UTAH DEPARTMENT of ENVIRONMENTAL QUALITY
WATER QUALITY

Utah Comprehensive Assessment of Stream Ecosystems (UCASE)

Field Operations Manual

Version: 3.0 (implemented July 2021)

West Fork White Rocks River; Uinta Mountains; 2020

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Revision Page

Year Revised	Summary of Changes	Sections	Comments
2015	Abolished the Legacy Tree Form and procedures from the manual.	Previously in Section 5	
2015	Added a component to manual regarding the presence of clams at sites. Added a field on Site Assessment Form to capture whether clams were observed at a site.	6.1.2	This was added to assist with growing concerns of ammonia on clam/snail populations.
2015	Added an acronym, abbreviations, and measurement units page	Pages *(vii, viii)*	
2015	AFDW changed to AFDM	Entire document	
2018	Reformatted front cover. Added newer DWQ logo	Cover page	
2018	New format to track versions	Entire document	
2018	Updated contact information	1.0	
2018	Re-wording	Section 1	Grammar and punctuation update
2018	Re-wording	Section 4	Grammar and punctuation update
2018	Updated bottle requests	Section 5	
2018	Re-wording	Section 5	Grammar and punctuation update
2018	Abolished the qualitative periphyton sampling procedures	Section 5 and 6	
2021	Removed map drawing, updated benthic macroinvertebrates, language and grammar updates.	Section 5 and Whole Document	
2021	Changed versioning structure	NA	

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Acronyms, Abbreviations, and Measurement Units

AFDM	Ash-Free Dry Mass
amps	Amperes
BOD	Biochemical Oxygen Demand
bpj	Best Profession Judgment
cm	Centimeter
CWA	Clean Water Act
dbh	Diameter at Breast Height
DWQ	Division of Water Quality
DC	Direct Current
DO	Dissolved Oxygen
DO%	Percent Dissolved Oxygen Saturation
EMAP	Environmental Mapping and Assessment Program
EPA	Environmental Protection Agency
ft	Foot
gal	Gallon
GPS	Global Positioning System
GRTS	Generalization Random Tessellation Stratified Spatially-Balanced Survey Design
ha	Hectare
HZ	Hertz
ID	Identification
in	Inches
L	Liter
LWD	Large Woody Debris
m	Meter
mm	Millimeter
m^2	Square meters
mg/L	Milligram per liter
mS/cm	Millisiemens per centimeter
NAD	North American Datum
NHD	National Hydrology Database
NRSA	National Rivers and Streams Assessment
PCB	Polychlorinated biphenyl
PFD	Personal Flotation Device
pHab	Physical Habitat
ppm	Parts per million
psi	Pounds per square inch
QA	Quality Assurance
QAPP	Quality Assurance Protection Plan
QC	Quality Control
RBS	Relative Bed Stability
SOP	Standard Operating Procedure
TMDL	Total Maximum Daily Load
UCASE	Utah Comprehensive Assessment of Stream Ecosystems

µm	Micrometer
µS/cm	Microsiemens per centimeter
USGS	United States Geological Survey
V	Volts
VA	Volt-ampere

1 BACKGROUND

This manual describes the field protocols and daily operations for field crews to use in the Division of Water Quality's (DWQ) Utah Comprehensive Assessment of Stream Ecosystems (UCASE) sampling program. UCASE is a monitoring program with both probabilistic and targeted components designed to meet the following objectives:

- Assess the biological, physical, and chemical conditions of Utah's wadeable and partially wadeable streams
- Target pollution/contamination sources to Utah's streams and identify water quality impairments
- Establish a baseline to compare future stream surveys for trend assessments
- Assess the effectiveness and changes in stream habitats after restoration projects (i.e. Non-point Source (NPS) projects) in order to determine project effectiveness
- Establish reference site criteria

The majority of content in this manual is derived and structured from the 2009 Environmental Protection Agency (EPA) manual for the National Rivers and Stream Assessment (NRSA) protocol (USEPA. 2007. National Rivers and Streams Assessment: Field Operations Manual. EPA-841-B-07-009. U.S. Environmental Protection Agency, Washington D.C.). Procedures, objectives, and goals used for NRSA are very similar to the ones used for the UCASE program. The NRSA and UCASE protocols differ slightly from time to time (i.e. collection methods; processing methods; overall workflow for completing sites). However, in general, the UCASE protocol is designed to be as consistent with the NRSA protocol as possible.

This document is intended to be used as a guide for crews conducting UCASE methods while in the field. This manual briefly mentions some background information about the UCASE program, however, it does not go into great detail on specifics regarding the overall program design, approach, and analysis. For more information and specifics on the programmatic overview and design of the UCASE program, please refer to the Utah Strategic Monitoring Plan.

A variety of tools and equipment are used to make measurements for UCASE surveys. These often require training and have reference manuals from the manufacturer associated with them. These manuals will not be found in this document. Contact the Field Program Coordinator to find manuals from specific equipment manufacturers/vendors. Or, find the manufacturer on the internet and obtain the manual from there.

Supplemental information relating to the field component of UCASE can often be found on the DWQ network by following this folder pathway:
U:\PERMITS\MONITORS\UCASE

The following DWQ personnel can be contacted for more information relating to this document. These personnel will be referenced often throughout this manual.

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1.1 Survey Design

Utah's biological beneficial uses require the protection of fish (e.g., cold or warm-water species) and the organisms upon which they depend. In the past, DWQ has assessed these beneficial uses via water chemistry sampling and associated water quality standards that are assumed to protect aquatic organisms. However, DWQ developed a RIVPACS (River Invertebrate Prediction and Classification System) model that directly assesses attainment of biological beneficial uses by quantifying the "health" of macroinvertebrate assemblages. Measuring biological communities directly has the advantage that it integrates the combined effects of all pollutants. This allows for a direct examination of how interacting pollutants affect the condition of a stream ecosystem. Moreover, because aquatic macroinvertebrates spend the majority of their life in aqueous environments, they are capable of integrating the effects of stressors over time providing a measure of past, transient conditions.

An important component of the biological monitoring program is the identification and incorporation of reference sites, which serve as controls, or benchmarks, that are used to establish the chemical, physical, or biological condition expected in the absence of human disturbance. Ideally, all reference sites would be in near-pristine condition. However, few pristine sites remain in Utah, especially among streams in valleys, which have experienced a long history of human occupation. As a result, systematic protocols for quantifying the relative extent of human-caused alteration at both reach-and watershed-level spatial scales are being developed to help DWQ objectively identify suitable reference sites throughout Utah.

1.1.1 Target Population and Sample Frame

Target populations for UCASE sites consist of all streams that are at least 50% wadeable (1st through 5th order streams) and that have flowing water at some time during the sampling period, which is May 1st through October 31st.

Representative means sampling a site during "normal" flow regimes under conditions that represent the usual characteristics of the stream type during the sampling timeframe. Since Utah has several eco-regions, varying weather patterns, and varying stream types, certain sites can be sampled before or after the preferred dates. Depending on the location of particular sites, sampling periods can vary (i.e. a low-flowing ephemeral desert stream could be sampled in April or May whereas a high mountain stream could be targeted in July or August). Crews should not sample sites during spring runoff or after a major flash flood. Of note, it is especially important to consider the timing of monsoon events in certain regions of Utah. However, if a flash flood does occur before or while sampling, the crew should wait at least two weeks from the last major event before returning to sample. Determining whether sites can be sampled before or after the preferred sampling dates should be decided by the Field Program Coordinator prior to going out in the field (pre-season planning) using best professional judgment. All other sites can be sampled at any time during the season using best professional judgment as long as representative conditions exist for that site.

Historically, DWQ's biological program was largely based on a targeted approach. More recently, probabilistic surveys have been the preferred method for selecting sites for the UCASE program. More on this can be found in the Utah Strategic Monitoring Plan.

1.1.2 Probabilistic Sites

As in other probability-based population surveys, waterbody assessments based on randomly selected sites allow states to make statistical inferences as to the condition of all waters based on the conditions of those randomly selected and assessed. Probabilistic sites will help describe the status of waterbodies and help guide targeted monitoring efforts in specific basins to define impairments and pollution sources. Sampling criteria and scope is usually planned well prior to the field sampling season by the Biological Program Coordinator, Field Program Coordinator, and the Monitoring Section Manager. Depending on data needs, annual probabilistic sampling can range anywhere from 0-50 sites, typically. Historically, probabilistic sampling was based on a rotating basin sampling design. More on the rotating basin schedule and structure can be found in the DWQ Monitoring Plan. The current approach is based on a statewide design made up of targeted re-visit sites and probabilistic basin sites in conjunction with the NRSA program coursed over multiple field seasons.

1.1.3 Targeted Sites

UCASE sampling methods are also used for targeted sites where biological data are requested on a site specific basis. These sites are usually requested by cooperators (i.e. BLM, USFS, DWR, etc.) or DWQ personnel to determine pollution source identification; Total Maximum Daily Load (TMDL) analysis and development; pre and post restoration project success; 3A assessment purposes; 319 program effectiveness; establish reference sites; develop reference sites and criteria. Historic probabilistic sites may become targeted sites during future sampling cycles depending on data needs. These sites are usually pre-determined by data users/requesters where randomness is not a factor. The number of targeted sites sampled throughout a sampling season is unlimited as long as the monitoring section has the resources to perform the sampling. Site requests and site feasibility is determined by the Monitoring Section manager and the Biological Program Coordinator.

1.2 Selection of UCASE Sites

1st through 5th order streams are the target population for UCASE sampling, which is assumed will cover most wadeable waterways. Sites must be 50%-100% wadeable in order to sample. If sites are less than 50% wadeable they should be dropped from the sampling list or sampled later in the season when it can be sampled more adequately. Sites used for probabilistic sampling are generated by the Biological Program Coordinator using the Generalized Random Tessellation Stratified Spatially-Balanced Survey Design (GRTS). Detailed information on design information related to probabilistic surveys can be found by going this EPA website: https://archive.epa.gov/nheerl/arm/web/html/design_intro.html

When a UCASE site list is developed at any given time, a series of metadata is associated with every site (e.g. coordinates, site codes, stream names, stream order, county, weight value, etc.). There is a column that designates sites as “base” sites and “oversample” sites. Base sites are the first set of sites that should be sampled to meet the survey design objectives. If these sites cannot be sampled for any reason (e.g. safety concerns, non-target, dry, access permission denied by landowner, etc.) they will be replaced by oversample sites. There is usually some kind of unique methodology for selecting oversample sites depending on the scope of the project. This methodology should be organized by the Biological Program Coordinator and the Field Logistics Coordinator.

1.3 Selection of UCASE Indicators

In an effort to keep biological sampling efforts consistent with other projects, both state and nation-wide, DWQ has chosen to adopt similar indicators and methods used in the Enviornmental Monitoring and Assessment Program (EMAP) and NRSA program. Although UCASE procedures are similar to these national programs, some collection methods, logistics, lab methods, and labs used for analysis differ.

Indicators used for UCASE not only compare with national methods, but have also been designed to meet Utah-specific sampling objectives developed by DWQ. Biological

indicators sampled in this protocol help develop direct measures for aquatic life uses. For detailed information on biological indicators used for UCASE, see Utah's Strategic Monitoring Plan. The remainder of this section briefly describes the indicators that will be used for UCASE to assess water quality, ecological integrity, recreational value, and site characteristics.

1.4 Description of UCASE Indicators

Multi-Parameter Water Quality Measurements

Measurements for temperature, pH, dissolved oxygen (DO), and conductivity are taken with a calibrated water quality meter or multi-probe sonde within the designated sampling reach in the stream. This information is used to detect extremes with stream conditions that might indicate impairment.

Water Chemistry and Associated Measurements

Water chemistry measurements are used to determine concentrations of a variety of components in any given stream. Common analytes include dissolved and suspended solids, nutrient enrichment, metals loading, and general classification of water chemistry type.

Chlorophyll-a

Chlorophyll-*a* analysis helps determine the structure of photosynthetic organisms in a stream. It is the pigment that makes plants and algae green. Chlorophyll-*a* concentration measurement is used as a surrogate to determine algal biomass in the water. Chlorophyll is collected from both the water column and the periphyton sample.

Periphyton Assemblage

Periphyton are diatoms and soft-bodied algae that are attached or otherwise associated with different sorts of channel substrates. They can contribute to the physical stability of inorganic substrate particles and provide habitat and structure. Periphyton are useful indicators of environmental disturbances, including habitat destruction, contamination by nutrients, metals, herbicides, hydrocarbons, and acidification.

Benthic Macroinvertebrate Assemblage

Benthic macroinvertebrates (also referred to as benthos, inverts, macroinverts, or bugs) are bottom-dwelling animals without backbones (invertebrates) that are large enough to be seen with the naked eye (macro). Examples of macroinvertebrates include crayfish, snails, clams, aquatic worms, leeches, and the larval and nymph stages of many insects, including dragonflies, mosquitoes, and mayflies. Populations in the benthic assemblage respond to a wide array of stressors in different ways. Presence or absence of certain species in certain environments serves as a key tool to indicate the stressors that affect benthic macroinvertebrate assemblages. Many macroinvertebrates have relatively long

life cycles of a year or more and are relatively immobile, the structure of the macroinvertebrate assemblage is a response to exposure of present or past conditions.

Fish Assemblage

A fish assemblage component to any biological design is an integral component of many water quality management programs. This indicator will measure specific attributes of the overall structure of the ichthyofaunal community to evaluate biological integrity and water quality.

Fish Tissue

The fish tissue contaminants indicator, which measures bioaccumulation of persistent toxins, is used to estimate risks of fish consumption to humans. Fish from Utah's streams are primarily tested for mercury and selenium. At times fish tissue is analyzed for poly-chlorinated biphenyls (PCBs), but only rarely. Fish tissue samples are not collected at all UCASE sites, but might be collected upon request from members of the Utah Mercury Workgroup (http://www.mercury.utah.gov/).

Physical Habitat Assessment

The physical habitat assessment (pHab) of the sampling reach and the riparian zone (the region lying along the bank) will serve three purposes:

1. Habitat information is essential for the interpretation of what ecological conditions are expected in the absence of many types of anthropogenic impacts.

2. Habitat evaluation is a reproducible, quantified estimate of habitat condition serving as a benchmark against which to compare future habitat changes that might result from anthropogenic activities.

3. Specific aspects of the habitat information collected aid in the diagnosis of probable causes of ecological degradation in streams. For example, some of the data collected is used to calculate relative bed stability (RBS). RBS is an estimate of stream stability that is calculated by comparing the mean sediment size present to the sediment size predicted by channel characteristics and slope.

In addition to information collected in the field by the physical habitat assessment, the physical habitat description of each site includes many map-derived variables such as stream order and drainage area. Furthermore, an array of information, including watershed topography and land use, supplements the physical habitat information. Together with water chemistry, the habitat measurements and observations describe the variety of physical and chemical conditions that are necessary to support biological diversity and foster long-term ecosystem stability.

Site Characteristics

Observations and impressions about the site and its surrounding watershed by field teams are useful for ecological value assessment, development of associations and stressor indicators, and data verification and validation.

Table 1-1. Summary table of indicators for wadeable sites

Indicator	Specs/Location in Sampling Reach
Multi-parameter measurements (pH, DO, temperature, conductivity)	Measurements taken at any point within the designated sampling reach in the thalweg; readings are taken at least 0.3 m depth at the same time, but below where the water chemistry samples are collected.
Water chemistry (total chemistry suite; non-filtered nutrient suite; filtered (dissolved) metals suite; filtered (dissolved) nutrient suite)	Collected from a depth of 0.3 m within the designated reach in the thalweg before any other sampling activities occur upstream.
Chlorophyll-*a*	One sample is collected from composite periphyton samples and the second sample from the water column where water chemistry samples are collected.
Periphyton	Collected systematically from 11 locations at each transect and combined into a single composite sample
Benthic macroinvertebrate assemblage	Collected from 8 targeted riffles (edge habitat if sandy bottoms or no riffles exist) in the sampling reach and combined into a single composite sample
Fish assemblage	Sampled throughout the reach at specified locations.
Physical habitat assessment	Measurements collected throughout the sampling reach at each transect
Fish tissue	Collected anywhere within the reach. If desired number of samples cannot be obtained in reach, crew can shock 1 reach length above and/or below the original reach. Fish tissue samples are only collected at requested sites.
Drainage area	Done at desktop.
Characteristics of watershed	Desktop GIS reconnaissance verified by field crew members

Supplemental Material to the Field Manual

This field manual describes field protocols and daily operations for crews while performing sampling for the UASE program. Following these detailed protocols will ensure consistency across the State and reproducibility for future assessments. Before sampling a site, crews should prepare a packet for each site containing information pertinent to a successful sampling event. This packet might include road maps; topographic maps; a set of directions to the sampling site; land owner access forms (if applicable); sampling permits (if needed); site evaluation forms, and other information necessary to ensure an efficient and safe sampling day.

Key documents and their addresses on the DWQ network to be used in accordance with this UCASE Field Manual include:

- Equipment manuals
- DWQs standard operating procedures (SOPs) for field methods: https://deq.utah.gov/water-quality/quality-assurance-and-quality-control-program-monitoring-water-quality
- DWQs Quality Assurance Program Plan (QAPP) Put a link to the website: https://deq.utah.gov/water-quality/quality-assurance-and-quality-control-program-monitoring-water-quality
- Utah Monitoring Plan: https://deq.utah.gov/water-quality/strategic-monitoring-plan-water-quality

2 DAILY OPERATIONS SUMMARY

This section presents a general overview of the UCASE activities that a field team is to conduct during a typical 1-day sampling visit to a site. This section gives general guidelines for recording data using standardized field data forms and sample labels and also includes safety and health considerations related to day to day field operations.

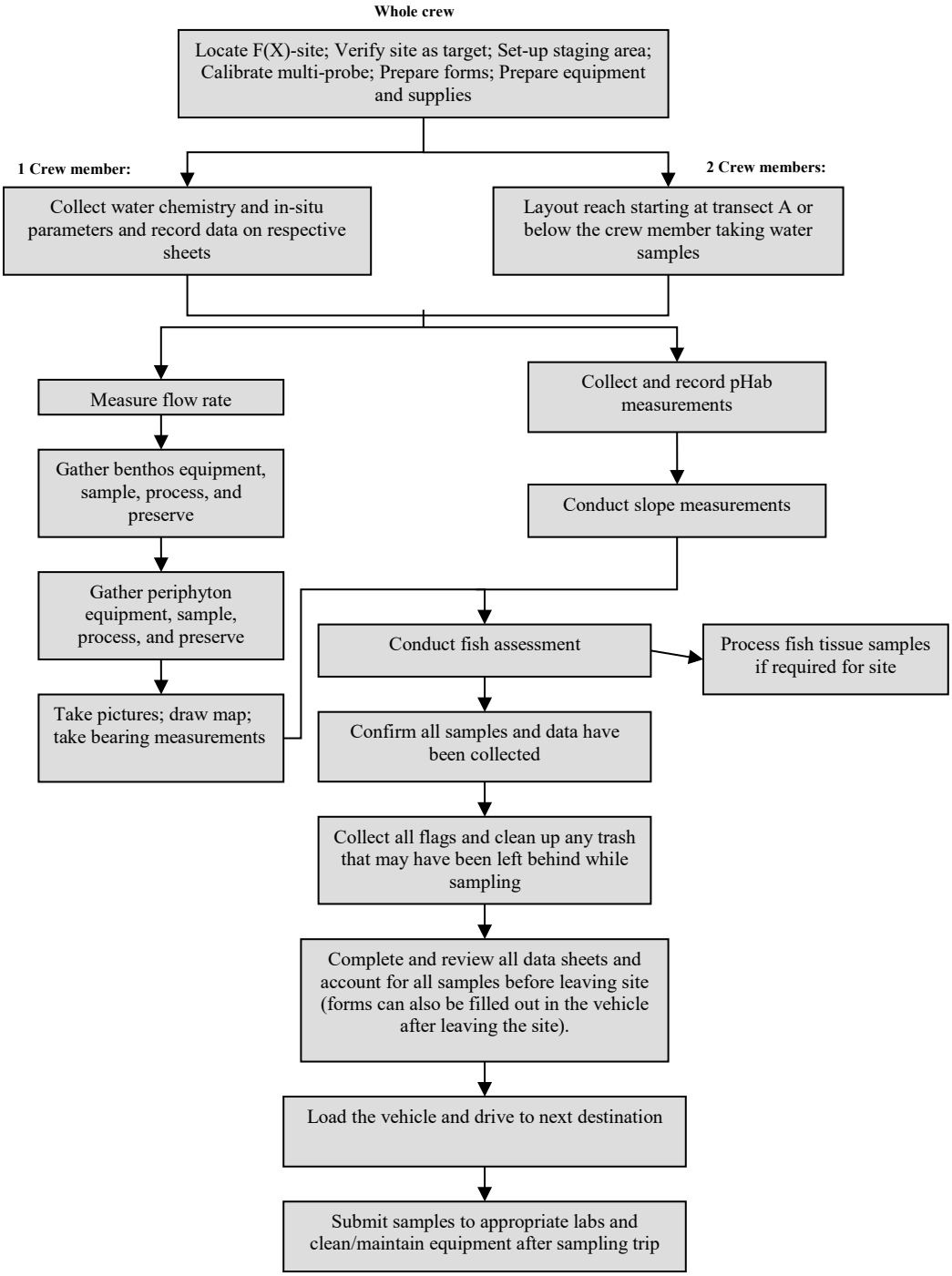
2.1 Sampling Scenario

The field methods for UCASE are designed to complete one site in one day including travel and sample processing time in most cases. Depending on the remoteness and accessibility, certain sites may require extra time to access, such as sites that can only be accessed by foot or horse. Remote sites with lengthy or difficult approaches may require more time and field crews will need to plan accordingly.

2.1.1 Field Crew

Field crews typically consist of three or four people depending on the location and size of stream. If a site is fairly remote more people may be needed to help pack in sampling gear and/or personal gear. Crews of less than three people are not recommended and generally require modifications to the protocol such as not electro-fishing. Site sampling should be postponed to a later date in the sampling period if three people are not available at that particular time. Having a crew of at least three people fosters better consistency, organization, and efficiency to complete a site. If any additional team members are present they can either help collect samples or remain on the bank for observational purposes or for logistical support. A daily field scenario suggesting how work load may be split between three crew members is presented in Figure 2-1. Dynamics between crew members and site conditions will inevitably yield variations of work flow organization as presented in Figure 2-1. It is to the crews' discretion to decide the most effective work assignments/flow as long as SOPs are being followed and all analytical samples are being sampled the same day.

Figure 2-1. Field sampling scenario



2.2 Recording Data and Other Information

All samples need to be identified and labeled in the field. If any tracking or recording of samples onto data sheets is necessary (i.e. hydrodata, number of bug jars, etc.), this should be done prior to leaving the site. To assist with tracking and label consistency, the label format in Figure 2-2 should be used for their respective samples. Label templates can be found on the DWQ network by following this folder pathway: U:\PERMITS\MONITORS\Labels.

It is imperative that field and sample information be recorded accurately, consistently, and legibly. The cost and time of a sampling visit coupled with a relatively short index period severely limits the ability to resample a site if the initial information was inaccurate or illegible. Guidelines for recording field measurements are presented in Table 2-1.

Data sheets and templates can be found on the DWQ network as well, by following this folder pathway: U\PERMITS\MONITORS\UCASE\Official Packet\Complete Packet.

Figure 2-2. Example of sample labels for sample tracking identification

BENTHOS Field preservation: alcohol. Lab: USU Bug Lab

Site Name:_____

_____

Site Code: _____ # Bottles: _____ of _____

Samplers: _____Date:_____

Equipment Type:_____ # of Kicks:_____

BENTHOS COMPOSITE SAMPLE
(Interior label)

Site Name:_____

_____

Site Code:_____

Collection Date:_____

Equipment Type:_____

# of Kicks:_____

Sampler (s):_____

# of Bottles:____OF____

Chlorophyll-*a* by SM10200H(2)(b) Field preservation: dry-ice. Lab: UPHL

SITE NAME: _____

_____

SITE CODE:_____

DATE: _____SAMPLERS:_____

FIELD METHOD (Circle One): WATER / ALGAE / NDS

SPECIES ID Field preservation: wet-ice. Lab: Rushforth

Circle one sample type: QUANTITATIVE or QUALITATIVE

Site Name:_____

_____Site Code: _____

DATE: _____ SAMPLERS:_____

SUB SAMPLE VOL: __50mL___COMPOSITE VOL: _____

FISH COLLECTION Field preservation: dry-ice. Storage: freezer in shop

Site Name:_____

Site Code: _____ Samplers: _____

Length (mm): _____ Weight (g): _____Date: _____

Fish ID: _____

(Species code-Fish sequence #)(ex: CTT01, CTT02, CTT03…)

AFDM by SM10300C–Periphyton Field preservation: dry-ice. Lab: UPHL

SITE NAME: _____

_____

SITE CODE: _____

DATE: _____SAMPLERS:_____

FIELD METHOD (Circle One): WATER / ALGAE / NDS

Table 2-1. Guidelines for recording field measurements and tracking information

Activity	Guidelines
Field Measurements	
Data Recording	Record measurement values and observations on data forms preprinted on water-resistant paper. Only use pencil to record information on forms. Record data and information using correct format as provided on data forms. Be sure to accurately record site names, sample numbers, and STORETs. Print legibly (and as large as possible). Clearly distinguish letters from numbers (e.g., 0 versus O; 2 versus Z; 7 versus T or F, etc.). In cases where information is recorded repeatedly on a series of lines (e.g., physical habitat characteristics), do not use "ditto marks" (") or a straight vertical line or leave values blank. Fill in every value necessary. When recording comments, print or write legibly. Make notations in comments fields only; avoid marginal notes. Be concise, but if you run out of space for comments, attach a sheet of paper with the additional information rather than trying to squeeze everything into the space provided on the form.
Data Qualifiers (Flags)	Use only the defined flag codes and record on data form in appropriate field. K = Measurement not attempted or recorded Q = Failed quality control check: re-measurement not possible U = Suspect measurement; re-measurement not possible F*n* = Miscellaneous flags (*n* = 1, 2, etc.) determined by the person recording data depending on the particular field sheet. Flags remain constant for **individual** data sheets and not the entire site (e.g. F1 would stand for something constant for the transect A sheet, but F1 would mark something else for the transect B sheet). Explain reason for using each flag in comments section on data form.
Sample Labels	Use adhesive labels with preprinted fields and follow the standard recording format for each sample type. Use a pencil to record information on label (ink will dissipate or bleed with contact to alcohol or water). Cover the completed label with clear tape.

Table 2-1. Guidelines for recording field measurements and tracking information *(cont.)*

Activity	Guidelines
Sample Collection and Tracking	
Review of Labels and Data Collection Forms	Compare information recorded on labels and sample collection form for accuracy before leaving site. Review labels and data collection forms for accuracy, completeness, and legibility before leaving site. The crew should review all labels and data forms for consistency, correctness, and legibility before submitting samples to lab and submitting field forms to field crew leader.

2.3 Safety and Health

Collection and analysis of samples can involve significant risks to personal safety and health. This section describes recommended training, communications, and safety considerations, safety equipment and facilities, and safety guidelines for field operations.

2.3.1 General Considerations

Important considerations related to safety are presented in Table 2-2. It is the responsibility of the Monitoring Section supervisor AND the field crew to ensure that the necessary safety courses are taken by all field personnel and that all safety policies and procedures are followed.

Field crew members should become familiar with the hazards involved with sampling equipment and potential natural/anthropogenic hazards at a particular site. Make sure all equipment is in safe working condition. Personnel must consider and prepare for hazards associated with the operation of motor vehicles, tools, electrofishers, natural hazards, etc. Crew members should be aware of the environment they will be working in prior to departure for the trip. This includes climate, weather forecast, site conditions, human caused hazards, etc. Prior to leaving for the trip, crews should always make sure they have a first aid kit, a shovel, and adequate food and water for the period of time spent away from a nearby city or town. Taking extra supplies on more remote trips is highly recommended (e.g. food, water, clothing, satellite phones, inverters, sleeping bags, shelter, etc.).

Ultimately, a safe trip will be dependent on the crew and the decisions they make together while out in the field. Unexpected hazards and conditions will routinely arise without warning at any time. The UCASE program often brings the highest risk to crews' personal safety. Awareness, planning, and common sense from the crew toward

potentially dangerous situations will inevitably determine the overall success of the trip. Some obvious safety measures have been put into place to assist with safety in the field, but overall, health and safety in the field will be determined by the crews' ability to effectively communicate and use best professional judgment between one another.

Primary responsibility for safety while fish shocking rests with the crew members, collectively. Electrofishing units may deliver a fatal electrical shock and should only be used by qualified experienced operators. Wearing insulated gloves and non-breathable chest waders is highly recommended for field crew members who partake in electrofishing. Breathable waders rarely protect crew members from getting shocked especially on hot summer days. Crew members should bring two pairs of waders on UCASE trips; one set of non-breathable waders used for electrofishing and another pair of breathable waders for sampling all other parameters. Avoid contact with the anode and cathode at all times to prevent a potential shock hazard. If certain crew members perspire heavily, they should attempt to wear polypropylene or some other wicking and insulating clothing instead of cotton. If it is necessary for a team member to reach into the water to grab a fish or something that has been dropped, do so only after the shocking unit has been turned off and anode is out of the water. Do not resume electrofishing until all crew members are clear of the shocking hazard. The electrofishing equipment is equipped with a 45º tilt switch that interrupts the current. Do not make any modifications to the electrofishing equipment that would hinder the safety switch. Do not shock a stream where people outside of the crew, pets, or livestock might come into contact with the electrical current. Discontinue activity during thunderstorms or rain. Crew members should stay in constant view and communication with one another while shocking occurs. Each team member has the responsibility to question and modify an operation or decline participation if it is unsafe.

For more information on product operation, safety, and manuals visit the following website: http://www.smith-root.com/downloads/product_manuals/ or http://www.halltechaquatic.com/category/support-documents.

Table 2-2. General health and safety considerations

Recommended Training
• First Aid/CPR/AED • Vehicle safety (e.g. operation of four-wheel drive vehicle) • Field safety (weather, personal safety, orienteering, site reconnaissance prior to sampling) • Equipment design, operation, and maintenance • Handling of chemicals and other hazardous materials
Communications
• Check in schedule with Monitoring Section supervisor • Sampling itinerary on Monitoring Section's calendar (vehicle used and description; time of departure and return; travel route) • Cell phone and/or satellite phone • Emergency services available near each sampling site and base location
Personal Safety
• Field clothing and other protective gear • Medical personal information (allergies, personal health conditions) • Personal contacts (family telephone numbers, etc) • Physical exams and immunizations • Bug repellent and sunscreen

A communication plan between the Monitoring Section supervisor and field crew should be established each week prior to going out in the field. The supervisor should be made aware of when and where the crew is leaving for the week and when they plan on returning whether it is verbally or through the section's Gmail calendar. Unless issues or emergencies arise throughout the course of the trip, the supervisor does not need to be contacted every day.

When sending an appointment to the Monitoring Section's Gmail calendar to plan a UCASE trip, the following should be included:

- Crew members going on trip (this includes personnel outside of the section)
- Dates and duration crew plans on being in the field
- Vehicle being used
- Towns or locations of where crew plans to stay overnight
- Site names and IDs that crew will be sampling for the trip
- If the site is remote and no cell phone service is available, include number for the satellite phone taken by crew.

Proper field clothing should be worn to prevent hypothermia, heat exhaustion, sunstroke, drowning, and other dangers. Hip or chest waders are highly recommended while sampling (required for electrofishing), but it is up to the individual if he/she would prefer sampling in shorts, sandals, or any other clothing.

Many hazards lie out of sight in the bottoms of rivers and streams. Broken glass or sharp pieces of metal embedded in the substrate can cause serious injury if care is not exercised when working in such environments. Infectious agents and toxic substances that can be absorbed through the skin or inhaled may also be present in the water or sediment. Personnel who may be exposed to water known or suspected to contain human or animal wastes that carry causative agents or pathogens must be immunized against tetanus, hepatitis, typhoid fever, and polio. Water contaminated with biological wastes can also be a threat in the form of viruses, bacteria, rickettsia, fungi, or parasites.
If a crew member has a medical condition like allergies or diabetes it is vital that other crew members are aware of this in case something happens unexpectedly in the field. It is important for fellow crew members to know where the appropriate medications are and how to use them in case the victim becomes unresponsive. If medical conditions are rather personal, at the very least, let the crew leader know so they can act to remedy the issue. Helicopter Use

If a helicopter is used to access remote sites the crew leader for the trip needs to send an informal flight plan in email format to someone in the Monitoring Section that will be reachable all day (office contact). The section manager should take precedent over others in the section, but if he/she is unable to attend to their phone all day or be near internet service to access information about the flight plan and sampling site, someone in the section who has access to these services should be used. The crew leader must include the following in their email:

- Personnel on flight
- Name of pilot and the company he/she works for
- Phone number to flight company (other than the pilot)
- Site name(s) including unique site IDs
- Planned departure time from the airport
- Planned arrival time back to airport
- Designate an "emergency time" that is later than the planned arrival time for when the office contact should call the flight company or emergency services in case communication has not occurred with field crew at designated arrival time (approximately 4 hours after arrival time if no contact has been made between crew leader and office contact).

Each field crew member on the flight should always bring plenty of food, water, appropriate clothing, and medications for themselves in case of unforeseen helicopter accident or mishap. In most cases each crew member is allowed to bring one personal medium sized backpack with their gear they find necessary to bring. Even though pilots usually carry emergency gear and rations, the sampling crew should attempt to have the following between them:

- Satellite phone
- Water purifier
- First Aid Kit
- Fire Starter
- Compass
- Knife

- Flashlight

On the day of the flight, the crew leader will make aware to the office contact that the flight will depart on time, be delayed, or be cancelled. The crew leader will immediately call the office contact once the flight has arrived back to let he/she know the flight is complete. The crew leader is responsible, to the best of their abilities, to contact the office contact as soon as possible if the flight will be delayed or an emergency has occurred. If the office contact has not heard from the crew by the designated "emergency time" they are to call the flight services contact or emergency personnel immediately and report the situation.

The section supervisor or crew leader should make aware to the crew the potential dangers, both on flight and ground, of accessing sites via helicopter. Helicopters have a certain risk of failure, which often present deadly and/or emergency situations. People often get sick or uncomfortable while flying. If a staff member is uncertain or uncomfortable with their flying abilities, it might be best they do not go on the trip.

2.3.2 Safety Guidelines and Field Operations

General safety guidelines for field operations are presented in Table 2-3. Personnel participating in field activities should be in sound physical condition and have experience with sampling or working in a variety of field conditions. All surface water and sediments should be considered potential health hazards due to potential toxic substances or pathogens. Crew members should become familiar with the health hazards associated with using chemical fixing and/or preserving agents. Chemical waste can be hazardous due to flammability, explosiveness, toxicity, causticity, or chemical reactivity. All chemical wastes associated with UCASE sampling must be stored until they can be disposed of properly at the State of Utah Health Laboratory.

During the course of field sampling activities, field teams may observe violations of environmental regulations, may discover improperly disposed hazardous materials, or may observe or be involved with and accidental spill or release of hazardous materials. In such cases it is important that the proper actions be taken and that field personnel do not expose themselves to something harmful. If field crews observe any of these activities they should immediately call his/her supervisor so that the proper people can be contacted to respond to the issue.

Field personnel should never disturb or retrieve improperly disposed hazardous materials from the field to bring back to a facility for disposal. To do so may worsen the impact, incur personal liability for the team members and/or their respective organizations, cause personal injury, or cause unbudgeted expenditure of time and money for proper treatment and disposal of the material. Notify the appropriate authorities so they may properly respond to the incident. In the event of a major environmental incident, call the DWQ Spill Line at 801 536 4123.

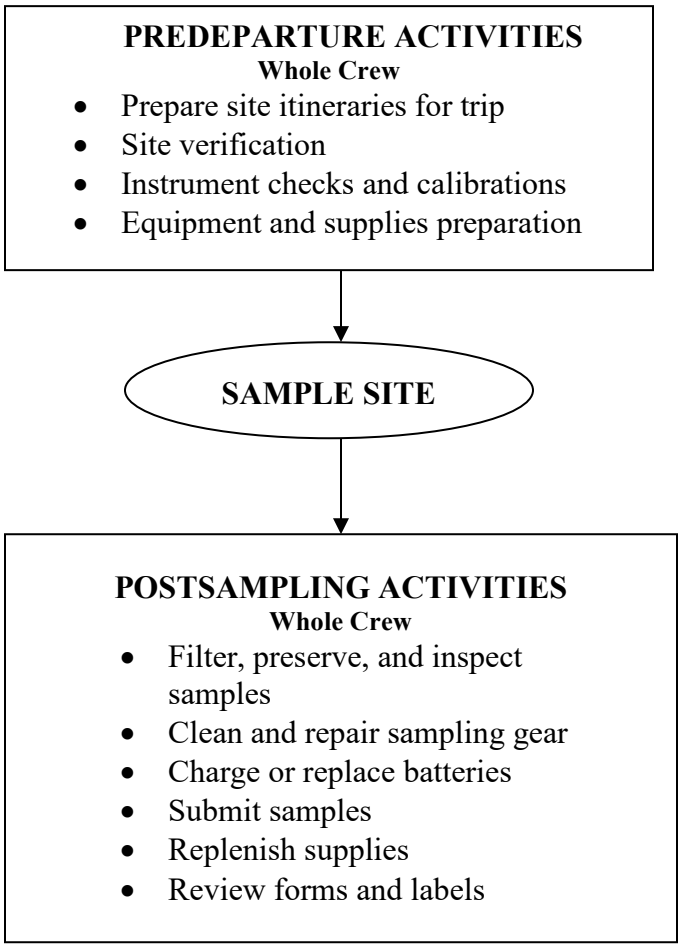
Table 2-3. General safety guidelines for field operations

- Crew members should never be left alone while in the field. Always travel together and remain within close proximity to one another while sampling.
- If site conditions make any one member of the crew uncomfortable, the sampling should be postponed to another day or the site should be dropped.
- Use caution when sampling in swift or deep water. If desired, crew members can wear a personal flotation device (PFD) at potentially deep sites.
- Field crew members using electrofishing equipment must be insulated from the water and electrodes via non-breathable waders and gloves.
- Use extreme care walking on rip-rap. Rocks can shift unexpectedly and serious falls are possible.
- Electrofishing equipment may deliver a fatal shock and should be used by experienced operators.
- Professional quality breathable waders with a belt are recommended for all sampling except for electrofishing.
- Exposure to water and sediments should be minimized as much as possible. Use gloves if necessary and clean exposed body parts as soon as possible after contact.
- All electrical equipment must bear the approval seal of Underwriters Laboratories and must be properly grounded to protect against electrical shock.
- Use heavy gloves when hands are used to agitate the substrate during collection of macroinvertebrate samples.
- Use appropriate protective equipment (e.g. gloves, safety glasses) when handling and using hazardous chemicals.
- Crews working in areas with poisonous snakes should know the whereabouts of the closest medical center in case of snake bite. Crew members should also be properly trained on how to treat a snake-bite victim.
- Any person allergic to bee stings, insects, or plants must take proper precautions, make crew members aware of allergy, and have any necessary medications on hand.
- Field personnel should also protect themselves from ticks because of the potential risks of acquiring pathogens that cause Rocky Mountain spotted fever and Lyme disease.
- Field crews should be familiar with the symptoms of hypothermia and know what to do in case symptoms occur. Hypothermia can kill a person at temperatures above freezing if he/she is exposed to wind or becomes wet.
- Field personnel should be familiar with the symptoms of heat/sun stroke and be prepared to move a suffering individual into cooler surroundings and hydrate immediately.
- Handle and dispose of chemical wastes properly. Do not dispose of chemicals in the field.
- Field crew members using a helicopter should be prepared for a variety of flying conditions and have direct communication with a designated office contact to inform progress of flight plan.

3 BASE SITE ACTIVITIES

Field teams conduct a number of activities from their base site. This is the location where a crew might travel from to access a site for the day (i.e. office, sampling site, hotel, the Shop, campsite, etc.). These include tasks that must be completed both before departure to the sampling site and after returning from the field (Figure 3-1). These activities are especially important for the field crew because they address where the crew will be sampling; review accessibility information; assure equipment and supplies are available and in working order to complete sampling efforts; assure samples are labeled and submitted properly; and assure that all required documentation is completed.

Figure 3-1. Overview of base-site activities



3.1 Pre-departure Activities

Pre-departure activities include the development of a trip itinerary (e.g. maps, directions, landowner permission forms, etc.), instrument checks and calibration, and equipment and supply preparation. Procedures for these activities are described in the following sections.

3.1.1 Daily Itineraries

At least one member of the crew should be put in charge of putting together itineraries for each site prior to leaving for the trip. This entails developing "site packets," compiling of maps, contact information, permission letters, access instructions, directions to medical facilities, etc. It is recommended that a crew member with the most familiarity with the vicinity of the site prepare directions and maps. None of these items are required for the site packet, but depending on the crews' knowledge of the site or area they will be traveling to will determine the content of the packet. Additional activities include confirming the best access routes, calling landowners or local contacts, confirm lodging plans, and coordinating rendezvous locations with individuals who choose to meet with field crew members before sampling a site. The following is a list of items a crew may want to include in their site packets:

- Topo maps of F-transect
- Google maps of F-transect
- Topo maps of watershed (showing roads/trails and other routes)
- Google maps of watershed (showing roads/trails and other routes)
- Directions to access the site
- Site IDs; site coordinates; site name
- List of emergency facilities in the area with directions
- Land owner information
- Permission letters if applicable
- Emergency contact information
- Anecdotal information

If the trip itinerary is delayed, changed, or postponed, crew members should inform the Field Coordinator of the situation.

3.1.2 Instrument Checks and Calibrations

Prior to leaving the base location, the crew should know if all of their gear is in working order. An attempt to calibrate the multi-parameter unit at the sampling site should always be made. However, some sites locations will not always allow for this to occur especially at remote sites. In these cases the crew can calibrate all probes except for DO, prior to leaving for the site wherever they may be. Because of the potential influence of altitude, DO calibration is to be performed only at the site. Crews can take a small jug of tap water to calibrate DO while they are at the site (This is required with Hydrolab type probes, In-Situ brand probes auto adjust for barometric pressure/elevation).

Other equipment to routinely check for operation quality are GPS units, flow meters, laser rangefinder, electrofisher, and geo-pumps. Prior to departure, field teams should:

- Turn on GPS unit and check the batteries to make sure it is functional. Replace the batteries if they are bad or low before going in the field.
- Turn on multi-parameter unit and make sure it is functional and charged. All meters should be calibrated at each site unless special circumstances apply. All units should be calibrated according to DWQ protocols (see https://deq.utah.gov/water-quality/quality-assurance-and-quality-control-program-monitoring-water-quality).
- Turn on electrofisher unit and check the batteries. Crews should bring an extra battery and battery charger with them on their trip.

3.1.3 Equipment and Supply Preparation

Field teams must check the inventory of supplies and equipment prior to departing the Shop using the equipment and supplies checklists provided in Appendix A. Crews have the option of taking an enclosed double-axle trailer with them on their trip if space is limited in the vehicle or if it will help with equipment organization. Crew members should consider how they pack gear into the vehicle and/or trailer to avoid personal injury during transport and avoid damage to equipment.
Sampling kits with any necessary supplies and gear for sampling will be built at the Shop in Salt Lake City every season before the sampling index period. These kits should be kept together throughout the season. It is each crew's responsibility to replenish supplies throughout the season as they get used or are broken/damaged. Crews should always return supplies to the kits after use so that the next crew to use it is not left without the gear they need. Typically, there will be two or three kits built each season and can be found in the UCASE gear area at the Shop.

3.2 Post Sampling Activities

After completely sampling a site, at least one crew member should check the site packet to make sure all information that must be collected at the site is accounted for. Site packets don't necessarily have to be completed at the site, but any component requiring on-site information needs to be accounted for before leaving the site (e.g. pHab sheets, benthos sheet, flow sheet, water chemistry sheets, etc).

3.2.1 Review Data Forms and Labels

Crew members are responsible for reviewing all data forms and labels for accuracy, completeness, and legibility. Ensure that written comments use no "shorthand" or abbreviations except those that are included in the Monitoring Manual (e.g. Ck for creek; R for river; Rd for road; Xing for crossing, etc.). The data forms must be completed before turning them into the UCASE field crew lead (APPENDIX B). All site packets should be completed the SAME day the site was sampled. It does not matter whether this is done at the site or while driving back to the base site but it must be done on the same day. Once the forms are submitted to the field crew leader, he/she will double check the sheets and initial them to indicate they have been proofread. Each sample label must

have the same site ID (MLID) as the site packet. Each sample label must be checked for accuracy, completeness, and legibility before leaving the site.

3.2.2 Inspect and Prepare Samples

All samples need to be inspected and appropriately preserved and packaged for transport. Check that all samples are labeled and that labels are completely filled out. Check that each label is covered with clear plastic tape to prevent it from getting wet. All samples, except for the macroinvertebrate samples, should be placed in water tight zip-top bags before going into a cooler. This prevents sample labels from getting soggy, fading, or falling off. Macroinvertebrate jars need to be sealed with a layer of black electrical tape and should be placed in a container such that they remain upright for the remainder of the trip. Check the integrity of each sample container and be sure there are no cracks or leaks. Make sure all sample containers are properly sealed. Make sure that all sample containers are properly preserved for storage or lab submittal. The processing and preservation processes for each sample type are discussed individually in their respective sections in the manual.

3.2.3 Equipment Clean-up and Check

All equipment and gear must be cleaned and/or disinfected between sites to reduce the risk of transferring nuisance species and pathogens. Some species of concern for Utah include Eurasian watermilfoil (*Myriophyllum spicatum*); zebra mussels (*Dreissena polymorpha*); New Zealand mud snails (*Potamopyrgus antipodarum*); *Myxobolus cerebralis* (sporozoan parasite that causes whirling disease in fish); and *Batrachochytrium dendrobatidis* (a chytrid fungus that threatens amphibian populations). Shoes and wading boots used for UCASE sampling must always have a rubber-based sole. Felt-bottomed footwear is not allowed. Crews should always carry a mixed solution of Sparquat (see product container for proper solution mixtures) to disinfect sampling equipment, waders, and boots. These items will be sprayed or submersed in a Sparquat solution and rinsed upon completion of each sampling site.
Several resources regarding nuisance species can be found online:

- Whirling Disease Foundation (www.whirling-disease.org)
- USDA Forest Service (http://fs.fed.us/invasivespecies/documents/Aquatic_is_prevention.pdf)
- USGS (http://www.protectyourwaters.net/htichhikers)
- Utah Division of Wildlife Resources (https://wildlife.utah.gov/invasive-mussels.html)

More information on disinfecting gear can be found the Utah Division of Wildlife Resources https://stdofthesea.utah.gov/Handle and dispose of disinfectant solutions properly and take care to avoid damage to equipment and property. Inspect and clean all equipment before going to the next site

Table 3-1. Post sampling equipment care

1. Clean for biological contaminants
- Prior to departing the site, drain all water from buckets used for benthos and fish sampling
- Before leaving the site, inspect and rinse periphyton sampling equipment, dip nets, d-nets, waders and boots with water. Allow this equipment to air dry if possible.
- If disinfection is required, the following methods can be used:
  - On-site: Air dry equipment for at least 4 hours.
  - Chemical disinfection: Sparquat is the preferred chemical solution to use on gear. Use the solution recommendations on the product container for proper concentrations. Bleach is not recommended to use as a disinfectant but can be used if it is the only option. Gear should sit in Sparquat for at least 5minutes and then rinsed.
  - Freezing: Gear can be placed in a freezer overnight.
  - Heating: Gear can be soaked in 120ºF water for at least 1 minute.

2. Clean and dry other equipment prior to storage
- Rinse coolers with water to clean off any dirt or debris on the outside and inside.
- Rinse periphyton sampling equipment with tap water at base location
- All equipment should be stored dry

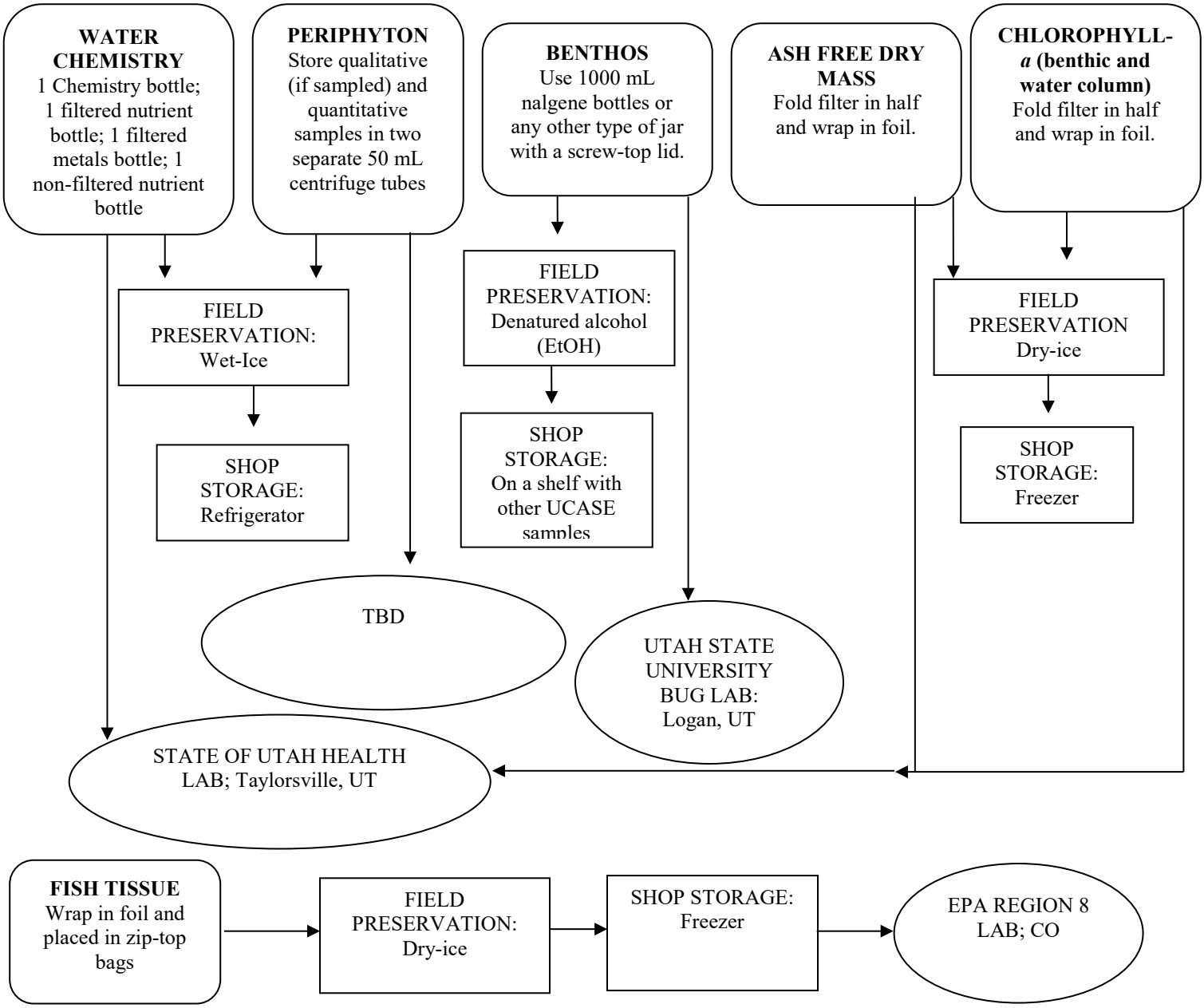
3. Inventory equipment and supply needs and relay purchase requests to the Field Coordinator.

4. Store gear in their proper locations at the Shop.

Sample Submittal

An attempt should be made to submit water chemistry and periphyton samples after each sampling trip. If this is not possible then those samples must be submitted the following week after the trip. Each sample except for the benthos and fish samples need to be submitted to their appropriate lab no later than two weeks from its collection time (except for water chemistry samples, which need to be submitted within one week of collection). If samples cannot be submitted after the trip they will be stored at the Shop. The water chemistry samples should be stored in a refrigerator. The ash-free dry mass and chlorophyll samples need to be stored in a freezer. Qualitative and quantitative periphyton samples will be stored in the refrigerator as well. Benthos samples should always be stored at the Shop and are be submitted to the lab at the end of the field season in one batch. Fish tissue samples should be frozen immediately until they can be processed. Figure 3-2 shows preservation and handling procedures for each sample.

Figure 3-2. Sample handling process



4 INITIAL SITE PROCEDURES

When the crew arrives at a site, they should confirm they are at the correct site and determine if the site meets the criteria for sampling and data collection activities. Inspect the selected reach for appropriate access, safety, and general conditions. **Permission to access private property is required**. Decide whether the site is at base flow condition and not unduly influenced by rain events or snow-melt, which could affect the representativeness of field data and samples. If the crew determines that the site can be sampled, prepare to lay out the reach within which all sampling and measurements activities are conducted.

4.1 Site Verification Activities

4.1.1 Locating the F (X) Transect

Whenever a crew is given coordinates for a site location, that point is commonly referred to as the "F-transect" and occasionally as the "X-transect". The "F-transect" is the mid-point of the sampling reach and it will determine the location and extent for the rest of the sampling reach. The coordinates of the "F-transect" are listed on the yearly site list that is created by the Field Logistics Coordinator prior to the field season. When recording coordinates on field sheets, they need be taken from the F-transect and no other transect. The "F-transect" applies to both probabilistic and targeted sites.

Conditions encountered at streams across the state will vary tremendously. To orient crews and help anticipate sampling and access challenges, each site should be prepared with a site dossier, especially probabilistic sites. Targeted sites may be more difficult to plan because the crew will not always know exactly where they will be sampling beforehand. Each dossier should contain maps with the F-transect plotted and show general conditions at each site. Maps should include "watershed" scaled maps that show the position of the site on the landscape and stream network. Maps should also include "site-scaled" images that show the area around the site where samples will be taken. Other maps can include roadways/trails that the crew will use to access the site. Dossiers should also contain emergency contact information for the crew and locations/contact information for nearby medical facilities. Directions can also be added to the dossier if the crew is unfamiliar with how to get to a site.

Table 4-1 is the checklist for equipment and supplies required to conduct site verification protocols used to describe this section. It is a subset in Appendix A that is used at a base site to assure that all equipment and supplies are taken to and available at the site. While traveling from a base location to a site, record a detailed description of the route taken on the *Verification Form* Figure 4-1. This information will help others find the site again in the future. Upon reaching the F-transect, confirm its location and verify that you are at the correct stream. Use all available means to accomplish this, including map coordinates, location data from the GPS, and any other evidence such as signs or conversations with local residents, and record the information on the front page of the *Verification Form* Figure 4-1. Complete a verification form for each site sampled, following the procedures described in Table 4-2.

Table 4-1. Equipment and supplies list for site verification

For locating and verifying site	• Sampling permit and landowner access (if required) • Field Manual • Site dossier that includes directions, coordinates, directions, anecdotal information, nearby medical facilities, maps, etc • GPS unit with extra batteries • Flagging tape or pin-flags • Rangefinder • Measuring tape with reel that is at least 15 m long (if not using rangefinder)
For recording measurements	• Clipboard • Pencils • Forms • Indelible markers

Figure 4-1. Copy of Verification Form (front)

STREAM VERIFICATION FORM-WADEABLE STREAMS

Reviewed by (initial): BB

SITE NAME: Straight Canyon CK DATE: 10/05/2011 TEAM: ____

SITE/STORET ID: 494020 UCASE ID (if applicable): UT 09 ST- 741 VISIT: 0 (1) 2 3

STREAM VERIFICATION INFORMATION

Stream Verified by (fill in all that apply): ● GPS ○ Local Contact ○ Signs ○ Roads ● Topo Map ○ Crew Experience ○ Other (Describe here): ○ Not Verified (Explain in Comments)

Coordinates	Latitude North	Longitude South	Datum: (Circle one)
Map/Defined pre-trip (fill in only one unit) DMS	___ ___ ___.___	___ ___ ___.___	NAD 27
Map/Defined pre-trip (fill in only one unit) Decimal Degrees	38.42668	112.08698	(NAD 83)
Map/Defined pre-trip (fill in only one unit) UTM	_____	_____	
GPS/Actual at F(X)-SITE (fill in only one unit) DMS	___ ___ ___.___	___ ___ ___.___	Elevation (ft):
GPS/Actual at F(X)-SITE (fill in only one unit) Decimal Degrees	38.42665	112.08696	8711
GPS/Actual at F(X)-SITE (fill in only one unit) UTM	_____	_____	

DID YOU SAMPLE THIS SITE?

● **YES** If YES, circle-in one below

SAMPLEABLE (Choose method used)

- ● **Wadeable** - Continuous flowing water; 90%-100% wadeable
- ○ **Partial Wadeable** - Continuous flowing water; 50%-89% wadeable
- ○ **Wadeable Interrupted** - Flowing water not continuous along reach but ≥50% flowing
- ○ **Impounded by Beaver** - Beaver dam present at site but ≥50% of reach still in its channel and wadeable
- ○ **Altered Channel** - Stream present but not as on map

○ **NO** If NO, check one below

NON-SAMPLEABLE (Choose method used)

- ○ **Dry Channel** - Less than 50% site has flowing water
- ○ **Map Error** - No evidence channel/waterbody present
- ○ **Impounded by Beaver** - Too deep or water outside of its channel
- ○ **Impounded Stream** - Site underneath lake or pond (human caused or natural, discluding beaver)
- ○ **Wetland** - No definable channel
- ○ **Other** - Explain in comments

DIRECTIONS TO STREAM SITE: From Richfield head South towards Monroe. From Monroe go up Monroe Canyon (USFS 078). Follow 078 to USFS Rd 083. Turn onto 083.60 past Manning Meadow Res to the Dry CK Guard Stn. Go ~0.1 miles past guard station & look for ATV Trail 845/089 trailhead. Park here & walk ~75 yards & look for USFS trail 093 to the right (follows enclosure fence). Follow this trail to site.

UCASE Field Form Updated: 03/2011

4.1.2 Determining the Sampling Status of a Stream

After the crew confirms the location of the F-transect, evaluate the stream reach surrounding the F-transect and classify the stream into one of three major sampling status categories: sampleable, non-sampleable, or no access (Table 4-2). The primary distinction between "sampleable" and "non-sampleable" streams is based on the presence of a defined stream channel, water content, and adequate access to the site.
If the F-transect is dry upon arrival to the site it does not necessarily have to be dropped right away. Before deciding to drop the site the crew should determine if there is flowing water, if possible, above or below the site up to 5 stream miles without changing the stream order, stream characteristics, land use type, and if diversion inlets do not exist. Essentially, the site characteristics need to be very similar except that water is flowing. If the crew cannot find water or cannot find access to the stream in different areas, the site should be dropped or visited later when flowing water might exist. If isolated pools are present at the F-transect, but no flowing water exists, the site should be moved to flowing water or dropped as well. If water is flowing in certain locales near the F-transect, but goes sub-surface or into isolated pools the F-transect can be slid to the flowing water and sampled. ***There must be greater than 50% flowing water throughout the reach*** in order to sample the site.

Record the sampling status and the pertinent site verification information on the *Verification Form* Figure 4-1. If the site is non-sampleable or inaccessible no further sampling activities are conducted. If the site is part of a probabilistic design, replace the it with the next oversample site on the site list. Notify the Field Coordinator that the site was dropped.

Table 4-2. Site Verification Procedures

1. Find the stream location in the field corresponding to the F-transect coordinates and the "F" marked on the maps prepared for each site in the dossier. Record the routes taken and other directions on the *Verification Form* so that others can visit the same location in the future.
2. Use a GPS to confirm coordinates at the F-transect as marked on maps and the site dossier (most sites will be in NAD 83 datum, but at times could be in NAD 27).
3. Use all available means to insure you are at the correct stream as marked on the map, including USGS maps, topographic landmarks, road maps, signs, local contacts, etc.
4. Scan the channel upstream and downstream from the F-transect, decide if the site is sampleable, and mark the appropriate box on the *Verification Form*. If the channel is dry at the F-transect, determine if the site can be moved upstream or downstream. If not, then drop or re-visit the site. Record the category on the *Verification Form*.

SAMPLEABLE CATEGORIES

Permanent

- Wadeable: continuous water, 90%-100% wadeable.
- Partial: sampled by wading, ≥50%-89% of site wadeable.
- Wadeable Interrupted: Not continuous water along the reach but is ≥50% flowing.
- Impounded by Beaver: Beaver dam present in reach but ≥50% of reach still in channel and wadeable.
- Altered Channel: Stream channel present but differs from map. If stream is still wadeable and channel can still be defined despite a natural (not including beavers; see above) or man-made impoundments near the site.

NON-SAMPLEABLE CATEGORIES

Permanent

- Dry Channel: Less than 50% flowing water within the reach. Record as "Dry-Visited." If site was determined to be dry from another source and/or field verified before the sampling visit, record as "Dry-Not visited." In the comments section describe why the site might be dry (i.e. drought year, nearby diversions, ephemeral/intermittent segment, poor stream layer coverage from the GRTS design).
- Map Error: No evidence that a waterbody or stream channel was present at F-transect.
- Impounded by Beaver: Beaver dam present and has altered site by flooding the channel and making the site ≥50% non-wadeable.
- Impounded Stream: Stream is submerged under a lake or pond due to human influences or natural influences (beaver impoundments not included; see above). If the impoundment is still wadeable, record it as "Altered," sample the site, and explain conditions in the comments section.
- Wetland: Standing water present, but no definable stream channel or too deep to sample.
- Other: Examples would include underground pipelines/culverts, permanently too swift or deep, or a non-target canal. A sampling site must meet both of the following criteria to be classified as a non-target canal:
    - The channel is constructed where no natural channel has ever existed.
    - The sole purpose/usage of the reach is to transfer water. There are no other uses of the waterbody by human (e.g. fishing, swimming, boating, etc.).
    - If crew is unsure of canal characteristics, coordinate with the Field Logistics Coordinator and/or the Biological Program Coordinator to decide if the canal could be sampled or not. The reason why the canal was or was not sampled should be described in the comments section.

Table 4-2. Site Verification Procedures *(cont.)*

NO ACCESS TO SITE CATEGORIES

- Access Permission Denied: Crew is denied access to the site by landowners.
- Permanently Inaccessible: Site is unlikely to be sampled by anyone due to physical barriers that prevent access to the site (e.g. cliffs, weather, etc.).

5. Do not sample non-target or "Non-sampleable" or "No Access" sites. Fill in the "NO" circle for "Did you sample this site?" and check the appropriate circle in the "Non-Sampleable" or "No Access" section of the *Verification Form*; provide an explanation in the comments section.

4.1.3 Sampling During or After Rain Events

Avoid sampling during or after high flow rainstorm events. It is often unsafe to be in the water during such times and sample representativeness can decrease. Biological and chemical conditions can vary quite differently from those during baseflow during these events. On the other hand, sampling cannot be restricted to only strict baseflow conditions. It would be next to impossible to define "strict baseflow" with any certainty at an unstudied site. Such a restriction would also greatly shorten the index period when sampling activities can be conducted. Thus, some compromise is necessary when determining whether or not a stream should be sampled because of a storm event. To a great extent, this decision is based on the judgment of the field team. Some guidelines to help make this decision are presented in Table 4-3. The major indicator of the influence of the storm events will be the condition of the stream itself. If you decide a site is unduly influenced by a storm event do not sample the site that day. Notify the Field Coordinator to reschedule the site. Crews should give at least 2 weeks for the site to "heal" after any flood event before coming back to re-sample. If an unusually large flood occurs, consider giving the site a longer time to recover.

Crews can continue sampling during small to medium rain events as long as baseflow conditions are not being compromised and all crew members are still comfortable sampling. If a large rain event occurs while sampling where flows and conditions might become dangerous, the crew should leave the site and seek safety. If the crew is forced to leave a site when it has only been partially sampled, they have the option to come sample the site at a later date with certain restrictions. All analytical samples must be collected on the same day (i.e. water chemistry, hydro-data, macroinverts, and periphyton). If any of these samples cannot be collected at the site the same day with the others, then the crew will have to come back and resample all parameters another day. If the crew collects all analytical samples before flow conditions change, but does not finish pHab or electrofishing, they can come back at a later date when the site is safe again.

Table 4-3. Guidelines to determine the influence of rain events

- If it is running at bankfull discharge or water seems much more turbid than typical for the class of stream, do not sample it that day.
- Do not sample that day if it is unsafe to be in the water.
- Keep an eye on the weather reports and rainfall patterns. Do not sample a stream during periods of prolonged heavy rains.
- If the stream seems to be close to normal summer flows and does not seem to be unduly influenced by storm events, sample it even if it recently rained or is raining.

4.1.4 Sampling Areas Influenced by Beaver

If beaver activity has created unsampleable conditions (i.e. deep ponds or a choked stream) then the site should be dropped and replaced with a new site. If beaver activity is present, but the site is still ≥50% wadeable and a channel is still present then continue with sampling. Crews should still attempt to target their bug samples in riffle habitat as presented in the macoinvertebrate collection methods. If the site is predominantly an impounded pool or glide and no riffles exist then crews should target "edge-habitat" for their bug samples by making sweeps along banks and bank-side vegetation instead of kicking bottom substrates. For periphyton sampling, if no substrates can be found to scrub then the crew will use a syringe on fine substrates, sticks, or vegetation. These methods are explained in depth in their respective field method sections (Section 5.5 and 5.6.)

4.1.5 Site Photographs

An attempt should be made to take photos at every site using a digital camera. The date and time-stamp function on the camera should be on. Before taking any photos of the site, a crew member should take a picture of something that identifies the site first (e.g. completed *Site Verification Form* that has site ID and name; a sample bottle that has been properly labeled; the site packet that has site information on it; or even scratch-paper that shows the site IDs and site name). The photographer will then take a picture upstream and downstream at A, F, and K transects. Before taking pictures of the stream at these transects the photographer will take a shot of the flag indicating which transect is being photographed. The photographer should also take 2 or 3 shots that characterize the landscape near the site. Pictures should also be taken of anything that influences site characteristics or is unique or interesting to the site. Photos of fish found at the site should also be taken. Details on taking photos of fish are described in detail in the fish assemblage section (Section 5.7). An abundance of photos is not necessary for each site. The purpose of photos is to associate a visual with the data, not to capture every characteristic that has already been recorded on the data sheets. Somewhere around 10-15 photos per site is sufficient. Photos should be downloaded after every trip into their appropriate site folders in the DWQ network (U:\PERMITS\MONITORS\UCASE).

4.2 Laying out the Sampling Reach

Unlike chemistry, which can be measured at a point, most of the biological and habitat structure measures require sampling a certain length of a stream to get a representative picture of the ecological community. A length of 40 times the channel width is necessary to characterize the habitat and several biotic assemblages associated with the sampling reach. Establish the sampling reach from the F-transect using the procedures described in Table 4-4 and Figure 4-3.

Scout the potential sampling reach to make sure it is clear of obstacles that would prohibit sampling and data collection activities. Record the channel width used to determine the reach length and the sampling reach length upstream and downstream of the F-transect on the back side of the *Verification Form* shown in Figure 4-2. This figure illustrates the principal features of the established sampling reach for wadeable sites including the location of 11 cross-section transects used for collecting samples and physical habitat. If bars or islands are found in the reach always choose the channel with the most flow to layout the sampling reach. Note any landmarks/directions that can be used to find F-transect for future visits.

Table 4-4. Laying out the reach

1. If the crew decides the reach needs to be slid, whether before or after laying out the reach, they should regroup and discuss where to move the F-transect. Some of the reasons as to why the F-transect might need to be moved include the following: tributaries are present with higher order streams (downstream), a change to a lower order streams (upstream), impoundments (lakes, reservoirs, and ponds), physical barriers (falls, cliffs, diversion structures), access restrictions to a portion of the initially-determined sampling reach (private property), dry or non-flowing channel. Refer to Table 4-5.

2. Once the crew determines the best spot for the F-transect, two crew members will determine an average wetted width using a tape measure or rangefinder. The two team members will measure the stream channel at 5 places of "typical" width within approximately 5 channel widths upstream and downstream from the F-transect. Average the 5 readings together and round to nearest 1 m. If the average width is <4 m, use 150 m as a minimum reach length. If the average widths are ≥4 m, multiply the average by 40 and divide the result by 10. This result will be the length of each transect. Record the wetted width used to determine the reach as well as the final reach length on the backside of the Verification Form Figure 4-2.

3. Once the transect lengths have been determined the two crew members will layout the reach by going downstream first (Transects F-A). Transects should be measured by a tape measure or rangefinder assuring not to "cut corners" or bends in the stream. At each transect the team member will place a pin-flag on the bank or tie surveyor's flagging onto vegetation with the appropriate lettered transect they are establishing written on the flagging material. He/she should make every attempt to not disrupt the condition of the stream while laying out the reach. Disrupting the site might yield unrepresentative conditions while sampling. He/she should check the conditions of the site while setting flags to observe anything that might require the site to be slid or to observe any safety concerns (e.g. deep holes, rebar, loose rocks, etc.).

4. Once Transects A-F have been established, the two crew members walk back up to the F-transect and layout Transects F-K only after making sure all water chemistry samples and hydro-data have been collected.

5. If islands exist (Section 4.3.4) at any particular transect then the crew will need to establish an "extra" transect for that side channel.

Figure 4-2. Copy of Verification Form (back)

STREAM VERIFICATION FORM-WADEABLE STREAMS (cont.) Reviewed by (initial): BB

SITE NAME: Straight Canyon Ck DATE: 10/05/2011 TEAM: ____

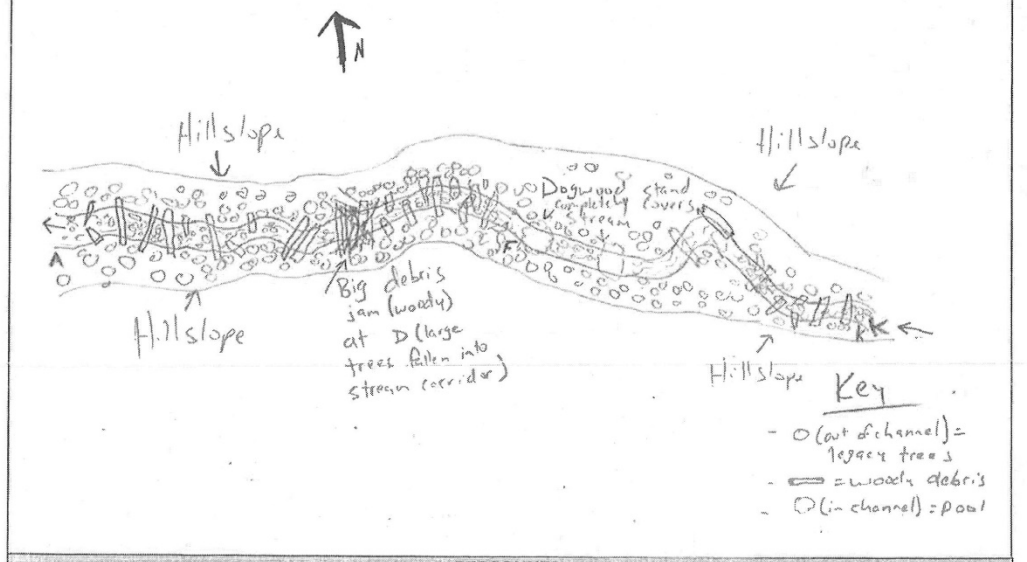
SITE/STORET ID: 494 1020 UCASE ID (if applicable): UT 09 ST- 741 VISIT: 0 (1) 2 3

LAND OWNERSHIP INFO/GENERAL COMMENTS ABOUT SITE LOCATION: Site is on Fishlake NF. Hike required to get to site. Hike is ~1.2 miles long but an established USFS trail exists the whole. Overall, hiking conditions are moderate in difficulty

STREAM REACH DETERMINATION

Channel Width Used to Define Reach (m)	Total Reach Length Intended (m)	COMMENTS
<1	150	

SKETCH MAP - Arrow indicates North

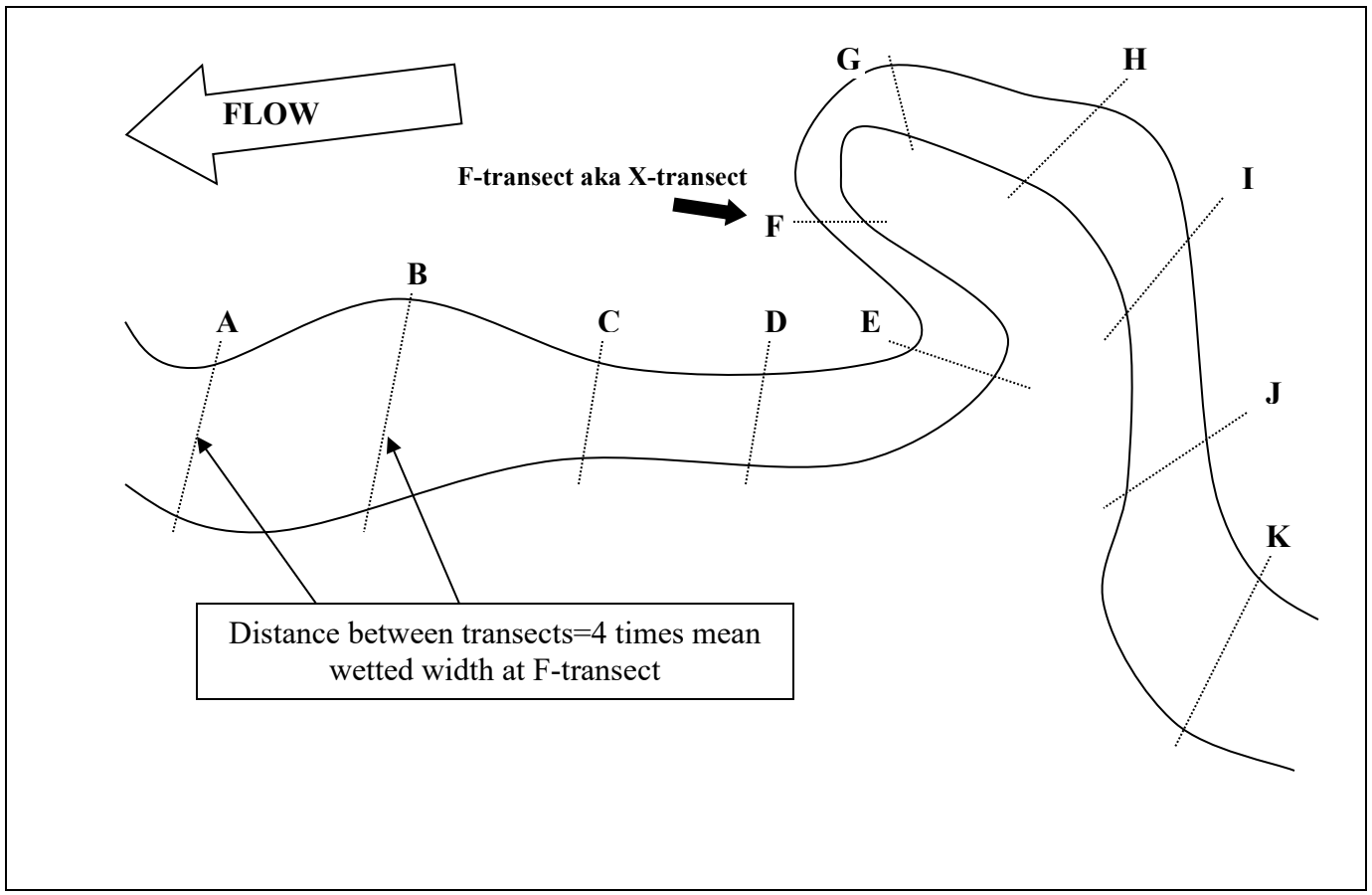


PERSONNEL

NAME	DUTIES Bio/Chem Sampling	Habitat
B. Holcomb	○	●
S. Tubir	○	●
R. Brown	●	○
	○	○
	○	○

UCASE Field Form Updated: 03/2011

Figure 4-3. Sampling reach features for a wadeable site



4.2.1 Conditions for Sliding or Moving a Reach

There are some conditions that may require moving or "sliding" the reach to avoid features we do not wish to sample or physically cannot sample. When a site is moved, reach lengths and procedures remain the same but the new coordinates need to be recorded in the "GPS/Actual" column on the front of the *Verification Form* Figure 4-1 and an explanation for the move needs to be discussed in the comments section. Table 4-5 describes when crews should and should not slide the reach.

Sometimes, due to remoteness, crews will not always be able to reach the exact coordinates for a site. If this is the case, crews should make an attempt to get as close to the sites as possible, whether it be hiking, horse-packing, or helicopter. If crew is certain the stream order does not change, the land use is similar, and the geologic/geographic features are consistent, they have the option of attempting to get as close to the site as possible and relocating it. If a crew is hiking to the site and they know for certain they will not be able to reach it for whatever reason, they can hike for 30 minutes from the closest access point and establish the new point there. 30 minutes must be used at any site where this is the case in an attempt not to bias site selection.

For targeted sites, crews should avoid sampling a site where a tributary is present and changes the order of the stream. Since targeted sites are hand-picked the randomization concerns discussed above do not apply.

Table 4-5. Moving or "sliding" the sampling reach

1. Slide the reach if you run into an impoundment (lake, pond, or reservoir), not including beaver ponds.
2. Slide the reach if you run into an impassable barrier (e.g. falls, culvert, etc).
3. When you are denied access permission to a portion of the reach, you can slide the reach to make it entirely accessible; use the point of access restriction as the endpoint of the reach.
4. Note the distance of the barrier, confluence, or other restriction from the F-transect, and move the site such that transect A begins right above the restriction or transect K is right below the restriction.
5. If moving the reach forces a change in stream order, do not sample site.
6. For probabilistic sites, do not slide a reach to target personal preferences or avoid man-made obstacles, unless access or safety is an issue.
7. For probabilistic sites, do not slide reach to gain better habitat for fish and/or benthos.
8. If a probabilistic site cannot be accessed effectively due to its remoteness crews can hike for 30 minutes from the nearest access point and relocate the site there.

4.3 Modifying Sample Protocols for High or Low Flows

4.3.1 Streams with Interrupted Flow

Often, especially with randomized sites, crews will find very shallow and low flowing sites across the State. Even though these sites might appear insignificant for intensive sampling efforts, they are indeed very important for assessment purposes for the State. These sites will commonly bring confusion and uncertainty to crews. Crews that come across 'interrupted' sites (≥50% of site is flowing, but might be intermittent in a few locales) are still able to sample the site by making few modifications by following Table 4-6. Note that no data should be collected from streams that are completely dry. Interrupted streams will have some cross-sections amendable to biological sampling and habitat measurements and some that are not. To be considered, target streams must have ≥50% flowing water in the reach length within the channel. Sites that are non-flowing isolated pools are not considered target sites like they are in the EMAP or NRSA protocols. Water chemistry samples (Section 5.1) can be collected anywhere in the reach to obtain an adequate sample. If flow is too shallow to collect water chemistry samples, crew members can dig a small hole deep enough to fill a bottle and collect there. Crew members should let water flow through to hole for at least five minutes before sampling to allow settling to occur.

Collect data for physical habitat indicators along the entire reach from interrupted streams regardless of the amount of water present at transects. Obtain depth measurements along the main channel of flow in the channel, also known as the "thalweg" (this does not

necessarily mean the deepest part of the channel). Thalweg readings (Section 5.2.4) provide a record of the "water" status of the stream for future comparisons (e.g. the percent of length with intermittent pools or no water). Other measurements associated with characterizing riparian condition, substrate type, etc., are useful to help infer conditions in the stream when water is flowing.

Table 4-6. Reach layout modification for interrupted streams

- Streams with ≤50% of flowing water in the reach are considered dry and are not sampled.
- Upon arrival to the F-transect, if it is dry, but the prospective reach has more than 50% of flowing water the site should be still be sampled to protocol.
- If F-transect is dry but the reach potential still has ≥50% of flow, use a representative wetted width average from that potential area as your wetted width average. These sorts of sites will almost always have ≤3 m width therefore the reach will be 150 m.
- If filling water chemistry bottles is difficult due to a lack of water, a small pool can be dug in order to fill samples. The hole should have at least five minutes of continuous flow before sampling.

Physical Habitat and Benthic Macroinvertebrates

DRY CHANNEL: If ≤50% of surface water anywhere in potential reach is not present, then drop the site or look for flowing water above or below the reach.

DAMP CHANNEL: If no flowing water at site, but small puddles of water exist throughout the potential reach, drop the site.

WATER PRESENT: Potential reach has several isolated pools, but not flowing, drop the site.

INTERMITTITENT SHALLOW WATER ≥50% FLOWING:

- Obtain a complete thalweg profile for entire reach. At points where channel is dry, record depth as 0 cm and wetted width as 0 m.
- Still sample at any dry transect. Depths will be zero, but substrate type and embeddedness should still be recorded.
- If a suitable habitat is lacking for benthos collection, make multiple kicks or sweeps at one location.
- Fish shocking can be skipped if water levels are not deep enough to support fish colonization. Be sure to discuss this in the comments section.

4.3.2 Partially Wadeable Sites

Since wadeable sites will inevitably have sections that are too deep or swift to wade safely, at times it will be impossible to do all of the wadeable sampling protocols at every transect. Partially wadeable is defined as ≥50% of site is wadeable. If less than 50% of site cannot be sampled by wading the crew should drop the site or revisit it when flows are less. At partially wadeable sites, keeping safety a priority, try to do as much sampling and data collection as you can with wadeable procedures. The amount of sampling that can actually be done while wading will depend on the extant conditions. When certain

spots cannot be sampled use the correct flagging on data sheets that show the sample could not be taken. Crew members should discuss these sorts of sampling conditions in the comments section.

4.3.3 Braided Streams

Although rare in Utah, crews may encounter braided channels especially in desert systems. Braided streams are characterized by numerous sub-channels that are generally small and short and often have no obvious dominant channel. Figuring the mean width of extensively braided streams for purposes of setting up a sampling reach can be challenging. In these systems, measure the mean wetted and bankfull widths the same as normal, defined in the pHab protocols (Section 5.2). Bars should always be included in your bankfull and wetted width measurements (do not measure bar widths separately and subtract them from overall bankfull or wetted width measurements). If crew members feel the mean wetted width creates the reach to be exhaustingly long (i.e. bars adding to the overall average wetted width), using best professional judgment, they can reduce the reach to a length that is more appropriate. If this is the case, crews should make sure they capture at least 2 to 3 meander cycles or layout the reach by measuring a mean wetted width as if no bars existed.

4.3.4 Islands

At certain sites crews will be faced with islands and bars (Section 4.3.5). It is very important to know the difference between the two as it might influence how the reach is laid out. Extra pHab forms may have to be filled out as well. An island is characterized as a piece of land in the middle of a stream that creates two or more channels and is at bankfull height and higher. There is often a change of vegetation type at this level compared to areas below the bankfull height as it is not impacted by high flows as frequently (e.g. dense willow stands or birch might be present). Soil is often much drier in this area. When islands are present at any of the cross-sections (e.g. A-K) and at least 15% of water is flowing through the secondary channel, crews must fill out an extra *Channel/Riparian Cross-Section Form* (Figure 5-10) for that transect and mark the 'X-tra Side Channel' bubble on the sheet. In cases where this situation exists, the primary channel (channel with the most flow) will only get the 'Transect' bubble filled for its respective transect whereas the secondary channel (channel with less flow) will get the 'Transect' bubble filled AND the 'X-tra Side Channel' bubble as well. If there is < 15% flow in the side-channel a second sheet does not have to be completed for that transect. When filling out the pHab forms the left side of the island essentially becomes the right bank for the primary channel and the right side of the island becomes the left bank for the secondary channel. Often, an island will exist where the two riparian vegetation plots from the primary and secondary channels will overlap because it is a narrow. In these situations, the island might not be big enough to fulfill the vegetation plot requirements. In these cases, emphasize the Barren, Bare Dirt, or Duff category because most of the plot will be water and be sure to flag these circumstances in the comments section. It is acceptable for crew members to sample macroinvertebrates and periphyton in side channels if there is enough flow (See Section 5.2.5.2 and Section 5.2.5.8).

4.3.5 Bars

A bar is characterized as a piece of land in the middle of a stream that creates two or more channels and is at bankfull height and lower. Bars usually have very sparse to no vegetation growing on them. When vegetation is present it is usually in the form of thin stands of grasses and small forbs (pioneer species). Otherwise, bars are usually sandy or have exposed barren substrates. If soil is present it is usually damp. When bars are present crews do not have to fill out any extra pHab sheets like they might when islands are present (Section 4.3.4). Crew members will follow the channel with the most flow as they continue to work upstream while taking measurements and samples. When making cross-sectional measurements it is important to include the bar in the wetted width and bankfull width measurements (do not subtract them out of measurement) See Section 5.2 for further instructions.

5 WADEABLE STREAM SITE SAMPLING

5.1 Water Quality

This section describes the procedures and methods for the field collection and analysis of the water quality indicators (multi-parameter measurements and water chemistry) from wadeable streams.

5.1.1 Multi-parameter Measurements of Dissolved Oxygen, pH, Temperature, and Conductivity

5.1.1.1 Summary of Method

Crews will measure dissolved oxygen (DO), percent dissolved oxygen (DO%), pH, temperature, and conductivity by using a multi-parameter water quality meter (or sonde). Take all measurements with this device at any point in the reach at 0.5 m depth or greater. These measurements should be taken at the same time, but downstream, with the water chemistry samples.

5.1.1.2 Equipment and Supplies

Table 5-1 provides the equipment and supplies needed to measure DO, pH, temperature, and conductivity. Record the measurements on the UCASE *Channel Constraint and Field Chemistry Form* seen in Figure 5-1

Table 5-1. Equipment and supplies-DO, pH, temperature, and conductivity

For taking measurements and calibrating the water quality meter	• Multi-parameter water quality display, cord, and sonde with DO, pH, temperature, and conductivity probes. • Battery charger • Tap water • Calibration cups and standards • SRM (QC) calibration standards • Maintenance kit • Paper towels • Barometer or elevation chart for calibration (Hydrolab brand only)
For recording measurements	• DWQ calibration form • Pencils/pens

Figure 5-1. Channel Constraint and Field Chemistry Form

UCASE CHANNEL CONSTRAINT AND FIELD CHEMISTRY COLLECTION FORM-WADEABLE STREAMS

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ DATE:___ ___/___ ___/ 2 0 ___ ___

MULTI-PARAMEMTER UNIT MEASUREMENTS

	Comments
STREAM TEMP (°C): ___ ___.___	RESIDUAL CHLORINE (optional):
pH: ___ ___.___ ___	
SPECIFIC CONDUCTIVITY (µS/cm) ___ ___ ___ ___ ___ ___	
STREAM DO mg/L: ___ ___.___	
STREAM DO%: ___ ___ ___.___	
TIME OF DAY: ___ ___ ___ ___	

CHANNEL CONSTRAINT

CHANNEL PATTERN (Check one)

- ○ **One Channel**
- ○ **Anastomosing (complex) channel:** (Relatively long major and minor channels branching and rejoining)
- ○ **Braided channel:** (Multiple short channel branching and rejoining – mainly one channel broken up by numerous mid-channel bars)

CHANNEL CONSTRAINT (Check one)

- ○ **Channel very constrained in V-shaped valley** (i.e. it is very unlikely to spread out over valley or erode a new channel during flood)
- ○ **Channel is in Broad Valley** but channel movement by erosion during flood is **constrained by incision** (Flood flows do not commonly spread over valley floor or into multiple channels)
- ○ **Channel is in Narrow Valley but is not very constrained,** but limited in movement by relatively narrow valley floor
- ○ **Channel is Unconstrained in Broad Valley** (i.e. during flood it can fill off-channel areas and side channels, spread out over flood plain, or easily cut new channels by erosion)

CONSTRAINING FEATURE (Check all that apply)

- ○ **Bedrock/Sandstone** (i.e. channel is constrained by bare bedrock or sandstone)
- ○ **Hillslope** (i.e. channel constrained in narrow V-shaped valley)
- ○ **Terrace** (i.e. channel is constrained by its own incision into stream gravel/soil deposits)
- ○ **Human Bank Alterations** (i.e. constrained by rip-rap, landfill, dike, road, etc.)
- ○ **No constraining features**

PERCENT OF CHANNEL LENGTH WITH MARGIN IN CONTACT WITH EACH CONSTRAINING FEATURE (percentage fields should add up to 100%):	**Constraining Feature 1** (primary)	**Constraining Feature 2** (if applicable)	**Constraining Feature 3** (if applicable)
	Type:_____	**Type:**_____	**Type:**_____
	Percentage: ___ ___ ___ %	**Percentage:** ___ ___ ___ %	**Percentage:** ___ ___ ___ %

Valley floor width (Visual Estimated Average): ___ ___ ___ ___(m)

If you cannot see the valley borders, record the distance you can see and mark this circle ○

Comments:

Updated: 11/2012

5.1.1.3 Multi-Probe Sonde

Any multi-parameter unit being used for UCASE must be calibrated prior to sampling the first site for the day. It is recommended the unit be calibrated at the sampling site, but can be done at the base site location if remoteness is a factor (crew does not need to carry calibrations standards on a hiking trip or helicopter trip). DO is an exception to this rule. The DO probe should always be calibrated in the field at or near the site prior to sampling. If the site is remote, crews should take some tap water to calibrate this sensor once they get to the site (only necessary for Hydrolab sondes).

DWQ uses a variety of multi-parameter units (Hydrolab, YSI, In-Situ) for data collection. Any probe can be used for UCASE sampling depending on crew preference and availability. All calibration protocols and unit manuals can be found on DWQ's website [insert link here from above] Crews should always follow the DWQ calibration SOP as the unit manual might not touch on every detail directly related to the DWQ monitoring program. An example of a completed *Calibration Form* can be found in Figure 5-2.

Figure 5-2. DWQ Calibration Form

Sonde-Cal-Report_11.07.2017 11/21/2017

UTAH DEPARTMENT of ENVIRONMENTAL QUALITY
WATER QUALITY

DWQ Multi-Parameter Probe Calibration Report

Run (Trip ID): JRKLI 120417
Date: 12-4-17
Time: 08:05

Analyst: Ryan Parker
Instrument Make & Model: In-Situ SmarTROLL MP
Instrument ID Number: 468563

Specific Conductance (SpC)					
CALIBRATION		QA/QC			
SpC Calibration Standard Solution Value:	SpC Calibration Standard Solution Expiration Date:	SpC Reference Solution Value:	SpC Reference Solution Expiration Date:	SpC Measured Value:	Measured Value ±10% of Reference Solution Value?
500	7-30-18	1413	10-30-18	1373.1	☒ Yes ☐ No[1]

pH							
CALIBRATION				QA/QC			
pH Calibration Solution 1 Value[2]:	pH Calibration Solution 1 Expiration Date:	pH Calibration Solution 2 Value[2]:	pH Calibration Solution 2 Expiration Date:	pH Reference Solution Value:	pH Reference Solution Expiration Date:	pH Measured Value:	Measured Value ±5% of Reference Solution Value?
7.00	3/2019	10.00	6/2018	9.00	10/2018	9.01	☒ Yes ☐ No[1]

Dissolved Oxygen (DO)				Equipment QA/QC			
CALIBRATION			QA/QC				
Barometric Pressure (BP) Used to Calibrate DO?	Calibration Value (%):	Displayed Value (%):	Displayed Value ±5% of Calibration Value?	Instrument Date:	Instrument Time:	Instrument Date and Time Correct?	Does Instrument Battery Have Adequate Charge?
☒ Probe auto-accounts for BP	100.0	100.41	☒ Yes ☐ No[1]	12-4-17	08:04	☒ Yes ☐ No	☒ Yes ☐ No

General Comments:

Calibration Checks					
MLID	Which Probe is Being Checked?	Reason for Calibration Check?	Calibration Value	Measured Value[3]	Measured Value Within Range of Calibration Value? (i.e., SpC, pH, or DO)
	☐ SpC ☐ pH ☐ DO				☐ Yes ☐ No
	☐ SpC ☐ pH ☐ DO				☐ Yes ☐ No
	☐ SpC ☐ pH ☐ DO				☐ Yes ☐ No
	☐ SpC ☐ pH ☐ DO				☐ Yes ☐ No
	☐ SpC ☐ pH ☐ DO				☐ Yes ☐ No
	☐ SpC ☐ pH ☐ DO				☐ Yes ☐ No
	☐ SpC ☐ pH ☐ DO				☐ Yes ☐ No
	☐ SpC ☐ pH ☐ DO				☐ Yes ☐ No
	☐ SpC ☐ pH ☐ DO				☐ Yes ☐ No
	☐ SpC ☐ pH ☐ DO				☐ Yes ☐ No

[1] If no, use a different probe or perform maintenance

[2] When using a Hydrolab brand probe, be sure to correct for temperature when calibrating pH (see chart on back)

[3] If measured value is not within acceptable range of calibration value, perform a recalibration using a new calibration sheet

Sampling Procedure

Table 5-2 presents step-by-step procedures for measuring DO, pH, temperature, and conductivity

Table 5-2. Sampling procedure-temperature, pH, conductivity, and DO

1. Check meter and probes and calibrate according to DWQ specifications 2. Measurements are taken anywhere in the reach at a depth ≥0.5 m. 3. Put the sonde in the water and measure DO, pH, temperature, and conductivity. 4. Record the measurements on the Field Chemistry Form. 5. Flag any measurements that need further comment (or when a measurement cannot be made).

5.1.2 Water Chemistry Sample Collection and Preservation

5.1.2.1 Summary of Method

The water chemistry samples will be analyzed for a variety of parameters (e.g. phosphorus, nitrates, heavy metals, total suspended solids (TSS), etc.). There are four different bottles that are used for water chemistry analysis at UCASE sites. All UCASE water chemistry samples should be submitted to the State of Utah Health Lab using the following bottles and parameter requests:

- Chemistry: CL, TDS, TSS, TVS
- Non-Filtered Nutrients: NO32, NH3, TN TPO4
- Filtered Nutrients: NO32, TN, TPO4
- Filtered Metals: HG, AG, AL, AS, B, BA, BE, CA, CD, CR, CU, FE, MG, MN, NI, PB,SE, ZN
- CH-A (Water): PHEO (water column chlorophyll)

All water chemistry samples need to immediately be put on ice in a cooler after being collected. If having a cooler near the site is not possible, samples can be put in a small low-flowing pool, preferably shaded, while the rest of the sampling activities occur. Crew members should make sure the samples are placed in a way where they will not get swept away while sampling.

5.1.2.2 Equipment and Supplies

All samples need to be labeled correctly prior to taking the sample using an indelible pen or marker. Wearing nitrile gloves while sampling is not required, but they can be used if the person collecting the samples chooses to. It is necessary to fill a transfer bottle in order to filter and fill sample water into the filtered nutrients and filtered metals bottles. This bottle can be used multiple times for filtering at different sites. A triple rinse is required before filling at the next site. An amber bottle (or something that does not allow light penetration) must be used to collect the water column chlorophyll sample. This bottle can be used at multiple sites, but needs to be rinsed 3 times with site water before

filling. Procedures for filtering and processing these samples is discussed further in Section 6.

5.1.2.3 Sampling Procedure

Table 5-3 presents step-by-step procedures for collecting water chemistry samples at wadeable stream sites. Water chemistry samples should always be sampled right away once the reach has been located. Crew members should never wait to collect water chemistry samples at any time while sampling the site. These samples should be taken upstream concurrently with the multi-parameter unit. Whoever is collecting water samples must be cognizant of the location of their fellow crew members to assure they are not disrupting the site upstream of the sampling location. Crew members should always make sure no one goes above the water chemistry collection point until all samples have all been collected. Once samples have been collected, record which bottles were, or were not (if applicable) collected on the *Water Chemistry Sample Collection Form* (Figure 5-3).

Table 5-3. Sampling procedure for water chemistry sample collection

1. Correctly fill-in bottle labels with an indelible pen or marker as per the Monitoring Program SOP.
2. Taking a chemistry bottle, non-filtered nutrient bottle, and a transfer bottle wade to a flowing portion of the stream.
3. Unscrew the lid of the chemistry bottle making sure not to touch the inside of the bottle or cap with your hands, and dip and fill the bottle in the stream.
4. Screw the cap back on the bottle.
5. Follow step 3 and 4 for the non-filtered nutrient sample bottle.
6. Rinse both the transfer bottle and amber bottles, along with their caps 3 times, with stream water and discard the rinse downstream.
7. Fill and cap the transfer and amber bottles.
8. If site is not near a vehicle, place all bottles in a shaded pool where they will not get swept away while sampling activities continue to occur. Once crew arrives back to a vehicle continue on to step 10.
9. Leave the stream and go back to the vehicle where the filtered bottles and pump are located.
10. Properly set up a geo-pump using the glass-fiber filters (GFF) and filter water from the sample bottle into the filtered metals and filtered nutrients bottles individually.
11. The water-column sample in the amber bottle will be processed later when the periphyton samples are processed.
12. Put all samples on ice in a cooler and dump out the water from the transfer bottle. The transfer bottle can be re-used for the next site.
13. Record which bottles were collected on the *Water Chemistry Sample Collection Form* (Figure 5-3).

Figure 5-3. Water chemistry and macroinvertebrate collection form

UCASE WATER CHEMISTRY AND MACROINVERTEBRATE SAMPLE COLLECTION FORM-WADEABLE STREAMS

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ ___ DATE:___ ___/___ ___/ 2 0 ___ ___

WATER CHEMISTRY

Sample Bottle	Sample Collected?	Sample Bottle	Sample Collected?	Process: See protocol Preservation: Place water chem on ice and chlorophyll-*a* on dry ice Lab: State of Utah Health Lab within one week of collection. Shop storage: Fridge and freezer	Comments/Flags
Total Chemistry	Y / N	Filtered Metals	Y / N		
Non-Filtered Nutrient	Y / N	Chlorophyll-*a* (water column)	Y / N		
Filtered Nutrient	Y / N		Volume filtered: _____mL		

TARGETED BENTHOS SAMPLE

No. of Jars (Primary)	Was a Replicate Sample Taken (if Yes, record no. of jars?	Collection Method (Choose one)	Comments/Flags
___ ___	Y / N ___ ___	○D-net ○Modified surber ○Other (indicate in comments)	

TRANSECT:		____		____		____		____	
Dom. Substrate	Channel	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.
Fine/Sand	Pool	○F	○P	○F	○P	○F	○P	○F	○P
Gravel	Glide	○G	○GL	○G	○GL	○G	○GL	○G	○GL
Coarse	Riffle	○C	○RI	○C	○RI	○C	○RI	○C	○RI
Other: Note in Comments	Rapid	○O	○RA	○O	○RA	○O	○RA	○O	○RA

SUBSTRATE SIZE CLASSES
F/S – ladybug or smaller (<2 mm)
G – ladybug to tennis ball (2 to 64 mm)
C – tennis ball to car sized (64 to 4000 mm)
O – bedrock, hardpan, wood, vegetation, leaf litter, undercut, macrophyhtes, etc.

		____		____		____		____	
		Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.
		○F	○P	○F	○P	○F	○P	○F	○P
		○G	○GL	○G	○GL	○G	○GL	○G	○GL
		○C	○RI	○C	○RI	○C	○RI	○C	○RI
		○O	○RA	○O	○RA	○O	○RA	○O	○RA

Note:
- Always perform 8 kicks at every site.
- Target riffle habitat primarily. If riffles are scarce or absent (e.g. low gradient, beaver ponds, etc.), target edge habitats (e.g. overhanging veg., undercut banks, etc.) and mark "O" in the substrate column and explain situation in comments.
- If riffles are present, but scarce, multiple kicks can be performed at the same riffles throughout the reach.
- If kicks are made in beaver influenced areas, please explain in the comments section.

Process: Fill a clean wide-mouth bottle with ~40% of composite sample (use multiple bottles if necessary).
Preservation: Fill rest of bottle with denatured alcohol (EtOH) and seal lid with electrical tape. Keep secure in upright position.
Lab: Utah State University Bug Lab; submit in large batch at end of field season.
Shop storage: Store on shelf with other bug samples.

BENTHOS COMMENTS:

Updated: 01/2013

5.2 Physical Habitat Characterization – Wadeable Streams

Physical habitat (pHab) in streams includes all those physical attributes that influence or provide sustenance to organisms within the stream. The physical habitat of a stream varies naturally, thus expectations differ even in the absence of anthropogenic disturbance. Within a given physiographic-climate region, stream drainage area and overall stream gradient are likely strong natural determinants of many aspects of stream habitat. This is because of their influence on discharge, flood stage, and stream power (the product of discharge times gradient). Kaufmann (1993) identified seven general physical habitat attributes important to influencing stream ecology:

- Channel dimensions
- Channel gradient
- Channel substrate size and type
- Habitat complexity and cover
- Riparian vegetation cover and structure
- Anthropogenic alterations
- Channel-riparian interaction

5.2.1 Components of Physical Habitat Characterization

There are five components of the physical habitat characterization process (Table 5-4). Measurements are recorded on 11 copies of a two-sided field form and separate forms for recording slope and bearing measurements, assessing the degree of channel constraint, and recording evidence of debris torrents or recent major flooding. The *thalweg profile* is a longitudinal survey of depth, habitat class, presence of deposits of soft/small sediments, and presence of off channel habitats at 100 equally spaced stations (150 for streams ≤3 m wide) along the centerline between the two ends of the sampling reach. *Thalweg* refers to the main flow path of the deepest water in a stream channel (this does not necessarily mean the deepest point of the channel). Wetted width is measured and substrate size is evaluated at 21 equally spaced cross-sections (at 11 regular transects [A through K] and 10 supplemental cross-sections spaced midway between each of these). Data for the second component, the *woody debris tally*, are recorded for each of 10 segments of stream located between the 11 regular transects. The third component, the *channel and riparian characterization*, includes measures and/or visual estimates of channel dimensions, substrate, fish cover, bank characteristics, riparian vegetation structure, and evidence of human disturbances. These data are obtained at each of the 11 equally-spaced transects established within the sampling reach. In addition, measurements of the stream slope and compass bearing between stations are obtained, providing information necessary for calculating reach gradient, residual pool volume, and channel sinuosity. The fourth component, *assessment of channel constraint, debris torrents, and major floods*, is an overall assessment of these characteristics for the whole reach, and is undertaken after the other components are completed. Historically, a form dedicated to large (legacy) trees was included in all site packets, but in 2015 it was decided that this form was not necessary for current assessment purposes.

Table 5-4. Components of physical habitat characterization

COMPONENT	DESCRIPTION
Thalweg Profile (Section 5.2.4)	• Measure maximum depth, classify habitat pool-forming features, and check presence of backwaters, side channels and loose, soft deposits of sediment particles at 10-15 equally spaced intervals between each of 11 transects (100 or 150 individual measurements along entire reach). • Measure wetted width and evaluate substrate particle size classes at 11 cross-section transects and midway between them (21 width measurements and substrate cross-sections).
Woody Debris Tally (Section 5.2.4.2)	• Between each of the channel cross-sections, tally large woody debris numbers within and above the bankfull channel according to specified length and diameter classes (10 separate tallies).
Channel and Riparian Characterization (Section 5.2.5)	• At 11 transects (21 for substrate size) placed at equal intervals along reach: • Measure: channel cross-section dimensions, bank height, bank undercut distance, bank angle, slope and compass bearing (backsight), and riparian canopy density (densiometer). • Visually categorize[a]: substrate size class and embeddedness; areal cover class and type (e.g. woody trees) of riparian vegetation in Canopy, Mid-Layer and Ground Cover; areal cover class of fish concealment features, aquatic macrophytes and filamentous algae. • Observe and Record[a]: Presence and proximity of human disturbances.
Assessment of Channel Constraint, Debris Torrents, and Major Floods (Section 5.2.6)	• After completing thalweg and transect measurements and observations, identify features causing channel constraint, estimate the percentage of the channel margin that is constrained for the whole reach, and estimate the ratio of bankfull/valley width. Check evidence of recent major floods and debris torrent scour and deposition.
Discharge (Section 5.3)	• Measure water depth and velocity at equally spaced intervals across one carefully chosen channel cross-section.

[a] Substrate size class is estimated for a total of 105 particles taken at 5 equally-spaced points along each of 21 cross-sections. Depth is measured and embeddedness estimated for the 55 particles located along the 11 regular transects A through K. Cross-sections are defined by using a measuring tape or rangefinder to span the wetted channel. Woody debris is tallied over the distance between each cross-section and the next cross-section upstream. Riparian vegetation and human disturbances are observed 10m upstream and 5m downstream from the cross-section transect. They extend shoreward 10m from left and right banks. Fish cover types, aquatic macrophytes, and algae are observed within the channel 10m upstream and 10m downstream from the cross-section stations. These boundaries for visual observations are estimated by eye.

5.2.2 Habitat Sampling Locations within the Reach

Measurements are made at two scales of resolution along the length of the reach; the results are later aggregated and expressed for the entire reach, a third level of resolution. Figure 5-4 illustrates the locations within the reach where data for the different components of the physical habitat characterization are obtained. Many channel and riparian features are characterized on 11 cross-sections and pairs of riparian plots spaced at 4 channel-width intervals (i.e. transect spacing = $1/10^{th}$ the total reach length). The thalweg profile measurements must be spaced evenly over the entire support reach. In addition, they must be sufficiently close together that they do not miss deep areas and major habitat units. Follow these guidelines for choosing the increment between thalweg profile measurements:

- Channel widths up to 3.5 m = increment of 1.0 m
- Channel width ≥3.5 m = increments of 0.01 x (reach length)

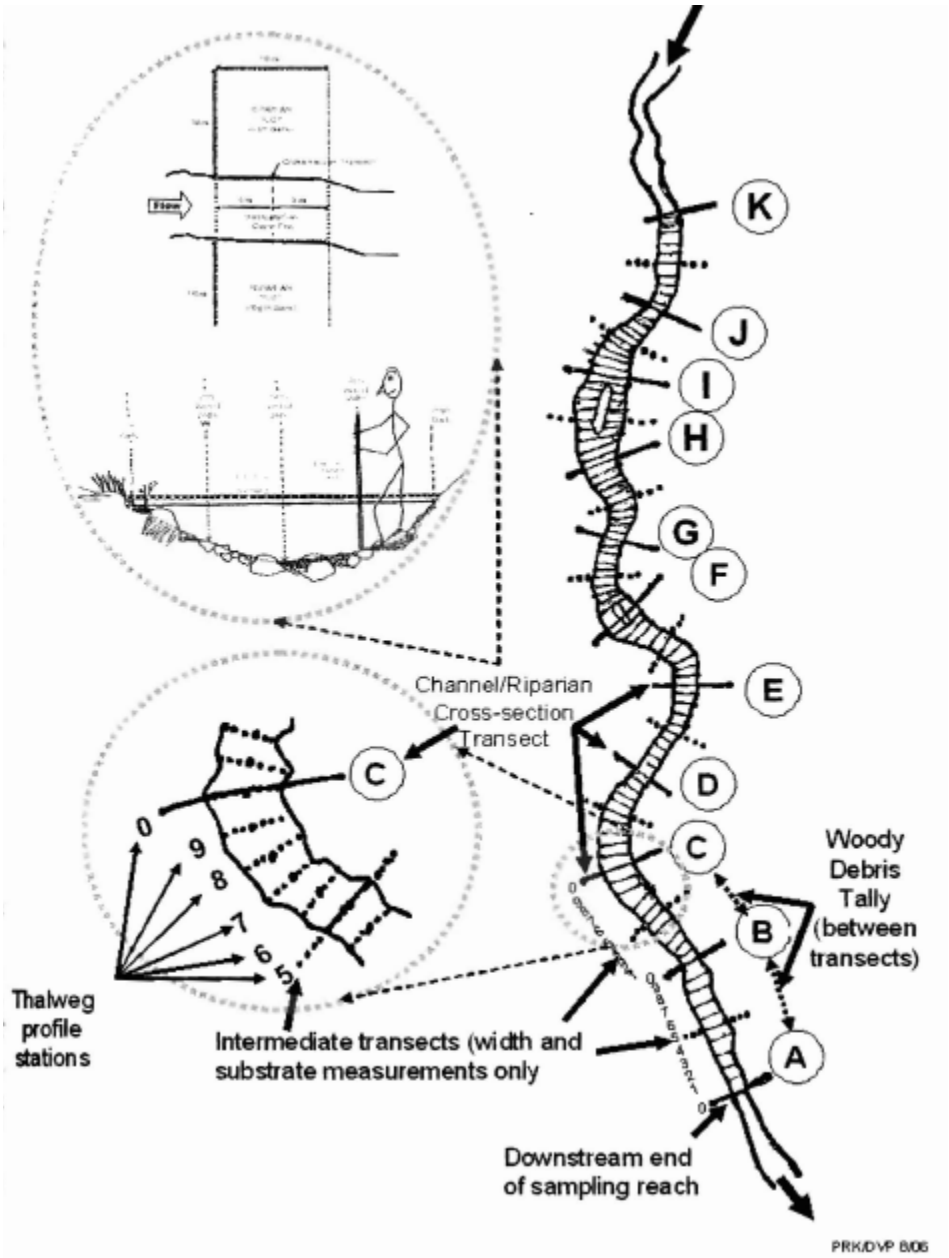
Following these guidelines, make 150 evenly spaced thalweg profile measurements in the smallest category of streams, 15 between each detailed cross-section. In all of the larger stream sizes (≥3.5 m), crews will make 100 measurements, 10 between each cross-section.

5.2.3 Logistics and Work Flow

The five components (Table 5-4) of the habitat characterization are organized into five grouped activities:

1. *Thalweg Profile and Large Woody Debris Tally* (Section 5.2.4). Two people proceed upstream from the downstream end of the sampling reach (Figure 5-4) making observations and measurements at the chosen increment spacing. One person is in the channel making width and depth measurements and determining whether soft/small sediment deposits are present. This person also classifies the channel habitat, indicates presence/absence of side channel and off-channel habitats (e.g. backwater pools, slough, alcoves, etc.). This person verbally calls out these measurements so that the other crew member can properly record them. The other crew member records measurements as they are called out and tallies large woody debris. Each time this team reaches a flag marking a new cross-section transect, they start filling out a new copy of the *Thalweg Profile and Woody Debris Form*. They interrupt the thalweg profile and woody debris tallying activities to complete data collection at each cross-section transects as it comes. When the crew member in the water makes a width measurement at channel locations midway between regular transect (i.e. A, B, K) he/she also locates and estimates the size class of the substrate particles on the left channel margin (0%) and at positions 25%, 50%, 75%, and 100% of the distance across the wetted channel. Procedures for this substrate tally are the same as for those at regular cross-sections, but data are recorded on the thalweg profile side of the field form.

Figure 5-4. Reach layout for pHab measurements (plan view)



2. *Channel/Riparian Cross-Sections* (Section 5.2.5). One person proceeds with the channel cross-section dimension, substrate, bank, and canopy cover measurements. The second person records those measurements on the *Channel/Riparian Cross-section Form* while making visual estimates of riparian vegetation structure, in-stream fish cover, and human disturbance specified on that form.

3. *Slope and Bearing* (Section 5.2.5.1). On a typical three person crew the bearing portion of the Slope and Bearing sheet are usually completed by the person who collects benthos and periphyton mainly mostly for efficiency reasons. As mentioned earlier in the manual, the exact order with how sheets are completed or which crew member fills them out does not matter just as long as they are filled out with completeness in the correct format. The crew members who do pHab together generally take slope readings after they are done taking pHab measurements at the entire site.

   Slope is measured by measuring the differences in elevation between each transect. Bearing is determined by back-sighting to the previous transect. Supplementary points may need to be located and flagged if the stream is extremely brushy, sinuous, or steep to the point that you cannot sight for slope and bearing measures between two adjacent transects.

4. *Channel Constraint and Torrent Evidence* (Section 5.2.6). After completing observations and measurements along the thalweg and at all 11 transects, the field crew completes the overall reach assessments of channel constraint and evidence of torrents/flash floods.

5. *Stream Discharge* (Section 5.3). Discharge measurements are made after collecting the water chemistry sample at the start of sampling the site. The flow measurement is done at a chosen optimal cross-section anywhere along the reach (does not necessarily have to be done at a particular transect).

5.2.4 Thalweg Profile and Large Woody Debris Measurements

5.2.4.1 Thalweg Profile

Thalweg refers to the **flow** path of the deepest water in a stream channel (not necessarily the deepest reading at a particular point, moreover, the reading is taken in the channel with the most flow volume). The thalweg profile is a longitudinal survey of the maximum flow path depth and several other selected characteristics at 100 or 150 equally spaced points (termed *stations*) along the length of the reach measured along the centerline of the channel. Data from the thalweg profile allows calculation of indices of residual pool volume, stream size, channel complexity, and the relative proportions of habitat types such as riffles and pools. One person walks upstream carrying a surveyor's rod (stadia rod or a calibrated rod (i.e. ski pole, broom handle, billiard cue, etc.) ranging from 1 m to 8 m). A second person on the bank or in the stream carries a clipboard with 11 copies of the field data forms.

The procedure for obtaining thalweg profile measurements is presented in Table 5-5. Record data on the *Thalweg Profile and Woody Debris Data Form* as shown in Figure 5-5. Use the surveyor's rod or calibrated rod to make the required depth and width measurements at each station and to measure the off distance between stations as you proceed upstream. You may need to make minor adjustments along each 10th measurement to be one increment short of the next transect. In streams with average wetted widths less than 3.4 m (round up or down when determining the average wetted width), make thalweg measurements at 1-meter increments. Because the minimum reach length is set at 150 m, there will be 15 measurements on a field data form: Station 0 at the transect, plus 14 additional stations between it and the next transect upstream. Use the five extra lines on the thalweg profile portion of the data form Figure 5-5 to record these measurements.

Table 5-5. Thalweg profile procedure

1. Determine the increment distance between measurement stations on the wetted width used to determine the length of the reach. Using a laser rangefinder or surveyor's rod:
    a. For widths ≤3.4 m establish stations every 1m (100 total).
    b. For widths ≥3.5 m establish stations at increments equal to 0.01 times the reach length (100 total).
2. Complete the header information on the *Thalweg Profile and Woody Debris Form*, noting the transect pair (downstream to upstream). Record the increment distance determined in Step 1 in the *Increment* field on the field data form.
3. Begin at the downstream end (*station 0*) of the first transect (*transect A*).
4. Measure the wetted width at station 0 and at either station 5 (if the stream width defining the reach length is ≥3.5 m) or at station 7 (if the stream width defining the reach is ≤3.4 m). Wetted width is measured across and over mid-channel bars and boulders. Record the width on the field data form to the nearest 0.1 m. For streams with interrupted flow where no water is in the channel at the station or transect, record zeros for wetted width.

NOTE: If a mid-channel bar is present at a station where wetted width is measured, measure the wetted width across including the bar to determine this value, but also measure the bar width separately and record it on the field data form.

5. At station 5 or 7 (see above) classify the size of the bed surface particle by physically reaching into the water. Whichever type of substrate is touched first should be written on the field form (crews members should not "guess" or "estimate" what the substrate is by simply looking at the stream bed and choosing what's most dominant. Smaller substrates tend to overlooked when this happens). If the water is deep where reaching into the stream to classify substrates is deeper than above the elbow or higher, crew members can use their surveyor's rod to classify substrates. Starting at either side of the wetted margin, substrates are classified at 0%, 25%, 50%, 75%, and 100% of the distance across the wetted width of the stream. This procedure is identical to the substrate size evaluation procedure described for regular cross-sections (transects A - K), except that for these midway supplemental cross-sections, substrate size is entered in the thalweg profile side of the field form.

6. At each thalweg profile station, use a calibrated pole or rod to located the deepest point within the deepest **flow path** (*the thalweg*), which may not always be found at mid-channel (and may not always be the absolute deepest point in every

Table 5-5. Thalweg profile procedure *(cont.)*

channel cross-section). Measure the thalweg depth to the nearest cm from the substrate surface to the water surface and record it on the thalweg profile form. Read the depth on the **side** of the rod to avoid inaccuracies due to the wave formed by the rod in moving water.

NOTE: For streams with interrupted flow – or there is not water at a transect, record zeros for depth.

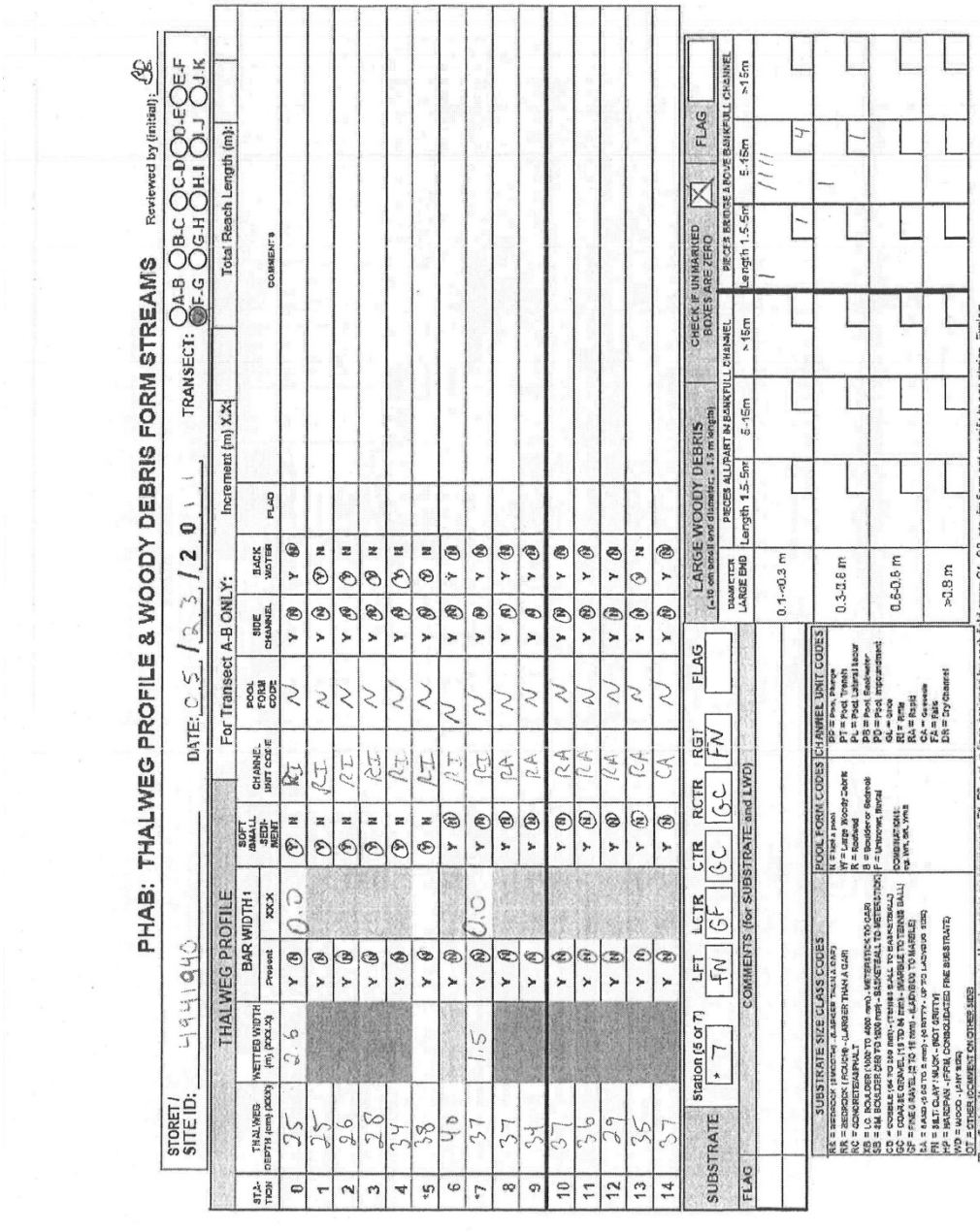
NOTE: Obtain thalweg depths at all stations. If the thalweg is too deep to measure directly one of two options are possible: 1) Estimate the depth to the best of your abilities and flag it with a "U" flag and explain in the comments that the value was estimated; 2) leave the depth reading blank and flag it with a "K" flag and
explain in the comments that conditions were too deep or too swift to take the measurement.

7. At the point where the thalweg depth is determined, observe if unconsolidated, loose (*soft*) deposits of small diameter (≤16mm) sediments are present directly beneath your surveyor's rod or pole.
   Soft/ small particles are defined here as fine gravel, sand, silt, clay or muck readily apparent by "feeling" the bottom with your hand or the surveyor's rod. Record presence or absence in the *Soft/Small Sediment* field on the field data form. *Note: A thin coating of fine sediment or silty algae coating the surface of cobbles should not be considered soft/small sediment. However, fine sediment coatings should be identified in the comments section of the field form when determining substrate size and type.*
8. Determine the channel unit code and pool forming element codes for the station. Record these on the field data form using the standard codes provided. For dry and intermittent streams, where no water is in the channel, record habitat type as dry channel (DR).
9. If the station cross-section intersects a mid-channel bar, indicate the presence of the bar in the *Bar Width* field on the field data form.
10. Record the presence or absence of a side channel at the station's cross-section in the *Side Channel* field on the field data form.
    Record the presence or absence of quiescent off-channel aquatic habitats, including sloughs, alcoves, and backwater pools in the *Backwater* column of the field data form.
11. Proceed upstream to the next station and repeat steps 2 through 11.
12. Repeat steps 2 through 12 until your reach the next transect. At this point complete Channel/Riparian measurements at the new transect (Section 5.2.5). Then prepare a new *Thalweg Profile and Woody Debris Form* and repeat steps 2 through 12 for each of the reach segments until you reach the upstream end of the sampling reach (transect K). At transect K, you will have completed 10

copies of the Thalweg Profile and Woody Debris Form, one for each segment (A to B, B to C, etc.).

Measure thalweg depths at **all** stations. Missing depths at the end of the reach (e.g. due to the stream flowing into or out of a culvert or under a large pile of debris) can be tolerated, but those in the middle of the reach are more difficult to deal with. Flag any missing measurements using a "K" code and explain the reason in the comments section of the field data form.

Figure 5-5. Thalweg Profile and Woody Debris form



While recording the width and depth measurements and the presence of soft/small sediments, the second person evaluates and records the habitat class and the pool forming element (Table 5-6) applicable to each of the 100 (or 150) measurement points along the length of the entire reach. Make channel unit scale habitat classifications at the thalweg of the cross-section. The habitat unit itself must meet a minimum size criteria in addition to the qualitative criteria listed in Table 5-6. Before being considered large enough to be identified as a channel-unit scale habitat feature, the unit should be at least as long as the channel is wide. For instance, if there is a small deep (pool-like) area at the thalweg within a large riffle area, do not record it as a pool unless it occupies an area about as wide or long as the channel is wide. If a backwater pool **dominates the channel**, record *PB* as the dominate habitat unit class. If the backwater pool is a pool that **does not dominate** the main channel, or if it is an **off-channel** alcove or slough (large enough to offer refuge to small fishes), circle *Y* to indicate presence of a backwater in the *BACKWATER* column of the field form, but classify the main channel habitat unit type according to characteristics of the main channel. *Sloughs* are backwater areas having marsh-like characteristics such as vegetation, and *alcoves* (or *side pools*) are deeper areas off the main channel that are typically wide and shallow (Helm 1985, Bain and Stevenson 1999). When trying to identify the pool forming element for a particular pool, remember that most pools are formed at high flows, so you may need to look for elements that are dry at baseflow, but still within the bankfull channel (e.g. boulders or large woody debris).

Table 5-6. Channel unit and pool forming element categories

Channel Unit Habitat Classes [a]	
Class (Code)	**Description**
Pools: Still water; low velocity, a smooth, glassy surface, usually deep compared to other parts of the channel.	
Plunge pool (PP)	Pool at base of plunging cascade or falls
Trench pool (PT)	Pool-like trench in the center or along the banks of the stream
Lateral Scour Pool (PL)	Pool scoured along a bank
Backwater Pool (PB)	Pool separated from main flow off the side of the channel (large enough to offer refuge to small fishes). Includes sloughs (backwater with marsh characteristics such as vegetation), and alcoves (a deeper area off a wide and shallow main channel).
Impoundment Pool (PD)	Pool formed by impoundment above dam or constriction
Glide (GL)	Water moving slowly, with a smooth, unbroken surface. Low turbulence
Rapid (RA)	Water movement rapid and turbulent, surface with intermittent whitewater with breaking waves. Sound: continuous rushing, but not as loud as a cascade.
Cascade (CA)	Water movement rapid and very turbulent over steep channel bottom. Much of the water surface is broken in short, irregular plunges, mostly whitewater.
Falls (FA)	Free falling water over a vertical or near vertical drop into plunge, water turbulent and white over high falls. Sound: from splash to roar.
Dry channel (DR)	No water in channel or flow is submerged under the substrate (hyporheic flow).

[a] Note that in order for a channel habitat to be distinguished, it must be at least as wide or long as the channel is wide (except for off channel backwater pools, which are noted as present regardless of size).

5.2.4.2 Large Woody Debris Tally

Large Woody Debris (LWD) is defined here as woody material with a small end diameter of at least 10 cm (4 in.) **and** a length of at least (5 ft.). The procedure for tallying LWD is presented in Table 5-7 . The tally includes all pieces of LWD that are at least partially in the baseflow channel (Zone 1), in the bankfull channel (Zone 2, flood channel up to bankfull stage), or spanning above the bankfull channel (Zone 3), as shown in Figure 5-6. The bankfull channel is defined as the channel that is filled by moderate sized flood events that typically recur every one to two years. LWD in or above the bankfull channel tallied over the entire length of the reach , including the area between the channel cross-section transects. Pieces of LWD that are not at least partially within Zones 1, 2, or 3 are not tallied.

Table 5-7. Procedure for tallying LWD

Note: Tally pieces of LWD within each segment of stream while the thalweg profile is being determined. Include all pieces in the tally whose large end is found within the segment.

1. Scan the stream segment between the two cross-section transects where thalweg profile measurements are being made.

2. Tally all LWD pieces within the segment that are at least partially within the bankfull channel. Determine if a piece of LWD (*small end dimater ≥10 cm [4 in],* ***and*** *≥1.5 m [5 ft]*).

3. For each piece of LWD, determine the class based on the *diameter of the* ***large end*** (0.1 m to <0.3 m, 0.3 m to <0.6m, 0.6 m to <0.8 m, or >0.8 m), **and** the class based on the ***length*** *of the piece* (1.5 m to <5.0 m, 5 m to <15 m, or >15 m).

    - If the piece is not cylindrical, visually estimate what the diameter would be for a piece of wood with circular cross-section that would have the same volume.

    - When estimating length, include only the part of the LWD piece that has a diameter >10 cm (4 in.).

4. Place a tally mark in the appropriate diameter x length class tally box in the *Pieces All/Part in Bankfull Channel* section of the *Thalweg Profile and Woody Debris Form*.

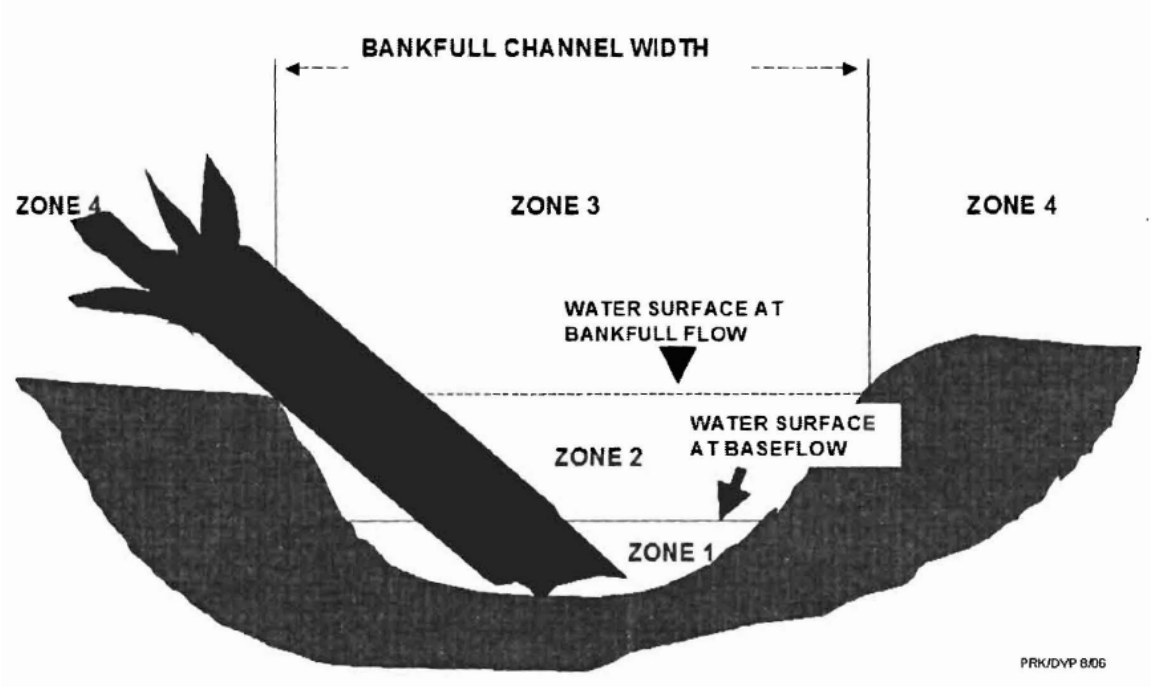
5. Tally all LWD pieces within the segment that are not actually within the bankfull channel, but are at least partially spanning (bridging) the bankfull channel. For each piece, determine the class based on the diameter of the **large end** (0.1 m to <0.3 m, 0.3 m to <0.6 m, 0.6 m to <0.8 m, or >0.8 m), **and** the class based on the **length** of the piece (1.5 m to <5.0 m, 5 m to <15 m, or >15 m).

6. Place a tally mark for each piece on the appropriate diameter x length class tally box in the *Pieces Bridge Above Bankfull Channel* section of the *Thalweg Profile and Woody Debris Form*.

7. After all pieces within the segment have been tallied, write the total number of pieces for each diameter x length class in the small box at the lower right-hand corner of each tally box.

8. Repeat steps 1 through 7 for the next stream segment, using a new *Thalweg Profile and Woody Debris Form.*

Figure 5-6. LWD influence zones (modified from Robison and Beschta).



5.2.5 Channel and Riparian Measurements at Cross-Section Transects

5.2.5.1 Slope and Bearing

Measure bearing by *sighting* between transects (e.g. transect B and A, C and B, etc.) as shown in Figure 5-7. To measure the bearing between adjacent transects, follow the procedure presented in Table 5-8. Record bearing data on the *Slope and Bearing Form* as shown in Figure 5-8. Slope and bearing measurements can be shot either from A-K or K-A, but the crew needs to make sure both measurements are done consistently with each other at the site. While filling the *Slope and Bearing Form*, crew members also need to be cognizant of which fields they are filling in depending on if they are taking measurements going upstream or downstream (if going upstream, the field-form needs to be filled in from top-bottom; if going downstream form gets filled from bottom-top). It is **highly recommended**, however, that crews shoot slope and bearing going upstream. Taking measurements downstream can be confusing to crew members who are not used to these measurements because the paper work has to be done backwards. Only experts who have taken these measurements several times and understand how the slope concept and field sheets work should sample in this direction.

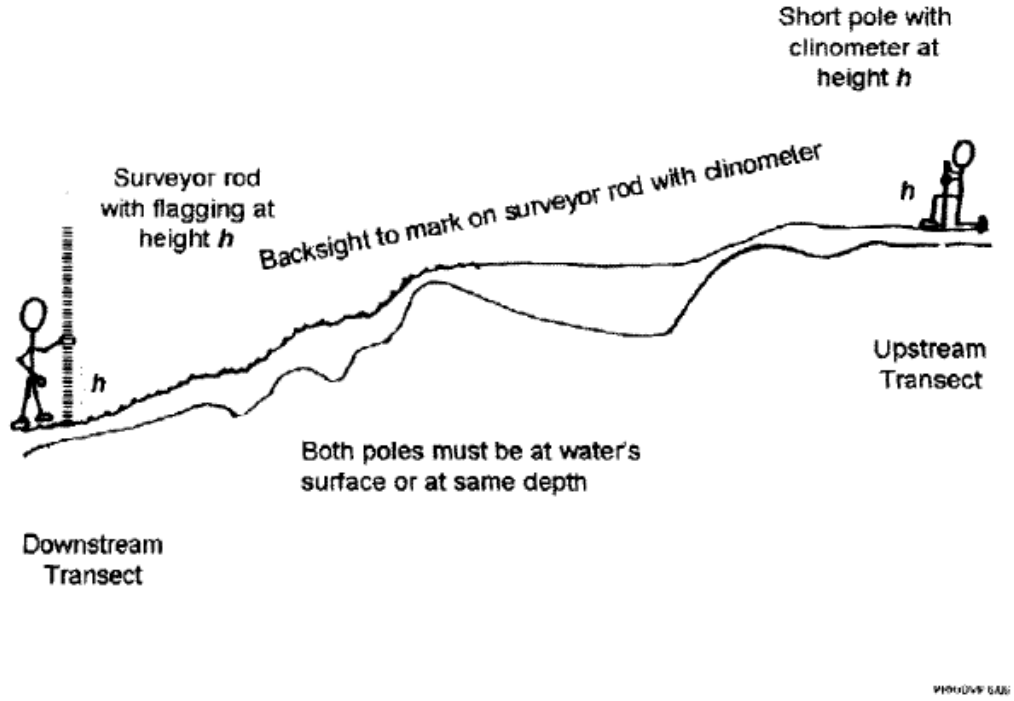
Slope is typically measured by two people, one holding a surveyor's rod and the second sighting through the surveyor's level. Be sure that the person is standing (or holding the marked pole) at the water's edge holding the rod at the surface of the water. The intent is to get a measure of the *water surface* slope, which may not necessarily be the same as the stream bed slope. The surveyor's level is leveled according to the manufacturer's

recommendation which is generally to adjust the three screw leveling feet until the bubble is centered. Level is checked in all planes to be measured. If the level does not "self-level" in all measured planes the user should check the instruction manual for suggested options. Elevation readings are made at each transect and the difference between each elevation reading is recorded as the change in elevation. NOTE: Multiple transect elevations can be made for each setup for the level, but every time the transit is moved it requires re-measuring the transect elevation from the last setup. You cannot use elevations from previous setups because the relative height of the transit has changed.

To calculate sinuosity from bearing measurements, it does not matter whether or not you adjust your compass bearings for magnetic declination, but it is important that you are **consistent** in the use of magnetic or true bearings throughout all the measurements you make on a given reach. Guard against recording *reciprocal bearings* (erroneous bearings 180 degrees from what they should be). The best way to do this is to know where the primary (cardinal) directions are in the field: (north [0 degrees], east [90 degrees], south [180 degrees], and west [270 degrees]), and ensure that your bearings "make sense."

As mentioned earlier, it may be necessary to set up intermediate (supplemental) slope and bearing points between a pair of cross-section transects if you do not have direct line-of-sight along (and within) the channel between stations (see Figure 5-7). This can happen if brush is too heavy or if there are sharp slope breaks or tight meander bends. *If you would have to sight across land to measure slope or bearing between two transects, then you need to make one or more supplemental measurements* (i.e. do not "short-circuit" a meander bend). Record these supplemental slope and bearing measurements, along with the proportion of the stream segment between transects included in each supplemental measurement, in the appropriate sections of the *Slope and Bearing Form* (Figure 5-8).

Figure 5-7. Channel slope and bearing measurements



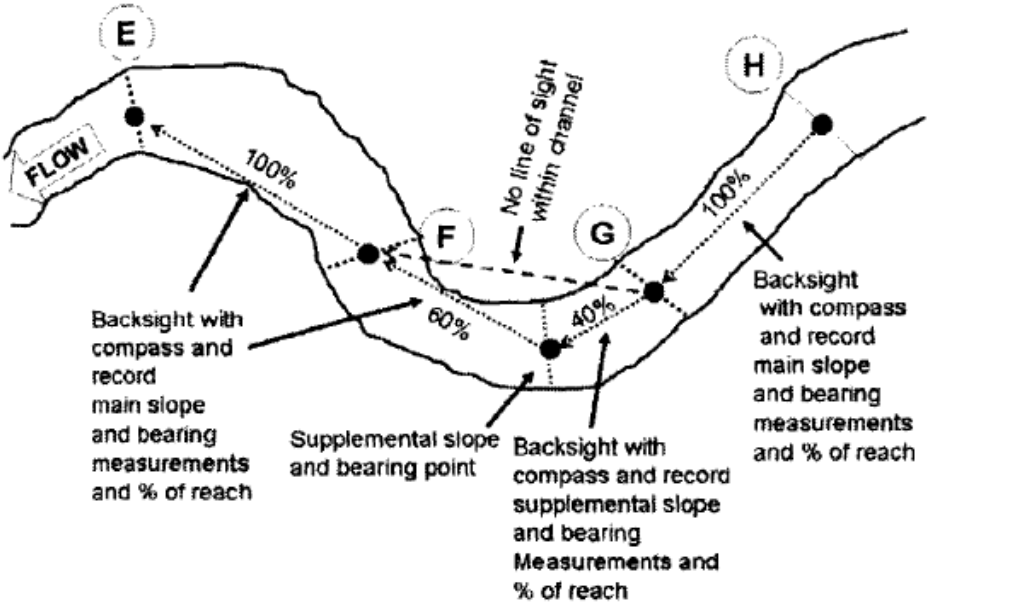
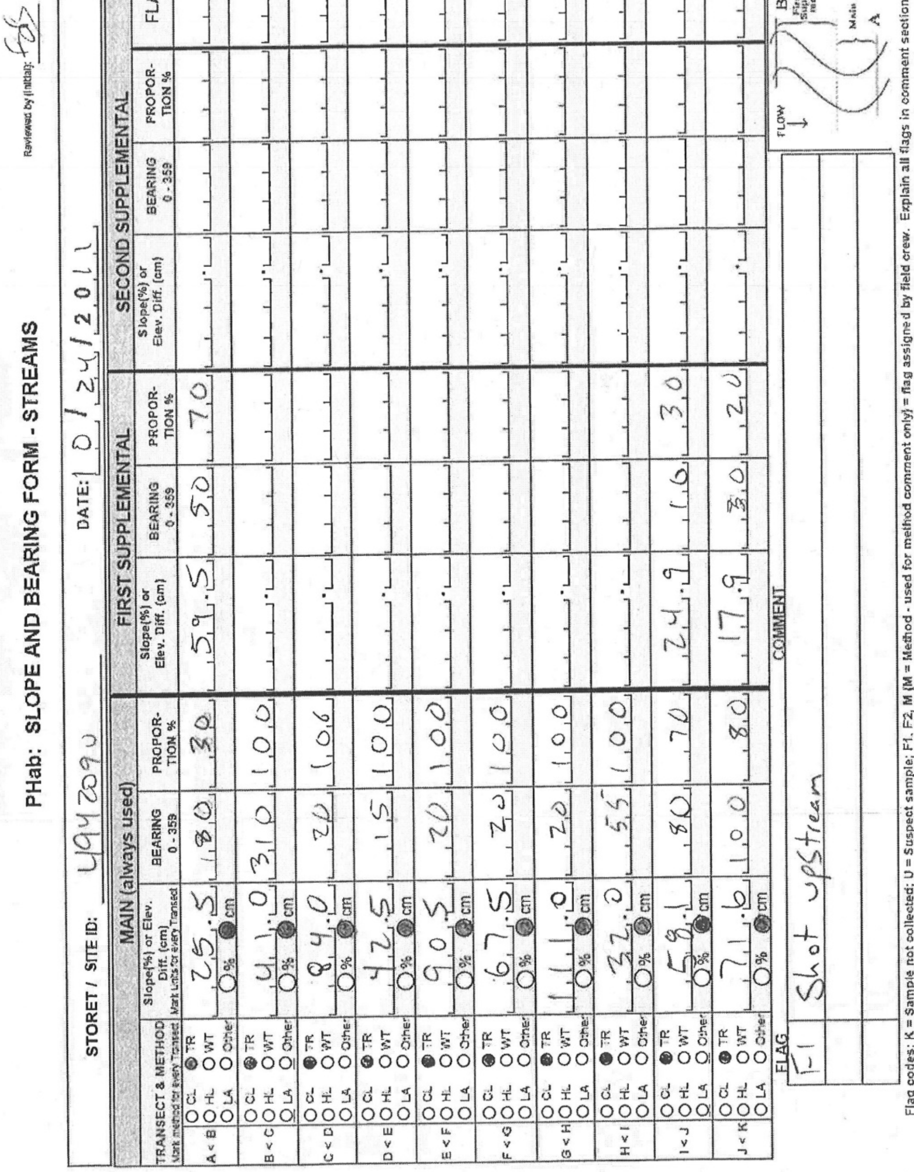


Figure 5-8. Slope and Bearing Form



PHab: SLOPE AND BEARING FORM - STREAMS

Reviewed by (initial): FGS

STORET / SITE ID: 4942090 DATE: 10/24/2011

TRANSECT & METHOD (Mark method for every Transect)	MAIN (always used) Slope(%) or Elev. Diff. (cm) (Mark Units for every Transect)	BEARING 0 - 359	PROPORTION %	FIRST SUPPLEMENTAL Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPORTION %	SECOND SUPPLEMENTAL Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPORTION %	FLAG
A < B (● TR)	25.5 (● cm)	180	30	59.5	50	70				
B < C (● TR)	41.0 (● cm)	310	100							
C < D (● TR)	84.0 (● cm)	20	106							
D < E (● TR)	42.5 (● cm)	15	100							
E < F (● TR)	90.5 (● cm)	20	100							
F < G (● TR)	67.5 (● cm)	20	100							
G < H (● TR)	111.0 (● cm)	20	100							
H < I (● TR)	32.0 (● cm)	55	100							
I < J (● TR)	58.1 (● cm)	80	70	24.9	10	30				
J < K (● TR)	71.6 (● cm)	100	80	17.9	30	20				

Method options: ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other; Units: ○ % ○ cm

FLAG	COMMENT
F1	Shot upstream

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, M (M = Method - used for method comment only) = flag assigned by field crew. Explain all flags in comment sections

03/2011 UCASE Phab Slope CL=Clinometer; HL=Hand Level; LA=Laser rangefinder with electronic clinometer; TR=Transit, surveyors level or total station; WT=Water Tubing.

Crews should make using a transit (and tripod) for this measurement as a standard. Other tools are listed on the *Slope and Bearing Form* (Figure 5-8), which are an option to use (i.e. laser and clinometer), but using a transit is the current preferred tool for this measurement. Clinometers are often more desired due to ease of portability, but due to inaccuracies with this particular measurement in the past they are not recommended. Nonetheless, if a transit is not available other methods can be used. Mark the appropriate method circle (identified in Figure 5-8) as well as the appropriate unit circle (cm or %). If the instrument measures change in elevation, *cm* should be used, if percent slope then *%* should be used.

Table 5-8. Procedure for obtaining slope and bearing data

1. Determine a location at transect A to hold a surveyor's rod that will be visible from a point between transect A and transect B:
   a) Set up the instrument at a point approximately halfway between points A and B and where a clear line of site is possible.
   b) Position the staff at point A, holding the bottom of the staff at the water level and the staff as vertical as possible and the numbers facing the instrument.
   c) Site the staff and record the reading to nearest centimeter.
   d) Move the staff to transect B and gently swivel the instrument to face the next reading. Hold the staff as before, vertically, with the bottom at the water level and the numbers facing the instrument.
   e) Site the staff and record the reading to the nearest centimeter.
   f) Repeat measurements between each transect.
   g) The difference in the readings is the height difference or gradient.

Note: In small streams with a clear line of site it may be possible to set the instrument up once and make readings to several transects from a single set up. Simply record the readings for each transect and do not skip transects.

- If you are backsighting from a supplemental point, record the bearing in the appropriate *Supplemental* section of the *Slope and Bearing Form*.

2. Proceed to the next cross-section transect (or supplementary point) and repeat steps a-g above.

Instrument Setup:

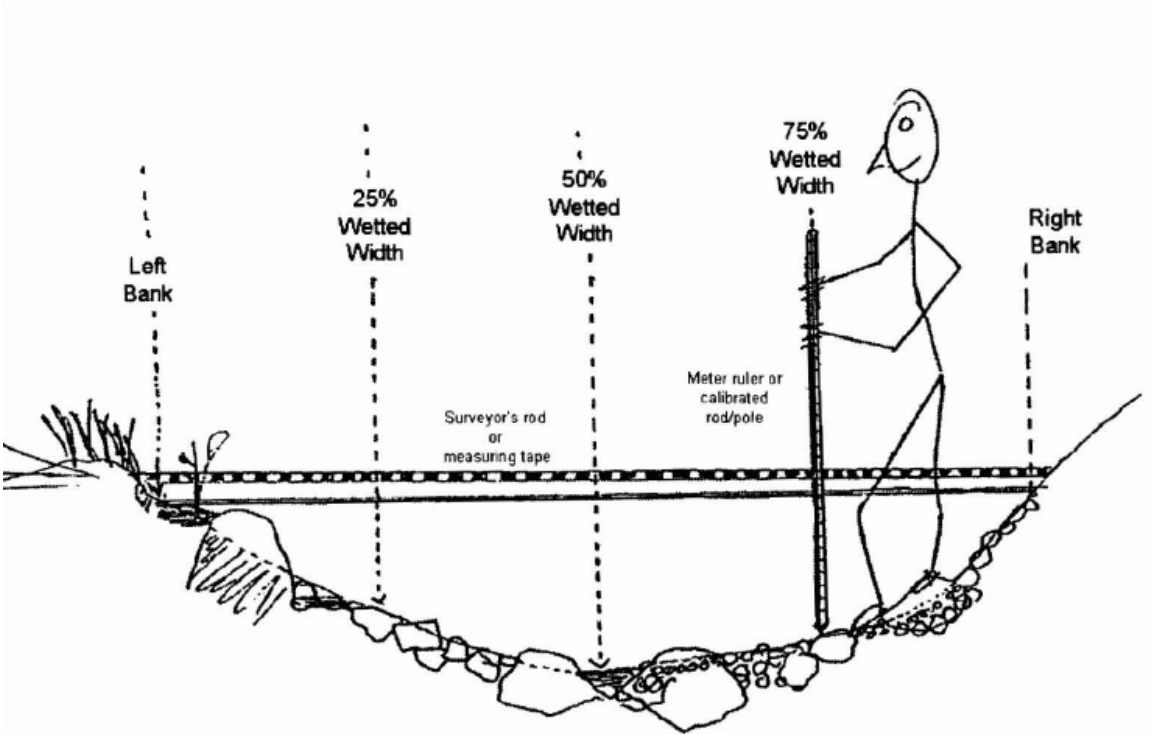
a) Extend the tripod legs to approximately eye level and set the legs firmly into the ground; adjust the legs so that they form a regular triangle and are firmly set with no wobble.
b) Hold the instrument in the tripod and start the centering screw. Ensure the adjustable feet are roughly evenly adjusted. While the centering screw is still loose slide the instrument on the base plate until the bubble is approximately centered in the circular level. Tighten the centering screw.
c) Adjust the leveling foot screws until the bubble is exactly level in the center circle.
d) The self leveling instrument can now be swiveled gently on the base plate and maintain level as long the tripod remains steady.
e) Adjust focus, brightness and parallax according to manufactures specifications.
f) The instrument is ready to make measurements.

Method codes are: CL=clinometer, TR=transit, HL=hand level, WT=water tube, LA=laser level, Other=method not listed (described in commencts section of form.

5.2.5.2 Substrate Size and Channel Dimensions

Substrate size and embeddedness are evaluated at 5 points at each of the 11 transects (refer to Figure 5-9). Substrate size is also evaluated at 10 additional cross-sections located midway between each of the 11 regular transects (A-K). In the process of measuring substrate particle sizes at each channel cross-section, the wetted width of the channel and the water depth at each substrate sample point are measured (at the 10 midway cross-sections, only substrate size and wetted width are recorded). If the wetted channel is split by a mid-channel bar, the five substrate points are centered between the wetted width boundaries regardless of the mid-channel bar in between. Consequently, substrate particles selected in some cross-sections may be "high and dry." *For cross-sections that are entirely dry, make measurements across the* ***unvegetated portion*** *of the channel*.

Figure 5-9. Substrate sampling cross-section



The substrate sampling points along the cross-section are located at 0, 25, 50, 75, and 100 percent of the measured wetted width, with the first and last points located at water's edge just within the left and right banks. The procedure for obtaining substrate measurements is described in Table 5-9 (including all particle size classifications). Record these measurements on the *Channel/Riparian Cross-section* side of the field form as shown in Figure 5-10. For the supplemental cross-sections midway between regular transects, record substrate size and wetted width data on the thalweg profile side of the field form. To minimize bias in selecting a substrate particle size classification, the person taking substrate measurements should submerge their hand into the water and actually touch the substrate whenever possible. The first particle that is touched is to be recorded on the data sheet (crew members should avoid determining this field by simply looking at what substrate is common in the area). If the water is too cold or deep, crews should use the tip of their surveyor's rod to determine substrate classifications. It is important to concentrate on correct placement of the measuring stick along the cross-section and to select the particle right at the bottom of the stick (not, for example, a more noticeable large particle that is just to side of the stick). Classify the particle into one of the size classes on the field data form (Figure 5-10) based on the middle dimension of its length, width, and depth. This *median dimension* determines the sieve size through which the particle can pass. When you record the size class as *Other*, assign an *Fn* flag on the field data form and describe the substrate type in the comments section of the field form as shown in Figure 5-10.

At substrate sampling locations on the 11 regular transects (A-K), examine particles larger than sand for surface stains, markings, and algal coatings to estimate embeddedness of all particles in the 10 cm diameter circle around the substrate sampling point. Embeddedness is the fraction of a particle's volume that is surrounded by (embedded in) sand or finer sediments on the stream bottom. By definition, record the embeddedness of sand and fines (silt, clay, and, muck) as *100 percent*, and record the embeddedness of hardpan and bedrock as *0 percent.*

Table 5-9. Substrate measurement procedure

1. Fill in the header information on page 1 of a *Channel/Riparian Cross-section Form*. Indicate the cross-section transect. At the transect, visually divide the wetted channel by 4 to locate substrate measurements points on the cross-section. In the *DistLB* fields of the form, record the distances corresponding to 0% (LFT), 25% (LCtr), 50% (CTR), 75% (RCtr), and 100% (RGT) of the measured wetted width. Record these distances at Transects A-K, but just the wetted width at midway cross-sections.

NOTE: If a side channel is present and contains 15% – 49% of the total flow, establish a secondary cross-section transect. Use a separate field data form to record data for the side channel, designating it as a secondary transect by marking both the *X-tra Side Channel* circle and the associated primary transect letter (e.g. XA, XB, etc.). Collect all channel and riparian cross-section measurements from the side channel (See Section 4.2.4)

2. Place your calibrated pole at the LFT location (0 m). Measure the depth and record it on the field data form (cross-section depths are measured only at regular transects A-K, not at the 10 midway cross-sections).
    - Depth entries at the left and right banks may be 0 if the banks are gradual.
    - If the bank is nearly vertical, let the base of the measuring stick fall to the bottom (i.e. the depth at the bank will be >0 cm), rather than holding it suspended at the water surface.
3. Submerge your hand in the water, without targeting any one particle, at the same place the depth was taken and pick up the substrate particle that is at the base of the meter stick (unless it is bedrock or boulder) and visually estimate its particle size according to the following table. If it is too deep or cold do your best with classifying substrates using the tip of your surveyor's rod. Classify the particle according to the **median diameter** (the middle dimension of its length, width, and depth). Record the size class code in the field data form (*Cross-section* side of form for transects A-K; special entry boxes on *Thalweg Profile* side of form for midway cross-sections).

Code	Size Class	Size Range (mm)	Description
RS	Bedrock (smooth)	>4000	Smooth surface rock bigger than a car
RR	Bedrock (rough)	>4000	Rough surface rock bigger than a car
HP	Hardpan	>4000	Firm, consolidated fine substrate
LB	Boulders (large)	>1000 to 4000	Yard/meter stick to car size
SB	Boulders (small)	>250 to 1000	Basketball to yard/meter stick size
CB	Cobble	>64 to 250	Tennis ball to basketball size
GC	Gravel (coarse)	>16 to 64	Marble to tennis ball size
GF	Gravel (fine)	>2 to 16	Ladybug to marble size
SA	Sand	>0.06 to 2	Smaller than ladybug size – gritty between fingers
FN	Fines	≤0.06	Silt Clay Muck (not gritty between fingers)
WD	Wood	Regardless of size	Wood and other organic particles
RC	Concrete	Regardless of size	Record size class in comment field
OT	Other	Regardless of size	Metal, tires, car bodies, etc. (describe in comments)

Table 5-9. Substrate measurement procedure *(cont.)*

4. Evaluate substrate embeddedness as follows at each transect. For particles larger than sand, examine the surface for stains, markings, and algae. Estimate the average % embeddedness of particles in the 10 cm circle around the measuring rod. Record this value on the field data form. For sand and smaller particles, you will not be able to pick up individual particles, but a "pinch" of fine particles between your fingers. Determine and record the dominant size of particles in the "pinch." By definition, sand and fines are embedded 100%; bedrock and hardpan are embedded 0%.

5. Move to the next location on the transect and repeat Steps 4-6 at each location. Repeat steps 1-6 at each transect, including any additional side channel transects established if islands are present.

5.2.5.3 Bank Characteristics

The procedure for obtaining bank and channel dimension measurements is presented in **Table 5-10.** Data are recorded in the Bank Measurements section of the Channel/Riparian Cross-section Form as shown in Figure 5-10. Bank angle and bank undercut distance are determined on the left and right banks at each cross-section transect. Figure 5-11 illustrates how bank angle is determined for several different situations. The scale at which bank angle is characterized is approximately 0.5 m. A short (approx. 1-m long) pole is used to determine bank angle (the same tool used for measuring depths can be used). The angle is determined based on the pole resting on the ground for about 0.5 m. Other features include the wetted width of the channel, the width of exposed mid-channel bars of gravel or sand, estimated incision height, and the estimated height and width of the channel at bankfull stage as described in Table 5-10. Bankfull height and incised height are both measured relative to the present water surface (i.e. the level of the wetted edge of the stream). This is done by placing the base of the measuring rod (stadia rod, ski pole, broom handle, etc.) at the edge of the wetted margin and measuring to the top of the bankfull and incised heights.

Figure 5-10. Channel/Riparian Cross-section Form

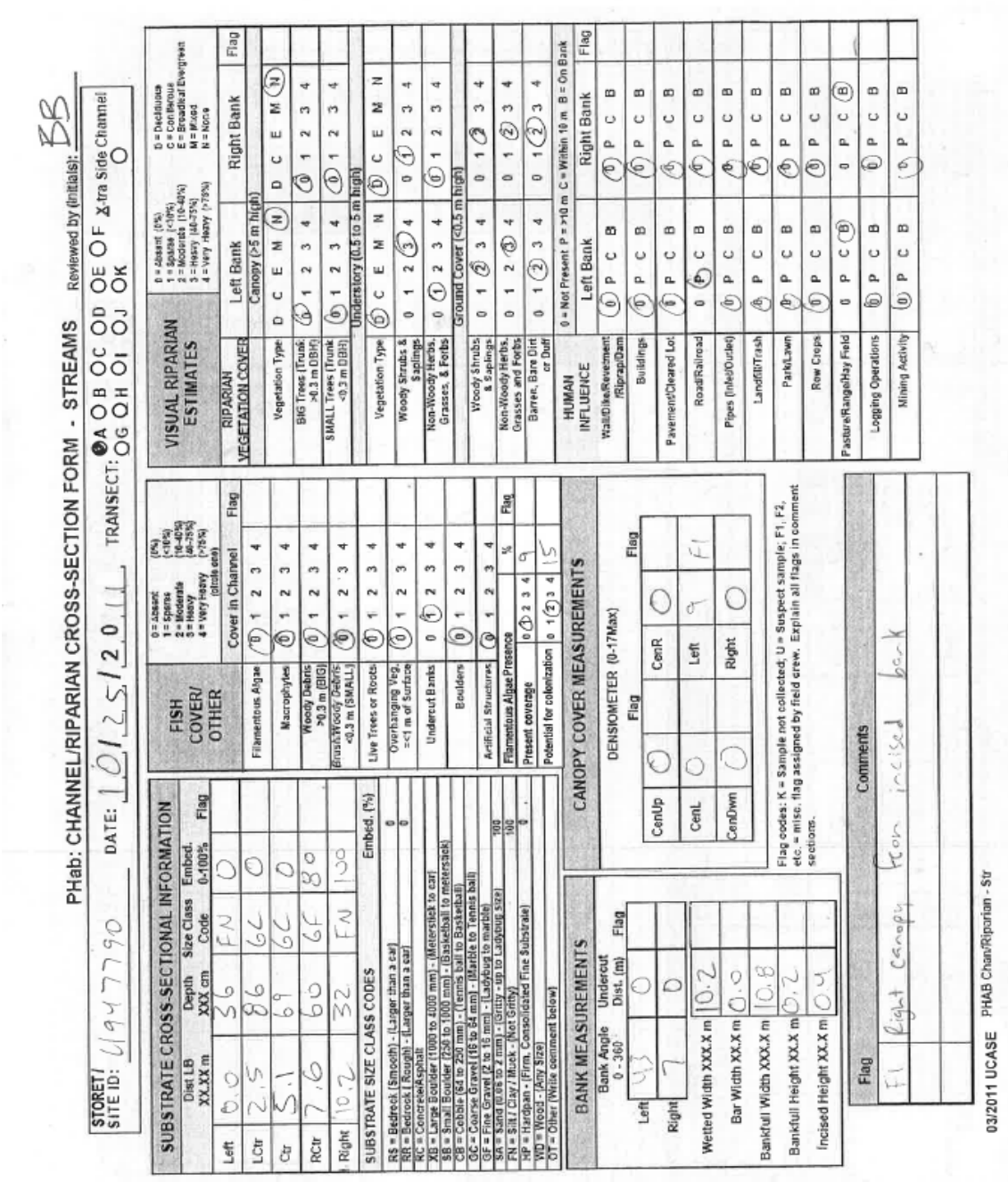


Table 5-10. Procedure for measuring bank characteristics

1. To measure bank angle, lay a rod of sorts (stadia rod, ski pole, broom handle, etc. approximately 1 m long) down against the left bank (determined as you face downstream) with one end at the water's edge. At least 0.5 m of the rod should be resting comfortably on the ground to determine bank angle. Lay the clinometer on the rod and read the bank angle in degrees from the external scale on the clinometer. Record the angle in the field for the left bank in the *Bank Measurement* section of the *Channel/Riparian Cross-section Form*.
    - A vertical bank is 90º, overhanging banks have angles >90° approaching 180°, and more gradually sloped banks have angles <90°. To measure bank angles >90°, turn the clinometer (which only reads 0 to 90°) over and subtract the angle reading from 180°.
    - If there is a large boulder or log present at the transect, measure angle at a nearby point where conditions are more representative.

2. If the bank is undercut, measure the horizontal distance of the undercutting to the nearest 0.01m. The undercut distance is the distance from the water's edge out to the point where a vertical plumb line from the bank would hit the water's surface. Record the distance on the field data form. Measure submerged undercuts by thrusting the rod into the undercut and reading the length of the rod that is hidden by the undercut.

3. Repeat steps 1 and 2 on the right bank.

4. Hold the surveyor's rod vertically with its base planted at water's edge. Examine both banks and measure the height of bankfull height above the present water level using the surveyor's rod (usually the first terrace above the water surface). Look for evidence on one or both banks such as:

    - An obvious slope break that differentiates the channel from a relatively flat floodplain terrace higher than the channel.
    - A transition from exposed stream sediments to terrestrial vegetation.
    - Moss growth on rocks along the bank.
    - Presence of drift material caught on overhanging vegetation.
    - A transition from flood- and scour-tolerant vegetation to that which is relatively intolerant of these conditions.

    Record the value in the *Bankfull Height* box on the bank measurement section of the field data form.

5. While still holding the surveyor's rod as a guide, examine both banks to measure and record the incised height from the water surface to elevation of the valley floodplain (Note: this is at or above the bankfull channel height). Often, the incised height is the next terrace above the bankfull height. Record this value in the *Incised Height* field of the bank measurements section of the field data form.

NOTE: Often, the incision on both the left and right bank may be at a different height. Always measure the lower of the two banks and record that value in the *Incised Height* field.

Table 5-10. Procedure for measuring bank characteristics *(cont.)*

6. Record the *wetted width* value determined when locating substrate sampling points in the *Wetted Width* field in the bank measurement section of the field data form. Also determine the *bankfull channel width* and the *width of exposed mid-channel bars* (if present). Record these values in the *Bank Measurement* section of the field data form.
7. Repeat Steps 1 through 6 at each cross-section transect, (including any additional side channel transects established when islands are present). Record data for each transect on a separate field data form.

Bankfull flows are large enough to erode the stream bottom and banks, but frequent enough (every 1 to 2 years) not to allow substantial growth of upland terrestrial vegetation. Consequently, in many regions, it has these flows that have determined the width and depth of the channel. Estimates of the bankfull dimensions of stream channels are extremely important in these surveys. They are used to calculate shear stress and bed stability (see Kaufmann et al., 1999). Unfortunately, we have to depend on evidence visible during low-flow sampling season. If available, consult published rating curves relating expected bankfull channel dimensions to stream drainage area within the region of interest. Graphs of these rating curves can help you get a rough idea of where to look for field evidence to determine the level of bankfull flows. Curves such as these are available from the USGS for some streams in Utah.
Estimate the bankfull flow level by looking at the following indicators:

- First look at the stream and its valley to determine the active floodplain. This is a depositional surface that frequently is flooded and experiences sediment deposition under the current climate and hydrological regime.
- Then look specifically for:
    - An obvious break in the slope of the banks.
    - A change from water-loving and scour-tolerant vegetation to more drought-tolerant vegetation.
    - A change from well-sorted stream sediments to unsorted soil materials.

In the absence of clear bankfull indications, consider the previous season's flooding as the best evidence available (note: you could be wrong if very large floods or prolonged droughts have occurred in recent years.). Look for:

- Drift debris ("sticky wickets" left by the previous seasons flooding).
- The level where deciduous leaf-fall is absent on the ground (carried away by previous winter flooding).
- Un-vegetated sand, gravel or mud deposits from previous year's flooding.

In years that have experienced large floods, drift material and other recent high flow markers may be much higher than other bankfull indicators. In such cases, base your determination on less-transient indicators such as channel form, perennial vegetation, and depositional features. In these cases, flag your data entry and also record the height of drift material in the comments section of the field data form.

We use the vertical distance (height) from the observed water surface up to the level of the first major valley depositional surface (Figure 5-12) as a measure of the degree of *incision or downcutting* of the stream below the general level of its valley. This value is recorded in the *Incised Height* field. It may not be evident at the time of sampling whether the channel is downcutting, stable, or aggrading (raising its bed by depositing sediment). However, by recording incision heights measured in this way and monitoring them over time, we will be able to tell if streams are incising or aggrading.

If the channel is not greatly incised, bankfull channel height and incision height can be the same (i.e. the first valley depositional surface is the active floodplain). However, if the channel is incised greatly, the bankfull level will be the level of the first terrace of the valley floodplain, making bankfull channel height less than incision height (Figure 5-13). Bankfull height is **never** greater than incision height. You may need to look for evidence of recent flows (within about a year) to distinguish bankfull and incision heights. In cases where the channel is cutting a valley sideslope and has oversteepened and destabilized that slope, the bare "cutbank" against the steep hillside at the edge of the valley is not necessarily an indication of recent incision. In such a case, the opposite bank may be lower, with a more obvious terrace above bankfull height; choose that bank for your measurement of incised height. Examine both banks more accurately to determine incision height and bankfull height. Remember that incision height is measured as *the vertical distance to the first major depositional surface above bankfull* (whether or not it is an active floodplain or a terrace). If terrace heights differ on left and right banks (both are above bankfull), choose the lower of the two terraces. In many cases your sample reach may be in a "V" shaped valley or gorge formed over eons and the slope of the channel banks simply extends uphill indefinitely, not reaching a terrace before reaching the top of a ridge (Figure 5-13). In such cases, record incision height values equal to bankfull levels and make appropriate comment that no terrace is evident. Similarly, when the stream has extremely incised into and ancient terrace, (e.g. stretches of the Escalante River, San Rafael River, Calf Creek, etc.) you may crudely estimate the terrace height if it is the first one above bankfull level. If you cannot estimate the terrace height, make appropriate comments describing the situation.

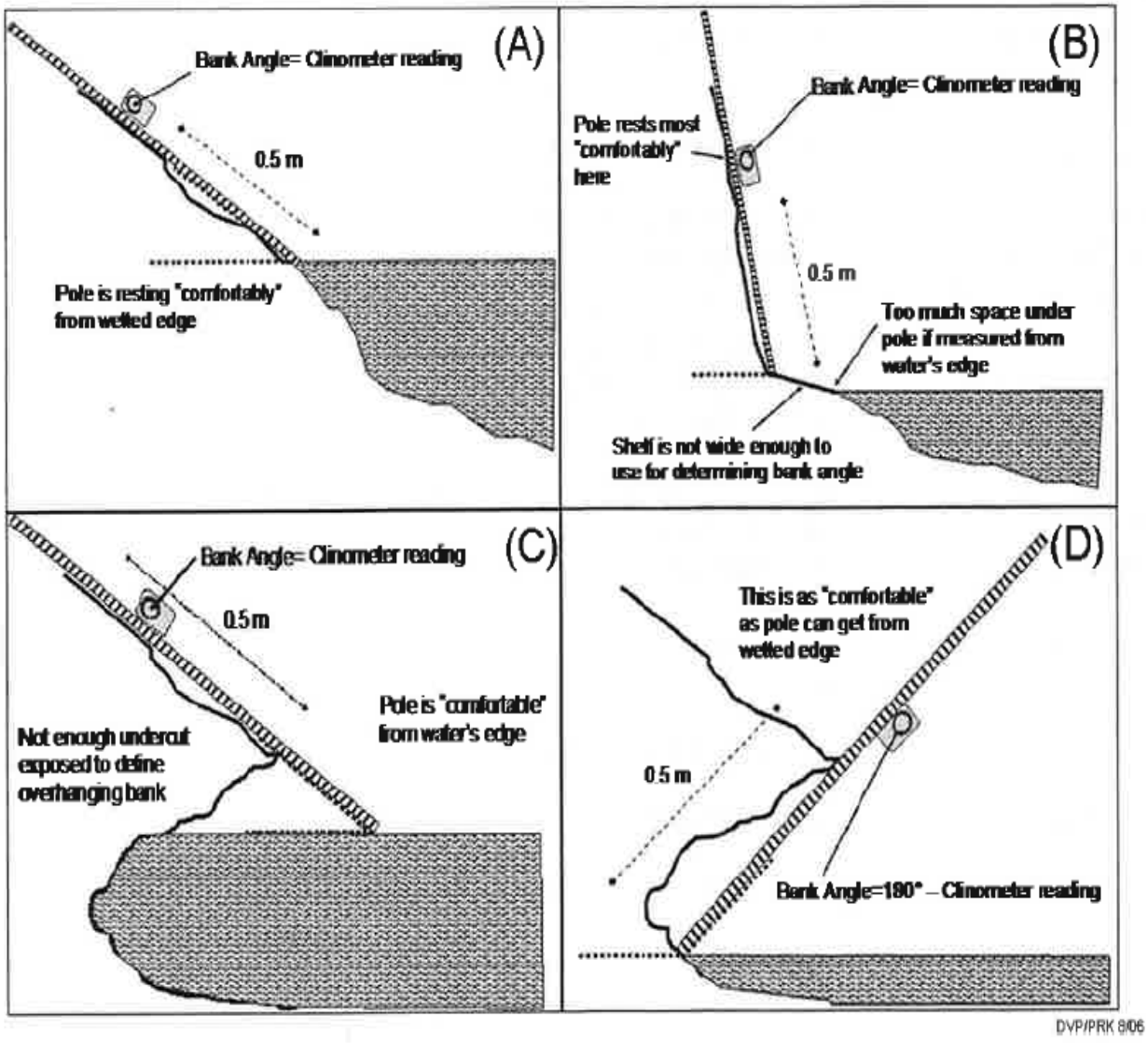


Figure 5-11. Bank angle under different kinds of conditions

Figure 5-12. Relationship between bankfull channel and incision

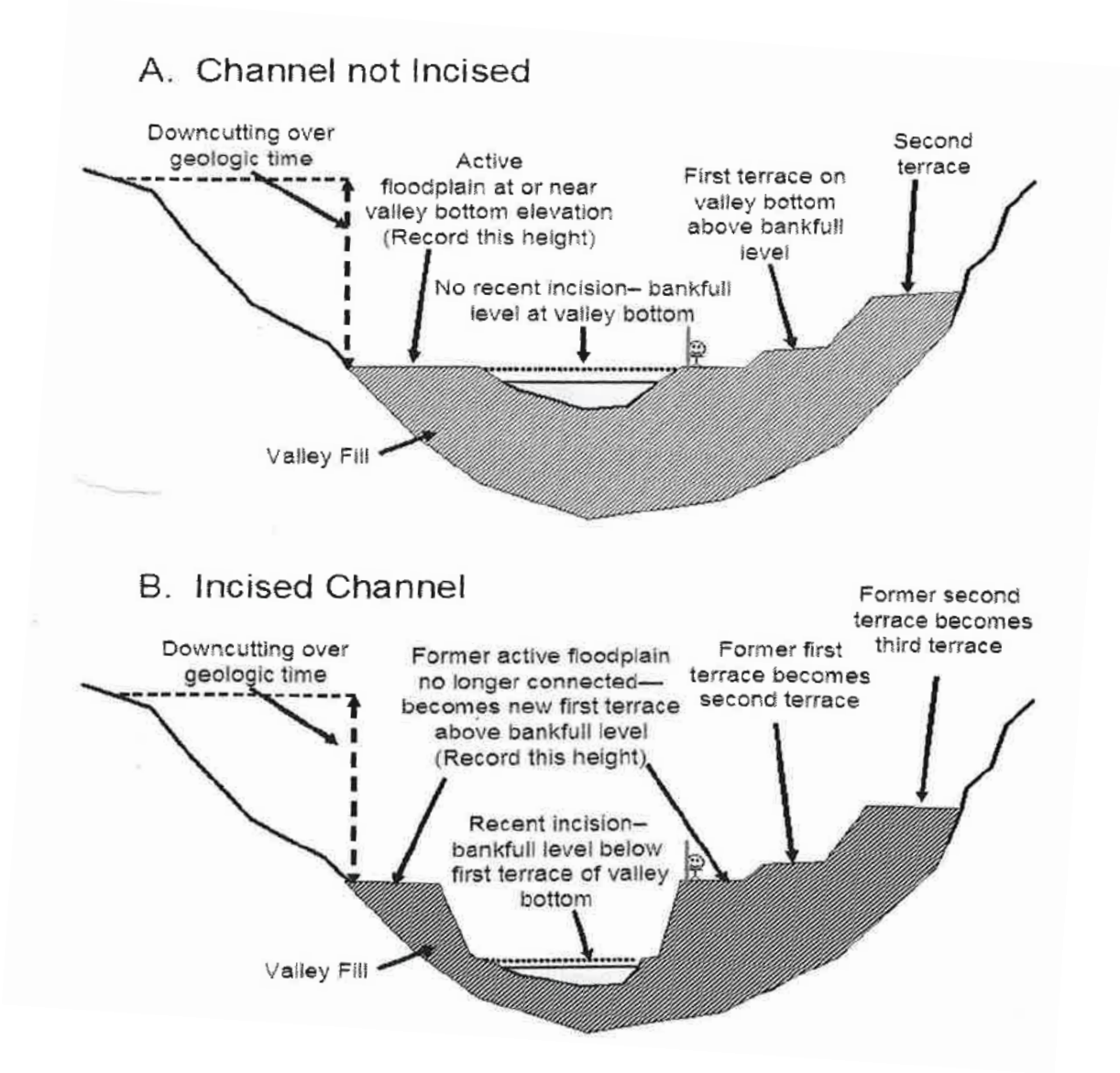
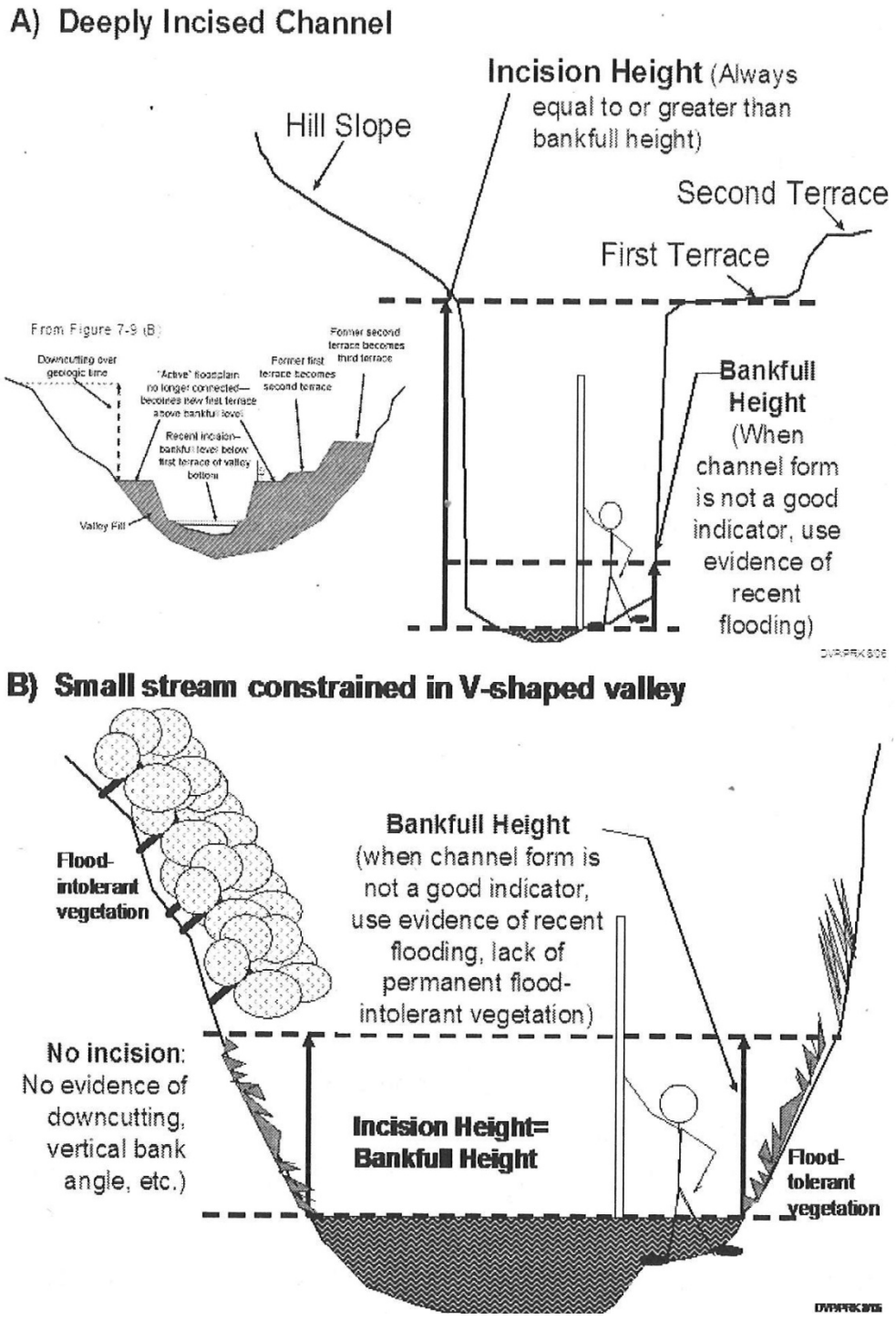


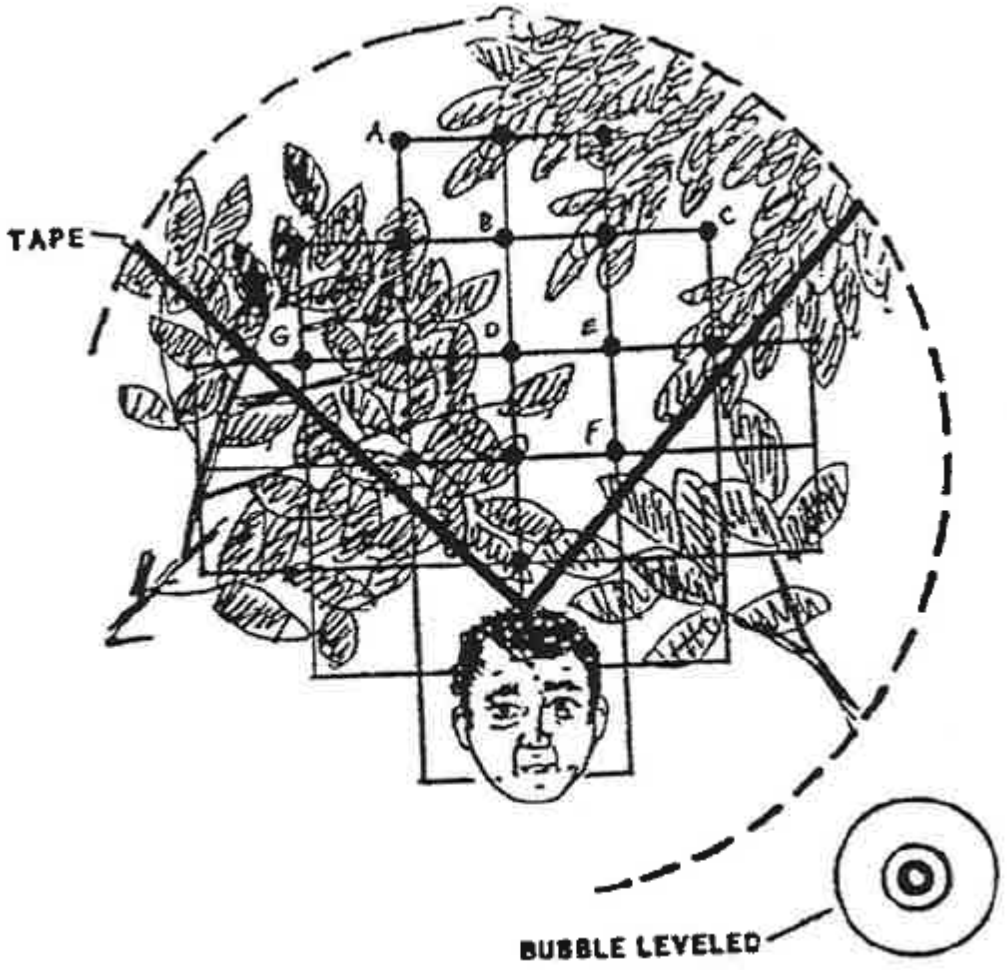
Figure 5-13. Determining bankfull heights for deeply incised channels and V-shaped valleys



5.2.5.4 Canopy Cover Measurements

Canopy cover over the stream is determined at each of the 11 cross-section transects. A spherical densiometer (model A-**convex** type) is used (Lemmon 1957). Mark the densitometer with a permanent marker or tape exactly shown in Figure 5-14 to limit the number of square grid intersections to read 17. Densiometer readings can range from 0 (no canopy cover) to 17 (maximum canopy cover). Six measurements are obtained at each cross-section transect (four measurements in each of four directions at mid-channel and one at each bank).

Figure 5-14. Schematic of modified convex spherical densiometer



The procedure for obtaining canopy cover data is presented Table 5-11. Hold the densiometer level (using the bubble level) 0.3 m above the water surface with your face reflected just below the apex of the taped "V", as shown in Figure 5-14. Concentrate on the 17 points of grid intersection on the densiometer that lie within the taped "V". If the reflection of a tree or high branch or leaf overlies any of the intersection points, that particular intersection is counted as having cover. For each of the six measurement

points, record the number of intersection point (maximum=17) that have vegetation covering them in the *Canopy Cover* Measurement section of the *Channel/Riparian Cross-section Form* as show in Figure 5-10.

Table 5-11. Procedure for canopy cover measurements

1. At each cross-section transect, stand in the stream at mid-channel and face upstream.
2. Hold the densiometer 0.3 m (1 ft) above the surface of the stream. Level the densiometer using the bubble level. Move the densiometer in front of you so your face is just below the apex of the taped "V".
3. Count the number of grid intersection points within the "V" that are covered by either a tree, leaf, or high branch. For objects that are not vegetation, but are appearing in densiometer, still count them, but make flags/comments in the appropriate fields (e.g. cut/incised banks, bridges, road crossings, power lines, etc.) explaining the objects that are creating cover, but not necessarily vegetation. Record the value (0-17) in the *CenUp* field of the canopy cover measurement section of the *Channel/Riparian Cross-Section and Thalweg Profile Form*.
4. Face toward the left bank (left as you face downstream). Repeat Steps 2 and 3, recording the value in the *CenL* field of the field data form.
5. Repeat Steps 2 and 3 facing downstream and again while facing the right bank (right as you look downstream). Record the values in the *CenDwn* and *CenR* fields of the field data form.
6. Move to the water's edge (either the left or right bank). Repeats Steps 2 and 3 again, this time facing the bank. Record the value in the *Lft* or *Rgt* fields of the field data form. Move to the opposite bank and repeat.
7. Repeat Steps 1 through 6 at each cross-section transect (including any additional side channel transects established when islands are present). Record data for each transect on a separate field data form.

5.2.5.5 Riparian Vegetation Structure

The previous section (5.2.5.4) described methods for quantifying the cover of canopy over the stream channel. The following visual estimation procedures supplement those measurements with a semi-quantitative evaluation of the type and amount of various types of riparian vegetation.

Riparian vegetation observations apply to the riparian area upstream 5 meters and downstream 5 meters from each of the 11 cross-section transects (refer to Figure 5-4). They include the visible area from the stream back a distance of 10 m (~30 ft) shoreward from both the left and right banks, creating a 10 m x 10 m riparian plot on each side of the stream (Figure 5-15). The riparian plot dimensions are estimated, not measured. On steeply sloping channel margins, the 10 m x 10 m plot boundaries are defined as if they were projected downs from an aerial view.

Table 5-12 presents the procedure for characterizing riparian vegetation structure and composition. Figure 5-10 illustrates how measurement data are recorded on the *Channel/Riparian Cross-Section Form*. Conceptually divide the riparian vegetation into 3 layers: The *Canopy* layer (> 5 m high), the *Understory* layer (0.5 to 5 m high), and the *Ground cover* layer (< 0.5 m high). Note that several vegetation types (e.g. grasses or woody shrubs) can potentially occur in more than one layer. Similarly note that some things other than vegetation are possible entries for the *Ground cover* layer (e.g. barren ground).

Figure 5-15. Riparian zone and instream fish cover plots for a stream cross-section transect

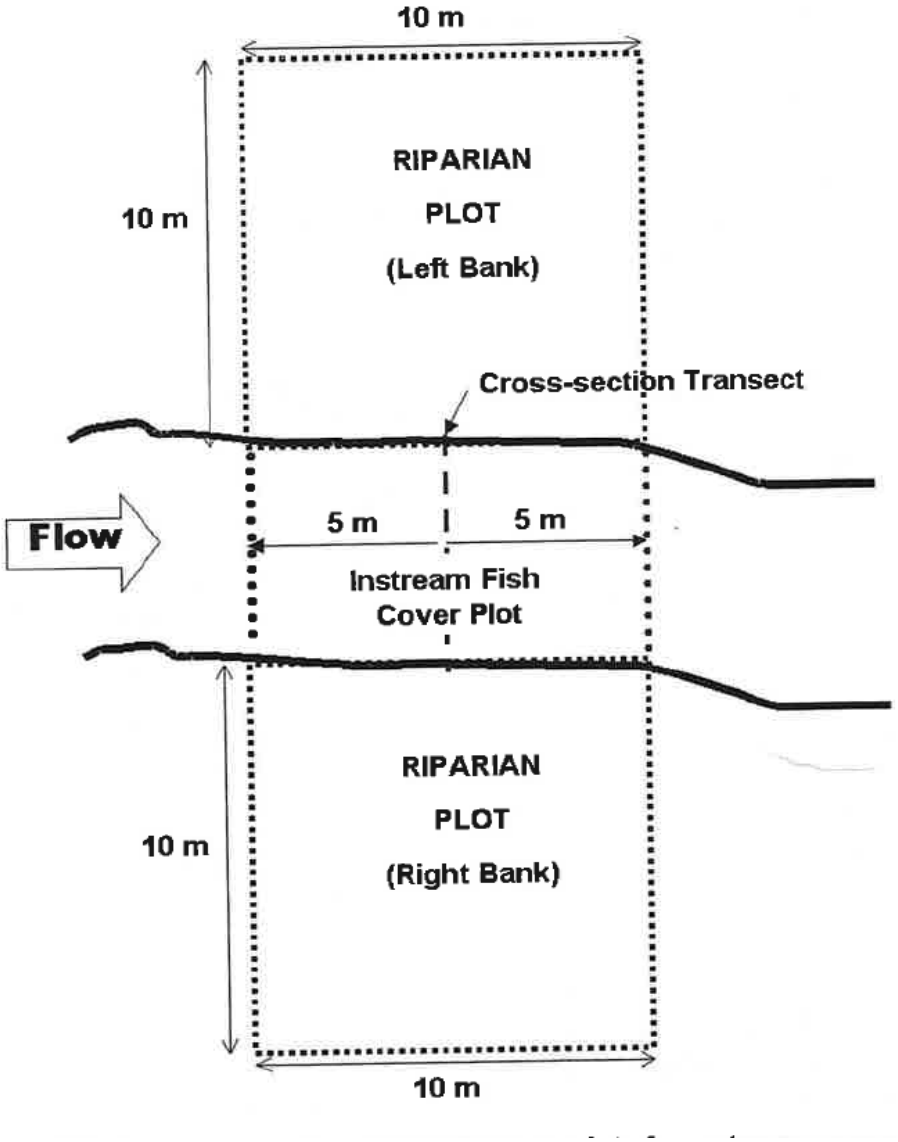


Table 5-12. Procedure for characterizing riparian vegetation structure

1. Standing in mid-channel at a cross-section transect, estimate a 5 m distance upstream and downstream (10 m total length).
2. Facing the left bank (left as you face downstream), estimate a distance of 10 m back into the riparian vegetation.
   On steeply-sloping channel margins, estimate the distance into the riparian zone as if it were projected down from and areal view.
3. Within this 10 m x 10 m area, conceptually divide the riparian vegetation into 3 layers: a *Canopy Layer* (> 5 m high), an *Understory* (0.5 m to 5 m high), and a *Ground Cover* layer (<0.5 m high).
4. Within this 10 m x 10 m area, determine the dominant vegetation type for the *CANOPY LAYER* (vegetation >5 m high) as either ***D****eciduous,* ***C****oniferous, broadleaf* ***E****vergreen,* ***M****ixed,* or ***N****one.* Consider the layer *Mixed* if more than 10% of the areal coverage is made up of the alternate vegetation type. Indicate the appropriate vegetation type in the *Visual Riparian Estimates* section of the *Channel/Riparian Cross-Section Form*.
5. Determine separately the areal cover class of large trees (≥0.3 m [1 ft] diameter at breast height [dbh]) and small trees (<0.3 m dbh) within the canopy layer. Estimate the areal cover as the amount of the shadow that would be cast by a particular layer alone if the sun were directly overhead. Record the appropriate cover class on the field data form (*0=absent*: zero cover, *1=sparse*: ,10%, *2=moderate*: 10-40%, *3=heavy*: 40-75%, or *4=very heavy*: >75%).
6. Look at the *UNDERSTORY* layer (vegetation between 0.5 m and 5 m high). Determine the dominant **woody** vegetation type for the undestory layer as described in Step 4 for the canopy layer. If there is no woody vegetation in the understory layer, record the the typ as ***N****one*.
7. Determine the areal cover class for woody shrubs and sapling separately from non-woody vegetation within the understory, as described in Step 5 for the canopy layer.
8. Look at the *GROUND COVER* layer (vegetation <0.5 m high). Determine the areal cover class for woody shrubs and seedlings, non-woody vegetation, and the amount of bare ground present as described in Step 5 for large canopy trees.
9. Repeat Steps 1 through 8 for the right bank.
10. Repeat Steps 1 through 9 for all cross-section transects (including any additional side channel transects established when islands are present). Use a separate field data form for each transect.

Before estimating the areal coverage of the vegetation layers, record the type of *woody* vegetation (broadleaf ***D****eciduous,* ***C****oniferous, broadleaf* ***E****vergreen,* ***M****ixed,* or ***N****one*) in each of the two taller layers (Canopy and Understory). Consider the layer *Mixed* if more than 10% of the areal coverage is made up of the alternate vegetation type. If there is no woody vegetation in the understory layer, record the type as *None*.

Esimate the areal coverage separately in each of the three vegetation layers. Note that the areal coverage can be thought of as the amount of shadow cast by a particular layer alone when the sun is directly overhead. The *maximum cover in each layer is 100%, so the*

sum of the areal covers for the combined three layers could add up to 300%. The four areal cover classes are *Absent, Sparse* (<10%), *Moderate* (10 to 40%), *Heavy* (40 to 75%), and *Very Heavy* (>75%). These cover classes and their corresponding codes are shown on the field data form (Figure 5-10). When rating vegetation cover types for a single vegetation layer, mixtures of two or more subdominant classes might all be given *Sparse* (*1*), *Moderate* (*2*), or *Heavy* (*3*) ratings. One *Very Heavy* cover class with no clear subdominant classes might be rated *4* with all the remaining classes rated as either *Moderate* (*2*), *Sparse* (*1*) or *Absent* (*0*). Note that within a given vegetation layer. Two cover types with 40-75% cover can be both rated *3*, but no more than one cover type could receive a rating of *4*.

5.2.5.6 Instream Fish Cover, Algae, and Aquatic Macrophytes

The procedure to estimate the types and amounts of instream fish cover is outlined in Table 5-13. Data are recorded on the *Channel/Riparian Cross-Section Form* as shown in Figure 5-10. Estimate the areal cover of all fish cover and other listed features that are in the water and on the banks 5 m upstream and downstream of the cross-section (Figure 5-15). The areal coverage classes of fish concealment and other features are the same as those described for riparian vegetation (Section 5.2.5.5).

The entry *FILAMENTOUS ALGAE* refers to long streaming alage that often occur in slow moving waters. *AQUATIC MACROPHYTES* are water-loving plants, including mosses, in the stream that provide cover for fish or macroinvertebrates. If the stream channel contains live wetland grasses, include these as aquatic macrophytes. *WOODY DEBRIS* are the larger pieces of wood that can influence cover and stream morphology (i.e. those pieces that would be included in the large woody debris tally [Section 5.2.3]). *BRUSH/WOODY DEBRIS* refers to smaller wood pieces that primarly affect cover but not morphology. *LIVE TREES OR ROOTS* are living trees that are within the channel – estimate the areal cover provided by the parts of these trees or roots that are inundated. *OVERHANGING VEGETATION* includes tree branches, brush, twigs, or other small debris that is not in the water but close to the stream (within 1 m of the surface) and provied potential cover. *BOULDERS* are typically basketball-to car-sized particles. *ARTIFICIAL STRUCTURES* inlcude those designed for fish habitat enhancement, as well as in-channel structures that have been discarded (e.g. concrete, asphalt, cars, or tires) or deliberately placed for diversion, impoundment, channel stabilization, or other purposes.

Table 5-13. Procedure for estimating instream fish cover

1. Standing mid-channel at a cross-section transect, estimate a 5 m distance upstream and downstream (10 m total length).
2. Examine the water and both banks within the 10 m segments of stream for the following features and types of fish cover: *filamentous algae, aquatic macrophytes large woody debris, brush and small woody debris, in-channel live trees or roots, overhanging vegetation, undercut banks, boulders, and artificial structures*.
3. For each cover type, estimate the areal cover. Record the appropriate cover class in the *Fish Cover/Other* section of the *Channel/Riparian Cross-Section Form*.
    - *0=absent*: zero cover,
    - *1=sparse*: <10%
    - *2=moderate*: 10-40%
    - *3=heavy*: >40-75%, or
    - *4*=very heavy: >75%.
4. Repeat Steps 1 through 3 at each cross-section transect (including any additional side channel transects established when islands are present). Record data from each transect on a separate field data form.

5.2.5.7 Human Influence

For the left and right banks at each of the 11 detailed Channel and Riparian Cross-sections, evaluate the presence/absence and the proximity of 11 categories of human influnces with the procedure outlined in Table 5-14. Relate your observations and proximity evaluations to the stream and riparian area within 5 m upstream and 5 m downstream from the station (Figure 5-15). Four proximity classes are used: In the stream or on the bank within 5 m upstream or downstream of the cross-section transect, present within the 10 m x 10 m riparian plot but not in the stream or on the bank, present outside of the riparian plot and absent. For more information on the proximity classes see Figure 5-16. Record data on the *Channel/Riparian Cross-Section Form* as shown in Figure 5-10. If a disturbance is within more than one proximity class, record the one that is the closest to the stream (e.g. *C* takes precedence over *P*).

A particular influence may be observed outside of more than one riparian observation plot (e.g. at both transects *D* and *E*). Record it as present at every transect where you can see it without having to sight through another transect or its 10 m x 10 m riparian plot.

Figure 5-16 Proximity Classes for Human Influences

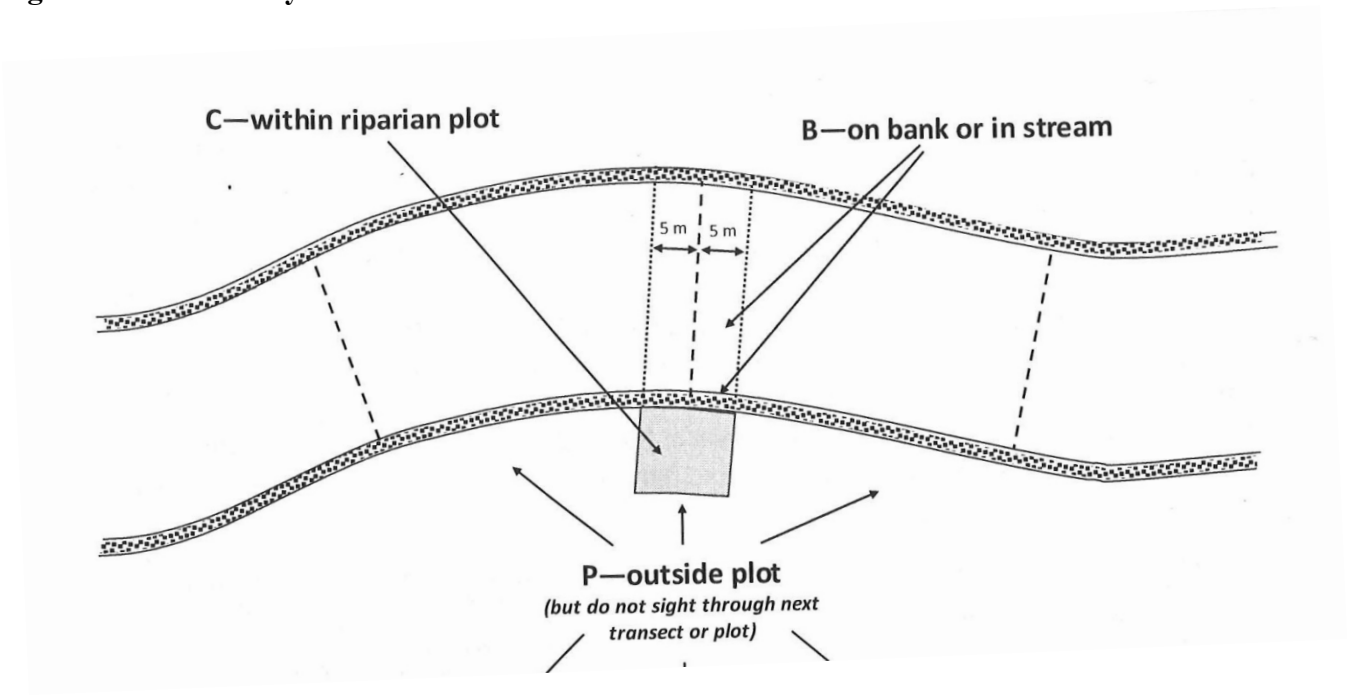


Table 5-14. Procedure for estimating human influence

1. Standing mid-channel at a cross-section transect, look toward the left bank (left facing downstream), and estimate a 5 m distance upstream and downstream (10 m total length). Also, estimate a distance of 10 m back into the riparian zone to define a riparian plot area.
2. Examin the channel, bank and riparian plot area adjacent to the defined stream segment for the following human influences: (1) *walls, dikes, revetments, riprap, and dams*; (2) *buildings,* (3) *pavement/cleared lots* (e.g. paved, gravelled, dirt parking lot, foundation); (4) *roads and railroads*, (5) *inlet or outlet pipes*; (6) *landfills or trash* (e.g. cans, bottles, trash heaps); (7) *parks or maintained lawns*; (8) *row crops*; (9) *pastures, rangeland, hay fields,* or *evidence of livestock*; (10) *logging*; and (11) *mining* (including gravel mining).
3. For each type of influence, determine if it is present at what its proximity is to the stream and riparian plot area. Consider human disturbance items as present if you can can see them from the cross-section transect. Do not include them if you have to sight throught another transect or its 10 m x 10 m riparian plot.
4. For each type of influence, record the appropriate proximity class in the *Human Influence* part of the *Visual Riparian Estimates* section of the *Channel/Riparian Cross-Section Form*. Proximity classes are:
    - *B (Bank)* Present within the defined 10 m stream segment and located in the stream or on the stream bank.
    - *C (Close)* Present within the 10 m x 10 m riparian plot area, but away from the bank.
    - *P (Present)* Present, but outside the riparian plot area.
    - *0 (Absent)* Not present within or adjacent to the 10 m stream segment or the riparian plot area at the transect.
5. Repeat Steps 1 through 4 for the right bank.
6. Repeat Steps 1 through 5 for each cross-section transect, (including any additional side channel transects established when islands are present). Record data for each transect on a separate field form.

5.2.5.8 Cross-section Transects on Side Channels

If the wetted width channel is split by an island and the estimated flow in the side channel is less than or equal to 15% of the total flow, the bank and riparian measurements are made at each side of the main channel (the minor side channel is ignored other than to note its presence on the thalweg profile form), so one riparian plot is established on the island as shown in Figure 5-17. If and island is present that creates a major side channel containing more than 15% of the total flow (Sections 4.3.4 and 5.2.5.2), an additional cross-section transect is established for the side channel shown in Figure 5-17. Separate substrates, bank and riparian measurements are made for side channel transects. Data from the additional side channel transect are recorded on a separate *Channel Riparian/Cross-Section Form* as shown inFigure 5-18. Riparian plots esblished on the island for each transect may overlap (and be < 10 m shoreward) if the island is less than 10 m wide at the transect.

Figure 5-17. Riparian and instream fish cover plots for a stream with minor and major side channels

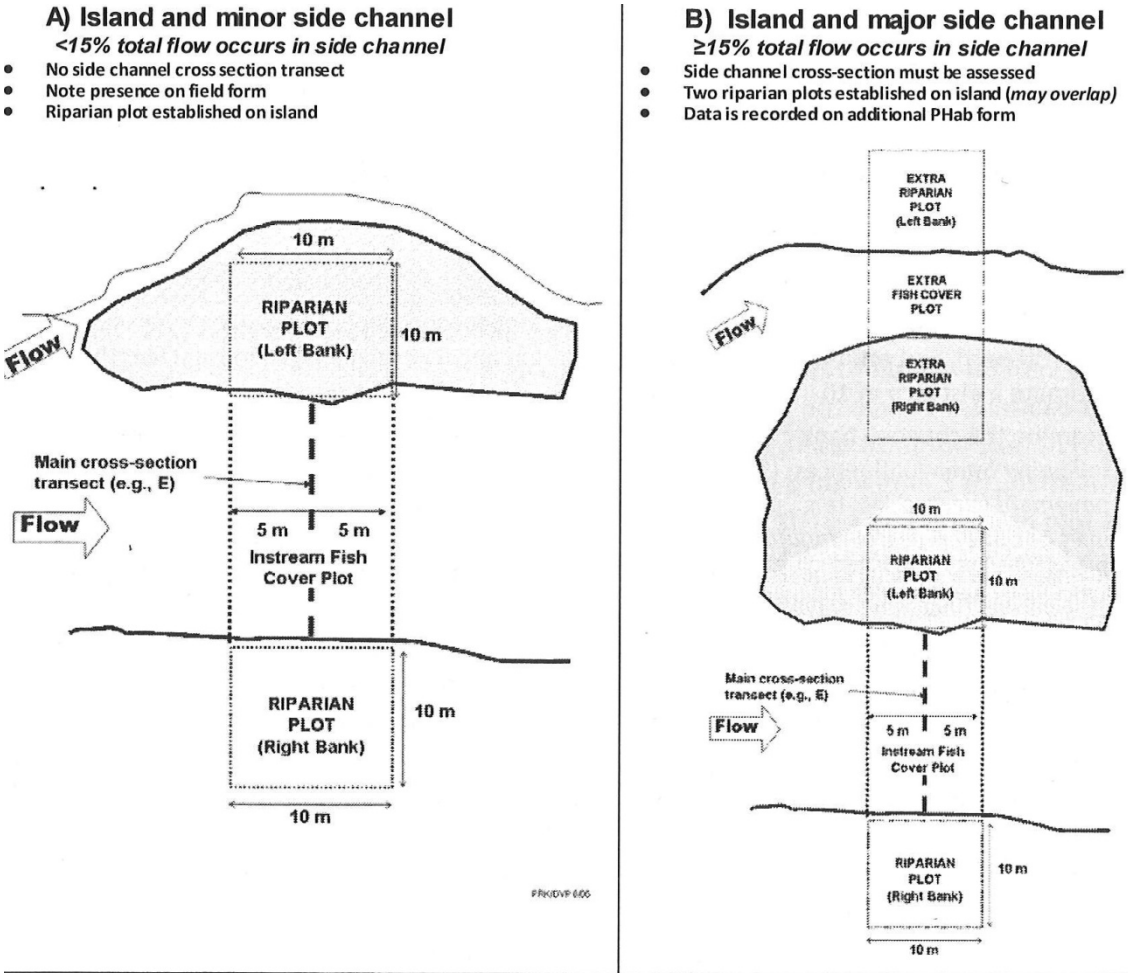
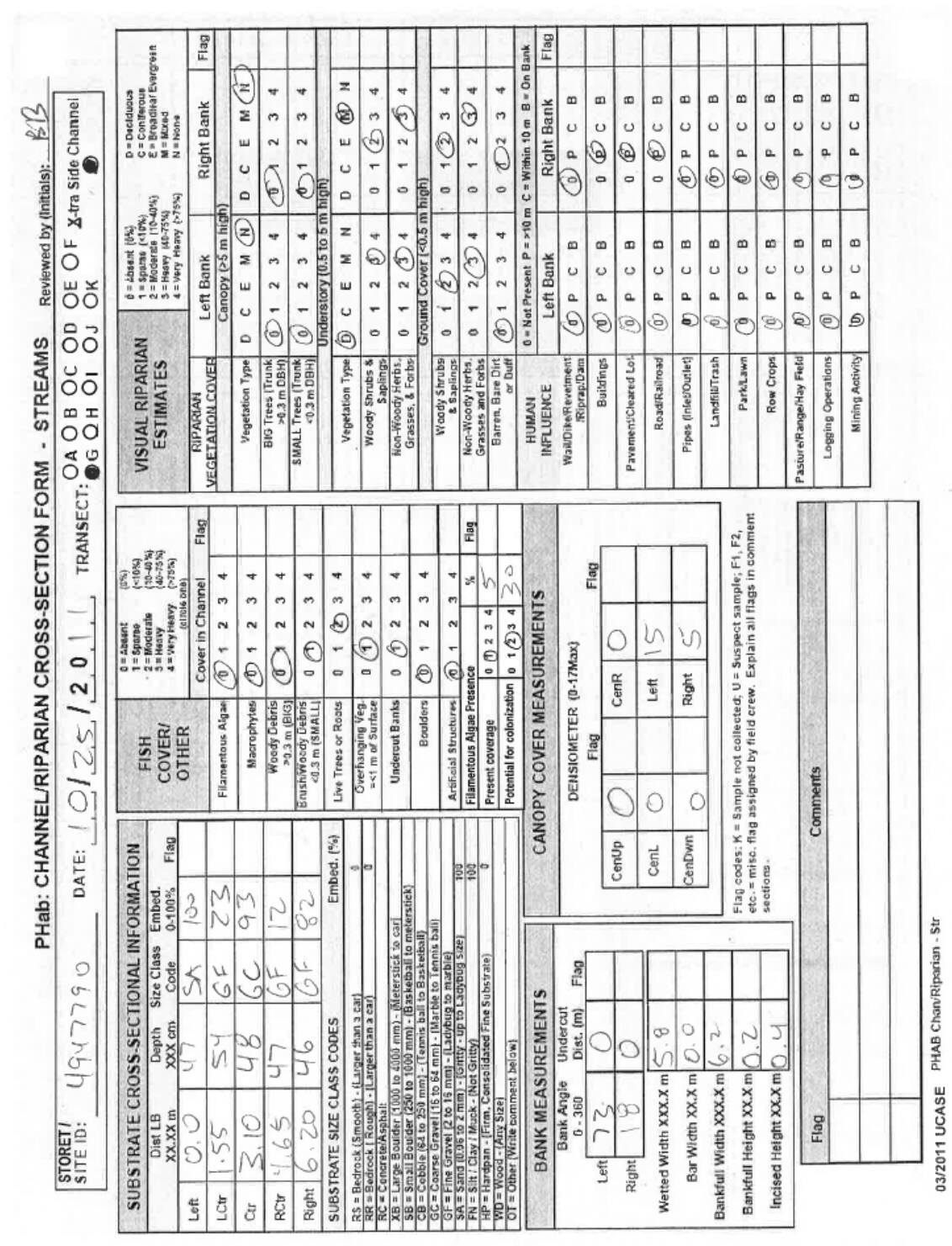


Figure 5-18. Channel/Riparian Cross-Section Form for an additional major side channel transect



5.2.6 Channel Constraint, Debris Torrents, Recent Floods, and Discharge

5.2.6.1 Channel Constraint

After completing the thalweg profile and riparian/channel cross-section measurements and observations, envision the stream at bankfull flow and evaluate the degree, extent and type of channel constraint, using the procedures presented in. Record data on the *Channel Constraint and Field Chemistry Collection Form* (Figure 5-19). First, classify the stream reach channel pattern as predominantly a *single channel*, an *anastomosing channel*, or a *braided channel* (Figure 5-20):

1. *Single channels* may have occasional in-channel bars or islands with side channels, but feature a *predominant single channel,* or a *dominant main channel with a subordinate side channel.*
2. *Anastomosing channels* also have relatively long *major and minor channels* (but no predominant channel) in a complex network, diverging and converging around many *vegetated islands*. Complex channel pattern remains even during major floods. An example of this in Utah would be high mountain streams especially in the Uintas or Boulder Mountain.
3. *Braided channels* also have multiple branching and rejoining channels, (but no predominant channel) *separated by unvegetated bars*. Channels are generally smaller, shorter, and more numerous, often with no obvious dominant channel. During major floods, a single continuous channel may develop. An example of this are desert streams in southern Utah similar to the Paria River.

After classifying the channel pattern, determine whether the channel is constrained within a narrow valley, constrained by local features within a broad valley, unconstrained and free to move about within a broad floodplain, or free to move about, but within a relatively narrow valley floor. Then examine the channel to ascertain the bank and valley features that constrain the stream. Entry choices for the type of constraining features are bedrock, hillslopes, terraces/alluvial fans, and human land use (e.g. a road, a dike, landfill, rip-rap, etc). Estimate the percent of the channel margin that is in contact with each of the constraining features if applicable (for unconstrained channels, this is 0%). To aid in this estimate, you may wish to refer to the individual transect assessments of incision and constraint. Next, estimate the width of the valley floor either with a map or visually. If you cannot directly measure the valley width (e.g. it is further than you can see, or if your view is blocked by vegetation), record the distance you can see and mark the appropriate circle on the field form.

Table 5-15. Procedures for assessing channel constraint

NOTE: These activities are conducted after completing the thalweg profile and littoral-riparian measurements and observations, and represent an evaluation of the entire stream reach.

***Channel Constraint*:** Determine the degree, extent, and type of channel constraint based on envisioning the stream at **bankfull flow**.

Classify the stream reach pattern as predominantly a **single** channel, an **anastomosing** channel, or a **braided** channel.

- ***Single channels*** *may have occassional in-channel bars or islands with side channels,* but feature a predominant single channel or a dominant main channel with a subordinate side channel.
- ***Anastomosing channels*** *have relatively long major and minor channels* branching and rejoining in a complex network separated by vegetated islands with no obvious dominant channel.
- ***Braided channels*** *also have multiple branching and rejoining channels*, separated by unvegetated bars. Subchannels are generally small, short, and numerous, often with no obvious dominant channel.

After classifying the channel pattern, determine whether the channel is constrained within a narrow valley, constrained by local features within a broad valley, unconstrained and free to move about within a broad floodplain, or free to move about, but within a narrow valley floor.

Then examine the channel to ascertain the bank and valley features that constrain the stream. Entry choices for the types of constraining features are bedrock, hillslopes, terraces/alluvial fans, and human land use (e.g. a road, dike, landfill, rip-rap, etc.). More than one of these features can be selected.

Estimate the percent of the channel margin in contact with each constraining feature selected for the site (for unconstrained channels, this is 0%). Record the value on the *Channel Constraint and Field Chemistry Collection Form*.

Figure 5-19. Channel Constraint and Field Chemistry

CHANNEL CONSTRAINT AND FIELD CHEMISTRY-WADEABLE STREAMS

Reviewed by (initial): BB

SITE/STORET ID: 4946990 DATE: 09/29/2011

MULTI-PARAMEMTER UNIT MEASUREMENTS

	Comments
STREAM TEMP (°C): 9.6	RESIDUAL CHLORINE (optional): Not sampled
pH: 8.04	
SPECIFIC CONDUCTIVITY (µS/cm) 454	
STREAM DO mg/L: 7.3	
STREAM DO%: 77.8	
TIME OF DAY: 1011	

CHANNEL CONSTRAINT

CHANNEL PATTERN (Check one)

- ● **One Channel**
- ○ **Anastomosing (complex) channel:** (Relatively long major and minor channels branching and rejoining)
- ○ **Braided channel:** (Multiple short channel branching and rejoining – mainly one channel broken up by numerous mid-channel bars)

CHANNEL CONSTRAINT (Check one)

- ● **Channel very constrained in V-shaped valley** (i.e. it is very unlikely to spread out over valley or erode a new channel during flood)
- ○ **Channel is in Broad Valley** but channel movement by erosion during flood is **constrained by incision** (Flood flows do not commonly spread over valley floor or into multiple channels)
- ○ **Channel is in Narrow Valley but is not very constrained,** but limited in movement by relatively narrow valley floor
- ○ **Channel is Unconstrained in Broad Valley** (i.e. during flood it can fill off-channel areas and side channels, spread out over flood plain, or easily cut new channels by erosion)

CONSTRAINING FEATURE (Check all that apply)

- ○ **Bedrock/Sandstone** (i.e. channel is constrained by bare bedrock or sandstone)
- ● **Hillslope** (i.e. channel constrained in narrow V-shaped valley)
- ○ **Terrace** (i.e. channel is constrained by its own incision into stream gravel/soil deposits)
- ○ **Human Bank Alterations** (i.e. constrained by rip-rap, landfill, dike, road, etc.)
- ○ **No constraining features**

PERCENT OF CHANNEL LENGTH WITH MARGIN IN CONTACT WITH EACH CONSTRAINING FEATURE (percentage fields should add up to 100%):	Constraining Feature 1 (primary)	Constraining Feature 2 (if applicable)	Constraining Feature 3 (if applicable)
	Type: Hillslope	Type:	Type:
	Percentage: 100 %	Percentage: %	Percentage: %

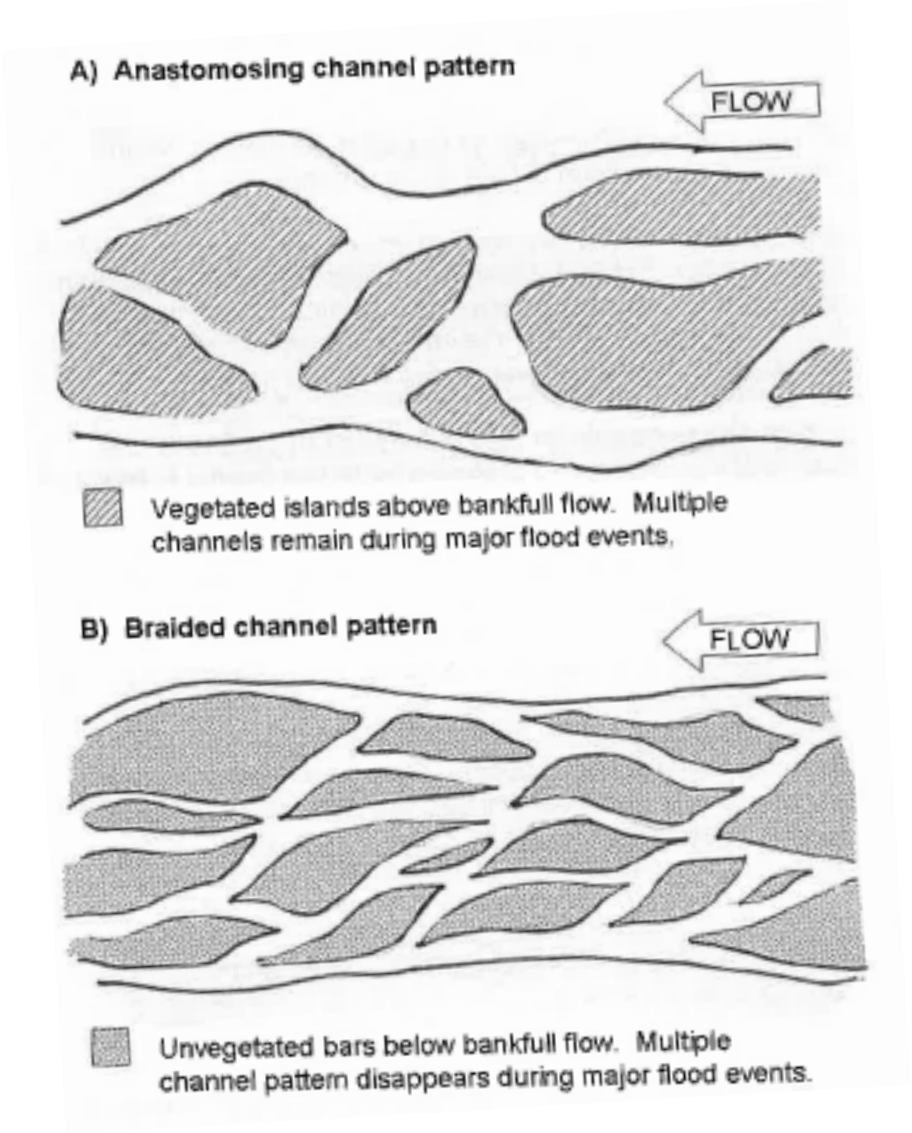
Valley width (Visual Estimated Average): 30 (m)
Note: Be sure to include distances between both sides of valley border for valley width.

If you cannot see the valley borders, record the distance you can see and mark this circle ○

Comments: width from valley floor

UCASE FIELD FORM Updated: 03/2011

Figure 5-20. Types of multiple channel patterns



5.2.5.1 Debris Torrents, Flash Floods, and Recent Major Floods

Debris torrents, differ from conventional floods in that they are flood waves of higher magnitude and shorter duration and their flow consists of a dense mixture of water and debris. On a Utah scale, these events are commonly known as flash floods. Their high flows of dense material exert tremendous scouring forces on streambeds. Summer monsoonal storms in Utah often bring sudden and destructive storms that cause torrent events and can devestate stream habitat. These events are able to move boulders the size of a medium sized car and large logs from high reaches of watersheds that would otherwise never be found in the desert. These events can wipe out structures, like homes, near the stream corridor with no warning because the storm was so sudden and local to a particular area and not necessarily another downstream. Even though these flooding/torrent events are almost always part of the natural process for stream characterization,

they often scour several portions of the stream, reduce channel complexity, and denude near-bank riparian vegetation. The massive disruption of the stream channel and its biota are transient and these intense events will often lead to new complex habitat development within years or decades.

Since these events are able to alter habitat and biota substantially, infrequent major floods and torrents can confuse the interpretation of measurements of stream biota and habitat in streams across the state. Therefore, it is important to determine if a debris torrent or flash flood has occurred recently in the past (2-3 years). After completing the thalweg profile and channel/riparian measurements and observations, examine the stream channel along the entire sample reach, including its substrates, banks, and riparian corridor, checking the presence of features described on the *Torrent/Flash Flood Evidence Form* (Figure 5-21). It may be advantageous to look at the channel upstream and downstream of the actual sample reach to look for areas of torrent scour and debris depostion to answer some of the questions on the field form.

Figure 5-21. Torrent/Flash Flood Evidence Form

TORRENT/FLASH FLOOD EVIDENCE ASSESSMENT FORM

Reviewed by (initial): BB

SITE/STORET ID: 4 9 4 8 8 4 3 DATE: 08 / 18 / 2011

TORRENT EVIDENCE

Please fill-in the circle if any of the following are evident while sampling.
Visit the following web-sites pre/post sampling to see if any recent major flow regimes occurred:
- **http://www.wrh.noaa.gov/slc/**
- **http://waterdata.usgs.gov/ut/nwis/current/?type=flow**

	EVIDENCE OF TORRENT EVENTS/FLASH FLOODS:
●	Stream channel has a devegetated corridor. This corridor lacks riparian vegetation due to a large flood event.
●	Vegetation along stream margin is depressed.
●	Riparian trees have fresh bark scars at many points along the stream at heights above bankfull level.
●	Riparian vegetation has fallen into the channel as a result of scouring near their roots.
●	Much of stream bed substrate is more loosely consolidated than expected (appears recently deposited).
●	There are locales along the reach where gravel or cobble deposits can be seen above bankfull level from a large flood event.
○	Isolated puddles of water are seen above bankfull level.
●	Deposits of debris (sticks, trash, gravel, vegetation, etc.) can be found above bankfull level.
●	Water appears more turbid than typically expected (considering the eco-region).
●	Overall abundance of benthos sample appears lower than otherwise expected, considering the eco-region, due to recent flood event.
●	Soil above bankfull height is damp.
●	Significant amounts of large woody debris within the stream or riparian corridor appear to have recently been moved.
	MAGNITUDE/DESCRIPTION OF EVENT:
○	Evidence of flooding present, but magnitude is common for this system on a yearly basis (i.e. desert streams after a monsoonal summer storm).
○	Evidence of a large flood present (10-20 year flood), but did not make massive changes to the system.
●	Evidence shows a massive event where signs like channel blow-outs, devegetation, and major changes to the stream are present (i.e. ≥100 year flood).
	OTHER:
○	Other evidence not covered in options above (explain observations in comments section)
	NO EVIDENCE:
○	No evidence of torrent or flash flood events.

COMMENTS
(if evidence of flooding is present, please explain when you think the event occurred, whether it be within the month, longer than a month, within the year, more than two years, etc.)

Upper watershed / tributaries had a massive fire in 2010, which increased magnitude of spring runoff. Area had abnormally high snowpack last winter. Massive flood from storm occurred 3-4 weeks prior to sampling.

UCASE FIELD FORM Updated: 03/2011

5.3 Stream Discharge

Stream discharge is equal to the product (multiplication) of the mean current velocity and vertical cross sectional area of flowing water. Discharge measurements are critical for assessing loading trends for any particular pollutant being sampled or to see if too much or too little volume is impairing the quality of the stream. Discharge should be measured at a suitable location within the sample reach that is as close as possible to the location where water chemistry samples were collected so that these data correspond. Discharge is usually determined after collecting water chemistry samples.

No single method for measuring discharge is applicable to all types of stream channels. The preferred procedure for obtaining discharge data is based on "velocity-area" methods. For UCASE sampling, there are five different methods that are acceptable to determine a flow rate at UCASE sites:

1. Sontek/YSI FlowTracker handheld-ADV® (Flow Tracker)
    - See Utah DWQ's SOP on Stream Flow for further information and instruction on how to operate this piece of equipment Add website link to SOPs
2. USGS gage or other properly maintained gage nearby site (county gages, etc.)
    - If this method is used, crew must be certain that there are no influences that might change the flow rate from where the site is located to where gage is located.
    - Record the gage number and agency who manages it in the comments section of the *Stream Discharge Form*.
    - USGS website: http://waterdata.usgs.gov/ut/nwis/current/?type=flow
3. Neutrally Buoyant Object
    - See Utah DWQ's SOP on Stream Flow for further information and instruction on how to conduct this type of flow measurement Add website link to SOPs
4. Estimated Flow
    - This is the least preferred method due to its inaccuracy, but is still preferred over having no flow recorded at all.
    - This method is used mostly when streams are so small that a hand-held device is unable to be submerged in water to take readings; when crew has limited staff to hike in gear to remote sites; or if equipment fails while in the field.

The flow rate is recorded and the method used is marked on the *Stream Discharge Form* in the UCASE site packet (**Figure 5-22**).

Figure 5-22. Stream Discharge Form (the Flow Tracker method was used at this particular site)

PERIPHYTON COLLECTION FORM
STREAM DISCHARGE FORM-WADEABLE STREAMS

Reviewed by (initial): RB

SITE/STORET ID: 4 9 4 0 4 1 0 DATE: 0 6 / 0 8 / 2 0 1 1

COMPOSITE PERIPHYTON SAMPLE

Composite Volume (mL) (Quantitative)	Number of transects sampled (0-11):	Qualitative Collection
5 0 0	1 1	Was a **qualitative** sample collected at this site: (Y) / N If yes, indicate # of vials submitted to the Rushforth Lab: 0 1

Species (Assemblage) ID	Chlorophyll	Ash-Free Dry Mass (Biomass)	Comments/Flags
Process: Fill vial with 50mL of water from composite sample. **Preservation:** Seal cap with electrical-tape, place sample in a zip-top baggie; keep cool on ice. **Lab:** Rushforth Phycology within 2 weeks of collection. **Shop storage:** Fridge	**Process:** Filter 25 mL from composite sample using a pre-filter. **Preservation:** Fold filter in half and place in a lightless canister (e.g. film canister) and keep frozen (dry ice) in a zip-top baggie. **Lab:** State of Utah Health Lab within two weeks of collection. **Shop storage:** Freezer	**Process:** Filter 25 mL from composite sample using a pre-filter. **Preservation:** Fold filter in half and place in a lightless canister (e.g. film canister) and keep frozen (dry ice) in a zip-top baggie. **Lab:** State of Utah Health Lab within two weeks of collection. **Shop storage:** Freezer	Please indicate if any aquatic vegetation samples were collected for ID at Utah State University Herbarium
Indicate volume if not 50 mL __ __ FLAG:	Indicate volume filtered if not 25 mL __ __ FLAG:	Indicate volume filtered if not 25 mL __ __ FLAG:	

●FLOW TRACKER

Discharge is determined directly in field, record value here:

Q= 4.74 ●cfs ○m^3/s FLAG:

○USGS Gage

USGS gage is near site and flow was looked up online:

_____ cfs FLAG:

○VELOCITY AREA

Distance Units ○ft ○cm	Depth Units ○ft ○cm	Velocity Units ○ft/s XX.X ○m/s X.XX	FLAG

○ NEUTRAL BOUYANT OBJECT

	FLOAT 1	FLOAT 2	FLOAT 3
Float Dist. ○ft ○m	__ __	__ __	__ __
Float Time (sec)	__ __ __	__ __ __	__ __ __
FLAG	__ __ __	__ __ __	__ __ __

Cross Sections on Float Reach

	Upper Section	Middle Section	Lower Section
Width ○ft ○m	__ __ . __	__ __ . __	__ __ . __
Depth 1 ○ft ○m	__ __ __	__ __ __	__ __ __
Depth 2	__ __ __	__ __ __	__ __ __
Depth 3	__ __ __	__ __ __	__ __ __
Depth 4	__ __ __	__ __ __	__ __ __
Depth 5	__ __ __	__ __ __	__ __ __

COMMENTS/FLAGS

If flow is estimated, please indicate value here:

UCASE Field Form Updated: 03/2011

5.4 Equipment and Supplies

Table 5-16 lists the equipment and supplies required to conduct all activities described for characterizing physical habitat. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3.0) to ensure that all of the required equipment is brought to the stream.

Table 5-16. Checklist of equipment and supplies for physical habitat

For taking measurements	• Surveyor's telescoping leveling rod (stadia rod) (square or round profile, metric scale, at least 5 m when extended • 50 m or 100 m measuring tape and reel • Laser rangefinder (400 ft. distance range) • Digital camera with memory card and battery • Meter stick or short rod or pole (e.g. marked ski pole or broom handle) with cm markings for thalweg measurements • Roll of colored surveyor's flaggin tape OR at least 15 pin flags • Convex spherical canopy densiometer (Lemmon Model A), modified with at taped/drawn "V" • Clinometer • Bearing compass (backpacking type) • Chest waders (bring multiple pairs/types if possible for variable conditions such as a breathable pair, a neoprene pair, a pair of hip waders) • Current velocity meter, probe, and operating manual (flow meter) • Neutrally buoyant object (e.g. rubber ball, stick, apple, orange) • UCASE Field Manual and quick reference guide • GPS unit
For recording Data	• Clipboards (2)-have at least one that has multiple compartments in case of wet weather to protect field forms • Soft (#2) lead pencils • 11 plus extras Channel/Riparian Cross-section Forms • 11 plus extras Thalweg Profile and Woody Debris Forms • 1+ extras field form: Stream Verification Form • 1+ extras field form: UCASE Reference Site List/Site Rating Field Evaluation Form • 1+ extras field form: Channel Constraint and Field Chemistry Form • 1+ extras field form: Torrent/Flash Flood Evidence Assessment Form • 1+ extras field form: Sample Collection Form • 1+ extras field form: Periphyton Collection Form/Stream Discharge Form • 1+ extras field form: Stream Assessment Form • 1+ extras field form: Slope and Bearing Form • 1+ extras field form: DWQ Electrofishing/Fish Tissue Collection

5.5 Periphyton

5.5.1 Summary of Method

Collect periphyton from each of the 11 transects within the sampling reach ("A" through "K"). Usually, the person that collects the macroinvertebrate sample also collectes the periphyton sample. Depending on the number of people on the crew this can be managed differently as preferred by the crew. The periphyton sampling should be done after the macroinvertebrate sample has been collected. Prepare one composite "index" sample of periphyton for each transect. At the completion of the day's sampling activities, but before leaving the site, prepare three types of laboratory samples: (a benthic-chlorophyll-*a* sample and a ash free dry mass sample (AFDM) otherwise known as dry weight), and a 50 mL pour-off of the composite sample for species ID from the composite periphyton sample.

5.5.2 Equipment and Supplies

Table 5-17 is a checklist of equipment and supplies required to conduct periphyton sample collection and processing activities. This checklist is similar to the checklist presented in Appendix A, which is used at the base location (Section 3.0) to ensure that all of the required equipment is brought to the site.

Table 5-17. Equipment and supplies list for periphyton at wadeable sites

For collecting samples	• Large funnel (15-20 cm diameter) • 12-cm^2 area dilimiter (3.8 cm diamater pipe, 1-3 cm tal • Standard stiff-bristle toothbrush, or small paintbrush with bristles cut short (cut short enough where bristles are firm enough to scrub objects). • Wash bottle for stream water • 50 mL centrifuge tubes • 500 mL plastic bottle for the composite sample • 60 mL plastic syringe with 3/8" hole bored into end • Field Manual and/or Quick Reference Guide
For recording measurements	• Periphyton Collection Form • Pencil for recording data on field forms • Sample labels (CHLA-Benthic(UCASE)-5162 and Dry Weight 5162) • Clear tape strips for covering labels

5.5.3 Sampling Procedure

At each of the 11 transects (A-K), collect a periphyton sub-sample from anywhere in the transect following a left, right, center method of distributing the samples across the stream. Be sure not to sample right at the transect cross-section since there might be a chance that other sampling activities have already occurred there by other crew members (i.e. laying out the reach, measuring pHab attributes, etc). Always try to sample a short distance (1-2 m) above or below the cross-section flags. While sampling, make an attempt to collect substrates from a variety locations in stream (i.e. left bank, center of stream, and right bank) instead of favoring one bank or another. Make and attempt not to collect substrates deeper than 0.5 m. If a sample cannot be collected somewhere within the transect because the location is too deep, skip the transect. The procedure for collecting samples and preparing a composite sample is presented in Table 5-18. Collect one sample from each of the transects and composite in one bottle to produce one composite sample for each site. Record the volume of the sample on the *Periphyton Collection Form* as shown in **Figure 5-22**.

Table 5-18. Procedure for collecting composite index samples of periphyton at wadeable sites

1. Starting with Transect "A", collect a single sample using the procedure below.

    a) Collect a sample from substrate (rock, wood, etc.) that is small enough (<15 cm diameter) and can be easily removed from the stream. Place the substrate in a plastic funnel which drains into a 500 mL plastic bottle with volume graduations marked on it.

    b) Use the area delimiter to define a 12-cm^2 area on the upper surface of the substrate. Dislodge attached periphyton from the substrate within the delimeter into the funnel by brushing with a stiff bristled toothbrush, paint brush, or wire brush for 30 seconds. Take care to ensure that the upper surface of the substrate is the surface that is being scrubbed and that the entire surface within the delimeter is scrubbed.

    c) Fill a wash bottle with river water (do not use DI water). Using a minimal amount of water from this bottle, wash the dislodge periphyton from the funnel into the 500 mL bottle. Also wash the brush by spraying it with the squirt bottle into the 500 mL bottle as well. If no coarse sediment (cobbles or larger) are present:
        - Use the area delimiter to confine a 12-cm^2 area of soft sediments.
        - Vacuum the top 1 cm of sediments from within the delimited area into a de-tipped 60 mL syringe.
        - Empty the syringe into the same 500 mL plastic bottle as above.

    d) If possible, put the sample bottle in a cooler with ice while you travel between transects and collect the subsequent samples (the sample bottle needs to be kept cool and dark because a chlorophyll sample will be filtered from the composite).

2. Repeat steps 1 for Transects "B" through "K". Place the sample collected at each sampling site into the single 500 mL bottle to produce the composite index sample.

3. If all 11 samples are not collected, record the number of transects collected and reason for any missed collection on the field forms.

4. After samples have been collected from all 11 transects, thoroughly mix the 500 mL bottle regardless of substrate type. Record the total estimated volume of the composite sample in the periphyton section of the *Periphyton Collection Form*.

5.5.4 Sample Processing in the Field

You will prepare two different types of laboratory samples from the composite index samples: a chlorophyll-*a* and a biomass sample (for ash-free dry mass [AFDM]). Please refer to Section 6.0 for processing the periphyton samples.

5.6 Benthic Macroinvertebrates

5.6.1 Summary of Method

Benthic macroinvertebrates are also commonly referred to as: benthos, inverts, macroinverts, macroinvertebrates, or simply "bugs". Collect benthic macroinvertebrate composite samples using a D-frame net with a 500 µm mesh opening. 8 "kick samples" will be made at different locations within the reach. The material collected in the d-net from each kick will ultimately be composited into one sample after processing. Riffle habitat should be targeted when sampling for bugs, but if a site is lacking this, then edge habitat should be sampled. Also try to target coarser substrates (coarse gravel and bigger) rather than finer ones. If coarse substrates are lacking look for woody debris, macrophyte, or leaf packs to sample in. Composite all sample material and field reserve with 95% ethanol (ETOH). Macroinvertebrate sampling should take place before or during periphyton sampling.

The reason riffle habitat is targeted is because these conditions tend to yield the greatest diversity of bug species in stream ecosystems. A key factor of the UCASE program, in general, is capturing bug diversity at sites. Riffles tend to have more food flowing through them, as well as consistent temperatures and oxygen levels. Because riffles tend to be the shallower areas of a stream system, they tend to get more light which yields higher algae and diatom development. Alage and diatoms are a large food source for certain bug species. Also, since riffles tend to be in more contsricted parts of the stream it forces objects to become more funnelled and concentrated thus creating more food potential.

5.6.2 Equipment and Supplies

Table 5-19 shows the checklist of equipment and supplies required to complete the collection of benthos. This checklist is similar to the checklist presented in Appendix A, which is used at the base location to ensure that all of the required equipment is brought to the site. Record collection data on the *Sample Collection Form* (Figure 5-3).

Table 5-19. Equipment and supplies list for benthic macroinvertebrate collection at wadeable sites

For collecting samples	• Modified kick net (D-frame with 500 µm) with at least a 4 ft handle or a modified surber with at least a 4 ft handle. • Watch with timer or stopwatch • Plastic buckets 8 to 10 qt. • Sieve bucket with 500 µm mesh openings • Plastic forceps • Wash bottle; 1 L capacity labeled "Stream water" • Sample jars suitable for use with ethanol; usually 1 L HDPE Nalgene sample bottles • 95% ethanol (ETOH) in proper container • Bottle caddy • Electrical tape • Scissors or knife • UCASE Field Manual or Quick Reference Guide • Waterproof neoprene gloves
For recording Measurements	• Composite benthic sample labels (BENTHOS 5163 or 5523) • Blank waterproof sample tags for inside benthos jars (BENTHOS JAR TAG (INTERIOR)) • Lead pencils • Fine tipped indelible markers • Sample Collection Form • Clear tape strips

5.6.3 Sampling Procedure

Table 5-20 explains how samples are collected at wadeable sites. The kick methods used from UCASE are derived from the EPA's Wadeable Streams Assessment, which provides continuity for DWQ to make statewide assessments similar to what EPA does on a nation-wide scale. Utah DWQ has made a few changes from the original Wadeable Streams Assessment protocol, but overall, methods are still consistent with the original project.

Collect a sample at 8 locations throughout the sampling reach making an attempt to target riffle habitat and coarse substrates (coarse gravel and bigger). If these conditions are lacking or not present at a site, due to slow moving water or a stream dominated by fines, sample edge (bank) habitat[1]. Edge habitat is areas along the stream banks that offer refuge for bugs. Often, overhanging vegetation, sticks, and other material will offer protection for bugs to colonize. If possible, try to sample at different locations in the stream at various locations (e.g. left bank, right bank, center of stream). Multiple kicks/sweeps can be sampled in one transect if desireable habitat is lacking in the reach. If desireable habitat lands near a cross-section, sample at least 1

m above or below it since chances are other members of the crew may have disrupted conditions from taking other measurements.

[1]In many cases, Utah streams will lack or be absent of riffles and coarse sediments (e.g. desert streams in Southern Utah predominantly characterized by glides and fine/sandy sediments). It is important to target edge habitat in these cases to get a representable invert sample.

At each of the 8 sampling points in the reach determine which transect the sample is being collected in, the channel unit type (pool, glide, riffle, or rapid), and the dominant substrate (fines/sand, gravel, coarse, or other). Other inlcudes things like wood, leaves, edge habitat, overhanging vegetation, bedrock, hardpan, etc. Record this information by filling out the *Sample Collection Form* as shown in Figure 5-3. For this method, it does not matter what sequence the individual kicks are collected in (i.e. do not have move in an upstream to downstream fashion or in sequential order of the transects), just as long as they are not being collected in an area that has been disturbed from other sampling activities. Combine all sub-samples into a solid bucket as you move from transect to transect. If the site has low detritus and not much material is being collected in the net after each kick, it is okay to leave the material in the net and not empty it into a 2.5 gallon solid bucket right away. However, if detritus is heavy, you may have to periodically empty the contents in to a bucket as you make kicks throughout the site.

5.6.4 Sample Processing in the Field

Once the composite sample from all stations is sieved and reduced in volume, store in a 1 L jar and preserve with 95% ethanol. Do not fill jars more than 40% of material. Multiple jars may be required if detritus is heavy (Table 5-20and Table 5-21). Try to use no more than 5 jars at one site. The number of jars will be recorded on the *Sample Collection Form*. If multiple jars are used for one site be sure to fill out the bottle labels (both interior and exterior) accordingly. Be sure the inside labels and outside labels describe the same sample. Cover the exterior labels with clear tape.

It is recommended that crews carry an extra sample bottle containing ethanol with them to the site if it is not within close proximity to a field vehicle (be sure to label the bottle properly so that it is not mistaken for water) to enable them to immediately preserve the invertebrates. Doing so will help reduce the chance of aggressive specimens consuming others and prevent damage prior to the end of the field day. It is okay if the jar(s) are not completely filled while at the site just as long as there is enough to dispatch the bugs. The jars should, however, be completely topped off once final processing occurs back at the field vehicle.

Table 5-20. Procedure for benthic macroinvertebrate sampling at wadeable sites

1. Once all of the required equipment has been gathered to conduct benthos sampling, inspect the d-net/modified surber and sieve bucket for holes or tears. If there are holes, be sure to use a different net or bucket (crews should make it practice to carry a back-up set on every run).
    - If holes or tears are found in this equipment be sure to repair or replace it as soon as possible. Or, at least let the project coordinator know about the defects so that he/she can fix or replace it as soon as possible.
2. Find an area downstream of the reach (below Transect "A") and wash all equipment with stream water. Visually inspect the nets and buckets and make sure no particles are present on/inside of them. If they are, continue to wash the gear until it is clean. Triple rinse the spray bottle and fill it with stream water.
3. Proceed to the sampling reach with at least a net and a clipboard with the *Sample Collection Form* (a scratch piece of paper can be used to record information as well if it is easier to manage; transfer the data later onto the field form). If the stream has lots of fines and/or detritus, carry a 2.5 gallon bucket with you as well.
4. While walking along the reach, look for desireable habitat, which are riffle/runs with coarse substrates (coarse gravel or bigger). If riffle/run habitat exists proceed to Step 5. If not, proced to Step 12.

Riffle/Run Habitats:

5. With the net opening facing upstream, quickly position the net securely in the stream bottom to eliminate gaps under the frame. Avoid large rocks that prevent the net for seating properly on the stream bottom.

   NOTE: This is easier said than done it most cases epecially in high gradient cobble-dominated streams. Do the best you can and make an attempt to get this net as flush with the substrates as possible. If there are gaps underneath the net and you are unable to do anything about it, continue to sample. If there are issues or concerns, record them on the *Sample Collection Form*.

   NOTE: If there is too little water to collect the sample with the bug net, pick up the rocks in the stream that lie in the 1 ft^2 quadrat you would be kicking in (see the next step) and wash the organisms off them into a bucket using stream water.
6. Holding the net in position on the substrate, visually define a quadrat that is one net width wide and long upstream of the net opening. The area within this quadrat is $1ft^2$.
7. Check the quadrat for heavy organisms, such as mussels and snails. Remove these organisms by hand and place them into the net. Pick up loose rocks or larger substrate particles in the quadrat. Use your hands to dislodge organisms and wash them into the net. Scrub all rocks that are golf ball-sized or larger and which are halfway into the quadrat. After scrubbing, place the substrate particles outside of the quadrat.
8. Hold the bug net securely in position. Starting at the upstream end of the quadrat, vigorously kick the remaining finer substrate within the quadrat for 30 seconds (use a stopwatch if desired).

Table 5-20. Procedure for benthic macroinvertebrate sampling at wadeable sites *(cont.)*

NOTE: For samples located within dense beds of long, filamentous aquatic vegetation (e.g. algage or moss), kicking within the quadrat may not be sufficient to dislodge organisms in the vegetation. Usually these types of vegetation are lying flat against the substrate due to current. Use a knife or scissors to remove only the vegetation that lies within the quadrat (i.e. not entire strands that are rooted within the quadrat) and place it into the net.

9. Pull the net up out of the water. Immerse the net in the stream serveral times to remove fine sediments and to concentrate organisms at the end of the net. Avoid having any material enter the mouth during this operation.
10. Go to the next area with sampleable habitat.
11. Go to 16.

Pool/Glide Habitats:

12. Visually define a quadrat that is one net width wide and long at the sampling point. The area within the quadrat is 1 ft^2.
13. Check the quadrat for heavy organisms such as mussels and snails. Remove these organisms by hand and place them into the net. Pick up loose rocks or other larger substrate particles in the quadrat. Use your hands to dislodge organisms and wash them into a net. Scrub all rocks that are golf ball sized or larger and which are halfway into the quadrat. After scrubbing, place the substrate particles outside the quadrat.
14. Vigorously kick the the remaining finer substrate within the quadrat with your feet while dragging the net repeatedly throughout the area just above the bottom in a figure-eight pattern. Continue kicking the substrate and moving the net for 30 seconds.

    NOTE: If there is too little water to use the kick net, stir up the substrate with your hands and use a sieve with 500 µm mesh size to collect the orgranisms from the water in the same way the net is used in larger pools.

    NOTE: If you are at a site where beaver ponds are common or a lack of desireable habitat is present (e.g. riffle, coarse substrates), sample edge habitat by stirring up the bank with your foot and then sweeping the area in a figure eight motion with the net for 30 seconds. Or, stir up any overhanging vegetation, sticks, or other material with your hands or feet and then sweeping them in a figure eight motion for 30 seconds. Be sure not to drag the frame of the net against the bed of the stream as you well scoop up lots of fines and muck. Keep the net about 1 inch from the bed while sweeping.
15. After 30 seconds, remove the net from the water with a quick upstream motion to wash the organsisms to the bottom of the net.

Table 5-20. Procedure for benthic macroinvertebrate sampling at wadeable sites *(cont.)*

All samples:

16. Invert the net into a solid plastic bucket and transfer the sample if the net is full of detritus and/or substrates. To prevent bugs from being damaged during transport in the bucket, rinse and remove any large substrates. To do this, carefully
inspect coarse substrates and wash off any organisms into the bucket (using stream water) found still clinging to it before discarding the object. If the net is not full move onto the next sampling location and make your next kick, leaving any material from the previous kick in the net.
17. Determine the predominant substrate size/type you sampled from within the sampling quadrat. Fill in the approprate circle for the dominant substrate type for the transect on the *Sample Collection Form*. If carrying a clipboard and the data sheets is too cumbersome, you can keep record data in a small notebook or scratch piece of paper and transfer it to the actual data sheet when you are done sampling.

    NOTE: If there are co-dominant substrate types, you may fill in more than one circle; note the co-dominants in the comments section of the form.

    - **F**ine/sand: not gritty (silt/clay/muck <0.06 mm diam.) to gritty, up to lady bugged sized (2 mm)
    - **G**ravel: fine to coarse gravel (ladybug to tennis ball sized; 2 mm to 64 mm)
    - **C**oarse: Cobble to boulder (tennis ball to car sized; 64 mm to 4000mm)
    - **O**ther: bedrock (larger than car sized; >4000 mm), hardpan (firm, consolidated fine substrate), wood of any size, aquatic vegetation, etc.). Note type of "other" substrate in comments on field form.

18. Identify the habitat type where the sampling quadrat was located. Fill in the appropriate circle for channel habitat type for the transect on the *Sample Collection Form*.
    - **P**ool; Still water; low velocity; smooth, glassy surface; usually deep compared to other parts of the channel.
    - **GL**ide: Water moving slowly, with smooth, unbroken surface; low turbulence.
    - **RI**ffle: Water moving, with small ripples, waves, and eddies; waves not breaking and surface tension is not broken; "babbling" or "gurgling" sound.
    - **RA**pid: Water movement is rapid turbulent; surface with intermittent "white water" with breaking waves; continuous rushing sound.

Record information for each composite sample on the *Sample Collection Form*. If a sample requires more than one jar, make sure the correct number of jars for the sample is recorded on

Sample Collection Form. Place the samples in a cooler (no ice), bottle tote, or other secure container for transporting.

Table 5-21. Procedure for preparing composite samples for benthic macroinvertebrates at wadeable sites

1. Empty the contents collected in the d-net into a 2.5 gallon solid bucket, if you did not empty it at each transect.
2. You may have to re-invert the net, flush any remaining material or organisms to the bottom of the net using the stream, and empty material into the bucket in order to effectively clean the net.
3. Inspect the net for any remaining bugs that may still be clinging to it. Using a wash bottle full of stream water and/or forceps, flush/pick them off the net and into the bucket.
4. Once the net is cleaned from any materials or organisms and into the bucket, this becomes the composite for the sample reach.
5. Fill the bucket with stream water a few inches above the material line. Start swirling the bucket in a slow, but consistent fashion for about 7 seconds so that lighter material (sticks, leaves, organisms) in the bucket come to the surface and heavier material (substrates) stay at the bottom. While the material in the bucket is still moving in a circular motion, slowly pour the water into the sieve-bucket making sure not to dump any of the heavier material at the bottom of the bucket with it. Repeat this step several times until no more bugs are seen crawling around in the 2.5 gallon solid bucket. This process is known as sieving.

   NOTE: If there is an abundance of pebbles of cobbles in your sample, you will need to rinse (scrub if necessary) these off in the bucket with ample amount of water and then discard them.

   NOTE: Do not attempt to sieve samples with an abundance of filamentous algae. If this is the case, simply include all of the algae into the sample jar since it is difficult to effictively process these kinds of samples in the field.

6. Ultimately, you will end up with a 2.5 gallon bucket with coarse gravel and fines in it and a sieve bucket with organsisms and detritus in it (it is okay if some fine sediments are present in the sieve bucket). Once the sieving process is complete, you can dump the heavy material left in the 2.5 gallon bucket into the stream or on the ground.
7. Place the material in the sieve bucket into a 1000 mL jar making sure not to fill it more than 40% full with sample material. Be sure not to grab such a large handfull where material will become dislodged on the mouth of the bottle when you are filling it. Use multiple jars if necessary, but avoid using more than 5 per site. Keep in mind some material will stick to your hands during each transfer. It is a good idea to rinse your hands in the sieve bucket each time you put material in a bottle.
8. As the volume of material becomes less abundant at the bottom of the sieve bucket you will need to to wash the remaining contents to one side of the bucket in order to get the rest of the sample by gently agitating the bottom outside portion of it.

9. Once you think you have gotten everything out of the sieve bucket, carefully examine it for any remaining organisms. If there are still visible organisms either use a a pair of forceps to pick the bugs out. Or, you can tip the sieve bucket upside

Table 5-21. Procedure for preparing composite samples for benthic macroinvertebrates at wadeable sites *(cont.)*

down and spray the bottom side of it with rinse water into a funnel that is in the sample jar so that the bugs get washed out.

NOTE: If you choose to spray the sieve bucket as a final precaution, but end up filling the sample jar with too much water (>1/3 full) pour it off into the sieve bucket and re-spray with a smaller volume of water.

10. Place a properly labeled waterproof tag (Figure 2-2) inside of each jar (each tag for the site should be filled out completely the same except for the Jars "N" of "X" line).

NOTE: Always fill out these labels using a pencil. Ink will fade due to the ethanol.

11. Completely fill jar with 95% ethanol (no headspeace). It is very important that sufficient ethanol be used or the organsisms will not be properly preserved. Existing water in the jar should not dilute the concentration of ethanol below 70%.

NOTE: Samples can be transported back to the vehicle before adding ethanol if necessary. However, if site is a fair distance from vehicle (i.e. crew had to hike into site a ways) a liter of ethanol should be taken to the site with you. Fill the bottles to at least the detritus line then completely fill the rest of the bottle once back at the vehicle.

12. Replace the cap on each jar. Slowly tip the jar to a horizontal postion, then gently rotate the jar to mix the preservative. Do not shake the jar. After mixing, seal each jar with electrical tape.
13. Place an exterior label (Figure 2-2) on the outsie of the jar making sure they coincide with the interior tag (e.g. Jar "N" of "X" match with both labels). Cover it with clear tape.
14. Store filled bottles in an empty cooler or bottle tote during transportation until they can be stored in the appropriate location at the Shop.

5.7 Fish

5.7.1 Summary of Method

The fish sampling method is designed to provide a representative sample of the fish community, collecting all but the rarest fish taxa inhabiting the site. It is assumed to accurately represent species richness, species guilds, relative abundance, and anomalies. The goal is to collect fish community data that will allow the calculation of an Index of Biotic Integrity (IBI) at each site. Backpack electrofishing is the preferred method. If electrofishing is not possible at the site due to safety concerns, high turbidity, extremes in conductivity, difficult shocking conditions, lack of

water, etc., write "Not Sampled" and why in the comments section of the *DWQ Fish Field Sheet* (Figure 5-23 and Figure 5-24).

An attempt should be made to electroshock every site especially the ones that are part of the probabilistic survey. In most circumstances, crews usually will electroshock Transects "A-F". However, if site conditions are better for both sampling and results, it is okay to sample Transects "F-K". Or, if the crew feels the entire reach should be sampled and have adequate time, it is okay to do so as well. No matter which transects are selected be sure to record the length of the reach that was sampled and which transects were sampled on the field form. Before electrofishing begins, the crew should decide together as to whether they want to shock half of the site ("A-F" or "F-K") or the entire site.

In all instances electrofishing in wadeable systems should proceed in an upstream direction using a single anode. The number of netters can be variable just as long as it is recorded on the data sheet in the proper field. There needs to be at least one netter at all times and this should not be the person operating the electroshocker.

Electrofishing should be done after all other samples have been collected at the site. This will usually be the last activity at the site besides site clean-up and sample processing.
An electrofishing permit must be obtained every season from the Division of Wildlife Resources (DWR). It is important that the field coordinator for the UCASE program applies for one of these early on in the year before sampling begins. There is a form available online that must be filled out known as a Certificate of Registration (COR) in order to receive a permit. DWR personnel from the Salt Lake City office can be contacted for more information regarding permit information.

It is important to take photos of all fish species found at sites even though the crew might be keen on their idenitifcation skills. Having photos from each sites serves as a useful reference in case of a possible misidentification found during data review far after sites have been sampled. Good photos can also be used as a guide for new crew members who might not be familiar with their fish identification. Photos can also be useful to confirm fish species with DWR officials to make sure crew members are identifying different species correctly. Not every specific feature of the fishes need to be captured in the photos, but photos of things like the broad side, top side, mouth parts, flared fins, and photos of any other unique identifiers will suffice. It is very important to take detailed photos of mouth parts on sucker species as this is often the easiest way to identify sucker species. Detailed pictures of young-of-year specimens are important as well. In some cases, DWR will request that crews do not sample certain streams due to sensitive fish populations, endangered fish, or unique locations. Always respect and follow the requests of DWR personnel and verbiage written on the permit.

At times, crews will be asked to collect fish samples for heavy metal contamination (i.e. mercury, selenium, and PCBs)(Section 5.7.4). Crews will be informed before leaving on the trip by the field coordinator as to which sites will need these sorts of samples collected. If crews are asked to collect fish tissue samples they must print and read DWQs "Fish Tissue Hg" SOP. Refer to this SOP on the DWQ network (U:\PERMITS\MONITORS\QAQC\SOPs\Final\CURRENT VERSION) for complete details on field sampling procedures and an equipment list.

5.7.2 Equipment and Supplies

Table 5-22 shows the checklist of equipment and supplies required to complete the fish assessment. This checklist is similar to the one presented in Appendix A, which is used at the base location to ensure that all of the required equipment is brought to the site. Record fish collection data on the *Fish Collection Form*.

Table 5-22. Equipment and supplies-fish assessment at wadeable sites

For collecting samples	• Electrofishing equipment (including the backpack unit, wiring cables, anode, cathode, dip nets, neoprene or linesman gloves, thick waders (during warmer months, breathable waders can often conduct current due to sweaty or wet legs even if the person is wearing pants underneath), and necessary safety equipment) • Electroshocker batteries • Electrofishing permit • Camera • Neoprene gloves • 10% formalin (optional for preservation of unidentifiable fish) • 1000 mL jars (optional for transport of unidentifiable fish) • Non-conducting dip nets with ¼" mesh • Measuring board • Fish ID keys • Field Operations Manual and/or Quick Reference Guide • Fish Tissue Collection SOP (for field procedures and equipment list)
For recording measurements	• DWQ Fish Field Sheet • Lead pencils

Figure 5-23. DWQ Fish Field Sheet (front)

Reviewed by (initial): _____
Updated: 03/2011

DWQ Electro-shocking/Fish Tissue Collection Field Sheet

Please use a new sheet for each site (do not combine multiple sites on one sheet)

Site Name:					
STORET # (not applicable for DWR personnel):					
County:					
Date:					
GPS Coordinates		Degrees	Minutes	Seconds	Other (decimal degrees/UTM)
Datum:	Latitude				
	Longitude				
Electro-shocking Reach Length (m):					
Shocker Settings					
Shocking Time (s):					
Volts:					
Pulse Rate (Hz):					
Pulse Width (ms):					

1. Tally final counts for each species and their appropriate sizes in the circles. 2. Species codes on backside of this sheet	ELECTRO-SHOCKING DATA				
	Size 1: 0-60mm (0-2.36 in)	**Size 2: 61-200mm** (2.40-7.87 in)	**Size 3: 201-300mm** (7.91-11.81 in)	**Size 4: 301-400mm** (11.85-15.74 in)	**Size 5: >401mm** (>15.78 in)
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯

Comments:	Number of Netters:

Figure 5-24. DWQ Fish Field Sheet (back)

Reviewed by (initial): _____
Updated: 03/2011

Fish Tissue Data (Hg)

Sample ID*	Length (mm)	Weight (g)	Comments
01			
02			
03			
04			
05			
06			
07			
08			
09			
10			
11			
12			
13			
14			
15			
16			
17			
18			
19			
20			
21			If there are more than 21 samples, use another sheet and staple all the sheets together.

*Sample ID=STORET-Fish Code (listed below)-Unique Sequence ID per Site (i.e. 4982100CTT01)

Collector(s) Names (for Hg collection only):

_____ _____ _____

Species Codes: _____ _____

Black bullhead (BBH)	Least chub (LEC)	Utah sucker (UTS)
Black crappie (BLC)	Leatherside chub (LSC)	Virgin spinedace (VSD)
Bluehead sucker (BHS)	Longnose dace (LND)	Walleye (WLE)
Bluegill (BLG)	Mountain whitefish (MWF)	Wiper (WIP)
Bonneville whitefish (BWF)	Mountain sucker (MTS)	White bass (WHB)
Bonneville cisco (BCI)	Mottled sculpin (MOS)	Woundfin (WFN)
Brook trout (BKT)	Paiute sculpin (PTS)	Yellow perch (YLP)
Brown trout (BRT)	Rainbow trout (RBT)	
Channel catfish (CCF)	Redside shiner (RSS)	
Common carp (CMC)	Red-shiner (RES)	Note: Not all spp found in Utah are listed here. If crew collects a spp that is not listed then hand write the spp name on the front side of the sheet under "Species code"
Cutthroat trout (CTT)	Roundtail chub (RTC)	
Desert sucker (DSS)	Smallmouth bass (SMB)	
Flannelmouth sucker (FLS)	Speckled dace (SPD)	
Fathead Minnow (FHM)	Splake trout (SPT)	
Green sunfish (GSF)	Striped bass (STB)	
Kokanee (KOK)	Tiger muskie (TGM)	
Lake trout (LKT)	Tiger trout (TGT)	
Largemouth bass (LMB)	Utah chub (UTC)	

5.7.3 Sampling Procedure

Once all other sampling activities have been completed at the site, the entire crew should gather to help conduct electrofishing procedures. The first reason is these efforts usually require more than 2 people to sufficiently complete the activity and the second is for safety reasons. No crew members should remain in the stream for whatever reason while other members are electrofishing. Even if certain crew members are not going to participate with electrofishing efforts, they should not be in any portion of the stream while this procedure is happening. It is important for crew members to let others know when they are ready to start electrofishing so that everyone is on the same page. If the site is situated in a public place where people are watching you or are in the water, it is important that crew members take the time to inform them that they need to keep a clear distance while you are electrofishing.

While one crew member wears the backpack shocking unit, another should carry a live well (5 gallon bucket), if necessary, and record counts. The third person should do the netting. If a fourth or fifth person is available they can help with netting as well. Crews will calibrate the backpack electrofishing unit below the transect they choose to start with (usually "A" or "F") and do a quick test run to make sure the unit is functioning properly. For information on operating procedures, maintenance, and troubleshooting reference the manufacturers operation manual (Smith-Root, Inc or Halltech Aquatic). Once the machine has been calibrated and is functioning properly, proceed to the cross section where you have chosen to begin, reset the internal timer on the machine, and begin shocking upstream. As fish are netted or identified the netter and shocker call out the species and size to the recorder that they can positively identify. Please note that not every fish has to be netted in order to be counted. As long as crew members can positively identify the species and size of the fish it can be legitimately be counted. Each fish does not have to be measured by a measuring board unless you are uncertain of the size. For DWQ shocking efforts, fish lengths are categorized into 5 size-classes. As crew members identify fish species and determine the length, instead of calling out the specific length they call all the size class the length falls in (i.e. 1-5); see the field sheet for more information.

Make an attempt to shock all of the areas in the reach. Sometimes, due to stream size, depth, and swiftness, this is not always possible. Make an attempt to sample all habitats to the best of your abilities. If there are areas of the stream that cannot be shocked record these comments on the field form.

While electrofishing activities are occurring, at least one person should be carrying a camera to take photos of different characteristics of all species found at the site. Once the crew has shocked at least half of the site, electrofishing can cease. The crew can continue shocking in the reach if they choose to just as long as the reach distance is properly recorded on the field form. Once the crew is finished shocking, make sure to record the final effort time and tally counts on the field form. Document any unidentified species in an organized manner such that they can be properly identified and recorded at a later date. Turn off the backpack electrofisher and properly store it for transport. Table 5-23 presents the procedure for electrofishing in wadeable streams.

Table 5-23. Procedure for electrofishing at wadeable sites

1. Review all collecting permits to determine if any sampling restrictions are in effect for the site. In some cases, you may have to cease sampling if you encounter certain listed species.

2. Shocking should occur at every site unless there are safety issues. If the crew as a whole is absolutely certain that no fish are present at the site (e.g. lack of flow, high salinity) they can choose not to shock the site, but must record why in the comments section of the field form. However, if crews do not have sufficient evidence that a fish population is not present they should never assume one is not present and will conduct a fishing survey. If no fish are found, indicate such in the comments section of the field form.

3. Electrofishing will always occur in an upstream manner.

4. Crews must sample at least half of the reach. It does not matter where they start just as long as the total shocking length is at least half the site reach and does not extend downward from transect A or upward from transect K. It is okay to target more desirable fish habitat for this survey.

5. Proceed a short distance (~3-5 m) below transect where survey will begin. Make sure there are no safety hazards or potential hazards in the area.

6. Turn on the electroshocker and calibrate the unit (see the Smith-Root user's manual for more direction) using the Quick Setup function. The Smith-Root LR-24 Electrofishing device automatically calibrates its settings depending on the water quality at the site. However, crew members should be familiar with the different output ranges the unit should be in depending on stream conditions. The unit should always be set to pulsed DC. For the most part, a 30 Hz pulse rate will always be used. If mostly small fish (length <200 mm) a pulse rate of 60-70 Hz might be used. Start the electroshocker, set the timer, and depress the switch to begin fishing. The Halltech unit does not calibrate itself entirely. Some technique is required to establish the correct pulse ranges to effectively shock fish. The manual will provide more information as well as an experienced crew member. If fishing success is poor, increase the pulse width first and then the voltage. Increase the pulse rate last to minimize mortality or injury to large fish. If mortalities occur, first decrease the pulse rate, then voltage, then pulse width. Start cleared clocks. Note, some electrofishers do not meter all the requested header data; provide what you can. If button time is not metered, estimate it with a stop watch and flag the data.

7. Once the settings on the electrofisher are adjusted properly to sample effectively and minimize injury and mortality, proceed to the transect cross-

section where you have chosen to start the survey. Depress the switch and slowly sweep the electrode (anode) from side to side. Sample all habitats and available cut-bank and snag habitat as well. Move the anode wand into cover with the current off, turn the anode on when in cover, and then remove the wand quickly to draw fish out. In fast, shallow water, sweep the anode and fish downstream into a net. Be sure that deep, shallow, fast, slow, complex, and simple habitats are all sampled. In stretches with deep pools, fish the margins of pool as much as possible, being extremely careful not to step or slide into deep water. Keep the anode near the cathode if fish catch is low.

8. There must be at least one person netting while shocking, but there is no limit to the maximum number of netters. Be sure to indicate the number of netters on the field form. The netters, with the net 1 to 2 ft from the anode, follows the operator, nets stunned individuals, and identifies the fish and calls out the species and size to the person recording data. Keep in mind the fish do not have to be
netted in order for them to be counted. As long as they can be positively identified in some way they can be counted. If fish are being stunned hard, place it in a half filled 5-gallon bucket that the recorder should carry so that the fish can recover appropriately before being released.

   Note: If a crew is unable to identify a particular species on site they will need to take photos of it or collect a specimen for identification at a later time. In this case, make up a unique identifier code that can be used temporarily on the field form and can be linked some way to that particular species (i.e. Unk Fish 1; Red-striped fish; etc). This way the crew can still recognize the fish and take counts without knowing the species name at the site. The field sheet will need to be updated once the fish is identified at a later date (Section 5.7.5).

9. Continue upstream until the last transect has been electro-shocked.

10. Record the time in seconds on the field from the electrofisher. Tally all species counts and record counts on the field form. Make sure the field form is legible and complete. These sheets will ultimately be submitted to and used by other agencies outside of DWQ.

11. Turn off the electro-fisher.

12. Release any fish that might still be in the 5-gallon bucket if applicable.

5.7.4 Sampling for Fish Tissue Specimens

If the crew is collecting fish for fish tissue analysis one of the crew members should be carrying a live well (usually a 5 gallon bucket) while electrofishing. Once electrofishing begins and crew members start netting fish, they should keep in mind they need to keep 5 fish of the largest size class of each species found at the site. While crews move in an

upstream fashion they should be keeping and replacing fish in the live well with fish in the largest size class found at the site. In some scenarios crews will often begin netting fish at the site collecting a particular size class of fish only to find a bigger size class as they continue up the reach. This is why it is important to keep fish right away in the live well and replace them with bigger fish as you continue shocking upstream. If crews are unable to obtain 5 fish samples of the same size class within the site reach, it is okay to target fish habitat beyond the UCASE reach, both upstream and downstream, in order to get the desired sample size of 5. However, crews should NOT include any fish counts on the field sheet that are outside the UCASE reach layout. If this is the case, crews should record their shocking effort given in seconds on the electrofisher once the reach has been completed then proceed upstream or downstream with the electro-fisher and shock only desirable fish habitat until the targeted sample size is collected. The shocking time does not need to be documented at all if electrofishing occurs outside of the UCASE sampling reach. If 5 fish of each species just cannot be obtained in a reasonable manner, keep the fish that have already been collected and process them according the Fish Tissue Sampling SOP Add website link to SOPs Unidentifiable Fish

Often, crews will be unable to identify fish species depending on the fish taxonomy experience amongst the crew. It is better to put 'Unknown' on the field form than guess what the species is. If crews are unable to identify the species, but can identify the genus, it is okay to record this on the field sheet rather than guess or leave a blank field. If a fish cannot be identified in the field, an effort should be made after sampling to identify the species whether it is at the base location or back in the office. There are resources through DWR that are willing to help us identify fish to their species. See the UCASE field coordinator for contacts to DWR field offices who can assist you further. If you choose to use DWR to help identify and unknown fish you must take a series of photos or submit a specimen. Photos are preferred as it reduces the chances of accidentally "taking" a designated sensitive, threatened, or endangered species from a site. If the crew is certain the species is not protected they can keep a specimen, preserve it, and submit it to DWR staff at a later date.

Photos:

If pictures are taken of a specimen it is important to take clear descriptive photos and not necessarily a lot of them. The key characteristics to capture on any fish species when taking photos are the following:

- A broad/overall shot of the topside and broadside of the specimen.
- A fanned view of the tail/caudal fin AND the dorsal fin.
- The mouth (especially the jaw lines and scraping features on sucker species).

Once the sampling trip is over, a member of the crew should submit these photos within two weeks of collection to a DWR representative. Once the results are returned, updates should be made to the field sheets as soon as possible. Photos should be ultimately stored in the electronic site folder packet in the UCASE folder in the Monitors directory.

Specimen Collection:

If crews decided to keep a specimen from the site, they need to place it in an appropriate sized container at the site (e.g. centrifuge tube, benthos bottle, etc.) containing a 10% buffered formalin solution in a volume equal to or greater than the total volume of specimens. Any type of container will suffice as long as it does not leak. Individuals larger than 200 mm in total length should be slit along the right side of the fish in the lower abdominal cavity to allow penetration of the solution. Anyone who is handling formalin should read the Material Safety Data Sheet (MSDS) written for this substance.

Formalin is a potential carcinogen and should be used with extreme caution, as vapors and solution are highly caustic and may cause severe irritation on contact with skin, eyes, or mucus membranes. Wear vinyl or nitrile gloves and safety glasses and always work in well ventilated areas.

For a safer preservative, crews can use ethanol (ETOH), which they should already have available for benthos preservation. However, ETOH will not preserve specimen characteristics as well as formalin. Often, specimens fade quickly when exposed to ETOH. If it is used, crews should submit the samples as soon as possible once preserved. In the field, crews should label a small piece of paper, in pencil, with the following information and place it in the bottle before preserving:

- Site Name
- Site ID
- Date Collected
- Unique identifier than can be linked to the field sheet (i.e. Unk Fish 1; Red-striped fish; etc).
-

A label, with the same information, should be placed on the outside of the jar as well. Place a piece of clear tape over the label and seal the lid with black tape. Once the sampling trip is over, a member of the crew should submit these jars within two weeks of collection to a DWR representative. Once the results are relayed from DWR back to the crew member updates should be made to the field sheets as soon as possible. The specimen can that be thrown out once the identification has been confirmed and properly recorded. Table 5-24 explains the proper way to photograph and track unidentifiable fish while in the field if preserving and taking a specimen is not an option.
Table 5-25 explains the procedure for collecting and preserving and unidentifiable species for lab submittal if this is the preferred choice.

Table 5-24. Procedure for taking photos for unidentifiable fish

1. While electrofishing, if an unidentifiable fish is collected crew members should first give the specimen a unique identifier so that it can be linked between the photos and field form at a later date (i.e. red-striped fish; minnow-like species; Unk 1, etc.).
2. Take a broad photo of the top-side, bottom-side, and broad-side of the specimen. Proceed with taking detailed photos of the dorsal and caudal fin/tail fin (detailed photos include being able to see individual spines of the fins). Then, take different photos of the mouth. If the specimen is a sucker species,

take adequate photos of the lips and scraping features of the mouth. These features are often key in distinguishing sucker species from apart.

3. Record the photo tag ranges on the field form.

4. Release the fish.

5. Once back at the base site or in the office, try to identify the specimen using field guides, keys, or online resources. If the specimen is identified go back to the field form and erase the unique identifier and replace it with the proper fish code. This completes this procedure.
   If there is not enough time to do this or crew members feel they have tried enough to key the species, move on to the next step.

6. Contact the UCASE field coordinator and ask him/her to refer you to a DWR contact in the region where the fish was found. Send all pictures of the specimen, coordinates, and a brief site description to the person in an email format.

7. Once correspondence has occurred and results are given, update the field form.

   Note: All fish photos and site photos should be saved to the electronic folder in the UCASE folder in the Monitors directory once the crew arrives back to the office and follows data management procedures (U:\PERMITS\MONITORS\UCASE)

Table 5-25. Procedure for collecting an unidentifiable fish specimen

1. While electrofishing, if an unidentifiable fish is collected crew members should first give the specimen a unique identifier so that it can be linked between the label on the specimen jar and field form at a later date (i.e. red-striped fish; minnow-like species; Unk 1, etc.).

2. Keep the fish in a live well (like a 5-gallon bucket).

3. Once electrofishing has ceased, dispatch the fish.

4. Place the specimen in a jar that is larger than the volume of the specimen. It does not matter what kind of jar is used as long as it does not leak.

5. Label a small piece of paper in pencil with the site name, site ID, date collected, and the unique identifier and place it in the jar.

6. Place the specimen in the jar.

7. Fill the jar with a 10% buffered formalin solution in a volume equal to or greater than the total volume of specimens. Individuals larger than 200 mm in total

length should be slit along the right side of the fish in the lower abdominal cavity to allow penetration of the solution. If denatured alcohol (ETOH) is preferred over formalin, go to step 8.

Note: If fish are going to be retained with the crew for longer than 2 weeks they need to change out the formalin solution in the jar. Following the fixation period of 2 weeks, the volume of formalin should be properly discarded and replaced with tap water for soaking specimens over a 4-5 day period. Soaking may require periodic water changes and should continue until the odor of formalin is barely detectable. Final storage of specimens is done in 45%-50% isopropyl alcohol or 70% ethanol. **Formalin is a potential carcinogen and should be used with extreme caution, as vapors and solution are highly caustic and may cause severe irritation on contact with skin, eyes, or mucus membranes. Wear vinyl or nitrile gloves and safety glasses and always work in well ventilated areas.** Formalin must be disposed of properly. Contact the Utah State Health Lab to properly handle and dispose of formalin.

8. If denatured alcohol (ETOH) is the preservative of choice, after step 6, fill the jar full of ETOH and move on to step 9. Keep in mind that although ETOH is a simpler and safer way to preserve fish, it also does not preserve key characteristics of fish as well as formalin. When specimens are very small and/or faded it is probably best to use formalin as ETOH will fade and maybe destroy the characteristics even more.

9. Fill out a label with the same information that was put on the interior label an tape if with clear tape. Seal the jar with electrical tape and store the jar in a safe upright position for the remainder of the trip.

10. Contact the UCASE field coordinator and ask him/her to refer you to a DWR

11. contact in the region where the fish was found. Set up a time and location to transfer the specimen.

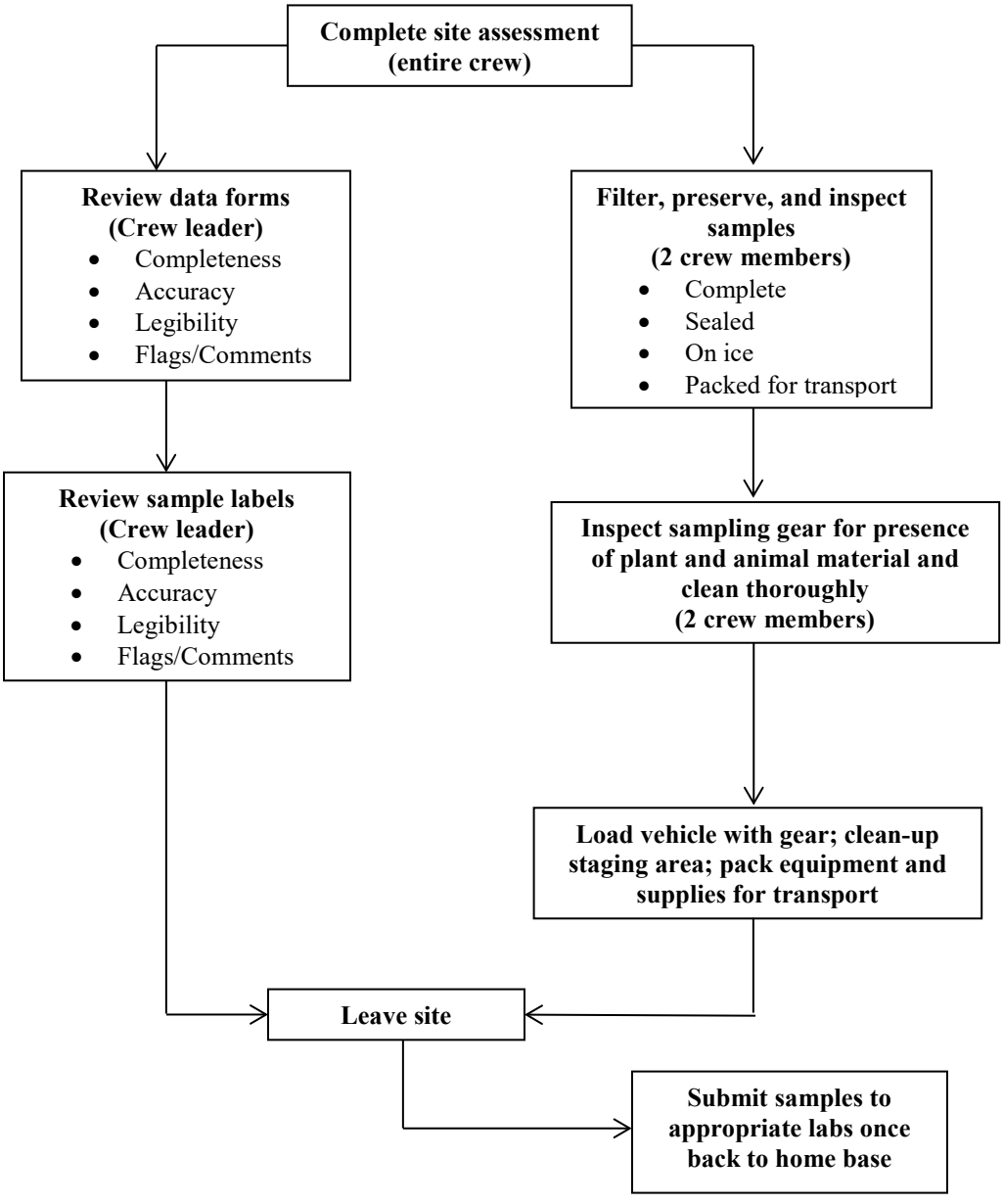
12. Once correspondence has occurred and results are given, update the field form. *Note:* All fish photos and site photos should be saved to the electronic folder in the UCASE folder in the Monitors directory once the crew arrives back to the office and follows data management procedures (U:\PERMITS\MONITORS\UCASE)

6 FINAL SITE ACTIVITIES

Prior to leaving the site, make a general visual assessment of the site and its surrounding catchment and watershed. The objective of the general site assessment is to record observations of watershed, catchment, and site characteristics that are useful for future data interpretation, ecological value assessment, development of associations, and verification of stressor data. Your observations and impressions are extremely valuable.

You will filter and process the chlorophyll-*a* and ash-free dry mass samples. Conduct a final check of the data forms, labels, and samples. The purpose of the second check of data forms, labels, and samples is to assure completeness of all sampling activities. Finally, clean and pack all equipment and supplies and do any clean-up of the staging area where post sampling activities have occurred, if need be (i.e. parking lot, campground, etc.). Activities described in this section are summarized in Figure 6-1.

Figure 6-1. Final Site Activities



6.1 General Site Assessment

Complete the *Site Assessment Form* (Figure 6-2) after sampling, recording all observations from the site that were noted during the course of the visit. The *Site Assessment Form* is designed as a template for recording pertinent field observations. It is by no means comprehensive, and any additional observations should be recorded in the "General Assessment" section.

6.1.1 Watershed Activities and Disturbances Observed

Record any of the sources of potential stressors in the "Watershed Activities and Disturbances Observed" section of the *Site Assessment Form* (Figure 6-2). Include those that were observed while on site, while driving or walking through the site and catchment/watershed, or while flying over the site and catchment/watershed. For activities and stressors that you observe, rate their abundance or influence as low (L), moderate (M), or heavy (H) on the line next to the listed disturbance. Leave the line blank for any disturbance not observed. The distinction between low, moderate, and heavy will be subjective. For example, if there are two to three houses on a site, circle "L" for low next to "Houses." If the site is ringed with houses, rate it as heavy (H). Similarly, a small patch of clear-cut logging on a hill overlooking the site would rate a low ranking. Logging activity right on the site shore, however, would get a heavy disturbance ranking. This section includes residential, recreational, agricultural, industrial, and stream management categories.

6.1.2 Site Characteristics

Record observations regarding the general characteristics of the site on the *Site Assessment Form* (Figure 6-2). When assessing these characteristics, look at a 200 m riparian distance on both banks. Rank the site between "pristine" and "highly disturbed," and between "appealing" and "unappealing." Document any signs of beaver activity and flow modifications as well as any signs of clams at the site. Clams populations are in rapid decline in stream ecosystems and are sensitive to ammonia. Their populations are in steady decline and or not as common in Utah stream systems as they once were. It is useful to know if populations are present at UCASE sites for documentation purposes and future monitoring. If clams are abundant at site or there are casings along the bank, collect and specimen in a baggie and submit it to the Field Coordinator. If it is not appropriate to collect a specimen take some photos of it and include them with the rest of the site photos. Also, record the dominant land type and forest age class near the site. Document the weather conditions on the day of sampling and any extreme weather conditions prior to sampling.

6.1.3 General Assessment

Record any additional information and observations in this narrative section. Information to include could be observations on biotic integrity, vegetation diversity, presence of wildlife, local anecdotal information, or any other pertinent information about the site or its catchment/watershed. Record any observations that may be useful for future data interpretation or observations that are unique to the site that are not usually seen elsewhere (e.g. observation of clam populations; Use this section to really describe the grazing characteristics of the site). Even though the Human Influence portion of the Physical Habitat sheet (Figure 5-10) rates the impact of grazing, it does not give enough detail on grazing activities in the area. Grazing is often the most common impact to UCASE sites. Use the *Site Assessment Form* to talk about specifics on grazing on what you observed at the site.

Figure 6-2. Site Assessment Form

UCASE QUALITATIVE SITE ASSESSMENT FORM

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ ___ DATE:___ ___/___ ___/ 2 0 ___ ___

WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED (Intensity: Blank=Not observed, L=Low, M=Moderate, H=Heavy)

L M H Residences	L M H Maintained Lawns	L M H Orchards	L M H Evidence of Wildfire	L M H Water Level Fluctuations
L M H Maintained Lawns	L M H Parks, Campgrounds	L M H Poultry	L M H Odors	L M H Fish Stocking
L M H Pipes, Drains	L M H Primitive Camping	L M H Irrigation Equip.	L M H Commercial Development	L M H Dams
L M H Dumping	L M H Trash/Litter	L M H Water Withdrawal	L M H Chemical Treatment	L M H Other:_____
L M H Roads/Motorized Pathways (i.e ATV)	L M H Surface Films	L M H Industrial Plants	L M H Angling Pressure	L M H Other:_____
L M H Hiking Trails	L M H Cropland	L M H Mines/Quarries	L M H Dredging	L M H Other:_____
L M H Bridges/Culverts	L M H Pasture	L M H Oil/Gas Wells	L M H Channelization	
L M H Sewage Treatment	L M H Grazing	L M H Power Plants		
		L M H Logging		

SITE CHARACTERISTICS (200 m radius from site reach)

Waterbody Character	**Pristine** ○5 ○4 ○3 ○2 ○1 **High Disturbed** **Appealing** ○5 ○4 ○3 ○2 ○1 **Unappealing**
Presence of Beaver	**Beaver Signs:** ○Absent ○Rare ○Common **Beaver Flow Modifications:** ○None ○Minor ○Major
Dominant Land Type	**Dominant Land Type at Site:** ○Forest ○Agriculture ○Range ○Urban ○Suburban/Town ○Recreation ○Other:_____
Presence of Clams	**Clam Signs:** ○Absent ○Rare ○Common ***Please collect a specimen if clams are abundant and submit to Field Coordinator. If a specimen cannot be collected please take photos and include them with the site photos.**
Weather (general description of weather over the last week)	

GENERAL ASSESSMENT (talk about things that you feel might not have fully been captured on the rest of the forms if there is a unique situation; biotic integrity; vegetation diversity; anecdotal information, etc.)

Updated: 01/2015

6.2 Processing Field Samples

6.2.1 Processing the Filtered Metals and Filtered Nutrient Samples

6.2.1.1 Equipment and Supplies

Table 6-1 lists the equipment and supplies needed to process the filtered metals and filtered nutrient samples.

Table 6-1. Equipment and supplies list for filtered metal and filtered nutrient sample processing

For filtering filtered metals and filtered nutrient samples	• Sample bottles • DI water • Forceps • Disposable cartridge filters • Geo-pump fitted with ~3 feet of tubing • Cooler • Ice
For data recording	• Lab sheets for State Lab • Pencil • Indelible Pen • Sample labels

6.2.1.2 Procedures for filtering the Filtered metal and Filtered nutrient samples

The filtered metal and filtered nutrient bottles are filled by filtering sample from the transport bottle that was filled with the other water chemistry samples by using a Geo-pump. These bottles are to be labeled the same as the chemistry and non-filtered nutrient bottles. Keep in mind that the chemistry and non-filtered nutrient bottles do not require processing besides proper labeling once they are filled. These are filled at the stream and then left alone after being put on ice. Filtered samples measure dissolved constituents in the water column at any given site. These samples should be filtered after they are collected as soon as possible. If the site is remote, then process these samples as soon as you get back to a vehicle. These samples are analyzed at the State Health Lab.
presents the procedure for filtering these samples. These procedures and a more in depth explanation of DWQ water chemistry sampling techniques can be found in the DWQ directory at: U:\PERMITS\MONITORS\QAQC\SOPs\Final\CURRENT VERSION.

Table 6-2. Procedure for filtering filtered metal and filtered nutrient samples

1. Rinse the outside of the intake tubing thoroughly with site water
2. Place the intake tubing into the transfer bottle
3. Turn on the pump
4. Flush the tubing with ~250 milliliters of site water, using the pump to pull the water through the tubing
5. Turn off the pump
6. Thoroughly mix the raw water sample to be filtered by gently inverting the sample container several times
7. Place a disposable cartridge filter on the outtake end of the tubing (opposite end)
8. Remove the caps from the sample bottles for the dissolved constituents
9. Turn on the pump and hold the filter holder over the sample bottle
10. Fill one bottle full, cap it and repeat for the other bottle
11. Be sure not to overfill the bottles. Overfilling would cause loss of the preservative and therefore some headspace must be remaining
12. Turn off the pump
13. Remove the used filter from the tubing and discard
14. Drain the rest of the water in the tubing by running the pump
15. Indicate which water chemistry samples were or were not collected on the *Sample Collection Form* by circling "Y" or "N"

6.2.2 Processing the Water Column Chlorophyll-*a* Sample

6.2.2.1 Equipment and Supplies (Water Column Chlorophyll-*a*)

This sample differs from the periphyton Chlorophyll-*a* sample in that it measures the chlorophyll particles suspended in the water column rather than what is scraped off rocks, sticks, and other material that is part of a composite sample. 2,000 mL is the targeted *and* maximum volume for this sample, but inevitably this will not always be possible due to turbid water. It is acceptable to filter less than 2,000 mL in this instance, however; it is very important to filter as much volume as possible to get good lab results. Record the volume of water processed for this sample on the *Sample Collection Form*. Table 6-3 lists the equipment and supplies necessary for processing Chlorophyll-*a* samples.

Table 6-3. Equipment and supplies list for Chlorophyll-*a* sample processing

For filtering chlorophyll-*a* samples	• Millipore 47 mm 0.7 micron glass fiber pre-filters • Filtration apparatus with graduated filter holder • Vacuum hand pump with tubing and pressure gage (geo-pump may be used if it has a gage) • Polypropylene filter flask; 250-500 mL • Analytical test filter funnels • #7 filter flask funnels with ½ inch hole • 50 mL centrifuge tubes • Aluminum foil • Forceps • Zip-top baggies • DI water in squeeze bottle • Electrical tape • Dry ice • Wet ice • Coolers
For data recording	• Sample collection form • Sample labels • Pencils • Indelible markers • Clear tape strips

6.2.2.2 Procedure for Processing the Water Column Chlorophyll-*a* Sample

This procedure can be carried out using a hand pump or a peristaltic pump (such as a Geopump) outfitted with hosing and a vacuum pressure gage. Regardless of what pumping method is used, the same filter flask and filter holder can be used, and operating pressures must not exceed 7 psi. The sampling procedure is presented in Table 6-4.

Table 6-4. Procedure for processing the Water Column Chlorophyll-*a* sample

NOTE: ***Decontamination Procedure:*** *Between samples, triple rinse the graduated cylinder, filter funnel, filter flask, and foreceps with DI water and shake dry as best as possible*

1. Attach tubing onto filter flask arm and pump.

2. Insert the funnel adapter into the #7 stopper; wetting the adapter may aid in this. Once the adapter is in the stopper it need not be disassembled unless a thorough cleaning of the entire apparatus is required.

3. Insert the filter funnel assembly into the top of the filter flask until it seals tightly

4. Remove the top of the filter funnel from the filter stage.

5. Using the forceps, place the glass fiber filter on the filter stage. Take care not to touch the filter or inside of the filter funnel with your hands to avoid contamination.

6. Wet the sides of the filter funnel and filter with DI water from the squeeze bottle to create a good seal on the filter.

7. Mix the water sample in its container multiple times to homogenize, and pour the sample into a graduated cylinder to measure 500 ml (or a smaller volume if sample is turbid).

8. Pour 250 ml (or a smaller volume) of sample water into the filter funnel and use the pump to pull the sample through the filter.

9. Continue filtering measured volumes of sample, being careful not to exceed 7 psi of vacuum pressure during filtration. When the filtration rate has slowed, or the filter has turned a green/brown color, sufficient amount of sample has been filtered. Do not exceed 2000 mL.

   NOTE: *If the sample is turbid, only filter small volumes of sample at a time. Try to estimate how much turbid water you can put through a filter by paying attention to how the flow decreases as you pull the water through the filter. If you overestimate the volume of water that can pass through the filter and water is left remaining on top of a clogged filter, filtering must be repeated with another volume of sample and a new filter.*

10. Rinse down the sides of the filter funnel with DI water and use the pump to pull the rinse water through the filter. You can stop filtering when the filter is still slightly wet; the filter need not be completely dry.

11. Remove the filter funnel being careful not to disturb the filter stage or filter.

12. Using forceps, remove the filter from the filter stage, being careful not to rip the filter or scrape off any green/brown residue.

13. Fold the filter in half with the residue facing the inside.

14. Place the folded filter onto a piece of aluminum foil and fold the foil to make a package for the filter.

15. Place the foil-wrapped sample into a small ziptop bag.

16. Fill out a sample label and stick/place it inside the baggie with the sample.

17. Place the sample into a larger ziptop bag with other chlorophyll samples from the trip and immediately store the samples on dry ice or place in the freezer.

6.2.3 Processing the Species ID-quantitative, Chlorophyll-*a*, and Dry Weight (AFDM) samples from the Periphyton Sample

6.2.3.1 Equipment and Supplies (Periphyton Sample)

Table 6-5 lists the equipment and supplies needed to process the periphyton samples.

Table 6-5. Equipment and supplies list for periphyton sample processing

For filtering periphyton samples	• Millipore 47 mm 0.7 micron glass fiber pre-filters • Filtration apparatus with graduated filter holder • Small graduated cylinder (i.e. 50 mL to 200 mL) • Vacuum hand pump with tubing (geo-pump may be used if it has a gage) • Graduated cylinder; 100 mL or less • Analytical test filter funnels • #7 filter flask funnels with ½ inch hole • 50 mL centrifuge tubes • Aluminum foil • Forceps • Zip-top baggies • DI water in squeeze bottle • Electrical tape • Dry ice • Wet ice • Coolers

For data recording	• Sample collection form • Sample labels • Pencils • Indelible markers • Clear tape strips

6.2.3.2 Procedures for Processing the Periphyton Samples

Three different types of laboratory samples are prepared from the composite index samples: a **Species ID-quantitative sample** (to determine taxonomic composition and relative abundances), a **Chlorophyll-*a* sample** and a **Dry Weight/Ash Free Dry Mass (AFDM) sample.**

6.2.3.3 Species ID-quantitative sample

Prepare the Species ID-quantitative sample as a 50 mL aliquot from the composite index sample, following the procedure presented in Table 6-6. Each sample container lid is taped with electrical tape and then placed in a zip-top baggie and placed in a cooler with ice. Be sure to completely fill out a Species ID label and stick it on a 50 mL centrifuge tube. Place a piece of clear tape over the label.

Table 6-6. Procedure for Species ID-quantitative sample of periphyton

1. Prepare a sample label for the Species ID-quantitative sample. The sub-sample volume should always be 50 mL from a 500 mL composite sample. If this is different for some reason, record the correct volumes on the labels AND the *Sample Collection Form.* Attach the completed label to a 50 mL centrifuge tube and completely cover the label with a clear tape strip.
2. Take the 500 mL composite periphyton sample and thoroughly mix it.
3. Rather quickly, poor off 50 mL of composite sample into the labeled centrifuge tube.
4. Preserve with a few drops of Lugols solution preservative.
5. Place the cap tightly back on the tube and seal it with a strip of electrical tape around the lid.
6. Place the sample in a plastic zip-top baggie and put it in a cooler with ice.

6.2.3.4 Chlorophyll-*a* Sample

Prepare the chlorophyll sample by filtering 25 mL aliquot of the composite index sample through a 47 mm 0.6 micron glass fiber pre-filter. The procedure for filtering chlorophyll samples is presented in Table 6-7. Chlorophyll can degrade rapidly when exposed to bright light. If possible, prepare the samples in subdued light (or shade), filtering as quickly as possible after collection to minimize degradation. Keep the glass fiber filters in a dispenser until use.

It is important to measure the volume of the sample being filtered accurately (+/- 1 mL) with a graduated cylinder. During filtration, do not exceed 14 inches of Hg (7 PSI) to avoid rupturing cells. If the vacuum exceeds 14 inches of Hg, prepare a new sample. If the filter clogs completely before all the sample in the chamber has been filtered, discard the sample and filter, and prepare a new sample using a smaller volume of sample. If this occurs, be sure to update the *Sample Collection Form* and sample label as both of these are pre-labeled to assume that 25 mL can always be filtered through (in only rare occasions can less than 25 mL be filtered through a filter occur).

Table 6-7. Procedure for preparing chlorophyll samples of periphyton

1. Using clean forceps (they can be re-used between sites, but they should be rinsed and cleaned with DI water from the previous site), place a Millipore 0.7-micron glass fiber pre-filter on the filter holder with the gridded side down.

2. Rinse the sides of the filter funnel with a small volume of DI water.

3. Rinse a small graduated cylinder 3 times with small volumes of DI water.

   NOTE: A clean centrifuge container works well for this step as it will have graduations on it.

4. Mix the composite sample thoroughly.

5. Measure 25 mL of aliquot into the filter funnel into the graduated cylinder.

   NOTE: For a composite sample containing fine sediment, allow grit to settle for 5-10 seconds before pouring the sample into the graduated cylinder.

6. Pour the 25 mL aliquot into the filter funnel, replace the cap, and pull the sample through the filter using the hand pump. Vacuum pressure from the pump should not exceed 14 inches of Hg (7 PSI) to avoid rupture of fragile algal cells.

   NOTE: If 25 mL of sample will not pass through the filter, discard the filter and rinse the chamber thoroughly with DI water. Collect a new sample using a smaller volume of sample, measure to +/- 1 mL. Be sure to record the actual volume sampled on the sample label and Sample Collection Form.

   NOTE: An electric pump (geo-pump) can be used for this process as long as it has a pressure gage on it and the same pressure limitations are used.

7. Remove the filter funnel from the filter holder being very careful to lift it straight up and not to the side as the sample will be scraped and ruined if this happens. Remove the filter from the holder with clean forceps. Avoid touching the colored portion of the filter. Fold the filter in half, with the colored sample (filtrate) side folded on itself. Place the folded filter onto a

small piece of aluminum foil and gently fold it into a square being sure not to ruin the filter or having it fold back open in the foil. Discard the filtered water.

8. Complete a chlorophyll-*a* (ALGAE) sample label and attach it to the inside of a plastic zip-top baggie. Place the aluminum foil square into the baggies and place it in a cooler with dry-ice.

6.2.3.5 Dry Weight/Ash Free Dry Mass (AFDM) Sample

This sample is prepared, processed, and handled the same exact way as the chlorophyll-*a* sample (Section 6.2.3.5). The only difference is that a different label is used. The following steps below are verbatim to Section 6.2.35 except for the label type:
Prepare the dry weight sample by filtering 25 mL aliquot of the composite index sample through a 47 mm 0.6 micron glass fiber pre-filter. The procedure for filtering dry weight samples is presented in Table 6-8. Keep the glass fiber filters in a dispenser until use. It is important to measure the volume of the sample being filtered accurately (+/- 1 mL) with a graduated cylinder. During filtration, do not exceed 14 inches of Hg (7 PSI) to avoid rupturing cells. If the vacuum exceeds 14 inches of Hg, prepare a new sample. If the filter clogs completely before all the sample in the chamber has been filtered, discard the sample and filter, and prepare a new sample using a smaller volume of sample. If this occurs, be sure to update the *Sample Collection Form* and sample label as both of these are pre-labeled to assume that 25 mL can always be filtered through (in only rare occasions can less than 25 mL be filtered through a filter occur).

Table 6-8. Procedure for preparing dry weight/AFDM samples of periphyton

1. Using clean forceps (they can be re-used between sites, but they should be rinsed and cleaned with DI water from the previous site), place a Millipore 0.7 micron glass fiber pre-filter on the filter holder with the gridded side down.

2. Rinse the sides of the filter funnel with a small volume of DI water.

3. Rinse a small graduated cylinder 3 times with small volumes of DI water. *NOTE: A clean centrifuge container works well for this step as it will have graduations on it.*

4. Mix the composite sample thoroughly.

5. Measure 25 mL of aliquot into the filter funnel into the graduated cylinder. *NOTE: For a composite sample containing fine sediment, allow grit to settle for 5-10 seconds before pouring the sample into the graduated cylinder.*

6. Pour the 25 mL aliquot into the filter funnel, replace the cap, and pull the sample through the filter using the hand pump. Vacuum pressure from the pump should not exceed 14 inches of Hg (7 PSI) to avoid rupture of fragile algal cells.

NOTE: If 25 mL of sample will not pass through the filter, discard the filter and rinse the chamber thoroughly with DI water. Collect a new sample using a smaller volume of sample, measure to +/- 1 mL. Be sure to record the actual volume sampled on the sample label and Sample Collection Form.
NOTE: An electric pump (geo-pump) can be used for this process as long as it has a pressure gage on it and the same pressure limitations are used.

7. Remove the filter funnel from the filter holder being very careful to lift it straight up and not to the side as the sample will be scraped and ruined if this happens. Remove the filter from the holder with clean forceps. Avoid touching the colored portion of the filter. Fold the filter in half, with the colored sample (filtrate) side folded on itself. Place the folded filter onto a small piece of aluminum foil and gently fold it into a square being sure not to ruin the filter or having it fold back open in the foil. Discard the filtered water.

   Complete a AFDM (ALGAE) sample label and attach it to the inside of a plastic zip-top baggie. Place the aluminum foil square into the baggies and place it in a cooler with dry-ice.

6.3 Data Forms and Sample Inspection

After the *Site Assessment Form* is completed, the Field Team Leader reviews all of the data forms and sample forms for accuracy, completeness, and legibility. The other team members inspect all sample containers and package them in preparation for transport and storage.

Ensure that all required data forms for the site have been completed. Confirm that the Monitoring Location ID and date of visit are correct on all forms. On each form, verify that all information has been recorded accurately, the recorded information is legible, and any flags are explained in the comments section. Ensure that written comments are legible. Make sure the header information is completed on all pages of each form. After reviewing each form initial the upper right corner of each page of the form.

Ensure that all samples are labeled, all labels are completely filled in, and each label is covered with clear plastic tape. Compare sample label information with the information recorded on the corresponding field data forms (e.g. the *Sample Collection Form*) to ensure accuracy. Make sure all samples are in a plastic zip-top baggies and are properly sealed.

6.4 Site Cleanup

While the Field Team Leader is reviewing data sheets, samples, and labels for completeness, other members of the team can inspect equipment for damages and cleanliness. If certain equipment is damaged, try fixing it. If it cannot be fixed in the field, prepare back up equipment to be used at the next site. If equipment is dirty, crew members should take the time to clean it. Not only does this help avoid the transfer of aquatic invasive species, but it also helps maintain the integrity of the equipment. Once this has occurred the vehicle can be loaded in preparation of leaving the site area. Keep equipment and supplies organized so they can be inventoried using the equipment and supply checklists presented in Appendix A. Lastly, be sure to clean up all waste and material at the staging area and dispose of or transport it out of the site if a trash can is not available.

7 FIELD QUALITY CONTROL

Standardized training and data forms provide the foundation to help assure that data quality standards for field sampling are met. These Standard Operating Procedures for field sampling and data collection are the primary guidelines for all field teams. It is important to follow these SOPs and not deviate from them even if a crew member has been taught (i.e. methods from another agency) or thinks they know a better method to take any particular measurement. QA/QC measures for UCASE are rather simple. These include performing equipment blanks, collecting replicate macroinvertebrate samples, and site re-visits at a later date in the field season.

7.1 Equipment Blanks

Equipment blanks are done once every UCASE trip (i.e. one per week). Equipment blanks are done on filtered metals and filtered nutrients bottles as well as chlorophyll-*a* processing techniques. These serve as a way to assure crews are keeping bottles and filtering equipment clean as well as following the proper rinsing/processing techniques for filtering as per the DWQ Water Chemistry Sample Collection SOP and the DWQ Chlorophyll-*a* SOP, respectively. Each set of blanks should be given their own lab sheet and submitted to the State Health Lab along with the rest of the water chemistry samples taken that week. The same Monitoring Location ID will be used for all blanks throughout the sampling season and will be assigned at the beginning of the field season by the Field Coordinator.

7.2 Replicate Macroinvertebrate Samples

At times, a request to collect replicate macroinvertebrate samples may occur depending on the season. When applicable, this will happen at a select few sites during the season, which are chosen by the UCASE Program Coordinator. These samples usually do not occur every season. Usually, the Program Coordinator will coordinate with the Field Coordinator at the beginning of the season to choose where to perform replicate macroinvertebrate samples, which will then be relayed to field crews. These samples are done to show if there is any variance with macroinvertebrate sampling from one sample to another. They can also show variance among crew members.

These samples are collected using the same methods as the primary Benthic Macroinvertebrate samples (Section 5.6). The crew member who is performing this sample should do the primary sample first, then go back to Transect A and do the replicate sample second. The field sampler will go to the same transects where the primary samples were taken and perform kicks next to the same location where the primary kick was made. The crew member will be sure to sample in an area as close to where the primary kick was made, but not in an area where disturbances are evident. The crew member will do this 8 times at the same transects the primary samples were collected at. The sample will be processed and labeled the same way as the primary (e.g. STORET/Monitoring Location ID and Site name) sample except the word “Replicate”

will be written on the bottle label. Indicate on the *Sample Collection Form* if a replicate sample was taken and how many bottles.

7.3 Site Re-visits

Site re-visits occur at a select few sites during the season and do not necessarily occur every season. These sites are chosen by the UCASE Program Coordinator and the Field Coordinator at the beginning of the season and relayed to the sampling crews. Crews will go to sites that have already been sampled under UCASE protocols earlier in the same field season, hence re-visit. Usually, only macroinvertebrates (Section 5.6), discharge (Section 5.3), water chemistry (Section 5.1.2, and multi-probe readings (Section 5.1.1) will be sampled at these sites. Crews should establish the X-site at the same location as the first visit. The crew does not necessarily need to layout the entire reach, but need to at least establish Transects A, F (X), and K. While 2 crew members layout the reach, the other crew member will collect water chemistry samples and multi-probe readings upstream of any activity. Once this is done the same crew member can take a flow measurement. Once the X-site has been established, one crew member will perform kick samples in the reach. These do not have to occur in the same locations as the primary visit. Once all of the samples have been collected the transect flags can be taken down and samples processed. The Targeted Benthos Section of the *Sample Collection Form* should be completed as well as a lab sheet for the water chemistry samples. Monitoring Location ID and Site names will be the same as the primary visit. Be sure to indicate on the macroinvertebrate label that this sample is a "Re-visit" sample. This, along with the date on the labels and sheets will separate the primary sample from the re-visit sample. The purpose of this sample is to capture seasonal variability as well as crew variability. The data that is collected from the primary visit will be used for analysis where the re-visit will be used for verification. At times, other parameters may be requested as part of the UCASE protocol on a site by site basis. However, the parameters listed above should always be collected at re-visit sites.

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Appendix A

List of Equipment and Supplies

EQUIPMENT AND SUPPLY LISTS

General Equipment

- Field Manual and/or Quick Reference Guide
- Site packets
- Lab sheets (for sites and blanks)
- Sample labels (dry weight, chlorophyll-*a*, benthos, species ID, fish collection)
- Clipboards
- Field forms and sample labels
- Clear tape strips for covering labels
- Pencils
- Fine-tipped indelible markers
- Digital camera with full battery
- Maps and access instructions
- Sampling permits and/or permission letters
- GPS
- 50 m or 100 m measuring tape with reel
- Surveyors flagging tape or pin flags
- Laser rangefinder
- Batteries
- Sparquat solution in a large tub for gear disinfection
- Calibration standards for multi-probe unit
- Electrical tape
- Scissors
- Plastic storage tub
- Cell phone and/or satellite phone
- Chest waders and/or hip waders
- Field book for fish identification

Sample/Data Collection

- Multi-parameter water quality meter with pH, DO, temperature, and conductivity probes
- Tape measure (metric and standard)
- Nitrile gloves
- Calibrated rod, 1-3 m in length, marked in 0.1 increments
- Convex spherical densiometer (Lemmon Model B), modified with taped “V”
- Clinometer
- Bearing compass (backpacking type)
- Surveyor’s telescoping leveling rod (Stadia rod) ate least 4 m in length
- Meter stick for bank angle measurements
- Flow-tracker flow meter
- Neutrally buoyant object (i.e. plastic golf ball, apple, rubber ball, stick)
- 2.5 gallon plastic bucket

Sample/Data Collection (cont)

- 5 gallon plastic bucket
- 60 mL plastic syringe with a 3/8" hole bored into the end
- Funnel
- 12-cm^3 are delimiter (3.8 cm diameter pipe, 2 cm tall)
- Stiff bristled brush (i.e. toothbrush; paint brush with 90% of bristles cut off)(no metal wire brushes)
- Modified kick net (D-frame, 500 micron mesh, 4 ft. handle
- Sieve bucket, 500 micron mesh (U.S. std No. 35)
- Plastic electrical tape
- Electrofishing equipment (backpack unit including anode and cathode, charged battery, and dip nets)
- Neoprene gloves that are elbow length
- Livewell/5 gallon bucket
- Non-conducting dip nets with ¼" mesh
- Measuring board (millimeter scale)-if collecting fish tissue samples
- Scale (in grams)-if collecting fish tissue samples
- 500 mL plastic bottle for periphyton composite sample
- Small graduated cylinder (up to 250 mL)
- Centrifuge tubes (delineations on tube used to measure small sub samples)
- Liter wash bottle for stream water
- Liter wash bottle for DI water

Sample Processing/Preservation

- Coolers
- Wet ice
- Dry ice
- 95% ethanol (ETOH)
- 10% buffered formalin (for fish preservation of desired)
- Filtration unit including filter funnel, cap, filter holder, filter funnel adapter, rubber stopper, and receiving chamber
- Electric pump (Geo-pump) with extra tubing, canisters, and gaskets
- Disposable forceps
- Millipore 47 mm 0.6 micron glass fiber pre-filters
- Millipore 47 mm 0.45 micron membrane filters
- Aluminum foil
- Large plastic bags
- Knife or scissors
- Plastic cable ties

Bottles/Supplies for Sample Storage

- 1 L sized bottles for benthos sample
- Water chemistry bottles (Big 4-Chemistry, Non-filtered nutrients, Filtered metals, Filtered nutrients, and a transport bottle)
- 50 mL centrifuge tubes with delineations
- Aluminum foil for chlorophyll-*a*, dry weight, and fish tissue samples if applicable
- Plastic baggies for chlorophyll-*a* and dry weight samples
- Large plastic baggies for fish samples if applicable
- Coolers

Appendix B

Field Forms

NOTE: Forms found in this appendix are to serve as a point of reference only! They should not be copied to be used as field forms as certain characters change frequently and these may not be current.

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NOTE: Forms found in this appendix are to serve as a point of reference only! They should not be copied to be used as field forms as certain characters change frequently and these may not be current.

UCASE STREAM VERIFICATION FORM-WADEABLE STREAMS (Front)

Reviewed by (initial):_____

SITE NAME: **DATE:**__ __/__ __/ 2 0 __ __ **TEAM:**_____

SITE/STORET ID:__ __ __ __ __ __ __ __ **UCASE ID (if applicable): UT**__ __**ST-**__ __ __ **VISIT:** 0 1 2 3

STREAM VERIFICATION INFORMATION

Stream Verified by (fill in all that apply): ○ **GPS** ○ **Local Contact** ○ **Signs** ○ **Roads** ○ **Topo Map** ○ **Crew Experience**

○ **Other (Describe here):** ○ **Not Verified (Explain in Comments)**

Coordinates	Latitude North	Longitude South	Datum: (Circle one)
DMS **Map/Defined pre-trip** (fill in only one unit)	__ __ __ __ __ __ . __	__ __ __ __ __ __ . __	NAD 27
Decimal Degrees	__ __ . __ __ __ __ __ __	__ __ __ . __ __ __ __ __ __	NAD 83
UTM	__ __ __ __ __ __ __	__ __ __ __ __ __	
DMS **GPS/Actual at F(X)-SITE** (fill in only one unit)	__ __ __ __ __ __ . __	__ __ __ __ __ __ . __	**Elevation (ft):**
Decimal Degrees	__ __ . __ __ __ __ __ __	__ __ __ . __ __ __ __ __ __	_____
UTM	__ __ __ __ __ __ __	__ __ __ __ __ __	

DID YOU SAMPLE THIS SITE?

○ **YES** If YES, circle-in one below

SAMPLEABLE (Choose method used)

- ○ **Wadeable -** Continuous flowing water; 90%-100% wadeable
- ○ **Partial Wadeable -** Continuous flowing water; 50%-89% wadeable
- ○ **Wadeable Interrupted -** Flowing water not continuous along reach but ≥50% flowing
- ○ **Impounded by Beaver -** Beaver dam present at site but ≥50% of reach still in its channel and wadeable
- ○ **Altered Channel -** Stream present but not as on map

○ **NO** If NO, check one below

NON-SAMPLEABLE (Choose method used)

- ○ **Dry Channel -** Less than 50% site has flowing water
- ○ **Map Error -** No evidence channel/waterbody present
- ○ **Impounded by Beaver -** Too deep or water outside of its channel
- ○ **Impounded Stream** - Site underneath lake or pond (human caused or natural, discluding beaver)
- ○ **Wetland -** No definable channel
- ○ **Other -** Explain in comments

DIRECTIONS TO STREAM SITE:

Updated: 11/2012

NOTE: Forms found in this appendix are to serve as a point of reference only! They should not be copied to be used as field forms as certain characters change frequently and these may not be current.

UCASE STREAM VERIFICATION FORM-WADEABLE STREAMS (Back)

Reviewed by (initial):____

SITE NAME: DATE:___ ___/___ ___/ 2 0 ___ ___ TEAM:_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ UCASE ID (if applicable): UT___ ___ST-___ ___ ___ VISIT: 0 1 2 3

LAND OWNERSHIP INFO/GENERAL COMMENTS ABOUT SITE LOCATION:

STREAM REACH DETERMINATION

Channel Width Used to Define Reach (m)	Total Reach Length Intended (m)	COMMENTS
___ ___ ___	___ ___ ___ ___ ___	

SKETCH MAP - Arrow indicates North

PERSONNEL

NAME	DUTIES Bio/Chem Sampling	Habitat
_____	○	○
_____	○	○
_____	○	○
_____	○	○
_____	○	○

Updated: 11/2012

NOTE: Forms found in this appendix are to serve as a point of reference only! They should not be copied to be used as field forms as certain characters change frequently and these may not be current.

UCASE Reference Site/Site Rating Field Evaluation Form

Reviewed by (initials):_____
Updated: 04/2011

Stream/Site Name:_____

STORET:_____ **Date:**_____ **Crew Members:**_____

UCASE ID:_____ **Lat/UTM:**_____ **Long/UTM:**_____

Best Professional Judgement Grades: After answering all other site evaluations, please grade site on the relative reference quality of the site on a WATERSHED scale and a SITE SPECIFIC scale and provide supporting comments if desired:

WATERSHED SCALE (overall grade of watershed upstream of site; Macro-assessment) circle one:

A) Ideal - wilderness area or virtually no human disturbance upstream of site.
B) Good - minimal disturbances upstream of site. Some activities present upstream in entire watershed but impacts are minimal.
C) Marginal - human disturbances present above site but stream still shows signs of integrity.
D) Sub-marginal - Considerable human disturbances above site in entire watershed; impairment from activities is obvious.
E) Poor - Watershed is heavily manipulated where activities are highly impactful to site; minimal signs of integrity present.
F) Very Poor - Extensive human disturbances in watershed. Integrity of stream is minimal or absent due to activities.

SITE SCALE (grade of site on a reach-wide scale; Micro-assessment) circle one:

A) Ideal - wilderness area and virtually no human disturbance at reach.
B) Good - minimal disturbance at or near site. Some human-based activities present near or at reach but impacts are minimal.
C) Marginal - human activities present at site while sampling. Signs of impacts are present, but site still functions properly.
D) Sub-marginal - Considerable human disturbance near or at site. Activities are having direct impact to site, integrity of site is reduced.
E) Poor - Site is heavily manipulated or impacted from human activities; minimal signs of integrity present.
F) Very Poor - Extensive human disturbances at or near site. Integrity is very minimal or absent.

Comments on grades:

Description of Habitat Features at Site: Accounting for natural factors related to climate, geology, stream size, stream confinement, eco-region, etc., please grade the site 1 through 5 after sampling whether the site has the following features (1=feature indicative of poor habitat….5=feature indicative of ideal habitat/reference conditions). If 1 or 2 is given, comment on why feature does not meet your expectations for this type of stream. Base ratings on ECO-REGION (don't compare a mountain stream to a desert stream). If any categories are undetermined by crew, put a **?** in the grade box. If description does not apply, put an NA in the grade box.

Grades (1-5)	*Description of Habitat Features*	*Comments*
	Riparian vegetation and structure (if non-natives present do not score a 5).	
	Historic riparian vegetation (e.g. legacy trees).	
	Bank conditions (if rip-rap, vegetation removal, car bodies present, etc. do not score a 5)	
	Availability of in-channel habitat (look for a mixture of substrates, undercut banks, overhanging veg, mixture of channel units, etc.)	
	Channel morphology (if channel is been re-routed or manipulated by humans, do not score a 5).	
	Natural flow regime (score accordingly if site is affected by upstream diversions/manipulation structures).	
	Sedimentation and turbidity for stream-type (if stream is naturally turbid, put NA).	
	Influence of human caused sheens; odors; or scum	
	Other:	

NOTE: Forms found in this appendix are to serve as a point of reference only! They should not be copied to be used as field forms as certain characters change frequently and these may not be current.

Potential Stressor Identification Checklist. Indicate the relative effects (i.e., alterations to the chemical, physical, or biological components of the stream ecosystem) of all activities that are present **within the stream channel or riparian corridor** at both the reach or watershed scale (include all tributaries in watershed no matter the size). If YES, describe and indicate intensity as follows:

Reach: **1** = historical activity most likely in vicinity of site but does not appear to have current effects; **2** = activity present but appears to have minimal current effects; **3** = activity present at or near site where impacts are evident but site is not being entirely degraded; **4**= activity present at or near site and has a significant impact to site but some integrity still exists; **5** = activity present on bank or within the stream channel creating obvious impairments to site.

Watershed: **1** = historical activity most likely above site but no signs of currents effects occur; **2** = activity present above the reach in a few limited locales and effects are minimal; **3** = activity is present upstream of the reach where impacts are evident but not creating a major impairment; **4** = activity is present upstream and has a moderate impact to site but some integrity still exists **5** = extensive signs of activity throughout the watershed resulting in major impacts.
Mark a '**?**' in the NO column, if after all feasible field observations have been made you cannot confidently tell whether a stressor is present.

	Reach		*Watershed*		**Comments**
	Yes	**No**	**Yes**	**No**	
Point discharge present?	1 2 3 4 5		1 2 3 4 5		
Hazardous waste sites, landfills?	1 2 3 4 5		1 2 3 4 5		
Mines or oil fields?	1 2 3 4 5		1 2 3 4 5		
Feed lots, poultry farms, or hatcheries?	1 2 3 4 5		1 2 3 4 5		
Urban, industrial, commercial, or residential land use?	1 2 3 4 5		1 2 3 4 5		
Channelization?	1 2 3 4 5		1 2 3 4 5		
Dams or impoundments (not beaver)[1]?	1 2 3 4 5		1 2 3 4 5		
Diversions (i.e., canal head gates)[2]?	1 2 3 4 5		1 2 3 4 5		
Logged or burned forests?	1 2 3 4 5		1 2 3 4 5		
Grazed lands[3]?	1 2 3 4 5		1 2 3 4 5		
Cropped lands?	1 2 3 4 5		1 2 3 4 5		
Non-Motorized Recreation use?	1 2 3 4 5		1 2 3 4 5		
Roads/Motorized Trails?	1 2 3 4 5		1 2 3 4 5		

[1]Score watershed a 2 if there is an impoundment small enough to not largely change natural flow or temperature regimes; [2]Score watershed a 2 if no evidence of daily fluctuations in flow, 3 if water withdrawals are minimal, 4 if water withdrawals are <1/3 flow and large fluctuations are infrequent, 5 if withdrawals >1/3 flow or highly variable; [3]Score reach a 2 if some evidence exists but vegetation looks intact **and** there is no evidence of stream habitat damage (i.e., collapsed banks).

Flow Regime at Site (circle one): Perennial Intermittent Ephemeral

Reviewed by (initials):_____
Updated: 04/2011
UCASE Field Form

NOTE: Forms found in this appendix are to serve as a point of reference only! They should not be copied to be used as field forms as certain characters change frequently and these may not be current.

UCASE QUALITATIVE SITE ASSESSMENT FORM

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ ___ DATE:___ ___/___ ___/ 2 0 ___ ___

WATERSHED ACTIVITIES AND DISTURBANCES OBSERVED (Intensity: Blank=Not observed, L=Low, M=Moderate, H=Heavy)

L M H Residences	L M H Maintained Lawns	L M H Orchards	L M H Evidence of Wildfire	L M H Water Level Fluctuations
L M H Maintained Lawns	L M H Parks, Campgrounds	L M H Poultry	L M H Odors	L M H Fish Stocking
L M H Pipes, Drains	L M H Primitive Camping	L M H Irrigation Equip.	L M H Commercial Development	L M H Dams
L M H Dumping	L M H Trash/Litter	L M H Water Withdrawal	L M H Chemical Treatment	L M H Other:_____
L M H Roads/Motorized Pathways (i.e ATV)	L M H Surface Films	L M H Industrial Plants	L M H Angling Pressure	L M H Other:_____
L M H Hiking Trails	L M H Cropland	L M H Mines/Quarries	L M H Dredging	L M H Other:_____
L M H Bridges/Culverts	L M H Pasture	L M H Oil/Gas Wells	L M H Channelization	
L M H Sewage Treatment	L M H Grazing	L M H Power Plants		
		L M H Logging		

SITE CHARACTERISTICS (200 m radius from site reach)

Waterbody Character	**Pristine** ○5 ○4 ○3 ○2 ○1 **High Disturbed** **Appealing** ○5 ○4 ○3 ○2 ○1 **Unappealing**
Presence of Beaver	**Beaver Signs:** ○**Absent** ○**Rare** ○**Common** **Beaver Flow Modifications:** ○**None** ○**Minor** ○**Major**
Dominant Land Type	**Dominant Land Type at Site:** ○**Forest** ○**Agriculture** ○**Range** ○**Urban** ○**Suburban/Town** ○**Recreation** ○**Other:**_____
Presence of Clams	**Clam Signs:** ○**Absent** ○**Rare** ○**Common** ***Please collect a specimen if clams are abundant and submit to Field Coordinator. If a specimen cannot be collected please take photos and include them with the site photos.**
Weather (general description of weather over the last week)	

GENERAL ASSESSMENT (talk about things that you feel might not have fully been captured on the rest of the forms if there is a unique situation; biotic integrity; vegetation diversity; anecdotal information, etc.)

Updated: 01/2015

NOTE: Forms found in this appendix are to serve as a point of reference only! They should not be copied to be used as field forms as certain characters change frequently and these may not be current.

UCASE QUALITATIVE SITE ASSESSMENT FORM (cont.)

Reviewed by (initial):_____

Updated: 01/2015

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UCASE CHANNEL CONSTRAINT AND FIELD CHEMISTRY COLLECTION FORM-WADEABLE STREAMS

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ **DATE:**___ ___/___ ___/ 2 0 ___ ___

MULTI-PARAMEMTER UNIT MEASUREMENTS

	Comments
STREAM TEMP (°C): ___ ___.___	RESIDUAL CHLORINE (optional):
pH: ___ ___.___ ___	
SPECIFIC CONDUCTIVITY (µS/cm) ___ ___ ___ ___ ___ ___	
STREAM DO mg/L: ___ ___.___	
STREAM DO%: ___ ___ ___.___	
TIME OF DAY: ___ ___ ___ ___	

CHANNEL CONSTRAINT

CHANNEL PATTERN (Check one)

- ○ **One Channel**
- ○ **Anastomosing (complex) channel:** (Relatively long major and minor channels branching and rejoining)
- ○ **Braided channel:** (Multiple short channel branching and rejoining – mainly one channel broken up by numerous mid-channel bars)

CHANNEL CONSTRAINT (Check one)

- ○ **Channel very constrained in V-shaped valley** (i.e. it is very unlikely to spread out over valley or erode a new channel during flood)
- ○ **Channel is in Broad Valley** but channel movement by erosion during flood is **constrained by incision** (Flood flows do not commonly spread over valley floor or into multiple channels)
- ○ **Channel is in Narrow Valley but is not very constrained,** but limited in movement by relatively narrow valley floor
- ○ **Channel is Unconstrained in Broad Valley** (i.e. during flood it can fill off-channel areas and side channels, spread out over flood plain, or easily cut new channels by erosion)

CONSTRAINING FEATURE (Check all that apply)

- ○ **Bedrock/Sandstone** (i.e. channel is constrained by bare bedrock or sandstone)
- ○ **Hillslope** (i.e. channel constrained in narrow V-shaped valley)
- ○ **Terrace** (i.e. channel is constrained by its own incision into stream gravel/soil deposits)
- ○ **Human Bank Alterations** (i.e. constrained by rip-rap, landfill, dike, road, etc.)
- ○ **No constraining features**

PERCENT OF CHANNEL LENGTH WITH MARGIN IN CONTACT WITH EACH CONSTRAINING FEATURE (percentage fields should add up to 100%):	**Constraining Feature 1** (primary)	**Constraining Feature 2** (if applicable)	**Constraining Feature 3** (if applicable)
	Type:_____	**Type:**_____	**Type:**_____
	Percentage: ___ ___ ___ %	**Percentage:** ___ ___ ___ %	**Percentage:** ___ ___ ___ %

Valley floor width (Visual Estimated Average): ___ ___ ___ ___(m)

If you cannot see the valley borders, record the distance you can see and mark this circle ○

Comments:

Updated: 11/2012

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UCASE TORRENT/FLASH FLOOD EVIDENCE FORM

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ ___ DATE:___ ___/___ ___/ 2 0 ___ ___

TORRENT EVIDENCE

Please fill-in the circle if any of the following are evident while sampling.
Visit the following web-sites pre/post sampling to see if any recent major flow regimes occurred:

- **http://www.wrh.noaa.gov/slc/**
- **http://waterdata.usgs.gov/ut/nwis/current/?type=flow**

EVIDENCE OF TORRENT EVENTS/FLASH FLOODS:	
○	Stream channel has a devegetated corridor. This corridor lacks riparian vegetation due to a large flood event.
○	Vegetation along stream margin is depressed.
○	Riparian trees have fresh bark scars at many points along the stream at heights above bankfull level.
○	Riparian vegetation has fallen into the channel as a result of scouring near their roots.
○	Much of stream bed substrate is more loosely consolidated than expected (appears recently deposited).
○	There are locales along the reach where gravel or cobble deposits can be seen above bankfull level from a large flood event.
○	Isolated puddles of water are seen above bankfull level.
○	Deposits of debris (sticks, trash, gravel, vegetation, etc.) can be found above bankfull level.
○	Water appears more turbid than typically expected (considering the eco-region).
○	Overall abundance of benthos sample appears lower than otherwise expected, considering the eco-region, due to recent flood event.
○	Soil above bankfull height is damp.
○	Significant amounts of large woody debris within the stream or riparian corridor appear to have recently been moved.
MAGNITUDE/DESCRIPTION OF EVENT:	
○	Evidence of flooding present, but magnitude is common for this system on a yearly basis (i.e. desert streams after a monsoonal summer storm).
○	Evidence of a large flood present (10-20 year flood), but did not make massive changes to the system.
○	Evidence shows a massive event where signs like channel blow-outs, devegetation, and major changes to the stream are present (i.e. ≥100 year flood).
OTHER:	
○	Other evidence not covered in options above (explain observations in comments section)
NO EVIDENCE:	
○	No evidence of torrent or flash flood events.

COMMENTS
(if evidence of flooding is present, please explain when you think the event occurred, whether it be within the month, longer than a month, within the year, more than two years, etc.)

Updated: 11/2012

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UCASE WATER CHEMISTRY AND MACROINVERTEBRATE SAMPLE COLLECTION FORM-WADEABLE STREAMS

Reviewed by (initial):_____

SITE/STORET ID:___ ___ ___ ___ ___ ___ ___ ___ DATE:___ ___/___ ___/ 2 0 ___ ___

WATER CHEMISTRY

Sample Bottle	Sample Collected?	Sample Bottle	Sample Collected?	Process: See protocol Preservation: Place on ice. Lab: State of Utah Health Lab within one week of collection. Shop storage: Fridge	Comments/Flags
Total Chemistry	Y / N	Filtered Nutrient	Y / N		
Non-Filtered Nutrient	Y / N	Filtered Metals	Y / N		

TARGETED BENTHOS SAMPLE

No. of Jars (Primary)	Was a Replicate Sample Taken (if Yes, record no. of jars?	Collection Method (Choose one)	Comments/Flags
___ ___	Y / N ___ ___	○D-net ○Modified surber ○Other (indicate in comments)	

TRANSECT:		____		____		____		____		Note: - Always perform 8 kicks at every site. - Target riffle habitat primarily. If riffles are scarce or absent (e.g. low gradient, beaver ponds, etc.), target edge habitats (e.g. overhanging veg., undercut banks, etc.) and mark "O" in the substrate column and explain situation in comments. - If riffles are present, but scarce, multiple kicks can be performed at the same riffles throughout the reach. - If kicks are made in beaver influenced areas, please explain in the comments section. **Process:** Fill a clean wide-mouth bottle with ~40% of composite sample (use multiple bottles if necessary). **Preservation:** Fill rest of bottle with denatured alcohol (EtOH) and seal lid with electrical tape. Keep secure in upright position. **Lab:** Utah State University Bug Lab; submit in large batch at end of field season. **Shop storage:** Store on shelf with other bug samples.
Dom. Substrate	**Channel**	**Sub.**	**Chan.**	**Sub.**	**Chan.**	**Sub.**	**Chan.**	**Sub.**	**Chan.**	
Fine/Sand	Pool	○F	○P	○F	○P	○F	○P	○F	○P	
Gravel	Glide	○G	○GL	○G	○GL	○G	○GL	○G	○GL	
Coarse	Riffle	○C	○RI	○C	○RI	○C	○RI	○C	○RI	
Other: Note in Comments	Rapid	○O	○RA	○O	○RA	○O	○RA	○O	○RA	
SUBSTRATE SIZE CLASSES F/S – ladybug or smaller (<2 mm) G – ladybug to tennis ball (2 to 64 mm) C – tennis ball to car sized (64 to 4000 mm) O – bedrock, hardpan, wood, vegetation, leaf litter, undercut, macrophyhtes, etc.		____		____		____		____		
		Sub.	**Chan.**	**Sub.**	**Chan.**	**Sub.**	**Chan.**	**Sub.**	**Chan.**	
		○F	○P	○F	○P	○F	○P	○F	○P	
		○G	○GL	○G	○GL	○G	○GL	○G	○GL	
		○C	○RI	○C	○RI	○C	○RI	○C	○RI	
		○O	○RA	○O	○RA	○O	○RA	○O	○RA	

BENTHOS COMMENTS:

Updated: 11/2012

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UCASE WATER CHEMISTRY AND MACROINVERTEBRATE SAMPLE COLLECTION FORM-WADEABLE STREAMS

Reviewed by (initial):_____

SITE/STORET ID:__ __ __ __ __ __ __ __ DATE:__ __/__ __/ 2 0 __ __

WATER CHEMISTRY

Sample Bottle	Sample Collected?	Sample Bottle	Sample Collected?	Process: See protocol Preservation: Place water chem on ice and chlorophyll-*a* on dry ice Lab: State of Utah Health Lab within one week of collection. Shop storage: Fridge and freezer	Comments/Flags
Total Chemistry	Y / N	Filtered Metals	Y / N		
Non-Filtered Nutrient	Y / N	Chlorophyll-*a* (water column)	Y / N		
Filtered Nutrient	Y / N		Volume filtered: _____mL		

TARGETED BENTHOS SAMPLE

No. of Jars (Primary)	Was a Replicate Sample Taken (if Yes, record no. of jars?	Collection Method (Choose one)	Comments/Flags
__ __	Y / N __ __	○D-net ○Modified surber ○Other (indicate in comments)	

TRANSECT:		____		____		____		____	
Dom. Substrate	Channel	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.
Fine/Sand	Pool	○F	○P	○F	○P	○F	○P	○F	○P
Gravel	Glide	○G	○GL	○G	○GL	○G	○GL	○G	○GL
Coarse	Riffle	○C	○RI	○C	○RI	○C	○RI	○C	○RI
Other: Note in Comments	Rapid	○O	○RA	○O	○RA	○O	○RA	○O	○RA
SUBSTRATE SIZE CLASSES F/S – ladybug or smaller (<2 mm) G – ladybug to tennis ball (2 to 64 mm) C – tennis ball to car sized (64 to 4000 mm) O – bedrock, hardpan, wood, vegetation, leaf litter, undercut, macrophyhtes, etc.		____		____		____		____	
		Sub.	Chan.	Sub.	Chan.	Sub.	Chan.	Sub.	Chan.
		○F	○P	○F	○P	○F	○P	○F	○P
		○G	○GL	○G	○GL	○G	○GL	○G	○GL
		○C	○RI	○C	○RI	○C	○RI	○C	○RI
		○O	○RA	○O	○RA	○O	○RA	○O	○RA

Note:
- Always perform 8 kicks at every site.
- Target riffle habitat primarily. If riffles are scarce or absent (e.g. low gradient, beaver ponds, etc.), target edge habitats (e.g. overhanging veg., undercut banks, etc.) and mark "O" in the substrate column and explain situation in comments.
- If riffles are present, but scarce, multiple kicks can be performed at the same riffles throughout the reach.
- If kicks are made in beaver influenced areas, please explain in the comments section.

Process: Fill a clean wide-mouth bottle with ~40% of composite sample (use multiple bottles if necessary).
Preservation: Fill rest of bottle with denatured alcohol (EtOH) and seal lid with electrical tape. Keep secure in upright position.
Lab: Utah State University Bug Lab; submit in large batch at end of field season.
Shop storage: Store on shelf with other bug samples.

BENTHOS COMMENTS:

Updated: 01/2013

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UCASE PHab: SLOPE AND BEARING FORM - STREAMS

STORET / SITE ID: _____ DATE: ___/___/ 2 0 ___ Reviewed by (Initial): _____

TRANSECT & METHOD Mark method for every Transect	MAIN (always used) Slope(%) or Elev. Diff. (cm) Mark Units for every Transect	BEARING 0 - 359	PROPOR-TION %	FIRST SUPPLEMENTAL Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPOR-TION %	SECOND SUPPLEMENTAL Slope(%) or Elev. Diff. (cm)	BEARING 0 - 359	PROPOR-TION %	FLAG
A < B ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
B < C ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
C < D ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
D < E ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
E < F ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
F < G ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
G < H ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
H < I ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
I < J ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									
J < K ○ CL ○ HL ○ LA ○ TR ○ WT ○ Other	○ % ○ cm									

FLAG COMMENT

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, M (M = Method - used for method comment only) = flag assigned by field crew. Explain all flags in comment sections

Revised: 11/2012 Phab Slope CL=Clinometer; HL=Hand Level; LA=Laser rangefinder with electronic clinometer; TR=Transit, surveyors level or total station; WT=Water Tubing.

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UCASE PHab: CHANNEL/RIPARIAN CROSS-SECTION FORM - STREAMS

STORET / SITE ID: _____ DATE: ____/____/20____ TRANSECT: A B C D E F X-tra Side Channel G H I J K

Reviewed by (Initials): _____

SUBSTRATE CROSS-SECTIONAL INFORMATION

	Dist LB XX.XX m	Depth XXX cm	Size Class Code	Embed. 0-100%	Flag
Left					
LCtr					
Ctr					
RCtr					
Right					

SUBSTRATE SIZE CLASS CODES — Embed. (%)

RS = Bedrock (Smooth) - (Larger than a car) 0
RR = Bedrock (Rough) - (Larger than a car) 0
RC = Concrete/Asphalt
XB = Large Boulder (1000 to 4000 mm) - (Meterstick to car)
SB = Small Boulder (250 to 1000 mm) - (Basketball to meterstick)
CB = Cobble (64 to 250 mm) - (Tennis ball to Basketball)
GC = Coarse Gravel (16 to 64 mm) - (Marble to Tennis ball)
GF = Fine Gravel (2 to 16 mm) - (Ladybug to marble)
SA = Sand (0.06 to 2 mm) - (Gritty - up to Ladybug size) 100
FN = Silt / Clay / Muck - (Not Gritty) 100
HP = Hardpan - (Firm, Consolidated Fine Substrate) 0
WD = Wood - (Any Size)
OT = Other (Write comment below)

FISH COVER/ OTHER

0 = Absent (0%)
1 = Sparse (<10%)
2 = Moderate (10-40%)
3 = Heavy (40-75%)
4 = Very Heavy (>75%)
(circle one)

	Cover in Channel					Flag
Filamentous Algae	0	1	2	3	4	
Macrophytes	0	1	2	3	4	
Woody Debris >0.3 m (BIG)	0	1	2	3	4	
Brush/Woody Debris <0.3 m (SMALL)	0	1	2	3	4	
Live Trees or Roots	0	1	2	3	4	
Overhanging Veg. =<1 m of Surface	0	1	2	3	4	
Undercut Banks	0	1	2	3	4	
Boulders	0	1	2	3	4	
Artificial Structures	0	1	2	3	4	

VISUAL RIPARIAN ESTIMATES

0 = Absent (0%)
1 = Sparse (<10%)
2 = Moderate (10-40%)
3 = Heavy (40-75%)
4 = Very Heavy (>75%)

D = Deciduous
C = Coniferous
E = Broadleaf Evergreen
M = Mixed
N = None

RIPARIAN VEGETATION COVER	Left Bank					Right Bank					Flag
Canopy (>5 m high)											
Vegetation Type	D	C	E	M	N	D	C	E	M	N	
BIG Trees (Trunk >0.3 m DBH)	0	1	2	3	4	0	1	2	3	4	
SMALL Trees (Trunk <0.3 m DBH)	0	1	2	3	4	0	1	2	3	4	
Understory (0.5 to 5 m high)											
Vegetation Type	D	C	E	M	N	D	C	E	M	N	
Woody Shrubs & Saplings	0	1	2	3	4	0	1	2	3	4	
Non-Woody Herbs, Grasses, & Forbs	0	1	2	3	4	0	1	2	3	4	
Ground Cover (<0.5 m high)											
Woody Shrubs & Saplings	0	1	2	3	4	0	1	2	3	4	
Non-Woody Herbs, Grasses and Forbs	0	1	2	3	4	0	1	2	3	4	
Barren, Bare Dirt or Duff	0	1	2	3	4	0	1	2	3	4	

HUMAN INFLUENCE	0 = Not Present P = >10 m C = Within 10 m B = On Bank										
	Left Bank					Right Bank					Flag
Wall/Dike/Revetment /Riprap/Dam	0	P	C	B		0	P	C	B		
Buildings	0	P	C	B		0	P	C	B		
Pavement/Cleared Lot	0	P	C	B		0	P	C	B		
Road/Railroad	0	P	C	B		0	P	C	B		
Pipes (Inlet/Outlet)	0	P	C	B		0	P	C	B		
Landfill/Trash	0	P	C	B		0	P	C	B		
Park/Lawn	0	P	C	B		0	P	C	B		
Row Crops	0	P	C	B		0	P	C	B		
Pasture/Range/Hay Field	0	P	C	B		0	P	C	B		
Logging Operations	0	P	C	B		0	P	C	B		
Mining Activity	0	P	C	B		0	P	C	B		

BANK MEASUREMENTS

	Bank Angle 0 - 360	Undercut Dist. (m)	Flag
Left			
Right			

		Flag
Wetted Width XXX.X m		
Bar Width XX.X m		
Bankfull Width XXX.X m		
Bankfull Height XX.X m		
Incised Height XX.X m		

CANOPY COVER MEASUREMENTS

DENSIOMETER (0-17Max)

		Flag		Flag
CenUp			CenR	
CenL			Left	
CenDwn			Right	

Flag codes: K = Sample not collected; U = Suspect sample; F1, F2, etc. = misc. flag assigned by field crew. Explain all flags in comment sections.

Flag	Comments

Revised: 11/2012 PHAB Chan/Riparian - Str

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Reviewed by (initial): _____
Updated: 03/2011

DWQ Electro-shocking/Fish Tissue Collection Field Sheet

Please use a new sheet for each site (do not combine multiple sites on one sheet)

Site Name:					
STORET # (not applicable for DWR personnel):					
County:					
Date:					
GPS Coordinates		Degrees	Minutes	Seconds	Other (decimal degrees/UTM)
Datum:	Latitude				
	Longitude				
Electro-shocking Reach Length (m):					
Shocker Settings					
Shocking Time (s):					
Volts:					
Pulse Rate (Hz):					
Pulse Width (ms):					

1. Tally final counts for each species and their appropriate sizes in the circles. 2. Species codes on backside of this sheet	ELECTRO-SHOCKING DATA				
	Size 1: 0-60mm (0-2.36 in)	**Size 2: 61-200mm** (2.40-7.87 in)	**Size 3: 201-300mm** (7.91-11.81 in)	**Size 4: 301-400mm** (11.85-15.74 in)	**Size 5: >401mm** (>15.78 in)
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯
Species code:	◯	◯	◯	◯	◯

Comments:	Number of Netters:

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Reviewed by (initial): _____
Updated: 03/2011

Fish Tissue Data (Hg)

Sample ID*	Length (mm)	Weight (g)	Comments
01			
02			
03			
04			
05			
06			
07			
08			
09			
10			
11			
12			
13			
14			
15			
16			
17			
18			
19			
20			
21			If there are more than 21 samples, use another sheet and staple all the sheets together.

*Sample ID=STORET-Fish Code (listed below)-Unique Sequence ID per Site (i.e. 4982100CTT01)

Collector(s) Names (for Hg collection only)**:**

_____ _____ _____

Species Codes: _____ _____

Black bullhead (BBH)	Least chub (LEC)	Utah sucker (UTS)
Black crappie (BLC)	Leatherside chub (LSC)	Virgin spinedace (VSD)
Bluehead sucker (BHS)	Longnose dace (LND)	Walleye (WLE)
Bluegill (BLG)	Mountain whitefish (MWF)	Wiper (WIP)
Bonneville whitefish (BWF)	Mountain sucker (MTS)	White bass (WHB)
Bonneville cisco (BCI)	Mottled sculpin (MOS)	Woundfin (WFN)
Brook trout (BKT)	Paiute sculpin (PTS)	Yellow perch (YLP)
Brown trout (BRT)	Rainbow trout (RBT)	
Channel catfish (CCF)	Redside shiner (RSS)	
Common carp (CMC)	Red-shiner (RES)	Note: Not all spp found in Utah are listed here. If crew collects a spp that is not listed then hand write the spp name on the front side of the sheet under "Species code"
Cutthroat trout (CTT)	Roundtail chub (RTC)	
Desert sucker (DSS)	Smallmouth bass (SMB)	
Flannelmouth sucker (FLS)	Speckled dace (SPD)	
Fathead Minnow (FHM)	Splake trout (SPT)	
Green sunfish (GSF)	Striped bass (STB)	
Kokanee (KOK)	Tiger muskie (TGM)	
Lake trout (LKT)	Tiger trout (TGT)	
Largemouth bass (LMB)	Utah chub (UTC)	

Appendix C

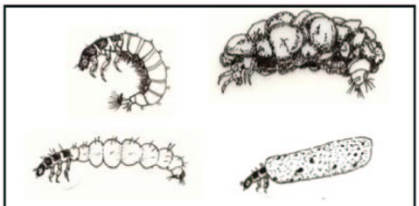
Macroinvertebrate/Benthos Guides

IOWATER

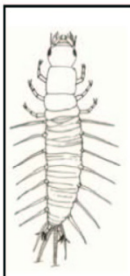
IOWATER, 502 E 9th St, Des Moines, IA 50319; www.iowater.net

IOWATER BENTHIC MACROINVERTEBRATE KEY

Pollution Intolerant (High Quality Group)

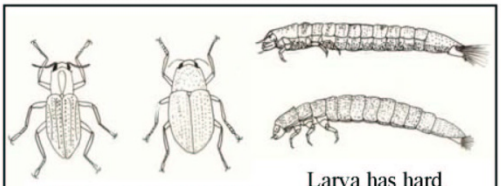


Caddisfly: 6 hooked legs on upper body, 2 hooks on end, may have stick, rock or leaf case, 2-40 mm in length.



Dobsonfly: 6 legs, 8 pairs of feelers and gill tufts on lower half of body, short antennae, 25-90 mm in length.

Mayfly: 6 legs, feathery or oval-shaped gills on lower body, 2 to 3 long tails, 3-30 mm in length.

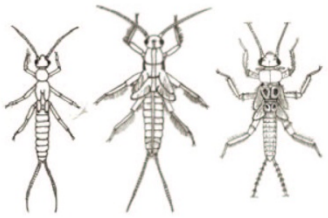


Riffle Beetle: Adult has 6 legs, body covered with tiny hairs, walks slowly underwater, 1-8 mm in length.

Larva has hard plates on each segment, 2-60 mm in length.

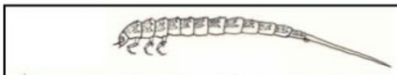
Snail (not pouch): When opening is facing you, shell opens on right, operculum (flap over opening) present.

Water Penny Beetle: Flat saucer-shaped body, 6 tiny legs and gills on underside, 4-6 mm.



Stonefly: 6 legs with hooked tips, antennae, 2 tails, gill tuffs under legs or no visible gills, 5-60 mm in length.

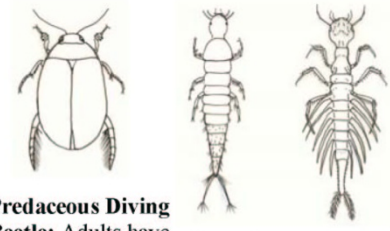
Somewhat Pollution Tolerant (Middle Quality Group)



Crawling Water Beetle: Larva has one long tail and legs with one hook-like claw, 2-10 mm in length. Adult is often patterned or spotted, 2-6 mm.

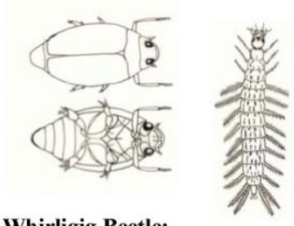
Backswimmer: Forelegs not as spoon-shaped as Water Boatman's, swims upside-down, body is V-shaped, 5-17 mm in length.

Water Boatman: Forelegs spoon-shaped and shorter compared to Backswimmer, 3-11 mm in length.



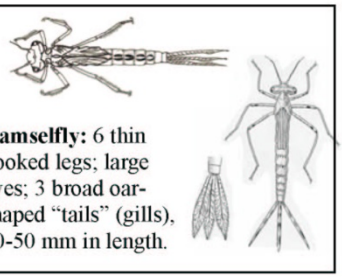
Predaceous Diving Beetle: Adults have an oval streamlined body, longer antennae than Whirligig Beetle, 1-80 mm in length.

Larva has many hairs on body, two feathery tails, large head, 5-70 mm in length.

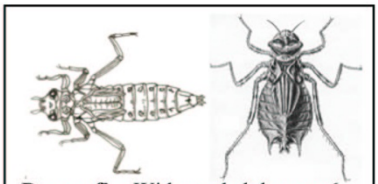


Whirligig Beetle: Flattened oval body, short, clubbed antennae, erratic swimmer, 3-15 mm.

Larva has many hairs on body, short tail or no tail, up to 30 mm in length.



Damselfly: 6 thin hooked legs; large eyes; 3 broad oar-shaped "tails" (gills), 10-50 mm in length.



Dragonfly: Wide oval abdomen, 6 hooked legs, large eyes, 10-60 mm in length.

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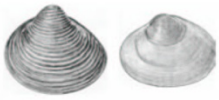
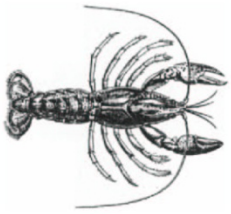
Somewhat Pollution Tolerant (Middle Quality Group) continued

Crane Fly: Milky, green, or light brown color, caterpillar-like segmented body, 4 finger-like lobes at back end, no visible head, 10-100 mm.



Mussels/Clams: Fleshy body enclosed between 2 clamped shells (bivalve), 2-250 mm.



Crawdad: 2 large claws, 8 legs, up to 6 inches long.

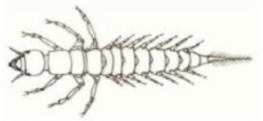
Scud: White to gray in color, more than 6 legs, swims sideways, body higher than wide, 5-20 mm in length.

Sowbug: Gray body wider than it is high, more than 6 legs, 5-20 mm.

Water Scorpion: Raptor-like forelegs for catching prey, long breathing tube, stick-like long body, 15-45 mm.

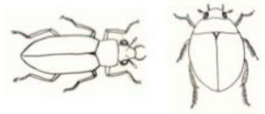
Giant Water Bug: Raptor-like forelegs for catching prey, leathery textured, oval body, 15-65 mm in length.

Water Strider: Slender body, long legs "walk" on water surface, 3-21 mm.

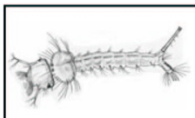


Alderfly: Looks like a small Dobsonfly but has one long tail and no gill tufts, 10-25 mm.

Pollution Tolerant (Low Quality Group)



Water Scavenger Beetle: Adult body may or may not be streamlined, most have no swimming hairs on legs, short clubbed antennae, 1-40 mm in length. Larva have short antenna, 8 soft body segments, 4-60 mm.



Mosquito: Head has small mouth brushes and short antennae; abdomen has breathing siphon, surfaces for air, 4-12 mm.

Pouch Snail: When opening is facing you, shell opens on the left, no operculum (flap over opening).

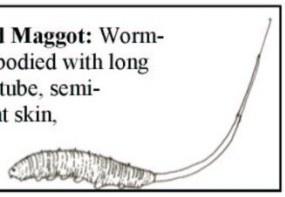
Flatworm (Planarian)**:** Flat, soft-bodied worm with arrowhead-shaped head, 1-30 mm in length.

Black Fly: One end of body wider, black head and suction pad on other end, 3-12 mm.

Midge Fly: Small, dark head, 2 tiny legs on each end, 2-20 mm.

Bloodworm: One type of midge fly, has a red body due to hemoglobin.

Leech: Brown, slimy body, suction pads on body, 5-400 mm.



Rat-tailed Maggot: Worm-like, soft-bodied with long breathing tube, semi-transparent skin, 4-70 mm.

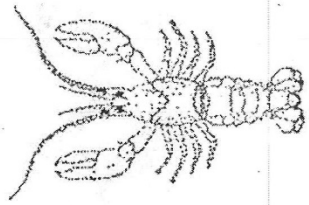
Aquatic Worm: Thin, worm-like body, 0.5-700 mm.

Revised: March 2004

Somewhat Sensitive

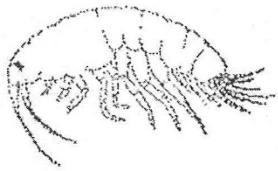
Macroinvertebrates That Are Somewhat Sensitive to Pollution

Found in Good or Fair Quality Water

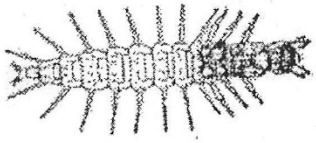


Crayfish

Alderfly



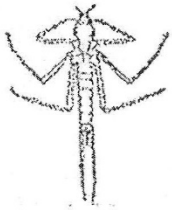
Scud



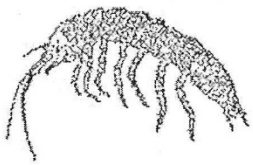
Fishfly

Crane Fly

Riffle Beetle Larva

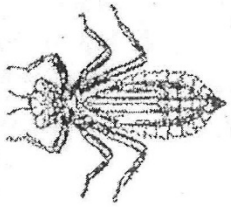


Damselfly

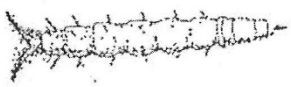


Sowbug

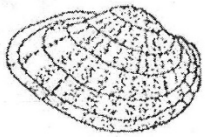
Whirligig Beetle Larva



Dragonfly



Watersnipe Fly

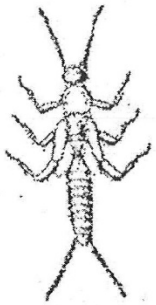


Clam or Mussel

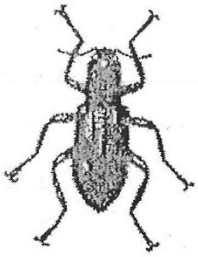
Very Sensitive

Macroinvertebrates That Are Sensitive to Pollution

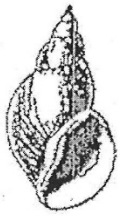
Found in Good Quality Water



Stonefly

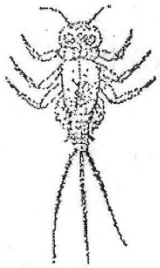


Riffle Beetle Adult

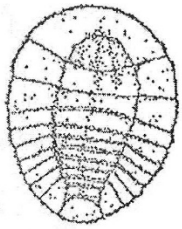


Gilled Snail

Planarian



Mayfly



Water Penny

Caddisfly

Hellgramite

Not Sensitive

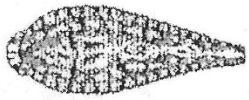
Macroinvertebrates That Are Tolerant of Pollution

Found in Any Quality Water

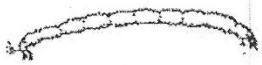
Aquatic Worm

Lunged Snail

Black Fly



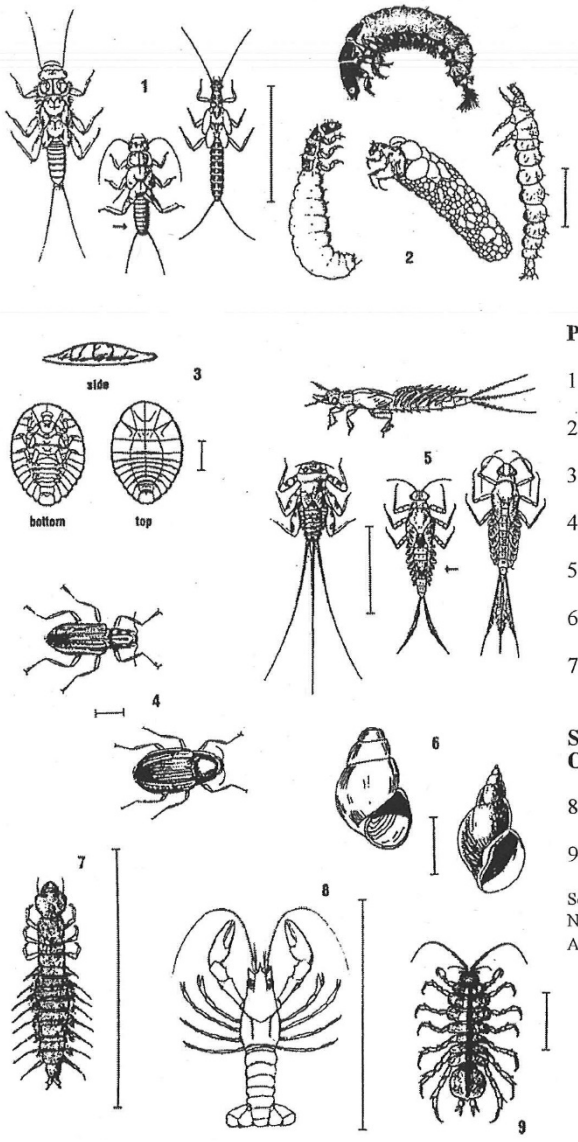
Leech



Midge Fly

From: The Stream Study based on material developed by the Save our Streams Program of the Izaak Walton League of America

Stream Invertebrate Species



Pollution Intolerant Organisms

1. Stonefly, Order Plecoptera
2. Cadisfly, Order Trichoptera
3. Water Penny, Order Coleoptera
4. Riffle Beetle, Order Coleoptera
5. Mayfly, Order Ephemoreptera
6. Gilled Snail, Order Prosobranchia
7. Dobsonfly (Hellgrammite) Order Megaloptera

Somewhat Pollution Tolerant Organisms

8. Crayfish, Order Decapoda
9. Sowbug, Order Isopoda

Source: Izaak Walton League of America published in NWCC Technical Note 99-1 Stream Visual Assessment Protocol, Dec 1998

Bar line indicates relative size

In this riparian assessment for those organisms that are listed as "Class," count those as one "order."

Somewhat Pollution Tolerant Organisms

10. Scud, Order Amphipoda
11. Alderfly Larva, Order Megaloptera
12. Fishfly Larva, Order Megaloptera
13. Damselfly, Order Odonata
14. Watersnipe Fly Larva, Order Megaloptera
15. Crane Fly, Order Diptera
16. Beetle Larva, Order Coleoptera
17. Dragon Fly, Order Odonata
18. Clam/Mussel, Class Bivalvia

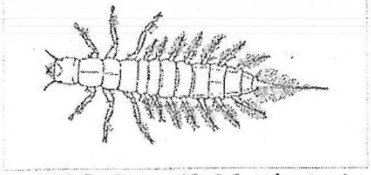
Pollution Tolernt Organisms

19. Aquatic Worm, Class Oligochaeta
20. Midge Fly Larva, Order Diptera
21. Blackfly Larva, Order Diptera
22. Leech, Order Hirudinea
23. Pouch Snail and Pond Snails, Order Pulmonata
24. Other Snails, Order Pulmonata

Bar line length indicates relative size

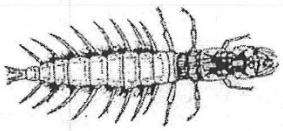
AQUATIC MACROINVERTEBRATE
Identification Guide

LOW POLLUTION TOLERANCE (PTI = 3)

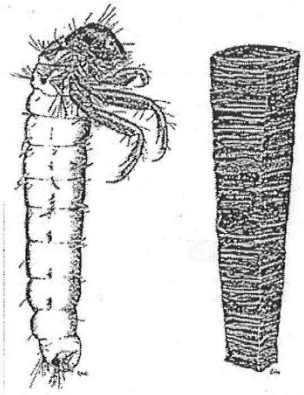


Fishfly Larva (O. Megaloptera)

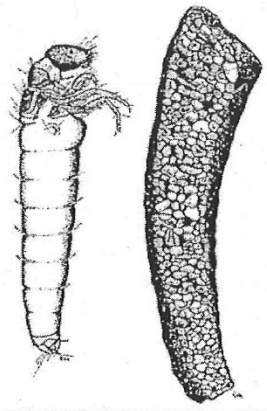
Snipefly Larva (O. Diptera)



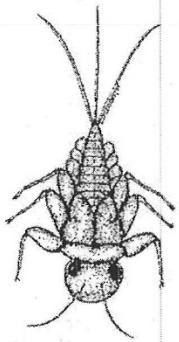
Dobsonfly Larva (O. Megaloptera)



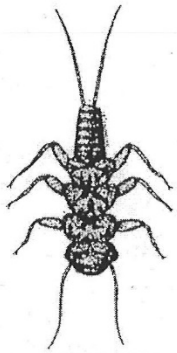
Caddisfly Larva (O. Trichoptera)
plant-matter case



Caddisfly Larva
gravel case

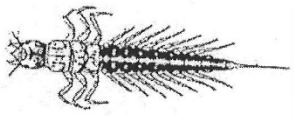


Mayfly Nymph
(O. Emphemeroptera)

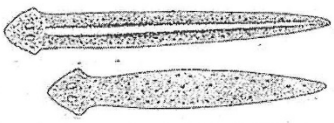


Stonefly Nymph
(O. Plecoptera)

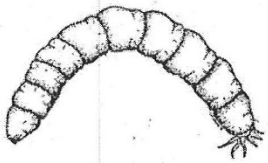
MEDIUM POLLUTION TOLERANCE (PTI = 2)



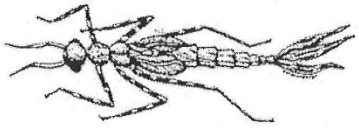
Alderfly Larva (F. Sialidae)



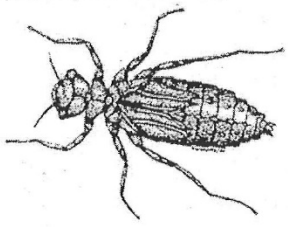
Flatworms (C. Turbellaria)



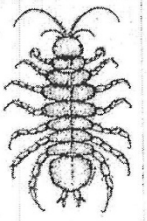
Cranefly Larva (F. Tipulidae)



Damselfly Nymph (O. Odonatia)



Dragonfly Nymph (O. Odonatia)



Aquatic Sowbug (O. Isopoda)

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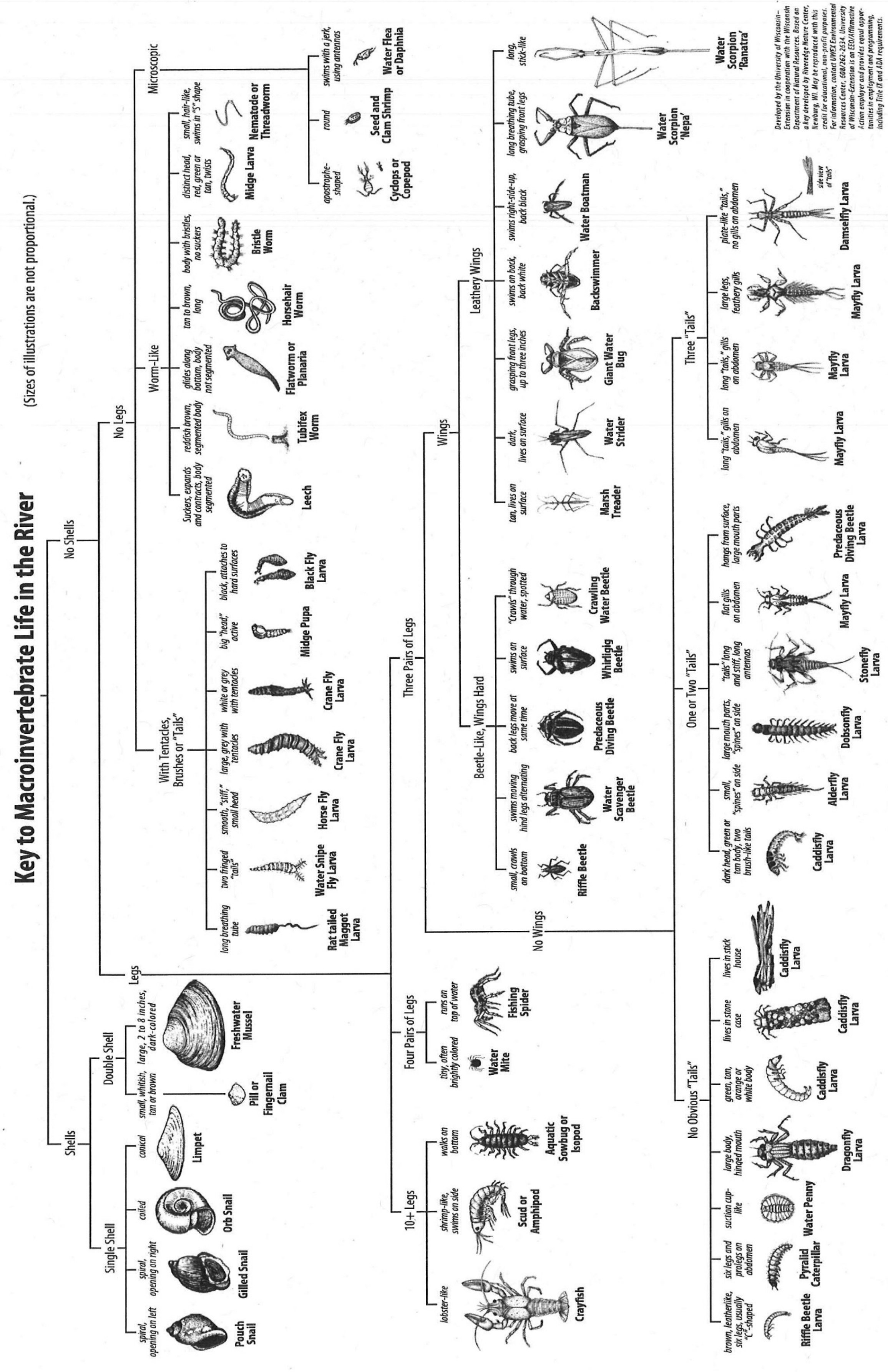
Key to Macroinvertebrate Life in the River
(Sizes of illustrations are not proportional.)
Shells
No Shells
Single Shell
Double Shell
spiral, opening on left
Pouch Snail
spiral, opening on right
Gilled Snail
coiled
Orb Snail
conical
Limpet
small, whitish, tan or brown
Pill or Fingernail Clam
large, 2 to 8 inches, dark-colored
Freshwater Mussel
Legs
No Legs
With Tentacles, Brushes or "Tails"
long breathing tube
Rat tailed Maggot Larva
two fringed "tails"
Water Snipe Fly Larva
smooth, "stiff," small head
Horse Fly Larva
large, grey with tentacles
Crane Fly Larva
white or grey with tentacles
Crane Fly Larva
big "head," active
Midge Pupa
black, attaches to hard surfaces
Black Fly Larva
Worm-Like
Suckers, expands and contracts, body segmented
Leech
reddish brown, segmented body
Tubifex Worm
glides along bottom, body not segmented
Flatworm or Planaria
tan to brown, long
Horsehair Worm
body with bristles, no suckers
Bristle Worm
distinct head, red, green or tan, twists
Midge Larva
small, hair-like, swims in "S" shape
Nematode or Threadworm
Microscopic
apostrophe-shaped
Cyclops or Copepod
round
Seed and Clam Shrimp
swims with a jerk, using antennas
Water Flea or Daphnia
10+ Legs
lobster-like
Crayfish
shrimp-like, swims on side
Scud or Amphipod
walks on bottom
Aquatic Sowbug or Isopod
Four Pairs of Legs
tiny, often brightly colored
Water Mite
runs on top of water
Fishing Spider
Three Pairs of Legs
Wings
No Wings
Beetle-Like, Wings Hard
small, crawls on bottom
Riffle Beetle
swims moving hind legs alternating
Water Scavenger Beetle
back legs move at same time
Predaceous Diving Beetle
swims on surface
Whirligig Beetle
"Crawls" through water, spotted
Crawling Water Beetle
Leathery Wings
tan, lives on surface
Marsh Treader
dark, lives on surface
Water Strider
grasping front legs, up to three inches
Giant Water Bug
swims on back, back white
Backswimmer
swims right-side-up, back black
Water Boatman
long breathing tube, grasping front legs
Water Scorpion 'Nepa'
long, stick-like
Water Scorpion 'Ranatra'
No Obvious "Tails"
brown, leatherlike, six legs, usually "C"-shaped
Riffle Beetle Larva
six legs and prolegs on abdomen
Pyralid Caterpillar
suction cup-like
Water Penny
large body, hinged mouth
Dragonfly Larva
green, tan, orange or white body
Caddisfly Larva
lives in stone case
Caddisfly Larva
lives in stick house
Caddisfly Larva
One or Two "Tails"
dark head, green or tan body, two brush-like tails
Caddisfly Larva
small, "spines" on side
Alderfly Larva
large mouth parts, "spines" on side
Dobsonfly Larva
"tails" long and stiff, long antennas
Stonefly Larva
flat gills on abdomen
Mayfly Larva
hangs from surface, large mouth parts
Predaceous Diving Beetle Larva
Three "Tails"
long "tails," gills on abdomen
Mayfly Larva
long "tails," gills on abdomen
Mayfly Larva
large legs, feathery gills
Mayfly Larva
plate-like "tails," no gills on abdomen
Damselfly Larva
side view of "tails"
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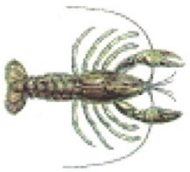
Stream Insects and Crustaceans ID Card

Lines under picture indicate the relative size of organisms

Aquatic Worm:
Class Oligocheata
¼" – 2", can be very tiny; thin, wormlike body, tolerant of impairment

Flat Worm:
Family Planaridae
Up to ¼", soft body, may have distinct head with eyespots, tolerant of impairment

Leech:
Order Hirudinea
¼" – 2", segmented body, suction cups on both ends, tolerant of impairment

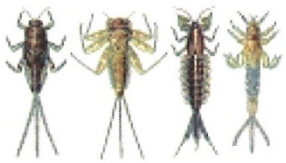


Crayfish: Order Decapoda
Up to 6", 2 large claws, 8 legs, resembles a small lobster, somewhat tolerant of impairment

Sowbug: Order Isopoda
¼" – ¾", gray oblong body wider than it is high, more than 6 legs, long antennae, somewhat tolerant of impairment

Scud: Order Amphipoda
¼", white to gray, body higher than it is wide, swims sideways, more than 6 legs, resembles small shrimp, somewhat tolerant of impairment

Stonefly: Order Plecoptera
½" – 1 ½", 6 legs with hooked tips, antennae, 2 hair-like tails, no gills on abdomen, very intolerant of impairment



Mayfly:
Order Emphemeroptera
¼" – 1", plate-like or feathery gills on abdomen, 6 hooked legs, 2 or 3 long hair-like tails, tails may be webbed together, very intolerant of impairment

Dragonfly and Damselfly:
Order Odonata
½ " – 2", large eyes, 6 hooked legs, large protracting lower jaw, 3 broad oar-shaped tails OR wide oval to round abdomen, somewhat tolerant of impairment

Hellgrammite, Fishfly, and Alderfly:
Order Megaloptera
¾" – 4", 6 legs, large pinching jaws, 8 pairs of feelers along abdomen, 2 hooks on tail end OR 1 single spiky tail, somewhat tolerant of impairment

Common Netspinners:
Family Hydropsychidae
Up to ¾", 6 hooked legs on upper 1/3 of body, 2 hooks at back end, underside of abdomen with white tufts of gills, somewhat tolerant of impairment

Most Caddisfly:
Order Trichoptera
Up to 1", 6 hooked legs on upper 1/3 of body, may be in stick, rock or leaf case, no gill tufts on abdomen, intolerant of impairment

Illustrations from: Voshell, J. R., Jr. 2001. Guide to the Common Freshwater Invertebrates of North America. MacDonald and Woodward Publishing Co. With permission of the author.

Group 1: These are sensitive to pollutants. Circle each animal found.


Stonefly Larva


Dobsonfly Larva

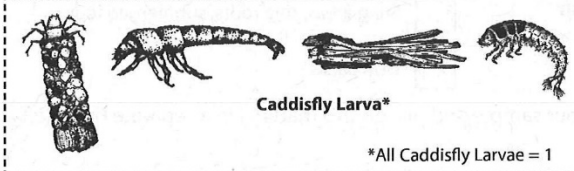

Alderfly Larva


Water Snipe Fly Larva

No. of group 1 animals circled: ☐

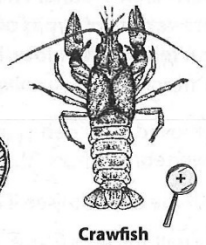
Relative Size Key:
(+) = larger than picture
(-) = smaller than picture

Group 2: These are semi-sensitive to pollutants. Circle each animal found.


Caddisfly Larva*
*All Caddisfly Larvae = 1

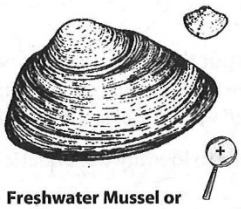

Dragonfly Larva


Water Penny

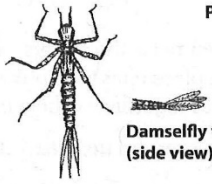

Crawfish

No. of group 2 animals circled: ☐


Crane Fly Larva


Freshwater Mussel or Fingernail clam


Mayfly Larva


Damselfly tail (side view)
Damselfly Larva


Riffle Beetle Larva*
Riffle Beetle Adult*
*All Riffle Beetles = 1

Group 3: These are semi-tolerant of pollutants. Circle each animal found.


Black Fly Larva


Non-Red Midge Larva


Snails: Orb or Gilled (right side opening)
*All Snails = 1


Amphipod or Scud

No. of group 3 animals circled: ☐

Group 4: These are tolerant of pollutants. Circle each animal found.


Pouch Snail (left side opening)


Isopod or Aquatic Sowbug


Bloodworm Midge Larva (red)


Leech


Tubifex Worm

No. of group 4 animals circled: ☐

For more information, call (608) 265-3887 or (608) 264-8948.
Download and print data sheets from
watermonitoring.uwex.edu/wav/monitoring/sheets.html

Water Action Volunteers

Appendix D

Model Numbers and Ordering Information for Select Items/Gear

Macroinvertebrate Sampling			
Product Description	**Place of Purchase**	**Item/Model Number**	**Comments**
D-Frame Dip Net 500 micron mesh with 52" handle (set)	www.wildco.com	425-D52	
D-Frame Dip Net Replacement Clips	www.wildco.com	425-L12	
Sieve Bucket 500 micron mesh	www.wildco.com	190-E25	
2.5 Gallon Plastic Bucket	DNR Warehouse or any standard supply store	NA	
D-Frame Dip Net 500 micron mesh (net only)	www.wildco.com	425-D50	
Denatured Alcohol 190 5 Gallon Tech	www.hvchemical.com	513700-5	Natured alcohol can also be purchased from USU
1000 mL Nalgene Wide-Mouth Bottles	www.amazon.com	332189	These items can be purchased from any vendor that supplies plastic-ware. Different size bottles can be used as well depending on crew preference

Periphyton Sampling			
Product Description	**Place of Purchase**	**Item/Model Number**	**Comments**
Corning Polypropylene Centrifuge Tubes	www.fishersci.com	430921	
500 mL Nalgene Wide-Mouth Bottles	www.amazon.com	332819	These items can be purchased from any vendor that supplies plastic-ware.

Physical Habitat Sampling			
Calibrated Rod 1-3 m			This item is usually home-made by staff members using things like a ski pole or broom handle.
Spherical Convex Densiometer	www.wildco.com	110-K10	
Surveyors Tripod	www.benmeadows.com	108495	
Clinometer	www.benmeadows.com	102204	
Automatic Level/Transit	CST/Berger	F 034 068 104	Can be ordered from any standard vendor online. Any model or brand can be used based on price and quality.

General/Other Supplies			
Buckets/Totes			These are part of the Monitoring Section's general inventory. See Field Program Coordinator
Field Book for Fish ID			These are part of the Monitoring Section's general inventory. See Field Program Coordinator
Digital Camera			These are part of the Monitoring Section's general inventory. See Field Program Coordinator
GPS			These are part of the Monitoring Section's general inventory. See Field Program Coordinator
Laser Rangefinder			These are part of the Monitoring Section's general inventory. See Field Program Coordinator
Satellite Phone			These are part of the Monitoring Section's general inventory. See Field Program Coordinator
Measuring Tape	www.wildco.com www.benmeadows.com	Depends on size needed	
Flagging Tape/Pin Flags	www.benmeadows.com	Staff preference	
Millipore Membrane Filters (0.45 micron)	www.fishersci.com	HAWG047S6	These are part of the Monitoring Section's general inventory. See Field Program Coordinator

General/Other Supplies (cont.)			
Millipore Glass Fiber Pre-Filters (0.2-0.7 micron)	www.fishersci.com	AP1504700	These are part of the Monitoring Section's general inventory. See Field Program Coordinator

Electrofishing Equipment *Note: All electrofishing equipment should be purchased through Smith-Root to keep parts consistent with manufacturer*			
Electrofisher	www.smith-root.com/ www.halltechaquatic.com	LR-24/HT-2000	
Battery	www.smith-root.com/ www.halltechaquatic.com		
Battery Charger	www.smith-root.com/ www.halltechaquatic.com		
Anode	www.smith-root.com/ www.halltechaquatic.com		
Cathode	www.smith-root.com/ www.halltechaquatic.com		
Fish Nets			These can be ordered through any vendor (online, DNR, wildco, etc.) depending on crew preference

NOTE: Any of these items can be purchased from any vendor beyond what's suggested above. What's listed above is where equipment has usually been purchased historically. If you find a more preferable vendor, or a piece of equipment that works better, inform the Field Program Coordinator and they will update it to this appendix.

Field Notes