# THE 2007 NMED/SWQB STANDARD OPERATING PROCEDURES FOR DATA COLLECTION


Prepared by

Surface Water Quality Bureau
New Mexico Environment Department

**STANDARD OPERATING PROCEDURES**
**FOR**
**Data Collection**
**Surface Water Quality Bureau**
**New Mexico Environment Department**
**July 26, 2007**

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| | |
|---|---|
| APHA | American Public Health Association |
| AU | Assessment unit |
| BLM | Bureau of Land Management |
| BTEX | Benzene, toluene, ethylbenzene, xylene |
| CDC | Centers for Disease Control |
| CFR | Code of Federal Regulations |
| cfs | Cubic feet per second |
| *chl a* | Chlorophyll a |
| $cm^2$ | Square centimeters |
| CWA | Clean Water Act |
| DIW | De-Ionized Water |
| DO | Dissolved Oxygen |
| DOE | U.S. Department of Energy |
| DTH | Depositional-Targeted Habitat |
| DTH | Epipsammic/Epipelic Habitats |
| EMAP | Environmental Monitoring and Assessment Program |
| EWI | Equal Width Increment |
| FISP | Federal Interagency Sedimentation Project |
| fps | Feet per second |
| FSP | Field Sampling Plan |
| ft/s | Feet per second |
| $ft^2$ | Square feet |
| $ft^2/s$ | Square feet per second |
| G | Grams |
| GF/A | Glass Fiber, Grade A |
| GPS | Global Positioning System |
| HCl | Hydrochloric acid |
| Hg | Mercury |
| KCl | Potassium chloride |
| KI | Potassium iodide |
| L | Liter |
| lbs | Pounds |
| LWE | Left wetted edge |
| $m^2$ | Square meters |
| MAS | Monitoring and Assessment Section |
| mg | Milligram |

| | |
|---|---|
| mg/L | Milligrams per liter |
| mL | Milliliter |
| mm | Millimeter |
| MSDS | Material Safety Data Sheets |
| mV | Millivolt |
| NaOH | Sodium hydroxide |
| NAWQA | National Water Quality Assessment |
| NIST | National Institute of Standards and Technology |
| NMAC | New Mexico Administrative Code |
| NMDGF | New Mexico Department of Game and Fish |
| NMED | New Mexico Environment Department |
| NPDES | National Pollutant Discharge Elimination System |
| NTU | Nephelometric Turbidity Units |
| ºC | Degrees Celsius |
| PBW | Pesticide Grade Blank Water |
| PC | Personal computer |
| PCB | Polychlorinated Biphenyls |
| ppb | Parts per billion |
| ppt | Parts per thousand |
| PSRS | Point Source Regulation Section |
| QA | Quality Assurance |
| QAP | Quality Assurance Program |
| QAPP | Quality Assurance Project Plan |
| QC | Quality Control |
| QMH | Qualitative Multi-Habitat |
| RBP | Rapid Bioassessment Protocol |
| QMP | Quality Management Plan |
| RID | Request Identification |
| RTH | Richest-Targeted Habitat |
| RWE | Right wetted edge |
| SC | Specific conductance |
| SLD | Scientific Laboratory Division |
| SOC | Soil organic carbon |
| SOP | Standard Operating Procedure |
| STORET | Storage and Retrieval database |
| SWQB | Surface Water Quality Bureau |
| TDS | Total dissolved solids |
| TKN | Total Kjeldahl Nitrogen |
| TL | Total length |

| | |
|---|---|
| TMDL | Total Maximum Daily Load |
| TN | Total nitrogen |
| TOC | Total organic carbon |
| TP | Total phosphorus |
| TSI | Trophic State Index |
| TSS | Total suspended solids |
| USACE | United States Army Corp of Engineers |
| USBOR | U.S. Bureau of Reclamation |
| USDA | U.S. Department of Agriculture |
| USDI | U.S. Department of Interior |
| USEPA | U.S. Environmental Protection Agency |
| USFWS | United States Fish and Wildlife Service |
| USGS | U.S. Geological Survey |
| v/v | Volume to volume |
| VBW | Volatile Organic Grade Blank Water |
| VOA | Volatile Organic Analysis |
| VOC | Volatile Organic Compound |
| W | Width |
| WinXSPRO | Windows-Based Stream Channel Cross-Section Analysis |
| WLA | Waste Load Allocation |
| WPS | Watershed Protection Section |
| WQCC | Water Quality Control Commission |
| WQS | Water Quality Standards |
| WWTP | Wastewater treatment plant |
| µg/L | Microgram per liter |
| µm | Micrometer |
| µmhos/cm | Micromhos per centimeter |
| µS/cm | Microsiemens per centimeter |

# 1.0 SCOPE AND APPLICATION

This Standard Operating Procedures (SOP) document serves as guidance to Surface Water Quality Bureau (SWQB) personnel engaged in monitoring of New Mexico surface waters.

Monitoring provides water quality data needed to accomplish the following:

- develop revisions to, or new, water quality standards (WQS);
- develop load allocations (LAs), waste load allocations (WLAs), and total maximum daily loads (TMDLs), where needed,
- develop water quality-based effluent limitations
- assess effectiveness of point and nonpoint source water pollution controls
- prepare biennial reports to the U.S. Congress as required under CWA §305(b).

Methods of data acquisition include:

- fixed-station monitoring,
- compliance monitoring of point source discharges,
- watershed-based surveys, including chemical, physical, and biological assessments.

## 2.0 WATERSHED-BASED SURVEYS

The SWQB performs surface water quality surveys, of streams, rivers, reservoirs, lakes, and playas, as part of a rotational watershed water quality monitoring strategy. The sampling regime for intensive water quality surveys typically extends over a three-season time period. Each survey involves an assessment of waterbodies and their associated habitats, biological communities, and chemical and microbiological water quality. When the bureau rotates back into a particular watershed or study area, additional stations are added as needed to bracket new point or nonpoint sources, to delineate contributions from several sources, or in response to other new information.

For each stream watershed or study area, a Field Sampling Plan (FSP) is prepared by the Project Lead and project team. Any deviations from the plan are documented. The FSP includes a complete list of sample stations along with a brief description of reasons for selection of each. It also includes a list of parameters to be studied at each station, sampling frequency, and addresses Data Quality Objectives (DQOs) for the survey. Currently, FSPs are not prepared for lake surveys.

The SWQB also conducts surveillance activities of waterbodies in response to citizen complaints and in response to spills, fish kills, and other emergencies.

## 3.0 PRE-SURVEY PLANNING

Refer to the QMP/QAPP for information on pre-survey planning.

# 4.0 SELECTING SAMPLING STATIONS

Refer to the QMP/QAPP to select the most appropriate sampling stations.

# 5.0 FIELD DATA MEASUREMENTS

## 5.1 Equipment

The primary field instruments employed by SWQB are the Models 6820, 6920 and 600 XLM multiprobe sondes manufactured by YSI, Inc.

YSI, Inc.
1700/1725 Brannum Lane
Yellow Springs, OH 45387
Phone: (937) 767-7241 or (800) 765-4974
Fax: (937) 767-9320
Email: info@ysi.com
Internet: <http://www.ysi.com>

The specific sonde models numbers are 6820, 6920, and 600XLM, all of which use Ecowatch software, which is a proprietary product of YSI, Inc. The Bureau currently uses Version 3.17.

Sonde models 6920 and 600XLM can be set to collect data automatically while deployed in streams for extended periods of time.

Sondes are typically equipped with:

- YSI 6560 Conductivity/ Temperature probe
- YSI 6561 pH probe
- YSI 6562 DO probe
- YSI 6026 or 6136 Turbidity probe, wiped (Models 6820 and 6920 sonde only)
- YSI 650 MDS data loggers

The thermographs that the Bureau uses are manufactured by:

Onset Computer Corporation
470 MacArthur Blvd.
Bourne, MA 02532
Mailing address: PO Box 3450, Pocasset, MA 02559-3450
Phone: (508) 759-9500 or (800) LOGGERS
Fax: (508) 759-9100
Email: loggerhelp@onsetcomp.com
Internet: <http://www.onsetcomp.com>

The specific models are called the "Optic StowAway® Temp" and the "HOBO® Water Temp Pro," both of which use BoxCar® Pro software. The Bureau currently uses Version 4.3. The Optic StowAway® is an older model and is typically used for air temperature logging, while the newer HOBO® is typically used for water temperature logging.

Scientific instruments used by SWQB are expensive and relatively delicate and must be treated with care to produce high quality data. For specific calibration and maintenance requirements, see Section 5.0 Field Data Measurements.

## 5.2 Field Data Documentation

Field data forms are taken into the field on each sampling trip. Each sampling station has a separate field data form.

### 5.2.1 Field Data Form

*Front:*

- Sampling Data (Date/Time, Staff, Sonde ID, Data Logger ID, Temperature, Specific Conductance, Salinity, D.O. (Dissolved Oxygen-mg/L), D.O. (local percent saturation), D.O. charge, pH, and Turbidity)
- Request identification (RID) number stickers that correspond to each sample container used
- Additional comments

*Back:*

- Designated uses and their attainment status, plus comments on probable sources or causes of non-attainment, which should be filled out whenever a probable source is observed
- Assessment comments and miscellaneous information
- A "nutrient screening observations" section to be filled in once per season. It includes percent algal cover, a rating of the thickness of periphyton on coarse substrate, and an indication if an anoxic sediment layer is present (see Chapter 9.2 Level I Nutrient Survey).
- A "flow condition rating" section to be filled in at each sampling event (this is a qualitative observation, on a scale of 0 – 5, of the flow level, with 0 being no flow and 5 being above bankfull)
- Additional comments

### 5.2.2 Field Notebooks

The use of field notebooks should be avoided. Use the appropriate datasheet or form to ensure information ends up in the administrative record, not on a bookshelf or in a desk drawer.

### 5.2.3 Field Observations

Upon arrival at a sampling site, record visual observations on the appearance of the water and other information related to water quality and water use in the "additional comments" section of the Field Data Form.

*Left bank* is defined as the bank to the left of the observer when facing downstream, and the *right bank* is to the right of the observer when facing downstream. Examples of observations include:

- **Water Appearance -** Color; unusual amount of suspended matter; debris or foam, etc.

- **Weather -** Recent meteorological events that may have impacted water quality such as heavy rains; cold front; very dry; very wet, etc.
- **Biological Activity** - Excessive macrophyte, phytoplankton, or periphyton growth (observation of water color and excessive algal growth may be important in explaining high chlorophyll *a* values); other observations such as presence of beaver, amphibians, fish, spawning fish, and/or dead or dying aquatic or semi-aquatic organisms.
- **Unusual Odors -** e.g. musty odor; sewage odor, hydrogen sulfide ($H_2S$) odor (NOTE: $H_2S$ can ruin the DO membrane. Continued exposure can ruin a DO probe. If $H_2S$ presence is suspected, long term deployment is not recommended. Extra thick membranes are available for grab samples.).
- **Watershed or In-stream Activities -** In-stream or drainage basin activities or events that may impact water quality such as bridge construction; shoreline mowing; livestock watering upstream; etc.
- **Specific Sample Information -** Specific comments about the sample itself that may be useful in interpreting analytical results. There is a space at the bottom of the SLD submittal form for comments that may be pertinent to the lab (e.g. highly turbid sample, etc.). If the sample was collected for compliance or a fish-kill, make a note of this in the observation section.
- **Missing Parameters -** If a scheduled parameter or group of parameters is not collected, note this and the reason for excluding it.

### 5.2.4 Site Condition Class Verification & Probable Source

Biological surveys require a site specific comparison to a reference site, or a group of reference sites that establishes a reference condition. These reference sites have relatively unaltered biology, chemistry, and physical habitat because of minimal human disturbance at both the watershed and stream reach scales. Some regions of New Mexico have widespread, historic anthropogenic impacts to the land which makes defining an unaltered state impossible. In this case, it is necessary to identify and rank the human disturbances found at survey sites in order to identify the least disturbed areas within a watershed or region. At the watershed scale, an analysis is done using GIS to distinguish categories of natural gradients such as elevation, as well as broad human disturbances such as land use and road densities. On the reach scale, sites need to be visited so that disturbances are observed and recorded. The Site Condition Class Verification & Probable Source Field Sheet (Appendix A) lists anthropogenic activities that could potentially alter a stream reach from the reference condition and provides the ability to score the activities based on proximity to the stream. This form should be filled out for all sites that are visited.

The Site Condition Class Verification & Probable Source Field Sheet (Appendix A) is also used to gather field observations on probable source of surface water impairment for inclusion on the Integrated 303(d)/305(b) Report and List of Assessed Waters. USEPA through guidance documents requires states to include a list of probable sources for each listed impairment. According to the 1998 305(b) report guidance, "…, states must always provide aggregate source category totals…" in the biennial submittal that fulfills CWA section 305(b)(1)(C) through (E) (USEPA 1997). "Sources" are defined as activities that may contribute pollutants or stressors to a water body (USEPA 1997). The 2006 Integrated Report guidance states that all states must

submit to EPA a description of the nature and extent of nonpoint source pollution, as well as state-level summaries of causes and sources of impaired waters (USEPA 2005).

The intent of documenting probable sources is to acknowledge any and all activities that could possibly be contributing to impairment of a waterbody. It is not intended to single out any particular land owner or single land management activity, and has therefore been labeled "Probable" on the Integrated List and generally includes several items. Probable sources listed for any particular water body may not be the only source(s) of identified impairment(s). It is generally based on a visual analysis combined with knowledge of known land management activities that have the potential to contribute to the identified impairment. One of the primary reasons we solicit public comment on the Integrated 303(d)/305(b) Report is so entities and individuals living and working in particular watersheds can provide specific information regarding probable sources of impairment that may have not been identified by SWQB staff.

In the past, data on probable sources has generally been gathered by SWQB Monitoring and Assessment Section staff, as well as SWQB Watershed Protection Section staff, during implementation of intensive watershed surveys and watershed restoration projects. This information is housed in the Assessment Database (ADB version 2). This database was developed by USEPA to help states manage information on surface water impairment and to generate integrated 303(d)/305(b) lists and associated statistics. More specific information on probable sources of impairment is provided in individual watershed planning documents (i.e., Total Maximum Daily Loads, Watershed Restoration Action Strategies, etc.) as they are prepared to address individual impairments by assessment unit.

To improve the accuracy of Probable Source lists, SWQB plans to expand on its existing efforts to develop probable source lists by soliciting input on the source list from watershed groups around the state during development of the 2008-2010 Integrated List. SWQB also plans to increase the use of available technologies such as GIS through the analysis of land use coverages.

## 5.3 Instantaneous Field Measurements in Streams and Rivers

Measurements are recorded for each visit on the Field Data Form, and subsequently transferred to the SWQB water quality database.

A multiprobe sonde is typically used to take measurements *in situ* for temperature, specific conductance (SC), dissolved oxygen (DO), pH, and turbidity. When field measurements are made with a sonde, place the sonde in the body of water to be sampled and allow it to equilibrate in "Run" mode while water samples are collected. This is done by enabling the "Wait for DO" feature and setting the "DO warm up" time for 40 seconds (Sonde menu\Advanced\Sensor).

Take field measurements at the centroid of flow, if the stream visually appears to be completely mixed from shore to shore. *Centroid* is defined as the midpoint of that portion of the stream width that contains 50 percent of the total flow.

### 5.3.1 Water Temperature

If a thermometer is to be used (rather than an electronic probe, such as on a sonde), allow the thermometer to stabilize for at least one minute, then without removing the thermometer from the water, read the temperature to the nearest 0.1ºC and record. Do not read temperature with the thermometer out of the water.

### 5.3.2 pH

The pH function does not need to be calibrated each day of use. The sonde should be checked daily against a pH 7.0 buffer and calibrated as necessary. Allow the pH probe to equilibrate before pH is recorded to the nearest 0.1 pH unit.

### 5.3.3 Dissolved Oxygen (DO)

**IMPORTANT:** Calibrate the DO sensor on the multiprobe sonde to the elevation of the first station. Recalibrate no less frequently than every 300 meters (1000 feet) of elevation change.

The DO probe must equilibrate for at least 40 seconds before DO is recorded to the nearest 0.1 mg/L (concentration) and the nearest tenth of a percent (saturation). Because DO takes longer than other parameters to stabilize, record DO after temperature, SC, and pH.

### 5.3.4 Specific Conductance (SC)

The primary physical problem in using a SC meter is entrapment of air in the SC probe chambers. The presence of air in the probe is indicated by unstable SC values fluctuating up to ±100 µS/cm. The entrainment of air can be minimized by slowly and carefully placing the probe into the water; and quickly moving it through the water to release any air bubbles when the probe is completely submerged.

### 5.3.5 In a Bucket

When measurements cannot be conducted in-stream, they may be measured in a container (bucket) used for water sample collection. Use a bucket only when all efforts to obtain an *in situ* measurement have been exhausted. Consider building small dams or depressions to create water deep enough to submerse the sonde. If a bucket must be used, make clear notes on the field sheet indicating exactly what was done.

For measuring field parameters from a bucket:

- Select a bucket that is large enough to allow full immersion of the probes or thermometer.
- Bring the bucket to the same temperature as the water before it is filled.

## 5.4 Additional Field Data Collected Outside of EMAP Procedures

### 5.4.1 Densiometer Field Procedure

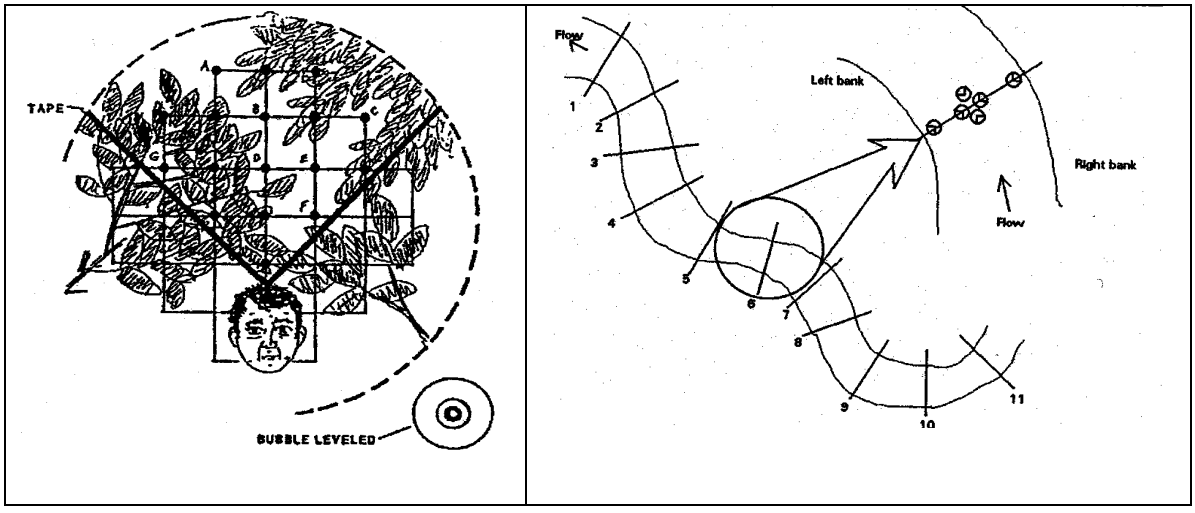
Typical Densiometer Equipment:

- Densiometer

- Measuring Tape
- GPS
- EMAP-based Densiometer Only Field Worksheet

The purpose of this section is to determine a Total Shade value for SSTEMP, a temperature model used in TMDL development, **when a full EMAP habitat survey <u>is not</u> or <u>will not</u> be scheduled** within a particular assessment unit. Ideally, measurements would be taken during seasonal low flow periods to minimize the effects of varying wetted widths. Low flow conditions are usually a time of critical temperature stress to aquatic organisms and stream shade is important. Also, measurements should be taken during a time of year when deciduous plants have leaves.

Use the GPS to find the location of the site's latitude and longitude (lat/long), record the data on the **EMAP–based Densiometer Only Field Worksheet**, and mark that spot. Measure the wetted width of the stream in 3-4 representative locations to find the average wetted width. Multiply the average wetted width by 40 to determine the reach length. The marked lat/long spot is the center of the reach (Transect 6 – refer to figure below). Walk downstream half the length of the reach (measured or paced out); this is the location of Transect 1. Stand at LEW, facing left bank. Hold V-marked densiometer level 0.3 m above the surface of the stream. Move the densiometer in front of you so the top of your head is just below the apex of the "V" (see below). Count and record the number of grid intersection points within the "V" that are covered by a tree, a leaf, or high branch (maximum is 17). Repeat for all six positions in the transect. Divide the reach length by 10 and move upstream this distance (measured or paced out) to locate Transect 2. Record all six densiometer measurements at Transect 2. Continue pacing out or measuring transect distances according to the method described above and record densiometer readings at each transect. Check that your measuring/pacing agree with the marked lat/long spot at Transect 6. To determine % total shade at each transect, sum the six densiometer measurements from the transect. To calculate the average % total shade, sum all transect totals and divide by the number of transects. After walking the length of the reach, circle the dominant substrate of the stream (bedrock, boulders, cobble/gravel, or sand/silt) on the **field worksheet**.



**Schematic of Modified Densiometer and Study Reach.**

In this example, 10 of the 17 intersections show canopy cover, giving a densiometer reading of 10. Note proper positioning with the bubble leveled and the head reflected at the apex of the "V." (Mulvey et al. 1992). Study reach with 11 sample transects and example of 6 densiometer measurements taken at each transect.

## 5.5 Unattended Sampling

### 5.5.1 Thermograph Deployment

One individual within SWQB shall be designated as the "Thermograph Manager" (manager). The manager is responsible for keeping thermographs, associated hardware, software, instruction manuals, and accessories in working order and available for staff to use. In addition, the manager is responsible for thermograph data management. A second individual in the Bureau is designated as the "Alternate Thermograph Manager" (alternate) and fulfills the manager's responsibilities when the manager is unavailable. The alternate performs all duties on the manager's computer in order to avoid confusion and/or misplacement of data.

The manager keeps a record of the disposition of each thermograph. The record includes the thermograph serial number, current status of each thermograph (available for use, launched and awaiting deployment, deployed, or retrieved and awaiting downloading), deployment location, staff member responsible for the thermograph (e.g., survey lead), date of checkout, and any other pertinent information. The manager also keeps a record of the disposition of all thermograph-related accessories, such as optic shuttles and optic couplers.

When staff requires use of one or more thermographs, a request is made to the manager. The manager sets the parameters (e.g., recording interval, triggered or delayed start, etc.) of the thermograph according to the staff member's specifications. This process is known as "launching" the thermograph.

Upon thermograph deployment, field staff fill out a deployment field sheet as completely as possible, including GPS latitude/longitude (in decimal degree format). Copies of the deployment field sheet are provided to the manager. After the thermograph is retrieved, it is given to the manager who downloads the data.

Monitoring for temperature should generally be conducted from May through September for consistency with periods when incident solar radiation angles are high and ambient air temperatures are most likely to be at maximums. Knowledge of regional patterns is important if monitoring duration must be limited to periods shorter than the interval described above. Monitoring should always include the period of maximum expected critical temperatures.

One thermograph should be placed in each Assessment Unit (AU), at locations representative of ambient stream conditions. The thermographs should not be placed in shallow riffles or in deep pools. The thermograph should be deployed in a transition between a riffle/run and a pool, and if possible, it should be placed at the toe of a pool as it becomes shallower, prior to entering a run or riffle. The thermograph should be placed such that, under expected flow conditions, it will be continually submerged. Actual situations encountered during thermograph deployment will vary.

Typical thermograph deployment/retrieval equipment:

- Plastic wire ties (8 inch length, preferably dark color)
- Surveyor's flagging tape
- #4 (1/2 inch diameter) steel rebar stakes of various lengths (18 inches to four feet)
- Steel T-posts (6 and 8 foot) and driver
- Sledgehammer (3 to 4 pounds)
- Tie wire
- Diagonal pliers and lineman's pliers
- Digital camera
- GPS unit
- 30 m measuring tape
- Metal detector (for retrieval)
- Modified mattock (for rebar retrieval)
- Thermograph Deployment/Download/Retrieval Field Sheets

Precautions against vandalism, theft, and accidental disturbance should be considered when deploying equipment. In areas frequented by the public, it is advisable to secure or camouflage equipment.

The most common technique for thermograph deployment is to drive a length of rebar into the stream bed using a small sledgehammer. The thermograph is then secured to the rebar using at least two plastic wire ties. Steel T-posts may be used for soft sediment or deeper streams in the same manner.

Using a GPS unit, record the thermograph lat/long location on the deployment sheet. Take a digital photograph showing the position, typically with a field technician pointing to the instrument's location. Clearly indicate direction of flow and left/right bank. Flag the locale with surveyor's tape to facilitate location upon return. Draw a clear map so that staff not present at deployment will be able to easily locate the thermograph for retrieval.

The SWQB maximum interval for monitoring for standards attainment using a thermograph is one hour. Shorter intervals provide a more precise estimate of the duration of daily maxima, and they may be used without a negative impact to data quality. A one hour interval is considered adequate for typical assessment purposes.

Because thermographs are subject to loss for a variety of reasons beyond staff control (e.g. vandalism, theft, high flows), it is advisable to download data periodically (typically once a month) during the sampling season, using a lap top computer in the field. A file name, consisting of the station name and thermograph serial number, is assigned to each data set upon the initial interim download. Upon return from the field, these data are copied to the manager's computer (and, optionally, to the survey lead's computer). The data may be left in raw form (i.e., not exported to a spreadsheet) at this time if desired. The thermograph is left running during interim download so that a full data set is copied at each download, obviating the necessity to combine files. The interim download date and time are recorded on the deployment field sheet.

### 5.5.2 Sonde Deployment

One individual within SWQB is designated as the "Sonde Data Manager" (manager). A second individual in the Bureau is designated as the "Alternate Sonde Data Manager" (alternate) and fulfills the manager's responsibilities when the manager is unavailable. The alternate performs all duties on the manager's computer in order to avoid confusion and/or misplacement of data.

Upon sonde deployment, field staff fill out a deployment field sheet as completely as possible, including GPS latitude/longitude (in decimal degree format). Copies of the deployment field sheet are provided to the manager. After sondes are retrieved, they are given to the manager who downloads the data.

Upon retrieval from deployment or after elongated storage periods, sondes should be cleaned and calibrated, particularly the DO probe. After changing the membrane and calibrating DO, the sonde should be left to run in "unattended" mode for at least 10 minutes, and DO re-calibrated (after 3-5 minutes of "cool down") if any drift is detected. This is to check for any problems (e.g. hole in membrane) before deployment.

Typical sonde deployment/retrieval equipment:

- Surveyor's flagging tape
- Steel T-posts (6 and 8 foot) and driver
- Tie wire
- Chain/cable and lock
- Diagonal pliers and lineman's pliers
- 4 inch diameter perforated PVC tube ("sonde cover")
- Digital camera
- GPS unit
- 30 m meter measuring tape
- Sonde Deployment/Download/Retrieval Field Sheets

Typically, the sonde is mounted inside a "sonde cover" and chained to a tree in the most secure location available. Other methods include mounting to a T-post, suspending from fence posts or bridges, and attaching to USGS gauging station structures.

The unattended sampling setup is as follows:

650 Main Menu
    System setup
        Disable "Power sonde" (un-check circle)

Sonde menu
Advanced
    Setup
        Enable "Auto sleep RS232" (check circle)

Sonde menu (same menu as above, just escape once to select Sensor menu)
Advanced
Sensor
Enable "wait for DO" (check circle)

Sonde menu (escape out of Advanced menu for this Sensor menu)
Sensor
Enable (check circles) the following sensors: Time, Temperature, Conductivity, Dissolved Oxy, ISE1 pH, Optic-T Turbidity, Battery

Sonde menu
Main
Run

- Unattended sample → Unattended setup
- Set interval, start date, start time, duration
- Create file name (up to 8 characters), site (number only)
- Scroll to "Start logging" and press enter key → Start Logging
- "Are you sure?" Enter "yes"

Detach cable from the sonde and cover the connection with the protective cap.

## 5.6 Sonde and Thermograph Calibration and Maintenance

### 5.6.1 Sonde Calibration and Maintenance

Since the SWQB utilizes only YSI multiparameter sondes, this section was written to address YSI equipment calibration and maintenance. Manufacturer's maintenance and calibration instructions for all instruments are kept for reference in the SWQB office. All calibration and maintenance activities must be recorded. Generally, calibration is recorded on the YSI Sonde Calibration Worksheet. The worksheets are stored in a 3-ring binder that is kept at the SWQB lab. Additionally, each sonde has its own logbook in which maintenance activities (e.g., probe replacement, software updates) are recorded. The logbooks must be taken into the field whenever the sonde is used in the field. Dissolved oxygen field calibration to adjust for changes in altitude may be recorded in the logbook (i.e., a Sonde Calibration Worksheet need not be completed for altitude adjustment calibrations).

As of 2007, YSI 6920, 6820, and 600XLM sondes are in service. Each is supplied with an operations manual. Operators shall refer to these manuals and follow the annotated instructions for calibration and maintenance. Recalcitrant problems should be referred to designated SWQB staff. In addition, updates and operational tips are available at https://www.ysi.com/ysi/Support.

#### *5.6.1.1 Surface Water Quality Monitoring Multiparameter Calibration Logbook*

Multiparameter Calibration Logbooks are kept on file. They must not be discarded. Additional blank logbooks are available from the SWQB. Each instrument has its own logbook. A

Multiparameter Calibration Logbook includes the following information, written in pencil or waterproof ink, and dark enough to permit photocopying.

- Date and time of calibration; initials of person conducting the calibration.
- Out-of-region instrument maintenance including: date shipped for repair; date returned from repair; and description of repair/work.
- In-region instrument maintenance including: date of maintenance; description of maintenance activity, i.e., battery replacement, probe cleaning, membrane replacement, and turbidity wiper maintenance. Include periodic observations made on the DO charge and notes on any sensor cleaning.

#### *5.6.1.2 Calibration Procedures*

All probes are calibrated in the SWQB lab before taking a sonde out into the field. During non-continuous daily use, the only probe that is regularly re-calibrated in the field is the DO sensor. This sensor is calibrated daily and with every gain or loss of elevation greater than 300 m (1,000 ft). Other probes are re-calibrated if the readings indicate a problem. In between long-term sonde deployments, all probes are cleaned if necessary (slow or unstable response) and re-calibrated prior to re-deployment. The DO membrane is also replaced due to the potential for bio-fouling during long-term deployment. Calibration supplies and tools for all probes are maintained in staff field kits.

*Dissolved Oxygen Sensor*

*DO Calibration Procedure for Instantaneous Sampling*

A. When using model 600XLM, 6820, or 6920 for instantaneous sampling, the auto sleep must be disabled. From the Main Menu, select 8-Advanced and then 2-Setup. If the Auto Sleep functions are enabled, select 5-Auto Sleep RS232 and 6-Auto Sleep SDI12 and press Enter to disable.

B. Place approximately 1/8 inch of water in the bottom of the YSI calibration (transport/storage) cup. Place the cup on the sonde body. Make certain that the DO and temperature probes are not immersed in the water. Wait at least 10 minutes for the air in the calibration cup to become water saturated and for the temperature to equilibrate. After 10 minutes, loosen the calibration cup such that only one thread of the calibration cup is engaged. This is to ensure that the DO probe is vented to the atmosphere (i.e., pressure inside the cup is equal to ambient atmospheric pressure).

C. From the Calibrate Menu, select 2-DO, then 1-DO percent to access the DO percent calibration procedure.

D. Obtain the estimated barometric pressure for your current elevation from the elevation/pressure table in appendix of the manufacturer's operations manual, or enter the current barometric pressure in mm of mercury (Hg) (inches of Hg × 25.4 = mm Hg) derived from a field barometer.

E. Observe the temperature and DO readings and when there is no significant change for approximately 30 seconds, press Enter. The screen will indicate that the calibration has been accepted and press Enter again to return to the Calibrate Menu.

F. Record calibration information (barometric pressure, DO charge, initial and calibrated DO % saturation values and DO gain (Sonde Menu → Advanced → Cal Constants)) in the logbook or calibration worksheet.

**NOTE**: Remember to use barometric pressure readings that have not been corrected to sea level (i.e., absolute barometric pressure). Weather reports report barometric pressure corrected to sea level.

*DO Calibration Procedure for Unattended Sampling (Logging)*

A. When using the Model 600XLM or Model 6920 in the unattended mode, the "auto sleep" (Sonde Menu → Advanced → Setup → Enable Auto Sleep RS232) must be enabled, as must "wait for DO" (Sonde Menu → Advanced → Sensor). "Power Sonde" (650 Main Menu → System Setup → Power Sonde) must be disabled.

B. Follow the calibration procedure described in A – F above.

DO warm-up time should not be decreased; it may be increased to yield better results. Contact YSI before adjusting the DO warm-up time.

*DO Charge*

DO charge must be within the range of 25 millivolts (mV) to 75 mV. Low charge likely indicates dilute electrolyte solution (perforated membrane). Replace electrolyte and membrane, then recalibrate DO (allow probe to pulse for a minimum of 15 minutes before calibrating). There are two likely causes of high (>75 mV) charge: moisture in the probe socket and a short in the probe itself. Inspect socket for moisture; if socket is wet, rinse with DI water, then 95% ethanol, blow out with canned air and allow to air dry for 24 hours. If socket is dry, replace probe and return the old probe to the sonde maintenance manager for reconditioning or disposal.

*Conductivity Sensor*

Conductivity standards are very sensitive to contamination. The date the container is opened must be inscribed on the label and the solution should be discarded one month after opening. Conductivity calibration should always be performed before pH calibration. Be sure that the probe and calibration cup are clean and rinsed with the calibration standard.

**NOTE**: standards with conductivities less than 10 mS/cm (10,000 µS/cm) are not recommended.

A. Rinse the sensor twice with the conductivity standard. Place the probe into the standard solution and make sure that the probe is completely immersed, past the vent holes. Gently tap the side of the calibration cup to dislodge any air bubbles from the probe.

B. Allow for temperature equilibration to occur before proceeding.

C. From the Calibrate Menu, select 1-SpCond to access the SC calibration procedure. Enter the calibration value of the standard and press Enter. The current values of all enabled sensors will appear on the screen and will change with time as they stabilize.

D. Observe readings under SC and when they show no significant change for approximately 30 seconds, press Enter. The screen will indicate that the calibration has been accepted; press Enter again and return to the Calibrate menu.

E. Record calibration information (see "YSI Calibration Worksheet"). Calibration error limit for SC is ± 5 percent. Rinse probe in tap or DI water

**NOTE**: This procedure calibrates SC, salinity, and total dissolved solids.

*pH Sensor*

The pH system is calibrated with a buffer of pH 7.0 and either 4.0 for acidic waters or pH 10.0 for alkaline waters. The pH buffers contain high concentrations of phosphate. Care must be taken during calibration to avoid leaving traces of buffer on equipment or at the work place that could contaminate water samples. The date the container is opened must be inscribed on the label. Buffer solutions prepared from reagent powder or concentrate are labeled with date of preparation. All buffer solutions are discarded after one month.

*Two-Point Calibration:*

<u>pH 7:</u>
<u>Calibration to pH 7 is always performed first.</u>

A. Place enough pH 7 buffer into a clean, dry or pre-rinsed calibration cup to immerse the tip of the pH probe and thermistor (i.e., the temperature sensor on the conductivity probe). Allow the temperature to equilibrate before reading.

B. From the Calibrate Menu, select 4-ISE 1 pH to access the pH calibration choices; then press 2-2 Point (or 3-3 Point). Press Enter and input the value of the buffer at the prompt. Press Enter and the current values of all enabled sensors will appear on the screen.

C. Record the pH mV reading. This value should range from -50 to +50. Observe the pH reading and when it shows no significant change for approximately 30 seconds, press Enter. The display will indicate that the calibration is accepted.

D. After the pH 7 calibration is complete, press Enter again to continue. Rinse the probes in water and the next buffer to be used before proceeding.

<u>pH 4 and 10:</u>

E. Next, place enough pH 10 buffer into a clean, dry or pre-rinsed calibration cup to immerse the tip of the pH probe and thermistor. Allow the temperature to equilibrate before reading. Observe the pH mV reading. This value should range from +177 mV from the pH 7 reading in pH 4 buffer to -177 mV from the pH 7 reading in pH 10 buffer.

F. Press Enter and input the value of the second buffer at the prompt. Press Enter and the current values of all enabled sensors will appear on the screen.

G. Observe the pH reading and when it shows no significant change for approximately 30 seconds, press Enter. After the second calibration is completed, press Enter again. If performing a 2-Point Calibration the screen will return to the Calibrate Menu.

H. Record calibration information (see "YSI Sonde Calibration Worksheet"). Rinse the probes with water. Rinse the calibration cup for future use.

**NOTE:** If the range of expected pH values is < 7.0, use pH 4 buffer in steps E through H for the 2-point calibration instead of pH 10 buffer. Observe the pH mV reading. This value should be +177 mV from the reading obtained in pH 7 buffer.

*Temperature Sensor*

Thermistors cannot be calibrated. Annually, or when a malfunction is suspected, check the temperature reading against a National Institute of Standards and Technology thermometer to ensure suitable instrument performance.

*Turbidity Sensor*

The date the turbidity standard container is opened must be inscribed on the label and the solution should be discarded one month after opening. Prior to using any turbidity standard it is critical to gently agitate the standard to re-suspend the formazin. Failure to do so will not only bias your calibration (high), But all those that follow (low). Be sure to use a calibration cup with the same color bottom as the turbidity probe (i.e., either black or gray).

A. Observe and record wiper park position on calibration record form.

B. Perform a 2 point test (DI water for 0 NTU and 100 NTU standard).

C. Double rinse the YSI sonde calibration cup with DI water.

D. Remove excess liquid from probe by blotting probe with Kimwipe between procedures.

E. Fill cup with DI water to the top of the narrow, bottom portion of cup.

**Test 1:** Enter 0 at the prompt; and press Enter

Record actual and calibrated values (NTUs), press Enter to accept the calibration. Discard the DI water

**Test 2:** Enter 100 NTU at the prompt;

Gently swirl/mix standard (do not create air bubbles), rinse storage cup twice with a small amount of standard;

Gently pour standard down side of cup, fill to the top of the narrow portion of cup, loosely screw the calibration cup onto the sonde and press Enter. Press Enter again to accept the calibration. and

Record actual and calibrated values (NTUs).

*Calibration Error Limits*

The manufacturer-stated calibration error limits for the YSI Multiparameter sonde are as follows:

- DO ±0.5 mg/L
- pH ±0.5 standard units

- SC ±5 percent
- Temperature ±1 °C
- Turbidity is the greater of +/- 5% or 2 NTUs

If calibration values fall outside the error limits for DO, pH, and SC, the data collected does not meet SWQB Program QA. These data should not be reported. If calibration measurements do not consistently fall within the error limits (and after in-house trouble shooting, Table 1.2 in the Care and Maintenance section) of the manufacturer's operation manual, the instrument should be returned to the manufacturer for maintenance.

#### *5.6.1.3 Care and Maintenance*

Please refer to the instrument manual or manufacturer for detailed maintenance requirements specific to YSI instruments.

*O-Rings*

If the O-rings and sealing surfaces on the sondes are not maintained properly, it is possible that water can enter the battery compartment and/or sensor ports of the sonde. If water enters these areas, it can severely damage the battery terminals or probe ports causing loss of battery power during a deployment, inaccurate readings, and corrosion to the contacts. Therefore, when the battery compartment lid is removed from 600XLM or 6920sondes, the O-rings that provide the seal should be carefully inspected for contamination (e.g. hair, grit, etc.) and cleaned if necessary using the instructions provided below. The same inspection should be made of the O-rings associated with the probes, port plugs, and field cable connectors when they are removed. If no dirt or damage to the O-rings is evident, then they should be lightly greased (see below) without removal from their groove. However, if there is any indication at all of damage, the O-ring should be replaced with an identical item from the YSI 6570 Maintenance Kit supplied with the sonde. At the time of O-ring replacement, the entire O-ring assembly should be cleaned as described below.

***To remove the O-rings:***

Use a small, flat-bladed screwdriver or similar blunt-tipped tool to remove the O-ring from its groove. Check the O-ring and the groove for any excess grease or contamination. If contamination is evident, clean the O-ring and nearby plastic parts with lens cleaning tissue or equivalent lint-free cloth soaked in a mild detergent solution.

***CAUTION:***

Using alcohol on O-rings may cause a loss of elasticity and may promote cracking. Do not use a sharp object to remove the O-rings. Damage to the O-ring or the groove itself may result. Before re-installing the O-rings, make sure that you are using a clean workspace, clean hands, and are avoiding contact with anything that may leave fibers on the O-ring or grooves. Even a very small bit of contamination (hair, grit, etc.) may cause a leak.

***To re-install the O-rings:***
Place a small amount of lubricant supplied in the YSI 6570 Maintenance Kit or food-grade silicone grease between your thumb and index finger. (More grease is NOT BETTER!) Draw the O-ring through the grease while pressing the fingers together. Use this action to place a VERY LIGHT covering of grease to all sides of the O-ring. Place the O-ring into its groove making sure that it does not twist or roll. Use the previously grease-coated finger to once again lightly go over the mating surface of the O-ring. DO NOT use excess grease on the O-ring or the O-ring groove. The grease is a lubricant, not a sealant.

***CAUTION:*** Do not over-grease the O-rings. The excess grease may collect grit particles that can compromise the seal. Excess grease can also cause the waterproofing capabilities of the O-ring to diminish, potentially causing leaks into the compartment. If excess grease is present, remove it using lens cloth or lint-free cloth.

*Sonde Probe Ports*

Whenever you install, remove, or replace a probe or port plug, it is extremely important that the entire sonde and all probes and plugs be thoroughly dried prior to the removal This will prevent water from entering the port. Once you remove a probe or plug, examine the connector inside the sonde probe port. If any moisture is present, rinse both the port and the probe with DI water, remove the water with three rinses of 95% ethanol and dry thoroughly with compressed air. Equipment subjected to this procedure must air dry for at least 24 hours before re-assembly. If the connector is corroded, return the sonde to the sonde maintenance manager. When you reinstall a probe or port plug, lightly grease the O-ring with lubricant supplied in the YSI 6570 Maintenance Kit or food-grade silicone grease.

*Cable Connector Port*

The cable connector port at the top of the sonde should be covered at all times. When a communications cable is not connected to the cable connector port, the pressure cap supplied with the instrument should be securely tightened in place.

If moisture has entered the connector port, dry it completely using 95% ethanol and compressed air. Never attempt to dry the connector port with a rag or paper towel as this may bend the pins. Apply a very thin coat of lubricant from the 6570 Maintenance Kit or food-grade silicone grease to the O-ring inside the connector cap before each installation.

*Sonde Probes*

*6562 DO Probes*

For best results, we recommend that the potassium chloride (KCl) solution and the Teflon membrane at the tip of the 6562 probe be changed prior to each sonde deployment and at least once every 30 days during the use of the sonde in sampling studies. In addition, the KCl solution and membrane should be changed if:

- Bubbles are visible under the membrane;
- If the DO charge is outside a range of <25 mV and >75 mV;

- Significant deposits of dried electrolyte are visible on the membrane or the O-ring; or
- Probe shows unstable readings or is slow to stabilize.

After removing the used membrane from the tip of the 6562 probe, examine the electrodes at the tip of the probe. If either or both of the silver electrodes are discolored or pitted, the probe should be resurfaced using the fine sanding disks which are provided in the 6035 reconditioning kit. To resurface the probe using the fine sanding disk, follow the instructions below.

A. Dry the probe tip completely with lens cleaning tissue.

B. Hold the probe in a vertical position, place one of the sanding disks under your thumb, and stroke the probe face lightly in a direction parallel to the gold electrode (located between the two silver electrodes). The motion is similar to that used in striking a match. Usually 10-15 strokes of the sanding disk are sufficient to remove black deposits on the silver electrodes. However, in extreme cases, more sanding may be required to regenerate the original silver surface.

C. Repeatedly rinse the probe face with clean water and wipe with lens cleaning tissue to remove any grit left by the sanding disk. After cleaning, thoroughly rinse the entire tip of the probe with distilled or DIW followed by KCl solution and install a new membrane.

**IMPORTANT**: Be sure to:

- Use *only* the fine sanding disks provided in the 6035 maintenance kit in the resurfacing operation;
- Avoid any contamination of the contacts with fluids;
- Sand in a direction parallel to the gold electrod*e. Not adhering to either of these instructions can seriously damage the electrodes.*

**NOTE:** If this procedure is unsuccessful, as indicated by improper probe performance, it may be necessary to return the probe to the sonde maintenance manager for evaluation.

*6560 Conductivity/Temperature Probes*

The openings that allow fluid access to the conductivity electrodes must be cleaned if response is slow or the reading fails to stabilize. The small cleaning brush included in the 6570 Maintenance Kit is provided for this purpose. Dip the brush in a mild detergent solution and insert it into each hole 15-20 times; rinse well. Never use anything but mild detergent to clean a turbidity probe. After cleaning, check the response and accuracy of the conductivity cell with a calibration standard.

**NOTE:** If this procedure is unsuccessful, or if probe performance is impaired, it may be necessary to return the probe to the sonde maintenance manager for evaluation.

*Temperature*

The temperature portion of the probe requires no maintenance.

*6561 pH and 6565/6566 Combination pH-ORP Probes*

Cleaning is required whenever deposits or contaminants appear on the glass and/or platinum surfaces of these probes or when the response of the probe becomes slow or unstable.

A. Soak the probe in a dilute detergent solution for 10 minutes. Using a soft cloth or cotton swab dipped in the detergent solution, gently wipe the bulb and reference electrode. Rinse thoroughly.

B. Soak the probe in dilute hydrochloric acid (1 molar) for 10 minutes. Using a soft cloth or a cotton swab dipped in the dilute hydrochloric acid, gently wipe the bulb and reference electrode. Soak the probe in clean water for one hour.

***CAUTION:*** When using a cotton swab with the 6561 or 6565, be careful NOT to wedge the swab tip between the guard and the glass sensor. If necessary, remove cotton from the swab tip, so that the cotton can reach all parts of the sensor tip without stress.

If biological contamination of the sensor is suspected, or if good response is not restored by the above procedures, perform the following additional cleaning step:

A. Soak the probe for approximately 1 hour in a 1 to 1 dilution of commercially-available chlorine bleach and DI water.

B. Rinse the probe with clean water and then soak for at least 1 hour in clean tap water with occasional stirring to remove residual bleach from the sensor. (If possible, soak the probe for period of time longer than 1 hour in order to be certain that all traces of chlorine bleach are removed.)

C. Re-rinse the probe with clean water and retest.

*Optical Probes–6026 and 6136 Turbidity*

The 6026 and 6136 probes require only minimal maintenance. After each deployment, the optical surface on the tip of the turbidity probe should be inspected for fouling and cleaned if necessary by gently wiping the probe face with moist lens cleaning paper. In addition, for the 6026 and 6136 probes, we recommended replacing the wiper pad when it becomes discolored. Do not discard wiper arms. The frequency of this replacement depends on the quality of water under examination.

A replacement wiper is supplied with the probes, along with the small hex driver required for its removal and reinstallation. Follow the instructions supplied with the probe to ensure proper installation of the new wiper assembly. Spare wipers and pads are kept in stock by SWQB.

**Table 5-1 Sensor troubleshooting**

| Symptoms | Possible Cause | Action |
|---|---|---|
| DO reading unstable or | Probe not properly calibrated | Follow DO calibration procedures |
| | Membrane not properly installed | Follow 6562 setup procedure |

**Table 5-1 Sensor troubleshooting**

| Symptoms | Possible Cause | Action |
|---|---|---|
| inaccurate | DO probe electrodes require cleaning | Follow DO cleaning procedure. Use 6035 maintenance kit |
| | Water in probe connector | Dry connector; reinstall probe |
| | Algae or other contaminant clinging to DO probe | Rinse DO probe with clean water |
| | Calibrated using improper barometric pressure | Repeat DO calibration procedure using proper barometric pressure |
| | Calibrated at extreme temperature | Recalibrate at (or near) sample temperature |
| | DO Charge too high ( >75)<br>1. Anodes polarized (tarnished)<br>2. Probe left on continuously | Recondition probe with 6035 Maintenance Kit. Follow DO cleaning procedure. |
| | DO Charge too low (<25)<br>1. Insufficient electrolyte.<br>2. DO probe has been damaged<br>3. Internal failure | Replace electrolyte and membrane.<br>Replace 6562 probe<br>Return sonde for service |
| pH, ORP, readings are unstable or inaccurate. Error messages appear during calibration. | Probe requires cleaning, | Follow probe cleaning procedure |
| | Probe requires calibration | Follow calibration procedures |
| | pH probe sensor has dried out from improper storage. | Soak probe in tap water or buffer until readings become stable |
| | Water in probe connector | Dry connector; reinstall probe |
| | Probe has been damaged | Replace probe |
| | Calibration solutions out of spec or contaminated with other solution | Use new calibration solutions |
| | Internal failure | Return sonde for service |
| Conductivity unstable or inaccurate. Error messages appear during calibration. | Conductivity improperly calibrated. | Follow calibration procedure |
| | Conductivity probe requires cleaning | Follow cleaning procedure |
| | Conductivity probe damaged | Replace probe |
| | Calibration solution out of spec or contaminated | Use new calibration solution |
| | Internal failure | Return sonde for service |

**Table 5-1 Sensor troubleshooting**

| Symptoms | Possible Cause | Action |
|---|---|---|
| | Calibration solution or sample does not cover entire sensor. | Immerse sensor fully. |
| Temperature, unstable or inaccurate | Water in connector | Dry connector; reinstall probe |
| | Probe has been damaged | Replace the 6560 probe |
| Turbidity probe: general | Probe requires cleaning. | Follow probe cleaning procedure |
| | Probe requires calibration | Follow calibration procedures |
| | Probe has been damaged | Replace probe |
| | Water in probe connector | Dry connector; reinstall probe |
| | Calibration solutions out of spec | Use new calibration solutions |
| | Wiper is not turning or is not synchronized. | Activate wiper. Assure rotation. Make sure setscrew is tight. |
| | Wiper is fouled or damaged. | Clean or replace wiper. |
| | Internal failure. | Return probe for service. |
| Installed probe has no reading | Sensor has been disabled | Enable sensor |
| | Water in probe connector | Dry connector; reinstall probe |
| | Probe has been damaged | Replace the probe |
| | Report output improperly set up | Set up report output |
| | Internal failure | Return sonde for service. |

#### *5.6.1.4 Short Term Storage*

No matter what sensors are installed in the instrument, it is important to keep them moist without actually immersing them in liquid, which could cause some of them to drift or result in a shorter lifetime. For example, the sensor of a pH probe must be kept moist to minimize its response time during usage, but continued immersion in pure water may compromise the function of the glass sensor and/or result in long term leaching of the electrolyte through the reference junction.

YSI recommends that short term storage of all multiparameter sondes be done by placing approximately 3 mm (1/8 inch) of water in the calibration / storage cup that was supplied with the sonde, and by placing the sonde with all of the probes in place into the cup.

The key for interim storage is to use a minimal amount of water so that the air in chamber remains at 100 percent humidity. The water level has to be low enough so that none of the sensors are actually immersed. Use clean tap water for storage between sampling runs. If the storage water is inadvertently lost during field sampling studies, environmental water can be used to provide the humidity. Do not use DI water, as this will degrade the performance of the pH probe.

Interim sonde storage is easy. Simply remember the following key points:

- Use enough water to provide humidity, but not enough to cover the probe surfaces.
- Make sure the storage vessel is sealed to minimize evaporation.
- Check the vessel periodically to make certain that water is still present.

#### *5.6.1.5 Long Term Storage*

The following recommendations are applicable for sondes with typical sensor configurations.

**600XLM –** Remove the pH or pH/ORP probe from the sonde and store it according to the instructions found in the following section on individual sensors. Seal the empty port with the provided plug. Leave the conductivity/temperature and the DO probe in the sonde with a membrane and electrolyte on the DO sensor. Place enough deionized, distilled, or tap water in the calibration cup to cover the sensors, insert the sonde into the vessel, and seal with the cap/O-ring to minimize evaporation.

**6820, 6920 –** Leave the conductivity/temperature, turbidity, and DO probes in the sonde with a membrane and electrolyte on the DO sensor. Remove the pH probe from the sonde and store according to the instructions found in the following section on individual sensors. Seal the empty ports with the provided plugs. Place enough deionized, distilled, or tap water in the calibration cup to cover the sensors, and tighten the threaded cup to attain a good seal and minimize evaporation.

**All Sondes with Batteries** – Because batteries can degrade over time and release battery fluid, it is extremely important to remove the batteries from all 600XLM and 6920 sondes prior to long term storage. Failure to remove batteries can result in corrosive damage to the battery compartment and terminals if the batteries leak.

*Probes*

The following sections provide additional details on the storage of individual sensors associated with instruments in the 6-Series product line from YSI. Probes should be cleaned prior to being placed in long term storage.

*Temperature*

No special precautions are required. Sensors can be stored dry or wet, as long as solutions in contact with the thermistor probe are not corrosive (for example, chlorine bleach).

*Conductivity*

No special precautions are required. Sensors can be stored dry or wet, as long as solutions in contact with thermistor probe and conductivity electrodes are not corrosive (for example, chlorine bleach). However, it is recommended that the sensor be cleaned with the provided brush prior to long term storage.

*Dissolved Oxygen*

Rapid pulse DO sensors should always be stored with a membrane and electrolyte in place and in such a way that drying of the electrolyte on the probe face is minimized. For long-term storage, the medium should be water rather than the moist air used in interim storage. For the 600XLM, 6820, and 6920, two long-term storage methods are equally acceptable.

- Remove all probes other than DO, conductivity, and turbidity from the sonde and seal the vacant ports with the provided port plugs. Leave the electrolyte and membrane in place on the DO sensor. Fill the calibration cup half way with tap water and insert the sonde. Make certain the water level is high enough to completely cover the DO sensor. Seal the vessel to prevent evaporation of the water. At the end of the storage time, remove the existing membrane and re-membrane the probe.

*pH*

The key to pH probe storage, short or long-term, is to make certain that the reference electrode junction does not dry out. Junctions which have been allowed to dry out due to improper storage procedures can sometimes be rehydrated by soaking the sensor for several hours (overnight is recommended) in a 2 molar potassium chloride solution. If potassium chloride solution is not available, soaking the sensor in commercial pH buffers may restore probe function. However, in some cases the sensor may have been irreparably damaged by the dehydration and will require replacement. It is also important to remember not to store the pH sensor in distilled or DIW as the glass sensor may be damaged by exposure to this medium and the electrolyte will be depleted through the reference electrode.

*Turbidity*

No special precautions are necessary for either the short or long-term storage of the turbidity probe. However, for long-term storage, the user may wish to remove the probe from the sonde, replace it with a port plug, and store the probe in dry air to minimize any cosmetic degradation of the probe body and to maximize the life of the wiper.

#### *5.6.1.6 Scheduled Maintenance*

Minimum maintenance must be conducted, regardless of the use that the instruments get. Please refer to the instrument manual or manufacturer for scheduled maintenance requirements specific to YSI instruments.

After each sampling trip, post-calibrate the instrument after general cleaning and maintenance. Following post calibration, rinse off the sensors and store them in tap water. Do not use distilled or DIW for storage. These are important steps in preventive maintenance that are done each day the instrument is used.

#### *5.6.1.7 Replacement Parts*

See appropriate staff member for replacement parts. Do not discard any malfunctioning parts, as these may be under warranty.

### 5.6.2 Thermograph Accuracy Verification and Maintenance

A National Institute of Standards and Technology (NIST) traceable thermometer, with a resolution and accuracy of 0.1°C or better, is used to verify thermograph accuracy. The NIST

traceable thermometer itself should be verified annually, with a minimum of two temperatures. The New Mexico Department of Health, SLD, can certify a thermometer for accuracy and NIST traceability at no cost to SWQB.

Accuracy of the thermograph must be tested annually, at a minimum of two calibration temperatures between 0°C and 25°C. Testing is done using a stable thermal mass, such as an ice water bath or other controlled water bath. The stable temperature of the insulated water mass allows direct comparison of the unit's readout with that of the certified thermometer. Accuracy should be within ±0.5°C.

A log by the thermograph coordinator is kept that documents each unit's calibration date, temperature discrepancy, and reference thermometer used. Thermographs that fall outside the acceptable accuracy range are returned to the manufacturer. Each year prior to field season, the thermograph coordinator prepares the thermographs for deployment utilizing the following procedures:

- Program thermographs to record intervals of (approximately) fifteen minutes.
- Place thermographs in a cooler or other insulated container. Fill the container part way with ice and add enough cold water so that most of the spaces between the pieces of ice are filled, but the ice is not floating above the thermographs.
- Record the water temperature as indicated with a NIST traceable thermometer. It should be at or near 0°C.
- Place the container in a walk-in refrigerator and allow the thermographs to equilibrate for at least twenty minutes.
- After equilibration, allow the thermographs to record for one hour. At the end of this procedure, the ice should not have melted to the point that it is floating above the thermographs. If ice has melted, return to the first procedure and begin again. The thermographs should have recorded a temperature ±0.5°C of the NIST recorded temperature.
- Next, set up a well-mixed warm water bath that is near room temperature.
- Place the thermographs in the warm water bath and allow them to stabilize for at least twenty minutes.
- Check the temperature of the water with a NIST traceable thermometer, taking care to ensure that the bath is well mixed and has not stratified by the time the thermographs are set to record (all the thermographs should be set to record simultaneously). Allow the thermographs to record for four intervals.
- The thermographs should have recorded a temperature ±0.5°C of the NIST recorded temperature.

**FORMS**

Field Data Sheet
EMAP–based Densiometer Only Field Worksheet
Thermograph Deployment/Download/Retrieval Field Sheets

Sonde Deployment/Download/Retrieval Field Sheets
Sonde Calibration Worksheet

## REFERENCES

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# 6.0 MEASURING FLOW

The measurement of flow in conjunction with surface water and wastewater sampling is essential to almost all SWQB activities. Activities such as water quality surveys, NPDES permit compliance monitoring, TMDL development, nonpoint source monitoring, assessment of fluvial geomorphological conditions, planning, and research rely on accurate flow measurement data.

A complete discussion of all available flow measurement techniques and the theory behind them is beyond the scope of this manual. However, most of the common techniques in current use are covered.

Flow measurements of surface waters are open-channel flow measurements. Most surface water flow measurements are made using classical stream-gauging techniques involving the velocity × area open-channel technique. Personnel may refer to standard stream flow measurement techniques outlined in USGS publications to select flow measuring sites, perform stream-gauging procedures, and calculate flow (Buchanan and Somersto 1969).

Where permanent gauging stations are not present, surface water flow should be measured utilizing classical stream-gauging techniques. If data are to be collected from a particular station several times during a water quality survey, a rating curve may be developed for that station by making a series of independent flow measurements and simultaneous cross-sectional profile measurements at the same location. See Section 6.3.5 for techniques for developing flow rating curves. Flow measurements should bracket the lowest and highest flows encountered during the study. A rating curve may be constructed by plotting stage against flow on a semi-log graph.

## 6.1 Using Flow and Current Meters

See Section 6.3 for alternative methods of determining flow.

### 6.1.1 Measuring Velocity with the Marsh-McBirney Flow Meter

The following information is a summary of the Marsh-McBirney, Inc. Flo-Mate 2000 Portable Flowmeter instruction manual (1990). The Marsh-McBirney flow meter measures velocity with an electromagnetic sensor and displays results in either feet per second (ft/s) or meters per second (m/s).

#### *6.1.1.1 Marsh-McBirney Assembly and Calibration*

- Remove sensor and a comfortable length of cable from the flow meter case. Attach the sensor to the English standard wading rod on the mounting shaft at the bottom and tighten the thumbscrew.

- The unit should always power up in real time operating mode, as opposed to memory recall. The display should be set to Fixed Point Averaging (FPA), where the readings are stabilized by averaging velocities over a fixed period of time (the alternative is time

constant filtering [rC]). Pressing the up and down arrows simultaneously will alternate between rC and FPA displays and the FPA display will show the letters FPA when you first switch over and afterwards will be indicated by the time period bar at the bottom of the display. The fixed period of time default is 40 seconds and may be altered with the up or down arrows. SWQB uses 40 seconds as its time period standard.

- Calibrate the sensor at the beginning of each field season. To calibrate the sensor, first clean it with soap and water. If there are nonconductive coatings, such as oil or grease, errors may occur due to noise or conductivity loss. Do not use any hydrocarbon solvents for cleaning. Place the sensor in a 5-gallon bucket filled with water at least 3 inches away from the sides and bottom of the bucket (use the wading rod to hold it in the appropriate position). Wait 10 or 15 minutes to make sure that the water is not moving. Set the time period to 5 seconds. The zero stability is ± 0.05 ft/sec. In order to reset the zero, press STO and RCL at the same time while the sensor is still in the bucket (unmoved). The number 3 will display. Decrement the 3 to 0 with the down arrow. Then the number 32 will display and the unit will decrement itself to zero and turn off. It is now calibrated. Make sure you return the time period to 40 seconds.

Note: There is a 5 second time limit between keys when zeroing. If you wait too long and ERR 3 is displayed, you need to turn the power off and start over.

#### *6.1.1.2 Marsh-McBirney Operation*

1. Place the wading rod at the appropriate location (see Section 6.2 for details on taking measurements through a cross section) then stand several inches downstream from the tag line and about 18 inches to one side of the meter to avoid disrupting the current near the meter. For small streams, where your feet would occupy a significant portion of the wetted cross section, stand on the bank, entirely out of the water. Ensure that the wading rod is vertical (if unsure, hold a rod level against the wading rod to check).

2. Turn on the meter and check all of the settings. Press clear (ON/C) at your first location and wait for the reading to stabilize. Record velocity. If the reading does not stabilize, wait for the time bar to fill completely and record the velocity. Move to the next location, press clear to reset the averaging, and repeat the procedure.

#### *6.1.1.3 Marsh-McBirney Error Messages*

- *Noise* – Indicates that there is excessive electrical noise in the water, and the display will blank out. It is normal for this message to appear for a few seconds when the sensor is first submerged.
- *Con Lost* – Indicates that the electrodes are out of the water or have been coated with grease and conductivity is lost. If this continues after washing with soap and water, the electrodes can be cleaned with very fine (600 grit) sandpaper. After 5 minutes of this message, the unit will shut down.
- *Low Bat* – Replace the 2 D-cell batteries in the bottom of the unit. Usually when this flag appears the battery will have 15-30 minutes of life left.

### 6.1.2 Measuring Velocity with the Price Current Meter (Pygmy or AA)

The following information is a summary of, and intended for use in conjunction with, the Scientific Instruments, Inc. Model 1205 Price Type "Mini" Current Meter user's manual. All "Price-type" current meters operate essentially the same way regardless of size, method of measurement, suspension means, or data collection methods.

A precisely balanced bucket wheel is mounted on a vertical pivot and is turned by flowing water. Water velocity is determined by counting the number of revolutions of the bucket wheel over a given period of time. A "cat whisker switch" produces a signal that is either manually counted or recorded by a counting instrument. This value, or count, is then compared on a cross reference chart, or rating chart, to yield water flow velocity.

- For velocities less than 3 ft/s, use the Mini (Pygmy) current meter.
- For non-wadeable flows or velocities greater than 3 ft/s, use the Price AA current meter.
- According to the USGS, a current meter should not be used in velocities less than 0.2 ft/s, unless there is no other option.

#### *6.1.2.1 Price Meter Assembly and Calibration*

- *Inspect/assemble the meter*. Remove the meter from the travel case and inspect the buckets for damage. Any visual damage, such as a bent cup, will yield an erroneous measurement. Replace the brass travel plug with the stainless steel pivot. Inspect the pivot for fractures, roughness, or other evidence of wear (use a new one as necessary). Put a drop of oil on the pivot. Insert the pivot firmly against the pivot-adjusting nut (the pivot has a flat side that should be positioned beneath the set screw that secures the pivot).

- *Ensure that the pivot is not too loose*. The rotor (bucket wheel) should have a very small amount of vertical play, such that it is not impaired by the pivot while rotating. Loosen the pivot nut set screw, adjust the pivot nut out one-eighth turn, and re-tighten the set screw. Loosen the pivot set screw, insert the pivot against the new adjusting-nut position, and re-tighten this set screw. Check rotor play again and repeat this step as necessary.

- Do not turn the bucket wheel clockwise. This can damage the contact wires, causing inaccurate readings. Always rotate counterclockwise as viewed from above.

- *Ensure that the pivot is not too tight.* The rotor should spin freely (be sure it's right-side up, because it will not spin freely when upside down). If the rotor tends to stop abruptly, inspect the pivot and pivot bearing (on the rotor) for damage. If the pivot bearing is too tight, loosen it by turning the pivot-adjusting nut out (use one-eighth-turn increments, as described above).

- *Conduct the spin test* This procedure must be performed completely out of the wind, such as in the vehicle. After assembling the meter, attach it to the wading rod. Tighten the mounting screw and attach the sensor wire to the binding post, ensuring that the electrical

connection does not short-circuit from the post to the meter housing. Attach the counter or flow computer to the plug at the top of the wading rod. While holding the wading rod so that the rotor is horizontal (as used in the water), give the rotor a brisk flip with your finger (always counterclockwise; spinning clockwise may damage the sensor) and ensure that the counter is registering the spinning rotor. Ensure that the rotor spins for at least one minute and comes slowly to a stop. If it doesn't, readjust the pivot or identify and fix the meter damage.

#### *6.1.2.2 Price Meter Operation*

1. Place wading rod at the appropriate location (see Section 6.2 for details on taking measurements through a cross section), then stand a couple of inches downstream from the tag line and about 18 inches to one side of the meter to avoid disrupting the current near the meter. For small streams, where your feet would occupy a significant portion of the wetted cross section, stand on the bank, entirely out of the water. Ensure that the wading rod is vertical (if unsure, hold a rod level against the wading rod to check). Give the meter a few moments to stabilize in the flow (several seconds or longer in velocities less than one foot per second).

2. Using the Model 921 Revolution Counter™, press "on" and start the stopwatch simultaneously. Count revolutions during a period of 40 to 70 seconds. End the measurement at some convenient number of revolutions (generally a multiple of five, as listed across the top of the rating table included with the meter). Using the elapsed time (down the left side of the rating table), find the water velocity.

3. Using the CMD 9000™ computerized sensor, turn the meter on, select the appropriate sensor, and adjust the other settings ("English" or "metric," etc.). When ready, press "start/stop." If the instrument is functioning properly, it will automatically count and determine the velocity.

#### *6.1.2.3 Price Meter Troubleshooting*

Some waters cause an intermittent failure of the meter. This seems to be a mechanical failure of the whisker sensor associated with high turbidity resulting from some forms of silt-clay. SWQB staff have not found a solution to this problem, except to use a different measurement method (such as the Marsh-McBirney meter). Following exposure of the Price to such waters, it must be disassembled and rinsed well (it appears to begin functioning properly after about ten minutes use in clear water).

## 6.2 Flow Measurements

When establishing the cross section the flow across should have the following characteristics:

- The cross section should be located in a straight run or gentle riffle. Do not select a cross section through a pool. Where the length of straight run is limited, the length upstream from the cross section should be twice the downstream length.

- The channel should be free of flow disturbances. Look for protruding pipe joints, sudden changes in stream width, contributing side streams, outgoing side streams or obstructions.
- Depths should ideally be greater than 0.5ft, and velocities mostly greater than 0.5 ft/s.
- The flow should be laminar (i.e., free of swirls, eddies, vortices, backward flow, or dead zones). Avoid areas immediately downstream from sharp bends or obstructions.
- The ideal streambed is "U" shaped, stable, and free from large rocks, weeds, and protruding obstructions that would create turbulence or interfere with the current meter. The investigator may modify the channel by removing cobbles that interrupt flow, or to temporarily make the channel deep enough to measure. Alternatively, consider using the timed-fill method, which is more accurate at very low flows (Section 6.3.1).
- If necessary, build small dikes to keep stream water from flowing around the cross section through a secondary channel (although choosing a better site is strongly recommended in this situation).

String a tape measure or lay a survey rod (tag line) across the stream at the chosen cross section. The tag line should be staked at or beyond bankfull on both sides of the channel, perpendicular to the direction of flow (not necessarily at right angles to the whole channel), and about one foot above the water level. Place the zero at left bankfull or beyond when facing downstream and make sure the tape is tight and level (Marsh-McBirney 1990).

### 6.2.1 Establishing Cross Sectional Windows

After the cross section location has been established, measure the stream wetted width. Divide this width into at least 20 partial sections (windows). When necessary to adjust for changing stream bed dimensions and velocities, shorten or widen the window width. Measurements should always be taken in the center of the window. It is not necessary to have equal window widths – the goal is to have no more than 5% of the total flow in each window.

If the channel is too narrow to establish 20 windows, then flow may be estimated using fewer windows. These data should be flagged as "estimated flow."

### 6.2.2 Determining Appropriate Method for Flow Measurement

For very low flows, use the timed-fill method described in Section 6.3.1.

To calculate flows in less than 2 inches (0.15 ft) of depth, use the $0.9 \times U_{max}$ method (Marsh-McBirney 1990):

1. Take a series of point velocity measurements throughout the entire flow.
2. Identify the fastest velocity.
3. Multiply the fastest velocity by 0.9 for approximate mean velocity ($\bar{U}$).
4. Use this velocity with a measured cross section to find stream flow.

When depth is more than 0.15 ft but less than 2.5 ft, the velocity measurement is taken at a depth that is equal to 60% of the total depth when measured from the surface of the water. This is automatically calculated when using the USGS type top-set wading rod, which is calibrated to quickly set the 60% point by moving the sliding rod so that the foot measurement on it lines up with the tenth of a foot indication on the scale at the top of the fixed rod. For example, if the

stream depth is measured at 1.4 feet, the one foot indication on the sliding rod would be moved up until it corresponds with the 4 on the scale at the top of the fixed rod.

If the depth is greater than 2.5 ft, the two-point method should be used. In this method, two velocity measurements are taken at depths that are equal to 20% and 80% of the total depth when measured from the water surface. This also does not have to be calculated when using the USGS type top set wading rod. To set the 20% point, double the water depth measurement and then move the sliding rod so that the foot measurement on it lines up with the tenth of a foot indication on the scale. To set the 80% point, halve the water depth measurement and move the sliding rod so that the foot measurement on the sliding rod lines up with the tenth of a foot indication on the scale.

### 6.2.3 Taking Measurements

Starting from the left edge of the water (LEW) while facing downstream, read the tape measure at the water's edge. Take the first measurement of depth and velocity in the center of the first window. For example, for a window width of 2.0 ft with the LEW at a tag line distance of 3.0 ft, take the first measurement at 4.0 ft. Then, from this first measurement, add the window width of 2.0 ft. to find the next measurement location at 6.0 ft.

At each measured depth, record tagline distance, depth, and velocity on the Flow Field Sheet.

See Sections 6.1.1 and 6.1.2 for specific procedure on individual flow meters.

#### *6.2.3.1 Calculating Total Flow*

The flow in each section equals the average water velocity multiplied by the cross-sectional area of each window. To determine the cross-sectional area of a window, measure the depth in the middle of the window and multiply it by the window width.

For the Model 921 Revolution Counter on a Price meter, look up the velocity on the rating table supplied with the meter. Locate the column corresponding to the number of revolutions, and then locate the row corresponding to the number of seconds. Record these water velocities (f/s). This step is not required if the CMD 9000 computerized sensor or Marsh-McBirney is used.

For each window, calculate the window width based on the meter locations preceding and following. That is, if you placed the meter at 3.0, 5.0, and 8.0 ft (along the tag line), then the window width at the "5-foot" section window would be 2.5 ft, calculated as follows:

$$\textit{Window width} \quad = \quad \left(\frac{8-5}{2}+\frac{5-3}{2}\right) \quad = \quad \left(\frac{8-3}{2}\right) \quad = \quad 2.5 \ \textit{feet}$$

Calculate the flow through each window by multiplying the velocity by the window width by the window depth (velocity by cross-sectional area). **Table 6-1** illustrates these data and results (NOTE: The non-bolded items are directly measured in the field; the bolded items are calculated).

**Table 6-1 Example of velocity by cross-sectional area calculations for determining flow**

| Tag line location (ft) | Window width (ft) | Depth (ft) | Revolu-tions | Time (seconds) | Velocity (f/s) | Flow (cfs) | Comments |
|---|---|---|---|---|---|---|---|
| 3.0 | 1.0 | 0.0 | NA | NA | 0.000 | 0.00 | LEW |
| 5.0 | **2.50** | 0.7 | 15 | 44 | 0.361 | **0.63** | |
| 8.0 | **2.10** | 0.8 | 25 | 43 | 0.596 | **1.00** | Rock |
| 9.2 | **1.35** | 1.3 | 40 | 43 | 0.937 | **1.64** | |
| 10.7 | **1.05** | 1.9 | 60 | 41 | 1.460 | **2.91** | |
| 11.3 | **0.85** | 2.1 | 150 | 40 | 3.690 | **6.59** | Thalweg |
| 12.4 | **1.35** | 1.4 | 80 | 45 | 1.760 | **3.33** | |
| 14.0 | **1.80** | 1.0 | 20 | 47 | 0.444 | **0.80** | |
| 16.0 | **1.00** | 0.2 | NA | NA | 0.200 | **0.40** | REW |
| **13.0 ft** | | | | | | **17.3 cfs** | **TOTALS** |

Notes:
f/s = Feet per second cfs = Cubic feet per second
LEW=Left Edge of Water REW = Right Edge of Water
NA = not applicable

A MS Excel spreadsheet has been developed to calculate flow based on tag line, depth, and velocity field measurements. Upon return from the field, use the file named Flow Calculator located in SWQB Public to determine flow. Enter these data on the field form and provide to the Project Lead for entry into the SWQB water quality database.

## 6.3 Additional Flow Calculation Methods

Velocity meters are the most common tool used by the SWQB to measure stream velocity in order to determine flow. At times, however, these meters may not be available or stream conditions may require additional or alternate methods. Also, in some instances there may be a staff gauge to measure depth and determine flow based on previously measured channel geometry. The following methods are approved by the SWQB for additional flow measurements and described below:

- Timed-fill
- Surface floats
- Tracers
- Manning Equation
- ISCO 4230 Bubbler Flow Meter
- Rating Curve

### 6.3.1 Timed-Fill

At low flows, when velocity meters are unable to function due to surface water depths <0.15 ft., the timed-fill method may be utilized. Use a stopwatch to measure the time it takes to fill a 5 gallon bucket. 5 gallons per unit time may be converted to cfs (5 g = 0.6684 $ft^3$, so

0.6684/elapsed time (s) = cfs). Obviously, the entire flow of the stream must be collected in the bucket (e.g., below a waterfall or weir).

### 6.3.2 Surface Floats

A very rough method for preliminary estimates of time-of-water travel consists of dropping a neutrally buoyant object (such as an orange or a rubber ball) in the current of the stream reach under observation and noting the time required for it to travel some known distance. Surface float velocity estimates may be too inaccurate for use in interpretation of data or final reporting, but they can be useful in preliminary planning of studies and in conducting subsequent, more precise, measurements.

Procedures are conducted as follows:

1. Measure and mark two points, one upstream and one downstream, at least two channel widths apart.
2. Two observers are best, one upstream and one downstream. The upstream observer tosses the float into the channel above the marker and calls out when it crosses the upstream point, at which point the downstream observer starts a stopwatch.
3. The downstream observer sights across the stream at the lower point. When the float passes the downstream point, the downstream observer stops the stopwatch and records the elapsed time.
4. Repeat the procedure 5 to 10 times. Each toss of the float should be a different distance from the bank to get a rough average of velocities across the channel. Average the values to get the mean surface velocity and then multiply it by a velocity adjustment coefficient of 0.85 (to account for friction) to calculate the mean velocity of the entire cross section.
5. Using the previously measured cross-sectional area, multiply velocity times area to find flow (Q=VA). Record it on a data sheet with date, time, etc. (Harrelson 1994).

### 6.3.3 Manning's Equation

Velocity through an open channel can be predicted based on the physical characteristics of the channel. These characteristics include the slope, streambed roughness, and channel shape. Several empirical methods are available to use these characteristics to estimate stream velocity at a variety of stages. This relationship can be used to develop a stage-flow rating curve.

To estimate flow using the Manning equation, cross-sectional and bed slope data are used to obtain the hydraulic radius and water surface slope. A roughness coefficient is estimated from tables readily available in literature (Brooks 1997). Flow can then be calculated by multiplying velocity by cross-sectional area.

$$V=(1.49/n)R_h^{2/3}s^{1/2}$$

where $R_h=A/WP$

where: V = average velocity in the stream cross section (ft/sec); $R_h$ = hydraulic radius (ft); A = cross-sectional area of flow ($ft^2$); WP = wetted perimeter (ft); s = energy slope as approximated by the water surface slope (ft/ft); n = roughness coefficient.

Wetted perimeter and cross-sectional area may be correlated to a stage (gauge height) and flow may be estimated in the field using a staff gauge (Brooks, 1997).

### 6.3.4 ISCO

See Chapter 12 Stormwater Sampling.

### 6.3.5 Rating Curve

A flow rating, or rating curve, is the relationship of flow to stage (or gauge height). It is constructed by plotting successive measurements of flow and gauge height on a graph. This relationship is then used to convert records of gauge height into flow rates. Due to changing controls, curves must be checked periodically to ensure that the relationship between flow and gauge height has remained constant. Scouring of the stream bed or deposition of sediment in the stream can cause the rating curve to change so that the same recorded gauge height produces a different flow. A constant relationship between water level and flow rate at a given site can be assured by constructing a flow control device of known dimensions in the stream, such as a sharp crested weir or flume (Chow, 1988).

The rating analysis is a process where data from a series of flow measurements are plotted (typically in MS Excel), a curve is defined by the measurements plotted, and a table is produced from the curve. “Simple” ratings–the most common design–involve only the relation of flow to stage at one location, rather than “complex” curves, which require a stage-flow relation curve plus one or more supplementary curves based on stream morphology (USGS 1969).

The quality of the stage-flow relation determines the quality of computed stream flow data. Hydraulic theory helps in determining the general form of the rating curve. In a long straight channel, where channel friction control operates, a curve is described by the equation:

$Q=C(h+a)^N$
where

Q = flow
C and N = constants
h = stage
a = stage at which flow is zero

Values of N for different cross section shapes are:

Rectangular: N = 1.67 (assuming width >20 × depth)
Parabolic: N=2.17 (assuming width >20 × depth)
Triangular: N=2.67

Because natural channels are often approximately parabolic in cross section, a value of about 2 for the exponent N is appropriate where there is channel friction control. Where there is a series of natural controls for different ranges of stage, different values of C, a, and N may apply for each range of stage. While rating curves are developed in correlation to flow and stage, this equation is useful to help with extrapolation of ratings, helping to identify the causes of changes in the slope of the rating curve, identifying when scour or aggradation has occurred, and checking for mistakes.

There is no standard number of measurements necessary for rating curve development. It is important to obtain data from low, moderate, and high flow conditions to develop an accurate curve. Increased vertical variability in the channel will naturally require more measurements to explain flow variability than in a trapezoidal channel. The ISO requires that 95% of flows measured must fit the curve to an accuracy of ± 8% of the rated value and a frequency specified by reference to flood event frequency, bed stability, and historical evidence.

A rating curve may be used in conjunction with a stream flow measurement station (commonly called a gauging station). Water surface elevations are recorded at regular time intervals. These are converted to flow using the rating curve in order to describe flow conditions over a length of time. A variety of hardware is required to set up a continuously-recording gauging station. ISCO (1983) gives standard procedures for gauging station design (Maidment, 1992).

## 6.4 Safety Concerns

### 6.4.1 "Rule of 10"

Wading across a stream bed can be very dangerous depending on flow and substrate conditions. Do not attempt to wade into a stream if the depth (in ft) multiplied by the velocity (in ft/s) equals or exceeds 10 square ft per second ($ft^2/s$). For example, a stream that is 2 ft deep, and has velocities of 5 ft/s or more, should be considered too dangerous to wade (NMED 2005, appendix C). If you unknowingly start to take measurements and discover part of the way across that you are/will violate the rule of ten, return to the nearest bank and note "too fast/deep to measure" on the field form.

### 6.4.2 Streambed Concerns and Obstacles

Some channels have quicksand-like areas, deep holes, sharp rocks, excessive fallen logs, etc., that can lead to foot entrapment, injury, or falls. The wading rod (without the current meter attached) can be gently used for stabilization and to probe the streambed when conditions are uncertain. Staff should use best professional judgment to assess risks involved with data collection.

**FORMS**

Flow Field Sheet
Flow Calculator

# 7.0 CHEMICAL SAMPLING

The results of a water sampling may have legal and regulatory ramifications. It is incumbent on staff to perform their tasks in as rigorous a manner as possible. Equipment must be in good working order and properly calibrated. Sample containers must be free from contaminants and appropriate to the sample suite collected in them. Sampling techniques must assure that samples are not contaminated during collection and processing.

## 7.1 Equipment

### 7.1.1 Stream Sampling Equipment

The following categorized list includes equipment necessary for typical surface water sampling in lotic waters:

*General*
Access authorization documents
Camera
Cell phone
Coolers
Extra batteries (AA, C, D)
First aid kit
Flashlight
GPS unit
Maps
NMED ID/business cards
Shovel
Toolbox
Vehicle Kit

*Sample Collection*
Calculator
Cartridge filters
Acid-washed, individually-bagged silicon tubing
Clipboard
Callibration standards fro sonde
Cubitainers (liters, gallons) or other sample containers
D.O. calibration values table
D.O. probe repair kit
Field forms
Geo pump, with appropriate power cords
Kimwipes

*Sample Collection (continued)*
Sulfuric acid
Trash bags
Turbidity standards
Wash bottles
Water carboy (w/DI water)

*Flow*
Bank pins
Flow clipboard
Flow forms
Flow meter
Hammer
Measuring tape
Wading rod

*E. Coli collection*
AC/DC Inverter
Black Light
Camp table
DC Cable
DC/AC Inverter
DC/DC Connector
Deep Cycle Battery
IDEXX Bottles
Incubator
Quanti-tray
Reagent (18-hr and 24-hr)
Sealer with Rubber Tray & Cord

NaOH (for cyanide)
Nitric acid
Organics sample bottles (vials/bottles)
Pencils
Pipetter tips
Pipetters (and repair kit)
Extra RID Stickers
Sharpies
Site list/directions
SLD submittal forms
Sonde maintenance kit
Sonde, data logger, probe guard, cable, manual, log book, calibration forms

*Personal/Optional*
Binoculars
Boots/waders/sandals
Field guides
Field notebook
Hat/sun protection
Neoprene gloves
Rain gear
Sunglasses/polarized glasses

7.1.2 Lake Sampling Equipment

See Chapter 14 Lakes

## 7.2 Cleaning of Surface Water Sampling Equipment

Equipment for water samples should be as clean as practicable before contacting the sample. The goal of equipment cleaning is to minimize the chance that equipment is a source of foreign substances that could affect the ambient concentrations or chemistry of target analytes in samples. Sampling equipment requiring cleaning consists primarily of sample tubing, equal-width-increment sampling equipment, processing equipment, such as churn splitters and automated sampling devices (e.g., ISCO® Automatic sampler). Disposable sample collection containers do not require cleaning or rinsing prior to use.

Sample collection and sample processing equipment should be cleaned before use to remove manufacturing residues from new equipment, dust and other foreign substances from equipment that has been in storage, and substances adhering to equipment from previous sampling.

In the laboratory, after the sampling run is completed, sampling equipment (e.g., coolers, buckets, cups, nets, flow equipment, etc.) that came in contact with surface waters must be hosed off outdoors. Immerse all porous material (e.g., nets) in a 10% bleach solution for 10 minutes. Rinse thoroughly with tapwater. Dry and put coolers away. At the end of the field season, scrub all coolers clean.

7.2.1 Cleaning Procedures

A. Field

1. In the field, it is adequate to thoroughly rinse equipment with DIW as soon as possible after use. At each new sampling station, thoroughly rinse equipment with sample water before collecting a sample.

B. Preparation at the laboratory

1. Prepare an area for cleaning and drying cleaning supplies, sample-collection, and sample-processing equipment.

    a. Gather cleaning supplies, equipment to be cleaned, and plastic bags or other material with which to wrap cleaned equipment. See **Table 7.1** for cleaning supplies needed.

    b. Place clean paper over the work surface.

    c. Put on disposable, powderless gloves,

    d. For most situations prepare a 0.1-0.2% solution of Liquinox. Use a higher concentration for dirtier equipment.

    e. Prepare a 10% v/v dilution of American Chemical Society (ACS) trace-element-grade hydrochloric acid (HCl) in de-ionized water (DIW). **SAFETY NOTE: Always add acid to water, never add water to acid.**

2. Clean the items used to clean the equipment.

    a. Fill wash basins with the Liquinox solution. Put wash bottles, scrub brushes, and other small items used for cleaning into a wash basin. **Soak for 30 minutes.**

    b. Scrub interior and exterior sides of basins and standpipes with soft scrub brushes. Fill wash bottles with a soapy solution and shake vigorously.

    c. Rinse all items thoroughly with tap water. to remove detergent residue. No detergent bubbles should appear when fresh tap water is agitated in the basin, standpipe, or wash bottle.

3. Disassemble dirty sampling equipment.

B. Detergent wash and tap water rinse

    a. Place small equipment parts into wash basin labeled for detergent and fill with Liquinox solution. Soak equipment for 30 minutes.

    b. Scrub exterior and interior of equipment surfaces to the extent possible using a firm sponge or soft brush to remove any adhering material such as oil and grease, sediment, algae, or chemical deposits. Pay particular attention to grooves and crevices, O-rings, nozzles, and other spaces where inorganic or organic materials might be trapped.

    c. Rinse equipment thoroughly with warm tap water.

C. Check equipment

1. Nonmetal equipment or equipment with removable metal parts: remove any metal parts and go to Step D.
2. Metal equipment components: go to Step E, DIW rinse.

D. Acid soak/rinse

For equipment constructed primarily of glass, fluorocarbon polymer, or other plastic, soak in a 10% HCl solution, or, using a wash bottle, rinse surfaces that contact sample water with 10% HCl.

1. Place nonmetal equipment and tubing into the wash basin labeled “acid solution.”

2. Fill basin with dilute HCl solution. Soak equipment and tubing for 30 minutes. Swirl the acid solution several times during the 30-minute soak.

3. Carefully pour the used acid solution into a black stone sink

E. DIW rinse — Laboratory or Field

1. Place equipment into the cleaned wash basin.

2. Rinse exterior and interior of each piece of equipment thoroughly. For pieces of equipment too large to submerse in the wash basin, rinse surfaces that contact sample water by spraying with DIW from a wash bottle.

3. Place on a clean surface to dry.

F. Storage of clean equipment

Place dry, clean equipment inside plastic bags. For small equipment or parts, use sealable plastic bags.

7.2.2 Sample Tubing Preparation, Cleaning and Storage of Filtration Tubing

Remove new tubing from the factory packaging, cut to appropriate length and placed in 10% “Trace Metal” grade (or equivalent) HCl/DIW water solution for not less than 1 hr and not more than 2 hrs*. At the end of the acid bath period, remove the tubing from the acid bath, rinse with DIW inside and out, coil and place the tubing in an unused zip-lock type, 1 gal. capacity plastic bag. **It is imperative that the person handling the acid bath basket and the tubing wear gloves at this stage, and touch nothing but the interior of the basket, tubing or the interior of the bag.** An assistant is required to open and seal the bag.

On returning from the field, soak the used tubing first in a hot water/Liquinox solution for a minimum of 15 minutes then rinse with hot, flowing tap water before placing the tubing in the acid bath and processed as above.

*The tubing is not acid resistant; exposure to HCl for extended periods will cause it to depolymerize and fail.

### 7.2.3 Multi-probe Sondes

In a manner appropriate to each device, decontaminate all exposed surfaces, including any containers. 70% isopropyl alcohol or a solution of ¼ cup bleach to 1 gallon tap water is suitable for most disinfecting (YSI, Inc.).

**Table 7-1 Supplies for cleaning equipment used for water-sampling activities (modified from USGS 1998)**

| Item | Description and Comments |
|---|---|
| Acid solution(a) | Hydrochloric: ACS trace-element grade (10 % by volume in DIW) |
| Aluminum foil | Organics only: Heavy duty, for work surfaces and equipment |
| Bags, plastic or fluorocarbon polymer | Sealable bags with uncolored closure strips, various sizes. Recyclable trash bags are recommended for large equipment storage. |
| Brushes and sponges | uncolored; plastic components needed for inorganic work. |
| Distilled water/DIW | Maximum specific electrical conductance, 1 mS/cm |
| Laboratory-produced organic-grade DIW | Usable only as a cleaning solution and only as specified in the text. Must not be used to substitute for PBW or VBW. |
| Detergent | Nonphosphate laboratory soap (for example, Liquinol®). |
| Gloves, disposable | Powderless, noncolored vinyl, latex, or nitrile (latex or nitrile for use with methanol), assorted sizes. |
| Safety equipment and guidelines | For example, MSDS, safety glasses, chemical spill kit, apron, emergency phone numbers. |
| Tap water | If quality is questionable, substitute DIW. Tap water is more effective for initial and rapid removal of detergent residue. |
| Wash bottles | Labeled to indicate contents (for example, ACID, DIW, TAP). Fluorocarbon polymer needed for methanol, PBW, VBW, and IBW. |

Notes:
(a) Hydrochloric acid is required if analyzing for nitrogen species; otherwise, nitric acid is acceptable.
(b) DIW = Deionized Water
(c) mS/cm = MilliSiemens per centimeter
(d) MSDS = Material safety data sheet

## 7.3 Routine Water Chemistry Sampling

For Lake sampling protocol, see Chapter 13 Lakes

It is necessary to notify SLD prior to sample collection if either bacterial samples will be processed there, or if organic samples will be collected. In the case of organics samples, arrangements should be made by phone at least two weeks in advance to reserve room for anticipated samples in the que for extractions to ensure that extractions will be performed in time to meet holding time requirements. For bacteria, reservations should also be made by phone as far as possible in advance to determine if the lab can accommodate the anticipated sample volume, and to prepare culture media if necessary. If *E.coli* samples will be processed using SWQB equipment, reservations for necessary equipment should be made in the Checkout Folder in the SWQB public folder on Magneto.

Water samples should be collected before other work is done at the site. Other work, (e.g., sediment sample collection, flow measurement, or biological/habitat) will disturb the stream, making it difficult to collect representative samples. If it is imperative that other work be done simultaneously with, or prior to, the collection of water samples, the water samples should be collected at a sufficient distance upstream to prevent interference or contamination.

The most common causes of sample contamination include poor sample handling techniques, atmospheric input, inadequately cleaned equipment, incorrect sample preservation, and use of equipment constructed of materials inappropriate for the targeted analytes. Contamination of samples from these sources can be prevented or minimized by adhering to good field practices (Table 7.2). Collection of equipment blanks and field blanks is necessary to help identify potential sources of sample contamination. The same equipment that is used to collect and/or process environmental samples is to be used to collect and/or process blank samples.

**Table 7-2 Good field practices for collection of water quality samples**

**(modified from USGS 1992)**

- Be aware of and record potential sources of contamination at each field site.
- Avoid hand contact with contaminating surfaces (such as equipment, coins, food).
- Use equipment constructed of materials that are inert with respect to analytes to be collected.
- Use only equipment that has been cleaned thoroughly.
- Field rinse equipment, but only as directed. Some equipment for some analytes are not to be field rinsed, e.g., organics samples.
- Use correct sample-handling procedures:
    - Minimize the number of sample-handling steps.
    - Obtain training for and practice field techniques under supervision before collecting water samples.
- Collect a sufficient number of appropriate types of quality-control samples.

### 7.3.1 Collecting a Water Sample

Unless otherwise indicated, samples are collected in clean, 1L polyethylene containers (Cubitainer). Samples are filtered for dissolved concentrations or not filtered for total concentrations. Filters, if used, are rinsed with sample water.

In most streams, near-surface water is representative of the water mass. In these cases, a water sample may be collected by immersing the container beneath the surface of flowing water.

A bucket or disposable bailer and nylon rope may be used to collect water if sampling containers cannot be dipped directly in the water. Care must be taken to avoid contaminating the sample with debris from the rope and bridge (or other sampling platform). The first bucketful of water is

used to rinse the bucket and the sampler's hands. Samples are collected from the second and third buckets of water. A fourth bucket of water is used to measure temperature, pH, DO and SC.

Every effort should be made to take sonde readings from flowing water. Extra long cables should be used to get readings from the thalweg whenever possible. When all else fails, sonde readings can be taken from a bucket, but a note to this effect should be made on the field sheet. A metal bucket must be used when collecting organics samples and a plastic bucket when collecting metals samples. The probe cluster on sondes has metallic components, which should not touch the sample water prior to collecting samples for metals.

All samples for dissolved constituent analyses are filtered using a 0.45-micron pore-size disposable in-line filter cartridge (Geotech dispos-a-filter®). Use filters only once to prevent contamination, except when preparing an equipment blank immediately before filtering an environmental sample.

### 7.3.2 Collecting for Specific Parameters

A summary of the containers, sample preservatives, and holding times for parameters is included in Table 7.4 (from 40 Code of Federal Regulations [CFR] Part 136.3). Rinsing new, plastic sampling containers with distilled or sample water before collecting the actual sample is unnecessary and it is imperative not to rinse pre-acidified containers. Sample containers may be preacidified at the laboratory. Fill pre-acidified containers by dipping a clean bucket or other container in flowing water and decanting water into the containers.

**Table 7-3 SWQB environmental sample handling and holding times**

| Sample Type | Sample Container | Preservation (1) | Maximum Holding Time |
|---|---|---|---|
| **Inorganic Tests:** | | | |
| Ions – full suite (2) | 1-liter polyethylene cubitainer | Cool, 4°C | 7 days TSS – 14 days other |
| Ions – SWQB suite (3) | 1-liter polyethylene cubitainer | Cool, 4°C | 7 days TSS – 14 days other |
| TDS and TSS only | 1-liter polyethylene cubitainer | Cool, 4°C | 7 days TSS – 14 days other |
| Total Nutrients (4) | 1-liter polyethylene cubitainer | 2.0 mL $H_2SO_4$, Cool, 4°C | 28 days |
| Dissolved Nutrients (5) | 1-liter polyethylene cubitainer | Filtered, 2.0 mL $H_2SO_4$, Cool, 4°C | 28 days |
| Cyanide (6) | 1-liter polyethylene cubitainer | 5-7 pellets NaOH, 0.6g ascorbic acid if chlorine present, Cool, 4°C | 14 days |
| **Metals:** | | | |
| Total Metals (7) | 1-liter polyethylene cubitainer | 5.0 mL $HNO_3$, Cool, 4°C | 28 days mercury – 6 months other |
| Dissolved Metals (8) | 1-liter polyethylene cubitainer | Filtered, 5.0 mL $HNO_3$, Cool, 4°C | 28 days mercury – 6 months other |
| **Microbiological Tests:** | | | |
| Coliform: total, fecal, and *E. coli* (9) | 100-mL polystyrene vessel (IDEXX) 100-mL LDPE vessel (lab) | 0.0008% $Na_2S_2O_3$, Cool, 4°C | 6 hours |
| **Organic Tests (10):** | | | |
| Method 8270 – Base/Neutral Acid Extractables (11) | Two 1-liter amber glass bottles (lab) | Cool, 4°C | 7 days |

**Table 7-3 SWQB environmental sample handling and holding times**

| Sample Type | Sample Container | Preservation [1] | Maximum Holding Time |
|---|---|---|---|
| Method 8260 – Volatile Organic Compounds [11] | Two 40-mL glass vials (lab) | 5-10 drops HCl (HCl provided by lab), Cool, 4°C | 14 days |
| **Radiological Tests:** | | | |
| Radionuclides [12] | Two 1-gallon polyethylene cubitainers | 17.5 mL $HNO_3$, Cool, 4°C | 6 months |
| **Biological Tests:** | | | |
| Ambient Toxicity (acute and chronic) in water and sediment | 1-gallon polyethylene cubitainer (water) and/or Two 1-quart, wide-mouth glass containers (sediment) | Cool, 4°C | 36 hours |
| Chlorophyll *a* (streams/rivers) | 1-liter polyethylene cubitainer | Place in cooler with dry ice and keep frozen | 3 months |
| Chlorophyll *a*, *b*, *c*, and phaeophytin (lakes) | Filters placed in 60-mL light-proof septum vials | 35 mL of 90% acetone mixed with 4 drops of saturated aqueous magnesium chloride | 48 hours |
| Phytoplankton (lakes) | 1-liter polyethylene cubitainer | 10-25 mL Acid Lugol's Solution, Cool, 4°C | indefinitely |
| Diatoms (lakes) | glass or plastic vials, <100 mL | Cool, 4°C | indefinitely |
| Periphyton community composition (streams/rivers) | 50-mL plastic vial | 2-4 mL of 10% formalin | indefinitely |
| Macroinvertebrates | glass or polypropylene jar(s), size varies | fill jar with 95% ethanol; remove air bubbles | not applicable |
| Fish | plastic bags | 10% formalin to cover; remove air bubbles | not applicable |

Notes:

1 Given the field conditions, preserve samples as soon as reasonably possible, preferably immediately after sample collection. Contract labs may provide pre-preserved sample containers.
2 Ions (full suite) include calcium, magnesium, potassium, sodium, hardness, alkalinity, bicarbonate, carbonate, sulfate, chloride, TDS, and TSS.
3 Ions (SWQB suite) include TDS, TSS, hardness, fluoride, chloride, and sulfate.
4 Total Nutrients include nitrate + nitrite, ammonia, total Kjeldahl nitrogen, and total phosphorus.
5 Dissolved nutrients include nitrate + nitrite, ammonia, orthophosphate, and dissolved phosphorus.
6 Maximum holding time is 24 hours when sulfide is present. Optionally all samples may be tested with lead acetate paper before pH adjustments in order to determine if sulfide is present. If sulfide is present, it can be removed by the addition of cadmium nitrate powder until a negative spot test is obtained. The sample is filtered and then NaOH is added to pH 12.
7 Total metals include mercury and selenium at a minimum.
8 Dissolved metals include aluminum, antimony, arsenic, barium, beryllium, boron, cadmium, chromium, cobalt, copper, lead, mercury, molybdenum, nickel, selenium, silver, thallium, uranium, vanadium, and zinc.
9 $Na_2S_2O_3$ is included in containers provided by IDEXX.
10 Various other organic analyses are available upon request. Refer to the SLD Organic Chemistry section (505 841-2571) or other contract labs for sample container, preservation, and holding time information.
11 Refer to 40CFR136 for the list of parameters analyzed using methods 8270 and 8260.
12 Radionuclides generally include gross alpha/beta and Ra-226 + Ra-228.

#### *7.3.2.1 Major Ions*

Samples collected for major cations and anions are not filtered or preserved, and are stored and shipped on ice at a temperature not to exceed 4°C.

#### *7.3.2.2 Total Suspended and Total Dissolved Solids (TSS/TDS)*

Samples collected for TSS and TDS are not preserved, and are stored and shipped on ice at a temperature not to exceed 4°C.

*7.3.2.3 Nutrients*

Samples collected for ammonia, nitrate plus nitrite, Total Kjeldahl Nitrogen (TKN), and/or phosphorus, are preserved with 2 milliliters (mL) sulfuric acid ($H_2SO_4$) per L, and are stored and shipped on ice at a temperature not to exceed 4°C. Orthophosphate samples are filtered immediately and not preserved, and are stored and shipped on ice at a temperature not to exceed 4°C.

*7.3.2.4 Metals*

Select equipment with components made of fluorocarbon polymer or other relatively inert and uncolored plastics or glass if components will directly contact samples to be analyzed for inorganic constituents. **Do not use metal or rubber components for trace-element sampling.**

Because total metals samples are analyzed only for mercury and selenium, and these metals are not included in the dissolved metals suite, it is not necessary to obtain both total and dissolved metals samples from a one gallon Cubitainer. Collect total metals samples directly from flowing water. Unless using pre-acidified sampling containers, filter the dissolved metals sample from a sample collected in the total metals container. Refill the total metals container for the total metals sample. When using pre-acidified sampling containers, fill a 1L Cubitainer from flowing water and filter into the preacidified dissolved metals sampling container. Samples for trace-metal analyses are preserved with five mL/L of ultratrace metal grade nitric acid. Both are stored and shipped at 4°C.

*7.3.2.5 Radionuclides*

Water samples for determinations of radiochemical parameters (e.g. gross α, gross β, radium-226, radium-228 and gamma) are collected in two, one-gallon Cubitainers. It is not necessary to rinse containers before collecting the sample, and rinsing must be avoided with preacidified containers. These samples are preserved with sufficient nitric acid to adjust the pH to below 2. Typically, 7-10 ml is adequate. Check pH using pH paper on an aliquot of the acidified sample. Preserved samples are stored and shipped at 4°C.

*7.3.2.6 Cyanide*

When collecting samples for cyanide analysis, field personnel should estimate the potential for presence of chlorine in the water being sampled. Oxidizing agents such as chlorine will decompose most cyanides. If chlorine is suspected to be present it must be removed. Collection points from open waters in streams which are NOT immediately downstream from outfalls present little chance that chlorine is present. **The decision to test for chlorine and to omit ascorbic acid is made based on the circumstances at the time of collection.**

- Cyanide Sample Treatment in Presence of Chlorine

  Test a drop of the sample with potassium iodide (KI)-starch test paper. A blue color indicates the need for treatment. Add ascorbic acid, a few crystals at a time, until a drop of sample produces no color on the indicator paper. Then add an additional 0.06 grams (g) of ascorbic acid for each L of sample volume. An excessive amount of

ascorbic acid can result in interferences in samples, where ammonia is high or in brines (e.g., effluent, brine water discharges). Therefore, add only a few crystals (0.06 g) at a time, until the field test for chlorine is negative. Ascorbic acid crystals are non-toxic and can be carried to the collection site.

Sodium hydroxide (NaOH) is used to preserve samples for cyanide analysis. NaOH is caustic and will burn skin or clothing on contact.

Add ascorbic acid before NaOH. The initial pH of the sample is determined on an aliquot of the sample using pH test paper supplied by the SLD and noted. The sample must be preserved with sufficient NaOH to produce a pH of 12 or greater. Most samples of surface water will require about 0.5 g solid NaOH per liter, approximately five pellets. Handle the pellets very carefully with wooden or plastic utensils, or pour directly from the container. After collection and preservation, cyanide samples are stored and shipped at 4°C.

#### *7.3.2.7 Organic Chemicals*

Because organic compounds tend to concentrate on the surface of the sampling device or container, **the sampling device and sample container are ***not*** to be rinsed with native water before being filled.** Select equipment with fluorocarbon polymer, glass, or metal components if components will directly contact samples to be analyzed for organic compounds. **Do not use plastics other than fluorocarbon polymers.** Wear a clean pair of Nitrille gloves when sampling for organics.

Collect water samples for determinations of pesticides and other organic chemicals in analysis-specific containers provided by the SLD Organics section or contract lab. If the samples will be submitted to SLD, the sampler must consult the SLD Organics section prior to the sampling event in order to notify SLD of the types, number, and scheduling of samples that will be delivered, and also to obtain the proper sampling containers, labels, and preservatives. The containers, preservatives, and information to be contained on the request forms are subject to change, as are recommendations for optimal sample-collection techniques. Sample containers for volatile organics samples should be stored at 4°C after being obtained from the SLD and prior to sampling. After collection and proper preservation, store and ship all organics samples at 4°C.

#### *7.3.2.8 Volatile Organic Compounds*

The sample containers for volatiles are Volatile Organics Analysis (VOA) vials. Fill two 40 mL VOA vials to overflowing and cap without trapping any air bubbles. If possible, collect directly from flowing water, keeping the vial submersed during the entire collection process. To keep the vial full while reducing the chance for air bubbles, cap the vials under the water surface. Fill one vial at a time and preserve on ice. If benzene, toluene, ethylbenzene, or xylene (BTEX) is requested, add 2-4 drops of HCl to eliminate microbial degradation. VOA bottles may come pre-acidified from contract labs, in which case the sample must be collected with a clear glass container and decanted into the 40 ml vials. Keep the vials together in a small plastic whirl pack.

#### *7.3.2.9 Pesticides/ Herbicides*

The sample container for pesticides and herbicides is a new, 1L, clean, unused amber colored glass jar with a Teflon lined cap. Collect 1L of water for each of the three sample types (Organophosphorus Pesticides, Organochlorine Pesticides and Chlorinated Herbicides). **EACH**

**SAMPLE TYPE REQUIRES A SEPARATE JAR**. Minimize the air space in the top of the jar. Preserve immediately after collection by placing on ice in darkness.

*7.3.2.10 Semivolatile Organics*

The sample container for semivolatile organics should also be a new, 1L, glass, amber colored bottle with a Teflon lined cap. Fill two 1L jars to the top and place on ice in the dark. If in-stream chlorine residual is suspected, measure the chlorine residual using a separate water subsample. Free chlorine will oxidize organic compounds in the water sample even after it is collected. If chlorine residual is above a detectable level (i.e., the pink color is observed upon adding the reagents), immediately add 100 milligrams (mg) of sodium thiosulfate to the pesticides, herbicides, semivolatiles, and VOA samples; invert until the sodium thiosulfate is dissolved. Record the chlorine residual concentration on the fieldsheet. If chlorine residual is below detectable levels, no additional sample treatment is necessary.

## 7.4 Ambient Water Toxicity Testing

The USEPA Houston laboratory conducts aquatic toxicity tests of water as part of the EPA Region 6 Ambient Toxicity Monitoring Program. The USEPA Houston laboratory follows the standard laboratory test procedures for *Ceriodaphnia dubia* (water flea) and *Pimephales promelas* (fathead minnow) and statistical data analyses specified in *Short-term Method for Estimating the Chronic Toxicity of Effluents and Receiving Waters to Freshwater Organisms* (USEPA 2002a) and *Methods for Measuring the Acute Toxicity of Effluents and Receiving Waters to Freshwater and Marine Organisms* (USEPA, 2002b).

### 7.4.1 Sample Collection

Using the standard grab sample collection method, fill one one-gallon Cubitainer with water, put on ice, and cool to 4ºC. A quantity of fine sediment sufficient to supply 1L of sample is composited from sites at the sample station, thoroughly mixed, and a 1L aliquot is transferred to a clean, wide-mouth container for transport to the analytical facility within 24 hrs of collection. Label the containers with the date, time, and sample site location. SLD Reference ID stickers are not required for samples shipped to the USEPA Houston Laboratory. Before scheduling toxicity sample collection, notify the laboratory. **Sample collection must be coordinated with the laboratory to guarantee appropriate scheduling.**

## 7.5 Blanks

Blanks are collected at intervals specified in the SWQB QAPP during field sampling operations. They are designed to quantify background levels of chemical analytes present on sampling equipment, in containers, and that may be picked up by samples during transport, storage or processing. As analytical detection limits are reduced due to advances in methodology, blanks are increasingly important to insure that reported analytical results represent the state of water sample when it was collected. In general, blanks consist of the cleanest appropriate water, preserved, transported and analyzed in a manner identical to the corresponding environmental sample.

### 7.5.1 Ions Blanks

Fill a 1L Cubitainer (or other container, if submitting to a contract lab) with DIW directly from a carboy of DIW obtained from SLD. The Cubitainer should be filled in the field. Transport and store exactly like an ions sample.

### 7.5.2 Nutrients Blank (also referred to as a reagent blank)

Fill a 1L Cubitainer (or contract lab equivalent) with DIW directly from an SLD carboy in the field and acidify with 2.5 mL of sulfuric acid if the container is not preacidified at the same time other nutrient samples are collected and preserved. Transport and store exactly like a nutrients sample.

### 7.5.3 Total Metals Blank

Fill a 1L Cubitainer (or contract lab equivalent) with DIW directly from an SLD carboy in the field and acidify with 5 mL of nitric acid if the container is not preacidified at the same time other total metals samples are collected and preserved. Transport and store exactly like a total metals sample.

### 7.5.4 Dissolved Metals Blank (also referred to as an equipment blank)

Fill a 1L Cubitainer with DIW directly from an SLD carboy. Set up a GeoPump with clean tubing and a new filter. Rinse the tubing and filter with approximately 50 mL of DIW by running the pump for several seconds. Discard the rinse water. Pump the remainder of the DIW into a 1L Cubitainer (or contract lab equivalent) and acidify with 5 mL of nitric acid if the container is not preacidified at the same time other dissolved metals samples are collected and acidified. The tubing and filter may be subsequently used for a sample after rinsing with approximately 50 ml of sample water.

### 7.5.5 Organics Blanks

**For organics blanks, do not to use DIW from SLD carboys!** Obtain de-ionized, distilled water (DDIW) from the SLD organics section in new glass jars with Teflon lids.
Take the DDIW into the field and fill the appropriate container type at the same time environmental samples are collected. Transport and store exactly like the corresponding environmental samples. Discard any remaining DDIW after preparing blanks.

### 7.5.6 VOA Trip Blank

Trip blanks are required for VOA only. VOA trip blanks are samples prepared in the laboratory with DDIW, preserved as required. They are transported to the sampling site, handled like an environmental sample, and returned to the laboratory for analysis. At the sample collection site, trip blanks should be taken out of packaging and the cooler at the same time environmental samples are to expose them to identical conditions. Repackage, store and transport trip blanks exactly like a VOA sample.

### 7.5.7 E. coli Blanks

DIW, DDIW or sterile water are all adequate for preparing *E. coli* blanks. Take a container with the water into the field and prepare the blank at the same time, and in the same manner, that you collect a sample.

### 7.5.8 Other Types of Equipment Blanks

For an equipment blank, DIW or DDIW (depending on the type of blank- see above) is poured into or over a sampling device (e.g., Van Dorn, Kemmerer, churn splitter, etc.) or pumped through a sampling device. It is collected in the same type of container as the environmental sample, preserved in the same manner, and analyzed for the same parameters. The analysis of equipment blanks should yield values less than the reporting limit. When ambient values are very high, blank values must be less then 5 percent of the lowest value of the batch. If Field Equipment Blanks are consistently less than the reporting limit, a set of Field Equipment Blanks are submitted with every tenth sample. If less than 10 samples are collected in a month, submit one set of blanks per month.

## 7.6 Equal-Width-Increment (EWI) Sample Collection Method

If the stream is large and not well mixed due to inflows from large tributaries, ephemeral channels, waste water treatment plants (WWTPs), and/or agricultural return flows, depth and width integrated sampling techniques are utilized. The preferred sampling technique is the EWI method (USGS 1999). The below isokinetic sampling equipment and depth-integrated procedures are most commonly used to collect high quality representative samples from open channel flow for analysis of suspended-sediment concentration (FISP 2006). Whether to sample only suspended sediments using EWI methods or to sample additional parameters using EWI methods is at the discretion of the study lead based on the level of mixing in the channel and the objectives of the study.

### 7.6.1 Determination of Verticals

A measuring tape or tagline is strung from bank to bank. The stream width is determined. Between 10 and 20 verticals are established, depending on the extent to which the stream is not well mixed, by dividing the stream width by the desired number of verticals. For example, if the stream width is 160 feet and the required number of verticals is 10, the width (W) of each sampled increment would be 16 feet. The sample station within each width increment is located at the center of the increment (W/2), beginning 8 feet from the bank nearest the initial point of measurement in this example. If the width increment results in a fraction, the number is rounded to the nearest integer to determine the initial station (Figure 7.1) (from USGS 1999).

### 7.6.2 Determination of Transit Rate

The transit rate is the speed of lowering and raising the sampler in the sampling vertical. EWI methods require that all verticals be traversed using the transit rate established at the deepest and fastest vertical in the cross section. The descending and ascending transit rate are equal during the sampling traverse of each vertical, and are the same at all verticals, in order to collect a volume of water proportional to the flow in each vertical (Figure 7.2) (from USGS 1999). The

transit rate must not exceed 0.4 times the mean ambient velocity in the vertical while being sufficiently fast to keep from overfilling the sample bottle. Mean stream velocity (measured or estimated), deepest sampling depth, and transit rate diagrams are used to approximate the necessary transit rate based on the equipment and nozzle used (FISP 1999). See below sections for details regarding determination of transit rate depending on equipment utilized.

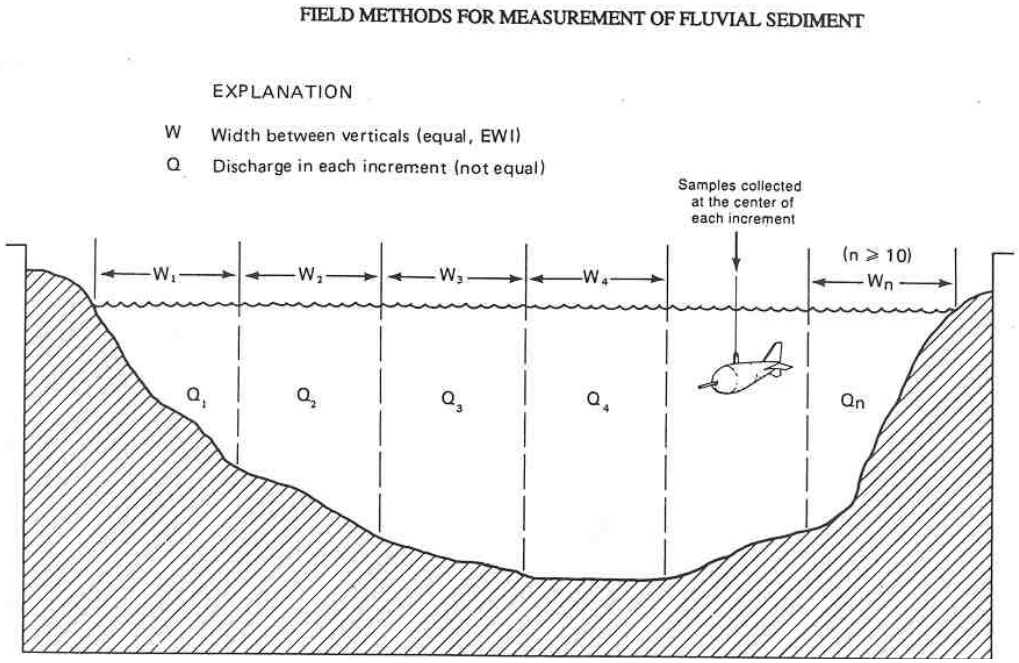



**Figure 7-1 Equal-width-increment verticals**

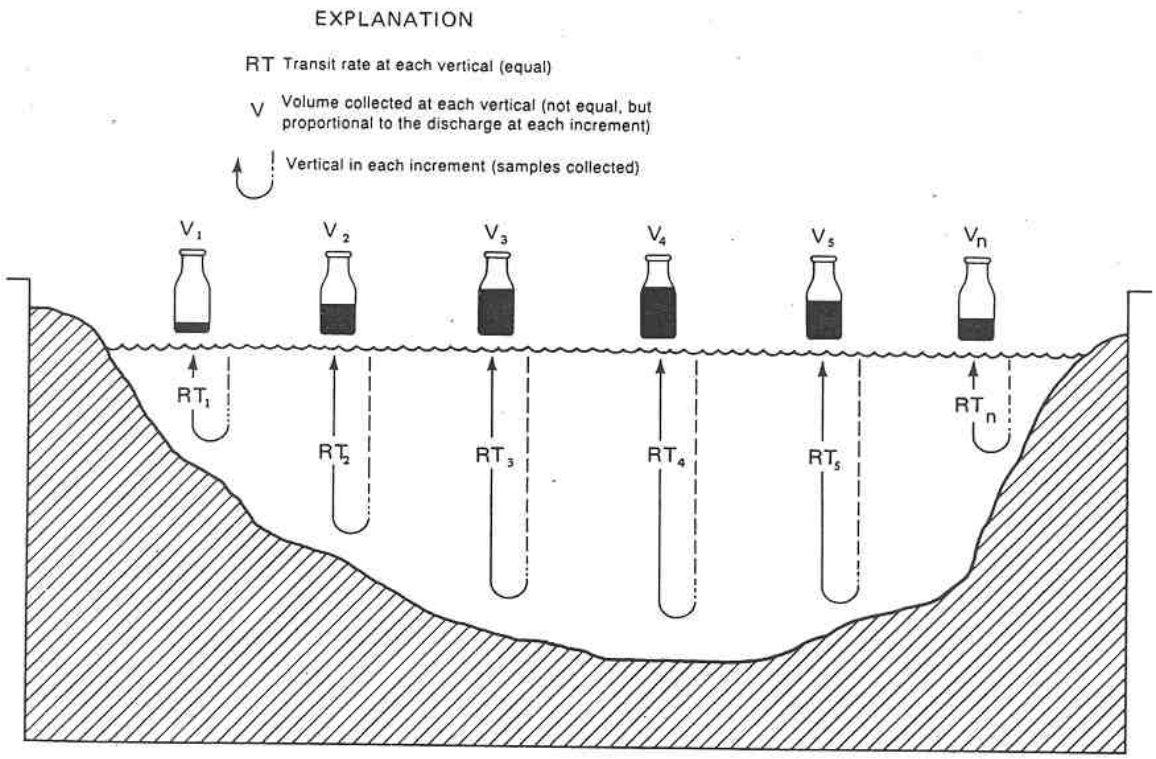



**Figure 7-2 Sample collection at wadeable sites using DH-48 sampler**

Sampling procedures for the DH-48 are detailed in FISP 2000c. A cross section displaying relatively stable hydrologic characteristics and uniform depths over a water range of discharge is selected. The sampling device utilized by SWQB is a hand-held US DH-48. It is designed to sample isokinetically, meaning that water and sediment enters the nozzle at the same velocity as the stream being sampled in order to collect a representative sample. A pint milk bottle is held in place and sealed against a rubber gasket by a hand operated, spring tensioned clamp at the rear of the sampler. A brass 1/4-inch internal diameter intake nozzle extends horizontally from the nose of the sampler body. A standard 1/2-in diameter wading rod is threaded into the top of the sampler body for suspending the sampler. The DH-48 can sample to within 3.5 inches of the stream bed and can be used in velocities ranging from 1 to 9 ft/sec (FISP 2000c). Specifications and sampling instructions for the alternative DH-81 handheld sampler can be found in FISP 1999.

Maximum safe wading depth is determined by multiplying the depth of the stream times the stream velocity. For maximum safety, a stream should not be waded when this calculated value is greater than ten.

Prior to sampling, the transit rate is determined using the procedure described in Table 7.1 utilizing transit rate diagram for the US DH-48 presented in Figure 7.3 (from FISP 2000c). The wading rod should be held vertically with the sampler and nozzle pointing upstream and as far away from the sampler as possible to avoid interference with the flow from the person taking the

sampler. In order to sample with a consistent transit rate, the person collecting the sample determines a reference point above the water surface at the deepest vertical to be sampled at which the sampler is started and stopped. This reference point is used to start and stop all verticals, allowing the same amount of time to elapse during the round trip traverse of the sampler regardless of the stream depth encountered in the vertical. The USGS recommends a reference point on the body of the person collecting the sample, such as his/her hip. Using the determined transit rate, the sampler lowers the DH-48 until it touches the stream bottom, then immediately reverses the direction to raise the sampler until it clears the water surface. To avoid overfilling, the person collecting the sample periodically checks the volume that has been collected and switches out the sample bottle when the volume is approximately 420 mL.

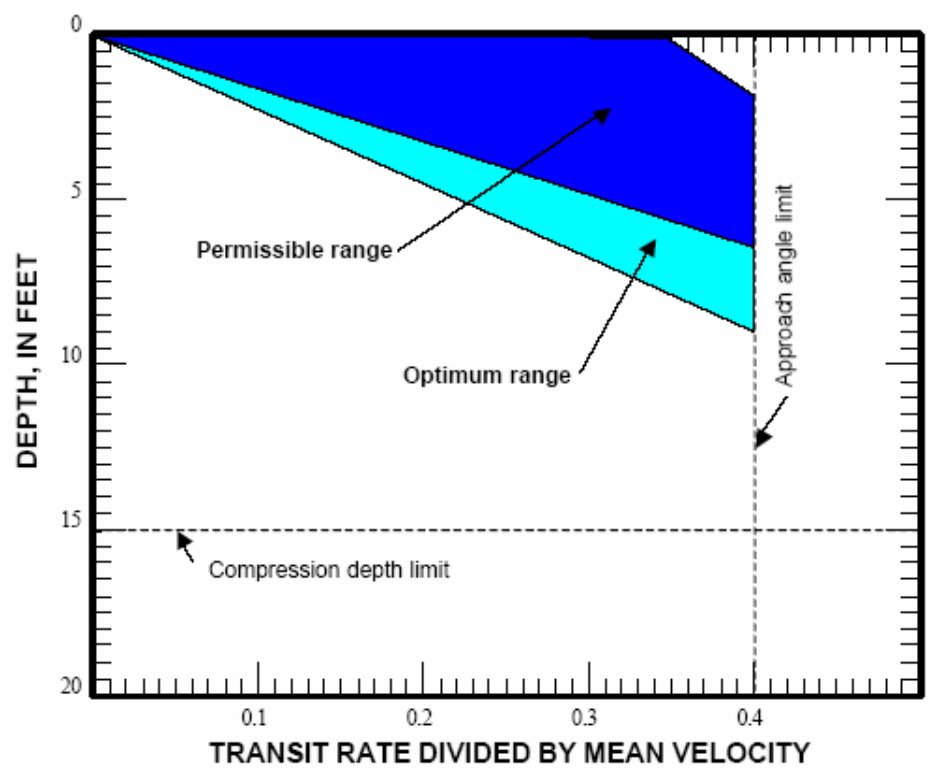
Compositing and/or multiple traverses of the cross section may be necessary. The person collecting the sample returns to the bank and empties the contents of the sampling device into a decontaminated churn or other clean sampling container depending on equipment availability, parameters to be analyzed, and necessary sample volume while agitating the sample to ensure particles are in suspension during the transfer. Before returning to the bank, the person collecting the sample makes notes of or marks his/her last collection vertical on the tagline with a clothespin or similar device. After emptying the sample bottle, the person collecting the sample returns to the marked location and resumes sampling. The procedure is repeated until the necessary volume has been collected and composited. The composited sample is then dispersed and filtered as necessary depending on the parameters to be analyzed. The agitator on the churn is slowly moved up and down while dispensing sample water from the spigot into the appropriate lab sample containers.

### Table 7.1 Determining transit rate using transit diagram

Using the appropriate transit diagram, draw a horizontal line from the maximum sampling depth on the Y-axis to the middle of the optimum range. Move vertically to intersect with the X-axis. Multiple this number on the X-axis times the mean stream velocity to determine the transit rate.

**EXAMPLE FOR DH-48:**

- Mean stream velocity = 4 ft/sec
- Maximum sampling depth = 2.5 feet
- Using transit diagram Figure 7.3 (from FISP 1999), 0.15 = corresponding x-axis value = " transit rate divided by mean velocity"
- 0.15 x 4 ft/sec = 0.6 ft/sec transit rate
- Therefore, a transit rate of ~0.6 ft/sec should be maintained at every sample vertical.



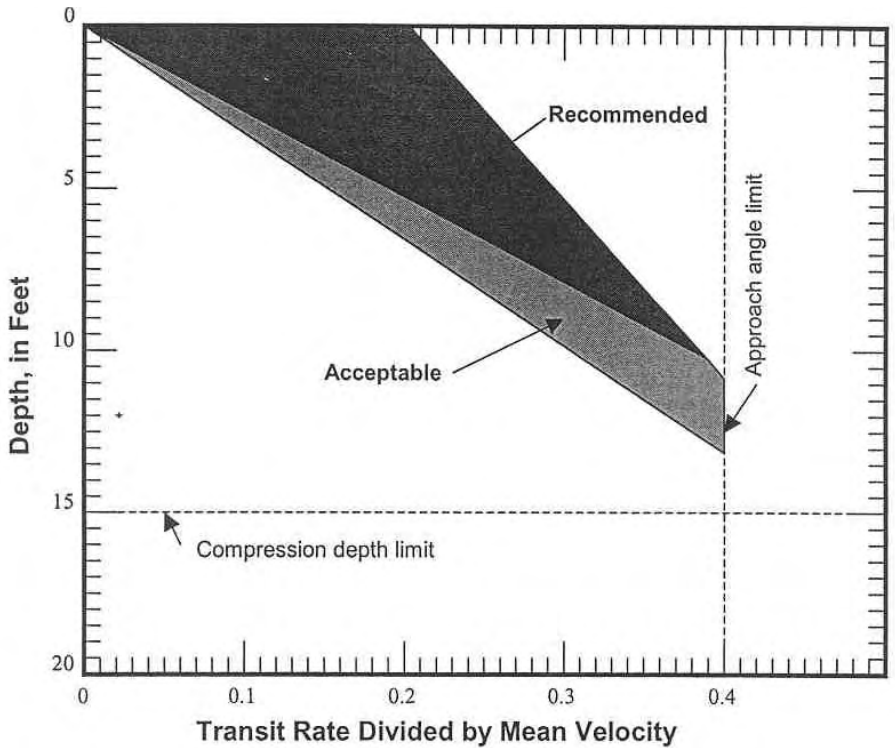

Note: The following volumes were used to produce this diagram: The total volume of the sampler container was 470 mL. The maximum recommended volume was 420 mL. The minimum optimum volume was 300 mL.

**Figure 7-3 Transit rate diagram for US DH-48 sampler**

### 7.6.3 Sample Collection at Non-Wadeable Sites using DH-95 Sampler

When sampling from a bridge or cableway, SWQB utilized a US DH-95 with a 5/16-inch nozzle (FISP 2000a). The maximum sampling depth for this configuration is 13.3 ft and sampler velocity range is from 2.2 ft/sec to 7.0 ft/sec (FISP 2000b). The sampling device is connected to a hanger bar. The hanger bar is connected to a suspension cable on an "A" reel supported by a bridgeboard. The nozzle is screwed hand tight into the cap. The O-ring is lifted in order to place a triple rinsed Teflon or polypropylene 1L bottle into the sampler cavity. The O-ring fits over the neck of the bottle to hold it in place. The bottle-cap configuration is rotated until the air vent hole in the cap is vertical. The nozzle intake and air vent hole are checked for any obstructions. The person collecting the sample has a lid and extra sample bottle available. The transit rate is determined using the procedure described in Table 7.1 and the appropriate transit rate diagram (Figure 7.4) (from FISP 2000b). The sampler is lowered to immediately above the water surface without submerging the nozzle. Using the transit rate determined by the above procedures, the sampler is smoothly lowered into the flow. When the sampler almost touches the streambed, the reel direction is quickly reversed and the sampler is raised to the surface using the same constant transit rate (FISP 2000b). The person collecting the sample avoids hitting the streambed to prevent stirring up loose sediment that could bias the sample. To avoid overfilling, the person collecting the sample periodically checks the volume that has been collected and switches out the sample bottle when the volume is approximately 800 mL.

Compositing and/or multiple traverses of the cross section may be necessary. The person collecting the sample retrieves the sampler and empties the contents of the sampling device into a churn or other clean sampling container depending on equipment availability, parameters to be analyzed, and necessary sample volume while agitating the sample to ensure particles are in suspension during the transfer. Before retrieving the sampler, the person collecting the sample makes notes of or marks his/her last collection vertical on the tagline with a clothespin or similar device. After emptying the sample bottle, the person collecting the sample returns to the marked location and resumes sampling. The procedure is repeated until the necessary volume has been collected and composited. The composited sample is then dispersed and filtered as necessary depending on the parameters to be analyzed. The agitator on the churn is slowly moved up and down while dispensing sample water from the spigot into the appropriate lab sample containers.




Note: The following configuration and volumes were used to produce this diagram. The total volume of the sampler container is 1265 mL., which includes a Teflon bottle, bottle adapter and US D-77 cap. The maximum recommended sample volume is 800 mL. The maximum acceptable sample volume is 1,000 mL.

**Figure 7-4 Transit rate diagram for US DH-95 with 5/16 inch Teflon nozzle**

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# 8.0 SEDIMENT SAMPLING

Determination of constituent concentrations in bed sediments is a widely used approach to monitor and assess contaminant distributions in streams (Shelton and Capel 1994). Sediment chemistry provides information regarding both trends in contaminant loading and the potential for adverse effects on sediment and water biota. Contaminated sediments can become a storage sink and possible resource for contaminants to be released to the environment. Suspended contaminated sediments can move through a water system to be deposited, sometimes over a wide area, in previously clean waters. Contaminants from sediments have been shown to move into the food chain, and thus be available for consumption by fish, wildlife and humans. Contaminants in sediments can also affect the base of the food chain by affecting existing resident biota communities.

## 8.1 Site Selection and Selection of Depositional Zones

The objective in selecting a sample site is to obtain recently deposited fine sediment. Many of the chemical constituents of concern are adsorbed onto fine particles which typically collect in depositional zones, which are locations in streams where the energy regime is low and fine-grained particles accumulate in the stream bed. Fine-grained particles deposited in these zones are natural accumulators of trace elements and hydrophobic organic compounds (Shelton and Capel 1994).

Depositional zones include areas on the inside bend of a stream or areas downstream from obstacles such as boulders, islands, sand bars, or simply shallow waters near the shore. These zones can cover large areas at some sites and small pockets at other sites. Wadeable depositional zones are preferred because they are easy to identify and to sample.

Fine-grained surficial sediments are obtained from several depositional zones within a stream reach and composited to yield a sample representing average conditions. Avoid hard clay, gravel, disturbed, and/or filled areas. Any sediment that resists being scooped by a dredge or scoop is not fine, recently deposited material. The ideal site-planning procedure is to identify 5 to 10 depositional zones containing fine-grained particulate matter at each site. Select depositional zones that represent upstream influences and various flow regimes, namely left bank, right bank, center channel, and different depths of water if available. This will ensure that the sediment sample represents depositional patterns from various flow regimes and sources within the reach. The number of subsamples from each zone is based on the size of each zone (that is, the larger the areal extent of the zone, the greater the number of subsamples collected). Compositing the subsamples will smooth the local scale variability and represents the average sediment contaminant levels present at the site (Shelton and Capel 1994).

In order to compare samples over time and from site to site, they must be collected in a consistent manner. Sediment will vary from site to site and can vary between sample events at a particular site. In areas of frequent scouring there may not be sufficient sediment for collection during or following periods of high flow. Sediment collection during these times may prove unsuccessful and may have to be rescheduled.

## 8.2 Sediment Sampling Procedures

Devices used to collect sediment samples for chemical analyses should have (1) the ability to sample surficial sediments with minimal loss of the fine material in the sediment/water interface and (2) the ability to sample sediment without contaminating the trace elements or organic compounds (Shelton and Capel 1994).

Typically, only the aerobic layer is sampled, because this zone represents more recent deposits and is where a large portion of the benthic infauna live. If the sediment does not have an aerobic zone, collect the top two centimeters (cm) for analysis. If the aerobic zone is deeper than five cm, collect the sample for analysis from the top five cm to obtain the most recently deposited sediments. Composite three or more grabs for the sediment sample.

In areas where very little scouring normally occurs, such as in reservoirs, the sediment will be vertically stratified. Often the sediment is consolidated enough to be extruded into a flat pre-rinsed plastic or Teflon pan as a cube-shaped block of mud. Vertical stratification can then be observed. Typically, there may be light brown silt on top, followed by a gray colored aerobic zone overlying a typically black anaerobic layer. Because the thickness of these layers is variable, it is difficult to prescribe a certain thickness representing "recent deposits" to be sampled.

Between sample sites, thoroughly wash all sampling equipment and compositing containers with phosphate-free detergent to remove all sediment. After washing, rinse the equipment with copious amounts of tap water first (if available), then triple rinse with deionized water (Radtke 2005).

### 8.2.1 Collection at Wadeable Sites

For shallow water sites collect the sample using the following procedure:

A. Select appropriate collection equipment for the type of analytical suite planned: plastic or Nalgene® for metals, stainless steel or Teflon for organics.

B. From several representative depositional zones, composite sufficient material to make up the sample (preferably at least two times the necessary sample volume) into an appropriate mixing container after decanting any excess water over the back of the scoop.

C. Mix the sample well.

D. Transfer an aliquot of the mixed material to the final, labeled sample container (typically a wide-mouthed glass jar) and place on ice for transport to the analytical facility. If shipment cannot be accomplished in a timely manner, the sample should be frozen prior to shipment. Sediment samples are not preserved.

### 8.2.2 Collection at Non-Wadeable Sites

When sampling non-wadeable streams and rivers, collect sediment samples from depositional areas along the wadeable portions of both banks, if possible. Use an Eckman dredge from boats and bridges, or in deep water where it is not possible to collect samples near the banks.

#### *8.2.2.1 Eckman dredge procedures*

A. Slowly lower the Eckman dredge to the bottom with a minimum of substrate disturbance.

B. Release the messenger to close the dredge.

C. Retrieve the closed dredge at a moderate speed (less than two feet per second [ft/s]).

D. Upon retrieval, examine the grab to ensure that the sediment surface is undisturbed. The grab is rejected if it does not meet these criteria:

- Mud surface must not be pressing out of the top of the sampler. If it is, lower the dredge more slowly.
- Overlying water must not be leaking out along the sides of the sediment in the dredge. This ensures the surficial sediment is not washed out.
- Sediment surface is flat and level in the sampler. If it is not level, the dredge has tilted over before closing.

E. Gently decant the water overlying the sediment in the dredge by slightly tipping the dredge with the lid closed until the water runs out the top. The decanting process should remove all of the overlying water but not remove the surficial sediments. The laboratory reports percent water for the sample, so overlying water is not included in the sample container.

F. The sediment is examined for depth of penetration, color and thickness of top aerobic zone, and texture. These observations are recorded on the field sheet.

G. Transfer an aliquot of the material to the final, labeled sample container and place on ice for transport to the analytical facility. If shipment cannot be accomplished in a timely manner, the sample should be frozen prior to shipment. Sediment samples are not preserved.

When the suspended load in rivers and streams precipitates due to reduction of velocity, most of the resulting sediment will be "fresh." In such instances, the entire bite from the dredge subsample may be used.

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# 9.0 NUTRIENT SAMPLING

This section describes the procedures for monitoring the nutrient variables used to assess streams and rivers. Generally, streams refer to wadeable systems and rivers are those systems that have drainage areas greater than 2,300 square miles and cannot be monitored effectively with the biological and habitat methods developed for wadeable streams. Refer to the **Rivers/Streams MAS Equipment Checklist** for the necessary field equipment.

Definitions:
**Algae** are non-vascular plants with no true roots, stems, or leaves. They are mostly aquatic and range from tall stalks of kelp to fuzzy growths of green filamentous algae to microscopic silica encased diatoms.

**Assessment Unit (AU)** is a length of stream defined by the Surface Water Quality Bureau (SWQB) and used to assess waterbodies for the Integrated 303(d)/305(b) List.

**Biological Sampling Index Period (BSIP)** the time of year in which biological samples (fish, macroinvertebrates, periphyton) are collected for stream biological assessment.

**Filamentous algae** are algae that grow as fibers or strands of cells forming filaments or mats that are attached to substrate. The filaments can either be branched or unbranched.

**Macrophyte** is a general term that applies to many types of aquatic vegetation including flowering vascular plants, mosses, and ferns. Four categories of macrophytes are defined by their relationship to the air, water, and substrate: emergent, floating-leaved, submerged, and freely floating (USEPA 2000).

**Emergent macrophytes** grow on the banks of rivers and streams in depths of water generally less than a meter and are typically rooted in the sediment with a portion of the plant in the water and part extending into the air. Common emergent macrophytes include plants such as reeds (*Phragmites* spp.), bulrushes (*Scirpus* spp.) and cattails (*Typha* spp.).

**Floating-leaved macrophytes** are rooted to the river bottom with leaves that float on the surface of the water, such as waterlilies (*Nymphaea* spp.) and spatterdock (*Nuphar* spp.).

**Submerged macrophytes** are a diverse group that grow completely underwater and include mosses (*Fontinalis* spp.), stoneworts (*Nitella* spp.) and numerous vascular plants, such as various pondweeds (*Potamogeton* spp. and *Elodea canadensis*), tape-grass (*Vallisneria* spp.), and exotic species including hydrilla and Eurasian watermilfoil.

**Free-floating macrophytes** typically float on or just under the water surface with their roots suspended in the water column. These unattached macrophytes range in size from small duckweeds (*Lemna* spp.) and water ferns (*Salvinia* spp.) to larger surface floating plants such as water hyacinth (*Eichhornia crassipes*).

**Periphyton** is an assemblage of organisms that grow on underwater surfaces and includes algae, bacteria, fungi, protozoa, and other organisms.

**Phytoplankton** are small (often microscopic) aquatic plants that are suspended in water.

**Rapid Bioassessment Protocol** (RBP) is the protocol developed by the USEPA for collecting, processing, and assessing fish, benthic macroinvertebrates, and periphyton from wadeable streams (Barbour et al 1999).

## 9.1 Level I Nutrient Survey: Qualitative Measures and Data Review

Level I is a screening level survey that includes collecting on-site observations and measurements of chemical parameters as needed. It is intended to provide the data needed to conduct the Level I Nutrient Assessment. The assessment should be conducted prior to the biological sampling index period (BSIP) (August 15 – November 15) to allow sites that do not pass the Level I assessment to be surveyed more intensively during the index period. The Level I Nutrient Assessment uses a qualitative determination of algal biomass in addition to a number of chemical parameters including dissolved oxygen (D.O.) concentration, percent D.O. saturation at the local elevation, and pH as well as total phosphorus and total nitrogen. Typically, all of these parameters are measured as part of a general water quality survey. Sample collection methods for the parameters listed above are described in *Field Data Measurements* and *Chemical Sampling* sections of the 2007 NMED/SWQB Standard Operating Procedures for Data Collection (SOP). There is no form for the Level I Nutrient Survey as the data are recorded on the general water quality field sheets and compiled in the SWQB Water Quality Database.

### 9.1.1 Periphyton and Algae

A qualitative algal biomass survey is part of the Level I Nutrient Survey and is conducted by field crews as part of regular water quality surveys. The information is recorded on the second page (back side) of the field sheets. This survey should be conducted at each sample site and completed once per season, i.e. in spring, summer, and fall. These seasons refer not to the calendar, but to the character of the hydrograph for a given waterbody. Typically in New Mexico, spring is characterized by higher flows resulting from snow melt, while summer and fall are characterized by low flow conditions interrupted by spates from seasonal rain storms. During the summer nutrient survey, the study lead should note which, if any, sites might need to be assessed using a reference approach due to the apparent **naturally high** productivity. This will allow for scheduling of appropriate Level II Nutrient Surveys to be conducted at the test site and a suitable reference site during the BSIP.

In the context of this guidance, periphyton refers to the slime layer or biofilm growing on substrate. It is composed primarily of microscopic organisms while the algae, noted in the percent coverage field, are mainly macroalgae. Algae refers to the visible growth of non-rooted aquatic vegetation attached to the stream substrate. Percent algal coverage and a rating of periphyton growth is a qualitative indicator of algal biomass. An estimate of percent macrophyte coverage will also be recorded. Macrophytes are primarily the large aquatic vegetation rooted in the substrate, although they are also represented by small leaves with roots floating on the surface (duckweed and water fern).

Observations will be made in and around a riffle unless none are present or accessible or the velocity is so great that it limits algal growth (i.e. velocities exceed 1.75 ft/sec) and more algae are found in glides than in riffles. Riffle habitat was selected to help partition the variability in algal biomass and because benthic algal growth is stimulated by moderately high velocities (Barbour et al 1999, Biggs and Kilroy 2000). The Rapid Bioassessment Protocol (RBP) states that, "single habitat sampling should be used when biomass of periphyton will be assessed," and, "the recommended substrate/habitat combination is cobble obtained from riffles and runs with current velocities of 0.33 - 1.6 ft/sec" (Barbour et al 1999). If the velocity appears to be limiting algal growth, then select a glide (a length of stream with intermediate velocity) to survey. If access to the reach is limited, use a transect across the stream at the sample location. However, effort should be made to find a representative riffle whenever possible.

The methods for examining and rating algal and periphyton cover are described below and include surveys of both river and stream systems. As the aquatic vegetation observations are conducted, check under stones and in depositional areas for the presence of an anoxic layer. Anoxic conditions are characterized by a black color and rotten egg smell associated with hydrogen sulfide.

1) Select a suitable riffle near the water quality sample site.

2) Starting at either the top or bottom of the riffle, visualize a line running diagonally through the riffle. Select a landmark on the far bank that corresponds to the end of the diagonal transect. Note the midpoint, ¼, and ¾ distance. Five observations will be made along this diagonal: near the left bank, at ¼, ½, and ¾ the width, and near the right bank. In this manner, locations across the length and cross-section of the riffle will be observed.

3) From your starting point (on the bank at either the top or bottom of the diagonal through the riffle) observe the transect across the stream and estimate the percent algal cover in the wetted width. Look for strands of non-rooted aquatic vegetation attached to the substrate.

4) Take a step into the stream, reach down and pick up the cobble or large gravel nearest the front of your foot. Observe the particle and note the periphyton thickness and associated rating (see below).

5) Walk along the diagonal, stopping at ¼, ½, and ¾ the width, and near the far bank and make observations of both the algal cover across the wetted width and periphyton thickness on the substrate. If you cannot safely wade across the entire riffle, go as far as possible then turn back and continue observations on a diagonal back to the bank at the opposite end of the riffle from where you started. For reaches that you cannot wade halfway across, pick up and/or examine the substrate in 10 locations that you can reach and rate the algal and periphyton cover as described below. The observations should be made over an area that is approximately as long as the river is wide.

6) Once you reach the end of the transect or make 10 observations, record the average percent algal coverage and periphyton rating on the back of the water quality field sheet.

7) Note where in the stream growth is occurring (e.g., in low flow areas, only near seeps, on fine substrate, only on large stable substrate, etc). The physical characteristics of the substrate can influence the amount of algal coverage. As some substrates are too soft or unstable for algae to anchor to, the overall percent algal coverage may reflect the amount of stable substrate in the stream rather than the potential biomass.

Periphyton Rating: Look for the presence of slime on the coarse substrate (cobbles and gravel). Rate the periphyton thickness on the substrate using the following scale (Stevenson 1996):

- 0 indicates substrate is rough with no apparent growth;
- 1 indicates a thin layer of microalgae is visible (tracks can be drawn in the film);
- 2 indicates accumulation of microalgae to a thickness of 0.5-1 mm;
- 3 indicates accumulation of microalgae from 1 mm to 5 mm thick;
- 4 indicates accumulation of microalgae from 5 mm to 20 mm and
- 5 indicates layer of microalgae is greater than 20 mm.

### 9.1.2 Water Chemistry

Measurements of dissolved oxygen concentration (D.O.), percent D.O. saturation at the local elevation, and pH will be collected during water quality surveys. The assessment will be made with data exported from the NMED database. If the Assessment Unit (AU) being surveyed is not part of a current water quality survey, take measurements of D.O. concentration, percent D.O. saturation at the local elevation, and pH as described in the *Field Data Measurements* section of the SOP.

Total phosphorus, Total Kjeldahl Nitrogen, and Nitrate + Nitrite are also monitored during water quality surveys. Total Nitrogen is calculated by adding Total Kjeldahl Nitrogen plus Nitrate + Nitrite. In the event that Nitrate + Nitrite or Total Kjeldahl Nitrogen are below the detection limit, a value of one half the detection limit will be used (Gilbert 1987). The assessment will be made with data exported from the NMED database. If the AU being surveyed is not part of a current water quality survey, collect water samples for analysis of Total phosphorus, Total Kjeldahl Nitrogen and Nitrate + Nitrite as described in the *Chemical Sampling* section of the SOP. Also take flow measurements as described in the *Measuring Flow* section of the SOP if there are no gages in the AU.

## 9.2 Level II Nutrient Survey: Quantitative Measures

The Level II survey involves quantitative measurement of selected indicators. **A Level II Survey will be conducted if the Level I Assessment indicates that nutrient impairment may be occurring in an AU or if the AU was previously listed for nutrient impairment.** This survey should be conducted during the BSIP. Normally, during this time there is the potential to have higher concentrations of plant nutrients in the stream and to see the effects of higher water

temperatures and maximum solar gain from the summer conditions. Higher temperatures tend to enhance algal growth, photosynthesis, and respiration resulting in greater variation in diurnal DO and pH values (Fisher and Grimm 1983). If a scouring event has occurred, the Level II survey should be conducted at least 3 weeks after the event (Biggs and Kilroy 2000). Look for evidence of high flows on site and consult regional gage data. All data from the survey should be recorded on a **Level II Nutrient Survey Form (Streams)** or a **Level II Nutrient Survey Form (Rivers)**, depending on the type of waterbody being surveyed.

Before selecting a location for the survey, note the character of the stream while driving to the site and walk a couple of hundred meters of the stream to ensure that the sampling station is representative of the reach being characterized. Collect the following data from each study site.

### 9.2.1 Location and Photographs

Describe the site location and record a GPS reading for latitude, longitude, and elevation. Take photos of the reach being studied, including the stream, substrate, and riparian area. Include the date, time, and personnel as well as description of photo location and content.

### 9.2.2 Flow Conditions

Note the current flow condition and look for evidence of recent high flow events. Evidence of recent high flows includes flood debris on the bank or in riparian vegetation and vegetation on the banks that is bent over in the direction of flow. Also, look at gage data where available and make a note on the field sheet if gage data are available.

### 9.2.3 Site Description

The shaded areas on the Level II Survey Form can be filled in after the field visit from the habitat and field forms, if an EMAP survey was conducted. If an EMAP survey was not conducted, fill in the shaded areas using data from the riffle or other habitat where the periphyton sample was collected, not the whole reach. Refer to the *Measuring Flow* and the *EMAP Habitat Characterization* sections of the SOP to measure wetted width, depth of water, velocity of water, and canopy cover. Record the turbidity, in NTU, from the water quality field sheet and note the water color (green, brown, clear, etc.).

Note features that influence the supply, transport, and cycling of plant nutrients (Fisher and Grimm 1983). Note the presence and location of watercress in the stream as it can indicate the presence of springs that may be a natural source of nutrients. Watercress is an emergent macrophyte that can be identified by the following (USDA 1988): (1) it has small whitish flowers with four petals, several arranged closely, (2) has roots from stem nodes, and (3) has pinnately divided leaves with very large terminal leaflets on mature leaves (see photos below). Note the location of springs, wetlands, and upwelling areas. Upwelling areas may be indicated by patches of dense algal growth. Observe the riparian area, noting the height, density, and type of streamside vegetation. Also, note areas where riparian vegetation has been removed. Finally, fill out a Site Site Condition Class Verification & Probable Source Field Sheet and identify probable sources of plant nutrients to the survey reach.

### 9.2.4 Sonde Deployment

A multiparameter meter (sonde) should be deployed for at least seven days and set to record hourly D.O., pH, specific conductance, temperature, and turbidity values. Refer to the *Field Data Measurements* section of the SOP for notes on field calibration of sondes.

Where conditions allow, the sonde should be placed upright in the stream in a location with significant flow, placed in PVC pipe, and secured to a T-post, with nylon straps or hose clamps (see photo at right). The PVC pipe should have holes in it to protect the sonde from debris, while allowing water to flow past the probes. The sonde should not be touching the bottom of the stream. In addition, the sonde should be further secured with a chain or cable and padlocked to a tree or other stable object on shore, and should be placed where it is not easily detectable. If the sonde cannot be placed upright it may be laid on the substrate in the PVC pipe and secured to a stable object. If the sonde cannot be safely deployed due to potential vandalism or likely high flow events, it should not be deployed for long-term unattended logging. Refer to the **Sonde Deployment Instructions** form for instructions on deploying a sonde for unattended sampling. Fill out a **Sonde Deployment-Retrieval Field Sheet**.

### 9.2.5 Water Chemistry

Water samples should be collected for analysis of Total Suspended and Total Dissolved Solids (TSS/TDS), alkalinity, and Total Nutrient (phosphorus and nitrogen) concentrations. Refer to the *Chemical Sampling* section of the SOP for sample collection and preservation methods.

### 9.2.6 Periphyton Sampling

Observe the stream substrate and note the dominant and subdominant size classes (e.g. silt/clay, sand, cobble, etc.). Collect a sample of benthic algae from a known area of substrate for analysis of Chlorophyll *a* concentration, Ash Free Dry Mass (AFDM), and community composition. Record the sampling information in the following table found on the **Level II Nutrient Survey Form (Streams or Rivers)**. It is important to accurately measure and record the area and volumes so the final concentration per area of substrate may be calculated. The methods for collecting and handling periphyton samples are described in the *Biological Sampling* section of the SOP.

As the periphyton sample is collected, a qualitative survey of aquatic vegetation and periphyton may be conducted (Stevenson 1996). Each rock that is picked up for periphyton collection is rated for periphyton thickness and macro-algae cover. The transect where the periphyton sample was collected is rated for macrophyte cover. At the sample location, an estimate of water depth and a general rating of the velocity is recorded. The ratings are recorded on the **Aquatic Vegetation Evaluation** form. Rate the periphyton, macro-algal, and macrophyte cover separately using the following scales:

Periphyton:

- 0 indicates substrate is rough with no apparent growth;
- 1 indicates a thin layer of microalgae is visible (tracks can be drawn in the film with the back of your fingernail);
- 2 indicates accumulation of microalgae to a thickness of 0.5-1 mm;
- 3 indicates accumulation of microalgae from 1 mm to 5 mm thick;
- 4 indicates accumulation of microalgae from 5 mm to 20 mm and
- 5 indicates layer of microalgae is greater than 20 mm.

Macro-algal and macrophytes:

- 0 indicates no macrophytes or macro-algae present;
- 1 indicates some (but < 5% coverage) macrophytes or macro-algae present
- 2 indicates 5-25% cover of substratum by macrophytes or macro-algae
- 3 indicates 25-50% cover by macrophytes or macro-algae
- 4 indicates 50-75% cover by macrophytes or macro-algae and
- 5 indicates >75% cover by macrophytes or macro-algae

### 9.2.7 Algal Bioassays (OPTIONAL)

If stream observations indicate that algal biomass is a problem in the stream or an NPDES permit is being written for the Assessment Unit, a limiting nutrient analysis and algal growth potential

test may be performed (USEPA 1975 and 1978). Two one-gallon samples of water (no acidification) should be collected and immediately stored on ice. General methods for collecting surface water samples are described in the *Chemical Sampling* section of the SOP. Deliver samples to UNM Biology Department within two days of collecting the samples. Please call Dr. Barton @ 505-277-2537 before taking the samples to his lab. Attach results to the survey form when received.

9.2.8 Landscape Features

If a site appears to be impaired, estimate the extent of the impacted area (i.e. the distance of the stream that is impaired). Note where indicators of nutrient enrichment change and regional geology and landscape patterns that may influence nutrient loading and cycling (e.g., an alluvium to bedrock transition, open basin to canyon transition, etc.).

## FORMS

River/Streams MAS Equipment Checklist
Level II Nutrient Survey Form (Streams)
Level II Nutrient Survey Form (Rivers)
Probable Source Field Sheet
Sonde Deployment Instructions
Sonde Deployment-Retrieval Field Sheet
Aquatic Vegetation Evaluation

## REFERENCES

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USEPA. 1978. The Selenastrum Capricornutum Prinz Algal Assay Bottle Test. EPA-600/9-78-018.

USEPA. 2000. Nutrient Criteria Technical Guidance Manual: Rivers and Streams. EPA-822-B-00-002.

# 10.0 BIOLOGICAL SAMPLING

## 10.1 Benthic Macroinvertebrates

Benthic macroinvertebrates inhabit substrates of almost all waterbodies. Monitoring benthic macroinvertebrate communities is a direct measure of the biological condition of the waterbody. These assemblages are useful for assessing the status of a waterbody and detecting trends in ecological condition. Benthic macroinvertebrate communities respond to a wide array of stressors, can have relatively long life cycles (1+ yrs.), and are relatively immobile. Because macroinvertebrate community structure is a function of past and/or present conditions, monitoring may enable determination of the types of stressors that affect a macroinvertebrate community.

Benthic macroinvertebrate samples should not be collected less than 30 days after a bankfull flow event. If the flow event results in large amounts of sediment erosion and/or deposition, sampling goals should be carefully considered to determine if and when sampling should be conducted.

### 10.1.1 Wadeable Streams/Rivers

SWQB adopted the EPA EMAP habitat and macroinvertebrate protocols in 2006, recognizing that the "riffle only" method for collecting macroinvertebrate samples was not able to address all lotic water body types (i.e. sample reaches with no riffles present). **Field crews must read Section 11 (Klemm et al. Unpublished draft) in the Environmental Monitoring and Assessment Program – Surface Waters: Western Pilot Study Field Operations Manual for Wadeable Streams (Peck et al. Unpublished draft) in detail** (http://www.epa.gov/emap/html/pubs/docs/groupdocs/surfwatr/field/fomws.html)**.** This protocol uses a 30 cm wide D-frame kick net with a mesh size of 500 μm and a sample area of 0.09 $m^2$ (1 $ft^2$). Two types of samples are collected: a "reach wide" sample and a "targeted riffle". The reach wide sample is comprised of one individual sample from each equidistantly spaced transect throughout the reach, and composited into one. The targeted riffle sample is comprised of 8 individual samples from riffle habitat in the reach that are composited into one.

#### *10.1.1.1 Reach Wide Sample*

Establish transects following protocols in the Physical Habitat Characterization of this SOP. Figure 1 illustrates the sampling design for the reach wide sample.

Collect a sample from each of eleven transects after randomly establishing the sample location at the first transect (left, center, or right) using the riffle/run procedure. If there is not sufficient current to extend the net, the area is operationally defined as a pool/glide habitat and the sample is collected using the pool/glide procedure. Figure 1 illustrates the sampling design for the reach wide sample. For each transect sample, record the dominant substrate type (fine/sand, gravel, coarse substrate (coarse gravel or larger) or other (e.g., bedrock, hardpan, wood, aquatic

vegetation, etc.) and the habitat type (pool, glide, riffle, or rapid) for each kick net sample collected on the Sample Collection Form. As you proceed upstream from transect to transect, combine all kick net samples into a bucket or similar container labeled “Reach Wide”, regardless of whether they were collected using the “riffle/run” or “pool/glide” procedure.

If it is impossible to sample at the sampling point with the modified kick net following either procedure, spend about 30 seconds hand picking a sample from about 0.09 sq m (1 sq ft) of substrate at the sampling point. Place the contents of this hand-picked sample into the “Reach Wide” sampling container. For vegetation-choked sampling points, sweep the net through the vegetation for 30 seconds. If sample cannot be collected at the correct location, sample at the closest point possible and note on Sample Collection Form. If no sample can be collected for a transect, make a note on Sample Collection Form.

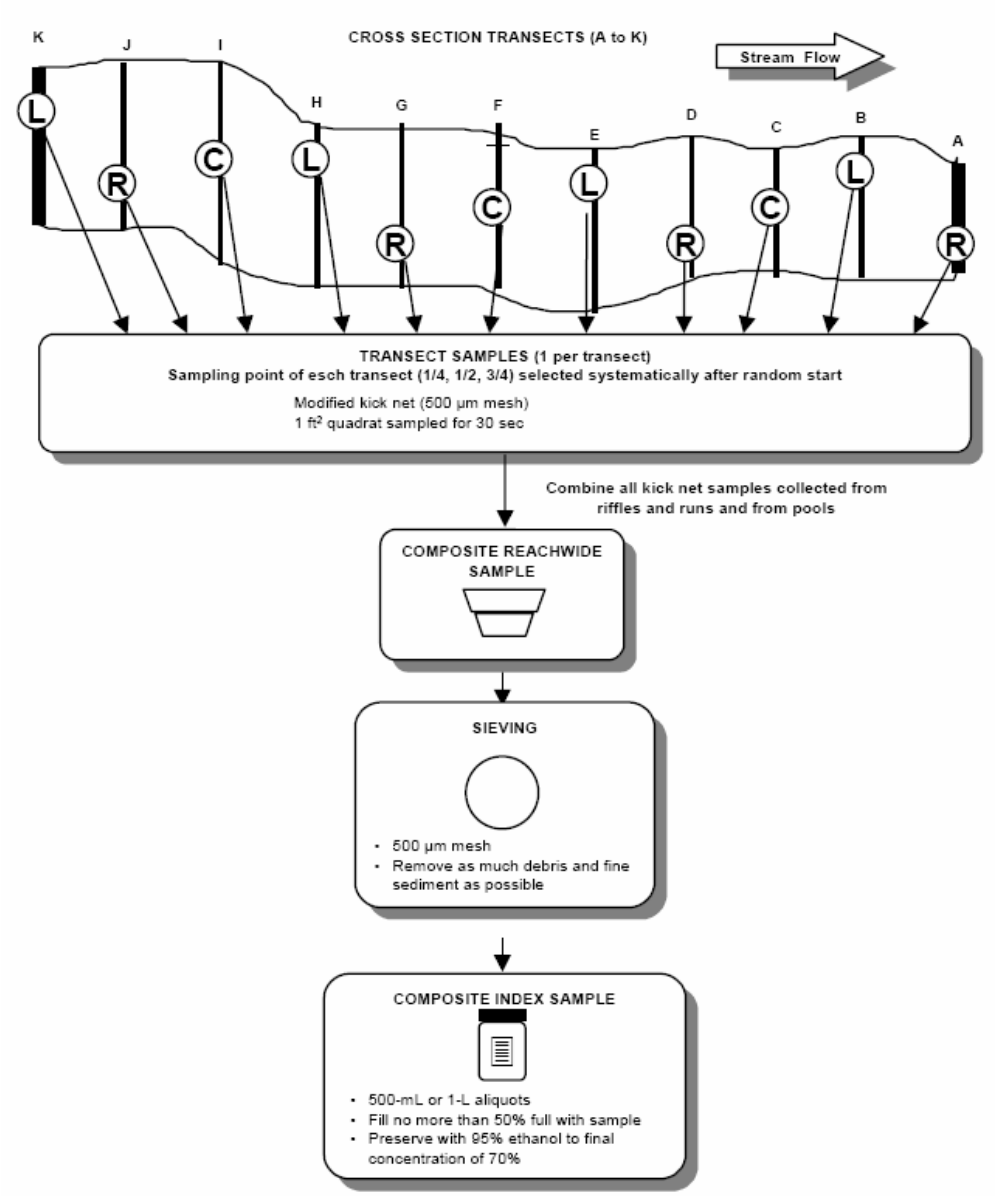


**Figure 10-1 Sampling design for reach wide benthic macroinvertebrate sample locations**

*10.1.1.2 Targeted Riffle Sample*

While laying out the reach and transect flags, survey the stream reach to visually estimate the total number (and area) of riffle macrohabitat units. Riffle habitat area must be greater than 0.09 $m^2$ (1 $ft^2$) to be considered a riffle macrohabitat unit for sampling. Figure 2 illustrates the sampling design for the targeted riffle sample. When total available area of riffle habitat is less than 0.74 $m^2$ (8 $ft^2$) (i.e., 8 non-overlapping kick net samples cannot be collected), do not collect a targeted riffle sample. There may be stream reaches where more than one 0.09 $m^2$ (1 $ft^2$) kick net sample is collected from a single riffle unit.

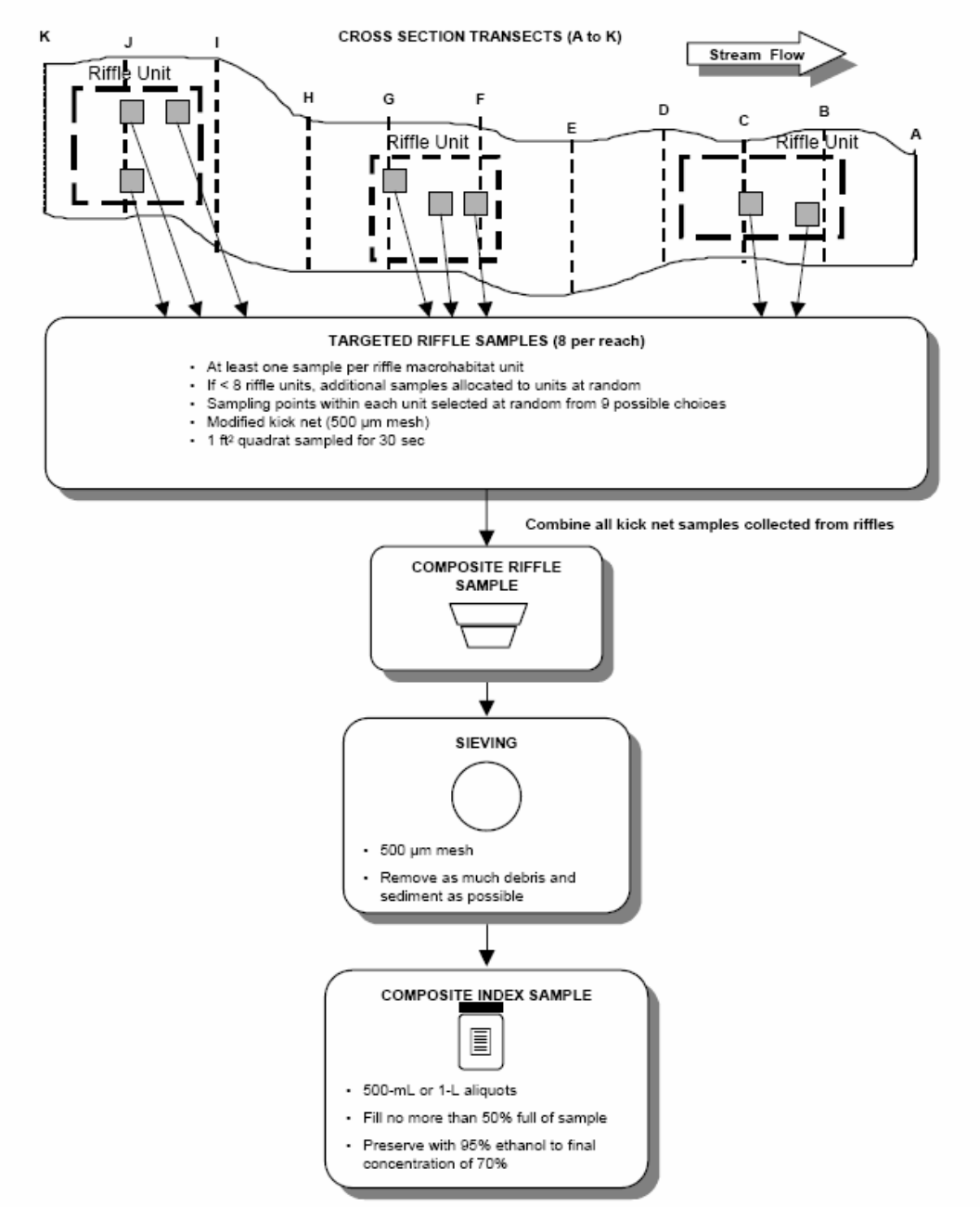


**Figure 10-2 Sampling design for targeted riffle benthic macroinvertebrate sample locations**

The objective for selecting sampling points within the available riffle macrohabitat units is to allocate points throughout the sampling reach as much as possible. Procedures for collecting a point sample using the kick net from riffle macrohabitat units are detailed in Klemm et al. (Unpublished draft). At each sampling point, a quadrat having a total area of 0.09 $m^2$ (1 $ft^2$) is sampled. Because the reach-wide and targeted riffle samples are collected in the order they are encountered during a single pass through the reach, it is very important to rinse the kick net thoroughly between samples to avoid carryover and possible cross-contamination of the targeted riffle sample and the reach-wide sample.

#### *10.1.1.3 Sample Processing*

After collecting kick net samples for both the reach wide and targeted riffle samples, prepare one composite sample from the contents of the "Reach Wide" bucket and one composite sample from the contents of the "Targeted Riffle" bucket. Record tracking information for each composite sample on the Sample Collection Form included in the EMAP field data form packet in the Habitat section of this SOP.

Place contents of bucket into sieve (500 μm mesh size) and wash off and discard large objects. Depending on how much debris was collected, this may need to be done in several steps, being careful not to overfill the sieve. Repeated rinsing should be done until rinse water is clear. When large amounts of sediment are collected, thoroughly agitate the contents by swirling and pour off all organic debris into the sieve. Refill the bucket with stream water to more than 1/3 – 1/2 full. Once organic debris has been removed, remove any large pebbles, inspect for attached macroinvertebrates (scrub off all that are present), and discard. Agitate the bucket contents by swirling, and immediately pour off the water into the sieve before lighter items in the water column can settle out. Be careful not to pour out any sediment. Repeat several times until you are confident that all macroinvertebrates have been removed. Visually inspect any remaining sediment for snails, clams, mussels, and macroinvertebrate cases and tubes. Remove the items and place in the sieve or directly into the sample jar.

Using a rinse bottle, wash the contents of the sieve into the sample jar. Do not fill the sample jar more than 1/2 full. Divide the sample into as many jars as needed and indicate jar number on the sample label (e.g., 1 of 3). Fill the jar with 95% ethyl alcohol (EtOH) to the top, leaving no headspace. Replace the cap on each jar. Slowly tip the jar to a horizontal position, then gently rotate the jar to mix the preservative. Do not shake the jar.

A completed sample label is required for all sample jars from a single composite sample. An example label for use inside the benthic macroinvertebrate sample jars is presented in Figure 10-3.

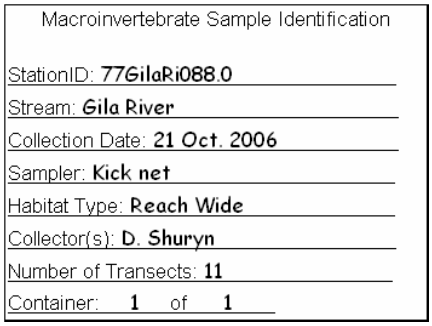


**Figure 10-3 Example label for macroinvertebrate sample jar**

### 10.1.2 Non-Wadeable Streams/Rivers

Macroinvertebrate sample collection procedures should follow EPA EMAP **Environmental Monitoring and Assessment Program - Surface Waters: Field Operations and Methods for Measuring the Ecological Condition of Non-Wadeable Rivers and Streams** (Lazorchak et al. 2000). The EMAP non-wadeable protocols should be supplemented with sampling of woody debris, uni-connected side channels, and off channel pools following the protocol in Table 1.

The following protocol has been used in the Middle Rio Grande during wadeable flow conditions. The protocol is based on EMAP wadeable methodologies and supplemented with USGS and other microhabitat sampling methodologies.

The label below (Figure 10-4) should be used for large river macroinvertebrate samples regardless of the protocols used, wadeable or non-wadeable. The Large River Sample Collection form should also be filled out as macroinvertebrate and periphyton samples are being collected.

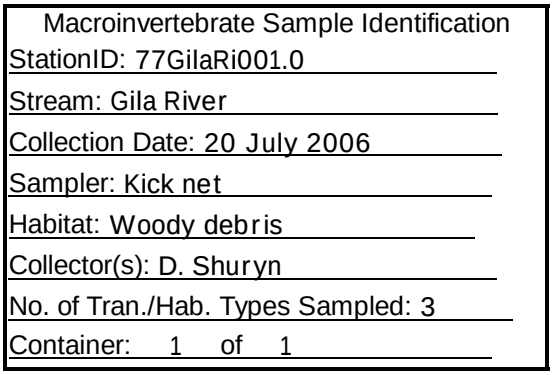


**Figure 10-4 Example label for macroinvertebrate sample jar**

**Table 10-1 Identifying macroinvertebrate sampling locations**

| Habitat Types | | Methods/Protocols |
|---|---|---|
| Continuous | | See EMAP Protocols (Peck et al., Unpubl. Draft) |
| Discrete | | 1. At transect A, after completing the continuous habitat sampling, look upstream and determine the closest discrete habitat of the following types:<br>▪ Uni-connected side channel (length > 1.0 m),<br>▪ Off-channel pool (depth > 0.03 m),<br>▪ Woody debris (> 0.03 m diameter; > 0.3 m in water)<br>2. Sample the first discrete habitat encountered and record on field sheet.<br>3. Do not sample poorly represented habitats. If the reach contains less habitat than detailed in 1 above, do not collect a sample.<br>4. After sampling the discrete habitat (e.g., woody debris), look for the next closest |

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| | | discrete habitat type that has not been sampled (e.g., uni-connected side channel, off-channel pool). Do not go upstream of transect B.<br>5. Collect one sample for each of type of discrete habitat encountered between transect A and B.<br>6. If there is no discrete habitat of a particular type (e.g., woody debris) between transect A and B, look for the closest discrete habitat of that type between transect B and C after sampling has been done at transect B.<br>7. Note on field sheet if none found.<br>8. If a discrete habitat type was sampled between transect A and B, then do not sample that habitat type between transect B and C.<br>9. Discrete habitat types should be sampled between transect A – B, C – D, E – F, G – H, and I – J.<br>10. Once a discrete habitat has been located, proceed to the appropriate protocol below to locate the sample collection site. |
| | ***Uni-connected side channel*** | 1. Locate the assigned sampling area by randomly determining if it is an upper (U), middle (M), or lower (L) sampling area in the channel (lower is considered to be the connected end). A die roll of 1 or 2 indicates U, 3 or 4 indicates M, and 5 or 6 indicates L (or use a digital wristwatch and glance at the last digit (1-3=L, 4-6=C, 7-9=R). Mark U, M, or L on the Sample Form. Assign sampling areas at each successive uni-connected side channel in order as U, M, or L after the first random selection.<br>2. Visually lay out the core area of the unit sampled into 9 equal quadrats (i.e., a 3 × 3 grid).<br>3. Select a quadrat for sampling using the method in Step 1 from the following list of locations (right and left are determined as you look downstream):<br>▪ Lower right quadrat<br>▪ Lower center quadrat<br>▪ Lower left quadrat<br>▪ Right center quadrat<br>▪ Center quadrat<br>▪ Left center quadrat<br>▪ Upper right quadrat<br>▪ Upper center quadrat<br>▪ Upper left quadrat<br>4. Collect the sample in the center of the selected quadrat.<br>5. If the sample point is too deep to sample, move to the closest area that can be sampled in that quadrat.<br>6. If the quadrat is too deep to sample, then randomly re-select quadrat. |
| | ***Off-channel Pool*** | 1. If the pool is entirely within visual range, proceed to Step 2. If the pool is long and extends out of visual range, locate the assigned sampling area by randomly determining if it is an upper (U), middle (M), or lower (L) sampling. A die roll of 1 or 2 indicates U, 3 or 4 indicates M, and 5 or 6 indicates L (or use a digital wristwatch and glance at the last digit (1-3=L, 4-6=C, 7-9=R). Mark U, M, or L on the Sample Form. If subsequent pools are long and extend out of visual range assign sampling areas at each successive pool in order as U, M, L after the first random selection.<br>2. Visually lay out the core area of the unit sampled into 9 equal quadrats (i.e., a 3 × 3 grid).<br>3. Select a quadrat for sampling using the method in Step 1 from the following list of locations (right and left are determined as you look downstream):<br>▪ Lower right quadrat<br>▪ Lower center quadrat<br>▪ Lower left quadrat<br>▪ Right center quadrat<br>▪ Center quadrat<br>▪ Left center quadrat |

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| | | ▪ Upper right quadrat.<br>▪ Upper center quadrat.<br>▪ Upper left quadrat<br>4. Collect the sample in the center of the selected quadrat.<br>5. If the sample point is too deep, to sample move to the closest area that can be sampled in that quadrat.<br>6. If the quadrat is too deep to sample, then randomly re-select a quadrat. |
| | ***Woody Debris*** | 1. Once the sample area has been determined for the woody debris habitat, inspect the woody debris for evidence that it has been submerged for an adequate amount of time (e.g., evidence of algal growth, partially decomposed vegetation in the debris, soft, waterlogged wood, etc.).<br>2. Inspect the woody debris for an area that can easily be trimmed off with pruning shears. |

**Table 10-2 Collecting macroinvertebrate samples**

Macroinvertebrate samples will be collected according to EMAP protocols for the continuous habitat types (transect method). Two types of invertebrate samples, semi-quantitative kick nets and semi-quantitative chironomid pupal exuviae (Chironomis spp.), will be collected. A semi-quantitative macroinvertebrate sample will be collected to provide a measure of relative abundance of the invertebrate taxa living within the reach. A semi-quantitative chironomid pupal exuviae sample will be collected to provide a measure of relative abundance and diversity of the chironomid taxa living in the reach.

The dipteran family, Chironomidae, is expected to dominate macroinvertebrate samples. Collection of a chironomid pupal exuviae sample will give a better idea of the taxa that make up the expected dominant family. Typically, a chironomid taxa list generated from a pupal exuviae collection is 2 – 3 times larger than a taxa list generated from kick net samples (Ferrington 1987). This is the result of two factors; 1) pupal exuviae allow for a finer taxonomic resolution and; 2) the method of collection allows for all habitats within approximately a 500 m reach to be sampled.

| Habitat Types | | Methods/Protocols |
|---|---|---|
| Continuous | | A kick net sample is collected from each of the eleven cross-section transects (Transects A through K) at an assigned sampling point (Left [25% of wetted width], Center [50% of wetted width], or Right [75% of wetted width] facing downstream). The sampling point at Transect A is assigned at random using die or other suitable means (e.g., digital watch). A die roll of 1 or 2 indicates U, 3 or 4 indicates M, and 5 or 6 indicates L (or use a digital wristwatch and glance at the last digit (1-3=L, 4-6=C, 7-9=R). Once the first sampling point is determined, points at successive transects are assigned in order (Left, Center, Right). At each sampling point, determine if the habitat is lotic-erosional or lotic-depositional. Areas where there is not a sufficient current to extend the net, and sandy bottom substrates are not mobile, are operationally defined as lotic-depositional habitat. To collect a kick net sample from a sampling point classified as lotic-erosional habitat, follow the procedure presented below. To collect a kick net sample from a sampling point |

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| | | classified as lotic-depositional habitat, follow the procedure presented below. Record the habitat type and sampling point on the Sample Collection Form for each kick net sample collected, as shown below. As you proceed upstream from transect to transect, combine all kick net samples collected from lotic-erosional habitats into a bucket or similar container labeled “RIFFLE”. Combine kick net samples collected from lotic-depositional habitats into a second bucket or container labeled “POOL”. Fill in the checklist as individual activities are completed. If a sample can not be obtained at the sampling point with the kick net following either procedure, a sample may be hand picked from about 0.09 m2 of substrate at the sampling point. Place the contents of this hand picked sample into the appropriate "RIFFLE" or "POOL" bucket. |
| | Lotic-Erosional | 1. At each cross-section transect, beginning with Transect A, locate the assigned sampling point (Left, Center, or Right as you face downstream). Roll a die to determine if it is a left (L), center (C), or right (R) sampling point for collecting periphyton and benthic macroinvertebrate samples. A die roll of 1 or 2 indicates L, 3 or 4 indicates C, and 5 or 6 indicates R (or use a digital wristwatch and glance at the last digit (1-3=L, 4-6=C, 7-9=R). Mark L, C, or R on the Sample Collection Form. Assign sampling points at each successive transect in order as L, C, R after the first random selection.<br>2. Determine if there is sufficient current in the area at the sampling point to fully extend the net. If so, classify the habitat as lotic-erosional and proceed to Step 3. If not, use the sampling procedure described for Lotic-depositional habitats.<br>3. Record the dominant substrate type (fine/sand, gravel, coarse substrate (coarse gravel or larger) or other (e.g., bedrock, hardpan, wood, aquatic vegetation, etc.) and the habitat type for each kick net sample collected on the Sample Collection Form.<br>4. With the net opening facing upstream, position the net quickly and securely on the stream bottom to eliminate gaps under the frame. Avoid large rocks that prevent the sampler from seating properly on the stream bottom.<br>5. Holding the net in position on the substrate, visually define a rectangular quadrat that is one net width wide and one net width long upstream of the net opening. The area within this quadrat is 0.09 $m^2$.<br>6. Check the quadrat for heavy organisms, such as mussels and snails. Remove these organisms from the substrate by hand and place them into the net.<br>7. Hold the net securely in position while using both feet to vigorously agitate the substrate within the quadrat for 30 seconds (use a stopwatch).<br>8. After 30 seconds, hold the net in place with your knees and pick up any loose rocks within the quadrat. Use your hands to rub any clinging organisms off the rocks (especially those covered with algae or other debris) in front of the net. Also, place any additional mussels and snails found into the net. Remove the net from the water with a quick upstream motion to wash the organisms to the bottom of the net.<br>9. Invert the net into a bucket labeled “RIFFLE”, which is about half full of water, to rinse organisms out of the net. Inspect the net for clinging organisms. Use watchmaker’s forceps to remove any organisms from the net and place them in the bucket. Carefully inspect any large objects (such as rocks, sticks, and leaves) in the bucket and wash any organisms found off of the objects and into the bucket before discarding the object. Remove as much detritus as possible without losing any organisms.<br>10. Place an “X” in the appropriate habitat type and sampling point boxes for the |

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| | | transect on the Sample Collection Form.<br>11. Proceed upstream to the next transect and repeat Steps 1 through 9. Combine all kick net samples from lotic-erosional habitats into the "RIFFLE" bucket.<br>12. Process the sample following the steps presented in Macroinvertebrate Sample Processing (Table 3). |
| | Lotic-Depositional | 1. At each cross-section transect, beginning with Transect A, locate the assigned sampling point (Left, Center, or Right as you face downstream). Roll a die to determine if it is a left (L), center (C), or right (R) sampling point for collecting periphyton and benthic macroinvertebrate samples. A die roll of 1 or 2 indicates L, 3 or 4 indicates C, and 5 or 6 indicates R (or use a digital wristwatch and glance at the last digit (1-3=L, 4-6=C, 7-9=R). Mark L, C, or R on the Sample Collection Form. Assign sampling points at each successive transect in order as L, C, R after the first random selection.<br>2. Determine if there is sufficient current in the area at the sampling point to fully extend the net. If so, use the sampling procedure described for lotic-erosional habitats. If not, classify the habitat as lotic-depositional and proceed to Step 3. NOTE: If the pool is too deep (much more than 1 m) to sample safely at the designated spot, move downstream until a safe sampling spot is found.<br>3. Record the dominant substrate type (fine/sand, gravel, coarse substrate (coarse gravel or larger) or other (e.g., bedrock, hardpan, wood, aquatic vegetation, etc.) and the habitat type for each kick net sample collected on the Sample Collection Form.<br>4. Visually define a rectangular quadrat that is one net width wide and one net width long at the sampling point. The area within this quadrat is 0.09 $m^2$.<br>5. Inspect the stream bottom within the quadrat for any heavy organisms, such as mussels and snails. Remove these organisms by hand and place them into the net or into a bucket labeled "POOL".<br>6. Vigorously agitate the substrate within the quadrat with both feet while dragging the net repeatedly through the disturbed area just above the bottom. Keep moving the net all the time so that the organisms trapped in the net will not escape. Continue agitating 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 gloved hands and use the U.S. Standard #30 sieve to collect the organisms from the water in the same way the net is used in larger pools.<br>7. After 30 seconds, hold the net between your legs and partially submerged. Pick up any loose rocks within the quadrat. Rub or brush any organisms found on them into the net. Also recheck the area for any additional snails or clams and place them in the net.<br>8. Invert the net into a bucket labeled "POOL", which is about half full of water, to rinse organisms out of the net. Inspect the net for clinging organisms. Use watchmakers' forceps to remove any organisms from the net and place them in the bucket. Carefully inspect any large objects (such as rocks, sticks, and leaves) in the bucket and wash any organisms found off of the objects and into the bucket before discarding the object. Remove as much detritus as possible without losing any organisms.<br>9. Place an "X" in the appropriate habitat type and sampling point boxes for the transect on the Sample Collection Form.<br>10. Proceed upstream to the next transect and repeat Steps 1 through 8. Combine all kick net samples from lotic-depositional habitats into the "POOL" bucket.<br>11. Process the sample following the steps presented in Macroinvertebrate Sample Processing (Table 3). |
| | Pupal Exuviae | Chironomidae pupal exuviae samples will be collected according to Ferrington et al. (1991). This method of collection allows for |

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| | | sampling of chironomids from a broad range of microhabitat areas upstream of the sampling point (Ferrington et al. 1991).<br><br>1. Pupal exuviae collections will be made between cross-section transects A - B, E - G, and J - K. Sample downstream of eddies and slack water areas formed by emergent or partially submerged objects such as rocks, riparian vegetation, trees, aquatic macrophytes, and filamentous algae.<br>2. Sample these areas by dipping one edge of a shallow pan just beneath the water's surface and skimming the floating debris.<br>3. After the pan has filled with water, pour the contents through a U.S. Standard Testing Sieve # 80 with a mesh size of 180 μm.<br>4. Repeat Steps 2 and 3 for five minutes.<br>5. After sampling, rinse all detritus out of the sieve with 95% EtOH into a sample container.<br>6. Label the sample bottle with the following information: site ID, collection date, and sample type (15 min. P.E.).<br>7. Repeat Steps 2 – 6 in the other two sampling areas (E - G, and J - K). Sample contents from these two areas are composited with the first sample. |
| Discrete | | Macroinvertebrate samples from discrete habitats (uni-connected side channels, off-channel pools, and woody debris) are collected using a combination of USGS-NAWQA and USEPA EMAP protocols. |
| | Uni-connected side channels and Off-channel pools | 1. Record the dominant substrate type (fine, sand, gravel, coarse substrate (coarse gravel or larger) or other (e.g., bedrock, hardpan, wood, aquatic vegetation, etc.) and the habitat type for each kick net sample collected on the Sample Collection Form.<br>2. Visually define a rectangular quadrat that is one net width wide and one net width long at the sampling point. The area within this quadrat is 0.09 $m^2$.<br>3. Inspect the stream bottom within the quadrat for any heavy organisms, such as mussels and snails. Remove these organisms by hand and place them into the net or into an appropriately labeled bucket e.g. "OCPOOL" or "USCHANNEL".<br>4. Vigorously agitate the substrate within the quadrat with both feet while dragging the net repeatedly through the disturbed area just above the bottom. Continuously move the net so that the organisms trapped in the net will not escape. Continue agitating 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 gloved hands and use the U.S. Standard #30 sieve to collect the organisms from the water in the same way the net is used in larger pools.<br>5. After 30 seconds, hold the net between your legs and partially submerged. Pick up any loose rocks within the quadrat. Rub or brush any organisms found on them into the net. Also recheck the area for any additional snails or clams and place them in the net. Handpick invertebrates from various substrates when necessary to collect fragile or tightly adhered invertebrates.<br>6. Invert the net into a bucket appropriately labeled e.g. "OCPOOL" or "USCHANNEL", which is about half full of water, to rinse organisms out of the net. Inspect the net for clinging organisms. Use watchmaker's forceps to remove any organisms from the net and place them in the bucket. Carefully inspect any large objects (such as rocks, sticks, and leaves) in the bucket and wash any organisms found off of the objects and into the bucket before discarding the object. Remove as much detritus as possible without losing any organisms.<br>7. Place an "X" in the appropriate habitat type and sampling point boxes on the Sample Collection Form. |

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|---|---|---|
| | | 8. Process the sample following the steps presented in Macroinvertebrate Sample Processing (Table 3). |
| | Woody Debris | 1. Identify at least five (if present) woody snag locations present throughout the reach. Woody snags that are suitable for sampling should have been submerged for an extended period during which there is clear evidence of invertebrate and periphyton colonization.<br>2. At each woody snag location, select at least two snags that will be sampled. [Note: depending on the size of the woody snags, additional snags may need to be sampled to obtain enough sample material for laboratory processing.] However, woody debris can be found in clumps of several small(er) diameter snags. When this is the case, sample the entire clump.<br>3. Place a collection net downstream of the woody snag to capture dislodged invertebrates.<br>4. Vigorously disturb the woody debris for 30 seconds with hand, foot, brush and/or net to dislodge invertebrates.<br>5. Place sample contents into a 19-L (5-gal) plastic bucket labeled "Woody Debris".<br>6. To sample for macroinvertebrates that may be living inside the woody debris, place a collection net downstream of the woody snag to capture and minimize loss of mobile or loosely attached invertebrates.<br>7. Remove two 0.03 m sections of the woody snags by using a pruning shears.<br>8. Remove attached organisms by handpicking, brushing, and rinsing the surface of each cut section of the branch into a 19-L (5-gal) plastic bucket. After the initial brushing and rinsing process is complete, place the woody snags in a separate, empty bucket for about 1 hour. As the snags dry in the bucket, missed organisms will crawl out of crevices and then can be collected. Add these organisms to the rest of the sample material.<br>9. Determine the substrate type where the woody snag was positioned. Measure the depth from the surface of the water to the woody snag and stream bottom, and current velocity. Record these data on the Sample Collection Form.<br>10. Combine the sample material collected from each woody snag and process the composite sample following the steps presented in Macroinvertebrate Sample Processing (Table 3). |

**Table 10-3 Methods for processing macroinvertebrate samples**

| |
|---|
| Macroinvertebrate sample processing has been adapted and modified from the USGS (Moulton et al. 2002). Procedures for reducing sample volume to 750 mL have been eliminated. Inorganic debris is reduced as much as possible by handpicking, washing, swirling, and decanting liquid contents and organic debris through a 500-µm mesh sieve. Organic debris, such as leaves and small pieces of wood, are washed and discarded. Organic debris that cannot be washed, filamentous algae, and tiny pieces of debris, are deposited in the sample container. |
| 1. Remove large debris (e.g., cobbles, filamentous algae, leaves, twigs) from each discrete collection and inspect for attached invertebrates, especially small, cryptic invertebrates (e.g., microcaddisflies, water penny beetle larvae).<br>a. Carefully remove any attached invertebrates by rinsing and using forceps, and place them in the appropriate sample bottle.<br>b. Discard the large debris.<br>2. Elutriate each discrete collection onto a 500-µm mesh sieve to separate invertebrates and organic debris from the inorganic debris. Doing this will minimize damage to organisms and maximize effective elutriation.<br>a. Place each discrete collection in a separate 19-L (5-gal) plastic bucket filled about one-third full with water.<br>b. Carefully stir the contents of each bucket by hand to suspend as much of the sample material as possible.<br>c. Swirl the suspended sample material and carefully decant it onto a 500-µm sieve that is held over a dishpan. The dishpan provides secondary containment to catch any decanted sample that is spilled outside of the sieve. Pour any sample spilled in the dishpan directly onto the sieve unless it includes a large volume |

of inorganic debris. [Note: use a large-diameter (30-cm) sieve to facilitate subsequent sieving of the sample to remove fine sediments.]

d. As decanting proceeds, watch the advancing inorganic sediment front to avoid pouring it onto the sieve.
e. Stop decanting when the inorganic sediment front reaches the lip of the bucket.
f. If sample is spilled in the dishpan, pour the dishpan water (except fine sediment) back into the 19-L (5-gal) bucket. Otherwise, discard the dishpan water.
g. Repeat elutriation about three to five times for each discrete collection until the inorganic debris (elutriate sample component) appears free of detritus and organisms. Check inorganic debris for heavy organisms. [Note: heavy organisms, such as mollusks and some case-building caddisflies, might settle too fast during decantation and will not be able to be elutriated.]
h. Set the elutriate sample component contained in the bucket aside for later inspection.
i. Invertebrates and organic debris that are retained on the 500-µm sieve represent the composited main-body sample component.
j. Repeat the bucket elutriation for each discrete collection by elutriating onto the same sieve. Ensure sieve has been thoroughly rinsed before processing a different habitat sample.

3. Inspect the elutriate sample component remaining in the bucket for invertebrates, particularly heavy organisms, such as mollusks and some case-building caddisflies, that often do not decant onto the sieve during elutriation.
   a. Place a small amount of the elutriate sample component in a shallow white tray containing enough water to float debris to facilitate detection of invertebrates. Alternatively, the elutriate sample component can be inspected in a sieve.
   b. If necessary, sieve the elutriate sample component through a pair of nested large-meshed (e.g., 2- and 4-mm) sieves to separate larger gravel and pebble material and expedite inspection and removal of invertebrates.
   c. Place cryptic invertebrates (e.g., elmid beetle larvae, case-building caddisflies) in the appropriate habitat sample bottle.
4. Inspect the debris for large, rare invertebrates (e.g., mussels, crayfish, hellgrammites) that might prey upon or damage other invertebrates in the collection.
   a. Remove only these organisms and place them in a separate bottle appropriately identified with the habitat sample it came from. Match the bottle size to the size and quantity (typically no more than 20) of organisms. [Note: if these organisms are not present in the collection, then do not prepare a large-rare sample component.]
   b. Do not add clumps of sample debris to the bottle. This debris often contains other invertebrates, especially early instar specimens, and interferes with laboratory analysis.
5. Wash the sample component remaining on the 500-µm sieve by dipping and swirling the sieve repeatedly in the stream or a dishpan. Washing and sieving removes fine sediment from the sample component, which facilitates laboratory analysis.
6. Place a sample label and sample component in the sample bottles. Labels should be completed only with archival quality India ink, or pencil, on 100% cotton acid free paper and are placed within the jar. Any label placed on the outside of the jar is only for convenience in managing samples. All samples must have a label on the inside of the sample container. Fill sample bottles with sample contents no more than 1/2 full. Reserve half the sample container for preservative.
7. Fill sample bottle to the top (as little headspace as possible, without over filling).
8. Record collection and sample information on the appropriate field data sheet corresponding to a particular sample type.

### 10.1.3 Lakes and Reservoirs

Lake and reservoir sampling methodologies are being evaluated.

### 10.1.4 Wetlands

For wetlands monitoring, EPA (2002) recommends:

- Sampling a stratum of the wetland that contains most of the macroinvertebrates: often the shallow areas have emergent or submerged vegetation; very small wetlands can be sampled all around the edges.
- Sampling once during the season, after determining when the maximum development of invertebrates occurs, and take more than one sample at that time; if resources permit, sample in two seasons taking more than one sample on each visit date.
- Sampling all habitats within the stratum or zone, or sample selected habitats if they are known to have a wide representation of invertebrates.
- Using a dip-netting method with a standardized and repeatable protocol; combine this with activity traps to collect the motile predators (see discussion of these and other methods below).

A wetlands sample index period has not been investigated in New Mexico. However, given the constraints of the hydrograph, staff timing, and program resources, the sample index for macroinvertebrates will likely be June – August. Methods are provided below for both dry and wet sampling.

#### *10.1.4.1 Sample Location Designs*

Wetlands will have five sampling areas that should be placed perpendicular to the stream/river, or from edge of wetlands, and spaced proportionally from one end of the wetland to the other (See Figure 10-5 and Figure 10-6).

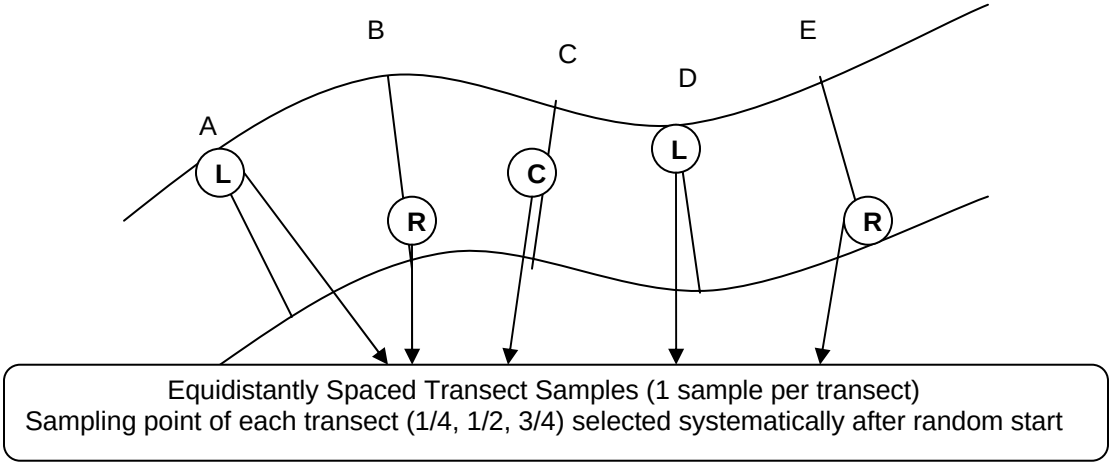


**Figure 10-5 Sample design for riverine type wetlands**

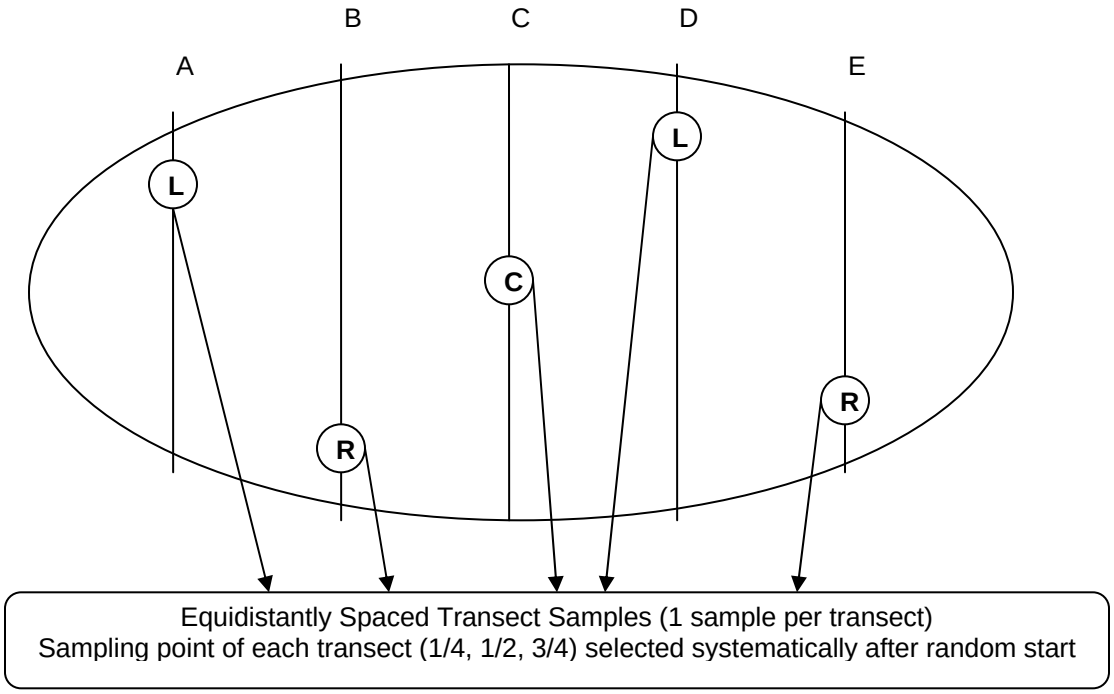


**Figure 10-6 Sample design for depressional type wetlands**

If the wetland is longer than 250 m in length, establish transects no more than 50 m apart. Establish five equidistantly spaced transects perpendicular to the bank. Randomly select the sample location, left, center, or right, on the first transect. Sample at location using the appropriate habitat (emergent/submergent vegetation, substrate, woody debris) sampling methodology and note on field form what habitat types were sampled. If not wadeable, establish one end of transect at a water depth of 1 m with the other end of transect at the edge of the wetland (See Figure 10-7).

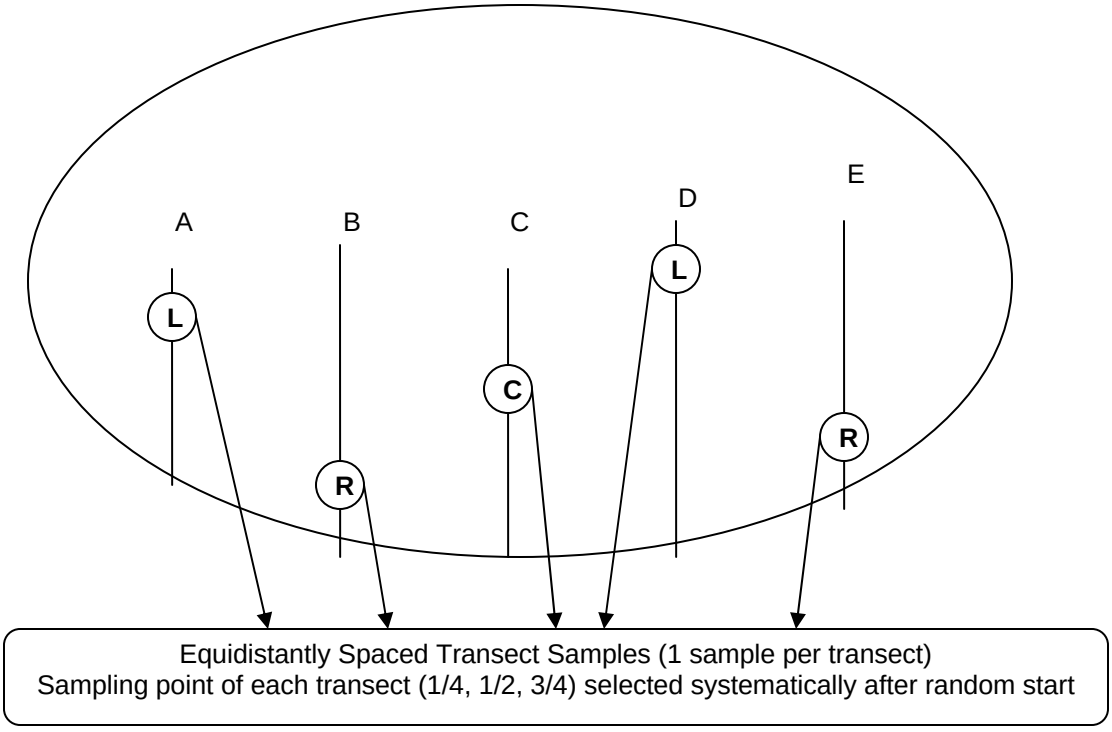


**Figure 10-7 Sample design for non-wadeable wetlands**

*10.1.4.2 Wetland Sampling and Processing Methods*

Given the hydrology of wetlands and the arid climate in New Mexico there is, in many instances, little or no standing water. Typically when there is standing water it is only present during a brief time in early spring and/or monsoon season, July - October. Sampling methods for the various types of wetlands are listed in Table 4.

Riverine, depressional, and slope wetlands that are not inundated should have macroinvertebrate samples collected from leaf litter and/or shallow groundwater from the hyporheic zone. Diverse macroinvertebrate assemblages have been documented to occur in the hyporheic zone with substrates ranging from sand to cobble (Case 1995, Ferrington 1984, Ferrington and Goldhammer 1992, Goldhammer and Ferrington 1992, Olsen and Townsend 2003, Palmer 1990, Stanford and Ward 1993, Stanford et al. 1994, Stanford and Ward 1988).

**Table 10-4 Sampling methods for different wetlands (wet or dry)**

| | | Sampling Method | | | | | |
|---|---|---|---|---|---|---|---|
| | | Transects | | | | HGM Vegetation Plot | Multi-habitat |
| | | Hyporheic | Emergent/ Submergent Vegetation | Substrate | Woody Debris | Leaf Litter | Live Trap |

| | | | | | | | |
|---|---|---|---|---|---|---|---|
| Riverine | inundated | X | X | X | X | | X |
| | Not inundated | X | | | | X | |
| Depressional | inundated | | X | X | | | X |
| | Not inundated | | | | | X | |
| Slope | inundated | X | X | X | X | | X |
| | Not inundated | X | | | | X | |
| Vernal Pool | inundated | | X | X | | | X |
| Fens/Bogs | inundated | | X | X | X | | X |

The utilization of leaf litter macroinvertebrates for wetlands assessment is a relatively new tool and has been shown to respond to a range of wetland conditions (Hatfield et al. 2006). Pioneering investigations have been done in New Mexico with regards to leaf litter invertebrates in riparian wetland areas (Cartron et al. 2003, Bess et al. 2002, Ellis et al. 2002, Ellis et al. 2001, Ellis et al. 2000).

All wetlands that are inundated should have macroinvertebrate samples collected from habitat types on the transect, emergent/submergent vegetation with sweep nets, substrates with kick net, and water column with live traps. The sweep net is the same net as the kick net and is 12 in wide with a mesh size of 500 μm. Ten minute chironomid pupal exuviae samples will also be collected.

A completed sample label is required for all sample jars from a single composite sample. An example label for use inside of the benthic macroinvertebrate sample jars is presented in Figure 8. Sampler types (and Habitat Type) will be one of the following Core (hyporheic), Pump (hyporheic), Kick net (sub/emergent vegetation, substrate) Shovel (leaf litter), or Live Trap (water column). Labels should be completed only with archival quality India ink or pencil on 100% cotton acid free paper and are placed within the jar. Any label placed on the outside of the jar is only for convenience in managing samples. All samples must have a label on the inside of the sample container.

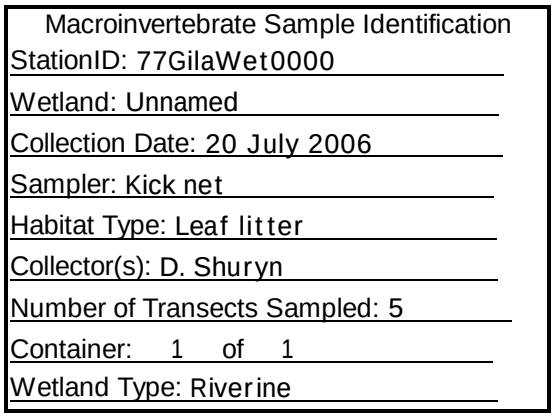


**Figure 10-8 Example label for wetland macroinvertebrate sample jar**

*10.1.4.3 Hyporheic Zone*

Methods for hyporheic zone sampling are being investigated. Likely methods include pumping water from 20 – 40 cm depths and filtering or core freezing and extraction of the first 20 – 40 cm.

**Table 10-5 Advantages and disadvantages of core freezing versus pumping.**

| Method | Advantages | Disadvantages |
|---|---|---|
| Pumping | o Ease of use<br>o Relatively simple equipment | o Under samples taxa richness<br>o Selective for small(er) invertebrates<br>o May be slow process depending on infiltration rate |
| Core Freeze | o Collects all invertebrates in freeze area<br>o Provides core profile | o Difficulty of extraction<br>o Special equipment including liquid nitrogen |

Hunt and Stanley (2000) and Scarsbrook and Halliday (2002) provide excellent analyses and comparisons of these two methods.

*10.1.4.4 Emergent/Submergent Vegetation*

Vegetation sampling should be done by using the kick net as a sweep net. Vigorously agitate the vegetation with the kick net by sweeping through vegetation for approximately one meter using a forward jabbing motion. Then quickly sweep the net through the water column in the disturbed area. Repeat this step quickly three or four times. Empty net contents into a 19 L (5 gal) bucket labeled "vegetation". After all sampling is complete, evenly distribute material from the vegetation bucket onto a 6 mm (¼ inch) mesh wire screen. Make sure material is distributed so that macroinvertebrates can crawl and drop off of material through the screen. Leave suspended for approximately five minutes over a cooler with approximately 25 mm (1 in) of water in the bottom. Turn coarse material over and let stand for another five minutes. Material should not be allowed to dry completely. Remove the pan of water and replace with a 500 μm mesh sieve. Rinse material thoroughly over the sieve and place back on screen. Place material in sieve into

sample container. Inspect material for any attached cases or macroinvertebrates, pick off and place into sample container. Discard vegetation back into wetland.

#### *10.1.4.5 Substrate*

Substrate sampling should be performed by jabbing the substrate vigorously in a forward direction and to a depth of approximately one centimeter for a distance of one meter. Then quickly sweep the net through the water column above and around the disturbed substrate. Repeat this step quickly three or four times. Empty net contents into 500 micron mesh sieve and wash sample repeatedly until fine sediments are gone. Return contents to a 19 L (5 gal) bucket that is ⅓ full of water. Elutriate organic debris from sediment by swirling the bucket around and quickly pouring off suspended material onto the sieve. Repeat until all organic debris has been removed. Carefully inspect sediment for cases, shells, snails, bivalves, and large invertebrates and remove and place in sample container. Evenly distribute organic material on a 6mm (¼ in) mesh wire screen. Make sure material is distributed so that macroinvertebrates can crawl and drop off of material through the screen. Leave suspended approximately five minutes over cooler with approximately 25 mm (1 in) of water in bottom. Turn coarse material over and let stand for another five minutes. Material should not be allowed to dry completely. Remove the pan of water and replace with a 500 μm mesh sieve. Rinse material thoroughly over a 500 μm mesh sieve and place back on screen. Place material on sieve into sample container. Inspect material for any attached cases or macroinvertebrates, pick off and place into sample container. Discard substrate back into wetland.

#### *10.1.4.6 Woody Debris*

Woody debris sampling should be done by vigorously agitating the woody debris with the kick net and rapidly sweeping the kick net through the water column near the disturbed area several times to capture dislodged macroinvertebrates. Additionally, 5 pieces of woody debris should be picked up and placed on a 6 mm (¼ in) mesh wire screen suspended over a cooler of water. If possible, break woody debris into smaller pieces and make sure material is distributed so that macroinvertebrates can crawl and drop off of wood pieces through the screen. Let stand ten minutes suspended over cooler with approximately 25 mm (1 in) of water in the bottom. Remove the pan of water and replace with a 500 μm mesh sieve. Rinse material thoroughly over a 500 μm mesh sieve and place back on screen. Place material on sieve into sample container. Inspect woody debris for any attached cases or macroinvertebrates, pick off and place into sample container. Discard woody debris back into wetland.

#### *10.1.4.7 Leaf Litter*

For macroinvertebrate sampling, two plots are randomly selected from each of the vegetation transects or plots. Intensive vegetation sample plots are excluded due to disturbance associated with vegetation sampling. In each of the randomly selected plots, vegetation type, and cover are evaluated and macroinvertebrate plots are placed so as to represent heterogeneity within the larger 10 × 10 m plot. A total of four 0.5 $m^2$ macroinvertebrate plots are placed in each larger 10 × 10 m plot. Leaf litter sampling methods have been modified from Hatfield et al. (2006).

Within each of the 0.5 $m^2$ plots, all duff layer material within the plot is collected. Soil and large plants are not collected. When present, roots are collected, but excess vegetative matter is discarded. All rotting log and twig pieces in the square are broken apart (over the container) and

collected. Samples are then placed in a loose-weave cotton bag with an identification label and stored in the shade until transported to the lab.

In the lab, litter samples are placed into Berlese funnels. The funnels are constructed of a 70 cm tall by 25 cm diameter metal cylinder. A 6.35 mm (¼ in) mesh screen is placed 35 cm from the top of the cylinder and a collection funnel is placed below the screen. Location, time, and depth of litter in the funnel are recorded. A 75 watt light bulb is placed over the funnel and a collection jar containing 95% EtOH is placed under the funnel. The funnels are kept in place and checked daily, until the litter is dry. Once the litter is dry, check for organisms, then discard litter and dismantle the funnel. Retrieve specimen jars, decant alcohol, and replace with fresh 95% EtOH.

For samples where the litter is deep, the top surface of dry litter is carefully removed exposing the damp litter underneath. Funnels are also checked to ensure they are not clogged. The same procedure is done when litter is completely dry. In the event that samples collected are not placed in Berlese funnels the same day, they should be stored in a cold area (cooler) and held for a maximum of two days.

#### *10.1.4.8 Live Trap*

Previous studies have shown that dip net sampling captures the greatest richness of invertebrates, but the actively swimming or night active predators may be under represented by using this method alone (Helgen et al. 1993, Serieyssol et al. 2005). Therefore, activity traps are placed in the wetland for 24 hours to collect active night predators and swimmers. The bottle trap method described below was modified from Minnesota Pollution Control Agency (MPCA 2006).

Bottle traps work as passive funnel traps to collect organisms that swim into the funnel and pass through the neck into the bottle. The traps are made from clear 2-liter plastic beverage bottles free of labels or opaque parts. Traps must be completely clear so that they are nearly invisible underwater. The bottle traps are designed with four two inch grooves cut into the funnel edge by a hot wire to allow the funnel to be inverted and snap into the bottle opening without the use of clips or visible straps (Figure 9). The traps are supported on a 1.2 m (4 ft) rod, and attached with a flexible half section of 3 in (72 mm) thin wall PVC pipe, which allows for raising or lowering the activity trap on the rod (Figures 9 and 10).

If wetland size allows, ten bottle traps are placed in each wetland for 24 hours within the near shore zone, preferably in emergent vegetation, if any exists. The ten activity traps are set out in pairs, with each trap in a pair located approximately 3 - 4 m apart and on the same transect. In the shallowest water (15 cm), the traps are placed just under the surface of the water, but should not be resting on the bottom to avoid filling the bottle trap with sediment. In deeper water (> 50 cm), traps are placed horizontally about 15 - 20 cm under the surface. Traps are not placed at the deeper edge of the vegetation in the open water area because capture efficiency goes down as the water gets deeper. The traps are backfilled with water leaving no air bubbles inside in order to reduce predation within the trap. The wingnut should be tightened enough so the trap remains horizontal (see Figure 10a).

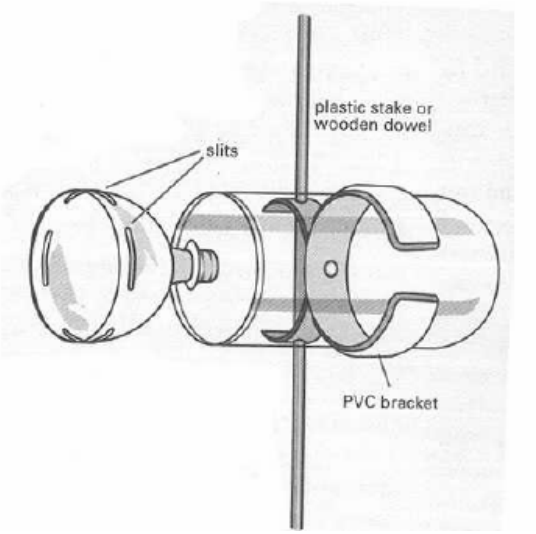


**Figure 10-9 Bottle trap design illustrating adjustable PVC bracket and funnel grooves (from Minnesota Pollution Control Agency).**

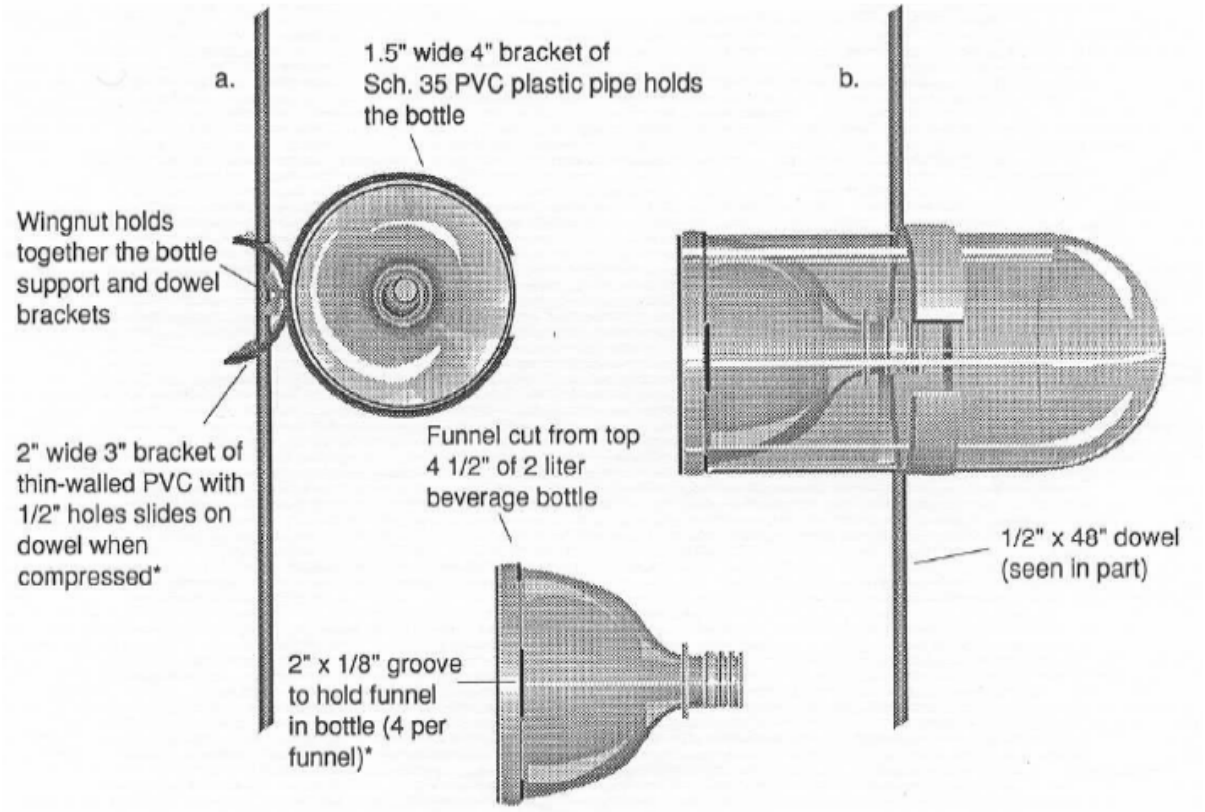


**Figure 10-10 Bottle trap design, a.) view into funnel, b.) lateral view**

**(from Minnesota Pollution Control Agency).**

After the required 24 hour period, the traps can be collected by slightly loosening the wingnut in order to rotate the trap to a vertical position and slide it up the dowel by slightly compressing the dowel clamp. Then the funnel is removed and the contents of the trap are poured through a 200 μm sieve. The trap is squirted with tap water and the inside is rubbed to dislodge leeches and other invertebrates. Specimens attached to both faces of the funnel opening are also considered part of the sample. These dislodged specimens are then added to the contents of the sieve. The second trap of the pair is collected and its contents are poured into the same sieve. The sieve is back-flushed into a sample jar with 100% alcohol to a final concentration of 80%. Divide the sample between multiple jars (sample #, jar 1 of 2, jar 2 of 2, etc.) so that the jar has not more than 1/3 volume of invertebrates to alcohol. Labels with India ink or pencil on 100% cotton acid free paper are placed within the jar. Any label placed on the outside of the jar is only for convenience in managing samples.

## 10.2 Fish Health

The fish health portion of the bioassessment provides data that could be used to determine the presence of environmental contaminants and their effects on fish, warrant further investigations into fish consumption advisories, and determine overall biotic integrity of aquatic systems.

Biological integrity of aquatic systems can be reflective of overall health of organisms within that system. Because of their position in the food chain, fish are regarded as representative indicators of overall system health (Adams et al. 1992). Impairments of fish health may indicate contaminants within the aquatic ecosystem, some of which remain undetected by water quality monitoring. Changes in overall condition of the organism may corroborate findings from other biomarkers and environmental contaminant analysis.

The methods and field procedures used in this protocol were developed and used in U.S. Geological Survey (USGS) Biomonitoring of Environmental Status and Trends (BEST) program projects that began in 1995 (Schmitt et al. 1995) and 1997 (Bartish et al. 1997) from data collected at National Contaminant Biomonitoring Program (NCBP) stations (Schmitt et al. 1999), USGS National Water-Quality Assessment (NAWQA) Program (Hirsch et al. 1988) sites, and selected National Stream Quality Accounting Network (NASQAN) (Hooper et al. 1996) sites in the Mississippi (Schmitt 2002), Columbia (Hinck et al. 2004), Yukon (Hinck et al. 2004), and Rio Grande (Schmitt et al. 2004, 2005) basins. These health assessments are also developed from procedures described by Goede and Barton (1990).

Monitoring of fish health is performed at selected water quality sites. These site locations, rationales, selected data collection and analysis methods, and additional details are documented in study plans. Collected fish are examined for external and internal anomalies. Organ weights are used to calculate somatic and health assessment indices. Samples from organs, abnormal tissue, and plasma are submitted for laboratory analyses. Generally, studies have both field and laboratory components. This section addresses only the field portions with mention of suggested laboratory methods and the fish tissues involved.

### 10.2.1 Animal Welfare

The procedures described in this document adhere to the American Society of Ichthyologists and Herpetologists, American Fisheries Society, and American Institute of Fishery Research Biologists, "Guidelines for Use of Fishes in Field Research" (ASIH et al. 1998).

### 10.2.2 Equipment and Supplies

Equipment and supplies needed to conduct the procedures outlined in this section are listed in Table 10.1. The amount of supplies will depend on the number of sampling sites and number of fish, which should be indicated in the study plan.

**Table 10-6 Equipment and supplies for fish health bioassessment**

| | |
|---|---|
| **Safety** | |
| Disposable Gloves | 40 pair of various sizes |
| Canvas Gloves | 2 pair |
| Raingear, boots, waders, etc. | as needed |
| Safety glasses | 2 pair |
| **Data Collection and Reporting** | |
| Camera | 1 |
| Clipboards | 2 |
| Fish Health Examination Sheet | 1 per fish |
| Station Identification Sheet | 1 per fish |
| Sharpie (permanent marker) | 2 |
| Ball-point | 2 |
| Cryogenic | 1 |
| Pencils | 3 |
| SOP, sampling guide, study plan, field notebook | 1 each |
| **Cleaning** | |
| Acetone (reagent grade) | 0.5L, for rinsing instruments |
| Acetone squirt bottle | 1 |
| Beaker | 1, for acetone rinse |
| Bleach | optional, for disinfecting |
| De-ionized water | 4L, for rinsing instruments |
| Plastic trash bags | for waste disposal |
| **Fish Processing** | |
| Aluminum foil | 300ft, heavy duty |
| Bottles, 125mL polyethylene w/leak-proof tops | 1 per fish, histopathology samples |
| Ethanol, 100% | 1.5L |
| Fixative (10% neutral buffed Formalin) | 85mL, for histopathology bottles |
| Heparin solution | sodium salt, 10USP |

| | |
|---|---|
| | units/mL, optional |
| Needles (hypodermic) | 1 per fish, size may vary with species and size of fish |
| Sharps container (for used needles) | 1 |
| Scale envelopes | 1 per fish, for scales or spines |
| Small towels or cloth diapers | 6 |
| Syringes | 5.0mL, 1 per fish |
| Transfer pipettes | 1 per fish, for plasma |
| Vacuum containers | 1 per fish, for blood |
| Weigh boats | 1 per fish, small and large |

**Equipment**

| | |
|---|---|
| 120V AC power source (>1 amp) | 1 inverter, or outlet, for centrifuge |
| Blunt instrument for subduing fish | 1 |
| Calibration weights | 1 of appropriate mass for each balance |
| Centrifuge | 1, only if plasma is to be collected |
| Centrifuge tube rack | 1 |
| Dissecting tools | 1 kit |
| Electrofishing equipment | 1 |
| Ground-Fault Interrupting (GFI) extension cord | 1 |
| GPS unit | 1 |
| Hanging balance, for large fish | 1 |
| Measuring board (mm) | 1 |
| Portable electronic balances | 1 |
| Tubs or live wells, for holding fish | # and capacities varies with study plan |

**Fish Tissue Packaging**

| | |
|---|---|
| Coolers | # and capacities varies with study plan |
| Cloth tags | # varies with study plan |
| Cryogenic vials | # varies with study plan |
| Cryogenic vial storage boxes | # varies with study plan |
| Dry ice | amount varies with study plan |
| Dry ice cooler | # and capacities varies with study plan |
| Wet Ice | amount varies with study plan |
| Plastic bags (for carcasses/filets) | # and capacities varies with study plan |

Wide-mouth thermos (for quick freezing) 1

**Miscellaneous**
Cell phone
Duct tape
Field guides and other references
Fire extinguisher
First-aid kit
Flashlights
Headlamps
Material Safety Data Sheets (MSDS) for chemicals
Paper towels
Pliers
Portable table and chairs
Shade canopy
Thermometer/Sonde

### 10.2.3 Data Collection and Analysis

*Collection and analyses methods can vary depending on the individual study plan.*

- The target taxa and total number of fish per site should be determined in the study plan or protocol of the specific project. Larger, adult fish should be collected because it is easier to obtain the necessary amounts of tissues and blood, observations may be difficult to perform with small fish, and many biomarkers are neither valid nor well documented in juvenile fish.

- Collection efforts are conducted under stable flow conditions. Stable flow conditions are those with limited fluctuations.

- Collect fish by pulsed DC electrofishing and/or seining. Hold collected fish in live wells until processed. Process collected fish within a few hours after capture.

- At each site, examine collected fish and note any external and internal anomalies. Record species, length, weight, gender, and observations from the field examination on the Fish Health Examination datasheet (Appendix A). Complete a separate Fish Health Examination sheet for each specimen. The sex of each specimen is verified upon internal examination. Measure and record the weights of the liver (only for fish with a discrete liver, such as salmonids), gonads, and spleen.

- If blood samples are taken, keep them on ice until centrifuged. Once an even number of blood samples have been collected, centrifuge the uncapped vacutainers® at 3500 rpm for 10 minutes. Aspirate the plasma into a cryovial using a transfer pipette . Flash freeze the cryovials in a dry ice slurry and then keep frozen on dry ice. Maintain plasma samples

at -80º C until processed. During internal examination, remove samples of the liver, gonads, spleen, and kidney and preserve in 10% neutral buffered formalin for histopathological analysis. These analyses include microscopic examination of preserved tissues for the presence of lesions, which can provide an early indication of chemical exposure and assist in determining overall health of the organism (Hinton at al. 1992; Hinton 1993; Goodbred et al. 1997). Also, cryogenically freeze an additional liver sample from each fish for hepatic ethoxyresorufin-O-deethylase (EROD) activity analysis, a method of enzyme induction by planar hydrocarbons to detect the presence of polychlorinated biphenyls (PCBs), polycyclic aromatic hydrocarbons (PAHs), dioxins, and furans (Pohl and Fouts 1980; Kennedy and Jones 1994).

- Whole fish or filets may be kept for future analyses. For each fish, wrap the carcass and remaining tissues or filet in aluminum foil, label, and keep frozen in a -80 ºC freezer. Composit carcasses by station-species-gender. Future composite sample analyses may include organochlorine chemical residues, elemental contaminant concentrations, H4IIE rat hepatoma cell bioassay (used as a screening tool to determine the presence of PCBs, PAHs, dioxins and furans) (Tillitt et al. 1991), and stable isotopes of nitrogen (δ15N) (used to determine trophic position and nitrogen sources) (Cabana and Rasmussen 1996).

- Calculate organo-somatic indices for all fish sampled. Indices are generally expressed as percentages of total body weight to organ weight, calculated as [(organ weight/body weight)*100] (Busacker et al. 1990). Measurements of the liver, spleen, and gonads relative to fish length or weight may signify overall health and reproductive status. These indices can reflect the status of organ systems, which may change in size due to environmental factors more rapidly than organism weights and lengths increase or decrease (Busacker et al. 1990). Calculate condition Factor (CF) and Health Assessment Index (HAI) and utilized as general indicators of fish health (Carlander 1969, 1977). CF is the relationship of body weight to length, computed as weight/length3. This equation reflects the expected exponential gain in weight relative to length as fish grow. CF is an organism level response, reflecting increases or decreases in weight to factors such as nutrition, toxic chemical exposure, and pathogens (Schmitt and Dethloff 2000). The quantitative HAI utilizes observations of gross external and internal pathological disorders (Goede 1988, 1989, 1996; Adams et al. 1993). Studies have indicated that fish in severely polluted areas exhibit a higher frequency of gross lesions than those in similar, less polluted areas (Schmitt and Dethloff 2000). Developed and revised by Goede (1988, 1989, 1996), this systematic necropsy-based system has been used by fisheries personnel in the field and hatchery situations.

**Table 10-7 Selected methods for laboratory analyses**

| Method | Description | Tissue(s) examined | Sensitivity | Primary reference(s) |
|---|---|---|---|---|
| Histopathology | Microscopic examination for the presence of lesions; can provide early indication of contaminant exposure | Liver, gill, gonads, spleen, and kidney | Overall organism health and contaminants | Hinton et al. (1992); Hinton (1993); Goodbred et al. (1997) |

| Method | Description | Tissue(s) examined | Sensitivity | Primary reference(s) |
|---|---|---|---|---|
| Ethoxyresorufin-O-deethylase (EROD) activity | Enzyme induction by planar hydrocarbons | Liver | PCBs, PAHs, dioxins, and furans | Pohl and Fouts (1980); Kennedy and Jones (1994) |
| Macrophage aggregate (MA) analysis | Macrophages are important in the immune system, serving as a first line of defense for the organism and as an antigen processing cell | Spleen, hemopoetic kidney, and liver | Multiple contaminants including PAHs and metals | Blazer et al. (1994); Blazer (1997) |
| Vitellogenin (vtg) | A precursor of egg yolk, normally synthesized in the liver of female fish | Blood plasma | Endocrine modulating substances | Folmar et al. (1996) |
| Sex steroids (estrodiol and testosterone) | Determine reproductive health and status | Blood plasma | Endocrine modulating substances | Guillete et al. (1994); Goodbred et al. (1997) |
| Somatic indices | The relative mass of some organs is often indicative of chemical exposure | Gonads, spleen, and liver | Overall organism health | Grady et al. (1992) |
| Necropsy-based fish health assessment | Visual assessment of external/internal anomalies (e.g., lesions, parasites, tumors) | All | Overall organism health | Goede (1988, 1996); Adams et al. (1993); Adams (1990) |
| H4IIE bioassay | A screening tool to determine the presence of certain classes of planar halogenated compounds | Whole fish (composites) | PCBs, PAHs, dioxins, and furans | Tillitt et al. (1991) |
| Chemical analyses | Organochlorine chemical residues and elemental contaminants | Whole fish (composites) | Specific analytes | Schmitt et al. (1999) |

(Schmitt and Dethloff 2000).

### 10.2.4 Examination Procedures (Schmitt et al. 1999)

Before processing fish (procedures may vary by study plan):

1. Pre-label tags, vials, etc. Each fish should be assigned a unique identification number.
2. Setup the outdoor laboratory in the most level, shady, wind-sheltered area.
3. Calibrate balances.
4. Prepare fixatives and dry-ice slurry*.
5. Initiate a Station Identification sheet for the day's collection efforts (Appendix B).
6. If collecting plasma, treat the needles and syringes with heparin** (not needed if collecting serum).
7. Keep fish processing area clean.

*Dry-ice slurry for flash-freezing cryovial samples: use crushed dry-ice mixed with ethanol to create a slurry. Keep the mixture in a wide-mouth thermos. Keep the thermos closed to reduce evaporation.

**Heparin is a powerful anti-coagulant. Treat the needle and syringe by uncapping the needle and drawing a few millimeters of heparin solution into the syringe. Invert the syringe and pull the plunger to its fullest extent. This will coat the syringe. Dispense the heparin back into its container.

### 10.2.5 Dissection and Sample Preservation

External and internal examinations are performed by a trained fish biologist, in accordance with established procedures developed and implemented by the U.S. Geological Survey (Schmitt et al. 1999; Smith et al. 2002). The following is a summary of published procedures.

For protection against pathogens and parasites associated with either fish or water, disposable latex or vinyl gloves should be worn by persons handling the fish and/or tissues. A new pair of gloves should be used for each fish processed.

**NOTE**: The order in which the procedures are performed is important to ensure that activities conducted in one step do not compromise observations in subsequent steps. Procedures described here conform to the organization of the Fish Health Examination datasheet upon which the observations are recorded.

A) Blood Collection

1. Secure a live fish from holding tank.
2. Use wet lint-free cloth towel, or cloth diaper to wrap the fish.
3. Lay the fish on its side and expose the caudal peduncle. The posterior caudal artery and vein lie together just ventral to the vertebral column.
4. Remove a few scales from the narrowest portion of the caudal peduncle.
5. Insert the needle from the side, just below the lateral line. The needle should be inserted to about mid-sagittal depth, between the hemal arches and spines. Exert slight suction. Fill the syringe with blood. Record the volume of blood collected on the Fish Health Examination datasheet.
6. Remove the needle from the syringe and dispense the blood into a chilled vacutainer®. Keep blood on ice until centrifuged.

B) External Examination

1. Prepare work surface by covering it with a piece of aluminum foil (dull side up/shiny side down) large enough to wrap the fish being processed. Remember to use a new piece of foil for each fish processed.
2. Expose the anterior dorsal surface of the fish and, using a blunt instrument, subdue the fish with a sharp blow to the rear of its head.
3. Record the species on the Fish Health Examination datasheet.
4. Record the weight (g) and total length (mm) on the Fish Health Examination datasheet.
5. Observe each eye and record condition on the Fish Health Examination datasheet. Check all that apply: normal, exopthalmic, hemorrhagic, opaque, missing, emboli, other. If a sample of abnormal tissue is collected, place it in fixative and record number of pieces on the datasheet.

6. Observe the head and body surface and record condition on the Fish Health Examination datasheet. Check all that apply: normal, tumors, lesions, parasites, other. If a sample of abnormal tissue is collected, place it in fixative and record number of pieces on the datasheet.
7. Observe both opercula and record condition on the Fish Health Examination datasheet. Check all that apply: normal, slight shortening, severe shortening, other. If a sample of abnormal tissue is collected, place it in fixative and record number of pieces on the datasheet.
8. Observe both the gills and record the condition of each on the Fish Health Examination datasheet. Check all that apply: normal, frayed, clubbed, marginate, pale, parasites, other. If a sample of abnormal tissue is collected, place it in fixative and record number of pieces on the datasheet. A **normal** sample of gill should also be taken and placed in fixative.
9. Observe each fin and record the condition of each on the Fish Health Examination datasheet. Check all that apply: normal, mild erosion, severe erosion, frayed, hemorrhagic, emboli, other. If a sample of abnormal tissue is collected, place it in fixative and record number of pieces on the datasheet.

   **NOTE**: If photographs are taken of individual fish and/or anomalies, the fish ID tag or scale envelope can be used in the photo as an identifier. Record photos taken on the Fish Health Examination datasheet.

C) Internal Examination

Lay the subdued fish on the foil and open the abdominal cavity with a cut from the vent forward to the pectoral girdle. Do not insert the scissors so far as to damage internal organs. With a gloved finger, reach into the anterior portion of the exposed cavity and cut the esophagus. Gently remove the entire viscera.

**Liver:**
Observe the liver and record condition on the Fish Health Examination datasheet. Check all that apply: dark to light red (normal), tan, general (overall) discoloration, focal (mottled) discoloration, nodules, other.

For fish with a discrete liver (e.g., salmonids, largemouth bass, others), carefully remove the liver and weigh it to the nearest 0.1 g (without the gallbladder). If the gallbladder is punctured, wash contaminated liver with de-ionized water. For fish with a dispersed (several hepatic nodules) liver (e.g., common carp), write “NA” for the weight on the Fish Health Examination datasheet.

Collect 5 pieces of liver (1 $cm^3$ each) and place into fixative. Be sure to take samples from various regions of the liver. Fill two cryovials with liver pieces and flash freeze for EROD analysis. Record the number of pieces in fixative on the Fish Health Examination datasheet.

**Bile:**

Observe the gallbladder and determine the fullness and color of the bile. Record condition on the Fish Health Examination datasheet. Check one for each; *Fullness*: empty, partly full, full. *Color*: N/A (gallbladder was empty), yellow, light-grass green, dark green to blue green.

**Spleen:**
Observe the spleen and record condition on the Fish Health Examination datasheet. Check all that apply: red to black (normal), granular, nodular, enlarged, other.

Carefully remove the entire spleen and weigh it to the nearest 0.002 g.

Collect 1 or 2 pieces (1 $cm^3$ each) and place in fixative. Record number of pieces in fixative on the Fish Health Examination datasheet.

**Gonads:**
**NOTE**: The gross visible appearance of gonads varies among taxa and over the life of the fish. Generally, ovaries are granular in appearance and are orange, yellow, or pink in color. Testes are lighter in color, whitish to yellow, with a smoother appearance. If a sample of gonad tissue is collected, sex determined in the field can be verified histologically in the laboratory.

Observe the gonads and record the sex and development on the Fish Health Examination datasheet.

Carefully remove gonads in their entirety and weigh to the nearest 0.1 g. Record the weight on the Fish Health Examination datasheet.

Cut 1 $cm^3$ pieces and place into fixative. Be sure to take samples from various regions of the gonads. Record the number of pieces collected on the Fish Health Examination datasheet.

**Mesenteric Fat:**
Pyloric caeca are present in certain fish families (eg. Salmonidae, Centrarchidae). For fish without pyloric caeca, the mesentery should be observed for inflammation or hemorrhaging. Using a ranking system, observe and record the estimated amount of fat: no fat, slight fat, 50%, >50%, completely covered, parasites, other. If a sample of abnormal tissue is collected, place it in fixative and record number of pieces on the datasheet.

**Kidney:**
**NOTE**: In some fishes (e.g., common carp, centrarchids) the anterior (head) kidney and the posterior (hind) kidney are separate; in others (e.g., salmonids, ictalurids), they are continuous. Here, they are treated as separate for examination and sample collection.

Observe the head and hind kidney and record condition of each on the Fish Health Examination datasheet. Check all that apply: normal (dark red), swollen, mottled, granular, urolithiasis, other.

Cut a 1 $cm^3$ piece of the head and hind kidney and place into fixative. Record the number of pieces collected on the Fish Health Examination datasheet.

**Scales, Spines and Otoliths:**
**NOTE**: These structures are collected to determine the age of the fish. The pectoral fin spines are collected from fish without scales (ictalurids). The otoliths (ear bone) are collected from inside the head, on either side of the brain.

Collect the sample from the left side of the fish; if the left side is damaged, collect from the right side. For spiny-rayed fishes, collect the scales from the area of the appressed pectoral fin. In soft-rayed fishes, collect the scales beneath the anterior portion of the dorsal fin, above the lateral line. For ictalurids, remove the entire disarticulated spine, including the base. These structures should be placed in a small scale envelope, labeled with the fish ID number. Record the type of structure sampled and location on the Fish Health Examination datasheet.

D) Carcass or Filet Preparation

**Carcass:**
Prepare the carcass sample by wrapping it securely in aluminum foil and attaching the labeled fish ID tag to the outside of the wrapped fish. Retain carcass samples in a freezer until processed.

**Filet:**
If necessary, remove scales from the left side of the fish, from the caudal peduncle to the pectoral fin. Using a sharp knife, remove a filet and wrap in aluminum foil. Label the outside of the wrapped filet with the fish ID number. Keep filets frozen until processed.

E) Cleaning Equipment

1. Thoroughly rinse all contact surfaces with tap water.
2. After each fish, as well as at the end of the day, wash all dissecting equipment with de-ionized water, and follow with an acetone rinse. Dispose of acetone in a 5 gallon bucket, with lid, until it can be disposed of properly.
3. Remember to use a new piece of aluminum foil for each fish as its work surface.
4. Make sure all needles are securely disposed of in the Sharps container.
5. Remember to properly disinfect all field gear (waders, boots, live wells, etc.). (See Chapter 7.2)

F) Shipment of Samples

If collected tissue samples are being analyzed through a contract laboratory, make sure the proper chain-of-custody procedures are followed. Cryogenic samples must be sent in dry ice or in a liquid nitrogen dry-shipper. Follow proper federal regulations when shipping.

G) Data

1. Fish age should be determined using proper techniques developed for the structure (e.g., otolith, scale) collected.

2. Lab results and field data are housed in a database such as EDAS. All of the original datasheets (Fish Health Examination and Station Identification) are kept on file, even after the data are entered into a database.
3. All photographs taken are archived for future reference.
4. Comply with sampling permits, and remember to submit all collection numbers when necessary.

## 10.3 Bioassessments and Fish Consumption Advisories

Fish sampling, other than for fish health studies, is typically performed either for bioassessment purposes or for tissue collection for fish consumption advisories. Fish bioassessments are usually done in lotic waters (wadeable streams). Tissue collections for fish consumption advisories are usually, but not always, done in lentic waters (lakes or reservoirs).

Assistance of other agencies (e.g., NMDGF, USFWS) should be considered when necessary or desirable. Tissue collection for fish consumption advisories is typically done with the assistance of NMDGF.

### 10.3.1 Sampling Sites and Season

For bioassessment, reach length is dependent on stream size. A stream reach of 40 X wetted width will be sampled for fish, or a minimum of 150 m. All available habitat types (pool, riffle, or run) should be sampled. Sampling is best performed in late summer or fall after any spawning migration has ceased and when young-of-the-year fish are large enough to be reliably identified in the field.

For tissue collection for fish consumption advisories, it is desirable to target waters where public fishing pressure is highest and/or where known or suspected contamination issues occur. Furthermore, it is desirable to sample at the time when fishing pressure is high, but as a matter of efficiency, sampling is usually coordinated with NMDGF sampling efforts.

### 10.3.2 Equipment

For bioassessment, electrofishing is the preferred method of capture. One backpack electrofisher is usually adequate for streams up to approximately three meters in width. Larger streams may require two backpack electrofishers or a bank/tote barge shocker. If two electrofishers are used, they should be set at equivalent voltages and other settings (frequency, pulse width, pulse duration, etc.).

Equipment used must be suitable for the specific conductance (SC) of the water. Backpack electrofishers are usually adequate for SC up to 1,500 microsiemens per centimeter (µS/cm); bank or tote barge shockers are necessary for higher SC. Seining is an acceptable technique in streams with high SC and fine sediment substrates.

For tissue collection for fish consumption advisories, SWQB usually uses trammel nets for lentic waters and electrofishers for lotic waters. NMDGF usually employs a boat-mounted electrofisher system for lakes and reservoirs.

### 10.3.3 Safety Considerations

When electrofishing, it is strongly recommended that non-conductive gloves of adequate length be worn by all participants to prevent electrical shock. Also, care should be taken to ensure that waders used do not conduct electricity and have no leaks. Consideration should be given to stream flow conditions to ensure safe working conditions. All activities requiring the use of a boat must adhere to boating safety protocols. It is strongly recommended that all personnel engaged in boating activities take a boating safety course that is offered by New Mexico State Parks.

### 10.3.4 Sampling Methods

#### *10.3.4.1 Bioassessment*

A single pass is usually adequate for bioassessment purposes; multiple passes are required if population estimates are to be performed. When electrofishing, the reach is sampled in an upstream direction to prevent creating turbidity in the area to be sampled. Seining is best performed in a downstream direction. A block net with five mm (3/16-inch) mesh may be used at the upstream end of the sampled reach (if no natural migration barrier exists) for single-pass samples.

To capture stunned fish use dip nets with mesh small enough to prevent escape of fish 20 mm total length (TL) or larger. If the mesh is too small, it is difficult to move the net through the water fast enough to capture fish. A 5 to 6 mm (3/16 to ¼ inch) mesh is ideal. Water must be clear enough to enable effective capture in dip nets. If water is too turbid for effective dip net use, a seine with a 5 to 6 mm mesh may be used by stretching it across the stream and shocking a short distance above the seine, sweeping the electrode down into the seine, repeating this process in an upstream direction until the reach is completely sampled (indicate in field notes if this variation is necessary).

Indicate on the data forms the length of the reach sampled and its proportion of habitat types (pool, riffle, or run). If electrofishing, indicate the number of seconds the electrofisher was passing current through the water (indicated by the timer on the electrofisher) and the settings (voltage, pulse type, pulse duration, etc.). If seining, indicate the length, width, and mesh size of the seine.

Move the fish from the dip net or seine to a bucket for later identification and quantification. Identify and count fish frequently enough to prevent mortality due to oxygen stress or other causes. If possible, aerate the bucket using a portable battery operated aerator to reduce oxygen stress. If it is necessary to stop and count fish before the reach is completely sampled, it should be done at a break between habitat types (e.g., between a pool and a riffle). After processing, replace the fish in the stream well downstream (upstream, if seining) of the last sampled reach to reduce the likelihood of capturing the same fish multiple times.

Fish are identified in the field by a qualified fisheries biologist. Total number of each species captured is enumerated and recorded in the appropriate field on the data form. Record visual observations of external anomalies (deformities, fin erosion, lesions, tumors, etc.), and any other

pertinent notes or comments. A subsample of each species may be measured (to length and/or weight) if necessary, depending upon the objectives of the study. Typically, salmonids are measured to length (for calculation of certain metrics used in coldwater streams). Ensure that individuals measured represent an unbiased subsample. Indicate on data forms if standard, fork, or total length (TL) is taken (SWQB typically uses TL).

Take voucher specimens of all fish species captured, with the possible exception of State or federally listed species (the NMDGF and/or the USFWS should be contacted prior to sampling if any listed species may be encountered, as per the terms of the collection permits). In the event that listed species are not vouchered, photographs may be taken provided the fish are not unreasonably stressed. Vouchers are placed in a Whirl-pak® or equivalent type of container with an estimated 10 percent by volume formalin solution. Fish from each sampling station are kept in separate containers and clearly labeled with a piece of paper inscribed with the field number of the collection placed in the bag. Deposit the vouchers at the Museum of Southwestern Biology, Division of Fishes, at the University of New Mexico in Albuquerque, NM. Complete the Museum field collection forms (printed on archival paper, available from the Museum) with archival quality ink and submit with the voucher specimens. Contact the Division of Fishes at the Museum for details.

Fill out fish sampling, physical characterization/water quality, and habitat assessment field data sheets completely while in the field. These forms are slightly modified versions of those available in *Rapid Bioassessment Protocols for Use in Wadeable Streams and Rivers* (Barbour et al., 1999). Habitat assessment field data sheets may be excluded if a separate habitat survey has been performed in the sampled reach.

#### *10.3.4.2 Tissue Collection for Fish Consumption Advisories*

Fish tissue may be collected for analysis under the New Mexico Fish Consumption Advisory program. In this case, there is usually little or no need for water quality, population, or habitat data collection. Assistance of other agencies (e.g., NMDGF, USFWS) should be considered when necessary or desirable.

In wadeable streams, electrofishing (when and where conditions permit) is probably the most efficient means of collecting a sufficient sample size of fish. Seines may also be used when necessary. Mesh size of dip nets and seines can be larger than with bioassessments, as larger fish are targeted for contaminant analysis. Block nets or natural migration barriers are unnecessary, except to facilitate capture if necessary. Shock seconds and other metadata do not need to be recorded.

In lentic waters, trammel nets are preferred over gill nets, as gill nets tend to catch a smaller range of sizes of fish and with a higher mortality rate. Trammel nets should have a large size outer mesh (e.g., 300 mm or 12 inch square) and small size inner mesh (e.g., 40 mm or 1.5 inch square) to maximize the size range that may be effectively caught. Fine twine size (e.g., 139) for inner mesh is more difficult for fish to detect, thus increasing catch results. Net depth should be at least four feet, preferably six feet. Length should be 50 to 100 meters or yards, unless specific circumstances dictate the use of a shorter net. For most purposes, nets should be designed to rest on the bottom (as opposed to floating from the surface). Therefore, the lead line's weight should overcome the float line's buoyancy. Use of lead core line on the bottom and foam core line on

the top is preferable to individual lead weights and floats as this reduces the tendency for tangles. Anchor weights for the nets may be fashioned from three-pound coffee cans filled with concrete with an eyebolt embedded in the concrete (a washer fastened to the threaded end of the eyebolt will prevent it from pulling out). During deployment, nets should be stretched lengthwise and should have adequate slack in the line to the surface floats to prevent wave action from lifting the anchor weights, thereby allowing the ends of the net to gradually move toward each other. Nets should be thoroughly dried after each use.

Store fish to be kept for analysis stored in a live well. Wrap each individual in aluminum foil (shiny side away from the fish's skin), and place those fish to be composited together in a plastic bag. Place the foil-wrapped fish on ice as soon as possible. Return fish that are not needed for analysis to the water as soon as possible, and with as little trauma as possible. Upon returning from the field, freeze fish that are not immediately processed. After processing, freeze fish fillets (or whole fish) until analysis.

For studies pertaining to wildlife, whole fish are used. For studies relating to human consumption, fish fillets should be used. It is preferable to have the processing performed by the contracted analytical laboratory. Otherwise, filleting should be performed on a clean surface that is washed and rinsed between each individual fish to avoid cross contamination. Those species of fish that are typically eaten without the skin should have the skin removed. Some species (e.g., salmonids) may have the skin left on, if that is how they are typically eaten. Individual fish are identified to species by a qualified fisheries biologist, weighed, and measured (TL). This information is recorded on the appropriate form.

Composite samples are always of one species from one water body, and a single size class should be included in any given composite (i.e., the smallest individual should be at least 75 percent as long as the largest individual; the smaller the size difference, the better). Details on how to handle samples and organize composites can be found in the New Mexico Fish Consumption Advisory Program planning document.

#### *10.3.4.3 Other Sampling*

Fish are sometimes collected for other than the purposes described above. For example, it may be necessary to determine if, and what species of, fish are present in a water body to ascertain the appropriate designated or existing uses. In such cases, best professional judgment should be exercised to determine sampling methodology and to ensure that safety concerns are addressed.

## 10.4 Periphyton Sampling

Currently, the State of New Mexico has a narrative criterion to determine nutrient impairment, which states,

> *"Plant nutrients from other than natural causes shall not be present in concentrations which will produce undesirable aquatic life or result in a dominance of nuisance species in surface waters of the state"* (NMAC 2005).

This narrative criterion can be challenging to assess because the relationships between nutrient levels and impairment of designated uses are not defined, and distinguishing nutrients from "other than natural causes" is difficult.

To address these problems, the Surface Water Quality Bureau (in cooperation with the Environmental Protection Agency and the US Geological Survey) revised New Mexico's Nutrient Assessment Protocol for Streams in 2004 using a more robust weight-of-evidence approach. The weight-of-evidence approach uses both causal (total nitrogen and total phosphorus) and response variables (dissolved oxygen, pH, and periphyton chlorophyll *a*) for assessment purposes. Impairment threshold values were developed for each of the causal and response variables used in the assessment protocol. The values are used to translate the current narrative nutrient criterion into quantifiable endpoints, which can be used in the development of Total Maximum Daily Loads (TMDLs).

### 10.4.1 Periphyton Sampling in Lotic Waters

A Richest Targeted Habitat (RTH) approach is used (Moulton et al 2002). Selection of the appropriate sampling method is based on habitat availability and a number of these methods may be used at a site. Selection of the habitat where samples are collected is generally based on the following guidelines:

- Cobble from riffles in shallow streams with coarse-grained substrates (epilithic habitat)
- Woody snags in streams with fine-grained substrates (epidendric habitat)
- DTH (Depositional Targeted Habitat): organically rich clay, silt, or sandy depositional areas (episammic and epipelic habitats) in streams where coarse-grained substrate and woody snags are absent.

<u>Sampling locations and selection of the Richest Targeted Habitats</u>:

A. Select a representative riffle. If the velocity is so great that it limits algal growth, (i.e., velocities exceed 1.75 ft/s) and more algae are found in glides than in the riffles, then select a glide (a length of stream with intermediate velocity). If no riffles are found near the site, select the glide where the most woody snags are located.
B. Starting at either the top or bottom of the selected reach, visualize a line running diagonally through the area. Select a landmark on the far bank that corresponds to the end of this diagonal transect. Samples will be collected from a number of points along this diagonal; generally ten points are sampled.
C. Collect one sample at each of ten equidistant points along this diagonal, starting with a point near one bank and ending at a point near the opposite bank. If you cannot wade across the stream, use the method described in paragraph F (below). If the study design stipulates that other than 10 points be sampled, divide the transect by the required number of points and sample accordingly. If the stream width is less than 3 feet, the sample may be collected from 2 or 3 separate diagonal cross sections by collecting 3 to 5 samples from each.
D. Bend down to lightly touch the bed sediments without looking at what is there. Pick up the first stone that you touch. If it is too big to retrieve, then take the nearest one that can be picked up. If you touch a small silty, sandy, or gravely patch among the cobbles, take the nearest stone that can be picked up. If no cobbles are present near the location, sample

a nearby woody snag. Look for cobble, then woody snags, within the distance to your next sample point in any direction. However, if you must go further than your next equidistant point to find a suitable cobble or woody snag among finer sediment, use the appropriate method to sample the substrate at that location, (i.e., the Petri dish, suction, or gravel sampler methods).

E. Composite the samples from each of the points into a single sample. Record the number and type of samples collected (i.e., ring, gravel sampler, woody snag, suction, or Petri dish) as well as the diameter and length of woody snags sampled.
F. If you cannot safely wade across the entire width, go as far as possible, then turn back and continue to collect samples on a diagonal back to the bank at the opposite end of the reach from where you started. For reaches that you cannot wade halfway across, collect samples in 10 points that you can access. Use the RTH approach in selecting substrate to sample (i.e., collect cobble if present; if not, then woody debris or finer substrate). Samples should be collected over an area that is approximately as long as the river is wide.
G. Record sampling collection information on the **Level II Nutrient Survey Form (Stream or Rivers)**

10.4.2 Sampling Methods for Epilithic Habitats

Epilithic (cobble and gravel) habitats are sampled using one of two methods depending on the types of rock substrate being sampled. The *ring method* is used when sampling cobble surfaces. A *gravel sampler* is used to sample periphyton attached to gravel substrates when only gravel is present (i.e., no > 2-inch diameter substrate is present in riffles or runs to allow use of the ring method).

*10.4.2.1 Ring method*

A. Equipment:
    - delimiter – section of 2- inch diameter PVC pipe, about 1 inch long
    - toothbrush – bent back 90° and bristles trimmed to about 6 mm
    - tray – to carry rocks
    - funnel
    - 500-1000 mL sample container
    - scalpel or knife
    - scissors
    - wash bottle

B. Select a representative riffle. Starting at either the top or bottom of the riffle, visualize a line running diagonally through the riffle. Divide the transect by the required number of points depending on the study design and stream width and collect a cobble at each of those points (see Section 10.1.1). Place stones in a plastic tray, top side facing up (i.e., in the same orientation that they were found), and transport them to the bank to collect periphyton. Place the ring on top of the stone to define a circle in the center, or as near as possible to the center, of the stone.
C. Use a knife or scalpel to scrape off as much periphyton growth as possible from within the ring and rinse off the scalpel into an appropriately labeled sample container.

D. Scrub the defined area for 30 seconds with the modified toothbrush. Remove the slurry from within the circle by removing the ring, holding the rock at an angle over a funnel placed in the sample bottle and rinsing with a wash bottle of stream water. Thoroughly rinse the area but use a minimal amount of water. Thoroughly rinse the toothbrush into the container. (Note: only use small amounts of wash water to avoid running out of space in the sample container and making homogenizing the sample more difficult).
E. If the sampling point falls over a mat of filamentous algae, a slightly different approach is required for sample collection. Place the ring on the center of the rock. At the ring, cut any filaments passing into or out of the ring. Remove any loose filaments from outside the ring. Scrape off all of the filaments inside the ring and rinse it off the scalpel into sample container. Scrub the defined area for 30 seconds with the toothbrush, then remove the slurry from within the circle as described above.
F. Combine each discrete collection into the same 500-mL sample bottle (i.e., composite the samples from the ten stones).
G. Place the bottle on dry ice inside a latched cooler and keep in the dark if the sample is not to be processed immediately. The sample must be processed within 12 hours if it is not frozen upon collection.
H. Note the area sampled.
I. Process the periphyton sample following the steps described in Section 10.4.8 Sampling Processing and Analysis.

Total Sampling Area = $n\pi r^2$, where:
n = number of discrete collections,
$\pi$ = 3.1416, and
r = radius of the delimiter in centimeters (2.3 cm for 2 inch PVC pipe ).

*10.4.2.2 Gravel sampler method*

A. Equipment:
    - Gravel sampler – 1 inch section of 3-inch diameter, capped PVC pipe
    - Dishpan
    - Wash bottle
    - Funnel
    - 500-1000 mL sample container
    - Masonry trowel or spatula
    - Forceps
    - Scissors

B. Assemble the gravel sampler from a 3-inch (7.6 cm) diameter capped PVC pipe about 1 inch long. One end of the pipe should be beveled to improve the coring capability of the sampler. Attach the cap.
C. Select 10 sampling points along a diagonal transect through a riffle (see Section 10.1.1).
D. Press the beveled end of the sampler into the gravel until it is full of substrate. After the sampler is in place, carefully remove the gravel surrounding the outside of the sampler and insert the masonry trowel.
E. Slide the sampler onto the trowel/spatula and carefully lift it out of the water.

F. Quickly invert the sampler to contain the gravel and water in the sampler cap.
G. Pour each discrete collection into a dishpan and rinse the sampler with stream water before taking another discrete collection.
H. Repeat these steps to complete 10 (or other required number) discrete collections, which form the composited sample. Record the number of collections on the field sheet.
I. Extract macroalgal filaments (if present) from the gravel with forceps and then cut them into fine pieces.
J. Brush and rinse (with dishpan water) the gravel. Reuse the rinse water as needed to keep the sample volume less than 475 mL.
K. Pour the composited sample from the dishpan through a funnel into a 500-mL sample bottle.
L. Process the periphyton sample following the steps described in Section 10.4.8 Sample Processing and Analysis.

10.4.3 Sampling method for Epidendric Habitats

Collecting quantitative microalgal periphyton samples from epidendric (woody snags) habitats presents a challenge because they generally have an irregular surface and are difficult to remove without loss of algal biomass (Porter et al. 1993). If the woody snag can be removed from the water and has a smooth surface, it can be sampled in a similar manner to epilithic habitats using the ring method (see Section 10.4.2.1). Otherwise, periphyton is collected from woody snags by using the *cylinder scrape method*.

10.4.4 Cylinder scrape method

A. Equipment:
    - Pruning shears or saw
    - Dishpan
    - Toothbrush
    - Scalpel
    - Wash bottle
    - Funnel
    - 500-1000 mL sample container
    - Ruler

B. Select one woody snag in each of ten locations throughout the reach.
C. Identify the part of the woody snag that will be sampled for periphyton. Carefully remove a 10 to 20 cm long section with pruning shears or saw and place in a plastic dishpan.
D. Scrub the entire surface of each woody snag section in the dishpan with the toothbrush. Rinse the brush and each section of woody snag in the dishpan using stream water. Recycle rinse water to keep the sample volume less than 475 mL.
E. Pour the composited sample from the dishpan through a funnel into a 500-mL sampling bottle.
F. Measure the length and diameter of each cleaned woody snag section and calculate the total sampling area by using the following formula (assumes a cylinder):

Total Sampling Area = $\Sigma\pi d_i l_i$, where:
$\Sigma$ indicates summing of all individual sample areas,
$\pi$ = 3.1416,
$d_i$ = diameter (cm) of each woody snag section, and
$l_i$ = length (cm) of each woody snag section.

G. Process the periphyton sample following the steps described in Section 10.4.8 Sample Processing and Analysis.

10.4.5 Sampling methods for epipsammic/epipelic habitats (Depositional Targeted Habitat – DTH)

Quantitative microalgal periphyton samples are collected from the upper five- to seven-mm layer of epipsammic (sand) or epipelic (silt) habitat in depositional areas. If the depth and velocity are low, the *suction method* may be used.

*10.4.5.1 Suction method*

A. Equipment:
- Delimiter – section of 2-inch diameter PVC pipe, about 1 inch long
- Turkey baster
- Wash bottle
- Funnel
- 500-1000 mL sample container

B. At points in the diagonal cross section where depth and velocity are low and that have a depositional zone consisting of either sand or silt substrates, place the delimiter (PVC ring) onto the sediment. Push into the sediment to a depth of 1-2 cm.
C. Use a turkey baster to remove the entire top 5-7 mm of sediment and deposit into the sample container. Combine each discreet collection into the same container (i.e., composite the samples from the 10 points).
D. Note the area sampled, as described in the ring method section.
E. Process the sample following the steps described in Section 10.4.8 Sample Processing and Analysis.

*10.4.5.2 Inverted Petri dish method*

A. Equipment:
- Petri dish, about 47 mm diameter
- Spatula
- Wash bottle
- Funnel
- 500-1000 mL sample container

B. At points in the diagonal cross section that have a depositional zone consisting of either sand or silt substrates, hold a small plastic Petri dish (about 47 mm diameter) upside down in the water; rub the inside of the lid to remove air bubbles.

C. Without disturbing the sediment, turn the inside of the Petri dish toward the substrate that will be sampled.
D. Carefully and slowly press (in cookie cutter fashion) the dish into the sediment.
E. Slide a spatula under the dish to enclose the discrete collection. Holding the Petri dish tight against the spatula, carefully wash extraneous sediment from the spatula, and then lift out of the water.
F. Invert the lid and remove the spatula.
G. Rinse the sediment from the dish with stream water into the sample container.
H. Repeat this collection procedure at each additional sampling points in the transect.
I. Combine each discrete collection into the same sample container (i.e., composite the samples from the 10 points).
J. Note the total area sample
K. Total Sampling Area = $n\pi r^2$, where:
    n = number of discrete collections,
    $\pi$ = 3.1416, and
    r = radius (cm) of the Petri dish.
L. Process the sample following the steps described in Section 9.4.8 Sample Processing and Analysis.

### 10.4.6 Periphyton Sampling in Lentic Waters

See Chapter 13 Lakes

### 10.4.7 Sample Handling

All samples should be processed as soon as possible after collection. Samples must be either frozen or filtered and frozen at the end of each day, no more than 12 hours after collection. After collection, pour off a 50 mL sub-sample and preserve with 2 mL of formaldehyde for taxonomic identification. Place the labeled periphyton samples on dry ice in latched coolers until processing (the 50 mL sub-sample should **not** be packed on ice). It is important to keep the periphyton samples chilled and in darkness.

### 10.4.8 Sample Processing and Analysis

#### *10.4.8.1 Collection and preservation of taxonomic identification sub-sample*

Before homogenizing or freezing the composite sample, a 50 mL sub-sample should be removed, labeled, and preserved with 2 mL of formaldehyde for taxonomic identification. Periphyton and algal samples for taxonomic identification should be preserved in formaldehyde to enable the soft-bodied algae to be preserved. This preservative is active indefinitely.

#### *10.4.8.2 Sample preparation for Chlorophyll a analysis*

Most periphyton samples will contain clumps of filamentous algae or diatoms creating difficulties in sub-sampling. Representative sub-sampling is essential for accurate assessment of periphyton communities. Homogenizing the sample with a hand-held blender can significantly reduce variability in sub-sample analyses, particularly in samples dominated by filamentous algae. The blending process generally does not greatly damage cells. Certain green filamentous

algae do not break apart easily (e.g., *Cladophora* sp.) during blending and tend to get wrapped around the cutting blades. The use of sharp blades on the blender, only a small volume of sample, and a slightly longer blending time usually overcomes this problem (Biggs 1987).

A. Equipment:
    - Glass or plastic beaker
    - Squirt bottle with distilled water
    - Kitchen or laboratory blender
    - Graduated cylinder
    - Small sharp scissors
B. Pour the contents of the sample container into a graduated cylinder. Rinse out any sample residue from the container and lid into the cylinder. Rinse off, but do not transfer, sand and gravel. Distilled or DI water is preferred for rinsing and making up the sample volume, however, tap water can be used providing the water isn't heavily chlorinated. Measure and record the total sample volume. Remember to add the 50 mL that was removed before freezing for taxonomic identification. Pour the sample into a beaker (the width of the beaker should be only slightly greater than the width of the housing holding the blades of the blender).
C. Pick out any invertebrates, pieces of gravel, leaves, moss, etc. from the sample.
D. Ensure that there is enough water to fully cover the blender-blade housing.
E. Blend for about 30 seconds or until the mixture is free of obvious clumps of material. If the sample contains much filamentous algae, break the strands up by repeated cutting with a pair of sharp scissors. Every 10 seconds, or so, stop the blender and free any filaments that have become caught on the blades or blender housing and cut into lengths less than 5 mm. The end result must be a homogeneous solution suitable for sub-sampling.

#### *10.4.8.3 Filtration for Chlorophyll a analysis*

A. Equipment:
    - Vacuum filtration apparatus
    - Hand held vacuum pump with pressure gauge
    - Glass fiber filters, 47 mm Whatman GF/F
    - Forceps/tweezers
    - Wide aperture (~ 2 mm diameter) 5 mL pipette
    - Squirt bottle with deionized or distilled water
    - Aluminum foil and permanent marker
B. Set up filtering apparatus with a fresh filter.
C. Place a sub-sample in the filtering chamber. The sub-sample is made up of 3 aliquots drawn from the blended sample. The volume of the aliquots depends on the concentration of the sample and varies from 1 to 5 mL. Good color development on the filter is necessary, but not so much that extensive dilution will be required after extraction. To take aliquots, shake the bottle of blended sample or briefly turn on the blender and withdraw 1-5 mL with a pipette from halfway down the solution while the liquid is still agitated. Repeat this twice more to give the full sub-sample (i.e., the total sub-sample volume = 3-15 mL).

D. Apply suction pressure (be careful not to have a high pressure as this will rupture cells releasing the chloroplasts; <6 inches Hg vacuum is recommended).
E. If there is not an obvious coloring from periphyton on the filter, more aliquots should be filtered.
F. Check for any fragments of leaves, mosses, invertebrates, etc. on the filter and remove these with forceps.
G. Record the volume of sub-sample (i.e., number and volume of aliquots used).
H. Remove the filter from the filtering apparatus, fold in half, and place on a sheet of aluminum foil. Fold in the sides of the aluminum foil to seal in the filter. Label the wrapped filter with the location, date, and analysis to be performed (Chl *a*).
I. Repeat steps 1-8 until the desired number of replicates of Chlorophyll *a* sub-samples are filtered.
J. The wrapped, labeled filters should be placed in a re-sealable plastic bag which has been labeled with the location, date, volume filtered, total volume, and number of replicates (e.g., 1 of *n*, 2 of *n*, etc.). Freeze the wrapped, labeled filters. Samples should be processed as soon as possible after collection, however, the wrapped filters can be frozen for up to 6 months before analysis, if necessary.
K. Refreeze the remains of the sample for repeat analyses if needed.

Notes:

- Always check that clumps of algae or leaf fragments, etc., have not blocked the intake to the pipette.
- If the sub-samples are taking a very long time to filter, or the filter is darkly colored or covered with a thick layer of material, the sample probably needs dilution or use a smaller volume aliquot. Ensure that you record the degree of dilution and aliquot volume so that this can be used in the concentration calculations.

#### *10.4.8.4 Periphyton Chlorophyll a analysis*

The Chlorophyll *a* analysis is conducted using a modified method as described in "Method 445.0 - In Vitro Determination of Chlorophyll a and Pheophytin a in Marine and Freshwater Algae by Fluorescence" (USEPA 2000). Acetone is replaced by ethanol throughout the method, e.g., for washing cuvettes, calibration, and dilutions. This modification was made because ethanol is a very effective solvent, there is only a minor shift in the absorbance peak with acidification, interference from chlorophyll *b* is less, ethanol presents a much lower health and safety risk, and it is less expensive. Chlorophyll is extracted in a solution of 90% boiling ethanol (i.e., 78 °C) (Biggs and Kilroy 2000).

A. Equipment:
   - Ethanol, diluted to 90% with distilled water
   - Capped 15 mL centrifuge tubes in racks
   - Forceps/tweezers
   - 5 mL Pipettes
   - Water bath
   - Centrifuge
   - 5 mL pipeters and tips

B. Pre-heat the water bath to 78 °C.
C. Prepare tubes by labeling, placing 5 ml of 90% ethanol in each, and re-cap.
D. Remove the filter from the filtering apparatus or aluminum foil, fold in half, loosely roll up and place in tube. Make sure the filter is completely covered with ethanol and the tube is firmly sealed.
E. Immerse the rack of tubes in the pre-heated bath for exactly five minutes. Make sure the tubes are held firmly in the racks. Loosen, but do not remove, the tops to prevent them popping off as the ethanol boils.
F. Remove racks of tubes from the water bath and place in the refrigerator overnight.
G. If any of the samples have high chlorophyll concentration (i.e., are a very deep green), then dilute with 90% ethanol. The aim is to keep the chlorophyll concentration below 90% of the upper limit of the Linear Dynamic Range (LDR) of the fluorometer.
H. To dilute, add an additional 5 ml of 90% ethanol to the tube, cap and shake well. Note on the data sheet that the volume of extractant is 10 ml (instead of the usual 5 ml). If the solution still looks very concentrated, centrifuge the solution (as described below), then withdraw 5 ml from the tube and place in a separate tube, add another 5 ml of 90% ethanol, and shake well. Note on the data sheet that the (effective) volume of extractant is now 20 ml.
I. Using a glass rod or metal forceps, push the filter papers to the bottom of the centrifuge tubes, and cap firmly. Centrifuge at a speed of 6000 rpm for 10 minutes.

## FORMS

Fish Sampling Field Data Sheet
Fish Health Examination
Fish Health Station Identification
Fish Consumption Advisory Field Data
EMAP Sample Collection Form -Streams
Large River Macroinvertebrate Periphyton
Level II Nutrient Survey Form (Streams)
Level II Nutrient Survey Form (Rivers)

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# 11.0 BACTERIOLOGICAL SAMPLING

Containers used for sampling for E. coli depend on the testing agency. SLD requires sterile, wide-mouth 100-mL polypropylene bottles containing sodium thiosulfate, which are supplied by SLD. These are currently the only containers acceptable for bacteriological samples being submitted to SLD. Contract laboratories may supply slightly different containers. For in-house IDEXX testing use the containers supplied by the manufacturer. Do not rinse the containers before use, and do not remove the lids until immediately before sampling. Fill containers by submersing in flowing water and fill exactly to the 100 ml line. Decant excess water immediately rather than at the time of processing. After collection, cool the sample on ice to 4ºC and transport in ice chests. The Environmental Microbiology section at SLD must be consulted well in advance of the proposed sampling effort to assure availability of media and incubator space. When performing IDEXX testing in SWQB equipment, allow the samples to warm to room temperature before adding the reagent packet supplied by the manufacturer. When transporting *E. coli* samples they can be taken off ice approximately 30 minutes before arriving at the lab so that they can be processed immediately upon arrival.

Total coliform and *E. coli* samples may be delivered to SLD, processed at the SWQB laboratory, or processed in the field using portable equipment. Holding time is 6 hours from time of collection to the start of incubation for samples processed with SLD equipment, either in the field or in the SWQB lab, and 6 hours from time of collection until time of delivery at SLD.

The IDEXX Laboratories, Inc. Colilert®, Colilert®-18, Colilert®-24 procedures are used by SLD and the SWQB laboratory for enumeration of total coliform and *E. coli* concentrations by the most probable number (MPN) method.

## 11.1 Sample Collection

Collection procedure is similar to chemical sampling. **Collect the exact amount necessary for analysis using the 100 ml mark on the collection vessel.** This eliminates the need to pour off excess at the lab, avoiding complications if too much liquid is poured off. After collection, cool the sample on ice to 4ºC and transport in ice chests for delivery and processing within six hours of the collection time.

*SWQB Testing Materials for Samples Processed by SWQB Staff*

- IDEXX Colilert® Reagent “Snap Packs” for 24-28 hour incubation
- IDEXX Colilert®-18 Reagent “Snap Packs” for 18-22 hour incubation
- IDEXX Quanti-Tray®/2000 bacterial enumeration trays
- IDEXX Quanti-Tray®/2000 Most Probable Number Tables
- IDEXX MPN Generator V3.1 Program: 3.5” Floppy Disc
- IDEXX Shrink Banded Sample Bottles
- IDEXX Colilert® and Colilert®-18 Quanti-Tray®/2000 Comparator

There are two sets of E.coli enumeration equipment, a “lab” kit and a “field” kit. These are specialized for their respective environments.

FIELD KIT:

- Thermotote® portable Incubator with DC power plug for vehicle operation
- IDEXX 6 watt fluorescent UV lamp
- IDEXX Quanti-Tray® sealer, “field”
- IDEXX Quanti-Tray®/2000 rubber insert
- Deep cycle 12V batteries
- Power Converters
- AC/DC to power the portable incubator via a typical 120V outlet
- DC/AC 800 watt inverter: +/- alligator clips allow for connection to deep-cycle battery or vehicle alternator
- DC/DC adapter allowing vehicle power port conversion to +/- alligator clips for deep cycle battery power

LAB KIT:

- IDEXX 110V Incubator
- Generic bench-light18” fluorescent UV lamp
- IDEXX Quanti-Tray® sealer, “lab”
- IDEXX Quanti-Tray®/2000 rubber insert

## 11.2 Processing Samples In-House

IDEXX samples to be processed in the SWQB laboratory are enumerated using IDEXX Laboratories Colilert® or Colilert®-18 and Quanti-tray® techniques, as described in *Standard Methods*, protocol 8310 B. Results are reported in MPN/100ml.

1. For enumeration using Colilert® -18 or Colilert®-24 methods, allow chilled samples to adjust to room temperature. If samples are not at room temperature, the reagent will not dissolve properly.
2. Add one reagent packet directly to the 100-ml vial. Shake gently to dissolve completely. The solution must be dissolved completely before processing.
3. Pour the solution into a Quanti-Tray®/2000 incubation plate and allow any foam to settle. Gently tap the incubation plate to dislodge any bubbles that may be trapped in the cells.
4. Switch on the Quanti-Tray® Sealer and allow it to warm up until the green light on the cover activates.
5. Place the tray into the red rubber frame and insert the frame and tray into the bay in the front of the sealer.
6. Carefully ease the frame and tray forward until the sealer activates and feeds the tray automatically. The sealer may occasionally jam while processing the tray. If this occurs, press the reverse button on the cover and the track will reverse, depositing the frame on the bay. Wait until the green light activates again and repeat the loading process. The tray will be deposited on the counter behind the sealer.
7. Place the sealed tray into the incubator at 35˚C (+/- 1˚C) and incubate 24-28 hours for Colilert® reagent or 18-22 hours for Colilert®-18 reagent.

If read before the appropriate minimum incubation time has elapsed, the incubation may not be complete and false negatives may occur. If read after the appropriate maximum incubation time has elapsed, the non-coliform suppressant may have diminished effects and false positives may occur. Flag samples for which holding times are violated in the database. A thermograph should be kept in each incubator recording temperature throughout sample incubation and temperature data files should be stored as a permanent record for QA/QC. Also flag samples for which temperature requirements are violated (35˚C +/- 1˚C) in the database.

## 11.3 Processing Samples In-Field

A portable "field" kit has been assembled for sample processing in the field when the 6-hour holding time cannot be guaranteed to SLD or SWQB laboratories. *E. coli* samples to be processed by SWQB staff in the field are enumerated using IDEXX Laboratories Colilert® or Colilert®-18 and Quanti-tray® techniques, as described in *Standard Methods*, protocol 8310 B, Results are reported in MPN/100ml, and include a 95% confidence interval.

Sample procedures are the same as for in-house processing, with the exception of an incubator capable of operating on 12V power sources and power options for the sealer and incubator. There is a checkout sheet for the field kit in the SWQB public folders: SWQB PUBLIC\Sonde&Equipment Checkout. Please utilize this sheet when taking the kit from the building.

If it is necessary to seal trays in the field, the IDEXX sealer may be powered by the vehicle alternator. It is also possible to seal trays using a stand alone deep cycle battery, but the sealer draws excessive current and is only just able to operate with the low amperage supplied by the battery alone. To operate the 110V sealer, power must be converted from the 12V supplied by the vehicle alternator, and the vehicle must be running to supply sufficient amperage. A DC/AC 800 watt converter is used for this power conversion. Clip the alligator clips on the converter to the respective terminals on the vehicle battery and press "ON". If the vehicle is started after the converter is connected it will need to be turned ON again. Plug the sealer into the standard household outlet on the converter face. Turn the sealer ON with the switch on the rear panel. Then follow procedures for in-house processing.

The Thermotote® incubator is supplied with a vehicle DC power adapter that allows it to be powered by a vehicle's power outlets. However, it draws a high volume of amperage over time and if the vehicle is turned off with the incubator running, it will drain the vehicle battery. To avoid this, but still keep the incubator powered, use a 12V deep cycle battery. The DC/DC converter attaches to the incubator power plug on one end and to the battery via alligator clips on the other. On especially cold nights when the incubator is left outside, it may be necessary to link two 12V deep cycle batteries in parallel to ensure sufficient amp-hours (thus, it is preferable to bring the incubator indoors if possible). Batteries linked in parallel have their positive terminals linked together and their negative terminals linked together. There is also an AC/DC converter that allows the incubator to be powered by a standard home 3-prong electric outlet by receiving the vehicle plug on one end and inserting into the wall outlet on the other.

Insert a thermograph inside the incubator to record temperature during field use to provide information for QA/QC purposes.

It may also be necessary to count cells while in the field. The biggest challenge to this task is finding a dark place to discern which cells fluoresce. Service station washrooms, WWTP laboratories, and SWQB field offices are often used for this purpose.

## 11.4 Enumeration/MPN Table

- An IDEXX Colilert® and Colilert®-18 Quanti-Tray®/2000 Comparator is a Quanti-Tray®/2000 filled with borderline yellow and fluorescing comparator for distinguishing threshold positive results from negative results by comparison. Concentrations may be determined for total coliforms and *E. coli* MPNs.
- To quantify total coliforms, count the number of small vs. large cells that have changed from clear to yellow.
- To quantify *E. coli*, count the number of small vs. large cells that fluoresce under a UV "black" light.
- There is one outstanding cell at the top of the tray that is larger than the rest and it should be counted as a large cell.
- To determine MPNs, the number of large cells ($\leq$49) and number of small cells ($\leq$48) are compared using an MPN table stored with the equipment or the MPN generator software available on a 3.5 floppy disk and stored with the IDEXX documentation.

## 11.5 Documentation

Samples submitted to SLD are documented using "SLD Bacti Water Form 5299" available in PDF format from the SLD website:
http://www.sld.state.nm.us/lab/Documents/waterform.PDF

Samples processed by SWQB staff should be recorded on the formal *E. coli* Bacteria Record Sheet. This is an official record of bacteria collection date/time, incubation date/time, enumeration date/time, test reagent, temperature at time of enumeration, sampling station, Request Identification (RID) number, and results. All of these conditions should be recorded for QA/QC record keeping. Data are ultimately uploaded into the SWQB database for electronic storage.

**FORMS**
*E.coli* Bacteria Record Sheet

# 12.0 STORMWATER SAMPLING

Stormwater, due to its unpredictable nature, is sampled most often with deployed, automatic sampling equipment. Grab samples may be collected when staff are present during a storm event. Caution should be exercised when collecting storm-water runoff to avoid rapidly rising hydrographs and debris washing down watercourses.

For unattended surface water monitoring, the Surface Water Quality Bureau uses a Teledyne ISCO 3700 portable automated sampler, often referred to simply as “an ISCO,” or “the sampler,” linked to a model 4230 flow meter, “the bubbler,” and programmed to collect samples at preset intervals on a watershed specific hydrograph. Data are stored electronically on the flow meter RAM and available for retrieval through a model 581 rapid transfer device (RTD). A liquid level actuator (LLA) is available to trigger the sampler upon detection of water in the channel when a bubbler is not available.

D-Tec Corporation is the sole distributor and manufacturer of the Environmental Liquid Sampler (ELS). The ELS is a mechanically operated instrument, set-up independently from the ISCO equipment, that collects a "first flush" volume of liquid without a power source, then seals itself off to preserve the sample integrity. These are often deployed in conjunction with a crest gauge: a staff gauge mounted inside a perforated tube that records maximum stage height each time it is reset. Crest gauge stage readings provide a basis for manufacturing a general hydrograph and help to determine storm load values.

An evolution to the crest gauge is the addition of an electronic level logger to record stage height at preset intervals, producing a more complete hydrograph than a crest gauge is capable of doing. In-Situ, Inc. Level “Trolls” are used for this purpose.

## 12.1 ISCO 3700 Automated Sampler Setup

The following description provides the details for an initial set-up using an array of glass and polypropylene sampling bottles. This is only one of many sample arrays possible, and adjustments to the program can easily be made, although review of the ISCO technical manuals may be necessary. During subsequent field visits, most of the program options can be disregarded.

Chapter 4 of the Teledyne-ISCO, Inc. Model 3700 Portable Sampler Instruction Manual has a detailed description of each step in programming the ISCO. The following is a condensed version describing how to take multiple, non-uniform, timed samples during a storm event. Variations of this sample array may be made to accommodate various sample volumes as necessary for analysis.

Non-uniform, timed samples are typically used in runoff studies. Preferably, sample intervals are short during the early stage of a runoff event and increase during the later, trailing leg of the hydrograph.

*Set-up*

1. The ISCO carousel can accommodate 24 bottles. Load 6-350ml glass bottles into slots 1 through 6. Fill the remaining slots with 18-1L polypropylene plastic bottles. Sample water collected in the glass bottles will be analyzed for organic substances while the remaining water collected in the polypropylene bottles will be analyzed for inorganic substances.
2. Power the sampler from a 12v deep cycle battery or other power source. Then connect the ISCO to the flow meter with the 6-pin cable provided by the manufacturer.
3. Install the ISCO sampler, flow meter, and battery on relatively level ground above the flood plain, within 26 vertical feet of the channel (the maximum lift of the ISCO pump at optimal conditions). Often large tool/storage boxes are used for equipment protection in the field.
4. Attach a length of 3/8-inch diameter Teflon suction line to the ISCO intake and anchor the other end to the channel bottom. The proper placement of the suction line assures the collection of representative samples. Slope the line from the sampler to the sample point in a continuous downhill fashion allowing water to drain between sample intervals. Orient the line intake facing upstream and anchor it to the channel bottom in the main flow of the stream, not in an eddy or edge of the flow. Intake at the channel bottom may result in excess heavy solids, or bed load. Professional judgment determines the intake height above the channel bottom. For example, wide, low-flowing channels may require locating the intake within 2 inches above the bottom, while a narrow, high-flowing channel may allow placement of the intake higher in the water column.
5. Attach a length of ¼-inch diameter vinyl tubing to the flow meter, at the barbed fitting below the cable connectors, and anchor the end of the hose to the channel bottom near the ISCO sampler intake. Be aware that the stream channels we commonly monitor are unstable. They are often adjusting, aggrading and degrading, to changing flow regimes. Professional judgment is required in locating the ISCO tube ends at a secure and appropriate location.
6. Program the ISCO sampler first, place it in standby, and then program the flow meter.

The following sections describe step-by-step procedures for programming the ISCO sampler and accompanying flow meter. It is recommended that you walk through each step carefully and assure that all steps are programmed properly.

After programming the sampler, installing the equipment in the field, recovering samples from a recent event, or reloading bottles for a future event, press the start sampling key. SAMPLER INHIBITED will then be displayed on the screen. This display is vital in determining whether the ISCO has been properly programmed.

At each step, you are given the option to continue on to the next step or move backwards to a previous step by using arrow keys. Once programming has been completed, this allows you to

move through the program to a specific step for which you may wish to change settings. At each step, the option that will be highlighted on the ISCO screen is listed here under the Option column in capitalized letters. Pressing the Enter/Program key will put you into the step and enable you to program that portion. Once in a step, the arrow keys allow you to scroll between choices, while the Enter/Program key selects your choice. The ENTER/PROGRAM key will be referred to as the ENTER key during the remainder of this description.

## 12.2 ISCO Portable Automated Sampler Programming Instructions

This procedure will instruct the ISCO to initiate sampling when a signal is sent by the flow meter and to continue sampling at preset time intervals after that initial start.

To begin, turn the sampler ON and press Enter to begin the programming sequence.

Press ENTER to proceed to the ISCO CONFIGURE sequence.

**Table 12-1 ISCO configure sequence**

| STEP | OPTION | NOTES |
|---|---|---|
| 1 | [PROGRAM,CONFIGURE] SAMPLER | Choose CONFIGURE first; you will do PROGRAM later in the procedure. Use the arrow keys to move between PROGRAM and CONFIGURE; press the ENTER key when the appropriate selection is flashing. |
| 2 | SELECT OPTION: SET CLOCK | Press ENTER to set clock and date. Synchronize the time and date between the sampler, flow meter and appropriate stations (when available). Pressing the appropriate numbers on the keypad can change the flashing selection. Press ENTER if correct, or use the arrow keys to scroll through the remaining selections to the next program option. |
| 3 | SELECT OPTION: BOTTLES AND BOTTLE SIZES | Press ENTER to reset the bottle number and size or use the arrow keys to move on to the next program option. |
| | [PORTABLE, REFRIG] | Select PORTABLE, then ENTER (SWQB does not use refrigerated samplers at this time). |
| | [1,4,12,24] BOTTLES | Select 24 BOTTLES, and then ENTER. |
| | BOTTLE VOLUME IS # | Change bottle volume to appropriate volume using the number keypad: enter 1000ml. The option to account for the smaller glass bottles is available in the PROGRAM mode. |
| 4 | SELECT OPTION: SUCTION LINE | Press ENTER to reset the suction line size and length, or use arrow to move on to the next option. |
| | SUCTION LINE I.D. IS [1/4, 3/8] INCH | Select 3/8 for SWQB current inventoried tubing. Investigate size if new tubing is added to inventory. |
| | SUCTION LINE IS [VINYL, TEFLON] | Select TEFLON, then ENTER |
| | SUCTION LINE LENGTH IS | Measure the length of the suction line and enter the length IN FEET using the number keypad, then press ENTER. The ISCO will momentarily display “Calculating Pump Value Tables” as it computes the tubing volume. |
| 5 | SELECT OPTION: LIQUID DETECTOR | Press ENTER to activate the liquid detector located at the pump housing. |
| | [ENABLE, DISABLE] LIQUID DETECTOR | Select ENABLE, then ENTER |
| | RINSE CYCLES | Select 2 with the number keypad, then ENTER. The rinse cycles condition the suction line and reduce the cross-contamination potential. |
| | ENTER HEAD MANUALLY [YES,NO] | Select NO, then ENTER |
| | RETRY UP TO “#” TIMES WHEN SAMPLING | Select 0, then ENTER. If multiple attempts are made and sampler collects partial samples, a risk exists that the bottles will overfill, finally causing the sample distributor to jam. |

| | | |
|---|---|---|
| 6 | SELECT OPTION: PROGRAMMING MODE | Press ENTER |
| | [BASIC,EXTENDED] PROGRAMMING MODE | Select EXTENDED, then ENTER. This mode allows further site-specific programming to be achieved. |
| 7 | SELECT OPTION: LOAD STORED PROGRAM | Use the arrow key to pass this and the following selection "SAVE CURRENT PROGRAM." These selections allow the user to store and reuse individual sample programs. Circumstances may vary at each station and the programs are individualized. |
| 8 | SELECT OPTION: FLOW MODE SAMPLING | Press ENTER |
| | TAKE SAMPLE AT START TIME | Select NO, then ENTER |
| | TAKE SAMPLE AT SWITCH? [YES,NO] | Select NO, then ENTER |
| 9 | SELECT OPTION: NONUNIFORM TIME | Press ENTER |
| 10 | ENTER INTERVALS IN [CLOCK TIME, MINUTES] | Select MINUTES desired between individual sample collections, then ENTER. |
| 11 | SELECT OPTION: CALIBRATE SAMPLER | Press ENTER |
| | [ENABLE,DISABLE] CALIBRATE SAMPLER | Select ENABLE, then ENTER |
| 12 | SELECT OPTION: SAMPLING [STOP,RESUME] | Press ENTER |
| | [ENABLE,DISABLE] SAMPLING STOP,RESUME | Select DISABLE, then ENTER |
| 13 | SELECT OPTION: START TIME DELAY | Press ENTER |
| | # MINUTE DELAY TO START | Select 0 with the number keypad, then ENTER. The program will initiate when a signal is sent by the flow meter. |
| 14 | SELECT OPTION: ENABLE PIN | Press ENTER |
| | MASTER/SLAVE MODE [YES,NO] | Select NO, then ENTER, unless it is desirable to link two ISCOs to sample a 48 bottle array-see manual for instructions. |
| | SAMPLE UPON DISABLE [YES,NO] | Select NO, then ENTER |
| | SAMPLE UPON ENABLE [YES,NO] | Select YES, then ENTER. This allows the program to activate when a signal is provided by the flow meter. |
| 15 | RESET SAMPLE INTERVAL? [YES,NO] | Select YES, then ENTER |
| 16 | SELECT OPTION: EVENT MARK | Press ENTER |
| | [CONTINUOUS SIGNAL, PULSE] | Select CONTINUOUS SIGNAL, then ENTER |
| | DURING [PUMP CYCLE, FWD PUMPING ONLY] | Select FWD PUMPING ONLY, then ENTER |
| 17 | SELECT OPTION: PURGE COUNTS | Select 100 with the number keypad. This allows the pump to purge the sample tubing with a blast of air before sampling. Extraordinarily long suction line may require additional pump strokes to clear the line. |
| | # POST-SAMPLE COUNTS | Select 100 with the number keypad. This allows the pump to purge the sample tubing of sample water after sampling. |
| 18 | SELECT OPTION: TUBING LIFE | Use arrow to skip to the next step if you do not change pump tubing. During pre-season maintenance pump tubing is changed and the pump counts are re-set to 0. A warning is provided at this option by the ISCO sampler when counts approach 500,000-the recommended change condition. |
| 19 | SELECT OPTION: PROGRAM LOCK | Press ENTER |
| | [ENABLE,DISABLE] | Select DISABLE, then ENTER. Do not lock if you are sure no one will tamper with the settings. If you enable the program lock, select 3700 as the pass code. |
| 20 | SELECT OPTION: SAMPLER ID | Press ENTER |
| | SAMPLER ID NUMBER IS # | Select an appropriate ID number for the sampler with the keypad, then ENTER |
| 21 | SELECT OPTION: RUN DIAGNOSTICS | Press ENTER. The ISCO will proceed through a number of self-tests. This will take a short time. |
| | TEST DISTRIBUTOR [YES,NO] | Select YES, then ENTER. The ISCO will proceed through the distributor test checking its pathway around the carousal. If the test fails, realign the bottles, check that the end of the distributor hose does not extend below the distributor frame and then retest. |

| | | |
|---|---|---|
| 22 | RE-INITIALIZE [YES,NO] | ALWAYS ENTER "NO". If YES is selected, the sampler will re-set its program to the factory settings. |
| 23 | SELECT OPTION: EXIT CONFIGURATION | Press ENTER |

The sampler should go into "STANDBY" mode.

This concludes the configure sequence for the ISCO sampler. You should not have to enter the configure mode again unless you wish to change settings or test the distributor. When samples are collected and bottles replaced, the distributor should be checked using the subsequent Program Sequence. This sequence is also necessary to set various bottle sizes and sampling intervals.

Press ENTER to proceed to the ISCO PROGRAM sequence.

**Table 12-2 ISCO program sequence**

| STEP | OPTION | NOTES |
|---|---|---|
| 1 | [PROGRAM,CONFIGURE] SAMPLER | Select PROGRAM, then ENTER |
| 2 | [TIME,FLOW,STORM] PACED SAMPLING | Select STORM, then ENTER. The ISCO will momentarily display "TIME MODE FIRST BOTTLE GROUP." |
| | # MINUTE DELAY TO FIRST GROUP SAMPLE | Select 10 with the number keypad, then ENTER. The flood bore is seldom instantaneous, and 10 minutes generally allows the sample to be collected into the rising leg of the hydrograph. Review of historic gauge data may provide additional insight of stage change rate for each watershed. |
| 3 | TAKE # TIMED SAMPLE EVENTS | Select 1 with the keypad, then ENTER. One event is selected for the 6-350ml glass bottles used in this example. |
| | # BOTTLES PER SAMPLE EVENT | Select 6 with the keypad, then ENTER. |
| | SAMPLE VOLUMES OF # ml | Select 325 with the keypad, then ENTER. There is an approximate 10% error while filling the bottle. By selecting 325ml, the potential for overfilling the bottles is diminished. The ISCO will temporarily display "18 BOTTLES, 1800ml AVAILABLE." |
| | [TIME,FLOW] SECOND BOTTLE GROUP | Select TIME, then ENTER |
| | # MINUTE DELAY TO SECOND BOTTLE GROUP SAMPLES | Select 10 with the keypad, then ENTER. The ISCO will begin to fill the plastic 1000ml bottles immediately after the 6 glass bottles have been filled. |
| | SAMPLE INTERVALS OF # MINUTE | Select 90, then ENTER. Review of watershed flow dynamics is required to determine an adequate multi-sample array. This program will collect multiple samples during the first 4.5 hours of storm flow. |
| 4 | # BOTTLES PER SAMPLE EVENT | Select 4, then ENTER. 4-1L samples will be collected at 90 minute intervals. In this sample array, 2 bottles will be surplus. |
| 5 | # SAMPLES PER BOTTLE | Select 1 with the keypad, then ENTER. |
| 6 | SAMPLE VOLUMES OF # ml | Select 950 with the keypad, then ENTER. Choosing 950 rather than 1000 will diminish the potential of overfilling the bottles. |
| 7 | CALIBRATE SAMPLE VOLUME? [YES,NO] | Select YES, then ENTER. Calibrating the sample is only necessary during initial deployment and will be disregarded during the regular field season, unless the samples are over or under filled. |
| | PRESS MANUAL SAMPLE KEY WHEN READY | Submerge the intake end of the sample line into a container of clean water. The container volume should be at least the same volume as the container being calibrated, for example a 4L cubicontainer. Press the MANUAL SAMPLE key. |
| | | The ISCO will then run a routine of rinsing and then collecting 350ml of water. When completed, the ISCO screen will display "350 ml VOLUME DELIVERED." |
| | | Remove Bottle 1, the bottle that received the water, and measure its contents with a graduated cylinder. Enter that measurement into the ISCO with the |

| | | |
|---|---|---|
| | | keypad and it will compute the calibration. Replace Bottle 1 into the ISCO and repeat the process to confirm the calibration, or until the appropriate volume is delivered. |
| | | Replace Bottle 1 with a new (sterilized) bottle and continue programming by selecting NO when asked to CALIBRATE SAMPLE VOLUME |
| 8 | ENTER START TIME [YES,NO] | Select NO, then ENTER |
| 9 | Programming Sequence Complete | After displaying this response, the ISCO will return to the STANDBY MODE |

The ISCO 3700 Sampler is now programmed to collect a 2-liter sample for PCBs (6-300ml bottles) and four 4-liter samples (4-1L samples four times) that could be analyzed for metals, radionuclides, and other physical parameters. The PCB and the first 4-liter sample would be collected 10 minutes after the stream level rises above the predicted storm "activation" stage. The remaining three 4-liter samples would then be collected every 90 minutes encompassing 4 ½ hours of the event.

The flow meter must be programmed to activate the ISCO sampler program at a prescribed stage, and record the stream hydrograph and sample history.

## 12.3 ISCO Model 4230 "Bubbler" Flow Meter Programming Instructions

Turn on the flow meter and press the "ENTER PROGRAM STEP" key

Press ENTER to proceed to the Flow Meter SET-UP sequence.

**Table 12-3 Flow meter set-up sequence**

| STEP | OPTION | NOTES |
|---|---|---|
| 1 | SELECT OPTION [PROGRAM, SET-UP] | Select SET-UP, then ENTER |
| | A menu screen appears that includes all of the SET-UP options. Use the arrow key to scroll to individual options. When the desired option is flashing, press the ENTER key and adjust the parameter. Adjustments are made using the number keypad and the ENTER key. When all SET-UP options are adjusted, or you desire to return to the preceding screens, press the EXIT PROGRAM key. | |
| 2 | SET CLOCK | Press ENTER, adjust the date and time using the number keypad, and press ENTER. It should be synchronized with the ISCO sampler. |
| 3 | SITE I.D. | Synchronize with the site identification number used in the sampling program. |
| 4 | MEASUREMENT SET-UP | Press ENTER |
| | A menu screen appears with 4 options: LEVEL READING INTERVAL, DO/PH READING, PURGE INTERVAL, and SUPERBUBBLER MODE. Adjust each option using the arrow and ENTER keys. The following are recommendations and field requirements may necessitate further adjustments. Some options require greater demands on the battery. When the following options are adjusted, select EXIT PROGRAM and ENTER to return to previous menu. | |
| | LEVEL READING INTERVAL | Select CONTINUOUS, then ENTER |
| | DO/PH READING INTERVAL | Disregard this option. Dissolved oxygen or pH meters are not utilized with ISCO equipment by SWQB at this time. |
| | PURGE INTERVAL | Select 30 MINUTES, and ENTER. This establishes the time between air purges of the bubbling line. Streams with large suspended sediment loads, containing large amounts of debris, or that are aggrading may require purge intervals of greater frequency. In ephemeral streams, the purge interval could be greater, conserving battery life. |
| | SUPERBUBBLE MODE | Select ON, then ENTER. This mode supplies an additional, stronger blast of air to clear the air tube. It also requires additional demands on the battery. |
| 5 | STATUS | Press ENTER. The flow meter will momentarily display model, revision #, and identification information. Or press ENTER to proceed to the battery status. |

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| | The SUPPLY VOLTAGE should be approximately 12 volts (at least greater than 11.5 volts) and the PUMP DUTY CYCLE should be less than 2%. If either is out of conformity, field maintenance is required. Replace the battery, investigate air supply problems, and/or replace the flow meter. This step should be made during each field visit. | |
| 6 | ENABLE/ALARM HYSTERESIS | Disregard this option. The telemetry requirements for this step are not available through SWQB at this time. |
| 7 | OPTIONAL OUTPUTS | Disregard this option. The telemetry requirements for this step are not available through SWQB at this time. |
| 8 | REPORT SET-UP | Press ENTER |
| | A menu screen appears with 2 options, REPORT A and REPORT B. The following recommendation will provide a daily report and a weekly report. After REPORT A is revised, an option to revise REPORT B will be provided. First, select REPORT A, then ENTER. A menu screen appears with 4 options: FLOW, DO/PH, SAMPLE HISTORY, and FLOW METER HISTORY. | |
| | FLOW | Press ENTER and include LEVEL. Do not include FLOW RATE and RAINFALL in the report. The FLOW RATE will be developed from recorded level (stage) and channel dimensions by computer programs. This increases the storage capacity of the data collection device. Rainfall gauges are not available through SWQB at this time. |
| | PH OR DO IN REPORT | Press ENTER, select NO, and then ENTER again. A TEMPERATURE selection is then presented, select NO and press ENTER. Temperature, pH and DO meters are not available through SWQB at this time. |
| | SAMPLE HISTORY IN REPORT | Select YES, then ENTER. The sample history will include the bottle number, date, time and failed attempts for each sample. |
| | FLOW METER HISTORY | Select YES, then ENTER. The flow meter history will include the program, date and time of each adjustment made to the flow meter. |
| | When these options are adjusted, select EXIT PROGRAM, then ENTER to return to the previous menu. Select REPORT B and include the settings entered for REPORT A. The report frequencies will be entered later in the program. | |
| 9 | LCD BACKLIGHT MODE | Select KEYPRESS TIMEOUT, then ENTER. This selection conserves the battery. |
| 10 | PROGRAM LOCK | Select NO, then ENTER |
| 11 | PROGRAM | Press ENTER. This selection returns the flow meter to the original menu. |

This completes the set-up options. The flow meter is now ready to be individually programmed specifically to the site location

Press ENTER to proceed to the Flow Meter PROGRAM sequence.

**Table 12-4 Flow meter program sequence**

| STEP | OPTION | NOTES |
|---|---|---|
| 1 | SET-UP OPTION [PROGRAM, SET-UP] | Select PROGRAM, then ENTER |
| 2 | LEVEL UNITS OF MEASURE | Select IN (inches), then ENTER |
| 3 | FLOW RATE UNITS OF MEASURE | Select CFS (cubic feet per second), then ENTER |
| 4 | TOTALIZED VOLUME UNITS OF MEASURE | Select CF (cubic feet), then ENTER |
| 5 | RAINFALL UNITS OF MEASURE | Select NOT MEASURED, then ENTER |
| 6 | PH UNITS OF MEASURE | Select NOT MEASURED, then ENTER |
| 7 | DISSOLVED OXYGEN UNITS OF MEASURE | Select NOT MEASURED, then ENTER |
| 8 | TEMPERATURE UNITS OF MEASURE | Select NOT MEASURED, then ENTER |
| 9 | FLOW CONVERSION TYPE | Select MANNING, then ENTER |
| 10 | MANNING CHANNEL-CHANNEL SHAPE | Select U-CHANNEL for most channels experienced in SWQB storm water sampling, then ENTER. If the unit is deployed at a weir or flume, select the appropriate shape. |
| 11 | MANNING U-CHANNEL | Two selections will be adjusted. The channel SLOPE and its ROUGHNESS. Survey measurements are required for these, the channel width, and the maximum head. These values will subsequently be entered by the number keypad. For this exercise, select SLOPE, enter 0.015 (1.5%), and then ENTER. Enter 0.035 for ROUGHNESS, and then ENTER. These are common values for |

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| | | arroyos of northern New Mexico. |
| 12 | MANNING U-CHANNEL WIDTH | Enter the measured channel width at bankfull, in inches, and then ENTER. |
| 13 | MAXIMUM HEAD | Enter the measured channel depth at bankfull, in inches, and then ENTER. |
| | A momentary pause will occur while the flow meter computes the maximum flow that may occur for these dimensions. For this example, assume an arbitrary width of 96 inches (8 feet) and depth of 12 inches (1 foot). The meter will calculate flow at bankfull to be 14 cfs. Note this arbitrary flow, as it will be used later when adjusting the REPORTS option. Press ENTER. | |
| 14 | PARAMETER TO ADJUST | Select LEVEL, and the CURRENT LEVEL will be displayed. Water level measurements will be required for this adjustment. Enter the water level measured in inches above the air tube, and then ENTER. If the channel is dry, enter 0, and then ENTER. |
| | The screen will return to PARAMETER TO ADJUST. This time select NONE, and then ENTER. This adjustment is required during each visit to adjust for "drift" in the calibration. | |
| 15 | FLOW TOTALIZER: # cf | This is an information window screen and describes the total flow that has occurred since the flow meter totalizer was last reset. Press ENTER. |
| 16 | RESET FLOW TOTALIZER | Select YES, then ENTER |
| 17 | ENABLE TOTALIZER | This flow is used for pulse-generated sampling routines and can be disregarded, since this routine is established for time-generated samples once the initial pulse is sent to initiate sampling. Press ENTER and adjust the following RESET condition to 0, or select NO, and then ENTER. |
| 18 | SAMPLER PACING | Select DISABLE, then ENTER. This option is also for pulse-generated sampling that could enable the sampling rate. Samples could be collected based on flow volume. Due to the unpredictability of storm events in this area, SWQB uses time based sampling. |
| 19 | SAMPLER ENABLE MODE | Select CONDITIONAL, then ENTER. This option provides the logic command that will signal the sampler to begin its sampling program. |
| 20 | CONDITION | Select LEVEL, then ENTER |
| 21 | LEVEL | Select GREATER THAN, then ENTER |
| 22 | LEVEL GREATER THAN # in | Use the number keypad to select a level rise for a predicted storm event. For this example, select 5 and ENTER. The flow meter will signal the ISCO sampler to begin sampling once the water level rises 5 inches. In most instances, 5 inches will capture most storm events. There is a risk of missing a later, more significant event. This is a judgment that will be made more accurately as one becomes more familiar with individual watersheds. |
| 23 | OPERATOR | Select DONE, then ENTER |
| 24 | WHEN ENABLE CONDITION IS NO LONGER MET | Select KEEP ENABLED, then ENTER. Allowing the sampler to remain enabled once the stage drops below the 5 inch (for this example) activation stage provides the opportunity to continue collecting at the trailing leg of the hydrograph. |
| 25 | ENABLE CURRENTLY LATCHED, RESET? | Always choose YES. This will clear the enable signal to the sampler and prepare it to accept a new signal from rising water of the next storm event. |
| | This option occurs only if the sampler has been enabled, but failing to reset the latch is one of the most common and most serious mistakes made when reprogramming the ISCO sampling equipment. | |
| 26 | PLOTTER ON/OFF WITH ENABLE | Select YES, then ENTER |
| 27 | ALARM DIALOUT | Select DISABLE, then ENTER. The telemetry equipment for this option is not available through SWQB at this time. |
| 28 | PLOTTER SPEED | Select ½ in/HR, then ENTER. Greater plotter speeds create unnecessary battery and plotter paper supply demands. |
| 29 | INPUT FOR PLOTTER LINE A | Select LEVEL, then ENTER |
| 30 | PLOTTER LINE A FULL SCALE | Select 15 inches, then ENTER |
| | Select a value that is greater than the maximum head at bankfull and retains a grid easily read on a paper scroll. For example, the maximum head for this exercise is 12 inches and the grid contains 10 increments on the paper scroll. Therefore, by selecting 15 inches, each increment is equal to 1.5 inches. The plotter gives a hard copy of flow data to back up electronic copies collected with the rapid transfer device. | |
| 31 | INPUT FOR PLOTTER LINE B | Select FLOW RATE, then ENTER |
| 32 | PLOTTER LINE B FULL SCALE | Select 15 cfs, then ENTER |
| | The maximum head for this exercise is approximately 14 cfs. Choosing 15 cfs generates a hydrograph with an appropriate scale that can be easily read. | |
| 33 | INPUT FOR PLOTTER LINE C | Select OFF, then ENTER |
| 34 | REPORT GENERATOR A | Select ON, then ENTER |
| 35 | REPORT A DURATION TO BE IN | Select DAYS, then ENTER |

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| | The following options generate reports on daily and weekly periods. Useful information regarding maximum, minimum and average daily flows, sample history, and flow meter program adjustments are contained in these reports. | |
| 36 | REPORT A DURATION | Select 1 day with the number keypad, then ENTER |
| 37 | PRINT FIRST REPORT A AT | Adjust the date and time the first daily report will be expected. Commonly the following day at midnight is selected. |
| 38 | REPORT GENERATOR B | Select ON, then ENTER |
| 39 | REPORT B DURATION TO BE IN | Select DAYS, then ENTER |
| 40 | REPORT B DURATION | Select 7 days with the number keypad, then ENTER |
| 41 | PRINT FIRST REPORT B AT | Adjust the date and time the first weekly report will be expected. |
| 42 | PRINT FLOW METER HISTORY | Select NO, then ENTER |
| 43 | CLEAR HISTORY | Select NO, then ENTER |

This concludes the flow meter programming sequence. Return to the ISCO sampler. It should be in standby. Hit the START SAMPLING key. The sampler should display SAMPLER INHIBITED.

You are ready for the next storm event.

## 12.4 Collecting Samples and Resetting the ISCO

After a storm event activates the equipment and the system runs through its entire program, the sampler will display DONE, or PROGRAM HALTED. Press the DISPLAY STATUS key. The following logic sequence provides the date and times of the enable signal, and for each attempted sample bottle collection. It also provides abbreviated messages describing potential failures. Record this data in a field logbook or field sheet to backup data from the RTD.

Label each bottle with site ID, bottle number, and date and time. Cap with clean bottle lids. The lids are commonly stored in sealed containers that are stored within the ISCO carousel. The bottles are then replaced individually, or a fresh carousel loaded with clean sample bottles is installed. Remove the lids and store them in a sealed container within or near the sampler.

Review the RUN DIAGNOSTICS option under the configure menu to test the computer RAM/ROM and distributor. Occasionally the bottles become misaligned and prevent the sampler armature from rotating. Under CONFIGURE use the arrow keys to scroll directly to the RUN DIAGNOSTICS option, and then press ENTER.

If necessary, the ISCO program may be adjusted by scrolling through its menu options. The times, volumes, and sample array may be adjusted. Enter the PROGRAM menu, press the ENTER key to pass through each option, or adjust each setting that is required.

When this is completed and the sampler is back in standby, review the following flow meter program steps: the level adjustment, RESET TOTALIZER, and SAMPLER ENABLE. These steps are listed on the front of the flow meter and can be quickly accessed by using the GO TO PROGRAM STEP key on the meter. Adjust the level if necessary, reset the totalizer, and most importantly RE-LATCH the ENABLE function.

***Re-latch the enable function after resetting the sampler:***

Under the SET-UP menu, got to the STATUS option.

Check and record the battery and power supply status.
When the flow meter has been inspected, return to the sampler and press the START SAMPLING key.
The sampler should read 'SAMPLER INHIBITED". You are ready for the next storm event.

## 12.5 Rapid Transfer Device

The Rapid Transfer Device (RTD) is a field data collection unit that can retrieve data from multiple ISCO flow meters. The device uploads that information to a desktop through the ISCO Flowlink® program. Flowlink can then generate individualized reports including stream hydrographs, flow, and sample history.

The RTD automatically collects the available data reports from multiple instruments in the field and saves each report separately. The RTD connects directly to a PC with its own power cable and ISCO software. The data collected from every site will be uploaded to files on the PC's hard drive. When the data reports have been uploaded from the RTD at least once, the RTD is ready for use again in the field.

*Rapid Transfer Device Instructions*

The RTD has three colored indicators on one end. These lights report the status of the RTD and show the result of each data transfer.

**Collecting Data with the RTD:**

Plug the RTD into the instrument's interrogator connector. The interrogator connector is labeled with the interrogator icon, a small computer icon.

While the RTD is operating, the yellow light will blink to indicate power, and then the green light will blink as the data reports are collected.

The green light will remain constant when the transfer is successfully completed. With this light constant, you can be sure that all the data has been collected and stored within the RTD.

A red light reports two other RTD conditions: *Memory Full* or *Transfer Error*.

A constant red light indicates a *MEMORY FULL* condition. There is not enough RTD memory available to collect data reports from another instrument similar to the one now connected. Check the green light to see if the last transfer was successful. Uploading the data reports from the RTD to your PC files will allow the RTD to reuse the memory.

A blinking red light reports a *TRANSFER ERROR*. The RTD was unable to complete the transfer. Unplug the RTD, wait briefly, and repeat step 1. If that does not work after retrying, check the voltage level of the instrument's power supply, then make sure the connection between the RTD and the instrument is clean and dry. If this fails, call the ISCO Customer Service Department.

You can upload the data collected by the RTD to your personal computer with ISCO Flowlink software, currently available only to the DOE Oversight Bureau.

**Uploading Data to a PC:**

"Flowlink 4" uploads text reports and detailed data for management of your monitoring data. The data is controlled as site files in a database. It is used for analyzing, viewing, editing, and printing. In addition, custom graphs, summaries, and reports may be produced.

Your personal computer must have a serial RS-232 port and ISCO's Flowlink software installed. Connect the RTD to your PC with the RTD Power Cable. This cable powers the RTD through a wall-outlet connection, and links the RTD to the PC's serial port.

To upload data with ISCO's "Flowlink 4," Refer to the Flowlink Tutorial embedded in the software.

**RTD Data Capacity**

The RTD can collect data from a maximum of 255 ISCO instruments, or until the maximum storage capacity of over 900 kilobytes is reached. When you reach either maximum value, the RTD will not accept more data until the data already collected is uploaded to a PC. After the data reports are successfully uploaded from the RTD, the storage space occupied by those reports is made available to collect new data from field instruments.

## 12.6 Teledyne ISCO 1640 Liquid Level Actuator

The Liquid Level Actuator (LLA) is a device used in conjunction with an ISCO automated sampler to begin a sampling routine when the liquid level reaches a predetermined height. The LLA consists of a control box assembly connected to the end of a 22 ft. coaxial cable and is used when a flow meter is not available to activate the sampler. A crest gauge is then necessary to provide an element of event flow.

*Connect to ISCO Sampler*
The LLA is supplied with two Velcro strips: one permanently attached to the switch box, the other with an adhesive strip for permanent attachment to the sampler control box. After attaching the LLA switch box to the sampler control box, ensure the sampler is off and connect the military connector at the end of the LLA coaxial cable to the "flow meter" socket on the sampler.

*Mounting the Probe Assembly*
The LLA is supplied with an assembly clamp that allows for mounting the probe end in the stream channel. A stake or length of rebar should be inserted into the stream bed at a stable location, where it is unlikely to be washed out, buried by aggradation, or cluttered by litter.

Attach the assembly clamp to the stake at the appropriate height, where water depth should be measured. The probe is mounted over the stream with the stainless steel pin pointing down toward the liquid. A plastic rain deflector is provided. Position this so that it nearly covers the probe, preventing rain from wetting the probe and accidentally activating the sampler. The vent

hole in the side of the rain deflector must be kept unobstructed so air can escape as the liquid level rises inside of the rain deflector.

In most cases, The LLA will activate the sampler when the liquid touches the stainless steel ring inside of the rain deflector. The probe assembly should be mounted with the stainless steel ring positioned at the height where the sampler is to be activated. Conversely, if the sampler is powered by a 117 VAC power pack, the LLA will actuate the sampler when the liquid touches the probe tip, rather than the stainless steel ring.

*Operating the LLA*
The LLA is switch-selectable for either LATCH or TOGGLE/RESET operation. The switch is located on the switch box, which should be mounted to the sampler control box.

When the switch is set to LATCH, the LLA will activate the sampler when the liquid level rises to the stainless steel ring on the probe assembly. The sampler remains activated even if the liquid level recedes, thus completing the entire program and requiring the sampler be reset. This allows any minimal flow at the tailing leg of the hydrograph to be collected, but there may be insufficient flow to collect after the level drops below the ring. The sampler will continue trying to complete its timed programming sequence, thus prohibiting capture of a later surge from the same event.

When the switch is set to TOGGLE/RESET, the sampler takes samples only while the liquid is touching the probe assembly. When the liquid level rises to the probe assembly, the LLA activates the sampler, and the sampler will continue taking samples only as long as the liquid touches the stainless steel ring. If the liquid level recedes, the sampler will be inhibited until the liquid level again rises to the probe assembly. If the liquid level recedes while the sampler is taking a sample, it will finish taking the sample before shutting off. This allows the sampler to pause in the midst of its programming sequence and save any remaining bottles for a later storm surge.

The TOGGLE/RESET switch selection is also used to reset the LLA.

*Sampler Programming*
The sampler must be configured to respond to the actuator. All ISCO samplers can be used in either TIME or FLOW mode when used in conjunction with the LLA. If the FLOW mode is used, a “Y” connect cable will be necessary to connect both the LLA and a flow meter to the FLOW METER port on the sampler. For most SWQB activities, the sampler will be set to TIME mode when used in conjunction with a LLA.

To program the sampler and LLA when the actuator is set to LATCH:

1. Program the sampler for the time interval (or number of flow pulses, when desired) between samples following the ISCO instructions.

2. Momentarily set the LLA control switch to the TOGGLE/RESET position in order to reset the actuator.

3. Set the CONTROL switch to the LATCH position.

When first activated, the sampler will immediately take a sample. The sampler will then return to its prescribed time base and take samples at the programmed time intervals. If, after the sampler and LLA have been turned on, the liquid level rises to the probe before one time interval between samples (as programmed on the sampler manually) has passed, the sampler will wait until the end of the first time interval to take the first sample. Thereafter, it will take samples as programmed.

To program the sampler and LLA when the actuator is set to TOGGLE/RESET:

1. Program the sampler for the time interval (or number of flow pulses) between samples following the instructions given in the ISCO automated sampler SOP.
2. Set the LLA CONTROL switch to the TOGGLE/RESET position.

When the liquid level rises to the probe assembly, the LLA activates the sampler. The sampler will react just as it does to the LATCH setting. However, if the liquid level drops below the stainless steel ring, the sampler will stop collecting.

Additional information is available in the 1640 Liquid Level Actuator Installation and Operation Guide, published by Teledyne ISCO, December 2004.

## 12.7 D-Tec, Inc. Environmental Liquid Sampler

Prior to Environmental Liquid Sampler (ELS) installation, make sure that all activities are in compliance with NEPA requirements or facility specific permits and regulations (e.g. excavation permits). The methods described below were developed for sampling in contaminated soil in and around Los Alamos National Laboratories. Such thorough personal protection may not be necessary at all locations.

*Description*
The ELS is a mechanized, spring loaded cap that threads onto a sample container, allowing surface water to flow into the container until it is filled. The ELS then snaps shut as a float valve is activated, preventing mixing of the sample with the atmosphere and flow occurring later in the hydrograph. The container must be buried in the channel where the sample is to be collected, with the cap positioned at a level suitable for collection of stream flow.

*Location*
A location with soil deep enough to install the sampling jar 12 to 15 inches deep is necessary. Check the depth to bedrock by hammering a length of rebar into the channel bed.

*Excavation*
Disposable gloves must be worn to protect from potential contaminants during the excavation process. Use a large, heavy-duty plastic bag marked with the site identification number is used to contain the removed soil. The soil removed must be the same soil put back into the excavated hole when the project is completed. Pursuant to RCRA land disposal regulations or other

specific regulations or permits, no soil is to be casually disposed of at the site or in the watercourse (erosion channel) except to refill the excavated hole. A posthole digger is usually used to excavate; however, if the soil is very sandy, the hole may also need to be excavated with a trowel and/or by glove protected hand. Two sampling jar sizes can be used: a two-liter plastic or a four-liter glass jar. Excavate a hole deep enough so that the top of the sampling jar rests flush with the ground so when the ELS is installed on the sampling jar shallow sheet flow will be collected. The excavation must also be wide enough so to accommodate at least one inch of space around the jar. Remove any rocks that could possibly damage the sampling jar and remove any loose soil with trowels. Place all of the soil in the marked bag.

***ELS Placement***

Place the sampling jar in the excavated space. Do not remove the lid of the sampling jar until the ELS is ready to be installed. Make sure that the jar is placed in the hole so that it is centered, straight, and level. A mounting frame is provided to anchor the jar in the hole with a length of rebar. Carefully place some of the soil from the marked bag back into the space around the jar. Fill the space until it reaches the neck of the jar. If the ELS is ready to be installed, tamp the soil around the jar, and remove the lid and any soil from around the lip of the jar. Be careful that no soil falls into the sampling jar once it is open. Place removed jar lids in a separate plastic bag so that they can be decontaminated for reuse.

The person handling the ELS must be independent of the excavation process or have removed any contaminated gloves used in the excavation process. Open the sampling port of the ELS and set the trigger mechanism. Test the trigger mechanism to be certain it will close when a storm water sample is collected, and then reset it. Carefully place the ELS on the sampling jar and gently tighten it. The tread of the ELS and the tread of the jar must be dry for proper attachment of ELS and jar. Be careful not to trigger the mechanism while tightening, otherwise it must be set again. A person wearing disposable gloves must carefully remove any materials that will interfere with water flow or clog the ELS. Spread and firmly tamp the soil around the ELS to allow for unobstructed flow.

*Completion*

Prior to leaving the site, place all equipment in one area away from the ELS. Hammer a rebar into the ground in a spot near the sampling location but where it will not interfere with the water flow. Write the site identification number on a yellow pin flag and attach the pin flag to the rebar with duct tape.

Avoid walking upstream from the ELS so that no excess erosion will occur. Take the equipment and any trash back to vehicle.

While wearing gloves, use a clean paper towel thoroughly rinsed with distilled water to wipe all surfaces of the tools that came in contact with the soil. Wipe surfaces dry with a clean paper towel and place equipment in vehicle. Dispose of paper towels and gloves in a marked trash bag.

Record the date, the names of people doing the installation, GPS coordinates and the site location identification number (this identification number will be used to identify the samples collected at

that site). Indicate in the field notebook other pertinent information (e.g. describe ELS location relative to BMPs at the site).

## 12.8 Crest Gauges

The crest-stage gauge is a simple, economical, reliable, and easily-installed manual device used to obtain the elevation of the flood crest of streams. The design used most often is a vertical piece of 2-inch galvanized pipe containing a wood or aluminum staff held in a fixed position with relation to a datum reference. The bottom cap has six intake holes, facing upstream, and spaced 30º apart across the face of the cap, so as to keep the non-hydrostatic draw-down or inside the pipe to a minimum (Figure 11.1). Tests have shown this arrangement of intake holes to be effective with velocities up to 10 feet per second, and at angles up to 30 degrees with the direction of flow. The top cap contains one small vent hole. The bottom cap, a perforated tin cup, or copper screening formed into a cup shape and attached to the lower end of the staff contains regranulated cork. As water rises inside the pipe, the cork floats on its surface. When the water reaches its peak and starts to recede, the cork adheres to the staff inside the pipe, thereby retaining the crest stage of the flood. The gauge height is obtained by measuring the interval on the staff between the reference point and the flood mark. Scaling can be simplified by graduating the staff. A crest gauge should placed in a location that allows personnel to readily access the gauge and reset the cork. Gauges are usually affixed to bridge abutments, supported by T-posts in the channel, or attached to structures in the stream, such as framework associated with USGS or LANL gauging stations.

Calibrate the gauge by measuring the difference between the lowest point on the stream bed and the zero point on the scale marked on the staff. Note that the lowest point on the stream bed won't necessarily be the point where the gauge is located, but it must be along the same cross section. This "depth-of-stream factor" must always be added to the gauge readings. Recalibrate the gauge yearly, and also after severe episodes of erosion or deposition.

To read the crest gauge, remove the top end cap and take out the dowel. Use the scale on the staff to measure the level of the cork powder "ring". Don't forget to add the depth-of-stream factor. After taking the reading, wipe off the cork powder and replace the dowel. Crest gauges should be cleaned and reset after periodic floods. The crest gauge should also be inspected periodically for damage and level errors. Occasionally you may need to add more cork powder. After initial installation of the crest stage gage, a cross section through the crest stage gage location should be surveyed with the laser level and survey rod using the Reference Reach Cross Section Form (Appendix A). This level of information is necessary to develop a stage-flow relationship that relates this cross section to the recorded gage measurements. The cross section needs to be re-surveyed after channel-altering flow events to re-establish this relationship.

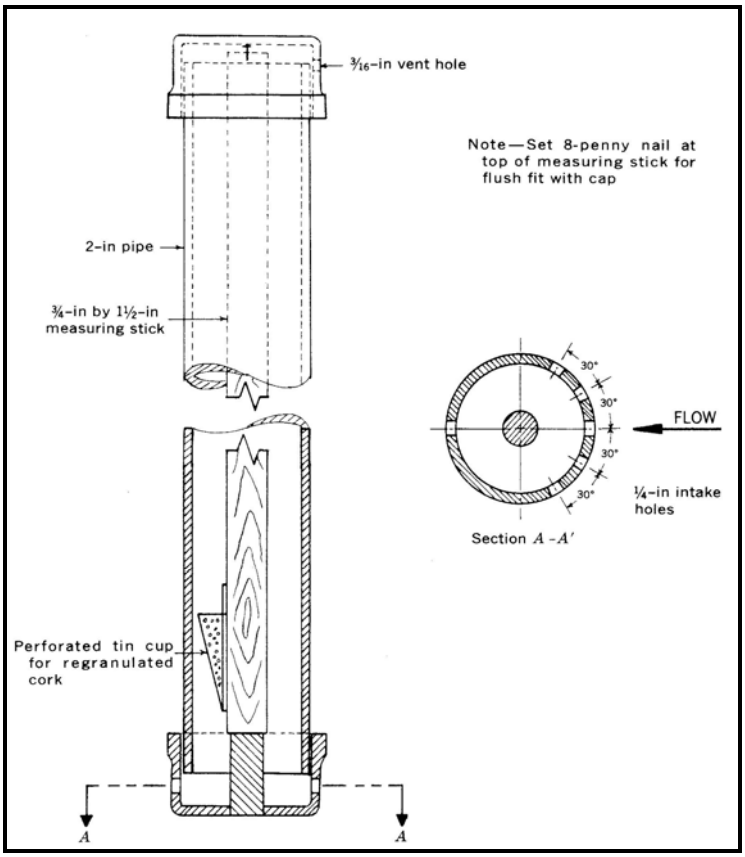



**Figure 12-1 Crest gauge design**

## 12.9 In-Situ, Inc. Level "Troll" Pressure Transducer

Level Loggers are small, cylindrical data loggers that have been equipped with a pressure sensor to detect the height of the water column above the sensor. This height is measured and stored at preset intervals depending on data needs: more memory for long periods between downloads or a more precise hydrograph with frequent measurements. SWQB uses In-Situ, Inc. "Level Troll 500" loggers.

For channel discharge estimates, loggers are installed into conduits that were originally designed for crest gauge operations (Figure 11.1). The transducer is attached to the dowel at the bottom of the casing and an offset is recorded to establish a datum at this arbitrary height-this offset is added to the logger's measured depth to find channel depth for discharge calculations.

Loggers may be programmed using a desktop PC, or in the field using a Pocket PC *Rugged Reader* that is produced by In-Situ. "Win-Situ" software is used to communicate with the devices and manage data files. A thorough description of system setup and operation is available from the *Level Troll Operator's Manual*, produced by In-Situ, Inc. (2007).

To initiate logging of a particular *Troll* using the *Rugged Reader*, use the following sequence:

1. Connect the PC to the *Troll* using a programming cable, with a DB9-pin serial port to the PC and a twist-lock connector to the *Troll*.
2. Launch the software using the stylus on the PC surface: Start → Programs → Win-Situ Mobile.
3. Ask the software to connect to the device: File → Connect. The screen should show a reading of each measured variable: temperature, pressure and depth.
4. Set up a data log to create a file for the data associated with a particular place and time: View → Logging → Expander (the blue arrow button "▲" on the bottom right corner) → New
5. Enter basic info: Site name (i.e. Pueblo Canyon above Acid Canyon) and Log File Name (i.e. May 12, 2007).
6. Tap Next "→"
7. Select parameter(s): Pressure, Temperature, & Depth.
8. Order and Units: select units "C", "ft", & "PSI". Tap Next →
9. Select Log Type: "Linear". Tap Next →
10. Measure and Store Values Every: 5 minutes. Tap Next →
11. Set Start/Stop Time: Scheduled start as soon as the unit is deployed. No Stop Time. Tap Next →
12. Select Log Wrap Mode: This selection should not be necessary, as there is sufficient memory on the *Troll* to extend through the monitoring season. However, as the memory should be downloaded after each storm event, select "Wrap Log When Full" to ensure new data is preserved by overwriting old data in the event the memory should become full-this will overwrite data that has supposedly been downloaded following the previous storm event. Tap Next →
13. Select Level Mode: Depth of Probe. Tap Next →
14. Select Specific Gravity: Fresh. Tap Next →
15. Summary: Review Settings. Tap Enter √
16. Upon completion of programming, disconnect the *Troll* recap the sensor. It will begin to log at the preset date and time and continue to do so until it is manually stopped.

To stop logging and retrieve a data file from the *Troll*, use the following sequence:

1. Connect the PC to the *Troll* using a programming cable, with a DB9-pin serial port to the PC and a twist-lock connector to the *Troll*.
2. Launch the software using the stylus on the PC surface: Start → Programs → Win-Situ Mobile.
3. Ask the software to connect to the device: File → Connect. The screen should show a reading of each measured variable: temperature, pressure and depth.
4. Tap View → Logging → Choose the Sensor from the list of Sensors that have been deployed (i.e. Pueblo above Acid) → Choose the specific log (i.e. May 12, 2007). Tap the Expander "▲"
5. Tap Download → Download All → Tap Enter √
6. Upon completion of Download, Tap Enter √

7. You may view the data now to assure it has downloaded properly or decline the option and move on. The data file is now stored on the PC.
8. A new log must be set-up to restart the *Troll*'s datalogging routine.
9. Upon synchronization with the desktop PC to which the *Rugged Reader* is associated, data files will be transferred to the desktop for permanent storage and assessment.

**FORMS**

Reference Reach Cross Section Form

# 13.0 PHYSICAL HABITAT MONITORING

Physical habitat monitoring will follow EPA EMAP-West Field Operations Manual for Wadeable Streams and EMAP Field Operations and Methods Manual for Non-Wadeable Streams for wadeable and non-wadeable waterbodies, respectively (EPA 2006, 2000).
(http://www.epa.gov/wed/pages/publications/authored/EPA620R-06003EMAPSWFieldOperationsManualPeck.pdf)
(http://www.epa.gov/emap/html/pubs/docs/groupdocs/surfwatr/field/R5_remap.pdf).
All other decisions made in the field to collect data outside of the EMAP procedures should be documented with available evidence on the EMAP Alteration Form. This includes data not collected at an entire site due to scour, dryness, or weather conditions. This also includes any alteration to protocol because of loss/damage of equipment, shortage of time, or any other reason.

Physical habitat parameters are selected to provide data on eight general physical habitat attributes important in influencing stream ecology (Stoddard, 2005):

- Habitat Volume/Stream Size
- Habitat Complexity and Cover for Aquatic Biota
- Streambed Particle Size
- Bed Stability
- Channel Riparian and Floodplain Interaction
- Hydrologic Regime
- Riparian Vegetation Cover and Structure
- Riparian Disturbance

These attributes were identified during EPA's 1992 national stream monitoring workshop as those that are essential to evaluate physical habitat in regional monitoring and assessment programs (Stoddard, 2005). Each of these attributes may be directly or indirectly altered by anthropogenic activities. Their expected values tend to vary systematically with stream size (drainage area) and overall gradient. There are 31 metrics that characterize these 8 attributes of physical habitat. Mean channel dimension, wood volume, substrate diameter, bed shear stress, and riparian vegetation cover and complexity are examples of metrics that allow quantification of attributes (e.g., lotic habitat quantity) through calculations of residual pool frequency and size distribution (Stoddard, 2005).

The habitat survey will be conducted concurrent with bug and periphyton sampling during the biomonitoring index period in mid-August to mid- November, and at least four weeks after a scouring flow event whenever plausible (Biggs, 2000). Bio sampling will be recorded on the Sample Collection form along with an additional form for the Periphyton – Nutrient Level 2. If a site is dry upon visitation, the visit should be recorded as such and no measurements taken. If a site has enough small pools to take biological samples, the full survey should be completed with zero depths recorded in dry cross section areas and notes taken on the level of low flow.

U.S. EPA's Environmental Monitoring and Assessment Program (EMAP) has a documented set of procedures for collection of field data and samples in both wadeable and non-wadeable rivers. The Western Pilot Study (EMAP-West) has refined these procedures for wadeable streams (stream order 1-3) in semi-arid and arid regions of the western United States. A brief description of the procedures for physical habitat monitoring will be listed, and before implementing this survey, the full physical habitat characterization procedure, section 7 of the EMAP Western Pilot Study Field Operations Manual for Wadeable Streams, should be read by all staff involved in the survey. Field staff should also have EMAP training in the field before being able to participate on an EMAP field crew. The training will be provided by the MAS EMAP Habitat Specialist.

Figure 12.1 illustrates locations within the sampling reach where data for components of physical habitat characterization are obtained. SWQB assesses habitat over stream reach lengths that are approximately 40 times their average wetted width at baseflow, but not less than 150 m long. This allows for adjusting the sample reach length to accommodate varying sizes of streams.

These procedures are most efficient when applied in low flow conditions, while terrestrial vegetation is still active. Generally, the latitude and longitude of the site (referred to X spot or F transect in EMAP procedures) is taken at the middle of 11 transects that create 10 segments, each of which is one-tenth the distance of the surveyed reach. There are five components of habitat characterization: thalweg profile; woody debris tally; channel and riparian characterization; assessment of channel constraint, debris torrents, and major floods; and discharge.

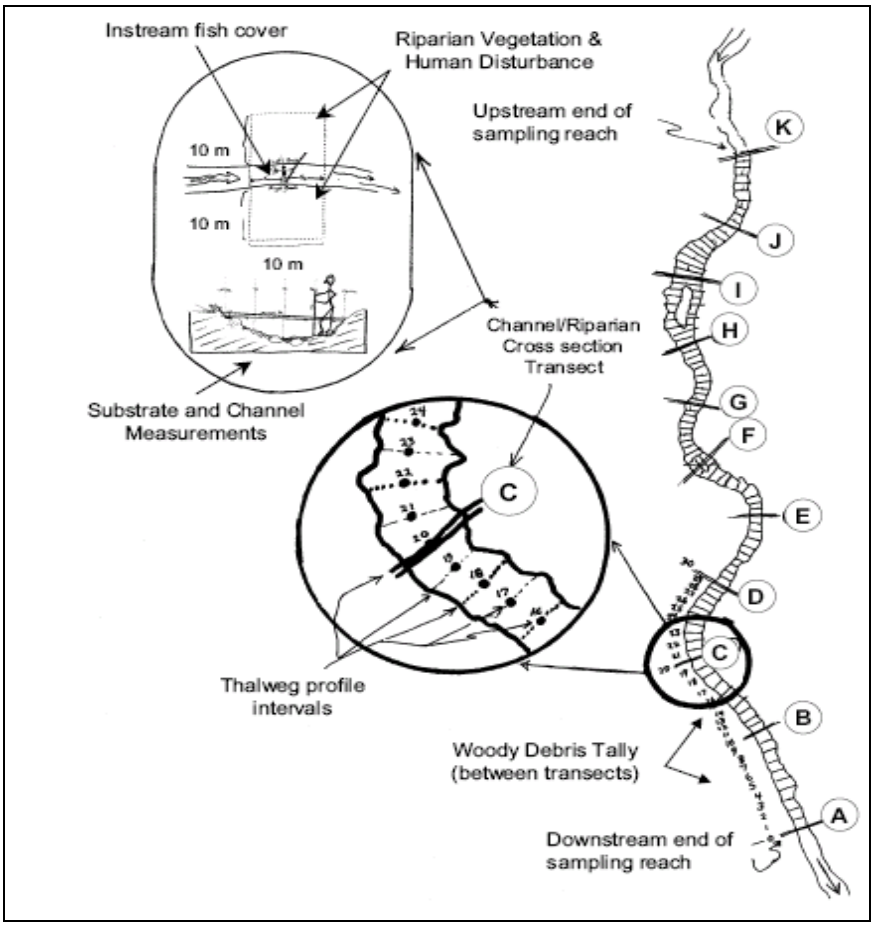



**Figure 13-1 Sampling reach layout for physical habitat measurements**

## 13.1 Equipment

- 2 surveyor's telescoping leveling rod (round profile, metric scale, 7.5 m extended)
- 1 50 m fiberglass measuring tape & reel
- 1 clinometer with percent and degree scales
- 1 lightweight telescoping camera tripod (necessary only if slope measurements are being determined by one person)
- 1 meter stick (alternatively, a short (1-2 m) rod or pole (e.g., ski pole) with cm markings for thalweg measurements, or the PVC pipe described for slope determinations can be marked in cm and used)
- 1 roll each of colored surveyor's plastic flagging (2 colors)
- 1 convex spherical canopy densiometer modified with taped "V"
- 1 bearing compass (backpacking type)
- 1 or 2 fisherman's vest with lots of pockets and snap fittings (used at least by person conducting the in-channel measurements to hold the various measurement equipment (densiometer, clinometer, compass, etc.); useful for both team members involved with physical habitat characterization)

- 2 pair chest waders (with felt-soled boots for safety and speed if waders are the "stocking" type); hip waders can be used in shallower streams
- Covered clipboards (lightweight, with strap or lanyard to hang around neck)
- Soft lead (#2) pencils
- 11 (plus extras) Channel/Riparian Cross-section & Thalweg Profile and Woody Debris Forms
- 1 (plus extras) Slope and Bearing Form; Riparian Legacy Tree and Invasive Alien Plant Form; Channel Constraint Assessment Form; Torrent Evidence Form.
- 1 set laminated sheets of procedure tables and/or quick reference guides for physical habitat

## 13.2 Logistics and Work Flow

Upon arrival at the site the Stream Verification and Stream Assessment forms should be completed. Total Nutrients and ion chemical samples should be taken from the water column and a sonde used to record physico-chemical measurements on a chemical field sampling form and SLD submittal sheet filled out as well. Also the qualitative Rapid Geomorphic Assessment and Rapid Habitat Assessment forms should be completed (See Appendix A for all forms) The five components of the habitat characterization are organized into four grouped activities that require approximately 4-5 hours to perform, depending on stream size:

1. **Thalweg Profile and Large Woody Debris Tally**. Two people (the "geomorphs") proceed upstream from the downstream end of the sampling reach (see Figure 12-1) making observations and measurements at the appropriate increment spacing. One person is in the channel making width and depth measurements, and determining whether fine sediment deposits are present. The other person records these measurements, classifies the channel habitat, records presence/absence of side channels and off-channel habitats (e.g. backwater pools, sloughs, alcoves), and tallies large woody debris. Each time this team reaches a flag marking a new cross-section transect, they begin 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 transect. When the crew member in the water makes a width measurement at channel locations midway between regular transects (i.e., A, B,...K), s/he also locates and estimates the size class of the substrate particles on the left channel margin 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 and Woody Debris field form.

2. **Channel/Riparian Cross-Sections**. One person will proceed 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, instream fish cover, and human disturbance as specified on that form. That person also makes observations to complete the Riparian "Legacy" Tree and Invasive "Alien" Plant field form. Slope and bearing are determined together by backsighting to the previous transect. Intermediate flagging (of a different color) may have to be used if the stream is too brushy, sinuous, or steep to site

for slope and bearing measures between two adjacent transects. (Note that the crews could tally woody debris while doing the backsight, rather than during the thalweg profile measurements.) Slope and bearing are recorded on the Slope and Bearing form,

3. **Channel Constraint and Torrent Evidence**. 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 debris torrents and major floods. Beginning in 2007, the flood prone width will be measured and recorded in the comments section of this page.

4. **Discharge**. After collecting chemistry samples (if any), discharge measurements are taken at an optimal cross section (but not necessarily at a transect) near the X-site and recorded on the Stream Discharge Form. However, do not use the electromagnetic current meter close to where electrofishing is taking place. Furthermore, if substantial channel disruption is necessary and sediment must be stirred up, wait until all chemical and biological sampling has been completed.

## 13.3 Thalweg Profile and Large Woody Debris Tally

### 13.3.1 Thalweg (longitudinal) Profile

The thalweg is the deepest flow path of water in the channel. In order to complete the thalweg profile, measurements should be taken at 100 - 150 evenly spaced intervals, or 10 - 15 intervals between each transect. This may be accomplished by relating interval length to channel width as follows:

1. If channel width is < 2.5 m, interval = 1.0 m
2. If channel width is 2.5 - 3.5 m, interval = 1.5 m
3. If channel width is > 3.5 m, interval = 0.01 reach length

Thalweg depth will be measured at each interval using a survey rod. If the thalweg is too deep to measure while wading, extend the rod at an angle to reach the deepest point. Then record the water level on the rod and measure the angle of the rod with the clinometer. The actual depth can then be calculated in the office by multiplying the trigonometric sin of the rod angle by the measured depth. Habitat is classified and recorded at each measurement, as well as the presence or absence of side channels and off channel habitats.

### 13.3.2 Woody Debris Tally

The large debris tally should be performed concurrent with the thalweg profile, where the recorder of depth measurements counts woody material in between the measurements. Large woody debris (LWD) is defined as that which has a small end diameter of at least 10 cm (4 in.) and a length of at least 1.5 m (5 ft). The tally includes all pieces throughout the entire reach that are at least partially within the bankfull channel (zone 1 or 2 in Figure 12.2) or bridging above it (zone 3 in Figure 12.2). Widths and lengths are visual estimations; if the debris is not cylindrical, estimate the average width.

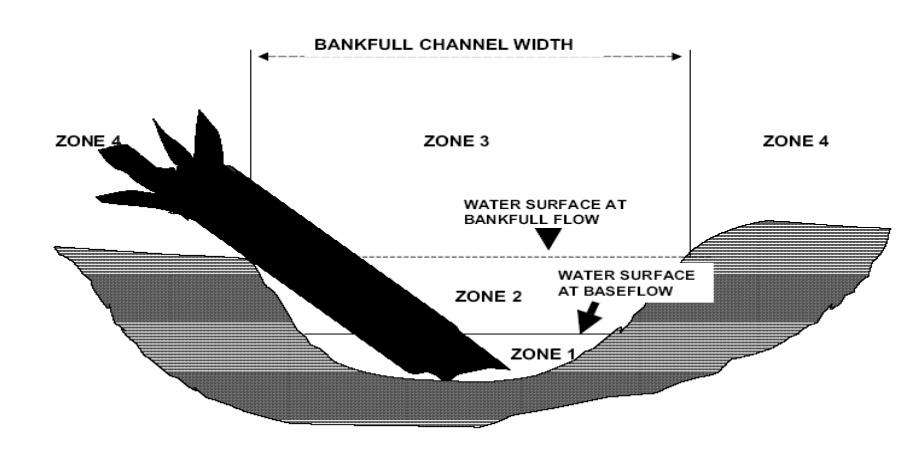
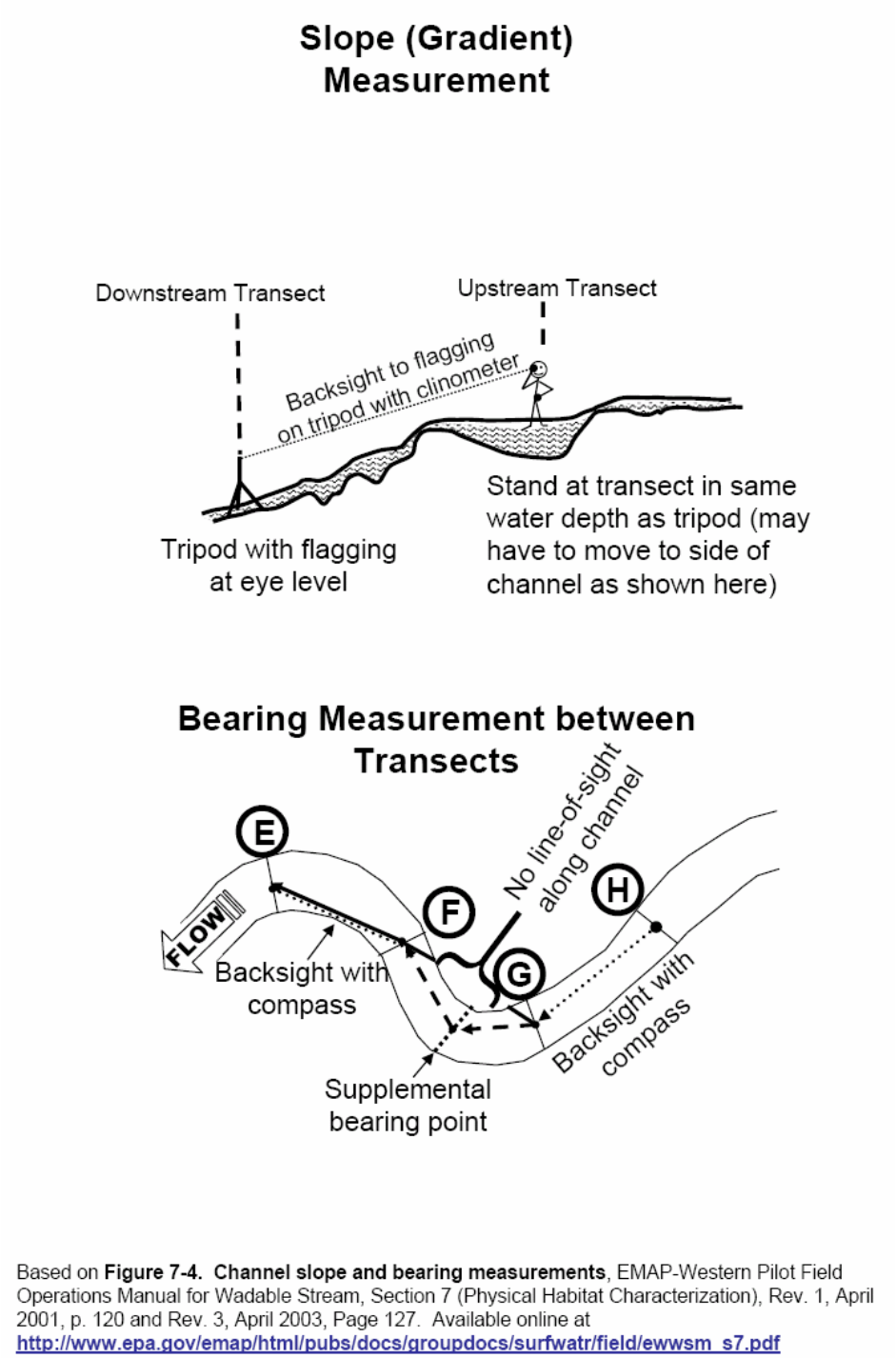



**Figure 13-2 Large woody debris in bankfull channel**

## 13.4 Channel and Riparian Measurements at Cross-section Transects

### 13.4.1 Slope and Bearing

The gradient and bearing of the stream will be measured by “backsighting” downstream between segments (see Figure 12.3).




Based on **Figure 7-4. Channel slope and bearing measurements**, EMAP-Western Pilot Field Operations Manual for Wadable Stream, Section 7 (Physical Habitat Characterization), Rev. 1, April 2001, p. 120 and Rev. 3, April 2003, Page 127. Available online at http://www.epa.gov/emap/html/pubs/docs/groupdocs/surfwatr/field/ewwsm_s7.pdf

**Figure 13-3 Slope and bearing measurements**

To measure slope, have two survey rods marked at the observer's eye level, one person at each end of the transect or one person and a tripod flagged at the observer's eye level (make sure you can see each other), and the survey rods held at the water surface so that the observer sights downstream with the clinometer to record the **percent** slope. In the same positions, the observer

will sight back with the compass and record the bearing (degrees) to the other person. Continue upstream and record measurements at each transect interval.

13.4.2 Substrate Size and Channel Dimensions

At the 11 transects, as well as the midpoint intervals between transects (for a total of 21 locations), there will be 5 pebbles selected from each wetted edge and at 25%, 50%, and 75% of the wetted width (see Figure 12.4). This yields a 105 particle pebble count (21 locations 5 particles) from the entire length of the reach. To minimize bias, place the survey rod at the correct measurement on the transect tape and select the particle directly at the bottom of the stick. EMAP protocols for this procedure call for depth and distances to be measured, but the particle sizes and embeddedness are visual estimates. **For SWQB purposes, particle sizes will be measured and individually recorded as F1 comments on the Channel/Riparian Cross Section and Thalweg Profile & Woody Debris forms.** Embeddedness percentages will be recorded as well. Sand and fines are recorded as 100% embedded by definition. In addition to this method, a modified Wolman Pebble count will be conducted in the designated riffle where periphyton was collected and recorded on a riffle pebble count field form. See the Additional Data Collection section for specific details of this additional pebble count.

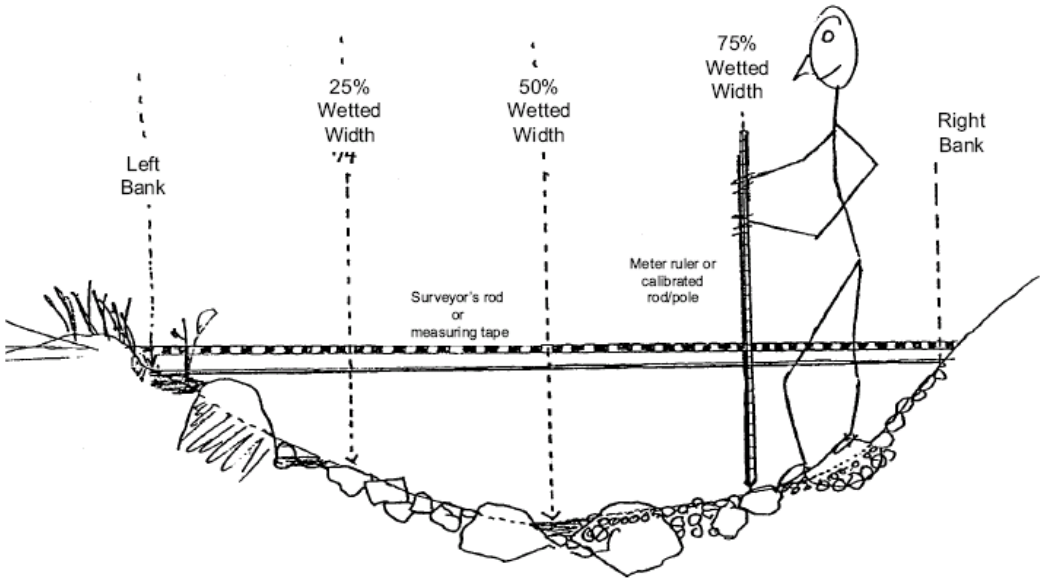



**Figure 13-4 Measuring pebbles**

13.4.3 Bank Characteristics

Bankfull or active channel is defined as that which is filled with moderate sized flood events that would typically occur every one or two years and do not usually inundate the floodplain. Bankfull levels can be identified by:

- 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 banks,
- Presence of drift material caught on overhanging vegetation
- Transition form flood- and scour-tolerant vegetation to that which is relatively intolerant.

To measure **bank angle**, lay the survey rod against the bank with one end at the water's edge, and lay the clinometer on top of the rod. Record the degrees of the angle. Measure both banks. If the bank is undercut, measure the **distance of undercutting** to the nearest 0.01m. Undercut distance is measured from the waters edge to the point where a plumb line from the bank would hit the water's surface.

A vertical bank is 90 degrees; undercut banks have angles >90 degrees approaching 180 degrees, and more gradually sloped banks have angles <90 degrees. To measure bank angles >90 degrees, turn the clinometer (which only reads 0 to 90 degrees) over and subtract the angle reading from 180 degrees.

Measure **incised height** by having one person holding the survey rod at the wetted edge and another person on top of the incision with a meter stick or ski pole with one meter marked on it. Then, holding the clinometer at the one meter mark on the ski pole, that person looks through the clinometer holding it at zero and reads the height on the survey rod at the wetted edge. Then subtract one meter from that measurement for incised height. This measurement is the distance from the water's surface to the elevation of the first terrace of the valley floodplain (above bankfull). This terrace height is only one measurement, therefore, if the distance is different on each bank, choose the lower one and flag the data. In the same position, visually estimate the distance from the water's surface to the bankfull height as described above. **Bankfull height is never greater than incision height.** At each transect also record **wetted width, bankfull width, and width of exposed mid-channel bars** (if present).

Remember that incision height is measured as the vertical distance to the first terrace above bankfull; 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, and the slope of the channel banks simply extends uphill indefinitely, not reaching a terrace before reaching the top of a ridge (Figure 12.5). In such cases, record incision height values equal to bankfull values and make appropriate comment that no terrace is evident.

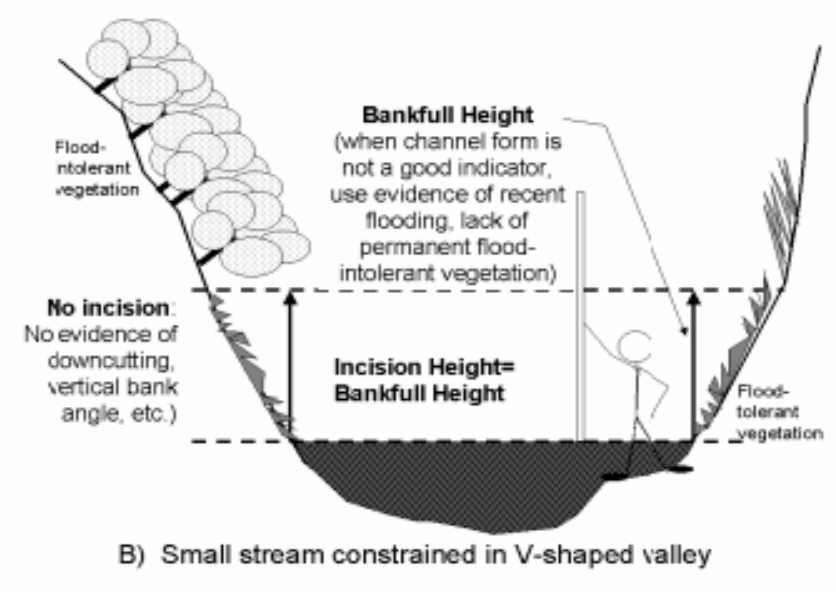



**Figure 13-5 V-shaped valley measurements**

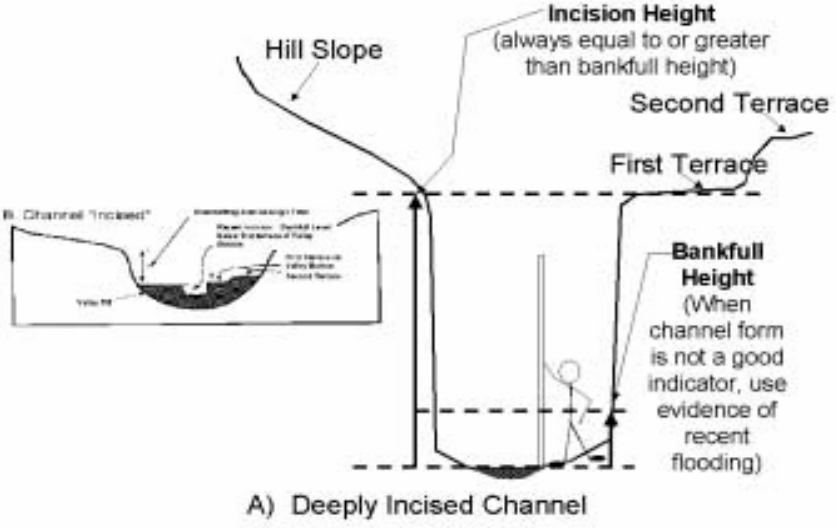



**Figure 13-6 Deeply incised channel**

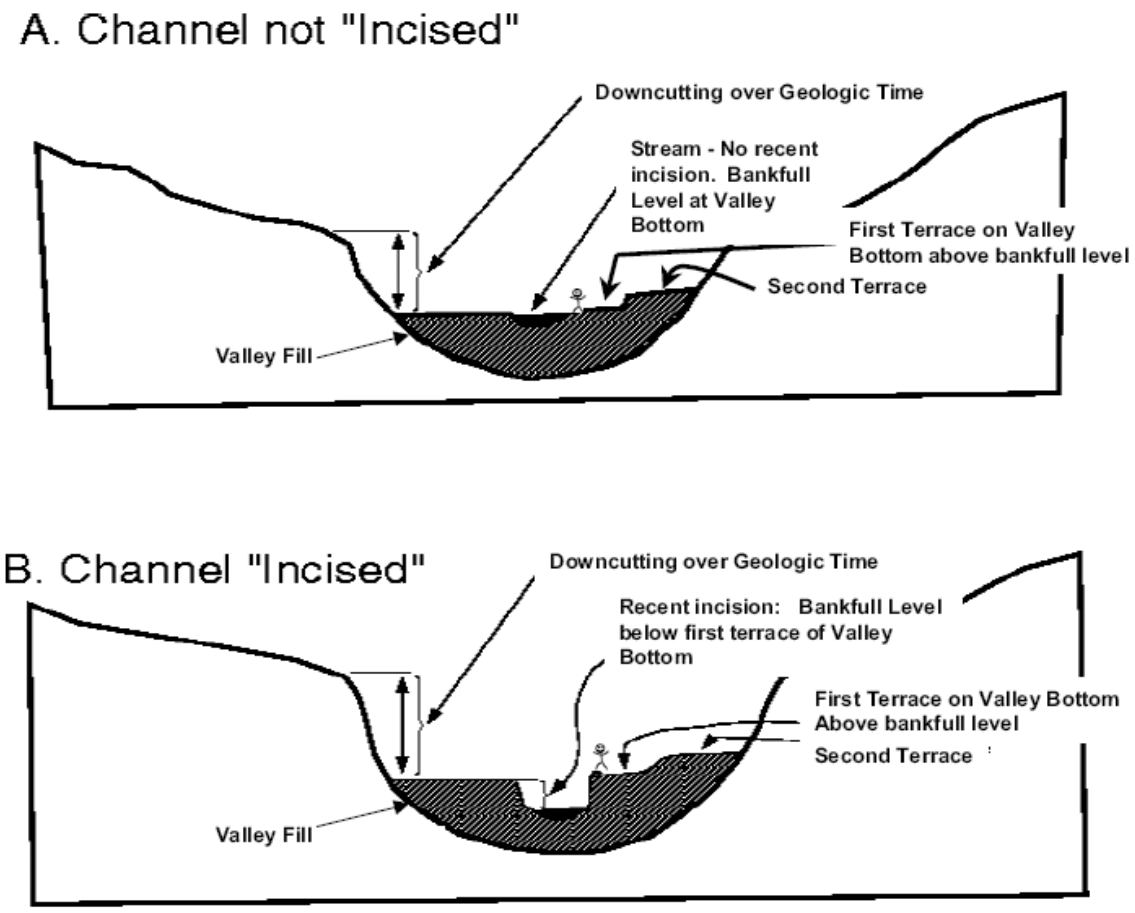



**Figure 13-7 Channel geomorphic descriptions**

13.4.4 Canopy Cover Measurements

Canopy cover is significant in controlling temperature as well as an indicator for bank stability and organic material input potential. 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 four directions at mid-channel and one at each bank). Densiometer measurements are taken at 0.3 m (1 ft) above the water surface, rather than at waist level, to (1) avoid errors because people differ in height; (2) avoid errors from standing in water of varying depths; and (3) include low overhanging vegetation more consistently in estimates of cover. Hold the densiometer level (using the bubble level) 1 foot above the water surface with your face reflected just below the apex of the taped "V." Count the number of grid intersection points within the "V" that are covered by any object that provides shade (vegetation, bank, etc.). Record the value (0 to 17) in the canopy cover measurement section of the Channel/Riparian Cross-section and Thalweg Profile Form.

### 13.4.5 Riparian Vegetation Structure

The following visual estimation procedures supplement canopy cover measurements with a semi-quantitative evaluation of the various types and amounts of riparian vegetation. These data are used to evaluate the health and level of disturbance of the stream corridor. They also provide an indication of the present and future potential for various types of organic inputs and shading.

Riparian vegetation observations apply to the riparian area 5 meters upstream, 5 meters downstream, and 10 meters into the visible floodplain from water's edge at each of the 11 cross-section transects (see Figure 12.8). Within this 10 m × 10 m area, conceptually divide the riparian vegetation into three layers: a **canopy layer** (> 5 m high), an **understory** (0.5 to 5 m high), and a **ground cover** layer (< 0.5 m high).

Determine the dominant vegetation type for the canopy layer (vegetation > 5 m high) as [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. Determine separately the areal cover class of large trees (> 0.3 m [1 ft] diameter at breast height [DBH]; DBH is defined as 4.5 ft [1.37 m] above the forest floor on the uphill side of the tree) and small trees (< 0.3 m DBH) within the canopy layer. Estimate areal cover as the amount of 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%).

Look at the understory layer (vegetation between 0.5 and 5 m high). Determine the dominant **woody** vegetation type for the understory layer. If there is no woody vegetation in the understory layer, record the type as "None". Determine the areal cover class for woody shrubs and saplings separately from non-woody vegetation within the under-story.

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. Repeat the above procedures for the right bank. Repeat the entire process for all cross-section transects, using a separate field data form for each transect.

### 13.4.6 Instream Fish Cover, Algae, and Aquatic Macrophytes

This portion of the EMAP physical habitat protocol is a visual estimation procedure that semi-quantitatively evaluates the type and amount of important types of cover for fish and macroinvertebrates. 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. Examine the water and banks within the 10 m segment 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. 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%). Repeat at each cross-section transect, recording data from each transect on a separate field data form.

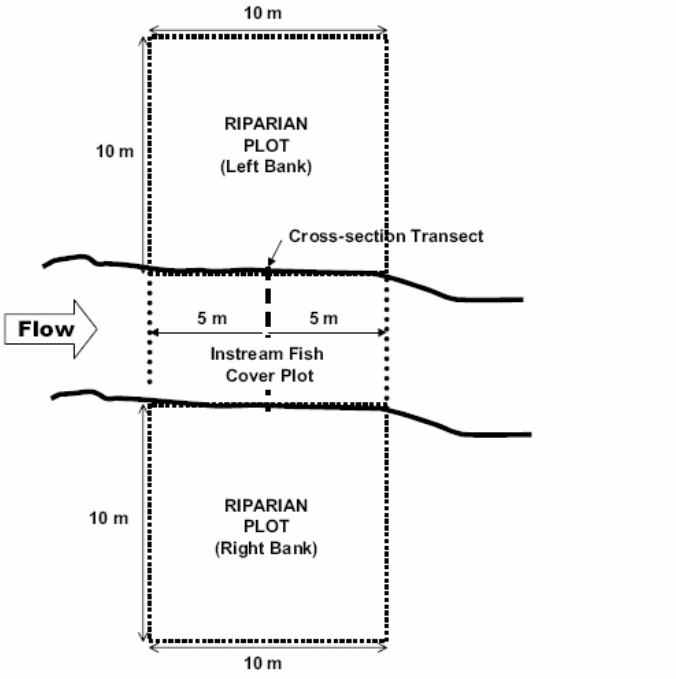



**Figure 13-8 Visual estimation boundaries**

The entry "Filamentous Algae" refers to long streaming algae that often occur in slow moving waters. "Macrophytes" are water-loving plants, including mosses, in the stream that could provide cover for fish or macroinvertebrates. If the stream channel contains live wetland grasses, include these as macrophytes. "Woody debris" refers to 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 [see Section 12.3.2]). "Brush/Woody Debris" refers to smaller wood pieces that primarily 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. For ephemeral channels, estimate the proportional cover of these trees that would be inundated during bankfull flows. "Overhanging Vegetation" includes tree branches, brush, twigs, or other small debris that is not in the water but is close to the stream (within 1 m of the surface) and provides potential cover. If "Undercut Banks" are present, they should be quantified as a proportion of the entire bank length (e. g., if 4 m are undercut out of a total of 20 m of bank, it should be considered 20%). "Boulders" are typically basketball- to car-sized particles. "Artificial Structures" include those designed for fish habitat enhancement, as well as in-channel structures discarded (e.g., concrete, asphalt, cars, or tires) or intentionally placed for diversion, impoundment, channel stabilization, or other purposes.

### 13.4.7 Human Influence

Human land use activities in the stream riparian area are evaluated in combination with mapped watershed land use information to assess the potential degree of disturbance of the sample stream reaches.

Examine 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 lot (e.g., paved, graveled, dirt parking lot, foundation); (4) roads or 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). For each type of influence, determine if it is present and what its proximity is to the stream and riparian plot area. Consider human disturbance items as present if you can see them from the cross-section transect. Do not include them if you have to sight through another transect or its 10 m × 10 m riparian plot. 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 × 10 m riparian plot area, but away from the bank.
- **P** ("Present") Present, but outside the riparian plot area.
- **0** ("Not Present") Not present within or adjacent to the 10 m stream segment or the riparian plot area at the transect.

Repeat the above procedure for the right bank. Repeat the entire process for each cross-section transect, recording data for each transect on a separate field form.

### 13.4.8 Riparian "Legacy" Trees and Invasive Alien Plants

**Riparian Legacy Tree:**

The Riparian "Legacy" Tree protocol contributes to the assessment of "old growth" characteristics of riparian vegetation, and aids the determination of possible historic conditions and the potential for riparian tree growth.

Beginning at Transect A, look upstream. Search both sides of the stream upstream to the next transect. At Transect K, look upstream for a distance of 4 channel widths. Locate the largest tree visible within 50 m (or as far as you can see, if less) from the wetted bank. Classify this tree as deciduous, coniferous, or broadleaf evergreen. Identify, if possible, the species or the taxonomic group of this tree from the list below.

| | |
|---|---|
| 1. Acacia/Mesquite | 11. Snag (Dead Tree of Any Species) |
| 2. Alder/Birch | 12. Spruce |
| 3. Ash | 13. Sycamore |
| 4. Cedar/Cypress/Sequoia | 14. Willow |
| 5. Fir (incl. Doug. Fir, Hemlock) | 15. Unknown/Other Broadleaf Evergreen |

6. Juniper
7. Maple/Boxelder
8. Oak
9. Pine
10. Poplar/Cottonwood
16. Unknown or Other Conifer
17. Unknown or Other Deciduous

**NOTE:** If the largest standing tree is dead, enter "Snag" as the taxonomic group.
Estimate the height of the potential legacy tree, its DBH, and its distance from the wetted margin of the stream. Enter this information in the appropriate column of the Riparian "Legacy" Trees and Invasive Alien Plants field form.

**Alien Invasive Plants:**
A trend of increasing concern along streams in many parts of the Western U.S. is the invasion of alien (non-native) tree, shrub, and grass species. At each transect, the presence of invasive plant species within the 10 m × 10 m riparian plots on either bank is recorded on the Riparian "Legacy" Trees and Invasive Alien Plants field form. Record the presence of any known alien species (common examples include salt cedar, Russian olive, and cheat grass) within the plot on either the left or right bank by marking the appropriate box(es) on the right hand column of the Riparian "Legacy" Trees and Invasive Alien Plants field form. If no known alien species is present in either of the plots at a given transect, check the box labeled "None" for that transect.

Repeat the procedure for each remaining transect (B through K). At transect K, look upstream a distance of 4 channel widths when locating the legacy tree.

## 13.5 Channel Constraint, Debris Torrents, and Recent Flood

### 13.5.1 Channel Constraint

The degree of channel constraint can strongly influence the quantity and quality of habitat for aquatic organisms. Constraint also influences the type and degree of stream channel adjustment to anthropogenic alterations in flow and sediment supply, or to direct channel manipulations (e.g., dredging, revetment, impoundment). To assess overall reach channel constraint, SWQB modified methods used by the Oregon Department of Fish and Wildlife in their Aquatic Inventories (Moore et al., 1993).

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

**Anastomosing channels** have relatively long major and minor channels branching and rejoining in a complex network.

**Braided channels** also have multiple branching and rejoining channels, but these sub-channels are generally smaller, shorter, and more numerous, often with no obvious dominant channel.

After classifying 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, hill slopes, terraces/alluvial fans, and human land use (e.g., road, dike, landfill, rip-rap, etc.). Estimate the percent of the channel margin in contact with constraining features (for unconstrained channels, this is 0%). Record this value on the Channel Constraint Form. Finally, estimate the "typical" bankfull channel width, and visually estimate the average width of the valley floor. Record these values on the Channel Constraint Form. As an added data point, measure the flood prone width whenever possible.

### 13.5.2 Debris Torrents and Recent Floods

Major floods are those that substantially overtop the banks of streams and occur with an average frequency of less than once every 5 years. Major floods may scour away or damage riparian vegetation on banks and gravel bars that are not frequently inundated. They typically cause movement of large woody debris, transport of bedload sediment, and changes in the streambed and banks through scouring and deposition. While they may kill aquatic organisms and temporarily suppress their populations, floods are an important natural resetting mechanism that maintains habitat volume, clean substrates, and riparian productivity.

In arid areas with high runoff potential, debris torrents can occur in conjunction with flash flooding from extremely high intensity rainfall. They may be nearly annual events in some steep ephemeral channels where drainage area is sufficient to guarantee isolated thunderstorms somewhere within their boundaries, but small enough that the effect of such storms is not dampened out by the portion of the watershed not receiving rainfall during a given storm. Because they may alter habitat and biota substantially, infrequent major floods and torrents can confuse the interpretation of measurements of stream biota and habitat in regional surveys and monitoring programs. Therefore, it is important to determine if a debris torrent or major flood has occurred within the recent past. After completing the Thalweg Profile and Channel/Riparian measurements and observations, examine the stream channel along the entire sample reach, including its substrate, banks, and riparian corridor, checking the presence of features described on the Torrent Evidence Assessment Form.

## 13.6 Additional Habitat Data Collected outside of EMAP Procedures

### 13.6.1 Riffle Pebble Count Procedure

The Wolman pebble count procedure is modified to perform the Surface Water Bureau's *Protocol for the Assessment of Stream Bottom Deposits on Wadeable Streams* (2004). This protocol is specifically designed to assess whether streams are biologically impaired due to excess sediment or stream bottom deposits that may "settle and damage or impair the normal growth, function, or reproduction of aquatic life or significantly alter the physical or chemical properties of the bottom" (NMWQCC, 2002). Pebble counts for this protocol need to be performed with and at the same time as benthic macroinvertebrate collections. In addition, the counts are limited to only the wetted width of the same riffle habitat units that the macroinvertebrates are collected. However, if the stream is small (i.e. less than 1 meter width) 5 particles from each of 20 transect is acceptable. Likewise, if the riffle is too short to fit ten transects another representative riffle may be sampled as well. The above-mentioned protocol

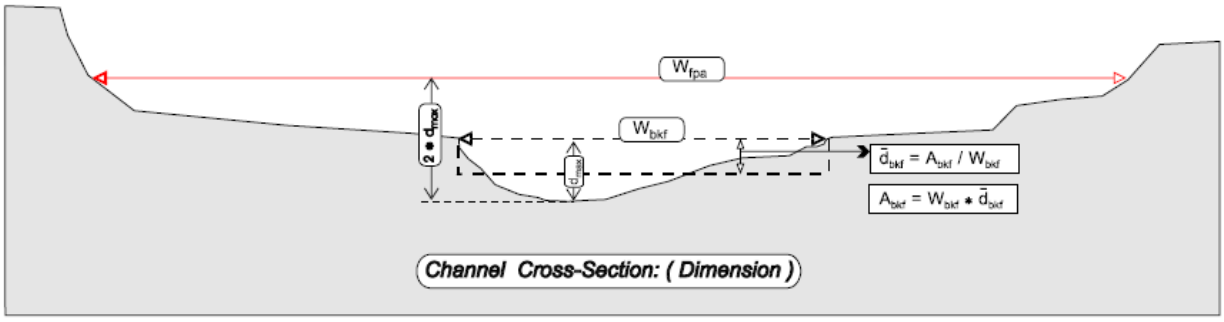
for stream bottom deposits needs to be consulted and read carefully when doing this assessment due to the modifications and details, which are beyond the scope of this document.

The observer should start each transect (start either at the top or bottom of the reach) at a random point near the wetted edge. Averting gaze, pick up the first particle touched by the tip of the index finger at the toe of the left (or right) wader and measure the intermediate axis with a metric ruler or classify (silt/clay and sand) it using a sand-gauge card. Immovable embedded substrate or substrate to large to lift should be measured in place (measure the smaller of the two exposed axis). The measurement should be called out and the note taker should tally it to the proper size classification and repeat the measurement back to the observer for confirmation. The note taker may also keep count of the particles measured in each transect and the reach by using a mechanical tally counter. After completing the first measurement, the observer should step along the transect toward the opposite bank adjusting the length of the stride into equal distances to insure at least 10 measurement points before wetted edge on the opposite side. Each measurement should be made as previously described; picking up and measuring the first particle touched by the index finger at the tip of the wader without gazing at this point. After inventorying at least 10 particles along the transect line, the observer should move to the next transect and repeat the process until all 10 transects have been competed and the requisite 100 particles (or more) have been measured. The raw data from the field sheet may then be processed using the Mecklenburg *Reference Reach (1999)* spreadsheet and software for channel materials. The software will not only calculate percentages of particle size categories and D values but will graphically display them.

### 13.6.2 Measurement of Floodprone width

Floodprone width will be measured at each site according to Rosgen (1996) so that a Rosgen Level 2 Classification may be completed to compare study and reference sites, and for monitoring of restoration projects. The flood prone width is one aspect of channel structure not measured in EMAP protocols. It describes the vertical containment of the stream and provides information as to how the stream can dissipate energy at high flows. Bankfull depth in EMAP protocol is measured from the water's surface. This depth should be added to the thalweg depth at transect A (or the most representative transect) to give a maximum depth. This maximum depth is multiplied by two, and the width at that elevation is the floodprone width. The floodprone width should be recorded in the comments section of the Channel Constraint and Field Chemistry Form. The figure below was taken from Rosgen's field survey procedures, found online http://www.wildlandhydrology.com/assets/Field_Survey_Procedures_for_Characterization_of_River_Morph.pdf.

*Figure 2. Channel Cross Section*




The red line indicates the floodprone width and the upper dashed line is bankful width.

**Figure 13-9 Rosgen's illustration of floodprone width measurement**

**FORMS**

EMAP Altercation Form
EMAP Sample Collection Form - Streams
Stream Verification
Stream Assessment
Field Data Sheet
Rapid Geomorphic Assessment
Rapid Habitat Assessment forms
Thalweg Profile and Woody Debris
Channel/Riparian Cross-section
Riparian "Legacy" Tree and Invasive "Alien" Plant
Slope and Bearing
Channel Constraint and Field Chemistry
Torrent Evidence Assessment
Stream Discharge Measurement
Riffle Pebble Count

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# 14.0 LAKES

Lentic waterbodies in New Mexico have historically been, and continue to be, studied following methods and approaches specified in *Clean Lakes Program Guidance Manual* (USEPA 1980). For purposes of consistency and comparability, classic limnological methods for lake trophic state classification and water quality standards attainment continue to be used in monitoring practices. The following will describe accepted approaches used in studies of lentic systems of the State. For purposes of this document, the term "lake" shall include natural lakes, as well as reservoirs, impoundments, and any other human-made lentic waters.

## 14.1 Lake Study Design

Lakes chosen for study are associated with intensive, rotating watershed studies scheduled for that year.

Typical lake water quality surveys consist of two sampling stations for large lakes (> 100 acres). One station will represent the deepest portion of the lake, which is typically near the dam. The second station will be located in a relatively shallow area, frequently two-thirds to three-quarters the length of the lake from the dam station. In small lakes (< 100 acres), one sampling station is established at the deepest point within the lake. Playa lakes are typically sampled from one station at the deepest point. During any intensive survey, additional stations may be established to evaluate conditions of concern.

Large lakes are monitored during three seasons, excluding winter, to better characterize seasonal variations in water quality. Small lakes are sampled, at the very least, during two seasons, which must include summer because summer coincides with the longest day-length, warmest temperatures, heaviest macrophyte and algal growth, and greatest recreational use.

Due to staffing and budgetary resource limitations, it is often necessary to prioritize lakes located within the respective watershed to ensure that the most significant waterbodies are studied. Factors that contribute to the selection process are size, Clean Water Act Section 303(d) listing status, designated or existing uses, and time since the lake was last studied. Large lakes generally attract more visitors than small lakes. In addition, large lakes commonly have multiple designated uses such as irrigation storage, municipal water supply, primary or secondary contact, wildlife habitat, and several aquatic life use designations. Consequently, large lakes are typically higher priority than small lakes.

## 14.2 Lake Sampling Typically Performed by SWQB

A Cubitainer® is a 1-liter polyethylene sampling container used to collect water samples from surface water. A Cubitainer® sampler is used for most routine sampling in lakes. Cubitainers® are never re-used. The following samples are collected in Cubitainers®:

- **Major cations and anions**
- **Total nutrients**: TP, NO2 + NO3, NH3 + NH4, and TKN.
- **Dissolved nutrients**: Ortho P, ammonia, nitrite + nitrate as N, dissolved P.

- **Total metals**: (most important to SWQB is Se and Hg).
- **Dissolved metals**: (please refer to the *State of New Mexico Standards for Interstate and Intrastate Surface Waters* (NMAC 2006) for list).
- **Cyanide**
- **Phytoplankton community composition**

The following samples are collected in other containers:

- **Organics** (usually base neutral extractable scan or others as conditions warrant) are collected in specialized glass containers (specified by SLD for each type of analysis); the sampling containers are *not* rinsed with native water prior to being filled.
- **Volatile Organic Compounds** are collected in two 40-mL VOA vials to overflowing and capped without trapping any air bubbles; the sampling containers are *not* rinsed with native water prior to being filled. If benzene, toluene, ethylbenzene, or xylene (BTEX) is requested, 2-4 drops of HCl are added to the sample to eliminate microbial degradation. See Section 7.0 regarding the VOA trip blank.
- **Radionuclides** (Gross alpha, beta, Ra-226 and Ra-228, and others as conditions warrant) are collected in two one-gallon Cubitainers®.
- **Diatom** samples are collected in clean, unused glass or plastic vials approximately 100 mL in volume.
- **E. coli** samples are collected in 125-mL sterilized bottles and placed on ice for transport and analyses within six hours of the sample collection; the sampling containers are *not* rinsed with native water prior to being filled.
- Filters containing **chlorophyll** samples are collected in 60-mL light-proof septum vials.

## 14.3 Equipment

Equipment and supplies use for lake survey trips are listed in the Lakes Checklist in Table 14-1.

## Table 14-1 Lake/playa sampling equipment checklist

**SAMPLE COLLECTION**

- ☐ Water carboy w/ deionized water
- ☐ Nitric acid & tips
- ☐ Sulfuric acid & tips
- ☐ Pipetter(s)
- ☐ Rubber gloves
- ☐ Safety glasses
- ☐ Cubitainers
- ☐ Septum vials for organics (8260)
- ☐ Amber liter bottles (8270)
- ☐ Bacti bottles
- ☐ Geofilter case
- ☐ Clean teflon tubing
- ☐ Inline filters
- ☐ Clipboard
- ☐ Lab forms (nut,met,sed,pest,rad,Cn)
- ☐ Washing bottle(s) w/ DI
- ☐ Marking pens
- ☐ Specimen/diatom vials
- ☐ **IDEXX** Equipment: Bottles, Sealer, Inoculants, Incubator, Black Light, AC and DC power cords.

**EQUIPMENT FOR FIELD MEASUREMENTS**

- ☐ YSI datalogger and sonde
- ☐ Lake line(s) for sonde (250', 25').....
- ☐ Standards for pH, Conductivity, Turb
- ☐ pH buffers (4,7, & 10)
- ☐ Turbidity meter (Hach) w/ standards
- ☐ Graduated cylinder
- ☐ Magnetic stirrer (AC or battery)
- ☐

**GENERAL**

- ☐ Coolers ( )
- ☐ Ice
- ☐ Tool box (LAKE)
- ☐ Kimwipes
- ☐ Extra batteries
- ☐ Topo maps
- ☐ Tape and rubber bands
- ☐ First aid kit
- ☐ Flashlight
- ☐ Backpack

**BENTHOS**

- ☐ D-net and/or kick screen
- ☐ Screen bucket
- ☐ Whirl Paks
- ☐ Alcohol
- ☐ Paper & pencil
- ☐ Hip waders; ☐Chest waders
- ☐ Forceps
- ☐ Shovel
- ☐ Buckets
- ☐ Porcelain pan
- ☐

**LAKE SURVEYS**

- ☐ Lugols Solution
- ☐ Kemmerer bottle
- ☐ Eckmann Dredge
- ☐ Glass-fiber filters
- ☐ Filter flask/funnel/hand pump/ filter holder/clamp (whew!)
- ☐ Tubes with acetone[1]
- ☐ Aluminum foil
- ☐ Secchi Disk
- ☐ Forel-Ule color comparator
- ☐ Underwater photometer
- ☐ Empty lake water carboy

**BOAT EQUIPMENT**

- ☐ Gas can / 2-cycle oil
- ☐ PFD's; Life jackets ( )
- ☐ Anchor(s) (Lg &/or Sm)
- ☐ Throwable flotation device
- ☐ Oars
- ☐ Depth Finder;(Lorance, Hummingbird, secchi)
- ☐ Spare trailer tire
- ☐ Boat keys
- ☐ 12-volt battery (2-3)
- ☐ Battery charger
- ☐ Electric motor
- ☐ 5 hp gas motor with gas can
- ☐ Raft ☐ Foot Pump ☐ Paddles
- ☐ Opt. Raft floor boards, seats

- ☐ Raincoats
- ☐ Field Guides (Mammals,birds,tracks)
- ☐ Binoculars
- ☐ **Camera and Cell Phone**
- ☐ Opt. Raft motor mount, repair kit
- ☐
- ☐

[1] 35 ml 90% acetone; 4 drops $MgCO_3$

## 14.4 Collecting Physical Data

At each station, a field-calibrated multi-parametric sonde and data logger are used to measure dissolved oxygen concentration, specific conductance, temperature, and pH at one-meter intervals. These measurements are recorded on the Lake and Reservoir Field Sheet located in Appendix A. Measurements are taken at the surface to within one meter of the bottom of the lake. Water color is determined at the lake surface using a Forel-Ule Color scale. Light attenuation is also read at one-meter intervals using an underwater photometer to the depth at which 1% of surface illumination remains. A Light Extinction Record Form, used to record these measurements can be located in Appendix A of this document. In those situations where lake size or access dictates, the empirical rule of 3 times the secchi depth is used to estimate the euphotic zone (Cole, 1983). The vertical region from the surface to this depth is defined as the euphotic zone.

## 14.5 Collecting Chemical Data

### 14.5.1 Field Rinsing of Equipment

Most equipment used for sample collection and processing is field rinsed with the water to be sampled (including DI water for blank samples) just before water samples are collected (see Section 13.2 for exceptions). Field rinsing procedures are summarized below for sampling devices or for sample compositing and sample splitting equipment.

To field-rinse a Cubitainer® or other sampler:

A. Partially fill the sampler with the water to be sampled, either lake water for water quality samples or DI water for blank samples.

B. Shake the sampler, ensuring that the rinse water thoroughly rinses all interior surfaces. Drain the rinse water from the sampler. This procedure is then repeated for a total of two rinses.

Samples are filtered for dissolved concentrations or not filtered for total concentrations. All samples for dissolved constituent analyses are filtered using a 0.45-micron pore-size disposable in-line filter cartridge (Geotech dispos-a-filter®). The filters and Cubitainers® used for these analyses should also be rinsed with the lake water to be sampled. To do this:

A. Attach an unused filter to a clean, acid-rinsed section of tubing.

B. Thread the tubing (with filter attached) through the geo-pump making sure the direction of flow is correct.
C. Place the end of the tube into the container with the sample water.
D. Turn the geo-pump ON to run the sample water through the tubing and filter. Turn the geo-pump OFF after the tubing and filter have been rinsed with sample water for a few seconds.
E. Place a Cubitainer® underneath the filter to capture the filtered water. Turn the geo-pump ON again for a few seconds. Turn OFF. Cap the Cubitainer® and thoroughly rinse the inside of the container with the filtered lake water. Dispose of the rinse water.
F. Repeat Step E for a total of two rinses.

Filters should only be used once in order to prevent potential contamination between samples. The only exception is when filtering blank water immediately before filtering an environmental sample. After filtering is complete, filters are disposed of but tubing is saved. The tubing is acid-rinsed in SWQB's laboratory and reused.

14.5.2 Collecting a Lake Water Sample

Five-liter samples from the top, middle, and bottom of the euphotic zone are collected for the water quality analyses listed in Section 13.2 using a rinsed Kemmerer® water bottle. The 5-liter samples are composited in a 5-gallon, acid-washed container.

When a thermocline is present (i.e., there is greater than 1°C difference between the surface and bottom of the lake), 5-liter samples are taken from the top, middle, and bottom of the entire water column instead of just the euphotic zone (Cole 1994). The 5-liter samples are composited in a 5-gallon, acid-washed container.

All water quality samples are poured off from the composited sample into their respective containers; either individually rinsed, 1-liter Cubitainers® or other containers as prescribed by the analysis to be performed. Dissolved nutrient and metal samples are filtered prior to being poured into individually rinsed, 1-liter Cubitainers®. After the composited sample is divided into individual containers, the samples for total and dissolved nutrients are preserved in the field with two milliliters of concentrated sulfuric acid per liter of sample water (to reduce pH to < 2). Samples for total and dissolved metal analyses are preserved with five milliliters of concentrated nitric acid per liter of sample water (to reduce pH to < 2). Samples for cyanide are preserved with 8-10 tablets of NaOH per liter of sample water (to increase pH > 12). Phytoplankton community composition samples are preserved with 10-25 mL of acid Lugol's solution per liter of sample water (after addition of the Lugol's solution, the sample should turn an iced tea color). Radionuclides samples are preserved with 20 mL of nitric acid per gallon of sample water (to reduce pH to < 2). (see Sections 7.4.2.6 and 7.4.2.7)

Unless specific concerns exist, samples for cyanide, organics, and radionuclides are initially collected during the summer season when irrigation and field application of herbicides and pesticides are most likely to occur. Subsequent sampling for these

parameters is conducted at sites where concerns are identified during initial sample review.

During every seasonal sampling run, samples for bacteriological analysis of *E. coli* are taken at each station. Bacteriological samples are collected in sterilized 125-mL bottles.

All samples are transported on ice and held at 4°C until analyzed.

## 14.6 Analyzing Water Samples

Sample types analyzed at SLD may include ions, nutrients, metals, organics, and radiological analyses, depending on concerns detailed in the study design. As required, certain uncommon analyses may be assigned to an appropriate outside contract laboratory. *E. coli* analyses are typically performed at the SWQB laboratory using the IDEXX Colilert® Quanti Tray method (IDEXX, USA).

SWQB staff analyze chlorophyll samples for chlorophyll *a*, *b*, *c*, and phaeophytin concentrations within 24 to 48 hours of collection using the trichromatic method described in USEPA's *Biological Field and Laboratory Methods* (1973) or an approved edition of Standard Methods for the Examination of Water and Wastewater, American Public Health Association (APHA 1998). For analyses performed by SLD, SLD transfers the results to SWQB staff who upload the data into the SWQB in-house database for storage and eventual upload to the USEPA Storage and Retrieval database (STORET).

## 14.7 Collecting and Processing Biological Samples

Macrophyte growth is a potential symptom of nutrient enrichment to determine whether recreational uses may be impaired by excessive growth. Staff should visually estimate percent macrophyte coverage in the lake and record the estimate on the bottom of the field notes.

Phytoplankton species and periphytic diatom communities are sampled, preserved, identified, and enumerated to assess community composition and dominant species, which may indicate undesirable symptoms of eutrophication (Likens 1975). Phytoplankton samples consist of a composited euphotic zone sample placed in a one-liter Cubitainer® and preserved with acid Lugol's solution, as explained in Section 13.5.2. Samples are concentrated according to the settling method and counted by the Sedgwick-Rafter method (APHA 1998). Laboratory identification and enumeration is accomplished using appropriate taxonomic keys. Results include major taxonomic units, a diversity index (Shannon and Weaver 1949), and community density (cells/mL). Results will be housed in the EDAS database.

Diatom samples from sediments are collected by Ekmann dredge or epiphytic scrapes collected from multiple substrates (e.g., aquatic plants, rocks, submerged wood, and materials on sandy or gravelly sediments). Diatom samples are typically processed by a contractor. If processed by SWQB staff, the samples are oxidized using the cold cook-down method (30% hydrogen peroxide with potassium dichromate) and cleaned using a

multiple rinse with distilled water (Van der Werff 1955). A minimum of four hours between each decant precedes the final concentration and storage of the materials in a clean, labeled vial. Permanent microscopic slides are prepared and mounted in Hyrax or equivalent mounting media with a refractive index of 4 or greater. Microscopic examination at 1000× of random paths across the specimen slide are conducted until a minimum of 200 frustules have been identified. The resulting community composition and diversity calculations are used to help determine overall water quality conditions and trophic state classification for lakes and reservoirs (see section 13.5). Analyses of diatom community composition performed by SWQB staff or an independent contractor will be performed using appropriate taxonomic keys (Cleve-Euler 1951-1955, Dodd 1987, Germaine 1981, Hustedt 1956, Patrick and Reimer 1966, Van der Werff 1963). Results will be housed in the EDAS database.

## 14.8 Chlorophyll Analysis, Trophic State, and Nutrient Limitation

For chlorophyll analysis, a volume of composited euphotic-zone water is filtered through Whatman Glass Fiber, Grade A (GF/A) filters until substantial color development is present on the filter medium, indicating adequate chlorophyll pigment is present. This is generally accomplished by filtering two liters of composited euphotic-zone water, but sometimes requires more. Once filtered, the glass fiber filters are placed in 60-mL, light-proof septum vials. The filters are covered with 35 mL of 90% acetone mixed with four drops of saturated aqueous solution of magnesium carbonate and stored on ice. The samples are analyzed for Chl *a*, *b*, and *c* and phaeophytin concentrations within 24 to 48 hours of collection using the trichromatic method described in *Standard Methods for the Examination of Water and Wastewater, 20th Edition* (APHA 1998). Results from the spectrophotometric analyses are recorded on the Chlorophyll and Pheophytin Analysis Form located in Appendix A.

Trophic states are evaluated using Carlson trophic state indices (TSIs) and lakes are categorized using a continuum ranging from oligotrophic to hypereutrophic (Carlson 1977). The univariable Carlson index is used to assess trophic state based on Secchi-disk depth, Chl *a*, total phosphorus, and total nitrogen concentrations. This is an absolute index whereby a ten-unit increase on a scale of 0 to 100 corresponds to a doubling in epilimnetic algal biomass. Thus, small differences in data values result in a larger change in TSI for eutrophic lakes (Carlson 1997). Each of the Carlson TSI values for a given lake is separately evaluated with preferential consideration given to TSI (Chl). Trophic-state boundaries are consistent with the USEPA index; trophic-state values exceeding 47 indicate a eutrophic lake and values less than 42 indicate an oligotrophic lake (USEPA 1974, Maloney 1979). The TSI values for secchi depth, chlorophyll *a*, and total phosphorus are calculated by year and seasons for each lake station. These values are averaged to provide the corresponding overall trophic state of the lake.

A similarly derived output is generated for nitrogen and/or phosphorus nutrient limitation from each lake station during each sampling visit. Total nitrogen must be calculated because SLD does not report it. Total nitrogen is defined as the sum of Nitrate+Nitrite ($NO_3$+$NO_2$), and Total Kjeldahl Nitrogen (TKN). The limiting nutrient is determined

using the ratio of total nitrogen to total phosphorus. Boundaries are defined by Forsburg (1980) as > 17 P-limiting; 10-17 N and/or P limiting; and < 10 N-limiting.

## FORMS

Lake and Reservoir Field Sheet
Light Extinction Record Form
Chlorophyll and Pheophytin Analysis Form

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## 15.0 WETLANDS

At this time the SOP for wetlands monitoring is in progress. Please refer to the QAPP for existing wetlands information.

# 16.0 PHOTOGRAPHIC DOCUMENTATION

Photographic documentation is an important component of the overall documentation of field conditions. It can be particularly useful for documenting and/or supplementing descriptions of physical habitat conditions, riparian structure, erosional and depositional features, and other aspects of water quality that are not effectively documented with numeric or verbal descriptions. A camera should be a standard piece of equipment in every field kit and should be carried on every field excursion. It should be used extensively, as any images that are later deemed unnecessary may be easily deleted.

Every station visited should be photographically documented at various flow stages throughout the sampling season. Other pertinent features that change through time should also be photo documented. Photos at each station should be taken from a consistent location to the extent possible. All photos should include an object of known size (e.g., person, ruler, field binder, or equipment) for scale. At the beginning of each field excursion, the date/time electronic stamp should be checked for accuracy and adjusted as necessary. A photo log with pertinent information about each image should be maintained for transcription to the permanent record upon return from the field. Do not depend on memory for this sort of information. It is also helpful to consistently take photos in the following order:

1. Station ID – The Station ID (and name, if desired) should be written on a blank sheet of paper and photographed close-up so that it is readable. An easy way to accomplish this is to use the macro function of the camera and photograph a field form, capturing the station name and ID. Remember to disable the macro function for subsequent photos.
2. Upstream
3. Downstream
4. Left bank
5. Right bank
6. Additional photos to document noteworthy conditions or features (e.g., evidence of recent high flow, habitat disturbances, recent fires, anthropogenic activities, substrate, abundant algal or macrophyte growth, etc).

For habitat and biological monitoring, the above photos should be taken first, followed by those listed below taken from representative areas:

7. Eroded banks – wide angle and close up to show bank material
8. Incisions
9. Head cuts
10. Old terraces and flood plains
11. Current terraces and flood plains
12. Riparian conditions
13. Valley type

# 17.0 EQUIPMENT CHECKOUT

## 17.1 Digital Camera Checkout

SWQB has five identical Nikon Coolpix L6 digital cameras for field investigations and outreach activities. Four of these cameras are located in Santa Fe and stored in the locked black file cabinet (locker) in front of Room Number N2103. One camera is housed in the Albuquerque Field Office, available through the SWQB staff person there. Each camera has been assigned a unique designation letter for identification and tracking purposes. Cameras A through D are in Santa Fe; Camera E is in Albuquerque. These new devices, along with the old Sony digital cameras 3, 4, and 5, will be subject to this checkout and operation SOP until further notice.

- Staff are required to use the spreadsheet that corresponds to the camera of interest found in the SWQB Public folders on Magneto under the CAMERAS CHECKOUT subdirectory. Information required for each blocked-out set of dates for checkout must include a contact name. Under most circumstances, cameras are available on a first-come first-serve basis and can be held in reserve as necessary without qualification.

- Staff will obtain and return the locker key (silver w/ round-head labeled “EQPT Cabinet” on keychain with white “Express Scripts” clear plastic ring bauble) from the key box located at the entrance to N2104 when checking out a camera and its accessories. Staff will repeat this process when returning a camera and all its accessories to the locker after use.

PLEASE NOTE: Concerning the new Nikon units, it is acceptable for staff to simply check out a camera with only its carrying case and without taking all of its accessories out into the field. On return, however, staff are required to obtain an audio-video transfer cable from the locker to facilitate uploading their images onto Magneto.

Checkout items for Nikon cameras include:

- Camera and carrying case (with shoulder strap in back zipper pocket)
- Audio-video transfer USB cable (required)
- System Box (discretionary), which contains:
    a. Quick Start Guide
    b. Instruction Manual
    c. Nikon PictureProject 1.7 CD-ROM
    d. SanDisk 1.0 Gb Memory Card information
    e. Nikon Limited Warranty contact information

Checkout items for Sony cameras include:

- Camera and carrying case.

PLEASE NOTE: Image transfers from Sony cameras will still require delivering the unit to Gary King in N2206 who is responsible for uploading all saved images to the appropriate public folder on Magneto, powering the unit, returning it to the locker, and finalizing the checkout process.

## 17.2 Cell Phone Checkout

The Surface Water Quality Bureau has five cell phones for remote staff activities. These cell phones are located in Santa Fe and stored in the locked black file cabinet (locker) in front of N2103, Marcy Leavitt's door. Each cell phone identified by its designated telephone number. These devices will be subject to this checkout SOP until further notice.

- Staff are required to use the spreadsheet which corresponds to the cell phone of interest found in the SWQB Public folders on Magneto under the CELL PHONE CHECKOUT subdirectory. Information required for each blocked-out set of dates for checkout must include a contact name. Under most circumstances, cell phones are available on a first-come first-serve basis and can be held in reserve as necessary without qualification.

- Staff will obtain and return the locker key (silver w/ round-head labeled "EQPT Cabinet" on keychain w/ white "Express Scripts" clear plastic ring bauble) from the key box located at the entrance to N2104 when checking out a cell phone and its accessories.

PLEASE NOTE: It is acceptable for staff to simply checkout a cell phone w/ only its carrying case and without taking all of its accessories (Verizon pamphlets, instruction manuals, and CD-ROMs) out into the field.

Checkout items for cell phones include:

- Cell phone and carrying case (w/ car power jack and wall power jack) (required)
- Samsung® Instruction manual (discretionary)
- Verizon® service brochure (discretionary)
- Verizon® CD-ROM (discretionary)
- Verizon® system plans (discretionary)

## 17.3 Global Positioning System (GPS) Checkout

The Surface Water Quality Bureau has six global positioning system (GPS) units for field investigation activities. These GPS units are located in Santa Fe and stored in the locked black file cabinet (*locker*) in front of N2103, Marcy Leavitt's door. Each GPS unit has been assigned a unique designation number, 1 – 6, for identification and tracking purposes. These devices will be subject to this checkout SOP until further notice.

- Staff are required to use the spreadsheet which corresponds to the GPS unit of interest found in the SWQB Public folders on Magneto under the GPS UNIT CHECKOUT subdirectory. Information required for each blocked-out set of dates for checkout must include a contact name. Under most circumstances, GPS units are available on a first-come first-serve basis and can be held in reserve as necessary without qualification.

- Staff will obtain and return the locker key (silver w/ round-head labeled "EQPT Cabinet" on keychain w/ white "Express Scripts" clear plastic ring bauble) from the key box located at the entrance to N2104 when checking out a GPS unit and its accessories.

PLEASE NOTE: It is acceptable for staff to simply checkout a GPS unit w/ only its carrying case and without taking any of its accessories (Instruction manuals, extra batteries, etc.) out into the field.

Checkout items for GPS units include:

- GPS unit and carrying case (required)
- eTrex Vista® Owner's Manual and Reference Guide (discretionary)
- Trimble® GeoExplorer II© Operational Manual
- Replacement batteries (discretionary)

**FORMS**
Camera and Photograph Field Form

# APPENDIX A- FORMS

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| Sonde Deployment Instructions | 9 |
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