	TAN	tangle net
TRB	rainbow trout	JOA	juvenile/adult	N	No scales collected					FWL	fishwheel	MIN	minnow trap, unbaited	TRL	trotline, unbaited
SSE	sockeye salmon	PAR	parr							FYK	fyke net	SCT	screw trap	TRLB	trotline, baited
		SMT	smolt							GND	gill net, drifting	SEN	seine net	OTH	other
												VOG	visual observation, ground		

NOTE: Fish recorded on this form should be counted, but not measured or weighed, on Form C or Form C-DMT.

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

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AEA SuWa FDA - River Productivity Study | **Fish Tissue, Gut, & Scale Sampling** | **Form B - Sample Coll'n (cont)**

Header Info

Site ID (FDA):	Date:	Gut Sample Tech(s):

Sample Collection *Target Species & Life Stages: SCK & SCO - JUV & CAR*; TRB - JUV & ADT; SCH, SPI, & SSE - CAR* (*CAR will be sampled for SICA only)*

Specimen ID (RP-A001)	*Species*	*Life Stg*	*Capt Meth*	*Coll'n Time*	*Pass #*	*FL (mm)*	*Weight (g)*	*Scales*	*Gut Contents*	*Tissue samples*	*PIT Scan*	*PIT Tag Code (recaptures only)*	*Comments (e.g., genetic sample type & bottle/vial ID, photo IDs)*

Species Codes		***Life Stage Codes***		***Scale Codes***		***Gut Content Codes***		***Tissue Sample Codes***		***Fish Capture Codes***		GNF	gill net, floating	SNK	snorkel
SCK	Chinook salmon	ADT	adult	L	Scales collected from left side (preferred)	Y	Collected	FC	Fin clip (ADT + JUV > 50 mm	ANG	angling	GNS	gill net, sinking	STL	set line, unbaited
SCH	chum salmon	CAR	carcass			N	Not collected	WB	Whole fish (JUV < 50 mm F	PEF	backpack electrofisher	HON	hoop net	STLB	set line, baited
SCO	coho salmon	FRY	fry	R	Scales collected from right side			DM	Dorsal muscle (CAR)	BEF	boat-mounted electrofishe	MINB	minnow trap, baited	TAN	tangle net
SPI	pink salmon	JUV	juvenile					N	Not collected	DIP	dip net	MIN	minnow trap, unbaited	TRL	trotline, unbaited
TRB	rainbow trout	JOA	juvenile/adult	N	No scales collected					FWL	fishwheel	SCT	screw trap	TRLB	trotline, baited
SSE	sockeye salmon	PAR	parr							FYK	fyke net	SEN	seine net	OTH	other
		SMT	smolt							GND	gill net, drifting	VOG	visual observation, ground		

NOTE: Fish recorded on this form should be counted, but not measured or weighed, on Form C or Form C-DMT.

Time Zone _____

QC1 Init Date _____
Data Entry Init Date _____
QC2 Init Date _____

APPENDIX 3. SUSITNA DATA STANDARDS

(significant changes since last version are in green)

AEA Susitna Project – Water Resources Programs

Field Data Collection, Processing, and Delivery Standards

version April 10, 2013 DRAFT

For questions and comments concerning this document,
contact the Susitna Project Data Resource Manager:
Dana Stewart, DES.IT,LLC
(datadana3@yahoo.com)

(significant changes since last version are in green)

AEA Susitna Project – Water Resources Programs
Field Data Collection, Processing, and Delivery Standards

Contents

Acronyms in This Document

ADNR	Alaska Dept. of Natural Resources
AEA	Alaska Energy Authority
MS	Microsoft
QC	Quality control
UAF GINA	Univ. of Alaska, Fairbanks – Geographic Information Network of Alaska

(significant changes since last version are in green)

A. Data Collection, Backup, and Delivery

In general, the process for preparing and submitting field data includes the following steps:

1. Create field forms and mobile device entry screens. Review with Dana Stewart and Judy Simon at least 2 weeks before field trip.
2. Project data resource manager creates data templates and dictionary from the field forms and delivers them to the consultant's data coordinator. The templates define the format for final data submittals by consultants to AEA for the project database. (Applies only to water quality and fish.)
3. In the field, record data on field forms or in mobile devices and do QC1. Data might also be entered to electronic format, which is QC2.
4. Backup field forms, field books, and mobile devices (ArcPad, Trimble, cameras, GPS, thermistors, etc.) nightly.
5. Submit these raw deliverables to AEA at least monthly, via AEA SharePoint. AEA considers these to be interim deliverables. Very large files can be submitted to AEA IT on external drives or DVDs.
6. Enter data to electronic format (QC2) and process the raw data as needed for the study: assign site IDs if not done in the field, flag unusable records, perform data reduction, etc.
7. A final review is done by a senior scientist (QC3).
8. Format data for submittal to the AEA project database, using data templates if provided.
9. Submit final QC3 data files to AEA SharePoint or via hard drive, as done for raw data. (Refer to the GIS User Guide for delivery of GIS data.)
10. For data being delivered for storage in the project database, data must be accompanied by a data dictionary.
11. For database submittals only, the project data resource manager will perform QC4 review and coordinate revisions with the consultant's Data Coordinator.
12. Data and dictionary are incorporated into the Susitna project relational database. No more revisions can be made in the data by consultants, as the data is considered Final for the study year.
13. If data revisions are needed later, such as for QC5, they'll be coordinated by the project data resource manager. The appropriate QC columns will be updated, which will serve as adequate documentation.

QC Protocol – Briefly

- 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 A: Data QC Protocol.
- 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 scientist 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.

File Paths / Names

- All delivered files should be named to clearly identify the source and type of data within. If helpful, these file names may include folder names to group files together by field event and data type, such as for photo collections.
- The maximum filename length is 250 characters, including folder names and the file extension.
- All delivered files must be accompanied by a **Letter of Transmittal** which will include the information below, expanding on codes / shorthand as needed to clearly identify the deliverable. The template for the Letter of Transmittal is provided in the Appendices.
- Include the following information within file path / names, in the order below:

Descriptor	Format / Example
project name	SuWa
submitting comp./agency	HDR, LGL, ADFG, R2, etc.
program name	FA-IFS, FAQ
study subject	ChanMorph, AqHabitat, FishRadioTelem, ButterflyCollection, etc.
beginning study date	YYYYMMDD
study area/location	MidRiver , DevilCanyon, RM180.4
deliverable type	Photo, FieldBk, FieldFrm, HoboDump, GPSDump, etc.
field form name	(if applicable) Title of the field form included
QC level	QC1, QC2, or QC3
equipment name	(if applicable) GPS name, thermistor serial number, camera name, etc.
Data Coordinator staff	initials
date submitted	YYYYMMDD (or date of photo)
sequential file name	(if applicable) photo numbers, etc. Original camera photo names are ok, IF unique within the folder. A catalog with more descriptive info is expected for photos.
file type	.xls, .mdb, .pdf, .jpg, etc.

Examples:
SuWa Golder FAQ SalmonLifeHist 201307 MidRiver Database QC3 DF 20130830.mdb

SuWa LGL FAQ FishRadioTelem 20120601 MidRiver\GPS dumps QC1\GPS12 MB 20120610.txt
SuWa R2 IFS Riparian\20120731 RM98\Photos QC1 JZ 20120831 \IMGP2041.jpg

Field Data Collection Guidelines

- Field forms and field books should be backed up after each day's field work, either by scanning to PDF and storing on a laptop or external drive (hard drive, thumb drive, or DVD), OR making a photocopy, OR taking pictures with digital camera and storing the images on a laptop or external drive.
- If equipment isn't available for backup, then a new field book should be used each day, or new loose leaf field book pages in a binder. Do not take used field books into the field if they haven't been backed up.
- Each field book should have the following information on the front cover: Study, consultant, date range.
- Each field book page should have a header of waypoint name, streamcode (if known), date, crew (if first page for the day), and page #.
- Each field form page should have a header of study name, waypoint name, streamcode (if known), date, and page # of #. The crew should be recorded on the first form of each site/date.
- Once the river miles and site identifiers have been identified for the project, these may be recorded in addition to or instead of waypoints.
- Photo descriptions can be included in field notes, and then entered into the photo catalog later, so that anyone looking at a photo knows what they are looking at.

Raw Data Delivery

- Raw data should be delivered on the first day of each month for all field events occurring in the previous 30 days. Special considerations for delivery schedules and requirements can be worked out for each study if needed.
- The table below lists general raw data deliverable requirements:

Data Source	QC Level	Delivery Schedule	Delivery Format
Field book scans	QC1	First day of each month.	.PDF
Field form scans	QC1	First day of each month.	.PDF
GPS dumps	QC1 – raw dump, no data cleanup	First day of each month.	.TXT
Lab reports	QC1 – as received from lab	First day of each month.	.PDF
Mobile data collector (ArcPad, etc.)	QC1 – raw dump, no cleanup	First day of each month.	.TXT or .CSV
Photos	QC1 – raw dump from camera, before cleanup	First day of each month.	.JPG
Telemetry dumps	QC1 – raw dump, no cleanup	First day of each month.	.TXT or .CSV
Thermistor dumps	QC1 – raw dump, no	First day of each month.	.TXT or .CSV

	cleanup		

- Photos should be accompanied by photo catalogs to enable users to find applicable photos as needed in the future.
- Raw video files may be submitted to Alaska DNR for storage at UAF GINA.
- Data submittals can be posted to the AEA SharePoint site, Library "SUWADATA", folder "2013 Field Data Deliverables", in the appropriate folder for the study. Upon posting, a **Letter of Transmittal** (Appendix B) should be emailed to the data managers listed on the Letter template to notify them of the delivery, so they may maintain a catalog of all deliveries for AEA.
- Upload times to AEA SharePoint have been tested; expect a 10 MB file to upload in less than 2 minutes, and a 30 MB file to upload in 4 minutes. If an upload exceeds 100 MB, please notify AEA IT Dept. (Sara Nogg) before posting to plan transmission and storage space.
- Once raw data have been archived, external hard drives may be returned upon request.

Final Data Delivery

- Data collected in the field will be processed and submitted to AEA, constituting final data delivery. Delivery schedules and final data format for each study will be agreed on by AEA, the consultant Data Coordinator, and the project data resource manager. Tabular data may be MS Excel or Access relational format, or a GIS database.
- Processed data should follow the Susitna QC protocol (refer to "Appendix A: Data QC Protocol"). All raw data intended for the Susitna project relational database must be processed: equipment dumps are not intended for database imports.
- Photos selected for final delivery should be delivered with a catalog providing further details on specific location, date, etc. The catalog can be an MS Excel or MS Access table.
- Final video submittals should be sent to Sara Nogg at AEA and Courtney Smith at ADNR. They will ultimately be stored at UAF GINA for user and AEA access.
- The table below lists final data deliverable requirements:

Data Source	QC Level	Delivery Schedule	Delivery Format
DIDSON data	QC1	Study due date	
Field tabular data	QC3 – loaded from field forms and equipment dumps, processed, cleaned up, senior review	Study due date	.XLS or .MDB
Lab tabular data	QC3 – loaded from lab format, standardized, senior review	Study due date	.XLS or .MDB
Modeling data	QC3 – data used to feed into a modeling application	Study due date	.XLS or .MDB
Photos	QC3 – renamed if desired, bad photos removed	Study due date	.JPG
Photo Catalog	QC3	Study due date	.XLS or .MDB
Videography	QC3 – processed and compressed	Study due date	contact UAF GINA

			manager Dayne Broderson & ADNR Courtney Smith

- All deliverables should be accompanied by a transmittal letter (Appendix B).
- Once data files are delivered to AEA, they should be archived at the consultant's office for at least 2 years.

(significant changes since last version are in green)

B. Data Attributes and Databases

Data Attributes

Standards are being established for the Susitna project for some data attributes, whether stored on field forms, MS Excel sheets, database tables, etc. These standards should be considered as much as is practical.

Attribute Naming Standards

These naming standards were previously listed in the SharePoint document "SuWa - Field Data Standards - Attributes DES20120511". That document has been retired and the applicable content moved here.

1. Tables and attributes may be given descriptive names up to 30 characters long and start with a letter.
2. Most attributes also need a 10-character abbreviated name to make datasets compatible with GIS shapefiles. Capitalize the first letter of each abbreviation for readability.
3. Measurement units should be included in the field name as a suffix.
4. Order values may be included in field names, to put attributes or records in a certain order. e.g. FloatTime1, FloatTime2. Some of these may be normalized to 1:M tables.
5. Attributes that contain "Lookup Codes" should be suffixed with "Cd" to help users understand that the values are short codes, and refer to a Lookup table.
6. (more detailed guidelines for naming are found below for specific subjects)

A list of data domains is provided as an appendix to this document. Contact the project data resource manager to get the most up-to-date list and to make revisions or additions.

Attribute Naming - Names Not Allowed

Too Generic

These field names are not allowed as standalone and need clarification within the name, usually with a subject prefix or initials. Some of these are also reserved words in database software, so mustn't be used alone.

Too Generic	Better Example
Class	AqHabClass
Code	FishSpecCd
Comment	FishCtCom
Date	RTTrackDat
Desc, Description, Note	TurbidDesc
End	TransectED
File	GPSFile
ID	RTTrackID
Name	SiteName

Parameter	LabParam, Analyte
Sample	SampleID
Start	TransectST
Temp	WaterTempC
Time	FloatTime1
Type	RosgenType
Unit	AqHabUnit
UOM	AnalyteUM

Database Reserved Words

Some words have special meaning within database engine software; some of these "reserved words" should be avoided as full names for attributes. For example, DATE and COUNT are database function names, so are disallowed as attribute names unless they are qualified with descriptors, such as SurvDate or FishCount.

AEA currently uses MS Access 2010 and Alaska Department of Natural Resources uses Oracle, so reserved words for these platforms should be considered in attribute naming. Some reserved words are found in the generic names list, but others to avoid include:
Current, Float, Group, Index, Key, Label, Limit, Memo, Nested, Note, Range, Recover, Report, Reset, Resource, Return, Set, Size, Table, Text, User, Value, Year, Zone.
Complete lists of reserved words can be found on Microsoft and Oracle websites, but those listed above seemed the most likely to be encountered in the Susitna project.

Attribute Data Values

Case

- Values may be upper or lower case or a mixture, for readability and reporting.
- Case should be applied consistently within a field.
- Some data systems can accommodate case sensitivity while others can't, so values should be assumed to be equivalent for upper and lower case. For example, a units code of M or m represents meters.
- Coded values should be upper case; this helps identify them as codes from lookup tables.

Comment, Note

- Field names for comments and notes should be named to reflect the entity, as it helps clarify data entry from field forms where multiple comments are recorded. Example: site comment and method comment may be recorded on the same field form, so these fields should be named differently.
- If a comment field is being used for a single attribute, then it should be named accordingly. Eg: Fish Count Comment (FshCntComm).

(significant changes since last version are in green)

Coordinates

- All coordinates must be WGS84 and in units decimal degrees NNN.NNNNN (5 – 6 decimals).
- Degree decimal minutes dumped from GPS are not allowed in final data. Consultants will convert coordinates before delivery.
- Coordinates should be Text data type, to help preserve appropriate decimal precision.

Dates and Times

- All dates are Text data type, format YYYYMMDD. (The DateTime type is problematic in GIS, so is not used.)
- Times should be stored in separate attributes from dates.
- Times are Text data type, 24-hour time
    - Time of Day format = HH:MM or HH:MM:SS, specified in the data dictionary.
    - Duration Time format = HH:MM or HH:SS, specified in the data dictionary
- If a time is for duration, try to reflect that in the attribute name with "Dur".
- Consider using a units field for durations, which can read as HH:MM or MM:SS.
- Field names should reflect the entity, so they are easily distinguished from other dates and times in reports and query output. For example: fish wheel dates might be FWLogDate and FWCatchDat.
- A time zone qualifier must be included in any tables that have time-of-day attributes. Use codes:
    - **AST** = Alaska Standard Time
    - **ADT** = Alaska Daylight Time.

Derived and Calculated Fields

- Data tables may contain calculated and derived fields. The formula must be provided in the data dictionary and list any other fieldnames used in the calculation.
- Calculated fields must be named to show their status, using a "Calc" as a name suffix, such as AvgWidCalc.
- At this point, the MS Access 2010 data type of Calculated is not used for the Susitna project.

Downstream / Upstream Orientation

- Whereas some disciplines may normally orientate as "looking upstream", the Susitna project has chosen a downstream orientation for all applications with deliverables to AEA.
- Any attributes that are specific to a left bank (LB) or right bank (RB) feature should be orientated as "looking downstream".

Location / Site Identifiers

- A linear route layer has been developed for the Susitna River mainstem for the current project. River miles along this route are name "PRM" (project river mile). Some studies and historic data may include "HRM" (historic river mile), calculated in the 1980s studies. When HRM is present, the historic source should be noted in the data dictionary and

possibly a field in a site table. A cross-reference table of PRM and HRM may be created by the GIS team.

- As of this document version, streamcodes and project river miles have been generated only for the Susitna River mainstem main channel and certain river features. Off channel and tributary sites are making use of lat/long for location identifiers, but naming conventions for them are being considered.
- Location names must be meaningful, and at least include a project river mile (PRM) if available, verified on the current linear reference. (A site name domain is being generated for some fish studies.)
- No cryptic site codes. Codes used in the field must be converted to site names in the GIS site domain before submittal.
- The following verbiage from the project implementation plans explains the use of River Miles in the project and applies to data as well:

  The Project River Mile (PRM) system for the Susitna River was developed to provide a consistent and accurate method of referencing features along the Susitna River. During the 1980s, researchers often referenced features by river mile without identifying the source map or reference system. If a feature is described by river mile (RM) or historic river mile (HRM), then the exact location of that feature has not been verified. The use of PRMs provides a common reference system and ensures that the location of the feature can be verified. The PRM was constructed by digitizing the wetted width centerline of the main channel from 2011 Matanuska-Susitna Borough digital orthophotos. Project River Mile 0.0 was established as mean low water of the Susitna River confluence at Cook Inlet. A centerline corresponding to the channel thalweg was digitized upstream to the river source at Susitna Glacier using data collected as part of the 2012 flow routing transect measurements. The resultant line is an ArcGIS route feature class in which linear referencing tools may be applied. The use of RM or HRM will continue when citing a 1980s study or where the location of the feature has not been verified. Features identified by PRM are associated with an ArcGIS data layer and process, and signifies that the location has been verified and reproduced.

Measurements: Numeric, Estimates, and Descriptive

- Attributes of a numeric nature should be NUMBER data type and cannot contain characters.
- Number fields are typically measurements such as count, width, velocity, etc. However, some measurement results require alphanumeric values, which can be accommodated in various ways.
- If estimated measurements must be stored, they go into the numeric field, with a TEXT flag to describe the nature of the estimate, such as EstFlag.

  Example:

  Count values that are <u>not</u> allowed: "~10", ">20", "many", "5-10"

  Use the following instead:

  <u>FishCount</u> <u>CntEstFlag</u>

10		this means exactly 10
10	~	this means about 10
20	>	this means >20

- If counts of "5-10" and "many" need to be allowed for some reason, we can employ a count description (CountDesc) field,TEXT datatype.
- Other descriptive measurements, such as some Turbidity, use a TEXT field named with "Desc", such as TurbidDesc. The domain for a field like this should be defined and enforced to allow for reporting.
- Queries and reports may need to include EstFlags and Desc fields, if they exist. Users need to know how to deal with measurements like this, so they should be documented in the dictionary.
- Use caution that the default value for numeric fields isn't set to zero (0). This will be checked during QC4 verification.

Measurements Units (UM)

- The Susitna project prefers that Units be included in field names where practical. However, some attributes may need units stored in a separate units of measurement (UM) field.
- Some attributes use varying units based on discipline, or the units can't be denoted within a 10-character field name. These will need a separate UM field. Examples may include:
  WetWid and WetWidUM
  RelatCond and RelCondUM
  SpecCond and SpecCondUM
- Some parameters will have standard measurement units for the project. These can be identified when reviewing field forms, but at least include:

water temperature:	degrees C
fish distribution:	metric units
Instream Flow (IFS):	English units
Habitat Suitability Criteria (HSC):	English units

- Unit values should never include special characters, as the Unicode character set could be misinterpreted during data imports and exports. For example, the Unicode symbol for micron "µ" should be represented with an ASCII "u".

Person / Staff Names

- Use first initial and last name (FLastname), such as DStewart.
- Avoid using a person's initials in the final data, to avoid an additional lookup and confusion of acronyms.
- Exception: Authors in the Bibliographic Database are Last, First M.

Special characters and symbols

- ASCII special characters are allowed within values. These are common in:
  long text fields like Comments

streamcodes with periods (SU 1.120.10)
multiple values separated by commas or semicolon (WeatherDes = wind, light rain)

- Values should never contain Unicode symbols, only ASCII characters.

Waypoint names

- Waypoints may typically be assigned sequential numbers within a GPS unit. More descriptive names may also be used.
- Waypoints don't need to be renamed in the project database, as they should always be accompanied by the GPS unit ID and GPS date which will create a unique waypoint list.

Relational Databases

If MS Access databases will be delivered as part of the final data deliveries, the following guidelines should be used.

Database Object Names

The Leszynski (Hungarian) naming convention is commonly used by MS Access developers and is adopted for the Susitna project, with some minor customization. Note that this convention isn't enforced by MS Access; it is implemented by the database administrator for easier maintenance and programming in Visual Basic for Access (VBA), where reference to an object name may not indicate its data type.

	Attributes (no prefix)
tbl	Table: data
tlu	Table: lookup, valid value, code
tmp	Table: temporary, can be deleted without adverse effect
qry	Query, view
(The next ones aren't typically delivered with a database by consultants.)	
frm	Form
rpt	Report
mcr	Macro
mod	Module

Other naming rules:

- Table names are restricted to a 30-character maximum, as required to meet GIS standards for this project.
- Attribute names are restricted to a 10-character maximum to accommodate GIS shapefile users.
- Table and attribute names can't start with a number, per project GIS standards.
- Attribute names must start with a capital letter.
- Contain only letters and numbers.

- Underscores may be allowed if necessary, but no spaces.
- Symbol fonts are never allowed in names.
- Name using Pascal case (camel case with the first letter capitalized). This is a mix of upper and lower case, where each new element of the name is capital, and is encouraged for readability.

The naming convention may be re-addressed if the database is later moved to another platform with case sensitivity issues between Oracle, MS Access, and SQL Server.

Attribute Data Types

The following field data types will be utilized in the Susitna database and are permitted in deliverables:

Boolean (True/False, Yes/No)
Hyperlink
Number
Text (make sure zero-length string properties are disabled in MS Access)

Data types that aren't permitted at this time in deliverables:

Attachment (OLE, BLOB)
AutoNumber (change to Text or LongInt for delivery)
Calculated (MS Access 2010 data type)
DateTime (dates and times must be Text)
Memo
Multi-valued (MS Access accdb format)

A naming convention for attributes to show the data type won't be implemented for the Susitna project, as we need to accommodate the shapefile attribute name limit of 10 characters. For example, we won't use prefix "int" for integer type attributes.

Unique Record Identifiers (Primary Keys)

- A logical / natural primary key must be identified for each dataset, whether MS Access table or MS Excel data sheet.
- If a synthetic / surrogate key is also desired, or in some situations required, then the key name must be descriptive; the name "ID" alone (a default name created by MS Access) is not allowed. Refer to the Susitna project Data Naming Conventions for descriptors.
- Surrogate keys may be text, numeric, or MS Access AutoNumber data types. Text keys should be upper case for portability to another platform.
- If the key contains information, it should be noted in the data dictionary so users can interpret it correctly. For example, SurveyID is year + study method + sequential number (2012RTTAG2).

C. Data Dictionary

The Program Lead team is tasked with compiling a comprehensive data dictionary document for all water resources studies. Ideally, a data dictionary utility with reporting capabilities will be employed, although this has not been decided yet. This may provide a more detailed and descriptive document than the GIS metadata, which is needed to meet GIS project standards.

For the Susitna project, we make a distinction between the terms "metadata" (refers to the GIS) and "data dictionary" (refers to the relational database). The metadata has standards that the GIS team and ADNR establish and enforce for the GIS. The relational database will be documented differently from the GIS, and its template doesn't resemble GIS metadata.

- (This item is in progress and will be updated.)
- When field data is submitted to the Program Lead team for level QC4, it should be accompanied by a data dictionary. This will provide a detailed, descriptive document to compliment the GIS metadata project standards.
- The dictionary will be reviewed for table naming and descriptions, identification of keys, field names, data types, and descriptions.
- Descriptions should not typically be terse, but rather detailed with an eye to being useful to scientists years later and without access to current scientists for explanation. Special handling of anomalies within tables or fields should also be described.
- The format for data descriptions can be MS Excel or MS Word until further notice. Storing field descriptions within MS Access table designs won't fulfill the dictionary requirements.

(significant changes since last version are in green)

Appendix A: Data QC Protocol

Introduction

The F&A Program Lead team is tasked with implementing a standardized QA/QC protocol, intended for use in all environmental field studies in 2012, including fish and aquatic, water quality, river ice, IFS, and others. This document will be presented to the leader and appointed Data Coordinator of each of these study teams.

Members of the Program Lead team can be contacted with questions and comments:

Dana Stewart – Data Resource Management
Judy Simon – Program Coordination
Joetta Zablotney – GIS-related QC

QC Levels

There will be 5 levels of data QC, named QC1 to QC5, each of which is tracked within the data. This allows for quick determination of the QC status of every data record. The first three levels are to be completed by the study team, the fourth level by the Program Lead team, and the final level by senior professionals during analysis and reporting.

QC1 – Field Review: QC review performed by the person collecting field data, whether recorded on paper field forms or directly into electronic data collection tools, and then by the field team leader. This is also the QC level of raw data collected via field equipment such as thermistors, cameras, GPS units, etc.
The goals of QC1 are to identify errors and omissions and correct them under similar field conditions prior to leaving the field, and to backup files in the field.
Review is done on 100% of data and includes completeness, legibility, codes, and logic on all information recorded. This is typically completed in the field daily. Once completed, QC1 notations are made directly on the field form in an entry named "QC1", containing the date and responsible staff and formatted as "YYYYMMDD FLastname" (example: "20120631 JDoe").

QC2 – Data Entry: Data from paper forms are entered into an electronic format, then data entry is verified by a second party against the field forms.
The goal of QC2 is to verify correct, complete, and consistent data entry.
Verification is done on 100% of data entered and includes extrapolation of shorthand codes that might be used in the field into longhand or standard codes during data entry. Data entry errors are corrected at this time, then QC is recorded in a column named "QC2", containing the date and responsible staff and formatted as "YYYYMMDD FLastname" (example: "20120631 JDoe").

QC3 – Senior Review: Data are reviewed by a senior professional scientist on the consultant team, checking for logic, soundness, and adding qualifiers to results if warranted.

Calculated results can also be added at this time (formulas must be documented in the data dictionary). Photo locations should be verified. This is the final review before submitting field data to the Program Lead, and is recorded in the "QC3" column in the same format as QC2. This is also the QC level of raw files that have been "cleaned up" or otherwise processed for delivery to AEA, such as photos.

QC4 – Database Validation: Electronic data files are submitted to and verified by the Program Lead's data resources manager. The deadline for this delivery is negotiated with the team Data Coordinator in consideration of the study due date.

Data are verified for completeness, project standards (codes, field name conventions, date formats, units, etc.), calculated and derived fields, QC fields, etc. The data files are incorporated into the project database schema, splitting into normalized tables as necessary and all primary and foreign keys checked. An error report is generated for the study consultant, who is expected to make corrections and resubmit data. The process is repeated until verification is clean and records are marked in column "QC4" (such as "20121001 DStewart").

QC5 – Technical Review: Data revision and qualification may be applied by senior professionals when analyzing data for reports, trends, and FERC applications. Data calculations may be stored with the data. Some data items may get corrected or qualified within the database, while others are only addressed in report text. QC5 may be iterative, as data are analyzed in multiple years.

If a data item is revised directly, it's recorded in 2 columns, QC5 (date and staff) and QC5Edit (what is revised and why). This will serve as adequate documentation of the revisions, so maintenance of additional documentation isn't usually necessary. QC5 revisions will be physically made by the Data Resource Manager, directed by the senior professional.

Data Collection Devices (e.g. ArcPad, Trimble)

Field forms should be reviewed and approved by the Program Lead team before use in the field. If mobile data devices (ArcPad and Trimble) are used to record field data directly, they must be accompanied by backup paper field forms in case of equipment failure, and both the paper forms and device entry screens should be approved by the Program Lead team.

Both paper and electronic field forms should be backed up nightly in the field by scanning and downloading to a storage unit or photocopy to paper.

Data Revisions

Once the processed field data (QC3) have been submitted by a consultant to AEA via R2, and it has been validated as ready for incorporation into the Susitna project database (QC4), the data are considered to reside with AEA, and subsequent revisions will only be made by the Program Lead team on their behalf. If a study team discovers that data require revisions, their Data Coordinator

can send a formal, written request (i.e. email) to the Data Resource Manager. Revisions will be made and the appropriate QC columns updated, which will serve as adequate documentation.

Appendix B: Letter of Transmittal

(next page)

(significant changes since last version are in green)

SUSITNA-WATANA HYDROELECTRIC PROJECT

LETTER OF TRANSMITTAL

To: ☐ Dana Stewart, DESIT
☐ Judy Simon, R2
☐ Sara Nogg, AEA
☐
☐
☐

Date:
Project:
Subject:

Transmitted via ☐ AEA SharePoint ☐ DVD ☐ Thumb drive ☐ External hard drive
☐ Other_____

are the following files: **Please specify file names and folder/file paths and include a brief description

As: ☐ Raw / QC1 ☐ Final/ QC3 ☐ Other _____

Remarks:

Please notify us if the enclosures are not received.

Submitted by:

Name:

Company

cc:

Appendix: Data Domains

(next page)

Appendix - Data Domains and Codes

Aquatic Habitat - Channel Location (ChanLocCd)

Code	Description
BB	bank-to-bank
LB	left bank, looking downstream
MID	mid-channel
RB	right bank, looking downstream

Aquatic Habitat - Fish Cover Type (CoverCd)

Code	Description
AqVeg	aquatic vegetation
BO	boulder
DP	water depth > 1 meter
LWD	large woody/organic debris
None	no cover
OV	overhanging vegetation
SWD	small woody/organic debris
UCB	undercut bank

Riparian Habitat (RipVegCd)

Code	Description
BFC	Broad Leaf Forest – Closed
BFO	Broad Leaf Forest – Open
CFC	Conifer Forest – Closed
CFO	Conifer Forest – Open
NHB	Nonforest-Herbaceous – Bog
NHE	Nonforest-Herbaceous – Estuarine
NHF	Nonforest-Herbaceous – Fen
NHO	Nonforest-Herbaceous – Other
NSA	Nonforest Shrub – Alder
NSO	Nonforest Shrub – other
NSW	Nonforest Shrub – Willow

Fish Behavior (FBehavCd)

Code	Description
MG	migrating
ML	milling
PS	post-spawning
SP	spawning

Fish Disposition Code (FishDispCd)

Code	Description
CHY	Chytrid samples taken (AK FRP)
ELA	elastomer marked (AK FRP)
FCAD	fin-clipped, adipose (AK FRP)
FCPT	fin-clipped, pectoral (AK FRP)
FCPV	fin-clipped, pelvic (AK FRP)
FHP	fin hole-punched (AK FRP)
GENP	genetic samples taken, non-lethal
GENW	genetic samples taken, lethal
IDRE	identified and released (AK FRP)
MERE	measured and released (AK FRP)
MORT	unintended mortality (AK FRP)
NAP	not applicable
PTG	pit-tagged (AK FRP)
RTG	radio-tagged, unspecified
RTGE	radio-tagged, esophageal (AK FRP)
RTGS	radio-tagged, surgical (AK FRP)
SCA	scale samples taken
SDSA	sacrificed and discarded sanitarily (AK FRP)
SDSI	sacrificed and discarded at site (AK FRP)
STG	spaghetti-tagged
TLA	transported live to aquarium (AK FRP)
VOG	visual observation, ground
VSID	voucher specimen, identification
VSMD	voucher specimen, metals, dissected
VSMW	voucher specimen, metals, whole fish

Fish Capture/Observation Method (FishMethCd)

Code	Description
ANG	angling
BEF	boat-mounted electrofisher
DIP	dip net
FWL	fishwheel
FYK	fyke net
GNF	gill net, floating
GNS	gill net, sinking/set
HON	hoop net
HOT	hoop trap
IPT	inclined plane trap
MIN	minnow trap, unbaited
MINB	minnow trap, baited
NON	no fish sampling occurred
OTH	other
PEF	backpack electrofisher
SCT	screw trap
SDD	sonar, DIDSON
SEN	seine net
SNK	snorkel
STL	set line, unbaited
STLB	set line, baited
TAN	tangle net
TRL	trotline, unbaited
TRLB	trotline, baited
USC	underwater still camera
UVC	underwater video camera, unbaited
UVCB	underwater video camera, baited
VOB	visual observation, boat
VOF	visual observation, fixed-wing aircraft
VOG	visual observation, ground
VOH	visual observation, helicopter
VOT	visual observation, tower
WER	weir

Fish Biosample Type (FBioSampCd)

Code	Description
GEAP	genetic sample, axillary process
GECF	genetic sample, caudal fin-clip
GEPV	genetic sample, pelvic fin-clip
GEWB	genetic sample, whole fish
ISFC	isotope sample, caudal fin-clip
MEFL	metal/mercury sample, filleted
MEWB	metal/mercury sample, whole fish
OTOL	otolith sample(s) taken
SCAL	scale sample(s) taken
STCN	stomach contents sample

Fish Length Method (FLenMethCd)

Code	Description
FL	fork length
MEF	mideye to fork (same as METF)
METF	mideye to tail fork (same as MEF)
SL	standard length
TL	total length

Fish Aging Method (FAgeMethCd)

Code	Description
OTO	otolith
SCA	scale

Appendix - Data Domains and Codes

VSOT	voucher specimen, otoliths

Fish Species (FishSpecCd)

Code	Description
CDV	Dolly Varden
CHR	char, undifferentiated
CLK	lake trout
DAL	Alaska blackfish
GBR	burbot
GRA	Arctic grayling
HER	herrings, undifferentiated
ISA	salmonid
KNS	ninespine stickleback
KSB	stickleback, undifferentiated
KTS	threespine Stickleback
LAR	Arctic lamprey
LMP	lamprey, undifferentiated
LPC	Pacific lamprey
NOS	longnose sucker
OEU	eulachon
OPS	pond smelt
OSM	smelt, undifferentiated
PIK	northern Pike
QQQ	other species not listed
SAM	Pacific salmon, undifferentiated
SCH	chum salmon
SCK	Chinook salmon
SCO	coho salmon
SPI	pink salmon
SSE	sockeye salmon
TRB	rainbow trout
TRT	trout, undifferentiated
UCR	coastrange sculpin
ULP	sculpin, undifferentiated
USL	slimy sculpin
VVV	no collection effort
WBC	Bering cisco
WHB	humpback whitefish
WHF	whitefish, undifferentiated
WRN	round whitefish
XXX	no fish collected or observed
ZZZ	general fish observation, no species info.

Fish Lifestage (FLifeStgCd)

Code	Description
EGG	eggs
FXA	alevin
FRY	fry
PAR	parr
SMT	smolt
JUV	juvenile
JOA	juvenile/adult
JAC	jack
ADT	adult
CAR	carcass
NAP	not applicable
NRD	not recorded

Fish Tag Insertion Location (FTagInsCd)

Code	Description
ABD	abdominal cavity
DOR	dorsal musculature or cavity
GIL	posterior to gill pore

Fish Survey Method (SurvMethCd)

Code
Boat
Foot
Heli

Sex (SexCd)

Code	Description
F	female
M	male
U	unknown

Appendix - Data Domains and Codes

Style Guide: Studies (StudyCd)

Code	Description
AQHAB	Characterization and Mapping of Aquatic Habitats
AQTRANS	Aquatic Resources Study within the Access Alignment, Transmission Alignment, and Construction Area
BARR	Study of Fish Passage Barriers in the Middle and Upper Susitna River and Susitna Tributaries
CIBW	Cook Inlet Beluga Whale Study
ESCAPE	Salmon Escapement Study
EUL	Eulachon Run Timing, Distribution, and Spawning in the Susitna River
FDAML	Study of Fish Distribution and Abundance in the Middle and Lower Susitna River
FDAUP	Study of Fish Distribution and Abundance in the Upper Susitna River
FHARV	Analysis of Fish Harvest in and Downstream of the Susitna-Watana Hydroelectric Project Area
GENE	Genetic Baseline Study for Selected Fish Species
GW	Groundwater Study
ICE	Ice Processes in the Susitna River Study
IFS	Fish and Aquatics Instream Flow Study
MERC	Mercury Assessment and Potential for Bioaccumulation Study
PASS	Study of Fish Passage Feasibility at Watana Dam
RESFSH	The Future Watana Reservoir Fish Community and Risk of Entrainment Study
RIFS	Riparian Instream Flow Study
RIVPRO	River Productivity Study
WQ	Baseline Water Quality Study

APPENDIX 4. DRAFT DATABASE TEMPLATES

Relationships for River Prod ERD
Wednesday, January 16, 2013