A MANUAL OF PROCEDURES FOR THE SAMPLING OF SURFACE WATERS

ADEQ
Arizona Department of Environmental Quality

Janet Napolitano, Governor
Stephen A. Owens, ADEQ Director

Prepared by

Hydrologic Support and Assessment Section
Surface Water Monitoring and Standards Unit
1110 W. Washington Street
Phoenix, Arizona 85007

Lin Lawson, Editor

The Arizona Department of Environmental Quality shall preserve, protect and enhance the environment and public health, and shall be a leader in the development of public policy to maintain and improve the quality of Arizona's air, land and water resources

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Steven Pawlowski,
Surface Water Monitoring and Standards Unit Manager

Staff from the Surface Water Monitoring and Standards Unit

Lee Johnson,

Doug McCarty

Kyle Palmer

Patti Spindler

Roland Williams

TMDL and Assessment Unit

Robert Scalamera,

Southern Regional Office`
Lin Lawson

PREFACE

A Manual of Procedures for the Sampling of Surface Waters provides a single source of information describing procedures used by personnel of the Arizona Department of Environmental Quality, Hydrologic Support and Assessment Section, for the collection and management of surface water data and related environmental information. It may be used as a guideline for all surface water sample collections performed by ADEQ personnel, ADEQ contractors, environmental organizations, private companies and corporations, and educators.

Any reference to specific brand names or model numbers is intended for the sake of clarification purposes and in no way represents an endorsement of such product.

This manual, or portions of it, will be updated whenever available technologies, procedures, or quality assurance protocols change. The Manual is posted on the ADEQ website and is available in printed form at the ADEQ library at 1110 W. Washington Street, Phoenix, Arizona. Updates to the Manual will be posted in a timely manner on the website and in printed form in the library.

INTRODUCTION

A Manual of Procedures for the Sampling of Surface Waters is a presentation of surface water sampling procedures and related activities by the Hydrologic Support and Assessment Section. The Manual is subdivided into six sections, each numbered separately from the others. A Table of Contents is given at the beginning of each section.

1) **Pre-Trip Administrative Activities** documents the actions required or necessary for the successful completion of surface water sampling activities. It describes how to select sampling sites, explains site safety while engaged in sampling activities, provides suggested equipment lists for various types of field work, presents the field data sheets used by ADEQ staff to collect field data, and explains the process of submitting water samples to the analytical laboratory.

2) **Equipment Calibration and Cleaning Procedures** explains the processes performed on sampling equipment before traveling to the sample site.

3) The **Field Procedures** section is the most extensive portion of the Manual and therefore has been subdivided into three parts. Part A, Basic Field Procedures, detail those activities directly involved in collecting field data for water quality, bacteria, macroinvertebrates and algae. Part B, Geomorphology Procedures, describe those activities that assess the physical properties of stream channels. Part C, Habitat Assessments Procedures, describe the methods used to collect and assess habitat and the biological condition of wadeable streams.

4) The **Post-Trip Procedures** relates the activities related to quality control for a water sampling multimeter.

5) **Data Management** is the process that details handling of collected data with emphasis on quality assurance and quality control.

6) **Supporting Material** is an appendix to the Manual and is a repository for procedures and processes indirectly related to sampling activities.

SECTION 1

PRE-TRIP ADMINISTRATIVE ACTIVITIES

TABLE OF CONTENTS

1.00 Pre-Trip Administrative Activities

1.10 Selecting and Naming Sample Sites

1.10.1 Selecting Sample Sites for Chemical and Physical Monitoring

Ambient surface water monitoring programs have been charged with the monitoring of state waterbodies. Previous historic data or general institutional knowledge as to the flow regime of proposed sampling sites should be employed during the planning and reconnaissance stages of the monitoring project. Walking the reach of the proposed sampling area is an invaluable exercise for understanding the geology, fluvial geomorphology, riparian condition and composition, and stream characteristics of the reach and should be done for any stream reconnaissance work prior to the selection of a sampling site. Any springs or tributaries contributing to the flow within several hundred feet of the selected sampling site should be noted. Additional inputs to the stream system (e.g. tributaries, non-point sources) have the potential to alter the chemical characteristics of water, downstream of the input and create collinear flows along one side of the channel. All relevant maps and data from legitimate sources pertaining to the site should be used during reconnaissance. When the objective is to sample perennial waterbodies and visits to the proposed site determines that there is an absence of any discharges, then site selection for the affected area should be reevaluated and the selection of another site considered.

Maximum accessibility and safety are principal considerations when selecting a sampling site; for example, higher water levels may limit access. Streams a few inches deep during the summer can easily fill to several feet deep during vernal runoff. Road access and road condition to sample sites throughout the year needs to be considered. Streams that may appear too deep or too swift to sample safely probably should not be considered for sampling. The “Rule of Nine” (maximum velocity x depth = <9) should be used as a guideline for entering flowing waters. A stream having a velocity times depth value of 9 or greater should not be entered.

Locating the sample site near a recognizable streamside landmark (large tree, rock outcrop, meander) will make it easier to recognize the site on subsequent visits. Any proposed site situated on private property requires permission to enter from the landowner. ADEQ does not have permission to sample any waters on Indian lands. On public lands, the managing agency (BLM, Trust Lands, USFS, etc) should be notified of the intent to sample.

Adherence to the following guidelines for site selection should ensure representative sampling.

- Consider the objectives of the project. What is being characterized? If the intent is to capture cumulative effects for a basin, sub-basin, or watershed, the site selected should be near the base of the catchment. If the intent is to characterize a particular reach or tributary, do not sample below the confluence that defines the terminus of the reach or tributary. If springs are in the potential sampling reach, it may be advisable, depending on project objectives, to relocate the site upstream of the spring input.

- Ideally, sites should have morphological features of stability. If stable sites cannot be found, select the best that's available. Avoid locations of incisement and aggradation. Bedrock-dominated areas are the best examples of stable channels, but they may not be suitable for other types of monitoring, such as macroinvertebrate collections. Other undesirable features for site selection are shifting channels, sand-dominated reaches, and locations downstream of excessive bank erosion.
- Areas of high turbulence and/or elevated gradients should be avoided. This may not be possible if one is working in an Aa+ or A stream type. Although these conditions can assure thorough mixing of water and representative samples, flow measurements may not be accurate. Areas of low to moderate turbulence are usually selected for water samples.
- For flow measurements, select a straight portion of channel having a smooth uniform bottom without obstructions.
- When cross-sections are required, avoid areas of braded channels and mid-channel bars.
- Do not sample in standing waters. Sometimes in drought conditions, there will be water in the channel, but it is pooled and not flowing.
- When existing sites become overgrown with aquatic vegetation, it may be advisable to temporally relocate the sample location as close as possible to the original site until the condition improves. The temporary sample location should not be more than 1/8 mile (200 meters) from the existing site and must share the same hydrologic characteristics as the established site.
- The ideal sampling site location is near an established USGS gauging station. Gauging stations are generally accessible and located in areas with stable channel morphology. Executing a streamflow discharge measurement at a gage site is an option that will allow for comparison between ADEQ measured and USGS gage determined flows (Section 3C, 3.183.22). Comparison of flow data is recommended on the initial site visit and at periodic intervals during the period of sampling.

1.10.2 Assigning Monitoring Site Identification Codes for Arizona Streams

In order to maintain consistency and avoid redundancy when giving monitoring sites their unique identification codes, a set of rules governing naming conventions in Arizona has been developed. The rules are to serve as a guideline when assigning monitoring site identification codes which will ensure maximum consistency and minimum redundancy between ADEQ surface water monitoring programs.

The process of assigning a unique monitoring site identification code means that three separate pieces of information need to be determined:

Watershed Code
Stream Identification Code
Miles on Stream from terminus

An example of the monitoring site identification code is:

VREVR025.30

Where:

VR = Watershed Code for the Verde River watershed
EVR = Stream identification code for the East Verde River
025.30 = Number of river miles, measured from the terminus (mouth), that pinpoints the location of the monitoring site on the stream

1.10.2.1 Stream Identification Code Naming Rules

Each site code clarifies its location and individuality from any other stream code in the state.

Rule 1 - Procedure for naming streams ending in "Creek"

A1 Creek ID codes are primarily derived from the first 3 letters of the creek name. If this results in a duplicate code or the last letter is a dedicated letter from the list in Rule 2, then the code comes from the first letter and next two previously unused consonants of the name; examples: Butcher Creek = BUT, and Butte Creek = BTT. (For naming purposes, the letter "Y" is always considered a consonant.)

A2 If the procedure specified in A1 results in a duplicate ID code, then the next consonant is used that doesn't result in a duplicate. If all the consonants result in duplicates, then the first vowel after the first consonant is used. This procedure continues until a discrete ID code is found; example: Concho Creek = CNH.

B. Creek ID codes should not end in a "C", with a few exceptions.

C. Creek ID codes cannot use the dedicated endings listed in Rule 2.

D. For creek names which consist of 2 letters, use "K" from the word creek for the third consonant (P B Creek = PBK).

Rule 2 - Procedure for naming streams not ending in "Creek"

A1 For stream names ending with the word Canyon or Canal, use a "C" as the dedicated ending. (These are streams without "Creek" being the last word in its name). The rest of the ID comes from the first letter and next consonant in its name; example: Peeples Canyon (Creek) = PPC. If the word "Canyon" is included in a Creek's name but is not the last word in the name, Rule 1 is used.

A2 For stream names ending with the word River or Run, use an "R" as the dedicated ending. The rest of the ID comes from the first letter and next consonant in its name; example: Blue River = BLR; Johnson Run = JSR.

A3 For stream names ending with the word Wash, use a "W" as the dedicated ending. The rest of the ID comes from the first letter and next consonant in its name; example: Vekol Wash = VKW.

A4 For stream names ending with the word Gulch, use a "G" as the dedicated ending. The rest of the ID comes from the first letter and next consonant in its name; example: Snake Gulch = SNG (Note that other codes may also end in "G").

B1 If the procedures listed in Rule 2. A1 through A4 results in duplicate ID codes, then the same procedure specified in Rule 1, A2 is followed until a unique ID code is found.

Rule 3 - Directional streams and streams having common names

A1 Whenever a waterway has a common word at the beginning of its name (such as East or San), only the first letter of the common name is used followed by the beginning letter of the second word and then the next letter or the dedicated ending consonant; examples: San Pedro River = SPR; East Clear Creek = ECL, Little Ash Creek = LAS.

A2 If the procedure listed in 3. A1 results in a duplicate ID code, then the same procedure specified in Rule 1, A2 is followed until a discrete ID code is found.

Rule 4 - Directional forks

A1 If the stream is a directional fork of another stream (East Fork Black River) and is not a creek, the first letter of the direction is used as the first letter in the ID code. The word "Fork" is ignored. The second letter for the ID is the first letter of the third word or main part of the stream name (e.g., East Fork Black River = EBR; North Fork of the East Fork Black River = NBR). The third letter in the code uses the dedicated endings as specified in Rule 2.

A2 If the stream is a creek, the first letter of the direction is used as the first letter of the ID code. The word "Fork" is ignored. The second and third letters of the code come from the first and second letters of the third word or main part of the stream name. If the stream name contains 3 or more words then use the first letter of the direction plus the first letters of the 2 words in the name (other than Fork) (example: North Fork Bear Wallow Creek = NBE). In this example, "W" could not be used because it is a code letter for Wash. Instead, by Rule 1, the letters "BE" are used for the stream code.

A3 If the procedure listed in 4. A1 results in a duplicate ID code, then the same procedure specified in Rule 1, A2 is followed until a discrete ID code is found.

Rule 5 - Streams with 3 or more words in the name

A1 If the stream is a creek, use the first two consonants of the first word and the first letter of the second word in the name. If this results in an inappropriate ending, then use the first letter of the first word and the first letter and second consonant of the second word (e.g. Copper Camp Creek = CCM). Note that the ending "C" could not be used because that is the designated ending for Canyon. The consonant "M" was used instead.

A2 If the stream is not a creek, use the first letter of the first and second word, ending with the stream type (e.g. Big Chino Wash = BCW)

Rule 6 - Streams named with numbers

A1 For streams which have a number as their sole name and which are listed as creeks, use the number as the stream code plus M or MI to indicate miles (Eightmile Creek = 8MI)

A2 For streams which have a number as their sole name and which are not listed as creeks, use the number plus the appropriate designated ending (Thirteen mile wash = 13W).

A3 For streams which have a number as their sole name and are triple digit numbers, use the number only for the stream code (One hundred and thirty mile creek = 130).

Rule 7 - Duplicate codes

A. When multiple stream codes occur and all letters of the stream name have been used, a number will be assigned for the middle digit of the name, beginning with number 1 through 9, then 0 (e.g. Sand Hill Wash = S1W).

Rule 8 - Canals and Laterals

A. Canals which are named by a single letter will be given a code consisting of the letter, a number from 1-9 which makes the stream code unique in that basin, and the last digit will be C for canal (A Canal = A1C).

B. Similarly, laterals which are named by a single letter will be given a code consisting of the letter, a number from 1-9 which makes the stream code unique in that basin, and the last digit will be L for lateral (Lateral A = A1L).

Rule 9 - Named Springs

A1 Named springs will be coded using a three-letter code representing the name of the spring plus the distance of the spring from its terminus at the nearest downstream

stream. Use the same naming rules for springs as for streams (Caddis Spring = CAD000.90). Springs should be coded so as not to duplicate existing stream codes.

Rule 10 - Unnamed Springs

A1 Unnamed springs will only be assigned codes when they become sampling sites; coding will not be done a-priori. The code will consist of an "S" for spring plus a two letter code. The two letters will come from the first two consonants of the name of the downstream tributary if named. If the downstream tributary is also unnamed, then an alphabetical code will be assigned (unnamed spring tributary to Bitter Creek = SBT, unnamed spring #1 tributary to unnamed stream = SAA; unnamed spring #2 = SAB). A master list will be kept by the database coordinator to track these codes.

Rule 11 - Unnamed streams

A1 Unnamed streams will only be assigned codes when they become sampling sites; coding will not be done a-priori. The code will consist of a "U" for unnamed stream plus a two letter code. The two letters will come from the first two consonants of the name of the downstream tributary if named. If the downstream tributary is also unnamed, then an alphabetical code will be assigned (unnamed stream tributary to Bitter Creek = UBT, unnamed stream #1 = UAA; unnamed stream #2 = UAB). A master list will be kept by the database coordinator to track these codes.

1.10.2.2 Miles on Stream from Mouth

Once the monitoring site has been located in the field with a GPS unit, its location is transferred to a GIS system. The GIS system is used to determine the number of miles on the stream from the mouth to the monitoring site. In all cases, monitoring points will be recorded to at least 1/100th of a mile. This process generates the number portion of the stream code.

1.10.2.3 Determining River Miles for New Sites

This procedure is undergoing a technical transformation at time of printing and is not available.

1.11 Safety Procedures

Personal safety of staff engaged in any field work activity (e.g., in transit, walking or hiking, and any field activities while at the sample site) is of primary importance. Staff should never place themselves in dangerous or risky situations. Any hazards (e.g. mine shafts, rattlesnake infested areas, etc.) that are known by field personnel should be communicated to other members of the field crew. It is recommended that the field work be postponed if there is indication that engagement in the field activity could cause bodily harm other than the normal risks associated with field work. All field work has risk of personal injury which is stated in the job classifications for field work employment; this is normal risk. Other than normal risk are hazardous conditions not typically encountered in routine field work. Examples are 1) lightening storms, 2) night work, 3) flash flood conditions, and 4) snowy weather. If any member of the field crew is uncomfortable with a reasonable self-determined hazardous field condition, it is that person's responsibility to bring this to the attention of the project lead and that person is not required to complete the work assignment. A "reasonable self-determined hazardous field condition" is defined as other than normal risk. The project lead shall not dismiss any person's spoken concerns that field conditions are too hazardous to complete the work assignment.

The following sub-sections provide specific guidelines regarding safety while fulfilling field work assignments.

1.11.1 Field Trip Routing and Telephone Check-in Procedure

Before any field trip is conducted, the Hydrologic Support and Assessment Daily Trip Routing and Site Routing Information form (Figure 1) must be completed. Copies should be given to other staff members assisting on the trip and to the Unit Manager or substitute supervisor.

1.11.2 Site Safety

The following guidelines apply to all field work by staff employed in the Hydrologic Support and Assessment Section.

- Field sampling crews should consist of at least two members unless otherwise approved by the supervisor.
- Be conscious of the whereabouts of rattlesnakes, mountain lions, and other dangerous animals.
- Wear protective footing when entering streams.
- Open body wounds are entry sites for infection; take the necessary precautions for self protection.
- Do not enter waters that have a depth x velocity factor greater than 9.
- If there is storm activity in the work area, wait for safer conditions to develop or postpone the work assignment. For stormwater sampling see Section 1.11.3.

Figure 1. Hydrologic Support and Assessment Daily Trip Routing and Site Routing Information Form.

Fixed Station Network Monitoring Daily Trip Routing

Vehicle⇨	License #		Year		Make		Color	

Employee on Trip		Emergency Contact ⇨	Home Phone	Work Phone
1				
2				
3				
4				

Daily Check-In With ⇨	Steve Pawlowski	Work Phone	602.771.4219	Home Phone	480.839.6379
Prepared by				Date	

Site Routing Information

❶	Date		Site	
Route to Site:				
Check-in Time		Lodging		Phone #

❷	Date		Site	
Route to Site:				
Check-in Time		Lodging		Phone #

❸	Date		Site	
Route to Site:				
Check-in Time		Lodging		Phone #

❹	Date		Site	
Route to Site:				
Check-in Time		Lodging		Phone #

❺	Date		Site	
Route to Site:				
Check-in Time		Lodging		Phone #

❻	Date		Site	
Route to Site:				
Check-in Time		Lodging		Phone #

- Do not sample at night without approval from the supervisor. If night sampling is approved see Section 1.11.3, Stormwater Sampling Field Safety Guidelines.
- Do not trespass on private property, Indian reservations, or posted restricted public lands without prior permission and written approval from property owner or administrator.
- Extreme caution must be taken when working at sites along or near the U.S./Mexican border. This zone has become increasingly violent.
- If strange or suspicious looking people are in the work area, either wait for them to leave or postpone the work to a later time. Do not force confrontations with strangers and back away from imposed confrontations.
- Take the necessary precautions against exposure to harmful weather conditions (e.g. heat, cold, snow, wind).
- The project lead is responsible for providing water at the work site for drinking and washing. However, it is each person's responsibility to provide enough drinking water for their own use on any work assignment. Do not rely upon others for water needs. Recommended amounts of water for summer work is two gallons per person in the field vehicle and at least one quart per person away from the vehicle.
- It is each person's responsibility to wear proper clothing for the type of work to be performed and the expected weather conditions at the work site.
- Walking in streams and along stream banks are slip-and-fall conditions and it is the observer's responsibility to take appropriate precautions against sustaining personal injury.
- A field routing form (Figure 1) must be completed before departure into the field.
- It is recommended that the project lead have a cellular or satellite phone while conducting field work.

1.11.3 Stormwater Sampling Field Safety Guidelines (Provisional Status)

These guidelines are in effect until a comprehensive stormwater sampling SOP is approved. Being "guidance," these are not official ADEQ policies. The guidelines presented are intended to aid staff in making careful decisions in the field. They do not address administrative issues such as overtime, liability, insurance, etc. These guidelines are provisional and subject to modification.

1.11.3.1 Administrative Guidelines

Every stormwater sampling project shall have an approved Site Safety Plan. Minimum requirements for the Plan are:

- Personal Protective Equipment requirements
- Required training qualifications (OSHA, MSHA, etc.)
- Known and potential hazards and precautions
- Directions and map to nearest hospital

- ❖ Emergency phone numbers for Law enforcement, medical emergencies, and private and public land owners
- ❖ A Sample Site List indicating hazard awareness and work prohibitions, such as excessive flash flood risk, excessive night hazards, etc.

1.11.3.2 On-Site Daytime Guidelines

A copy of the Site Safety Plan shall be provided to all on-site staff.

No sample or measurement is worth the risk of injury.

Carefully evaluate a given on-site situation to determine if the task can be performed safely. Consider potential hazards to avoid and prepare for worst-case scenarios.

Always respect the on-site opinions of co-workers regarding safety issues.

Use a personal flotation device when working around swift or deep waters.

When fording a stream, the following requirements are necessary for evaluating a safe passage; the depth and velocity of the water at the crossing, the vehicle limitations, and adequate experience and driving skills of the operator. Consider what the depth of water under worst conditions could be if a return crossing is necessary. Use extreme caution when entering water deeper than the truck's axle or where the water is higher than the bottom of the truck.

Do not use chest waders under any circumstances.

Do not enter waters deeper than just above the knee.

Do not enter waters that have a depth x velocity factor greater than 9.

1.11.3.3 On-Site Nighttime Guidelines

All daytime safety guidelines apply to nighttime sampling. Additional guidelines for nighttime sampling are listed.

Participation in nighttime stormwater sampling is voluntary.

Fording a stream at night under stormy conditions is dangerous and extreme caution should be exercised when doing so.

Wear a personal flotation device when entering a stream.

Use a headlamp.

Do not hike more than 500-feet from the field vehicle.

Take either a satellite phone or cell phone. A cell phone is sufficient if excellent cell phone coverage is available at the site. If cell phone coverage is insufficient, a satellite phone is required.

Call or leave a message with the supervisor before leaving after normal work hours and call-in no later than 0900 hours by the next day.

For sites considered too hazardous to sample at night, reevaluate those sites the next morning before proceeding to take samples.

1.12 Field Trip Equipment List

The following is a list of equipment and supplies typically required for field work. It may not be complete for all projects.

Meters	**Meter Calibration Supplies**
Hydrolab MiniSonde Hydrolab Scout2 Hydrolab MiniSonde Hydrolab Scout2 Marsh McBirney Model 2000 Flow Meter and wading rod Hach 2100P Turbidimeter and standards Hach DR/700 Colorimeter and packets GPS unit Barometer	KCl standards pH buffers - 4, 7, 10

Sampling Equipment	**Filtered Metals Accessories**
Churn splitters Ice chests and ice Wading boots Chemsets Fiberglass measuring tape and tent pegs Thermometer, digital Suspended Sediment Concentration Sieve DH-81 and sample bottles Backpack(s) Duffle bag Meter stick Spherical Densiometer pH test strips Camera and print film Clipboard	Peristaltic pump/hoses Capsule filters
	Safety and First Aid Gear
	Sunscreen Bug Repellent with DEET Raingear Drinking water Cellular or satellite phone Flashlight Tool kit and flares

Surveying Equipment	
Laser level	DH-76 or DH-74 SSC sampler w/ glass jars
Extendable Rod	3" Helly Smith bedload sampler w/ 500micron mesh bag
Laser receiving unit	Bedload jars or bags
Tripod	6" Helly Smith bedload sampler w/ 500micron mesh bag
Rolls of flagging	A" and "B" reels for use on cableways
Stake flags	Shovels, monuments, galvanized steel pipe and concrete
Field data sheets	Bucket setup for bar samples
GPS-garmin	Set of sieves and scale
GPS-GeoExplorer	Lathe stakes
Auto Levels	BEHI equipment (16' rod, meter stick w/ line level, angle measure or calculator)
Rods & rod levels	Metal tags
Measuring tape reel , 1/10 ft	Walkie talkies
Cross-section and caps	Clipboards w/ reference documents
Short & long-handled sledgehammers	Digital camera or 35mm Camera
Machetes	
Tightening strap and chaining pin	
Bank pins	
Toe pins and caps	
Scour chains, duckbills, and driver	
Boltcutters	

Forms and Accessories	**Cleaning Accessories**
Field forms	Tap water
Truck logbook	DI water
Routing form	Hydrochloric Acid
Pencils/waterproof pens	Drinking water
Calculator	Kimwipes/towels
Photo log	DI squeeze bottle
Laboratory submittal form	Soap
Laboratory continuation forms	Brush
Tracking, Purchase Order, PCA, and Index numbers	Rubber gloves
	Trash receptacle
	Acid waste bucket

Bacteria Sampling Accessories	**Macroinvertebrate Supplies**
Whirlbags Filter tops Filter bottoms Handpump/hose Membrane filters Tweezers Syringes Pipettes MTEC Media Buffered water Urea Broth Controls Wooden swabs Lighter/candle Alcohol Waterproof marker Bacti rack Peristaltic pump and hose Incubators and cords Antibacterial soap	Sieve, 500 micron mesh Plastic Spoon Dissecting Tray/white pan Forceps Isopropanol Formalin Nalgene Bottles Labels Bucket D-frame dip net (500 micron mesh) 2-3 gal bucket Magnifying loop

Algae, from Natural Substrates	**Habitat**
Flagging tape Exacto Knife Sample jars 9cm2 template Squeeze bottle Toothbrush Labels	Pebble count ruler Densiometer Tapes, Rod and ruler Calculator w/ inv Tangent function

Lakes Equipment	Wildco Sediment Sampler
GPS	Eckman Dredge
Depth Finder	Plant Press
Plum	Field notes
Boats	Vertical Plankton Net
Hydrolab H20 or similar multi-meter	Flow Meter
Surveyor Logger	Cameras
YSI-sonde	Bouy
Turbidimeter	Colilert equipment
Secchi Disk	Altimeter
Beta Bottle	GeoPump
Sample bottles	Filters/ tubing
MTBE Bomb	Thermometer
Churn Splitter	

TMDL Equipment	
Flow Meter, Marsh McBirney 200	Field Wind Speed (hand anemometer, estimate)
Flow Meter, Marsh McBirney 200-D	Field Wind Direction (compass, landmark, sun-moon)
Flow Meter, Marsh McBirney 200	Colilert equipment
Wading Rod	GeoPump
Fiberglass Tapes	Churn Splitter
Calibrated Volume Container (5 gal bucket for discharge)	Air Temp (themometer- stand alone, hydrolab, estimate)
Display & Data Retrieval for 4a Sondes (w/ memory), Hydrolab Surveyor 4a	Turbidity Meter, Hach 2100P Turbidity Meter
Multi-Parameter Probe (temp, SC, DO, pH, ORP) (no memory), Hydrolab MiniSonde 4a	Multi-Parameter Probe & Display (temp, SC, DO, pH, turb), Hydrolab Quanta Display/Sonde
Multi-Parameter Probe (temp, SC, DO, pH, ORP)(w/memory), Hydrolab MiniSonde 4a(4-battery)	Multi-Parameter Probe (temp, SC, DO, pH, ORP)(w/memory), Hydrolab MiniSonde 4a(8-battery)
Field Barometric Pressure (HL Surveyor, NWS, stand alone Inst)	Field Time (watch, GPS, PC, stopwatch)
Automatic Sampler, American Sigma 800	GPS, Garmin GPS III+, GPS, Trimble
Automatic Sampler, American Sigma 900 MAX	Pressure Transducer, Instrumentation Northwest
Automatic Sampler, ISCO 6712	Rain Gage - 8", J&S Instruments
Water Level Logger, Global WL-14	Wind Speed, RM Young
Water Level Logger, Global WL-15	Air Temperature, Onset Hobo Temp Pro
Water Level Logger, Dycor	Air Temperature, Campbell
Datalogger, DataTaker DT50	Air Temp/Relative Humidity
Datalogger, Campbell CR10X	Depth to Water Probe, Keck
Datalogger, OnSet HOBO	Soil Temperature, Campbell
Data Retrieval, OnSet HOBO Data Shuttle	Solar Radiation, Li-Cor
Data Retrieval, Dell Lattitude PC	Turbidity, FTS DTS-12
Rain Gage - 8", J&S Instruments	Depth Integrated Sampler, DH-81
Bedload, Helly-Smith	Coolers

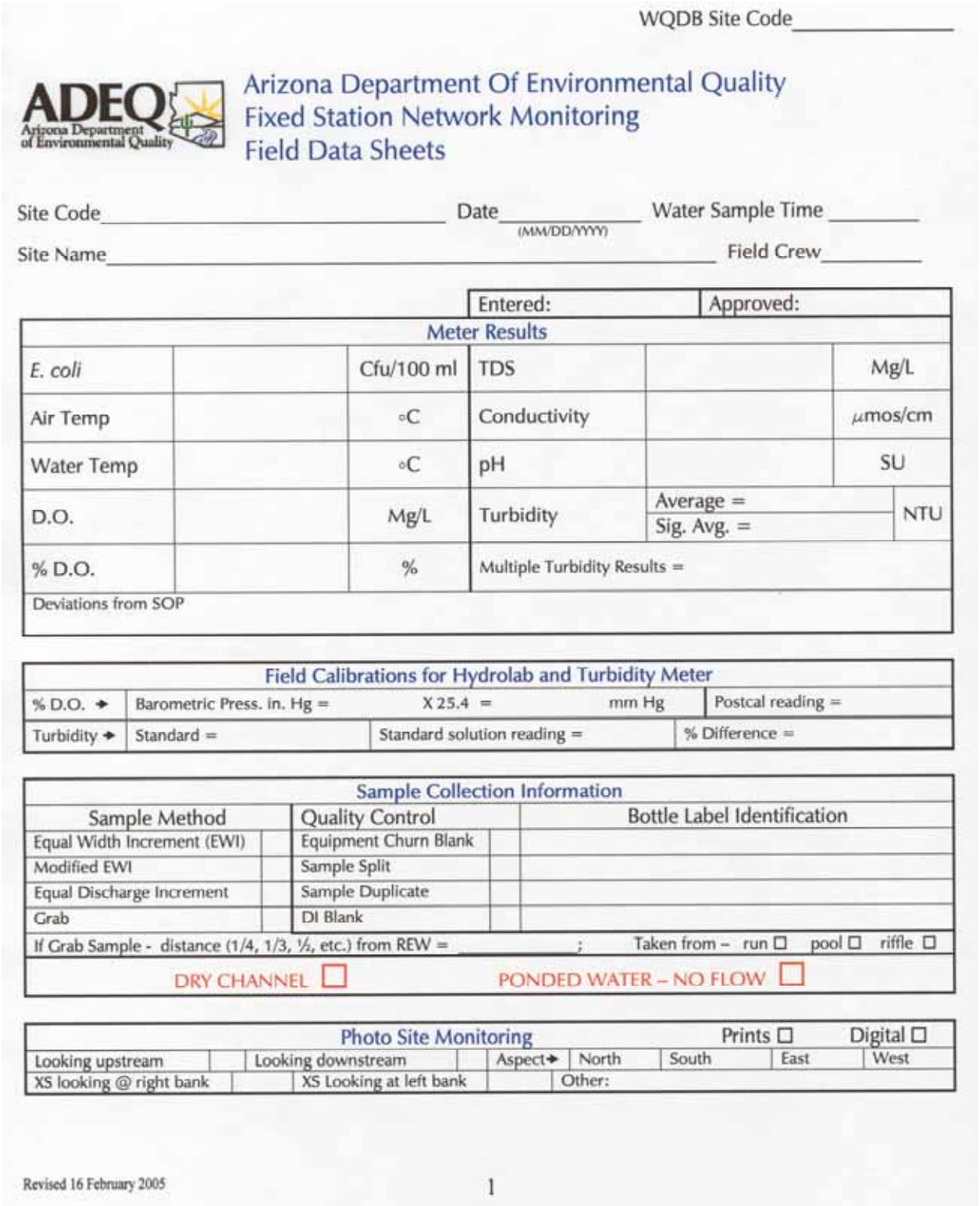
1.13 Field Data Sheets

The primary surface water quality program of ADEQ is the Fixed Station Network (FSN) Program. The primary purpose of the FSN program is to obtain data to characterize baseline water quality conditions of wadeable, perennial streams and to determine long-term water quality trends.

Each selected site is sampled four times a year. Data are collected on the physical characteristics, bacteria levels, and general water chemistry of the stream. An annual bioassessment and habitat assessment is conducted at each FSN site in the spring to assess the health of the biological community at each site.

The field data collecting form for the FSN program is the Fixed Station Network Monitoring Field Data Sheets (Figure 2). The biological and habitat assessment form is the Stream Ecosystem Monitoring Field Data Sheets (Figure 3). Both forms are presented on the following pages.

Figure 2. Fixed Station Network Monitoring Field Data Sheets.



WQDB Site Code_____

ADEQ Arizona Department of Environmental Quality

Arizona Department Of Environmental Quality
Fixed Station Network Monitoring
Field Data Sheets

Site Code_____ Date_____ (MM/DD/YYYY) Water Sample Time _____

Site Name_____ Field Crew_____

			Entered:	Approved:	
Meter Results					
E. coli		Cfu/100 ml	TDS		Mg/L
Air Temp		◦C	Conductivity		µmos/cm
Water Temp		◦C	pH		SU
D.O.		Mg/L	Turbidity	Average = Sig. Avg. =	NTU
% D.O.		%	Multiple Turbidity Results =		
Deviations from SOP					

Field Calibrations for Hydrolab and Turbidity Meter			
% D.O. →	Barometric Press. in. Hg =	X 25.4 = mm Hg	Postcal reading =
Turbidity →	Standard =	Standard solution reading =	% Difference =

Sample Collection Information				
Sample Method		Quality Control		Bottle Label Identification
Equal Width Increment (EWI)		Equipment Churn Blank		
Modified EWI		Sample Split		
Equal Discharge Increment		Sample Duplicate		
Grab		DI Blank		
If Grab Sample - distance (1/4, 1/3, ½, etc.) from REW = _____; Taken from – run ☐ pool ☐ riffle ☐				
DRY CHANNEL ☐		PONDED WATER – NO FLOW ☐		

Photo Site Monitoring					Prints ☐	Digital ☐
Looking upstream		Looking downstream		Aspect→ North	South	East	West
XS looking @ right bank		XS Looking at left bank		Other:			

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Field Conditions at Time of Visit
Flood Evidence w/in Last Month → None ☐; Fresh debris line in channel ☐; Grasses Laid Over ☐; Fresh debris line in bushes/trees ☐; Flood Width _____
Weather Conditions :
Precipitation at sample time : None ☐; Light ☐; Moderate ☐; Heavy ☐; Cloud Cover (%) =
Previous Precipitation (w/in 24 hrs.) : None ☐; Light ☐; Moderate ☐; Heavy ☐

Reach Observations	
General appearance in the channel (check all that apply)	No refuse visible ☐; Small refuse visible ☐; Small volume refuse common ☐; large volume refuse (tires, carts) rare ☐; large volume refuse common ☐
General appearance along the banks (check all that apply)	No refuse visible ☐; Small refuse visible ☐; Small volume refuse common ☐; large volume refuse (tires, carts) rare ☐; large volume refuse common ☐
Water Clarity	Clear ☐; Milky ☐; Light brown ☐; Dark brown ☐; Oily sheen ☐; Greenish ☐; Other _____
Water odor (check all that apply)	None ☐; Sewage ☐; Chlorine ☐; Fishy ☐; Rotten eggs ☐; Other _____
Appearance at water's edge (check one)	No evidence of salt crusts ☐; White crusty deposits rare ☐; Numerous white crusty deposits ☐; banks covered with white crusty deposits ☐
Fish presence	Absent ☐; rare ☐; Common ☐
Crayfish presence	Absent ☐; rare ☐; Common ☐
Sunfish presence	Absent ☐; rare ☐; Common ☐
Leopard frog presence	Absent ☐; Number observed alive _____; Dead _____
Floating leaves or other organic mater (not algae)	Absent ☐; rare ☐; Common ☐
Leaves or other organic matter on streambed	Absent ☐; rare ☐; Common ☐

Notes

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Flow Measurements					
Marsh-McBirney Flow Meter					
Measurement from: riffle ☐ run ☐ pool ☐					
Station	Distance from Initial Pt., ft.	Width, ft.	Depth, ft.	Velocity, ft/s	Discharge, cfs
1					
2					
3					
4					
5					
6					
7					
8					
9					
10					
11					
12					
13					
14					
15					
16					
17					
18					
19					
20					
21					
22					
23					
24					
25					
26					
27					
28					
29					
30					
	Total Width	QC width	Average	Average	Total Q

From USGS Gage	
USGS Gage Height =	USGS Discharge =

Float Method Discharge Measurement														
Timed Measurements, seconds														Avg. Time
Width, ft	X	Depth, ft	X	Dist., ft	X	Velocity, fps	X	0.85	=	Discharge, cfs				

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Albion Sample Documentation					
Metals	Composite ____ ; Grab ____ ; Blank ____ ; Duplicate ____ ; Split ____				
Hg	Composite ____ ; Grab ____ ; Blank ____ ; Duplicate ____ ; Split ____				
Deviations from 1669 protocol:	One person only processing the complete sample ☐		Re-used clean box ☐		
Delayed filtration/processing (not at site) ☐	Processed without a clean box, exposed to ambient atmosphere ☐				
No gloves or insufficient clean supplies (i.e. filter clogging, no replacement available) ☐			Other ☐		
Comments:					

E. Coli					
Collection Time ____	Distance (1/4, 1/3, ½, etc) from REW ____		From run ☐; riffle ☐; pool ☐		
Incubation Period is 24 hours for membrane filter technique and 18 hours for colilert technique					
Beginning Incubation Time ____ ; Enumeration Time ____					
Membrane Filter Results					
Dilution, ml	Number of colonies	Dilution Used in Calculation	Quality Control		
		☐		Pass	Fail
		☐	Equipment Blank	☐	☐
		☐	Technique Blank	☐	☐
Calculated Colony Forming Units/100 ml			Comments:		

Colilert Results			
Sample Number	Number Large Wells Positive	Number Small Wells Positive	Most Probable Number form Table
1			
2			
3			
4			
Average Most Probable Number =			cfu / 100 ml
Comments			

Field Meter Documentation			
Meter Type	Model	Serial Number	DEQ Name
Hydrolab			
Turbidimeter			
Incubator			
Flow Meter			
Camera Make :			
Chlorine Colorimeter			

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Figure 3. Stream Ecosystem Monitoring Field Data Sheets.

WQDB Site Number:_____

ADEQ Arizona Department of Environmental Quality

Stream Ecosystem Monitoring Field Data Sheets

Site Code_____ Date_____ (MM/DD/YYYY) Water Sample Time _____

Site Name_____ Field Crew_____

GPS: Latitude_____ Longitude_____

			Entered:	Approved:	
Meter Results					
E. coli		cfu/100 ml	TDS		mg/L
Air Temp		°C	Conductivity		µmos/cm
Water Temp		°C	pH		SU
Dissolved Oxygen		mg/L	Turbidity	Average =	NTU
				Signal Avg. =	
% D.O.		%			
Deviations from SOP					

Field Calibrations – Hydrolab and Turbidity Meter				
% D.O. →	Precal Reading =	Barometric Pressure: inches Hg =	X 25.4 =	mm Hg
	Postcal reading =			
Turbidity →	Standard =	Standard solution reading =	% Difference =	

Sample Collection Information				
Sample Method		Quality Control		Bottle Label Identification
Equal Width Increment (EWI)	☐	Equipment Churn Blank	☐	
Modified EWI	☐	Sample Split	☐	
Equal Discharge Increment	☐	Sample Duplicate	☐	
Grab	☐	DI Blank	☐	
If Grab Sample - distance (1/4, 1/3, ½, etc.) from REW = _____; Taken from – run ☐ pool ☐ riffle ☐				
DRY CHANNEL ☐		PONDED WATER – NO FLOW ☐		

Photo Reach Monitoring Log					Prints ☐ Digital ☐
Camera Make:		Model:		DEQ Name:	
☐	Upstream looking downstream	☐	Downstream looking upstream	☐	X-sec @ discharge location LDS ☐
☐	Upstream RB cross-section	☐	Downstream RB cross-section	☐	X-sec @ discharge location LUS ☐
☐	Upstream LB cross-section	☐	Downstream LB cross-section	☐	
☐	Upstream riffle substrate	☐	Downstream riffle substrate	☐	

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Flow Measurements					
Marsh-McBirney Flow Meter					
Measurement from: riffle ☐ run ☐ pool ☐					
Station	Distance from Initial Pt., ft.	Width, ft.	Depth, ft.	Velocity, ft/s	Discharge, cfs
1					
2					
3					
4					
5					
6					
7					
8					
9					
10					
11					
12					
13					
14					
15					
16					
17					
18					
19					
20					
21					
22					
23					
24					
25					
26					
27					
28					
29					
30					
	Total Width	QC width	Average	Average	Total Q

From USGS Gage	
USGS Gage Height =	USGS Discharge =

Float Method Discharge Measurement											
Timed Measurements, seconds											Avg. Time
Width, ft	X	Depth, ft	X	Dist., ft	X	Velocity, fps	X	0.85	=	Discharge, cfs	

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Albion Sample Documentation					
Metals	Composite ____ ; Grab ____ ; Blank ____ ; Duplicate ____ ; Split ____				
Hg	Composite ____ ; Grab ____ ; Blank ____ ; Duplicate ____ ; Split ____				
Deviations from 1669 protocol:	One person only processing the complete sample ☐	Re-used clean box ☐			
Delayed filtration/processing (not at site) ☐	Processed without a clean box, exposed to ambient atmosphere ☐				
No gloves or insufficient clean supplies (e.g. filter clogging, no replacement available) ☐	Other ☐				
Comments:					

E. Coli					
Collection Time ____	Distance (1/4, 1/3, ½, etc) from REW ____	From run ☐; riffle ☐; pool ☐			
Incubation Period is 24 hours for membrane filter technique and 18 hours for colilert technique					
Beginning Incubation Time ____ ; Enumeration Time ____					
Membrane Filter Results					
Dilution, ml	Number of colonies	Dilution Used in Calculation	Quality Control		
		☐		Pass	Fail
		☐	Equipment Blank	☐	☐
		☐	Technique Blank	☐	☐
Calculated Colony Forming Units/100 ml ⇨			Comments:		
Colilert Results					
Sample Number	Number Large Wells Positive	Number Small Wells Positive	Most Probable Number form Table		
1					
2					
3					
4					
Average Most Probable Number = ____ cfu / 100 ml					
Comments					

Field Meter Documentation			
Meter Type	Model	Serial Number	DEQ Name
Hydrolab			
Turbidimeter			
Incubator			
Flow Meter			
Camera Make :			
Chlorine Colorimeter			
Laser Level			

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Field Conditions at Time of Visit			
Flood Evidence within Last Month			
1. None ☐	2. Fresh debris line in channel above bankfull elevation ☐	3. Grasses laid over ☐	
4. Fresh debris line in bushes/trees ☐	5. Other ☐	6. Drought conditions prevailing ☐	Flood width
7. Recent flood event greater than baseflow but less than bankfull ☐		8. Riparian vegetation scoured away ☐	
Weather Conditions :			
Precipitation at sample time : None ☐; Light ☐; Moderate ☐; Heavy ☐; Cloud Cover (%) =			
Previous Precipitation (w/in 24 hrs.) : None ☐; Light ☐; Moderate ☐; Heavy ☐			

Reach Observations	
General appearance in the channel (check all that apply) (GAS)	No refuse visible ☐; Small refuse visible ☐; Small volume refuse common ☐; large volume refuse (tires, carts) rare ☐; large volume refuse common ☐
General appearance along the banks (check all that apply) (GAB)	No refuse visible ☐; Small refuse visible ☐; Small volume refuse common ☐; large volume refuse (tires, carts) rare ☐; large volume refuse common ☐
Water Clarity (WAP)	Clear ☐; Milky ☐; Light brown ☐; Dark brown ☐; Oily sheen ☐; Greenish ☐; Other_____
Water odor (check all that apply) (WOD)	None ☐; Sewage ☐; Chlorine ☐; Fishy ☐; Rotten eggs ☐; Other_____
Appearance at water's edge (check one) (AWE)	No evidence of salt crusts ☐; White crusty deposits rare ☐; Numerous white crusty deposits ☐; banks covered with white crusty deposits ☐
Fish presence	Absent ☐; rare ☐; Common ☐
Crayfish presence	Absent ☐; rare ☐; Common ☐
Sunfish presence	Absent ☐; rare ☐; Common ☐
Leopard frog presence	Absent ☐; Number observed alive _____; Dead_____
Floating leaves or other organic mater (not algae)	Absent ☐; rare ☐; Common ☐
Leaves or other organic matter on streambed	Absent ☐; rare ☐; Common ☐

Organic Debris / Channel Blockages in Active Channel			
☐	1. No organic debris or channel blockages	☐	6. Extensive, large debris dams either continuous or influencing over 50% of channel area. Forces water onto flood plain even with moderate flows. Generally presents a fish migration blockage.
☐	2. Infrequent debris, what's present consists of small, floatable organic debris.	☐	7. Beaver dams. Few and/or infrequent. Spacing allows for normal stream/flow conditions between dams.
☐	3. Moderate frequency, mixture of small to medium size debris affects less than 10% of active channel area.	☐	8. Beaver dams - Frequent. Back water occurs between dams - stream flow velocities reduced between dams.
☐	4. Numerous debris mixture of medium to large sizes - affecting up to 30% of the area of the active channel.	☐	9 .Beaver dams - abandoned where numerous dams have filled in with sediment and are causing channel adjustments of lateral migration, evulsion, and degradation etc.
☐	5. Debris dams of predominantly large material affecting over 30% to 50% the channel area and often occupying the total width of the active channel.	☐	10. Man made structures - diversion dams, low dams, controlled by-pass channels, baffled bed configuration with gabions, etc.

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Flow Regime	
☐	Perennial stream channel. Surface water persists all year long
☐	Intermittent stream channel. One which flows only seasonally or occasionally. Surface source includes springs, snow melt, and flows that reappear along various locations of a reach, then run subterranean (interrupted)
☐	Subterranean stream channel. Flows parallel to and near the surface for various seasons
☐	Ephemeral stream channel. Flows only in response to precipitation
Category	
☐	Seasonal variation in stream flow dominated primarily by snowmelt runoff
☐	Seasonal variation in stream flow dominated primarily by stormflow runoff
☐	Uniform stage and associated stream flow due to spring fed conditions
☐	Regulated stream flow due to diversions, dam releases, dewatering, effluent dominated, etc.
☐	Altered flows due to development, such as urban streams, cut-over watersheds, vegetation conversions (e.g. forested to grassland) that changes flow response to precipitation events

Stream Type Identification

Walk the reach and flag all likely bankfull indicators. Select the riffle with the best bankfull indicators to collect measurements. A measuring tape, stadia rod, and calculator are sufficient, although a laser level can also be used. Calculate the classification variables and use Rosgen stream type classification chart to identify stream type.

		Valley Type				
Watershed Area:		☐ I	☐ III	☐ V	☐ VII	☐ IX
Predicted Cross-section Area:						
Which regional curve used?	☐ Central / Southern ☐ Eastern AZ / New Mexico	☐ II	☐ IV	☐ VI	☐ VIII	☐ X

Measurements for Determining Stream Type

Measurement	Riffle Cross-section #1	Riffle Cross-section #2	Bankfull Indicators Used
Bankfull Width			☐ Top of point bars
Bankfull Max. Depth			☐ Change in particle size
Correction Factor			☐ Slope break
Bankfull Mean Depth			☐ Vegetation line
Cross-sectional Area			☐ Rock stains
Floodprone Width (2x BKF max depth)			☐ Undercut banks
STREAM TYPE =			☐ Presence of a floodplain at the elevation of incipient flooding

Notes/Comments

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Depositional Features				
Check off the feature that is most appropriate for the reach condition.				
☐ 1. Point Bars			☐ 5. Diagonal Bars	
☐ 2. Point Bars with Few Mid-Channel Bars			☐ 6. Main Channel Branching with Numerous Mid-Bars and Islands	
☐ 3. Numerous Mid-Channel Bars			☐ 7. Side Bars and Mid-Channel Bars with Length Exceeding 2 to 3 times Channel Width	
☐ 4. Side Bars			☐ 8. Delta Bars	☐ 9. NO bars
Illustrations from D. Rosgen, 1996. Applied River Morphology. Wildland Hydrology. Pagosa Springs, CO.				

Segment Habitat Quality				
Segment length equals 2 meander lengths or 20-30 times bankfull width of the stream. Use a minimum 300-feet reach to identify habitat types for large streams or rivers.				
Cobble	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Undercut banks	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Leaf packs	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Root masses	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Macrophyte beds	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Submerged logs / snares	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Sand dominated substrate	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Filamentous algae beds	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐

Reach Channel / Habitat Complexity		
Reach length equals 2 meander lengths or 20-30 times bankfull width. Use minimum of 300 foot reach to identify habitat types for large streams		
Habitat	Number of paces	Total
Pool		
Riffle		
Run		
Riffle / Run Ratio =		

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Site Sketch
Include location of riffles, pools, snags, submerged logs, undercut banks, areas of stable cobble habitat, type of bar formations, location and types of riparian vegetation, and areas with cut or eroding banks. Pace off length of eroding banks, length and width of riffles.

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Decision Criteria for Sampling Macroinvertebrates

The target stream habitat for collecting macroinvertebrates must be wadeable, perennial, contain riffle or run habitat, must contain heterogenous substrates, and must be sampled during the spring index period. Spring index period is April - May for warm water streams and May - June for cold water streams. Use the following specific specific decision criteria to determine whether to collect a macroinvertebrate sample. Circle the action taken regarding whether a sample was collected. Where you have found the stream conditions to be inappropriate for macroinvertebrate sampling, record a comment indicating the rationale for not collecting.

Parameter	Condition	Action to Take
Hydrologic Conditions	Baseflow conditions are occurring and it is approximately 4 or more weeks after a bankfull flow event. *	Collect macroinvertebrates
	A bankfull or greater magnitude flow event has occurred within 4 weeks of site visit. Or extreme high flow events have occurred resulting in deep scouring of the streambed and benthic community such that the macroinvertebrate community will not recover within the spring index period.	Do not collect macroinvertebrates
	Extended drought conditions have reduced flow from previously perennial condition to pools only or stagnant wetland habitat.	Do not collect macroinvertebrates
Substrate Type	A substrate consisting of a mixture of some of the following particle sizes is the target condition: boulder, cobble, gravel, sand, clay, silt, bedrock.	Collect macroinvertebrates
	Streams which have substrates dominated (consisting of >50% of that substrate type) by bedrock, travertine, or sand are considered non-target conditions.	Do not collect
Waterbody Type	The target waterbody type is a flowing stream with riffle or run (erosional) habitats present.	Collect macroinvertebrates
	We do not have methods developed for the following waterbody types and are not sampling them at this time: Effluent dependent streams, wetlands, ephemeral streams, lakes, seasonally intermittent streams.	Do not collect

Comments: (Indicate rationale for not collecting macroinvertebrate sample, if different from the above descriptions)

* Identification of bankfull and high flow elevation in the field: Using known watershed area, use appropriate Regional Curve and field bankfull indicators to estimate bankfull elevation. Look for debris lines and other high flow markers as an indicator of the most recent high flow stage. This procedure is explained in more detail and a copy of the regional curves is provided in the ADEQ Habitat Assessment Procedures (2005)

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Biological Sampling and Observations			
Types of Biological Samples and Sample Locations			
Macroinvertebrates	Riffle ☐ and riffle field split ☐; Pool ☐ and pool field split ☐; Edge ☐ and edge field split ☐		
Algae	Diatoms – riffle ☐/ pool ☐; artificial substrate ☐; filamentous riffle ☐; filamentous pool ☐; filamentous composite ☐		
Observations			
Filamentous Algae Covering Streambed throughout the reach	1) <1% 2) 1-25% 3) 26-50% 4) 51-75% 5) 76-100%		
Floating algae (detached clumps/mats) floating downstream	1) <1% 2) 1-25% 3) 26-50% 4) 51-75% 5) 76-100%		
Algal slime on rocks, wood, etc. (not filamentous)	Absent ☐; rare-thin coating ☐; common thick coating ☐		
Comments			
Macrophytes			
Macrophytes covering streambed throughout the reach	1) <1% 2) 1-25% 3) 26-50% 4) 51-75% 5) 76-100%		
Comments			
Identification of Algae (A) and Macrophytes (M)			
☐ A	Cladophora (hairlike feel, long beards)	☐ M	Watercress (Rorippa)
☐ A	Spirogyra (slimy to touch, bright green)	☐ M	Monkey flower (Mimulus, yellow flower)
☐ A	Nostoc (looks like jelly beans or round black to blue colored nodules)	☐ M	Pondweed (Potomogeton, submerged water grass)
☐ A	Blue-greens (blue-green to black in colo, e.g. Oscillatoria, Anabena)	☐ M	Columbine (yellow fower)
☐ M	Stoneworts (feels gritty, looks like a vascular plant, found in upwelling zones)	☐ M	White water buttercup (Ranunculus, white flower)
☐ M	Vaucheria (dark green felt-like mats)	☐ M	Eurasian water milfoil (Myriophyllum)
☐ M	Hydrodictyon (bright green, net forming algae)	☐ M	Hydrilla
☐ M	Praesiola (cold water algae, looks like sea lettuce)		

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Riffle Pebble Count

For transect method, tally 100-pebbles in riffle habitat only. Measure particles at equal increments across multiple transects within the wetted width of available riffle habitat throughout the reach.

Size Class	Size Range	Tally	Count	Percent	Cum. %
Silt/Clay *	<0.062				
Sand **	0.063 – 2.0				
Very Fine Gravel	3 – 4				
Fine Gravel	5 – 8				
Medium Gravel	9 – 16				
Coarse Gravel	17 – 32				
Very Course Gravel	33 – 64				
Small Cobble	65 – 96				
Medium Cobble	97 – 128				
Large Cobble	129 – 180				
Very Large Cobble	181 – 256				
Small Boulder	257 – 512				
Medium Boulder	513 – 1024				
Large Boulder	1025 – 2048				
Very Large Boulder	2049 – 4096				
Bedrock	>4097				
	Totals				

Comments:

% fines <2 mm	
# Size Classes	
D15	
D50	
D84	

Note: * Silt / clay particles feel slick when rubbing between thumb and forefinger.
** Sand Particles feel gritty when rubbing between thumb and forefinger.

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Riffle Embeddedness

Embeddedness measurements are collected concurrently with particle sizes within wetted width of 3 riffle transects. Count sand & fines as 100% embedded, bedrock & travertine as 100% embedded and gravel from a gravel patch as 100% embedded. Embeddedness is taken as a visual estimate. Keep a tally of embeddedness counts within each embeddedness category. Sum the counts and calculate a percentage for each embeddedness category. Then calculate weighted percent embeddedness as indicated. Take a sum of the weighted percents and divide by 100 for an average embeddedness value.

Embeddedness Category	Embeddedness Range (percent)	Tally	Count	Percent	Weighted Percent
Low	0 - 33				(% embed * 17)
Moderate	34 - 66				(% embed * 17)
High	67 - 100				(% embed * 17)
				Avg. % Embed	(sum weighted percents/100

Riffle Geometry

Riffle #	Length	Width	Length / Width ratio
1			
2			
3			
4			
		Average length / width ratio	

Notes

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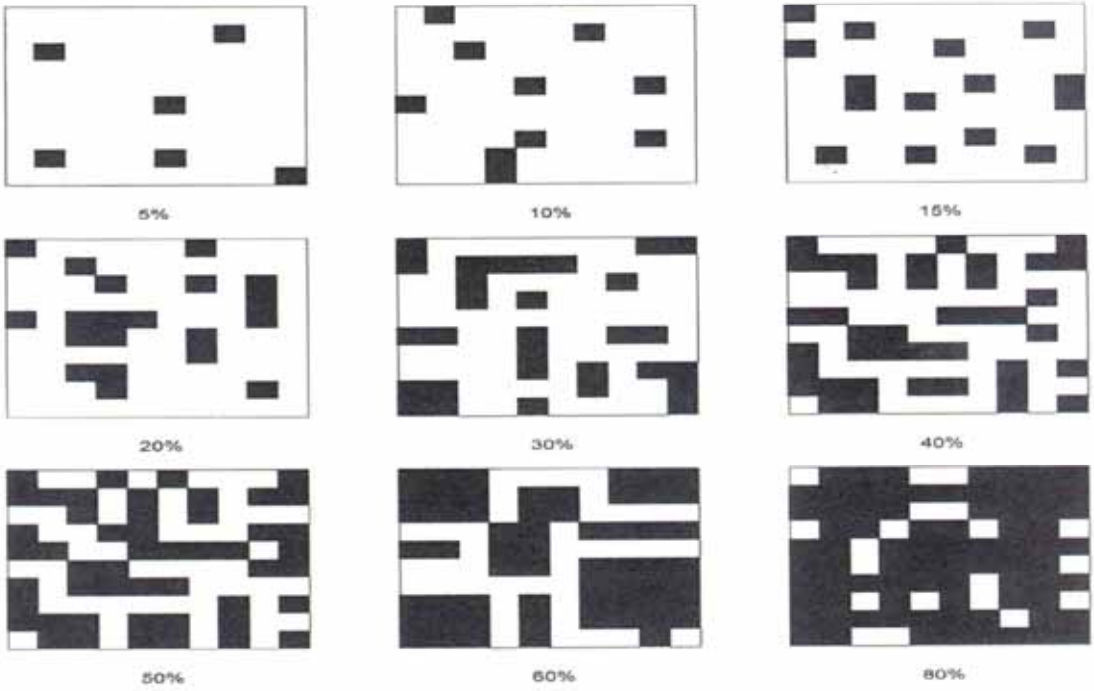
Riparian Vegetation Cover	
Record the percent cover of each vegetation type within the floodplain. Consider each vegetative layer separetly with a score of 0 – 100% for each. The object is to identify what vegetation type is holding the banks and floodplain together.	
Riparian Vegetation Cover	Estimated Percent Cover
Canopy of riparian trees > 15 feet high	
Understory of woody shrubs, saplings, herbs, grasses and forbs – 1.5 to 15 feet high	
Ground cover of woody shrubs, seedlings, herbs, and forbs - < 1.5 feet high	
Barren or bare dirt	



Riparian Association			
Place a check beside the most appropriate association using the riparian species list and elevation.			
☐	Sonoran riparian deciduous forest Cottonwood-Willow & Mesquite located at <3280' elevation	☐	Montane riparian deciduous forest mixed broadleaf species such as Big-tooth Maple; Narrowleaf Cottonwood; Box-elder; SW Choke Cherry; Arizona Alder; Pacific, Coyote, Red, or Bebb's Willow; located at 5740' – 8200' elevation
☐	Interior riparian deciduous forest Cottonwood-Willow & mixed broadleaf species such as Sycamore, Ash, Walnut, Alder, Soapberrry, and Hackberry located at 3280' – 5740' elevation	☐	Actic – Boreal forest Distinctive riparian communities are not present however there are some indicator species such as shrubby Scouler and Bebb's willows, Red Elderberry, Shrubby Cinquefoil, Goose-berry Currant, Raspberry, and Thin-leaf Alder located along streams of subalpine forests and meadows at >8200'.

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Riparian Species											
☐	Alder, Thinleaf	☐	Cinquefoil	☐	Hawthorn, River	☐	Tamarisk	☐	Willow, Peachleaf	☐	Equisetum
☐	Alder, Arizona	☐	Cottonwood, Fremont	☐	Maple, Big-tooth	☐	Walnut, Arizona	☐	Willow, Red	☐	Monkeyflower
☐	Ash, Lowell	☐	Cottonwood, Lanceleaf	☐	Maple, Rocky Mountain	☐	Willow, Arizona	☐	Willow, Scouler	☐	Phragmites
☐	Ash, velvet	☐	Cottonwood, Narrowleaf	☐	Mesquite	☐	Willow, Arroyo	☐	Willow, Seep	☐	Rushes
☐	Birch	☐	Elder, Blueberry	☐	Netleaf Hackberry	☐	Willow, Bebb	☐	Willow, Yewleaf	☐	Sedges
☐	Boxelder	☐	Elder, Mexican	☐	New Mexican Locust	☐	Willow, Bonpland	☐	Willow, Unknown	☐	Sacaton
☐	Buckthorn, Birchleaf	☐	Elderberry	☐	Raspberry	☐	Willow, Coyote	☐	Bamboo	☐	
☐	Buckthorn, California	☐	Elderberry, Desert	☐	Red-osier Dogwood	☐	Willow, Desert	☐	Carex	☐	
☐	Burro bush	☐	Gooseberry	☐	Soapberry	☐	Willow, Gooding	☐	Cattail	☐	
☐	Chokecherry	☐	Hawthorn, Cerro	☐	Sycamore, Arizona	☐	Willow, Pacific	☐	Deer Grass	☐	

Measuring Canopy Density			
	Number of Points Intercepted Along the Transect		
Position	Upper Reach	Mid-Reach	Lower Reach
Right Edge Water			
Middle – Looking Upstream			
Middle – Looking Downstream			
Left Edge Water			
Sum			

Mean Number of Points = Sum of the three columns _____ ÷ 3 =

If stream order <5	Percent Canopy Density = Mean Number of Points • 1.5 =	%
If stream order >5	Percent Canopy Density = Mean Number of Points • 0.75 =	%

Regeneration Potential of Riparian Trees					
	Species, in order of dominance	Mature Trees >16" @ 3 ft height	Young Trees –1 ¼" @ 3 ft. height	Saplings < 1 ¼"	Seedlings New growth
1					
2					
3					
4					
5					

Age Classes of Riparian Tree Species

(Classify according to species present, not just the dominant tree type of that plant association)

☐	Species abundant in 3 age classes	☐	Abundant in 2 age classes	☐	One age class present	☐	No regeneration evident, few mature trees present, no saplings or seedlings, or if present, they are heavily grazed

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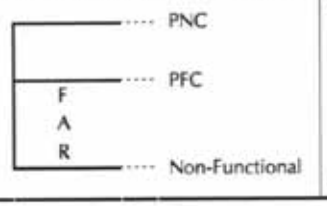
Proper Functioning Condition Worksheet			
Yes	No	N/A	Hydrologic
			1) Flood plain inundated in "relatively frequent" events (1-3 years)
			2) Active/stable beaver dams
			3) Sinuosity, width/depth ratio, and gradient are in balance with the landscape setting (i.e., landform, geology, and bioclimatic region)
			4) Riparian zone is widening or has achieved potential extent.
			5) Upland watershed not contributing to riparian degradation
			Vegetative
			6) Diverse (3) age structure of vegetation (Recruitment for maintenance/recovery)
			7) Diverse composition of vegetation (For maintenance/recovery)
			8) Species present indicate maintenance of riparian soil moisture characteristics
			9) Streambank vegetation is comprised of those plants or plant communities that have root masses capable of withstanding high streamflow events
			10) Riparian plants exhibit high vigor
			11) Adequate vegetative cover present to protect banks and dissipate energy during high flows
			12) Plant communities in the riparian area are an adequate source of coarse and/or large woody debris
			Erosion Deposition
			13) Flood plain and channel characteristics (i.e., rocks, coarse and/or large woody debris) adequate to dissipate energy
			14) Point bars are revegetating
			15) Lateral stream movement is associated with natural sinuosity
			16) System is vertically stable
			17) Stream is in balance with the water and sediment being supplied by the watershed (i.e., no excessive erosion or deposition)

Functional Rating

☐	Proper Functioning Condition	☐	Functional at risk, downward trend
☐	Functional at risk, upward trend	☐	Non-Functional
☐	Functional at risk, no apparent trend	☐	Unknown



PFC Remarks

Use reverse side for additional comments.

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GUIDELINES FOR COMPLETING THE PFC CHECKLIST

General guidance: If 75% or more of stream reach is PFC, classify entire reach as PFC. All "No" answers must have comments in notes section. Answers can go on the line between "Yes" and "No", but consider it a "No" and comment in notes section.

Q1. Instantaneous peak flows don't count. Inundation means to bankfull depth. Bankfull can be identified from top of the point bars, changes in vegetation, topographic break in slope, change in size of bank materials, evidence of an inundation feature such as small benches, exposed root hairs below an intact soil layer indicating exposure to erosive flow, and bank undercuts. "No" if channelization or entrenchment. "N/A" if a "V"-canyon without floodplain development.

Q2. Usually "N/A", but may be applicable at high altitude sites; also, consider the present environment (could they be present).

Q3. Based on the stream type expected & the regional curves, all three features must be present for a "YES". Use bankfull width, not wetted width. "NO" if straightness, excessive sediment, or entrenched channel.

Q4. Widening can mean encroaching on the channel as well as moving toward the terraces. The age of the vegetation in an indicator. "NO" if upland species encroaching on the floodplain or Kentucky bluegrass present. "YES" if recruitment of wetland/riparian species on new landforms. "N/A" if an A1 stream type.

Q5. Need to look at upland ground cover and erosion signs (e.g. plants on pedestals, debris dams around plants, rills, gullies). "NO" if side channel and mid-channel bars, gullies, fan shaped deposits from tributaries, braided channels, overloading of point bars, or cementing of streambed.

Q6. "YES" if 3 age classes (mature, young, saplings) present for a single species, or young and sapling classes if recruitment & replacement is occurring, or dense matting of herbaceous riparian/wetland plants. "NO" if individual plants. "N/A" if A1 Stream Type.

Q7. Maintenance means recruitment. Is it occurring? "YES" if several different species present (e.g. willows, rushes, sedges). It depends on the elevation and the potential natural community that might be present if all constraints are removed. In some environments, 2 species could be a "YES". Usually "NO" if 1 species present.

Q8. "YES" if sedges, rushes, willows, seep willows, alders, cottonwoods, etc. Don't consider quantity. Do you see any at all?

Q9. A high stream flow event is one that occurs once in 25-30 years. Q9 is similar to Q*, but you are now looking for quantity. "NO" if presence of upland species. "YES" if willows, alder, aspen, birch, cottonwood, sedge, rush, bulrush, and wetland grasses.

Q10. Are the plants healthy and dense? "NO" if yellow leaves, stunted plants, many dead stems and branches, a thin crown, infested with insects, diseased, or grazed down by browsers.

Q11. This is a quantity question. Use 80% cover as a guide. Look for riparian plants, herbaceous cover, salt cedar (tamarisk), seep willows, etc. "NO" if "NO" on Q9. If Q6-Q10 are "NO", this is probably a "NO".

Q12. "N/A" for meadows, desert streams, and probably intermediate elevation streams, or sedge/grass community streams. "YES" if fallen trees. For some locations consider living and dead trees and trees along banks out of the water.

Q13. "YES" if large boulders, roughness of the floodplain, large trees & dense vegetation along stream banks. "NO" if incision & no access of stream to floodplain.

Q14. "YES" if sedge/rush components. Consider potential, height and newness of the point bar. Sandy soils don't hold water well and there may be no potential for revegetation. A1 Stream Type is "N/A".

Q15. "NO" if straight channel, not confined geologically, and channel movement with every high flow event. "YES" if single channel, stable banks (especially on straight segments), & natural deposition.

Q16. "NO" if entrenchment, down cutting (some is natural), excessive aggradation, unstable vertical banks. "YES" if streambed is armored with large rock, bedrock, heavy gravel. Don't consider old down cutting. If a bedrock stream then "N/A".

Q17. "NO" if excessive sediment from side drainages, excessive aggradation, mid-channel bars, braiding, or unstable banks. "NO" if Q5 is "NO".

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PFC No Response Sheet – Comments and Notes

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Habitat Assessment Field Data Sheet				
Habitat Parameter	Condition Category			
	Optimal	Sub-optimal	Marginal	Poor
Habitat Quality	Large variety of habitats available for colonization which may include cobble, undercut banks, snags, submerged logs, leaf packs, root masses, macrophyte beds or other organic material.	Moderate variety of habitats which may include cobble, leaf packs, root masses, macrophyte beds or other organic material.	Habitat has minimal variety, substrate dominated by one particle size, may have some cobble, macrophyte beds, or algae beds.	Homogeneous substrate dominated by sand, shallow with uniform velocity, no shade on riffles, may have extensive filamentous algae beds.
Score ⇨	4	3	2	1
Extent of Riffle Habitat	Well developed riffle that is as wide as stream and its length extends 2x the wetted width of the stream.	Riffle is as wide as stream, but is less than 2x stream width; abundance of cobble; boulders and gravel are common.	Reduced riffle area does not extend across entire cross-section and is less than 2x width; gravel or large boulders and bedrock prevalent; cobble present.	Riffles virtually non-existent; sand, gravel, large boulders or bedrock prevalent; cobble lacking.
Score ⇨	4	3	2	1
Embeddedness of Riffles	Gravel, cobble, and boulder particles are 0-25% surrounded by fine sediment (bedrock is 0% embedded).	Gravel, cobble, and boulder particles are 25-50% surrounded by fine sediment.	Gravel, cobble, and boulder particles are 50-75% surrounded by fine sediment.	Gravel, cobble, and boulder particles are more than 75% surrounded by fine sediment (sand is 100% embedded).
Score ⇨	4	3	2	1
Sediment Depostion	Point bars in C type channel maintained, no mid-channel or side bars. No bimodal particle size distribution. No excess sediment in riffles and pools of A, B, or C type channels.	Point bars with few mid-channel bars or side bars in C type channels. No bimodal particle size distribution. Some filling in of pools in A, B, and C type channels.	Numerous mid-channel or diagonal bars in C type channels. Some loss of pool and riffle habitat in A, B, and C type channels. Bimodal distribution may be present with excess fines in the substrate.	Branched or braided C channel with numerous mid-channel bars and islands, some exceeding 2-3x channe width in length. Heavy deposites of finae material evident with bimodal particle distribution. Pools and riffles filled in, with run habitat dominating.
Score ⇨	4	3	2	1
Bank Stability within the active bankfull channel (score each bank)	Banks stable; no evidence of erosion or bank failure; <5% of bank affected.	Banks moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion.	Banks moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; 60-100% of bank has erosional scars.
Score_____ Left Bank ⇨	2	1.5	2	0.5
Score_____ Right Bank ⇨	2	1.5	2	.05
Sum of Habitat Category Scores_____ ⇨		Rating Category		
		0 – 7	8 – 14	15 - 20
		☐ Very Impaired	☐ Impaired	☐ Good Condition

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NON-POINT SOURCE CODES

Circle sources directly impacting the site, asterisk sources located in the watershed. Source Group is bolded, Category Code is italicized, and Sub-category Code is regular style font.

Code	Source Category	Code	Source Category	Code	Source Category
1000	**Agriculture** *(Agriculture)*	7350	Upstream impoundment	6600	Hazardous waste storage/disposal
1050	Crop-related sources	7400	Flow regulation/Modification/Diversions	8000	Highway salt storage/use
1100	Non-irrigated crop production	7550	Habitat Modification	8200	Storage tank leaks
1200	Irrigated crop production	7555	Erosion materials from tributaries	8250	Underground storage tank leaks
1300	Specialty crop production	7600	Removal of riparian vegetation	8275	Above ground storage tank
1350	Grazing-related sources	7700	Streambank modification or destabilization	**0100**	**Wastewater** *(Industrial Point Source)*
1400	Pasture grazing - riparian and/or upland	7750	Highway/Road/Bridge-erosion or aggradation	0110	Major industrial point source
1410	Pasture grazing - riparian	7800	Drainage/Filling of wetlands	0120	Minor industrial point source
1420	Pasture grazing - upland	7900	*Marinas and recreational boating*	0200	Municipal point source
1500	Range grazing - riparian and/or upland	7910	Boating with in-water releases	0210	Major municipal point source
1510	Range grazing - riparian	7920	Boating with on-land releases	0220	Minor municipal point source
1520	Range grazing - upland	**5000**	**Mining** *(Resource extraction)*	0230	Package plants (small flows)
1600	*Intensive Animal feeding Operations*	5075	Active Mining operation	*0300*	*Other Wastewater*
1620	Concentrated Animal Feeding Operations point source/permitted)	5100	Surface Mining	0400	Combined system (sewage and stormwater)
1640	Confined animal feeding operations (non-point source	5150	Sand and gravel operations	0500	Collection system failure
1700	*Aquiculture/Fish Hatchery*	5200	Subsurface mining	0900	Sewage lagoons
2000	**Forestry** *(Silviculture)*	5300	Placer mining	0975	Reuse (Effluent to lakes, golf courses, artificial
2100	Harvesting, restoration (residue management)	5400	Dredge mining	6500	Septic systems
2200	Forest management (fertilization, pesticide use)	5500	Petroleum activities	6700	Septage disposal (e.g. from septic tank trucks)
2300	Logging roads	5600	Mill tailings	**8100**	**Other** (Atmospheric deposition)
2500	Clear cutting	5650	Mill or mine tailings	8400	Spills
8610	Wildfires or controlled burns	5700	Mine tailings	8500	Contaminated sediments
3000	**Hydro/Habitat Modification/Runoff** *(Construction)*	5800	Acid mine drainage	8510	
3100	Highway/Road/Bridge construction	5900	Abandoned mining operation	8530	
3200	Land development/Land clearing	5950	Inactive mining operation	8540	
4000	*Urban runoff/Stormwater sewers*	**8700**	**Recreation** (non-boating)	8600	
4100	Non-industrial (NPDES) stormwater runoff	8710	Golf courses	8910	
4200	Industrial (NPDES) stormwater runoff	8720	Camping/Campground recreation	**Other Non-point Source Observations at the site or within the reach**	
4300	Other urban runoff	8730	All terrain vehicles/Off road vehicles/Biking		
4400	Illicit connections to stormwater sewers (dry weather flows)	**6000**	**Storage and Disposal** (Land disposal/Storage)		
4500	Urban Highway/Road/Bridge runoff	6100	Sludge disposal/storage		
4600	*Non-urban runoff/Erosion and sedimentation*	6300	Landfills		
8300	Non-urban (highway/Road/Bridge Runoff/Maintenance)	6350	Inappropriate waste disposal/Wildcat dumping		
7000	*Hydrological modifications*	6400	Industrial land treatment		

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1.14 Laboratory Sample Submittal Forms

ADEQ has contracts with several laboratories for sample analyses. Water samples constitute the majority of samples required for analysis and these are primarily submitted to the Arizona Department of Health Services State Laboratory Services. Two forms are necessary to submit water samples to the State Laboratory and they are presented on the following pages: Request for Chemical Analyses Form and Sample Continuation Form.

Other laboratory have there own unique sample submittal form. They are not covered in this manual because of the frequency of interchange of contract laboratories.

1.14.1 Completing the State Request for Chemical Analyses Form

On the Request for Chemical Analyses Form (Figure4), the box in the top left corner must be filled out in its entirety. The four codes (Tracking #, PO#, PCA, and Index) in the "ADEQ Codes Only" box must also be completed. The "Site Code" is only used when a single site has been sampled. For multiple sites, the Sample Continuation Form must be used (Section 1.14.2). An example of the "Program Name" would be "FSN".

All samples submitted to any laboratory are required to have a chain of custody. "Sample Matrix" for the FSN program is "Surface Water." The Priority for the FSN Program is typically "Third Priority and Routine Surveillance".

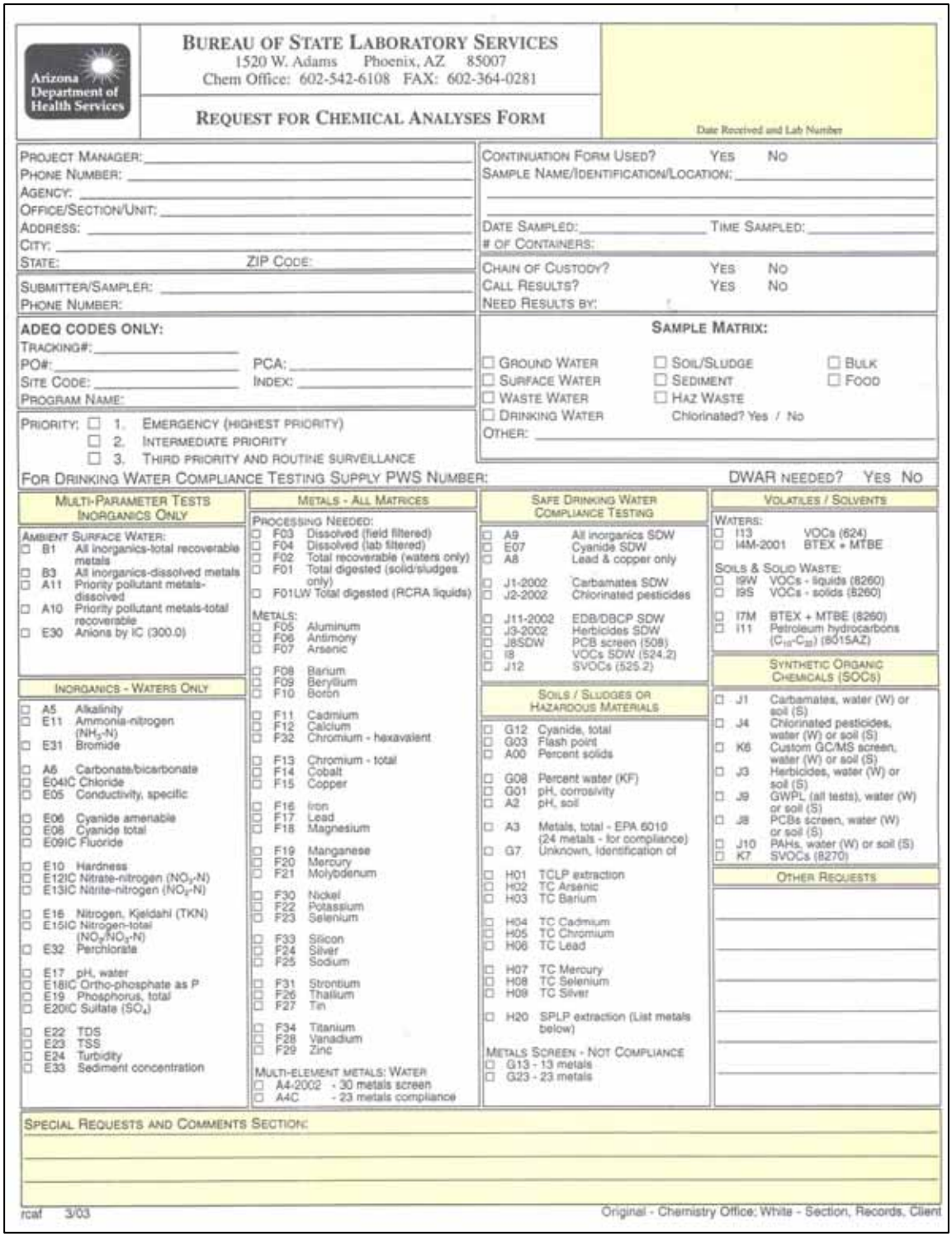
In the bottom portion of the form are the analyses that may be requested and any special requests or comments. For the FSN program, the required analyses information can be found in the "Surface Water Monitoring Sample Plan" published each year by the Hydrologic Support and Assessment Section, Surface Water Monitoring and Standards Unit.

Typically the FSN Program submits one bottle samples for inorganics, suspended sediment concentration, total metals, and dissolved metals. The request for analyses of dissolved metals requires a completed second Request for Chemical Analyses Form and Continuation Form.

1.14.2 Completing the State Laboratory Request for Chemical Analyses Sample Continuation Form

When multiple sites have been sampled, the Continuation Form (Figure5) must accompany the Request for Chemical Analyses Form**.** Sections on the top portion of the form that must be completed are**:** Sampler's Identification / Description, Date Sampled, Time Sampled, and Number of Containers. On the bottom portion, "Chain of Custody Needed?" must have a check mark in the "Yes" box. The Chain of Custody Record, on the bottom portion, must be completed for submittal to the State Laboratory Receiving Office.

Figure 4. Arizona Department of Health Services State Laboratory Services Request for Chemical Analyses Form.



Arizona Department of Health Services

BUREAU OF STATE LABORATORY SERVICES
1520 W. Adams Phoenix, AZ 85007
Chem Office: 602-542-6108 FAX: 602-364-0281

REQUEST FOR CHEMICAL ANALYSES FORM

Date Received and Lab Number

PROJECT MANAGER:
PHONE NUMBER:
AGENCY:
OFFICE/SECTION/UNIT:
ADDRESS:
CITY:
STATE: ZIP CODE:

SUBMITTER/SAMPLER:
PHONE NUMBER:

CONTINUATION FORM USED? YES NO
SAMPLE NAME/IDENTIFICATION/LOCATION:
DATE SAMPLED: TIME SAMPLED:
OF CONTAINERS:

CHAIN OF CUSTODY? YES NO
CALL RESULTS? YES NO
NEED RESULTS BY:

ADEQ CODES ONLY:
TRACKING#:
PO#: PCA:
SITE CODE: INDEX:
PROGRAM NAME:

SAMPLE MATRIX:
☐ GROUND WATER ☐ SOIL/SLUDGE ☐ BULK
☐ SURFACE WATER ☐ SEDIMENT ☐ FOOD
☐ WASTE WATER ☐ HAZ WASTE
☐ DRINKING WATER Chlorinated? Yes / No
OTHER:

PRIORITY: ☐ 1. EMERGENCY (HIGHEST PRIORITY)
☐ 2. INTERMEDIATE PRIORITY
☐ 3. THIRD PRIORITY AND ROUTINE SURVEILLANCE

FOR DRINKING WATER COMPLIANCE TESTING SUPPLY PWS NUMBER: DWAR NEEDED? YES NO

MULTI-PARAMETER TESTS INORGANICS ONLY

AMBIENT SURFACE WATER:
☐ B1 All inorganics-total recoverable metals
☐ B3 All inorganics-dissolved metals
☐ A11 Priority pollutant metals-dissolved
☐ A10 Priority pollutant metals-total recoverable
☐ E30 Anions by IC (300.0)

INORGANICS - WATERS ONLY

☐ A5 Alkalinity
☐ E11 Ammonia-nitrogen (NH_3-N)
☐ E31 Bromide
☐ A6 Carbonate/bicarbonate
☐ E04IC Chloride
☐ E05 Conductivity, specific
☐ E06 Cyanide amenable
☐ E08 Cyanide total
☐ E09IC Fluoride
☐ E10 Hardness
☐ E12IC Nitrate-nitrogen (NO_3-N)
☐ E13IC Nitrite-nitrogen (NO_2-N)
☐ E16 Nitrogen, Kjeldahl (TKN)
☐ E15IC Nitrogen-total (NO_2/NO_3-N)
☐ E32 Perchlorate
☐ E17 pH, water
☐ E18IC Ortho-phosphate as P
☐ E19 Phosphorus, total
☐ E20IC Sulfate (SO_4)
☐ E22 TDS
☐ E23 TSS
☐ E24 Turbidity
☐ E33 Sediment concentration

METALS - ALL MATRICES

PROCESSING NEEDED:
☐ F03 Dissolved (field filtered)
☐ F04 Dissolved (lab filtered)
☐ F02 Total recoverable (waters only)
☐ F01 Total digested (solid/sludges only)
☐ F01LW Total digested (RCRA liquids)

METALS:
☐ F05 Aluminum
☐ F06 Antimony
☐ F07 Arsenic
☐ F08 Barium
☐ F09 Beryllium
☐ F10 Boron
☐ F11 Cadmium
☐ F12 Calcium
☐ F32 Chromium - hexavalent
☐ F13 Chromium - total
☐ F14 Cobalt
☐ F15 Copper
☐ F16 Iron
☐ F17 Lead
☐ F18 Magnesium
☐ F19 Manganese
☐ F20 Mercury
☐ F21 Molybdenum
☐ F30 Nickel
☐ F22 Potassium
☐ F23 Selenium
☐ F33 Silicon
☐ F24 Silver
☐ F25 Sodium
☐ F31 Strontium
☐ F26 Thallium
☐ F27 Tin
☐ F34 Titanium
☐ F28 Vanadium
☐ F29 Zinc

MULTI-ELEMENT METALS: WATER
☐ A4-2002 - 30 metals screen
☐ A4C - 23 metals compliance

SAFE DRINKING WATER COMPLIANCE TESTING

☐ A9 All inorganics SDW
☐ E07 Cyanide SDW
☐ A8 Lead & copper only
☐ J1-2002 Carbamates SDW
☐ J2-2002 Chlorinated pesticides
☐ J11-2002 EDB/DBCP SDW
☐ J3-2002 Herbicides SDW
☐ J8SDW PCB screen (508)
☐ I8 VOCs SDW (524.2)
☐ J12 SVOCs (525.2)

SOILS / SLUDGES OR HAZARDOUS MATERIALS

☐ G12 Cyanide, total
☐ G03 Flash point
☐ A00 Percent solids
☐ G08 Percent water (KF)
☐ G01 pH, corrosivity
☐ A2 pH, soil
☐ A3 Metals, total - EPA 6010 (24 metals - for compliance)
☐ G7 Unknown, Identification of
☐ H01 TCLP extraction
☐ H02 TC Arsenic
☐ H03 TC Barium
☐ H04 TC Cadmium
☐ H05 TC Chromium
☐ H06 TC Lead
☐ H07 TC Mercury
☐ H08 TC Selenium
☐ H09 TC Silver
☐ H20 SPLP extraction (List metals below)

METALS SCREEN - NOT COMPLIANCE
☐ G13 - 13 metals
☐ G23 - 23 metals

VOLATILES / SOLVENTS

WATERS:
☐ I13 VOCs (624)
☐ I4M-2001 BTEX + MTBE

SOILS & SOLID WASTE:
☐ I9W VOCs - liquids (8260)
☐ I9S VOCs - solids (8260)
☐ I7M BTEX + MTBE (8260)
☐ I11 Petroleum hydrocarbons (C_{10}-C_{28}) (8015AZ)

SYNTHETIC ORGANIC CHEMICALS (SOCs)

☐ J1 Carbamates, water (W) or soil (S)
☐ J4 Chlorinated pesticides, water (W) or soil (S)
☐ K6 Custom GC/MS screen, water (W) or soil (S)
☐ J3 Herbicides, water (W) or soil (S)
☐ J9 GWPL (all tests), water (W) or soil (S)
☐ J8 PCBs screen, water (W) or soil (S)
☐ J10 PAHs, water (W) or soil (S)
☐ K7 SVOCs (8270)

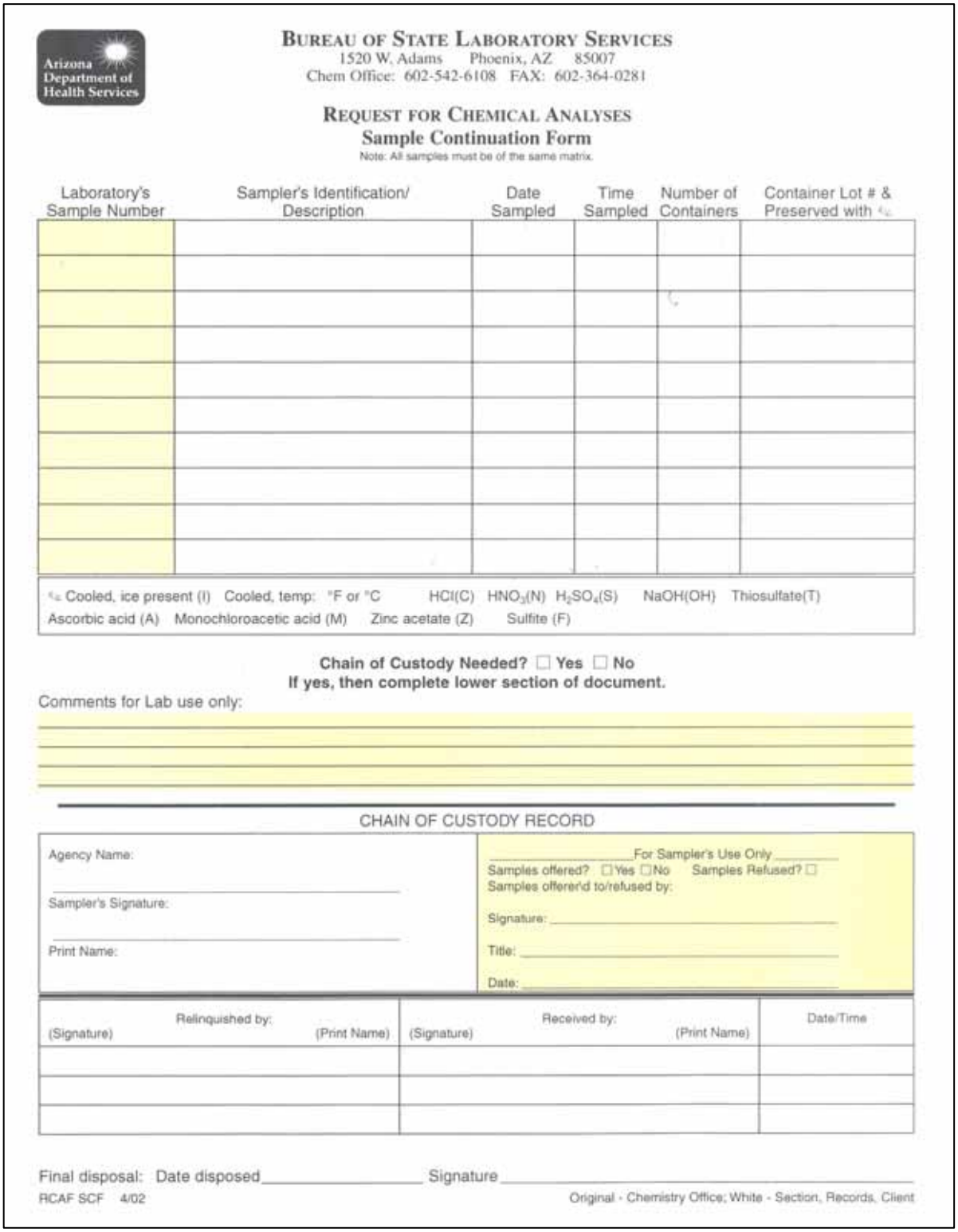
OTHER REQUESTS

SPECIAL REQUESTS AND COMMENTS SECTION:

rcaf 3/03

Original - Chemistry Office; White - Section, Records, Client

Figure 5. Arizona Department of Health Services State Laboratory Services Request for Chemical Analyses Continuation From.



Arizona Department of Health Services

BUREAU OF STATE LABORATORY SERVICES
1520 W. Adams Phoenix, AZ 85007
Chem Office: 602-542-6108 FAX: 602-364-0281

REQUEST FOR CHEMICAL ANALYSES
Sample Continuation Form
Note: All samples must be of the same matrix.

Laboratory's Sample Number	Sampler's Identification/ Description	Date Sampled	Time Sampled	Number of Containers	Container Lot # & Preserved with [1]

[1] Cooled, ice present (I) Cooled, temp: °F or °C HCl(C) HNO_3(N) H_2SO_4(S) NaOH(OH) Thiosulfate(T)
Ascorbic acid (A) Monochloroacetic acid (M) Zinc acetate (Z) Sulfite (F)

Chain of Custody Needed? ☐ Yes ☐ No
If yes, then complete lower section of document.

Comments for Lab use only:

CHAIN OF CUSTODY RECORD

Agency Name:

Sampler's Signature:

Print Name:

For Sampler's Use Only
Samples offered? ☐Yes ☐No Samples Refused? ☐
Samples offerer'd to/refused by:
Signature:
Title:
Date:

Relinquished by: (Signature)	(Print Name)	Received by: (Signature)	(Print Name)	Date/Time

Final disposal: Date disposed Signature

RCAF SCF 4/02 Original - Chemistry Office; White - Section, Records, Client

SECTION 2

EQUIPMENT CALIBRATION AND CLEANING PROCEDURES

TABLE OF CONTENTS

2.00 Equipment Calibration Procedures

2.10 Hydrolab® Multimeter

2.10.1 General Care of the Hydrolab

Hydrolabs or other multimeter instruments must be transported in containers that prevent damage to the equipment. When transporting the H20 Sonde, Scout read-out units, stirrers, cables, etc. (Figure 1), insert them in their soft padded cases. The Minisonde, Surveyor 4 read-out units, etc. (Figure 2) must be transported and stored in the hard plastic Pelican cases. If the units are stored in plastic bags, insure that the bags are anti-static bags. Static electricity generated from normal bags can cause damage the internal electronics of the units.

Figure 1. H2O Sonde and associated equipment.

Figure 2. Scout 4 Sonde and associated equipment.

Proper care of the probes is essential for accurate readings. Keep probes cleared of organic matter such as algal filaments and sand. An occasional pre-calibration cleaning and rinse with isopropyl alcohol and a very soft brush or cotton ball is recommended. Repeated use of the meter can accumulate iron filings on the magnetic dissolved oxygen stirrer. Regular cleaning will keep the build-up from becoming an operational problem. Figure 3 shows the location of the sensors on the H20 sonde units and Figure 4 shows the sensor locations on the Minisonde units.

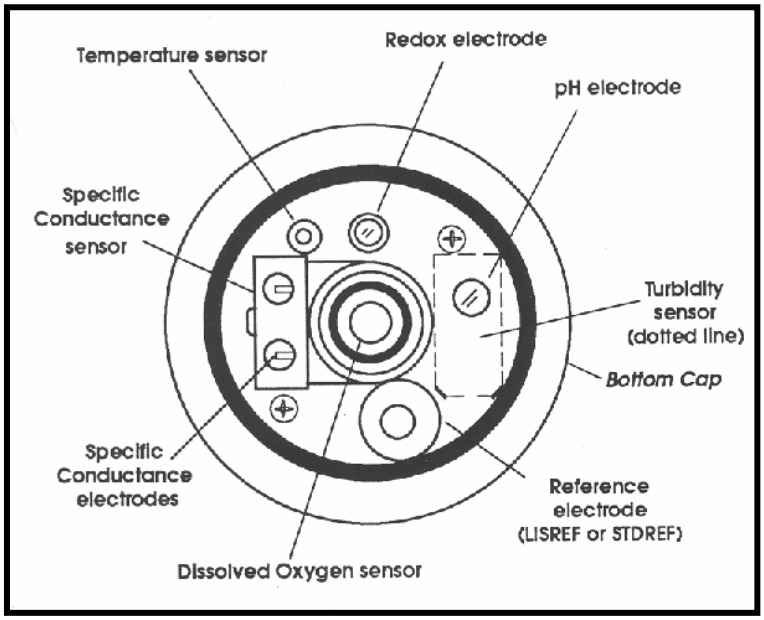


Figure 3. H2O Sonde sensor placement.

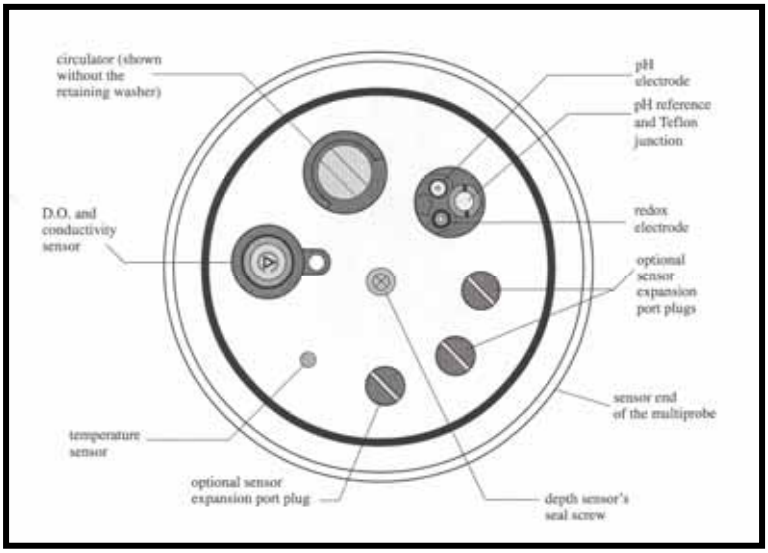


Figure 4. Minisonde sensor placement.

Maintain about 1/8 to 1/4 inch of tap water in the probe protective cup at all times; do not let the probes dry out; they will be permanently damaged and will have to be replaced. Never store deionized water in the probe protective cup. Probes should not be completely submerged in water.

Keep the connectors on the cable, probe and transmitter clear of debris. Do not let the cable ends fall into the dirt or water. Keep protector caps on the sensor connectors and cable connectors whenever they are not in use. Cleaning the connectors with isopropyl alcohol and a soft brush before taking the unit into the field should remove any dirt or moisture that may remain from previous use.

Avoid dropping the unit; it is rugged but there is an internal battery and electronics which can be damaged by shock.

Always check the battery reading before taking the unit into the field. The Scout 2 read-out units will normally show an internal battery level of about 16 volts with new batteries installed, and will usually function efficiently until the batteries reach about 9 volts or less. Keep spare batteries (10 AA batteries) and a phillips head screwdriver with the unit. If the unit is being stored for extended periods of time it is recommended that the AA batteries be removed and stored separately to prevent possible damage from battery leakage. The Surveyor 4 read-out units are equipped with a rechargeable nickel metal hydride battery. The fully charged battery holds a charge of 8.5 volts and should be recharged when the level reaches 6.5 volts. Nickel metal hydride batteries can be charged to full voltage at any time, no matter how low the voltage.

Turn the unit off immediately after use to preserve the battery charge. The stirrer unit is the largest draw on the battery, so use it sparingly. Some models will allow the observer to disable the stirrer when not needed. Stirrer should be turned on when flow velocity is less than 1 ft./sec.

Carefully coil the cable after use into a 6” to 12" diameter coil. Avoid damage to the cable by not stepping on the cable or allowing it to become kinked. If the pin connector to the probe or stirrer becomes difficult to slip on, lubricate the connector sleeve with the clear petroleum lubricant provided in the maintenance kit. The male connector pins should occasionally be cleaned with a weak solvent and a soft brush. After cleaning, always apply a thin coating of petroleum lubricant.

Protect units from temperatures greater than 122° F (50 °C). Units will automatically shut down at this temperature. The read-out units utilize a liquid crystal display and very cold or very hot temperatures will adversely affect the display read-out; therefore, bring it inside when these extreme temperatures are present.

2.10.2 Calibration Protocols

All equipment should be calibrated before and after each use in the field and recorded on the lab calibration form which is kept in a file specifically for the model number. This allows the sampler to determine the accuracy of the field parameters taken at the site and makes sure the instrument is ready for the next user. Some individuals may chose to calibrate at each site depending on the requirements of their sample plan. Each piece of equipment should have a calibration log-book that is kept in the case with the unit. Each time the instrument is pre-calibrated and post -calibrated, the results should be noted in the log-book along with the name of the user and the intended use of the equipment. Any problems with the unit should also be noted. As a general rule, the temperature and other parameter readings should be monitored for approximately a minute to ensure that the values stabilize before the readings are recorded in the unit's log book. There may be some drift between the two calibration values, depending on the amount of elapsed time between the pre-calibration and post-calibration. For guidance on the allowable percent differences between pre-calibration and post-calibration, refer to the appropriate Hydrolab® model operating manual.

This protocol covers calibration procedures for specific electrical conductance, pH, dissolved oxygen (DO), and turbidity. Other parameters are calibrated at the factory. For additional information about calibration, consult the Hydrolab® operating manual.

Maintenance work on the units is normally done on a quarterly basis by trained personnel and is covered in Section 4.10. Equipment that is not being regularly used should be maintained every six months. Prepare the unit by attaching the cable and transmitter. Some units use a calibration cup that is essentially a 250 milliliter Nalgene bottle with the bottom removed. A soft rubber cap covers the un-threaded end of the cup. The opposite end is threaded and has a white plastic cover. The cup threads onto the unit, covering the probes. The rubber cap can be easily removed to allow the calibration solutions to be added when needed. The Minisonde units utilize a calibration and storage cup with threaded cap. It can be used for storage of the unit and for calibration purposes. During the calibration procedure, the probes will need to be placed in a position that has them facing upward. Be sure to have a lab stand and the proper clamps in place to hold the unit securely while performing the calibrations (Figure 5).

Figure 5. Typical lab stand for holding the Hydrolab Sonde.

When calibrating the units, use the function and cursor keys on the Scout read-out units (Figure 6) and the Surveyor 4 read-out units (Figure 7) to navigate through the procedure.

If while calibrating the probes a message is displayed on the read-out unit that states "calibration out of range," there are problems with the device that need to be rectified. Try running through the calibration procedure again to see if the problem persists. If the problem occurs again, it may

be that the probe in question needs the reference solution replaced. Use fresh reference solution that is still within the expiration period and replace the probes old solution. If the probe in question will still not calibrate, make notes in the calibration book regarding the problem and have it shipped back to the manufacturer for repair. Do not put the faulty unit back into the cabinet.

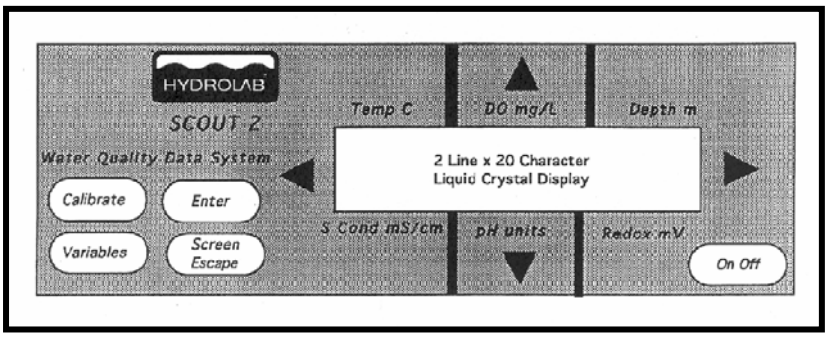


Figure 6. Scout read-out unit.

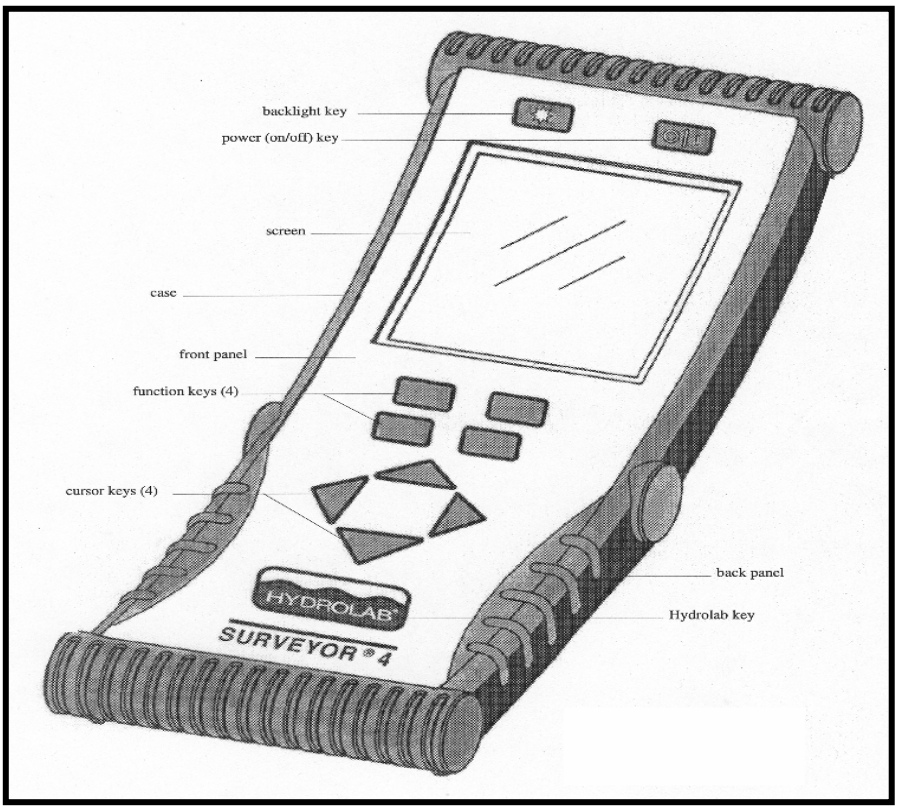


Figure 7. Surveyor 4 read-out unit.

2.10.2.1 Specific Electrical Conductance Calibration

There are normally three conductivity standards found in the lab area, 0.1 M, 0.01 M and 0.001 M potassium chloride (KCl) solutions. The State lab supplies ADEQ with KCl solutions that have been prepared, tested and assigned an acceptable range and usually a tested lab value. These three standards are typically sufficient for most conditions encountered in the field. If a

site has unusually high conductivity readings, it may be necessary to have the lab prepare a standard that more closely resembles the site conditions. The ranges and values assigned to the standards are in most cases given in micro Siemens per centimeter (µS/cm). When calibrating with the KCl standards, insure that the solutions have not exceeded the expiration date. Standard solutions can be disposed of in the lab sink, using tap water to dilute and rinse equipment.

1. Rinse the sensors three times with a half full calibration cup of deionized water to clear the probes of contaminants.

2. If a Minisonde unit is being pre-calibrated, obtain a paper towel or other non-abrasive absorbent material and dry the electrodes of the EC probe. When the electrodes are dry, use the calibration menu on the readout unit to set the EC value to zero and push enter to save. Rinse the sensors one additional time with DI water. If an H20 Sonde model is being calibrated, ignore this step (these models are zeroed at the factory).

3. Rinse the sensors two times with a small amount of either 0.001, 0.01, or 0.1Molar KCl standard solution. Try to use a solution that has a conductivity range somewhere near the conditions expected to be seen in the field.

4. Pour in the same KCl standard that was used when rinsing, until the conductivity electrodes are covered with the solution. Make sure there are no bubbles attached to the EC chamber; if bubbles are present, gently tap the cup to dislodge them.

5. Check the EC readings and look for stabilization of the value before continuing. EC readings are corrected to 25° C, regardless of the ambient temperature of the solution.

6. Use the calibration button on the readout unit to adjust the EC value to the conductance value of the KCl solution; press "Enter." This saves the calibration reading.

7. If other probes need calibrating, discard the KCl solution and rinse the cup and sensors 2 times with deionized water. When the EC calibration is completed, discard the KCl solution into the sink and flush with tap water. When storing the unit, add a small amount of tap water is in the cup to keep the probes moist.

2.10.2.2 pH Calibration

When performing a pH calibration, always check the buffer solutions being used and make sure that they are still within the expiration period specified on the container. pH calibration is essentially the matching of the meter to the current characteristics of the pH probe electrodes, and is generally performed by measuring separately two buffer solutions of differing and known pH values. This allows the sensitivity, or slope, to be determined. If acidic conditions are expected at the sample site, calibrate using the 4.0 buffer solution and the 7.0 buffer solution. If your sample site typically shows basic conditions of above pH 7, use the 7.0 buffer and the 10.0 buffer. Always use the 7.0 buffer first and then either the 4.0 or the 10.0 to establish the slope. Because of the basic nature of Arizona=s soils, most surface waters are usually above a pH of 7.0.

1. Rinse the probes three times with a half full cup of deionized water to clear the sensors of contaminants. Rinse the cup and sensors two times with a small amount of the 7.0 pH buffer solution.

2. Fill the calibration cup with enough of the 7.0 pH buffer solution to completely cover the pH electrode and the pH reference and Teflon7 junction.

3. Let the temperature and pH values stabilize before recording the current pH value.

4. Use the calibration menu to reset the pH value to 7.00. Save the new calibration value. Record the new value given by the unit in the log-book.

5. Rinse the cup and sensors 3 times with deionized water.

6. Repeat procedures 1 through 4 with a slope solution of either pH 4.00 or pH 10.00.

7. When the calibration is complete, replace the storage cup with a small amount of tap water and turn the unit off.

2.10.2.3 Dissolved Oxygen Calibration

Calibration of the dissolved oxygen (DO) probe is performed in the calibration cup of the H20 Sonde units and in the storage cup of the Minisonde units. Before beginning the calibration process, check the membrane on the DO probe for wrinkles and tears. There should not be any air bubbles present under the membrane. If any of these conditions are present, the membrane should be replaced with a new one before calibration. Hydrolab recommends that the unit be allowed to sit overnight after replacing the membrane. This allows the membrane to stretch and conform itself to the probe. Although calibration can be performed using a known DO concentration, it is easier to use percent saturation DO. Saturation of oxygen in water is determined by air pressure. ADEQ currently employs hand held altimeters capable of reporting ambient air pressure for calibration in the lab and at the sample site. Some Hydrolab units have the barometer built into the read-out unit.

1. Fill the calibration cup with tap water to below the DO probe membrane (water must not cover the membrane).

2. Blot the DO membrane gently with a lint-free absorbent cloth or tissue to remove any water droplets. Use a material that is non-abrasive.

3. Invert the cap and slide it over the top of the calibration cup, and let the unit sit for about 5 minutes to allow the conditions inside the cup to stabilize (Figure 8). Do not screw the cap back on. This will increase pressure inside the calibration cup.

4. Use the hand-held altimeter or the barometer in the unit to measure the ambient air pressure (the terms "air pressure" and "barometric pressure" are equivalent). See Section 2.10.3 for detailed instructions on barometer validation. If a barometer is not available or not functioning properly, contact the nearest Flight Service at a nearby airport. If the calibration site is at approximately the same elevation and is not too distant from the airport, the air pressure reading should be usable for calibration purposes. The reading will usually be reported as inches of Hg and needs to be converted to millimeters for entry into the calibration menu. Conversion to mm of Hg is accomplished using the formula: 25.4 * (inches of Hg) = mm of Hg. For the best calibration results, it is recommended that if the elevation of the sample site is known, the altimeter should be recalibrated to that altitude before using it.

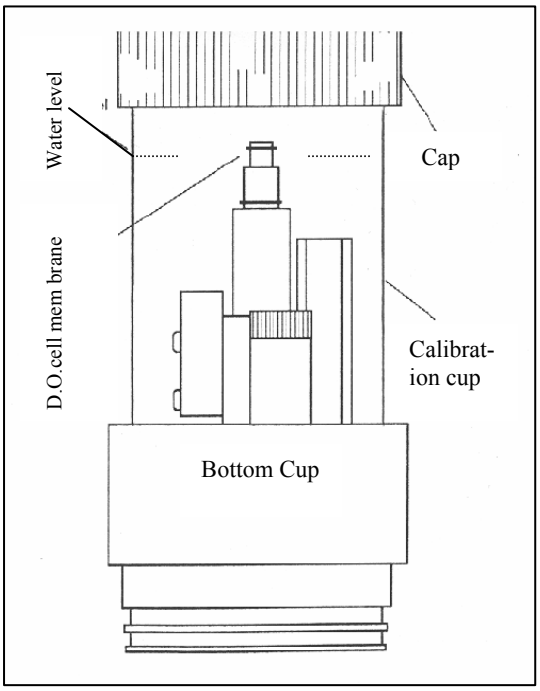


Figure 8. Calibration cup.

If an altimeter is not available or the altimeter on hand is not functioning properly, there is a basic rule of thumb that can be used to obtain air pressure. Standard pressure at sea level is 29.92 inches of Hg. Atmospheric pressure decreases with increasing altitude. For every increase of 1000 feet in elevation above sea level, air pressure will decrease approximately 1.0 inch of Hg. This simple formula can be useful, but may not hold up well in cases where there is a DO exceedence that cannot be attributed to naturally occurring conditions.

5. Before calibrating the DO probe, record the pre-calibration percent saturation value after the unit has stabilized. Using the calibration menu for % saturation, enter the barometric pressure when prompted. Record the new DO percent saturation reading, which should be at or near 100%. The reading should be stable for about 20 to 30 seconds. If DO is being measured at a number of sites and at substantially different elevations, the unit should be calibrated at each site. Most stream sampling will require DO calibration at each site (Figure 9).

Figure 9. Field calibration at sample site.

2.10.2.4 Turbidity Calibration

Calibration of the turbidity sensor on Hydrolab Sondes equipped with that sensor requires the use of a slope calibration standard that is freshly prepared. Preparation of the slope standard for turbidity requires:

Turbidity free water - The deionized (distilled) water obtained from the State Laboratory is satisfactory stock.

Formazin - Use a reagent grade 4000 NTU stock formazin. It may be a safety hazard since it is a suspected carcinogen. See the MSDS in the maintenance kit regarding this material.

The calibration for turbidity requires that about 200 ml of stock turbidity slope standard be prepared. The NTU value for the slope standard should be based on values of turbidity that are expected to be encountered in the field. The following formula is used to calculate the amount of stock needed to get a specific NTU value for the standard which you prepare.

Standard (in Nephelometric Turbidity Units (NTUs)) = $STOCK_{NTU} * STOCK_{VOL}/(STOCK_{VOL} + TF_{VOL})$

Where: $STOCK_{NTU}$ is the NTU value of the stock,
$STOCK_{VOL}$ is the volume of stock used, and
TF_{VOL} is the volume of the "turbidity free" water.

Pipette the required volume of stock into a volumetric flask. Fill to the determined volume line with turbidity free water.

1. Rinse with turbidity free water several times and dry the sensor as much as possible.

2. Fill the calibration cup with turbidity free water and wait for equilibrium (1-2 minutes).

3. Check for and remove bubbles trapped in the sensor by gently tapping the cup. Perform the calibration with a standard value of 0 NTUs.

4. Discard the turbidity free water and again dry the sensors.

5. Fill the calibration cup with the well-mixed NTU standard and wait the 1-2 minutes for equilibrium. Perform the turbidity calibration with the created NTU standard.

Turbidity measurements in situ at streams sites are unreliable and not recommended. For stream sites, use the HACH Turbidity Meter.

2.10.2.5 Source Material

Hydrolab Corporation. 1991. Scout 2, Multiparameter Water Quality Data System, Operating Manual. Austin, TX.

Hydrolab Corporation. 1991. H2O, Multiparameter Water Quality Data System, Operating Manual. Austin, TX.

Hydrolab Corporation. 1997. DataSonde 4 and MiniSonde, Water Quality Multiprobes, User's Manual. Austin, TX.

2.10.3 Validating the Barometer

The dissolved oxygen probe on the Hydrolab must be calibrated by a validated barometer. Barometers, either handheld or factory installed, should be validated for accuracy before each use. Some Hydrolab models require handheld barometers, while other models have built-in barometers. The specification for the factory installed barometer is ± 10 mm Hg. The barometric pressure from either the handheld or built-in models should be within ± 10 mm Hg of the reference barometer. A reference barometer is one that is located at a nearby airport or weather station.

The built-in Hydrolab barometer is "user-zeroed" (i.e., set to reflect the reading from a local reference barometer). If the built-in barometer generates a reading outside the reference barometric reading of ± 10 mm Hg, the built-in barometer electronics may be faulty and the factory should be consulted.

Most hand-held barometers cannot be user-calibrated for barometric pressure. These models have an altimeter that is set to a known elevation which then adjusts the barometric pressure. Consult the instruction manual for these procedures.

When calibrating the dissolved oxygen probe in the field and the built-in barometer generates a value seemingly out of the ordinary, locate a nearby reference barometer at a local airport or weather station. However, the reference barometric pressure reading may have been adjusted

(corrected) to compensate for altitude at its location. The corrected value cannot be used to zero the Hydrolab. Either request an uncorrected barometric pressure or the data necessary to calculate uncorrected pressure for the sample site.

2.10.3.1 Calculating an Uncorrected Barometric Pressure Reading

For pre-trip calibrations, a barometer checking worksheet (Excel) is available. Please note the date incorporated into the file name - newest is best. The worksheet calculates what the barometer should read based upon the current pressure at the Durango Station of the Maricopa County Flood Control District (URL: http://156.42.96.39/alert/Wx/wcurrent.txt). The spreadsheet can be used for calibrations outside the Phoenix metro area with readings from a local reference barometer.

Converting Corrected Pressure in Millibars to Uncorrected Pressure in Millibars

Pressure (uncorrected) in millibars =
[Pressure (corrected) in millibars x 0.750062] - [2.5 x ((elevation in feet x 0.2048)/100)]

Calculation Steps

1) Multiply the pressure (corrected) corresponding to the elevation of the barometer by 0.750062.
2) Multiply the elevation (in feet) by 0.2048, multiply the result by 2.5, divide this result by 100.
3) Subtract the result of step 2 from the result of step 1. This is the uncorrected pressure for the elevation of the barometer.

Converting Uncorrected Pressure in Millibars Hg to Pressure in Millimeters Hg or Inches Hg

Actual pressure for meter location in millimeters = [current uncorrected reference pressure in millibars - standard uncorrected reference pressure in millibars + standard uncorrected pressure for meter location in millibars] x 0.750062

Calculation Steps

1) Subtract the standard pressure (uncorrected) corresponding to the elevation of the reference barometer; e.g., Durango Station, from its current uncorrected pressure.
2) Add the standard pressure (uncorrected) corresponding to the elevation of the barometer being checked; e.g., ADEQ Lab.
3) Multiply the result by 0.02953 to convert to inches of Hg or 0.750062 to convert to mm of Hg. This is the reference pressure for the handheld or factory installed barometer.

2.11 Marsh-McBirney Flow Meter

The Marsh-McBirney Flow Meter should be zero calibrated before each field use.

1. Clean the sensor (with a mild detergent) because a thin film of oil on the electrodes can generate inaccurate field measurements.

2. Suspend the sensor in a five gallon plastic bucket of water. Keep it at least three inches away from the sides and bottom of the bucket. Wait approximately 5 minutes to allow for any water cross currents to dissipate.

3. Use a filter value of 5 seconds. Take the reading. Zero stability is ± 0.05 ft/sec.

4. To zero adjust, position the sensor as described in the zero check procedure.

5. To initiate the zero start sequence, press the "STO" and "RCL" keys at the same time; a number "3" will be displayed.

6. Decrement to zero with the ↓ key and press the "STO" and "RCL" keys a second time.

NOTE: If the ↓ key is not depressed within one second from the time the number "3" is displayed, the unit will display "Err," and steps 5 & 6 will have to be repeated.

7. Turn the unit OFF and back ON. Press STO and RCL keys to reinstate zero start sequence. The number "32" will be displayed and the unit will decrement itself to zero and turn off. The unit is now zeroed.

2.12 Hach Turbidity Meter

The Fixed Station Network Program uses the Hach Model 2100P Portable Turbidimeter (Fig. 10). It is recommended that the user become familiar with the manufacturer=s Instruction Manual before using the meter. The following procedure is an abbreviated version of that found in the instrument manual.

Equipment Required: Hach Model 2100P Portable Turbidimeter, three Gelex Secondary Reference Standards of varying NTU values, three sample vials, Formazin Primary Standard (4000 NTU) or similar calibrating medium, one 10 milliliter graduated cylinder, one 15 ml bottle of silicone oil, and a soft lint-free cleaning cloth.

Figure 10. The Hach Model 2100P Portable Turbidimeter.

2.12.1 Pre-Trip Procedures

The meter should be calibrated in the preparation room before field use.

Meter and Gelex Secondary Reference Standard Calibration Procedure

Refer to the Hach Turbidity Instruction Manual to calibrate the meter and the three reference standards. The meter should be calibrated quarterly to the Gelex Formazine Primary Standard (4000 NTU) or similar calibrating medium.

Optically Calibrating Sample Vials

Precise measurements for very low turbidity samples require optically calibrated sample vials. Refer to the Hach Turbidity Manual for this procedure.

Battery Voltage Check

For the battery voltage check, press the Diagnostic key identified as DIAG. The number displayed is the battery voltage.

2.12.2 Field Procedure for Measuring Turbidity

It is recommended that the meter be placed on a flat surface for taking measurements. Choose a Gelex Secondary Reference Standard that has a turbidity value close to that of the stream water. Thoroughly clean the outer surface of the Reference Standard vial of fingerprints, water spots, and evaporate by applying a thin coat of silicone oil with a soft cloth.

1. Insert the selected Reference Standard into the instrument cell compartment with the white triangle on the vial aligned to the raised orientation mark on the instrument (Fig. 11) and take the measurement. If the vials have been optically calibrated, align the orientation mark on the vial, which may not be the white diamond, to the raised orientation mark on the instrument. The displayed value should be within 5% of the calibration value.

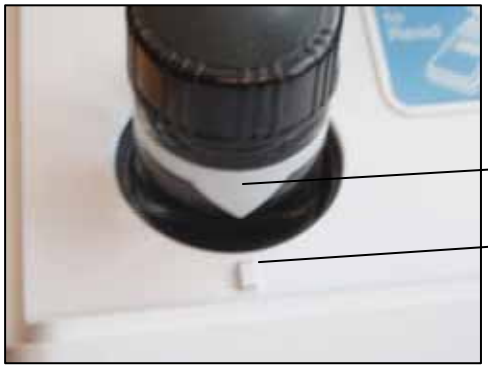


Figure 11. Instrument cell with vial.

If the difference between the measurement and the Reference Standard calibration value is greater than 5%, re-clean and re-oil the Gelex Reference Standard vial, and take another measurement. If the problem persists, record the values on the Field Data Sheet together with a description of the problem. The turbidity value should either not be entered into the water quality database or entered with qualifiers depending on the percent variation from the Reference Standard.

2. Rinse an empty sample vial several times with stream water. Fill the vial with stream water, replace the cap and wipe the outside surface clean and dry with a soft cotton cloth. For grab samples (Section 3A, 3.12.2), the location of the sample should be representative of the entire flow. For composite samples (Sections3A, 3.12.3 – 3.12.5), go through the rinsing process and take the water from the agitated churn splitter (Section 3A, 3.12.19) to insure complete mixing of the suspended matter. If there is any delay between when the vial is filled with stream or composite water and

the measurement, invert the vial several times before placing it into the instrument cell compartment.

3. FSN Field Data Sheets require an average of three turbidity readings. This can be accomplished with two methods; by the meter default or by use of the Signal Average Key. The default setting (Signal Average off) will internally average three measurements and display the result. The signal averaging (Signal Average on) mode averages 10 measurements every 1.2 seconds which compensates for measurement fluctuations caused by the drifting of sample particles through the light path. After 22 seconds, the average of the 10 measurements is displayed.

4. Record the displayed reading and measurement type onto the Field Data Sheet.

5. For very turbid waters, the meter may display a flashing A1,000" value or E-3 error message. This indicates that the turbidity value is greater then 1,000 NTUs. There are two options with this condition: 1) perform a dilution, or record the results as >1000 NTU. Note this on the field data sheet.

3.12.2.1 Performing a Dilution

a. It is recommended that a dilution factor (DF) of 10 be used for the turbidity calculation. The DF is the multiplier for the meter reading. For example, if the operator were to dispense 9 mL of deionized water into a 10 mL graduated cylinder and 1 mL of sample water, for a total of 10 mL, the DF is 10 (a ratio of 9:1); therefore, the turbidity value is the meter reading times 10.

b. For samples that are extremely turbid, it may be necessary to make more than one dilution to obtain a meter reading less than 1,000. For multiple dilutions, the procedure is the same as described above; however, for the second dilution, the 9:1 diluted sample becomes the sample to be diluted. If this is the case, and a 9:1 dilution is performed a second time, the DF is 100 (DF of 10 for the first dilution and DF of 10 for the second dilution). The turbidity value is simply the meter reading times 100.

2.12.3 Source Material

Hach Company. 1993. Model 2100P Portable Turbidity Instruction Manual. Loveland, CO.

2.13 Cleaning Equipment

2.13.1 Churn Splitter

Prior to field use, the churn splitter must be cleaned as follows.

Equipment Required: Hydrochloric acid, non-metallic long-handled brush, non-phosphate detergent, deionized water, pH paper, heavy duty trash bags. For field cleaning, a 5-gallon bucket with limestone chips is required.

1. Wash outside and inside surfaces of the churn thoroughly with tap water and a non-phosphate (e.g., Liquinox) detergent using a non-metallic stiff long-handled brush and let soak for thirty minutes. Before emptying container, run about 100 milliliters of the soap solution through the spigot.

2. Rinse all surfaces thoroughly with tap water.

3. Rinse inside surfaces thoroughly with 500 milliliters of 5% hydrochloric acid (HCl). Run some of the HCL solution through the spigot; however, if the churn splitter contains a metal spring in the spigot, do not open the spigot. For field cleaning, discard acid in a bucket with enough limestone or suitable material to neutralize the acid until it can be disposed of properly at the laboratory. For laboratory cleaning, discard used acid in a waste container labeled "HCL waste" or flush down sink with a copious amount of running water.

4. Rinse all surfaces thoroughly (at least twice) with de-ionized water.

5. After the second rinse, pour approximately 2 liters of de-ionized water into the churn. Swirl the water in the churn; then check the pH with test strip paper. If less than 5.5, discard rinse water and rinse again with de-ionized water.

6. Set cleaned churn splitter on a suitable drying rack in a contaminant free environment.

7. Double wrap the churn with clean heavy-duty trash bags to protect from contaminants during storage and transportation.

2.13.1.1 Source Material

U.S.G.S., 2004. National Field Manual for the Collection of Water-Quality Data. U.S. Dept. of the Interior, U.S. Geological Survey Techniques of Water-Resources Investigations, Book 9, Handbook for Water-Resources Investigations, Chapter A3. Cleaning of Equipment for Water Sampling. Reston, VA. URL http://Pubs.water.usgs.gov/twri9A/

2.13.2 DH-81 and Sample Collection Bottle

Equipment Required: A non-phosphate detergent, a non-metallic stiff short-handle brush, 5% hydrochloric acid, deionized water, pH with test strip paper, and heavy duty sandwich bags. For field cleaning a 5-gallon bucket containing limestone chips is required.

1. Wash all plastic surfaces of the DH-81 (e.g., US DH-81A Adaptor; US D-77 Cap; US D-77 Nozzle) and sample collection bottle thoroughly with tap water and a non-phosphate (e.g., Liquinox) detergent using a non-metallic stiff short-handle brush.

2. Rinse all surfaces thoroughly with tap water.

3. Rinse all surfaces thoroughly with 250 milliliters of 5% hydrochloric acid. For field cleaning, discard acid in a 5-gallon bucket with enough limestone or suitable material to neutralize the acid until it can be disposed of properly. For laboratory cleaning, discard used acid in a waste container labeled "HCL waste" or flush down sink with running water.

4. Rinse all surfaces thoroughly (at least twice) with de-ionized water.

5. After the second rinse, pour approximately 250 milliliters of de-ionized water into the sample collection bottle. Swirl the water in the bottle; then check the pH with test strip paper. If less than 5.5, discard rinse water and rinse with de-ionized water until pH is near neutral..

6. Set cleaned equipment on a suitable drying rack in a contamination free environment.

7. Place cleaned DH-81 parts in heavy duty sandwich bags to protect from contaminants.

SECTION 3

PART A

BASIC FIELD PROCEDURES

TABLE OF CONTENTS

3.00 Field Procedures

This section provides a comprehensive source of field procedures used to monitor surface waters in Arizona by ADEQ. It has a multi-functional purpose: 1) as a reference document to maintain consistency among staff and across years; 2) a training manual; and 3) as an information source for agencies, contractors, organizations, and educators for sampling surface waters.

3.10 Hydrolab Field Procedure

3.10.1 Dissolved Oxygen

At the sample site the dissolved oxygen probe must be calibrated before immersion into stream water. Record all parameter readings on the field data sheet.

1. Fill the calibration cup with tap water to below the DO probe (do not cover the membrane with water).

2. Blot the DO membrane gently with a lint-free absorbent cloth or tissue to remove any water droplets. Use a material that is non-abrasive.

3. Invert the cap and slide it over the top of the calibration cup, and then let the unit sit for about 5 minutes to allow the conditions inside the cup to stabilize. Do not screw the cap back on as this will increase pressure inside the calibration cup.

4. Determine ambient air pressure.

5. Before calibrating the DO, record the pre-calibration percent saturation value after the unit has stabilized.

6. Using the calibration menu for % saturation, enter the current air pressure when prompted.

7. Record the new DO percent saturation reading, which should be at or near 100%. The percent saturation reading should not drift for about 20 to 30 seconds after the calibration procedure.

8. After calibrating the DO probe, insure that the cables are securely attached to both the sonde (or minisonde) unit and the read-out unit.

9. Attach the probe guard (this is a part of the stirrer unit on the older sonde units) before placing the unit in the stream to avoid damage to the probes. If water flows are less than one foot per second, turn the stirrer on.

10. Check to see that all parameters are reporting and then place the sonde in the water body. In streams, this will be at the base of a riffle or other areas where the water is moving swiftly.

11. Allow a few minutes for the meter to stabilize and then record the DO readings.

3.10.2 Specific Electrical Conductivity

With stirrer or probe guard attached, allow meter to stabilize and take reading (µSm/cm).

3.10.3 pH

With stirrer or probe guard attached, allow meter to stabilize and take reading (reported as standard units, or SU).

3.10.4 Temperature

With stirrer or probe guard attached, allow meter to stabilize and take reading (°C).

3.10.5 Turbidity

With stirrer or probe guard attached, allow meter to stabilize and take reading (NTUs). The Hydrolab turbidity readings in streams can be extremely erratic and unreliable. It is recommended that turbidity be measured with a turbidity meter (Section 2.12).

3.10.6 Redox

The REDOX probe may not be present on all ADEQ units. With stirrer or probe guard attached, allow meter to stabilize and take reading.

3.11 Sample Collection Methods for Water Samples

ADEQ employs four techniques for collecting water samples: grab, equal width increment, modified equal width increment and equal discharge increment. For monitoring sites requiring ultra-clean sampling methods, see Section 3A, 3.14.

3.11.1 Collection Bottles

A one-liter wide-mouthed plastic sample collection bottle (Nalgene, or other similar type container as shown in Figure 1), is used for collecting water samples. The bottle must have been decontaminated by either the lab supplying the bottle or by ADEQ personnel.

Clean one-liter unpreserved sample bottles with water-tight caps are filled from the wide-mouth collection bottle. Use bottles made of either glass or a high density inert plastic. In most cases it will depend on the bottle type preferred by the lab. ADEQ currently uses High Density Polyethylene (HDPE) plastic bottles with either white or black caps (Figure 2).

Figure 1. One-liter wide mouth collection bottle.

Figure 2. HDPE plastic bottles with white and black caps.

The number of bottles required per site will be dependent on the parameters being analyzed. Water samples dispensed into white cap bottles are analyzed for parameters such as total inorganics, total dissolved solids (TDS), suspended sediment concentration (SSC), and do not require acid preservation. Water samples dispensed into black cap bottles are analyzed for parameters that require acid preservation, such as metals and nutrients.

Vials of nitric acid and sulfuric acid are provided by the Arizona State Laboratory for the preservation of water samples in the black capped sample bottles (Figure 3). The figure at right displays nitric acid vials with red labels and sulfuric acid vials with yellow labels. Nitric acid is dispensed into the black capped bottle for dissolved metals analysis. Sulfuric acid is used for the water to be analyzed for inorganics.

Figure 3. State Lab supplied nitric (left) and sulfuric acid

Each sample bottle must be labeled (Section 3A, 3.11.8) with permanent markers or preprinted labels for identifying the sample bottles. It is best to label the dry bottles prior to collecting the samples.

If the sample plan calls for the analysis of total cyanide, a sample bottle prepared with a solution of sodium hydroxide is required. When total sulfides are to be analyzed, a sample bottle that has been prepared with a solution of zinc acetate and sodium hydroxide is required. The Arizona State Laboratory or a contract laboratory can supply the preserved bottles upon request.

When sampling a water body that receives discharges of treated effluent, or is composed primarily of treated effluent discharges, a sample for biochemical oxygen demand (BOD) analysis may be required. A BOD sample bottle is typically black plastic or amber glass with an air-tight cap. This type of sample bottle can be supplied by the Arizona State Laboratory or a contract laboratory upon request.

CAUTION: Bottles containing acids or bases as preservatives must always be handled with care. Insure that sample bottle caps are tight before transporting. Acid spilt on skin or clothes must be rinsed and diluted immediately with clean water. When transporting acid vials or lab preserved sample bottles, keep them separated by preservative type. Some sample preservatives can be chemically incompatible and may react violently when mixed.

3.11.2 Grab Sample Method

Equipment Required: 1-liter wide mouth collection bottle (Nalgene, or other similar type container, High Density Polyethylene (HDPE) plastic bottles with white and black caps, nitric and sulfuric acid vials and labels, pH test strips, and zip-loc bags.

All water samples must be collected upstream of any activity that has occurred within the sample reach during field work. This refers primarily to those sampling techniques that involve physical disturbance to the stream bed (e.g. instantaneous flow measurement, pebble counts, macroinvertebrate collection, walking across the channel points in the reach, etc.).

Water samples should be collected after completion of field measurements. In those cases where the water sample cannot be practically taken at the end of the sample visit, it is important to not allow the temperature of the samples to rise significantly above the ambient temperature of the water body being sampled. If an ice chest is not readily available, place the sample bottles in a shaded location in the stream.

1. While facing upstream, collect about 2 liter of water from the middle of the representative stream flow using either the individual unpreserved sample bottles or the one-liter wide-mouthed sample collection bottle. When using lab preserved sample bottles, collect the water sample with the wide-mouthed collecting bottle. Rinse the bottle with stream water several times to flush out any contaminants that might be present. Collect the sample by inverting the bottle open end down, and lower to half the water column depth. Turn the bottle so that it is parallel to the stream bed, allowing

the air to escape and the bottle to fill. When a stream is in flood, locate a spot along the edge of the stream bank where the sample can be safely collected. Rinse the collecting bottle as described.

2. Fill the individual sample bottles. For the black capped bottles, leave space for the introduction of the acid preservative. If using lab preserved bottles, care must be taken not to overfill the sample container. Secure sample bottle caps tightly.

3. Upon returning to the vehicle, perform any sample processing as required. Insure the bottle labels are correct before placing in the ice chest.

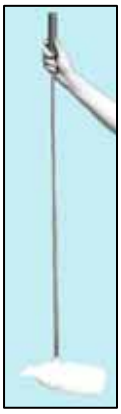
   For samples requiring acid preservation, segregate the bottles by type of preservation required. Proper gloves and eye protection should be in place before proceeding. Prior to adding the preservative, make sure that the bottle is either marked with the type of preservative used, or has a color-coded label that corresponds with the preservative vial being added. After adding the preservative vial to the sample bottle, replace the cap on the sample bottle tightly, and invert the sample bottle several times to mix the sample and preservative. When all sample bottles have been treated, place them in an ice-chest in an upright position.

   pH test strips can be used to identify sample bottles that, for some reason, may not have been preserved. Invert the bottle several times to make sure it is well mixed. Pour a small amount over the test strip. Do not place the strip into the bottle. The test strip should read a pH of less than 2.

4. To dispose of the emptied acid preservation vials, segregate the vials by acid type and place them into separate double-bagged zip-lock bags. Upon returning to ADEQ headquarters, flush the vials and caps with tap water and place them in the proper disposal area. Flush the receiving sink of any acid residues with tap water.

3.11.3 Equal Width Increment Sample Method

DH-81



Equal-Width Increment (EWI) sample collection and Equal-Discharge Increment (EDI) sample collection methods were developed and refined by the United States Geologic Survey (USGS). Both techniques utilize an isokinetic depth-integrating sampler (DH-81) that is designed to accumulate a representative water sample both continuously and isokinetically, meaning that the water approaching and entering the sampler intake does not change in velocity. EWI and EDI sampling techniques are commonly used in large flowing systems which cannot be adequately characterized with a grab sample. The purpose for collecting a EWI sample is to obtain a series of sub-samples, each representing a volume of water taken at equal vertical transit rates and at equal widths apart from each other at various intervals across the channel (Figure 4). This ensures obtaining a discharge weighted representative water sample from the entire flow passing through the channel.

Equipment Required: DH-81 sampler with nozzle and collection bottle, churn splitter, measuring tape, flowmeter and topsetting rod, 1-liter wide mouth collection bottle (Nalgene®, or other similar type container, High Density Polyethylene (HDPE) plastic bottles with white and black caps, and nitric and sulfuric acid vials and labels.

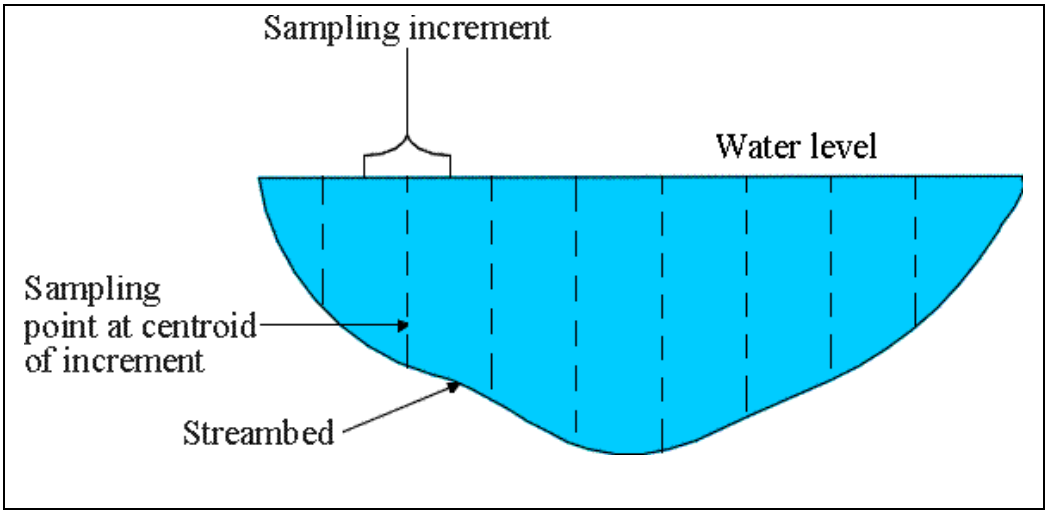


Figure 4. Depth-integrated samples collected using the equal width increment method (from http://sflwww.er.usgs.gov/publications/fs/).

3.11.3.1 Criteria for Using EWI

1) The stream has a depth equal to or greater than one foot for the majority of the stations, determined while in gauging the instantaneous discharge.

2) The stream has discharge velocities exceeding 1.0 ft/sec. for the majority of the discharge stations.

3) Locations where incoming tributary flows have not completely mixed with the main stream flow.

3.11.3.2 Preparing for Collecting the EWI Sample

Instantaneous discharge of the water body is determined with the Marsh McBirney flow meter and top-setting wading rod.

Samples are collected using a Isokinetic Depth-Integrating Sampler (US DH-81 hand-held sampler, Figure 5) which consists of four distinct parts: a three foot long metal rod with a plastic-vinyl handle on one end and machined threading on the opposite end; a US DH-81A molded Teflon adaptor which attaches to the threaded end of the wading rod; a US D-77 molded Teflon cap which has an internally molded air-vent tube; a machined Teflon US D-77 Nozzle with a 5/16 inch sample intake opening; and a 1-liter collecting bottle. There are three nozzles that can

be selected for use depending upon flow velocity. The isokinetic sampler should not be used when minimum flow velocities are less than 1.5 ft./sec.

Nozzle Size, inches	Minimum Flow Velocities, ft.sec.
3/16	2.0 to 6.2
1/4	1.5 to 7.6
5/16	2.0 to 7.0

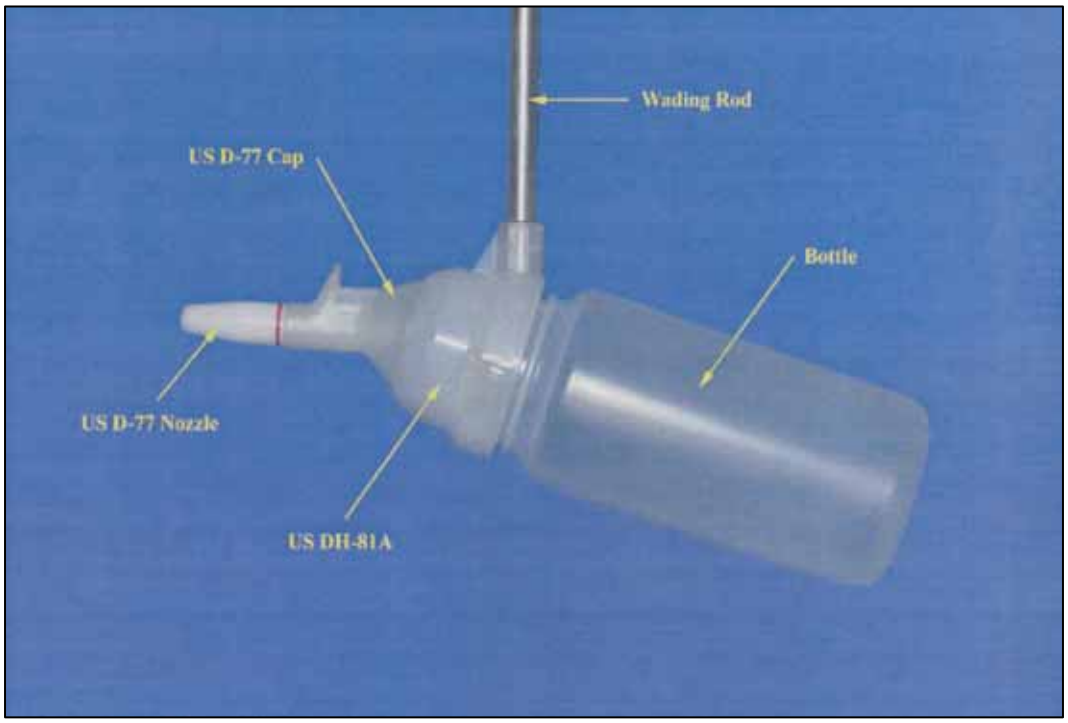


Figure 5. The Depth-Integrating Wading Type - US DH-81 water sampler.

Samples are composited in a clean churn splitter. The SS-1 churn sample splitter is available in 8 liter and 14 liter volume sizes. Ten sub-samples can be withdrawn from the 14 liter unit, and five can be taken from the 8 liter unit.

Upon arrival at the sample site, remove the churn splitter from its protective plastic bag and rinse it well two to three times with water from the stream. Fill to about 2 to : full and place the capped container in a shaded location and into the stream water. This will allow the churn splitter and ambient stream water to equilibrate prior to sampling. When ready to collect the sample, remove the cover and empty the churn splitter. Place the cover in the plastic bag to prevent contamination of the water sample when replacing the cover after the sample has been collected.

US SS-1 Teflon Churn Splitter

Bacteria samples should not be collected form the churn splitter.

After all the samples have been dispensed in the sample bottles, place the churn splitter back into the plastic bag to prevent cross-contamination with other equipment.

3.11.3.3 EWI Sampling Procedure

1. Extend a measuring tape transect across the stream channel and perpendicular to the flow at a sampling location not influenced by side-channel eddies.

2. Measure the instantaneous flow discharge as described in Section 3C, 3.18. This measurement is required to perform EWI.

3. The number of vertical intervals to be sampled is based on the following variables:

   - volume of sample needed for analysis (number of sample bottles to be filled)
   - size of the churn splitter
   - and, the depth and velocity distribution in the cross section at the time of sampling. The collection bottle may be completely filled at each vertical interval, if the flow at the individual discharge stations is consistent throughout the transect.

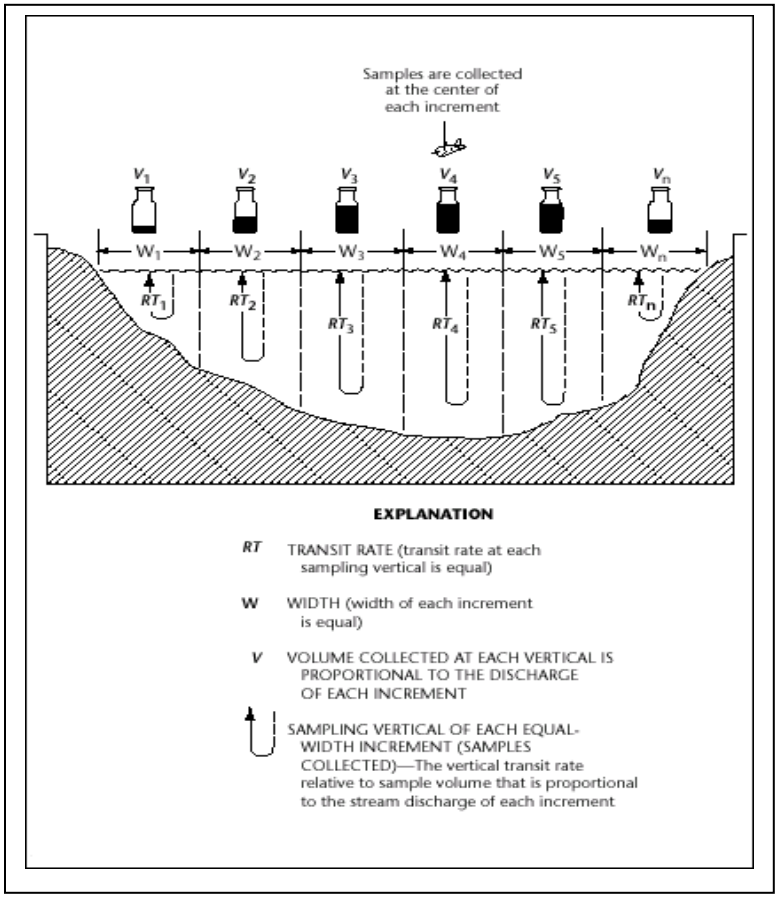
4. Establishing the transit rate. Determine the maximum discharge vertical obtained from the discharge measurement. In most cases this will be the deepest, fastest point identified along the transect line. Before beginning the transit, establish a starting reference point on the body of the observer, such as the belt buckle. All transits across the channel should begin at the same reference point regardless of the water depth.

   The observer begins the first transit by facing upstream at the selected vertical, while holding the DH-81 rod perpendicular to the surface and the nozzle parallel to the water surface. The bottle is lowered through the water column to the streambed and raised back to the surface, to the starting reference point, while maintaining a constant transit rate.

   To gage the transit rate, the observer should count from the beginning of the transit to the streambed and back to the beginning point. The count from the streambed to the reference point should be half the transit count. The correct transit rate is confirmed when the collected water is just below the shoulder of the DH-81 collecting bottle. It may require several attempts to perform the procedure correctly. The transit rate should be maintained throughout the transect regardless of the depth or rate of flow at the transect verticals. Figure 6 is a graphic illustration of the EWI method.

   It is recommended that the initial transit be located at the distant bank and work back to the bank where the equipment are located. It may be possible in some streams to sample at two or more verticals without emptying the sample collection bottle. If, during the sample collection process, the collection bottle is overfilled, the bottle must be emptied and all verticals, which contributed to that particular sub-sample, collected again.

Samples are collected at the center
of each increment



Explanation
RT = Transit Rate (transit rate at each sampling vertical is equal)
W = Width (width of each increment is equal)
V = Volume collected at each interval is proportional to the discharge of each increment
Ô = Sampling vertical of each equal width increment (samples collected). The vertical transit rate relative to sample volume that is proportional to the stream discharge of each increment.

Figure 6. Illustration of the equal width increment procedure.

5. Using the transit rate established in Step 4, go to either bank and prepare to collect the sub-samples at the established intervals and transit rate. The first vertical sub-sample should be at a point one half the distance of the selected increment width from the bank.

   - For example, if the stream width is 20-feet, and the required number of verticals is 10, then the width of each sampled increment would be 2 feet. The sub-sample station within each width increment is located at the center of the increment, beginning at a location of 1 foot from the bank. The verticals are then spaced 2 feet

apart, resulting in a sample station at 1, 3, 5, 7, 9, 11, 13, 15, 17 and 19 feet along the 20-foot width.

Face upstream at the selected sub-sample vertical and collect the sample with the DH-81 at the predetermined transit rate. A second observer should carry the churn splitter to receive the samples.

6. After all the sub-samples have been deposited into the churn splitter, the individual pre-labeled sample bottles can be filled. Prior to withdrawing the samples, place the churn splitter onto a flat, stable surface. Mix the composite sample by churning it with the handle of the agitator disk at a constant rate of approximately nine inches per second. The agitator disk should touch the bottom on every down stroke, and should not break the surface of the water on the upstroke. Complete mixing is obtained after ten strokes. During the agitation process, purge the churn splitter spigot by flushing about fifty to one hundred milliliters of the composite sample. Agitation of the sample should continue while dispensing the sample into the bottles.

3.11.4 Modified Equal Width Increment Sample Collection

The Modified EWI method is often used in small wide streams where depths are less than one foot, and horizontal mixing is insufficient for capturing a representative sample. It is also used in shallow streams where tributary inflows are not well mixed with the dominant mainstream flow. The procedure is designed to produce a sample that is more representative than a grab sample, but not discharge weighted. The sub-samples are collected by hand with the wide-mouth 1-liter collecting bottle at the determined vertical intervals.

Equipment Required: Churn splitter, measuring tape, 1-liter wide mouth collection bottle (Nalgene®, or other similar type container, High Density Polyethylene (HDPE) plastic bottles with white and black caps, 250-milliliter collection bottle, and nitric and sulfuric acid vials and labels.

3.11.4.1 Modified EWI Sampling Procedure

1. Determine the number of vertical intervals as described in Section 3A, 3.11.3.3, Step 5.

2. Holding the collection bottle with the mouth pointed down, the bottle is lowered vertically into the water column. When the mouth of the bottle is about two inches above the stream bed, turn the bottle to the horizontal position with the mouth facing upstream and raise the bottle to the surface. Determine an approximate transit rate by experimenting with the rate of ascent and the rate of turning the bottle to a horizontal position. The bottle should not be allowed to fill. Place the collected sub-samples into a churn splitter, mix the composite sample and dispense the samples as described in Section 3A, 3.11.3.3, Step 6.

3. For streams of shallow depth where the use of a 1-liter bottle and a churn splitter may be impractical, use a clean 250 milliliter bottle to collect the sample. Establish four sampling points along the transect that are approximately equal in distance from each other. Collect a sub-sample from each point, using a transit rate that fills the bottle completely and evenly at each point along the transect. Cap and invert the bottle several times to mix the sample.

3.11.5 Equal Discharge Increment Sample Collection

The EDI method produces a discharge-weighted sample that represents all the flow passing through the cross section by collecting a number of sub-samples, each representing equal volumes of discharge.

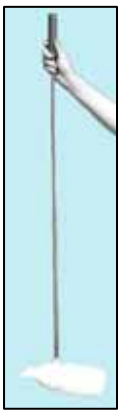
The flow in the cross section is divided into increments of equal discharge and then equal-volume. Depth-integrated samples are collected at the centroid of each increment along the transect. The term centroid refers to the location in the channel transect where discharge is equal on both sides. EDI sample collection is used by ADEQ principally for the collection of suspended sediment concentration (SSC) samples (Section 3.11.6). ADEQ typically collects four sample bottles at each site, each representing 25 percent of the total discharge of the stream being sampled.

3.11.5.1 EDI Sampling Procedure

The EDI sampling procedure for collecting SSC samples is described in Section 3.11.6.7.3.

3.11.6 Suspended Sediment Concentration Protocol

This protocol describes the equipment and field procedures for collection of suspended sediment concentration (SSC) samples in wadeable, perennial streams during normal flows. The SSC field protocol has been adapted from Field Methods for Measurement of Fluvial Sediment (Edwards and Glysson, 1999). The equipment and field methods described in this protocol are designed to yield a representative sample of a water/sediment mixture moving in a stream. The field procedures are limited to use in shallow streams at normal flows that can be safely waded. A general rule is that a stream's wadeability is questionable when the product of the stream depth and velocity in ft. / second equals 9 or greater. These field procedures do not apply to SSC sampling in non-wadeable streams and sampling during high flow events when it is not safe for the sampler to wade into a stream. Also, these field methods do not apply to sampling bed material or bedload.



Equipment: 250 micron sieve, plastic 1-liter sample unpreserved bottle, Nalgene sample collection bottle, DH-81 depth integrating sampler, 1/4" isokinetic nozzle, discharge measurement equipment (for Equal Discharge Increment method), churn splitter (for composite samples collected using Equal Width Increment method).

3.11.6.1 The DH-81 Depth-Integrating Sampler

The DH-81 depth integrating sampler is a hand-held sampling device that consists of a plastic nozzle ring attached to a 3-foot long metal rod. The plastic ring on the DH-81 sampler accepts any sample bottle having standard mason jar threads. For SSC sampling, ADEQ field personnel use a 1- liter Nalgene sample collection bottle that is threaded onto the nozzle ring. The DH-81 sampler is designed to isokinetically and continuously accumulate a representative sample from a stream vertical while transiting the vertical at a uniform rate. "Isokinetic" means that the intake velocity through the nozzle into the sample collection bottle and the ambient stream velocity are equal.

3.11.6.2 DH-81 Nozzles

The DH-81 depth integrating sampler is usually equipped with the 1/4 –inch nozzle (see Section 3.11.3.2 for nozzle selection) that is cut and shaped to ensure that the intake velocity of the water entering the nozzle is within 8% of the ambient stream velocity when stream velocity is greater than 1.5 ft./sec. (i.e., isokinetic).

3.11.6.3 DH-81 Sample Bottles

Nalgene 1-liter sample collection bottles with standard mason jar threads are used with the DH-81 sampler and for collecting grab samples. If subsamples are composited in a churn splitter, the sample that is withdrawn from the churn into a 1-liter unpreserved plastic bottle, provided by the ADHS State Laboratory.

3.11.6.4 Churn Splitter

An 8-liter or 14-liter plastic churn splitter is used to composite samples for SSC analysis. Samples totaling 10 liters can be withdrawn from the 14-liter churn splitter for whole water analyses. If a split sample will be done at a site, then the 14-liter churn should be used. Samples totaling 5 liters can be withdrawn from the 8-liter churn splitter. Thus, for routine sample analyses (i.e., a 4-bottle water chemistry set + SSC), an 8-liter churn splitter may be used. The sample for SSC analysis should be withdrawn from the churn splitter first.

3.11.6.5 250 Micron Sieve

A 250 micron sieve is used to make a visual determination as to whether sand-sized particles are suspended in the water column. Using a 1-liter sample collection bottle, the field person collects a grab sample at the deepest, fastest point in the stream channel. The sampler should take care not to sample any bed material. Slowly empty the water in the sample collection bottle into the sieve and visually inspect the sieve for sand-sized particles. If sand sized particles are not in the sieve, then the observer may collect a grab sample or use the Equal-Width-Increment (EWI) method to obtain a composite sample using a churn splitter. If sand-sized particles are in the

sieve, then the sampler may collect a grab sample if the stream is less than 10 feet wide or use the equal-discharge-increment (EDI) method to obtain multiple sample bottles for SSC analysis.

Tests performed by the U.S. Geological Survey demonstrate that a composite sample from a churn splitter can provide unbiased and acceptably precise (generally within 10% of the known value) SSC values as large as 1,000 mg/L when the mean diameter of sediment particles is less than about 0.25 mm. At SSC values of 10,000 mg/L or more, the bias and precision of SSC values in churn splitter sub-samples are considered unacceptable (Gray, et al., 2000).

3.11.6.6 Site Selection

The ideal SSC sampling site is defined as a cross-section displaying stable hydrologic characteristics and uniform depths over a wide range of stream discharges from which representative water quality and sediment data may be obtained that can be related to a stage discharge rating for the site. For this reason, the ideal SSC sampling site is located at or near an existing gage site.

Sites that may be affected by backwater conditions should be avoided whenever possible.

Sites that are immediately downstream of the confluence of two streams or a tributary stream should be avoided. Such sites can present problems because of incomplete mixing of flows; therefore, samples should be taken far enough downstream to ensure adequate mixing. The distance downstream from a confluence for complete mixing will depend on stream velocity, depth, and mixing width.

If suspended sediment concentration data is needed from the stream during periods of high flow, and thus not wadeable, then a site providing high flow access must be selected (i.e. bridge or cableway).

3.11.6.7 Selecting the SSC Sampling Method

When sampling for SSC, emphasis should be placed on acquiring a representative sample of the suspended sediment particles in transit. To acquire a representative sample, the sampler must obtain a sample that adequately defines the concentration of suspended sediment particles over the full depth of a sampled vertical. Also, a sufficient number of verticals must be sampled to adequately characterize the horizontal variation in a cross-section of the stream.

Several sampling methods may be used to obtain a representative sample depending on the flow conditions at the time of sample collection and the characteristics of the suspended sediment being sampled. In general, four sampling methods are available:

- Grab sampling
- Composite sampling using the equal width increment method (EWI)
- Modified equal width increment method
- Equal discharge increment sampling with multiple sample bottles (EDI).

Grab sampling for SSC may be used in shallow, low velocity streams. In general, grab samples may be taken when the stream that is being sampled is less than one foot deep for the majority of the channel and the stream velocity is less than 1.5 feet per second for the majority of the channel.

Composite sampling using the EWI method may be used when sieving indicates that sand-sized particles are not entrained in the water column and one or more of the following stream conditions is met:

- The stream depth is equal to or greater than one foot for the majority of the channel
- The stream velocity exceeds 1.5 ft / second for the majority of the channel, or
- The sampling site is located where tributary flows have not completely mixed with the main stream flow.

The modified equal width increment method is used when the water depth is less than one foot deep and a composite sample needed. The DH-81 sampler cannot be used shallow water, but a composite sample can be taken when a one liter bottle is filled at equal distances across the channel and deposited in a churn splitter. The number of samples is dependant upon the size of the churn splitter. On narrow streams it may be necessary to take replicate samples at the same locations to obtain the required amount of water to fill the churn splitter.

Equal discharge increment sampling using multiple sample bottles is used when sieving indicates that sand-sized particles are entrained in the water column and the stream is 10-feet wide or wider. If sand-sized particles are entrained in the water column and the stream is less than 10-feet wide, a grab sample may be taken from a single vertical in the deepest, fastest location in the channel.

3.11.6.7.1 Grab Sampling

A grab sample is collected by submerging a 1-liter sample collection bottle by hand into the stream. The observer faces upstream and collects a water sample from the deepest and fastest vertical available in the channel. The open mouth of the sample bottle should be held below the water surface, pointed upstream, and held at a 45° angle from the streambed. The bottle should be filled by moving the bottle vertically to the streambed. An un-sampled zone of about 3-inches above the streambed should be maintained to avoid sampling bed material.

3.11.6.7.2 Composite Samples: Equal Width Increment Method

Field personnel may use the EWI method to collect sub-samples from a stream for compositing in a churn splitter. In the EWI method, the sampler collects sub-samples using a DH-81 hand held sampler, a 1/4" nozzle, and 1-liter sample collection bottle. The sub-samples are collected at the center of equally-spaced increments using an equal transit rate at all sample verticals (Figure 4).

The number of verticals required for the EWI method depends on the sample volume needed to fill all sample bottles and the stream width, depth, and velocities in the cross-section at the time of sampling. USGS recommends that a minimum of 10 verticals be used for streams that are over 5-feet wide. For streams less than 5-feet wide, USGS recommends that as many verticals as possible be used as long as the increments are spaced a minimum of 3-inches apart to allow for discrete sampling at each vertical and to prevent overlaps. USGS recommends a maximum of 20 verticals for all but very wide and shallow streams. The width of each increment is determined by dividing the stream width by the number of verticals to be sampled.

The collection procedures for EWI sampling are as follows:

1. Place measuring tape across stream channel perpendicular to flow at the sampling site. Determine the stream width from the edges of water.

2. Determine the number of verticals to be sampled in order to obtain enough sample volume in the churn splitter for all required samples. Factors to consider are:

   - The number of sample bottles that need to be filled (e.g., four 1-liter bottles for water chemistry + one 1-liter bottle for SSC sample = 5 liters)
   - A 14-liter or 8-liter churn splitter (The churn splitter must be large enough to hold enough sample volume to fill all sample bottles. Up to 5 liters of sample may be withdrawn from an 8-liter churn. A 14-liter churn must be used if split samples are withdrawn for QA / QC purposes.)
   - The stream width, water depths, and the stream velocity distribution in the cross section at the time of sampling.

3. Take discharge measurements using Marsh McBirney flow meter to determine the point of maximum discharge in the stream.

4. Establish the proper transit rate: This is done by going to the point of maximum discharge in the stream obtained from the field discharge measurements (usually the deepest, fastest point in the stream channel). Using the DH-81 sampler and facing upstream, place the sample collection bottle on the water surface at the vertical to be sampled. To begin the process, position the bottom of the sample bottle on the surface, and while counting, descend and ascend the vertical using a transit rate (the count from beginning to end) that fills the 1-liter sample collection bottle without overfilling it (it may take several tries to establish the proper transit rate at the maximum discharge point). The rates of descent and ascent of the sampler while transiting the vertical must be equal. Once the proper transit rate is established, the sampler uses the same transit rate at all other verticals.

   It is sometimes difficult to maintain an equal transit rate while collecting samples at different verticals in the stream. USGS recommends the following procedure to minimize variations in the transit rate. When establishing the proper transit rate at the point of maximum discharge, the field person should note a reference point on the body where the downward and upward integration is started and finished (e.g., at the hip). This same reference point is used at each vertical regardless of the stream depth encountered at each vertical. This means the sampler will start the depth integration in

the air above the surface of the water in shallower parts of the stream. By starting at the same reference point, the sampler can better control the amount of time that elapses during the round trip of the sampler and the transit rate will remain relatively constant.

5. Go to the either the LEW (or REW) and prepare to collect sub-samples at the established increments. Sub-samples are collected at the centroid of each established increment. The first vertical is sampled at a point that is ½ the distance of the selected increment width from the edge of water. For example, if the stream width is 20 feet and the number of verticals determined to be necessary is 10, then the width of each increment is 2 feet. If starting at the LEW, the first vertical would be sampled at a point that is ½ of the increment width or 1 foot from the LEW. Each vertical is then spaced 2-feet apart, resulting in verticals that are sampled at 1, 3, 5, 7, 9, 11, 13, 17 and 19 feet (Figure 4).

6. Depending on the water depths, it may be possible to sample at more than one vertical without risk of overfilling the sample collection bottle. However, if there is a possibility that the sample collection bottle will overfill at a vertical, then the sample in the sample collection bottle should be emptied into the churn splitter. If the sample collection bottle is overfilled at a vertical, then the sample bottle must be emptied into the stream and all verticals that contributed to the contents of the bottle must be re-sampled.

7. Continue sampling at the centroid of each increment using an equal transit rate at each vertical until all verticals are sampled. Empty the sub-samples into the churn splitter as necessary.

8. Composite the sub-samples in the churn splitter and withdraw a 1-liter sample into an un-preserved sample bottle for SSC analysis. The SSC sample should be withdrawn first if other samples will be withdrawn for inorganics, total and dissolved metals, and nutrients.

3.11.6.7.3 Equal Discharge Increment Method

The objective of the EDI is to collect a discharge-weighted sample that represents the entire flow passing through the cross-section by obtaining a series of samples, each representing equal volumes of stream discharge. The EDI method requires that flow in the cross-section be divided into increments of equal discharge.

Collection procedures for EDI sampling are as follows:

1. Place measuring tape across stream channel perpendicular to flow at the sampling site. Determine the stream width from the edges of water.

2. Take discharge measurements using Marsh McBirney flow meter to determine the total discharge and the flow distribution across the channel at the cross section. When recording discharge measurements on the field form, the recorder should include a

cumulative discharge column and keep a running total of the cumulative discharge from the LEW.

3. Calculate the equal discharge increment. The equal discharge increment is determined by dividing the total discharge by the number of verticals to be sampled. For example if the total discharge of the stream is 86 cfs and 4 verticals are to be sampled, then the EDI method is 22 cfs (Table 1).

4. The location of the centroids of the equal discharge increment is determined from the cumulative discharge calculations. The first vertical is located at a point where the cumulative discharge from the LEW is half of the EDI. In the previous example, if the EDI is 22 cfs, then the first vertical is located at the point where the cumulative discharge = 12 cfs. Subsequent centroids are located by adding the EDI to the cumulative discharge at the first vertical. In our example the second vertical would be located at the point where cumulative discharge = 33 cfs relative to the LEW. The third centroid would be where cumulative discharge = 55 cfs and the fourth centroid would be at the point where cumulative discharge = 77 cfs.

5. At each centroid, a depth-integrated sample is collected using a DH-81 sampler. Each sample bottle collected is part of a multiple bottle set for the cross-section. The transit rate used to collect a sample must be constant in one direction. However, it is not necessary to maintain equal transit rates of ascent and descent within a vertical. Also transit rates can vary at different verticals in the cross-section. A single sample bottle is filled at each vertical. However, it is important that all sample bottles be of equal sample volume.

3.11.6.8 Sample Preservation

A chemical preservative is not required for SSC samples.

3.11.6.9 Sample Bottle Labeling

Each SSC sample bottle must be labeled prior to leaving a sample site. The following information should be marked on the outside of the sample bottle using a permanent, waterproof marker (Sharpie or other similar product):

Site identification number
Complete site name
Date of sample collection
Time of sample collection
Initials of the sampler
ADEQ
SSC

3.11.6.10 Literature Cited

Edwards, T.K. and G.D. Glysson. 1999. Field Methods for Measurement of Fluvial Sediment. U.S. Geological Survey, Open File Report 86-531, Reston, VA.

Gray, John R., et al. 2000. Comparability of Suspended Sediment Concentration and Total Suspended Solids Data. U.S. Geological Survey, Water Resources Investigations Report 00-4191 (August).

http://fisp.wes.army.mil/Instructions%20US_DH-81_010612.pdf

Table 1. Example of Equal Discharge Increment Method calculation for the location of sample vertical centroids.

Distance	Width	Depth	Velocity	Q	Cum. Q	EDI Sample
0	0.75	0	LEW	0	0.00	
1.5	1.5	0.15	0.14	0.03	0.03	
3	1.5	0.35	0.67	0.35	0.38	
4.5	1.5	0.5	0.91	0.68	1.07	
6	1.5	0.8	1.58	1.90	2.96	
7.5	1.5	1.05	1.97	3.10	6.06	
9	1.5	1.34	2.4	4.82	10.89	#1
10.5	1.5	1.35	3.85	7.80	18.68	
12	1.5	1.6	3.95	9.48	28.16	
13.5	1.5	1.7	3.4	8.67	36.83	#2
15	1.5	1.7	2.8	7.14	43.97	
16.5	1.5	1.7	3	7.65	51.62	
18	1.5	1.55	2.5	5.81	57.44	#3
19.5	1.25	1.4	2.53	4.43	61.86	
20.5	1.25	1.22	2.63	4.01	65.88	
22	1.5	1.3	2.88	5.62	71.49	
23.5	1.5	1.25	2.12	3.98	75.47	
25	1.5	1.18	1.1	1.95	77.41	#4
26.5	1.5	1.35	1.38	2.79	80.21	
28	1.5	1.52	1.07	2.44	82.65	
29.5	1.5	1.32	0.44	0.87	83.52	
31	1.5	1.4	0.23	0.48	84.00	
32.5	1.5	1.05	0	0	84.00	
34	2.15	0.4	0	0	84.00	
36.8	1.4	0	REW	0	84.00	

Where: Q = Discharge
Cum. Q = Cumulative Discharge
LEW = Left Edge of Water
REW = Right Edge of Water
Number of verticals to be sampled = 4

Calculation of Centroid Location
First Centroid = 84 / 4 = 22 / 2 = 12 cumulative cfs
Second Centroid = 11 + 22 = 33 cumulative cfs
Third Centroid = 33 + 22 = 55 cumulative cfs
Fourth Centroid = 54 + 22 = 77 cumulative cfs

3.11.7 Filtering Dissolved Metals

The analysis of dissolved metals requires filtration of the water sample prior to preserving the sample with nitric acid. An unpreserved water sample consists of two analytical components of interest: suspended metals and dissolved metals. Suspended metals are defined as the portion of a water sample that is unable to pass through a membrane filter with a 0.45 micrometer pore size. Dissolved metals are those in solution which are able to pass through the same membrane filter.

Equipment Required: A peristaltic pump, a short (2 to 3 feet) section of clean, ¼ inch diameter, silicon-based tubing, a groundwater capsule filter with 0.45 µm pore size, and an acid cleaned collecting bottle and an acid cleaned receiving sample bottle (Figure 7).

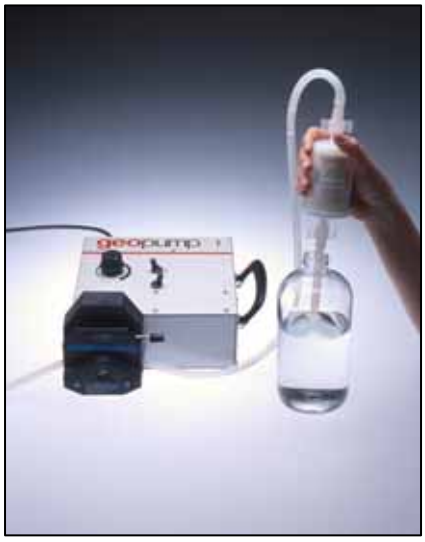


Figure 7. Filtering setup.

3.11.7.1 Peristaltic Pumps

ADEQ currently uses peristaltic pumps that operate from any external 12 volt DC or 120 volt AC power source, thus allowing the sample to be filtered in either the laboratory or the field. Typically the pumps are equipped with easy-load pump heads (as shown in the adjacent figure), but some pumps may be equipped with the factory supplied standard pump head design.

3.11.7.2 Filter Tubing

The silicon-based tube should be cut to length in the lab before going into the field. While cutting the tubing, wear a pair of clean lab gloves and make the cut with a ceramic knife to prevent contamination of the tubing. After cutting the pieces to length (one tubing per site, plus any extra needed for QAQC samples), place them in a clean, sealable plastic bag for transport to the monitoring sites.

3.11.7.3 Groundwater Sampling Capsule

The groundwater sampling capsule currently utilized by ADEQ (Figure 8) is able to filter waters with high suspended sediment concentrations. However, a smaller less expensive filter is available when filtering water with low sediment concentration. Transparent water can usually be filtered with a smaller filter. All filters should be quality certified by the supplier. Filters are designed for a single use and should be disposed of after each filtration.

Figure 8. Groundwater Sampling Capsule.

3.11.7.4 Filtering the Water Sample

1. Two clean sample bottles are required; one in which to collect the water, and the second to contain the filtered water. The second bottle should be labeled "Field Filtered."

2. Place the pump on a hard, flat surface (e.g. a table or the pickup tailgate). Position it such that the pump head of the mechanism extends over the edge of the stationary surface (Figure 9). Remove a section of the clean, pre-cut tubing from the re-sealable plastic bag. Always handle the tubing near the middle to prevent the tube ends from being contaminated.

3. Insert the tubing into the pump mechanism such that both ends are hanging loosely, but not in contact with any surface. Remove the filter from its packaging, taking care to not contaminate the nipple ends of the capsule. Securely attach the tubing to the filter.

4. Check the pump controls to insure the flow direction of the pump is congruent with the flow direction of the capsule filter. Place the end of the tubing without the filter into either the bottle or the churn splitter.

5. Turn the pump on and allow the filter to fill with water before filling the sample bottle. Allow about twenty five to fifty milliliters of the sample to run out of the out-flow opening to flush the filter. Place the out-flow end of the tubing into the open mouth of the pre-labeled filtrate bottle and fill to nearly full (Figure 10). If the bottle is pre-preserved with acid, do not overfill the bottle. If nitric acid is to be added after filtering, leave some space in the bottle for the addition of the preservative.

6. After placing the acid-preserved sample into the ice-chest for transport, properly dispose of the filter and tubing.

Figure 9. Positioning the peristaltic pump in preparation for filtering.

Figure 10. Filling the sample bottle.

3.11.8 Sample Bottle Labeling

Each water sample bottle must be labeled with a site code, a site location description, sample collection time and date, the initials of the observer collecting the water sample, and the agency name. Use a black or blue permanent marking pen, such as a Sharpie or other similar product, to label dry bottles. Handwriting must be precise and legible. Bottles are labeled in the order presented below.

Sample site identification code

- A sample site is given a code based on the water basin, name of the stream, and the river miles from the terminus of the stream. Section 1.10.2 describes the process for determining this code. Each code is unique for a given sample site. The site code is placed near the top of the bottle. An example of a site code is UGGLR505.96. "UG" is the Upper Gila River Basin. "GLR" is the identification designation for the Gila River. The site is located 505.96 miles from its terminus and confluence with the Salt River.

Site location description

- This is a brief generalized description that attempts to convey the location of the sampling point. The description will normally reference a permanent physical feature of some type. An example of this would be "Spring Creek below confluence with Dry Creek," or "Cienega Creek above Marsh Station Road Bridge." If a permanent physical feature is unavailable, the description may be as non-specific as "Trout Creek near Wikieup." Avoid using descriptions that are similar to other site descriptions.

Sample collection time and date

- Mark sample bottles with the collection time and date that appears on the field data sheet. Collection times are reported as sidereal (military) time (e.g., 9:45 a.m. = 0945 hours).

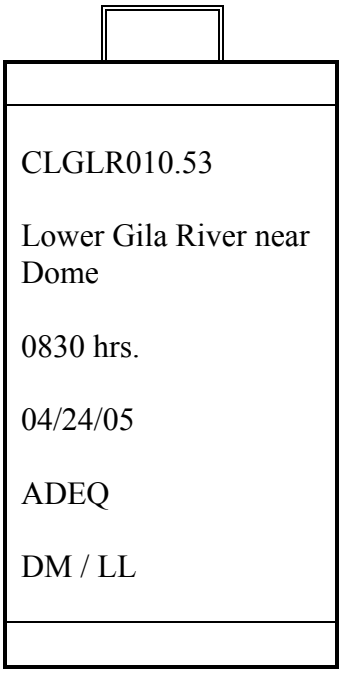
Agency name

- The placement of the agency name on the bottle informs the receiving lab of the billing entity. If samples have been collected and submitted for another agency or program that has interest in the sample site, label the bottle as "ADEQ for AGFD (or TMDL, etc.)."

Observer's initials

- The observer's initials indicate the person responsible for collecting and submitting the water sample. Initials of other field personnel may be applied to the bottle.

A properly labeled sample bottle is shown:



Miscellaneous Labeling Requirements

- Water samples that are field filtered must be labeled as "Field Filtered." When field filtering cannot be performed because of filtering equipment malfunctions, label the bottle "To Be Filtered" and make note on the laboratory submittal form.
- Water samples that are collected for suspended sediment concentration analysis are labeled prominently as "SSC."
- If problems arise with the acid preservative labels or there is a possibility of them wearing or falling off the bottles during transport, mark the bottles as either "sulfuric acid" or "nitric acid."

3.11.9 Operating the Churn Sample Splitter

1. Once the required volume of water has been dispensed into the sample splitter, churn the sample at a uniform rate of about 9 inches per second . The disc should touch the bottom of the churn on the down stroke and should not be allowed to break the water surface on the up stroke to prevent non-representative samples.

2. A minimum of 10 strokes is required before withdrawing the first subsample. Rinse the churn spigot with a small quantity of collected water. As subsamples are withdrawn, maintain an even churn rate of 9 inches per second. If the disk breaks the water surface while a subsample bottle is being filled, stop the filling process and stroke 10 times before continuing to fill the subsample bottle.

3.11.10 Ambient Sample Blanks

Ambient sample blanks are intended to determine if exposure of the sample to the environmental collection and processing procedures introduce measurable concentrations of target analytes. Different procedures can be used for collecting the ambient blank sample depending on the site characteristics or conditions being subjected to quality control.

The Fixed Station Network will typically use a three bottle chemset (one unpreserved for inorganics, one nitric-acid-preserved for total metals, and one sulfuric-acid-preserved for nutrients) as blanks. Unless specifically designated for a sample site or testing opportunity, a dissolved metals sample is not submitted to the Sate Lab.

Equipment that may be required for the quality control samples are a precleaned churn sample splitter (see Section 2.13), State Lab certified deionized water, a three bottle chemset, nitric and sulfuric acid vials, and precleaned sample bottles. See Section 3.11.9 for the proper operating procedure of the churn sample splitter and Section 3.11.13 for QC bottle labeling procedures.

3.11.10.1 Purpose of Blanks and Background Samples

Blank samples are a check for cross-contamination during sample collection and shipment, and in the laboratory. There are three types of quality control blanks: trip, field, equipment/churn. De-ionized water should be used for metal parameters. Blanks should be prepared by ADHS or the laboratory that supplies the sample containers. The blank should be numbered, packaged, and sealed in the same manner as other samples in the set.

3.11.10.2 Trip Blank

A trip blank is a water sample that remains with collected samples during transportation and is analyzed along with field samples to check residual contamination. A trip blank should not be opened by either the sample collector or sample handlers.

Procedure: Store the trip blank in a container or plastic bag such that the blank bottle is not exposed to ambient conditions during transport to the sample site. At the sample site, remove the trip blank from its container and place in the same cooler as the collected samples during transport to the analyzing laboratory.

3.11.10.3 Field Blank

A field blank is a water sample that travels with the sample set and is opened and exposed at the sampling point to detect contamination from air exposure. The field blank, usually deionized water, may be poured into appropriate containers to simulate actual sampling conditions. Contamination of the sample can be from air exposure during the collection process and during storage and transport.

Procedure: In the laboratory (ADEQ or State Lab), fill a clean sample bottle(s) with State Lab certified deionized water and transport it to the field. Place the bottle(s) in the processing area (e.g., truck tail gate) in which the environmental sample(s) are being processed. Open the blank sample bottle to expose the blank sample to the atmosphere for approximately the same period of time to which an individual environmental sample(s) is exposed during sample processing. Store and transport the blank(s) in the same container as the environmental samples.

3.11.10.4 Equipment/Churn Blank

This type of blank sample is normally collected after the sampling equipment has been used and subsequently cleaned. An equipment blank is used to detect contamination introduced by the sampling equipment either directly or through improper cleaning. Blank water is used to fill the sampling equipment and then poured into appropriate containers.

Procedure: While working in the sample site area (e.g., truck tail gate) in which the environmental sample(s) is being collected and processed, pour the State Lab certified deionized water directly into the precleaned churn sample splitter. Run some water through the spigot and fill the sample bottle(s). If a churn splitter is not being used, pour the deionized water from its container directly into the sample bottle(s). Cap the bottle immediately and label. Store and transport the blank(s) in the same container as the environmental samples.

Submit blank sample to a certified lab and request the same analyses as the environmental samples.

3.11.10.5 Source Material

U.S. Geological Survey, Collection of Water Samples, Quality-Control Samples, TWRI Book 9, Chapter A4, 1999. The URL on the World Wide Web is http://water.usgs.gov/owq/FieldManual/Chapter4/html/4.3.1.html .

3.11.11 Ambient Split Samples

Split samples are used in the ambient surface water monitoring programs to check laboratory handling and analytical procedures. Splits may be sent to the same lab, or preferably, to different labs. If sent to the same lab, the split sample bottles should be labeled such that they cannot be identified with the environmental samples (see Section 3.11.13).

Split samples are a set of identical samples collected from the same source at the same time. They may be equated to "identical twins" in that they each contain the exact same chemical makeup. They may be submitted to the same or different laboratories and should yield identical analytical results.

Collection Method

1. Splits are usually taken from a large sample compositor (churn splitter) that has been filled with numerous subsamples from the source. A 14-liter churn splitter should be used for split samples.

2. The composited sample is thoroughly mixed before withdrawing subsamples into two distinct chemsets of sample bottles for laboratory analysis.

3. Label appropriately, store and transport the splits in the same container as the environmental samples.

3.11.12 Ambient Duplicate Samples

Duplicate samples are used in the ambient surface water monitoring program to check laboratory handling and analytical procedures when splits are not practicable. They are collected at the same time as the environmental samples and may be equated to "fraternal twins." While they are not as accurate an indicator of potential problems as split samples, there are times when split samples are not feasible (when collecting organics, VOCs or grab samples), and duplicate samples are the best option. Duplicates may be sent to the same lab, or preferably to different labs. If sent to the same lab, the duplicate sample bottles should be labeled such that they cannot be identified with the environmental samples (see Section 3.11.13).

If a four or five bottle chemset is being collected for the environmental samples, the same chemset is collected for the duplicate samples, unless otherwise directed by the sample plan. It is advisable to collect paired chemset types in succession. For instance, fill the two unpreserved sample bottles as close in time as possible and in the same location.

Collection method

1. Using a precleaned collection bottle, collect one liter of sample water directly from the stream source.

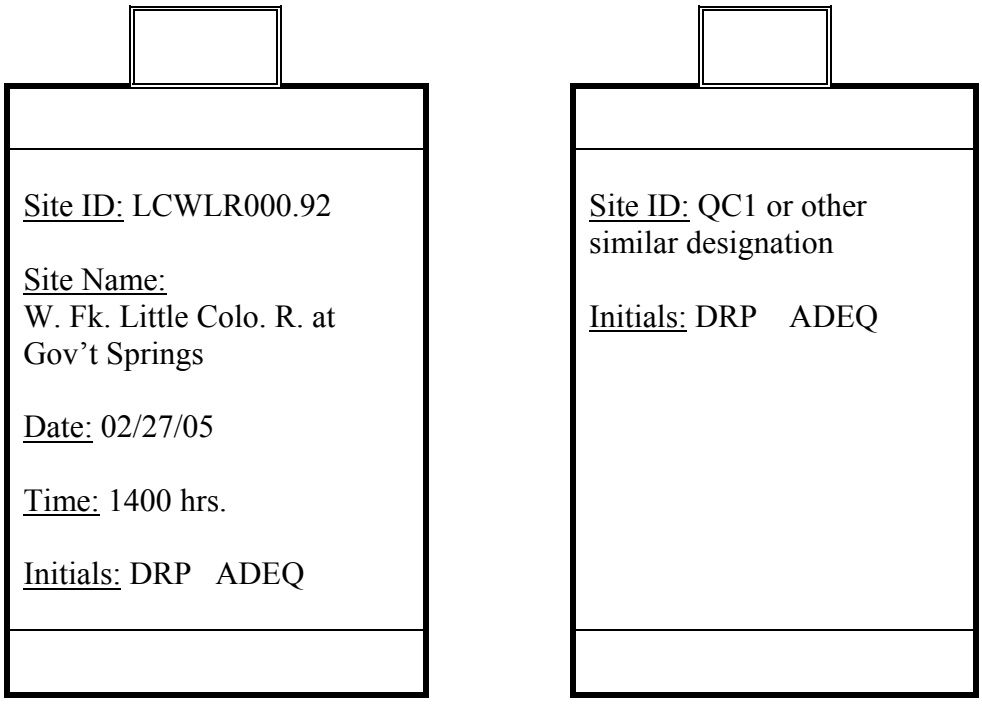
2. Pour the collected water into a sample chemset bottle.

3. If more than one analyte is being sampled for quality control purposes and additional modes of preservation are required, repeat steps 1 and 2 until necessary chemset bottles have been filled.

3.11.13 QC Sample Bottle Labeling

If both sample sets are to be analyzed by the same laboratory**,** the blank, split, or duplicate sample bottle set is labeled as a blind sample. This keeps the laboratory from biasing or correcting their analytical results based on what constituent concentrations they think should be in the sample. The labeling may look like the following example.

If the sample sets are to be analyzed by different laboratories, the QC sample bottle chemset should be labeled identical to the environmental sample bottle chemset.



3.12 Hach Colorimeter for Chlorine Analysis

This procedure describes how to measure free and total chlorine with the Hach DR / 700 Colorimeter (Figure 11).

Equipment Required: HACH Model DR/700 Colorimeter, HACH DPD Free Chlorine Reagent Powder Pillows, HACH DPD Total Chlorine Reagent Powder Pillows, two 20 ml sample vials, cotton cloth, and large nail clippers or scissors.

Figure 11. Hach DR / 700 Colorimeter.

3.12.1 Field Procedure for Measuring Free Available Chlorine

1. After powering on the instrument, a six digit number, such as 52.05.01, will appear at the bottom of the display window. This is the number of one of several filter modules that are factory installed. The filter module number required for the free chlorine analysis is 52.07.1. If a number other than the free chlorine module is displayed, press the up arrow key, in the edit box, until the correct number appears.

2. Inspect both sample vials provided with the meter for any discoloration of the glass surface or any evaporate remaining from the last analysis. The two vials should have been thoroughly washed with a chlorine free detergent and rinsed with deionized water before field use.

3. Rinse out the vial one or more times with sample water. Fill both sample vials to the 10-ml line with sample water and cap (Figure 12).

   One vial will be the calibration blank used for both the free and total chlorine measurements; the second vial is also used for the two analyses. Any fingerprints or water droplets on the outside surface must be wiped clean with a soft lint-free cotton cloth. Handle vials by the cap to avoid fingerprints. Do not use scratched vials.

Figure 12. Vials filled to the 10 mL line with sample water.

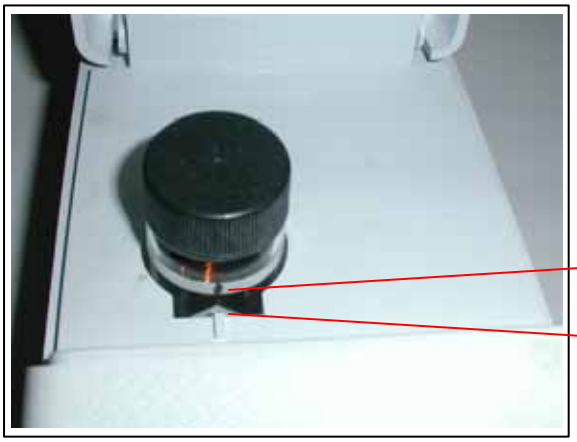


Figure 13. Aligning the vial to the meter for the optimum optical measurement.

Vertical line

Raised index

4. Place the blank sample in the vial holder of the instrument. Each sample vial should have been optically matched when the instrument was calibrated (Refer to the Hach Instrument Manual for this procedure). Vertical lines on the upper white band indicate the optimum position for measurement as determined by the optical calibration (Figure 13). Orient the positioning mark on the upper white band to the raised index structure on the vial holder. Close the cover.

5. Press **ZERO**. The display will count down to zero and the display will read 0.00 mg/l. The colorimeter is now calibrated to the sample water and ready for the first chlorine analysis.

6. Add the contents of one DPD free chlorine powder pillow to the second vial containing sample water. Cap the vial and shake for 20 seconds. A pink color will develop if free chlorine is present.

7. Within one minute of agitating the vial, place the prepared sample into the sample measurement compartment, aligning the vertical line on the upper white band with the raised index structure (Figure 13), and close the cover.

8. Press **READ**. The display will count down to zero and show the results in mg/l of free chlorine. Record the measurement on the Field Data Sheets.

3.12.2 Field Procedure for Measuring Total Chlorine

1. Change the chlorine filter module from 52.05.01 to 52.07.1 by pressing the up arrow key.

2. The meter needs to be recalibrated for the new filter module. Place the blank sample (same one used in the free chlorine analysis) in the vial holder and close the cover.

3. Press **ZERO**. The display will count down to zero and the display will show 0.00 mg/l. The colorimeter is now ready for the prepared sample.

4. Discard the sample used for the free chlorine measurement and rinse the vial with sample water several times to remove the remains of the first analysis. Fill to the 10-ml line with sample water and add the contents of one DPD total chlorine powder pillow to the sample vial.

5. Cap the vial and shake for 20 seconds. A pink color will develop if total chlorine is present.

6. Within three minutes, insert the prepared sample into the sample compartment. Align the calibration mark on the upper white band to the raised index structure on the vial holder.

7. Press **READ**. The display will count down to zero and the display will show the results in mg/l of total chlorine.

3.12.3 Source Material

The chlorine procedures have been adapted from the Hach Instrument Manual provided with the instrument.

Hach Company. 1993. Model DR/700 Portable Colorimeter Instrument Manual. Loveland, CO.

3.13 Bacteria Collection

This section describes the procedure for examining water samples for sanitary quality. The methods are intended to indicate the degree of contamination. The detection and enumeration of *E. coli* is the indicator organism for the community of pathogens commonly found in intestinal derived wastes.

Equipment Required for each sample site: Two six-ounce sterilized NASCO WHIRL-PAK© bags, one wide mouth collection bottle, a cooler and ice, powder-free Latex or Nitrile gloves (optional), ADHS prepared snaplid plates containing mTEC or similar media, sterile buffer dilution water, a control culture of *E. coli*, a wooden inoculation stick, urea broth, three empty Petri dishes, Nutropads, biohazard bags, two incubators, two sets of two incubator cords (one for DC and one for AC outlets), two stainless steel forceps, a small bottle containing alcohol, matches and a candle or butane lighter, filtering apparatus (one filter base, a middle adapter, and a funnel), membrane filters (0.45 μm, 47 mm, white, gridded), one portable peristaltic pump (AC/DC) or one hand pump, site field data sheets, one sterile polystyrene 10 ml in 1/10 serological pipette (Figure14), and one 50 milliliter syringe.

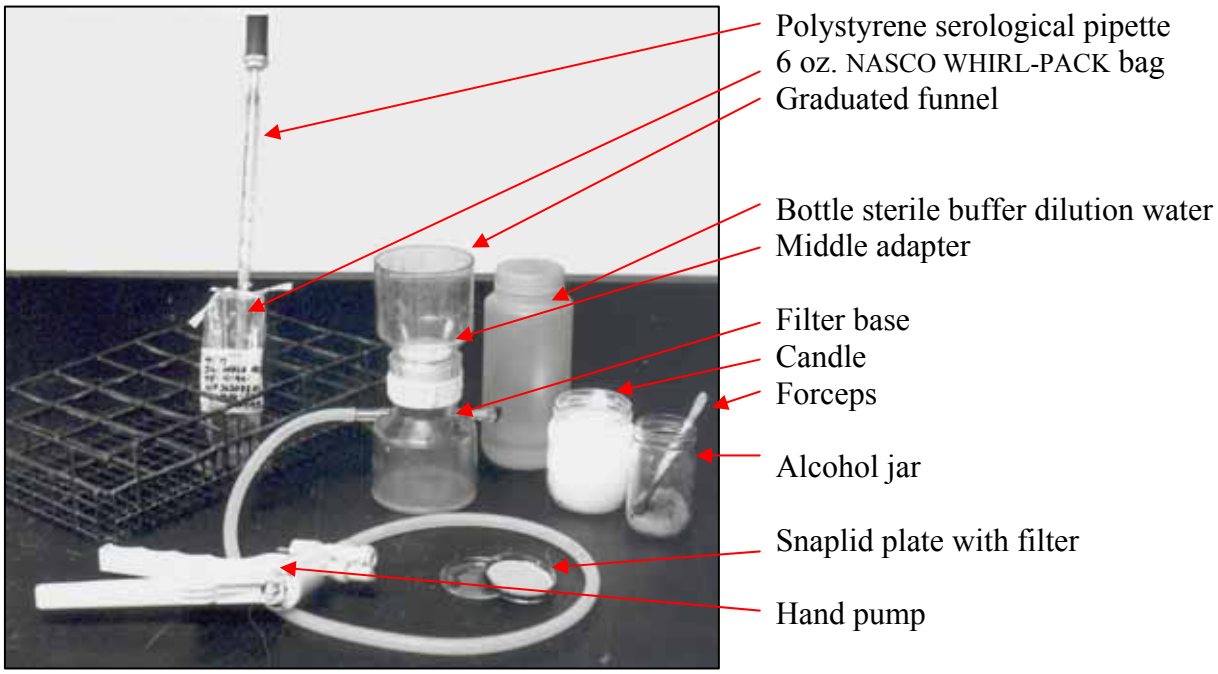


Figure 14. Typical bacteria processing set-up.

3.13.1 Field Trip Preparation

It is necessary to submit requests for bacterial analysis supplies to the ADEQ Laboratory Coordinator at least one week prior to the sampling trip. The Fixed Station Network (FSN) Program uses "snaplid plates." The media currently used for the analysis of *E. coli* bacteria is mTEC.

The media and control culture must be refrigerated and kept in the dark immediately after being obtained from the Arizona Department of Health Services (ADHS) Laboratory. Media should not be used beyond the expiration date.

Field incubators should be turned on prior to trip departure if any sample processing will be performed that day. Incubators must be "at temperature" by the time filtering is completed. Check incubators periodically to ensure that they are turned on and functioning properly.

3.13.2 Collection Procedure

Two six-ounce sterilized NASCO WHIRL-PAK© bags (or other similar product) are used to collect bacteria samples from the source water. Each bag contains a dechlorinating (sodium thiosulfate) tablet to neutralize chlorinated water. Two duplicate samples are taken at each site. Caution: Depending on the severity of pollution of the sample waters, it may be necessary to wear gloves while collecting the sample.

1. Label the WHIRL-PAK bags with the site identification using an indelible pen.

2. Tear off the tab at the top of the bag at the perforation line. Do not touch the inside of the bag.

3. Stand in the stream, facing upstream, at a location which is representative of the entire flow and upstream of any streambed disturbance.

4. Grasp the two small paper tabs on each side of the center of the bag.

5. With the bag still closed, submerge the bag to a depth which is approximately half the water column. With the open end of the bag facing upstream, move the bag in an upstream motion while simultaneously opening it with the paper tabs. This should be done in one continuous motion.

6. If the collected water is above the “fill line,” dispense the overflow back into the stream.

7. Using both hands, grasp the wire tabs on the sides of the bag and whirl the bag so that it winds around the wire tabs 1 to 3 times (face away from the sample while whirling the bag to avoid contaminating the face with stream water). Twist the wire tabs around each other with the tab ends pointed away from the bag to prevent bag puncture and leakage while in storage and transport.

3.13.3 Storing/Transporting Procedure

Place the two samples in a wide mouth collection bottle to insure their safety during transport. The collection bottle should be kept on ice until filtration is performed. The ideal temperature for preservation is 4 °C. Exposure of bacteria to direct sunlight should be minimized. If

samples are to be processed within one hour of collection, chilling them is unnecessary, however, it is still important to keep them out of direct light.

Bacteria samples should be processed within the six hour holding time. If the holding time is violated, note this on the Field Data Sheet and flag it in the water quality database.

3.13.4. Filtration and Plating Procedure

The wearing of powder-free Latex or Nitrile gloves is optional while filtering and processing the samples.

The FSN Program uses the membrane filter technique (MF) for processing water samples for bacteriological analysis.

1. Label the bottom of the snaplid container with an indelible pen indicating the site ID and the volume of filtered sample water.

2. The Field Data Sheets are designed for three sample volumes. The three volumes of sample water to be filtered should have been pre-determined. Typically for streams, the volumes are 10 ml, 25 ml, and 50 ml, The target volume is one that will yield 20 to 60 colonies per membrane filter. At sites with a sampling history, refer to historic Field Data Sheets at that site for filter volumes that have yielded ideal colony counts.

3. There are three parts to the filter apparatus; filter base, middle adapter, and funnel. The filter base collects the filtered water. The middle adapter fits onto the base opening and receives the screw-on funnel. The funnel is the receptacle for the sample water and sterile buffer water.

   Place a sterilized Millipore filter top and middle adapter on the filter base. The adapter and the funnel come as one unit in a sterilized package from the ADHS Laboratory. Do not touch the top surface of the middle adapter, or the inside of the funnel top. There are two spouts on the side of the base, 180° apart; one is covered with a soft plastic cap and the second is uncapped. Insert the unused sterile silicone tubing into the peristaltic pump, if an electric pump is being used. Attach one end of the tubing to the uncapped spout. The appropriate length of tubing is three to four feet. If a hand pump is being used, attach one end of the tubing to the hand pump and the other end to the uncapped base spout.

4. Pour a few milliliters of sterile buffered water onto the top of the porous surface of the middle adaptor.

5. Dip stainless steel forceps in alcohol and pass them through a flame. Let forceps cool for 10 seconds.

6. Open the sterile filter membrane package and grasp the edge of the membrane with the sterilized forceps.

7. For the 10 ml dilution, pour 20 to 50 milliliters of buffer solution into the graduated funnel. This step is not needed for dilutions of greater amounts.

8. Unscrew the funnel from the middle adapter and hold it in one hand above the work surface. With the free hand, place the membrane, grid side up, on the exposed middle adapter. Screw the filter top back in place. CAUTION: Over-tightening the filter top may cause the filter membrane to tear.

9. Vigorously shake the sample to be filtered 20 times in a large arcing motion with the elbow as the pivot point.

10. Starting with the smallest predetermined volume (e.g. 10 ml) aspirate the sample water into a 10 milliliter serological pipette and dispense to the filter funnel. For volumes greater than 10 milliliters, use the 50 milliliter syringe.

NOTE: On waters with known severe bacterial contamination where a 10 milliliter sample will likely generate a count that is to-numerous-to-count (TNTC), and smaller dilutions are required to achieve the ideal colony count, use a micro-pipette pump and 1 mL serological 1 in 1/100 mL pipette. For this small dilution volume, dispense 20 to 50 milliliters of sterile buffer water into the funnel before introducing the pipetted sample water.

10. Swirl the funnel solution after the transfer is made and filter under partial vacuum.

11. Rinse funnel walls 1 to 3 times, with 20 to 30 milliliters of buffer water. Filter each rinse separately under partial vacuum until the filter membrane appears dry. Remove the cap from the capped spout on the filter base to relieve air pressure on the filter. This will prevent the filter from tearing when the funnel top is unscrewed.

12. Unscrew filter top and hold in one hand above the work surface. Grasp the edge of the filter membrane with the sterilized forceps and drag it across the adaptor surface to remove excess water.

13. Replace the filter top on the middle adapter.

14. Place the filter membrane with the grid side up onto the mTec media in the snaplid plate. This is done by placing the filter, nearest to the forceps, on the edge of the snaplid plate. Slowly drag the filter across the snaplid edge until the opposing end of the filter falls onto the media, then lower the remaining membrane filter onto the media surface. If there are small air bubbles near the filter edge and between the filter and the media surface, carefully tap them out to the edge with the forceps. Restrict contact of the forceps to the edge of the filter surface. If large bubbles are present, remove the membrane filter from the media surface and re-apply.

15. Replace the top of the snaplid plate and record the time of incubation on the bottom of the plate and on the field notes. The snaplid plate must be placed in the incubator within 30 minutes after filtering.

16. Incubate *E. coli* plates lid down for 2 hours at 35.0 ± 0.5 °C, then transfer the plates to the warmer incubator (44.5 ± 0.2 °C) for 23 ± 2 hours.

17. Repeat the filtering process for the remaining predetermined dilution volumes.

Place used funnel, middle adaptor, syringes, and buffered dilution water bottles into a bio-hazard bag for autoclaving and used pipettes and WHIRL-PAK bags into a biohazard bag for disposal at the ADHS Laboratory.

3.13.5. Quality Assurance/Quality Control

To insure that proper procedures have been followed, an equipment blank and a technique blank should be processed each day of sample filtering.

- The equipment blank is processed before the first sample volume is filtered. This is a quality control for equipment sterility. No sample water is required. Decant 50 to 100 milliliters of sterile buffer water into the funnel and filter. The filter is incubated as described in 3.13.4, step 16
- The technique blank is processed after all samples dilutions have been processed. A technique blank is a quality control for adequate rinsing of the inside of the funnel. No sample water is required. Decant 50 to 100 milliliters of sterile buffer water into the funnel and filter. The filter is incubated as described in 3.13.4, step 16

A quality control for media viability and incubator temperature stability are monitored with cultured E. coli (positive control). This is done after all the samples have been filtered.

- The filtering apparatus, with an unused funnel, middle adaptor, and membrane filter are set up as described in the filtering and plating procedure. Pour approximately 25 milliliters of buffer water into the funnel. Open the *E. coli* inoculated test-tube, insert the end of a lab supplied wooden stick, and lightly touch the cultured media. Introduce the exposed end of the stick into the funnel buffer water and stir. Filter the solution and apply the filter to the mTec media. Incubate the filter as described in 3.13.4, step 16

After all samples have been processed and used equipment placed in the biohazard bags, thoroughly wash hands with antibacterial soap and sterilize the work surface with alcohol or antibacterial soap. The contaminated wooden stick contains viable *E. coli* and should be placed in a biohazard bag for disposal at the ADHS Laboratory. An alternative procedure is burning the contaminated portion of the stick in a flame.

3.13.6 Counting Bacteria Colonies

After the filter membranes have been incubated for the designated time period, colonies of *E. coli* may be present on the filters. Transfer the membrane filters from the mTEC media to a clean Petri dish containing an absorbent Nutropad that has been moistened, but not saturated, with urea broth. Allow the *E. coli* membranes to sit on the Nutropads at room temperature

(25 - 37 °C) for 15 to 20 minutes before counting colonies. The *E. coli* colonies will appear as a glowing light yellow mass to be differentiated from the cream or gray colored colonies that may be present.

Count colonies in each Petri dish using the grid squares on the membrane filter. Recount all colonies on a given membrane until replicate counts agree within 5% and record the results on the Field Data Sheets.

3.13.7 Computing Bacterial Densities

Results of bacterial analyses of water samples are reported in colonies forming units/100 milliliters sample. This is referred to as the "bacterial density. For bacterial analyses the ideal range is 20 and 60 "target colonies" per filter membrane.

Following are seven conditions that describe the results of counting colonies for bacterial analysis on filter membranes from single sample sites. Record all results on the Field Data Sheets.

Condition #1: Colonies on one filter membrane within the ideal range

If one of the three filter membranes contain a count within the ideal range (20 to 60), density is computed as:

$$\frac{\text{Colony Count} \times 100}{\text{Volume, mL}}$$

If the following results were obtained,

mL Filtered	Number colonies
10	5
25	33 (within the ideal range of 20 to 60)
50	87

then, $\frac{33 \times 100}{25\ \text{mL}} = 3300/25 = 132$ *E. coli* colony forming units/100mL.

Condition #2: Colonies on two filter membranes within the ideal range

If two or more filter membranes reveal counts in the ideal range, the density is calculated as:

$$\frac{(\#\ \text{of colonies membrane A} + \#\ \text{colonies membrane B}) \times 100}{\text{volume (mL) filter A} + \text{volume (mL) filter B}}$$

If the following results were obtained,

mL Filtered	Number Colonies
10	2
25	23 (within the ideal range)
50	56 (within the ideal range)

then, $\frac{(23 + 56) \times 100}{50 + 25 \text{ mL}} = 7900/75 = 105$ *E. coli* colony forming units/100mL

Condition #3: Colonies present on more than one filter membrane and all counts are out of the ideal range

If all filter membranes have colony counts outside the ideal range (20 to 60), but within the 1 to 200 range, calculate as:

$$\frac{(\#\text{ of colonies membrane A} + \#\text{ colonies membrane B} + \#\text{ of colonies on membrane C}) \times 100}{\text{volume (mL) filter A} + \text{volume (mL) filter B} + \text{volume (mL) filter C}}$$

If the following results were obtained,

mL filtered	# colonies
10	5
25	19
50	67

then, $\frac{(18 + 65 + 123) \times 100}{10 + 25 + 50 \text{ mL}} = 9100/85 = 107$ *E. coli* colonies/100mL

When entering the data into the Water Quality Database, us the data qualifier B (Colony counts outside acceptable range {20 – 60 CFU}).

Condition #4: Colonies on one or more filter membranes have a count less than the ideal range

Filters having a zero count are not included in the calculation. Compute the density as in Example #3.

If the following results were obtained,

mL filtered	# colonies
10	0
25	2
50	11

then, $\frac{(2 + 11) \times 100}{25 + 50 \text{ mL}} = 1300/75 = 17$ *E. coli* colony forming units/100mL

Condition #5: All filter membranes contain more than 200 colonies

When entering the data into the Water Quality Database, us the data qualifier A1 (TNTC; too numerous to count).

Condition #6: There are no colonies on any filter membranes

Reporting results are determined by the largest volume dilution used for the analysis. If the volumes used for the analysis were 10 mL, 25 mL, and 50 mL and no colonies were observed, the calculation is completed as follows:

A count of one is assigned to the 50 mL dilution and the calculation is

$$\frac{1 \times 100}{50 \text{ mL}} = 100/50 = 2 \text{ } \underline{E.} \text{ } \underline{coli} \text{ colonies/100mL.}$$

When entering the results in the Water Quality Database, the data qualifier ND (none detected) is assigned to the calculated value.

3.13.8 Disposal of used Materials and Personal Hygiene

Replace the lid on the snaplid plates and place those plates and the Petri dishes into a biohazard bag for disposal at the ADHS Laboratory.

Thoroughly wash hands with an antibacterial soap.

3.13.9. Source Material

American Public Health Association. 1995. Standard Methods for the Examination of Water and Wastewater. Washington, D.C.

3.13.10 Colilert® System

The Colilert system utilizes prepackaged reagents which include nutrients to support the growth of coliform bacteria in addition to specific compounds that react with coliforms in general and *E. coli* specifically. Colilert-18 is used for the simultaneous detection and confirmation of total coliforms and *E. coli* in fresh waters. When total coliforms metabolize Colilert-18's nutrient indicator reagent, ONPG (O-Nitrophenyl-β-d-galacotpyranoside), the reaction produces an easily recognized yellow color. When *E. coli* metabolizes Colilert-18 nutrient-indicator, MUG (4-Methumbelliferyl-β-d-glucoronide) the sample fluoresces. Colilert-18 can simultaneously detect these bacteria at 1 cfu/100 ml within 18 hours. The test is effective and free of interference in waters with population densities of other heterotrophic bacteria up to 10,000 cfu/100 ml. Non-

coliform bacteria that also have these enzymes are suppressed, for the incubation period, by other reagents in the media.

EPA refers to Colilert® as MMO-MUG and Standard Methods for the Examination of Water and Wastewater calls it a chromogenic substrate.

Materials: Catalog #WP020-18 and WP200-18 contain 20 and 200 Snap Packs respectively, each containing sufficient Colilert-18 reagent for a 100 ml water sample. The reagents should be stored at 4-25°C away from light.

3.13.10.1 Presence/Absence Procedure

1. Add contents of one pack to a 100 ml sample in a sterile, transparent, non-fluorescing vessel.
2. Cap vessel and shake.
3. If sample is not already at 33-38°C, then place vessel in a 35°C waterbath for 20 minutes or, alternatively, a 44.6 °C waterbath for a minimum of 7 and maximum of 10 minutes.
4. Incubate at 35 ±0.5 °C for the remainder of 18 hours.
5. Read results according to the Result Interpretation Table 2.

Table 2. Result Interpretation Table for Presence/Absence Procedure

Appearance	Result
Colorless or slight tinge	Negative for total coliforms and *E. coli*
Yellow equal to or greater than the comparator	Positive for total coliforms
Yellow and fluorescence equal to or greater than the comparator	Positive for *E. coli*

Note: 1. Look for fluorescence with a 6 watt, 365 nm, UV light within 5 inches (13 cm) of the sample. Face light away from your eyes and towards the sample.

2. Samples are negative if at any time after 18 hours there is no yellow and/or fluorescence.

3. Yellow or yellow/fluorescence observed before 18 hours is a valid positive. However, after 22 hours from inoculation, heterotrophs may overwhelm Colilert-18's inhibition system. Therefore, yellow or yellow/fluorescence first observed after 22 hours from inoculation is not a valid positive.

3.13.10.2 Quanti-Tray Enumeration Procedure

1. Add contents of one pack to a 100 ml, room temperature water sample in a sterile vessel. Vessel does not need to be transparent or non-fluorescing.
2. Cap vessel and shake until dissolved.
3. Pour sample/reagent mixture into a Quanti-Tray/2000 and seal in an IDEXX Quanti-Tray Sealer.

4. Place the sealed Tray in a 35 ±0.5°C incubator for 18 hours (this 18 hours includes the warming time).
5. Read results according to the Result Interpretation Table 2. Count the number of positive wells and refer to the MPN table (Table 3) provided with the Trays to obtain a Most Probable Number.

3.13.10.3 Quanti-Tray User Instructions

1. Use one hand to hold a Quanti-Tray upright with the well side facing the palm.
2. Squeeze the upper part of the Quanti-Tray so that the Quanti-Tray bends towards the palm.
3. Open the Quanti-Tray by pulling the foil tab away from the well side. Avoid touching the inside of the foil or tray.
4. Pour the reagent/sample mixture directly into the Quanti-Tray avoiding contact with the foil tab. Allow foam to settle.
5. Place the sample-filled Quanti-Tray onto the rubber tray carrier of the Quanti-Tray Sealer with the well side (plastic) of the Quanti-Tray facing down to fit into the carrier.
6. Seal according to Sealer instructions.
7. Incubate according to reagent directions.
8. Count positive wells and refer to Table 3 (MPN table) to find the Most Probable Number (MPN).
9. Dispose of media in accordance with good laboratory practices.

3.13.10.4 Quanti-Tray/2000 User Instructions

1. Use one hand to hold a Quanti-Tray upright with the well side facing the palm.
2. Squeeze the upper part of the Quanti-Tray so that the Quanti-Tray bends towards the palm.
3. Gently pull foil tab to separate the foil from the tray. Avoid touching the inside of the foil or tray.
4. Pour the reagent/sample mixture directly into the Quanti-Tray avoiding contact with the foil tab. Tap the small wells 2-3 times to release any air bubbles. Allow foam to settle.
5. Place the sample-filled Quanti-Tray onto the Quanti-Tray/2000 rubber insert of the Quanti-Tray Sealer with the well side (plastic) of the Quanti-Tray facing down.

Table 3. Most Probable Number Table for Quanti-Tray.

No. of wells giving positive reaction per 100 ml sample	Most Probable Number	95% Confidence Limits	
		Lower	Upper
<1	0.0	0.0	3.7
1	1.0	0.3	5.6
2	2.0	0.6	7.3
3	3.1	1.1	9.0
4	4.2	1.7	10.7
5	5.3	2.3	12.3
6	6.4	3.0	13.9
7	7.5	3.7	15.5
8	8.7	4.5	17.1
9	9.9	5.3	18.8
10	11.1	6.1	20.5
11	12.4	7.0	22.1
12	13.7	7.9	23.9
13	15.0	8.8	25.7
14	16.4	9.8	27.5
15	17.8	10.8	29.4
16	19.2	11.9	31.3
17	20.7	13.0	33.3
18	22.2	14.1	35.2
19	23.8	15.3	37.3
20	25.4	16.5	39.4
21	27.1	17.7	41.6
22	28.8	19.0	43.9
23	30.6	20.4	46.3
24	32.4	21.8	48.7
25	34.4	23.3	51.2
26	36.4	24.7	53.9
27	38.4	26.4	56.6
28	40.6	28.0	59.5
29	42.9	29.7	62.5
30	45.3	31.5	65.6
31	47.8	33.4	69.0
32	50.4	35.4	72.5
33	53.1	37.5	76.2
34	56.0	39.7	80.1
35	59.1	42.0	84.4
36	62.4	44.6	88.8
37	65.9	47.2	93.7
38	69.7	50.0	99.0
39	73.8	53.1	104.8
40	78.2	56.4	111.2
41	83.1	59.9	118.3
42	88.5	63.9	126.2
43	94.5	68.2	135.4
44	101.3	73.1	146.0
45	109.1	78.6	158.7
46	118.4	85.0	174.5
47	129.8	92.7	195.0
48	144.5	102.3	224.1
49	165.2	115.2	272.2
50	200.8	135.8	387.6
51	>200.5	146.1	infinite

6. Seal according to Sealer instructions.
7. Incubate according to reagent directions.
8. Count large and small positive wells and refer to the Quanti-Tray/2000 MPN table (Table 4) to find the Most Probable Number (MPN).
9. Dispose of media in accordance with good laboratory practices.

3.13.10.5 Procedural Notes

A slight tinge may be observed when Colilert-18 is added to the sample.

Colilert-18 can be run in any multiple tube format. Standard Methods for the Examination of Water and Wastewater MPN tables should be used to find Most Probable Number.

Some water samples containing humic material may have an innate color. If a water sample has some background color, compare inoculated Colilert-18 sample to a control blank of the same water sample.

Do not dilute sample in buffered water. Colilert-18 is already buffered.

Colilert-18 is a primary water test. Colilert-18 performance characteristics do not apply to samples altered by any pre-enrichment or concentration.

In samples with excessive chlorine, a blue flash may be seen when adding Colilert-18. If this is seen, consider sample invalid and discontinue testing.

Aseptic technique should be always followed when using Colilert-18. Dispose of in accordance with good laboratory practices.

3.13.10.6 Quality Control Procedures

The following quality control procedure is recommended for each lot of Colilert-18, or more often as regulation require.

1. Inoculate 3 sterile vessels filled with 100 ml sterile water with the following:
    A. one with Quanti-Cult** *E. coli* or a sterile loop of ATCC***25922 or 11775 (*E. coli*)
    B. one with Quanti-Cult *Klebsiella pneumoniae* or a sterile loop of ATCC 31488 (total coliform)
    C. one with Quanti-Cult *Pseudomonas aeruginosa* or a sterile loop of ATCC 10145 or 27853 (non-coliform).
2. Follow the P/A Procedure or Quanti-Tray Enumeration Procedure above.
3. Results should match the Result Interpretation.

3.13.10.7 Source Material

www.IDEXX.com.

Table 4. IDEXX Quanti-Tray/2000 MPN Table.

IDEXX Quanti-Tray®/2000 MPN Table (per 100ml)

# Large Wells Positive	# Small Wells Positive																								
	0	1	2	3	4	5	6	7	8	9	10	11	12	13	14	15	16	17	18	19	20	21	22	23	24
0	<1	1.0	2.0	3.0	4.0	5.0	6.0	7.0	8.0	9.0	10.0	11.0	12.0	13.0	14.1	15.1	16.1	17.1	18.1	19.1	20.2	21.2	22.2	23.3	24.3
1	1.0	2.0	3.0	4.0	5.0	6.0	7.1	8.1	9.1	10.1	11.1	12.1	13.2	14.2	15.2	16.2	17.3	18.3	19.3	20.4	21.4	22.4	23.5	24.5	25.6
2	2.0	3.0	4.1	5.1	6.1	7.1	8.1	9.2	10.2	11.2	12.2	13.3	14.3	15.4	16.4	17.4	18.5	19.5	20.6	21.6	22.7	23.7	24.8	25.8	26.9
3	3.1	4.1	5.1	6.1	7.2	8.2	9.2	10.3	11.3	12.4	13.4	14.5	15.5	16.5	17.6	18.6	19.7	20.8	21.8	22.9	23.9	25.0	26.1	27.1	28.2
4	4.1	5.2	6.2	7.2	8.3	9.3	10.4	11.4	12.5	13.5	14.6	15.6	16.7	17.8	18.8	19.9	21.0	22.0	23.1	24.2	25.3	26.3	27.4	28.5	29.6
5	5.2	6.3	7.3	8.4	9.4	10.5	11.5	12.6	13.7	14.7	15.8	16.9	17.9	19.0	20.1	21.2	22.2	23.3	24.4	25.5	26.6	27.7	28.8	29.9	31.0
6	6.3	7.4	8.4	9.5	10.6	11.6	12.7	13.8	14.9	16.0	17.0	18.1	19.2	20.3	21.4	22.5	23.6	24.7	25.8	26.9	28.0	29.1	30.2	31.3	32.4
7	7.5	8.5	9.6	10.7	11.8	12.8	13.9	15.0	16.1	17.2	18.3	19.4	20.5	21.6	22.7	23.8	24.9	26.0	27.1	28.3	29.4	30.5	31.6	32.8	33.9
8	8.6	9.7	10.8	11.9	13.0	14.1	15.2	16.3	17.4	18.5	19.6	20.7	21.8	22.9	24.1	25.2	26.3	27.4	28.6	29.7	30.8	32.0	33.1	34.3	35.4
9	9.8	10.9	12.0	13.1	14.2	15.3	16.4	17.6	18.7	19.8	20.9	22.0	23.2	24.3	25.4	26.6	27.7	28.9	30.0	31.2	32.3	33.5	34.6	35.8	37.0
10	11.0	12.1	13.2	14.4	15.5	16.6	17.7	18.9	20.0	21.1	22.3	23.4	24.6	25.7	26.9	28.0	29.2	30.3	31.5	32.7	33.8	35.0	36.2	37.4	38.6
11	12.2	13.4	14.5	15.6	16.8	17.9	19.1	20.2	21.4	22.5	23.7	24.8	26.0	27.2	28.3	29.5	30.7	31.9	33.0	34.2	35.4	36.6	37.8	39.0	40.2
12	13.5	14.6	15.8	16.9	18.1	19.3	20.4	21.6	22.8	23.9	25.1	26.3	27.5	28.6	29.8	31.0	32.2	33.4	34.6	35.8	37.0	38.2	39.5	40.7	41.9
13	14.8	16.0	17.1	18.3	19.5	20.6	21.8	23.0	24.2	25.4	26.6	27.8	29.0	30.2	31.4	32.6	33.8	35.0	36.2	37.5	38.7	39.9	41.2	42.4	43.6
14	16.1	17.3	18.5	19.7	20.9	22.1	23.3	24.5	25.7	26.9	28.1	29.3	30.5	31.7	33.0	34.2	35.4	36.7	37.9	39.1	40.4	41.6	42.9	44.2	45.4
15	17.5	18.7	19.9	21.1	22.3	23.5	24.7	25.9	27.2	28.4	29.6	30.9	32.1	33.3	34.6	35.8	37.1	38.4	39.6	40.9	42.2	43.4	44.7	46.0	47.3
16	18.9	20.1	21.3	22.6	23.8	25.0	26.2	27.5	28.7	30.0	31.2	32.5	33.7	35.0	36.3	37.5	38.8	40.1	41.4	42.7	44.0	45.3	46.6	47.9	49.2
17	20.3	21.6	22.8	24.1	25.3	26.6	27.8	29.1	30.3	31.6	32.9	34.1	35.4	36.7	38.0	39.3	40.6	41.9	43.2	44.5	45.9	47.2	48.5	49.8	51.2
18	21.8	23.1	24.3	25.6	26.9	28.1	29.4	30.7	32.0	33.3	34.6	35.9	37.2	38.5	39.8	41.1	42.4	43.8	45.1	46.5	47.8	49.2	50.5	51.9	53.2
19	23.3	24.6	25.9	27.2	28.5	29.8	31.1	32.4	33.7	35.0	36.3	37.6	39.0	40.3	41.6	43.0	44.3	45.7	47.1	48.4	49.8	51.2	52.6	54.0	55.4
20	24.9	26.2	27.5	28.8	30.1	31.5	32.8	34.1	35.4	36.8	38.1	39.5	40.8	42.2	43.6	44.9	46.3	47.7	49.1	50.5	51.9	53.3	54.7	56.1	57.6
21	26.5	27.9	29.2	30.5	31.8	33.2	34.5	35.9	37.3	38.6	40.0	41.4	42.8	44.1	45.5	46.9	48.4	49.8	51.2	52.6	54.1	55.5	56.9	58.4	59.9
22	28.2	29.5	30.9	32.3	33.6	35.0	36.4	37.7	39.1	40.5	41.9	43.3	44.8	46.2	47.6	49.0	50.5	51.9	53.4	54.8	56.3	57.8	59.3	60.8	62.3
23	29.9	31.3	32.7	34.1	35.5	36.8	38.3	39.7	41.1	42.5	43.9	45.4	46.8	48.3	49.7	51.2	52.7	54.2	55.6	57.1	58.6	60.2	61.7	63.2	64.7
24	31.7	33.1	34.5	35.9	37.3	38.8	40.2	41.7	43.1	44.6	46.0	47.5	49.0	50.5	52.0	53.5	55.0	56.5	58.0	59.5	61.1	62.6	64.2	65.8	67.3
25	33.6	35.0	36.4	37.9	39.3	40.8	42.2	43.7	45.2	46.7	48.2	49.7	51.2	52.7	54.3	55.8	57.3	58.9	60.5	62.0	63.6	65.2	66.8	68.4	70.0
26	35.5	36.9	38.4	39.9	41.4	42.8	44.3	45.9	47.4	48.9	50.4	52.0	53.5	55.1	56.7	58.2	59.8	61.4	63.0	64.7	66.3	67.9	69.6	71.2	72.9
27	37.4	38.9	40.4	42.0	43.5	45.0	46.5	48.1	49.6	51.2	52.8	54.4	56.0	57.6	59.2	60.8	62.4	64.1	65.7	67.4	69.1	70.8	72.5	74.2	75.9
28	39.5	41.0	42.6	44.1	45.7	47.3	48.8	50.4	52.0	53.6	55.2	56.9	58.5	60.2	61.8	63.5	65.2	66.9	68.6	70.3	72.0	73.7	75.5	77.3	79.0
29	41.7	43.2	44.8	46.4	48.0	49.6	51.2	52.8	54.5	56.1	57.8	59.5	61.2	62.9	64.6	66.3	68.0	69.8	71.5	73.3	75.1	76.9	78.7	80.5	82.4
30	43.9	45.5	47.1	48.7	50.4	52.0	53.7	55.4	57.1	58.8	60.5	62.2	64.0	65.7	67.5	69.3	71.0	72.9	74.7	76.5	78.3	80.2	82.1	84.0	85.9
31	46.2	47.9	49.5	51.2	52.9	54.6	56.3	58.1	59.8	61.6	63.3	65.1	66.9	68.7	70.5	72.4	74.2	76.1	78.0	79.9	81.8	83.7	85.7	87.6	89.6
32	48.7	50.4	52.1	53.8	55.6	57.3	59.1	60.9	62.7	64.5	66.3	68.2	70.0	71.9	73.8	75.7	77.6	79.5	81.5	83.5	85.4	87.5	89.5	91.5	93.6
33	51.2	53.0	54.8	56.5	58.3	60.2	62.0	63.8	65.7	67.6	69.5	71.4	73.3	75.2	77.2	79.2	81.2	83.2	85.2	87.3	89.3	91.4	93.6	95.7	97.8
34	53.9	55.7	57.6	59.4	61.3	63.1	65.0	67.0	68.9	70.8	72.8	74.8	76.8	78.8	80.8	82.9	85.0	87.1	89.2	91.4	93.5	95.7	97.9	100.2	102.4
35	56.8	58.6	60.5	62.4	64.4	66.3	68.3	70.3	72.3	74.3	76.3	78.4	80.5	82.6	84.7	86.9	89.1	91.3	93.5	95.7	98.0	100.3	102.6	105.0	107.3
36	59.8	61.7	63.7	65.7	67.7	69.7	71.7	73.8	75.9	78.0	80.1	82.3	84.5	86.7	88.9	91.2	93.5	95.8	98.1	100.5	102.9	105.3	107.7	110.2	112.7
37	62.9	65.0	67.0	69.1	71.2	73.3	75.4	77.6	79.8	82.0	84.2	86.5	88.8	91.1	93.4	95.8	98.2	100.6	103.1	105.6	108.1	110.7	113.3	115.9	118.6
38	66.3	68.4	70.6	72.7	74.9	77.1	79.4	81.6	83.9	86.2	88.6	91.0	93.4	95.8	98.3	100.8	103.4	105.9	108.6	111.2	113.9	116.6	119.4	122.2	125.0
39	70.0	72.2	74.4	76.7	78.9	81.3	83.6	86.0	88.4	90.9	93.4	95.9	98.4	101.0	103.6	106.3	109.0	111.8	114.6	117.4	120.3	123.2	126.1	129.2	132.2
40	73.8	76.2	78.5	80.9	83.3	85.7	88.2	90.8	93.3	95.9	98.5	101.2	103.9	106.7	109.5	112.4	115.3	118.2	121.2	124.3	127.4	130.5	133.7	137.0	140.3
41	78.0	80.5	83.0	85.5	88.0	90.6	93.3	95.9	98.7	101.4	104.3	107.1	110.0	113.0	116.0	119.1	122.2	125.4	128.7	132.0	135.4	138.8	142.3	145.9	149.5
42	82.6	85.2	87.8	90.5	93.2	96.0	98.8	101.7	104.6	107.6	110.6	113.7	116.9	120.1	123.4	126.7	130.1	133.6	137.2	140.8	144.5	148.3	152.2	156.1	160.2
43	87.6	90.4	93.2	96.0	99.0	101.9	105.0	108.1	111.2	114.5	117.8	121.1	124.6	128.1	131.7	135.4	139.1	143.0	147.0	151.0	155.2	159.4	163.8	168.2	172.8
44	93.1	96.1	99.1	102.2	105.4	108.6	111.9	115.3	118.7	122.3	125.9	129.6	133.4	137.4	141.4	145.5	149.7	154.1	158.5	163.1	167.9	172.7	177.7	182.9	188.2
45	99.3	102.5	105.8	109.2	112.6	116.2	119.8	123.6	127.4	131.4	135.4	139.6	143.9	148.3	152.9	157.6	162.4	167.4	172.6	178.0	183.5	189.2	195.1	201.2	207.5
46	106.3	109.8	113.4	117.2	121.0	125.0	129.1	133.3	137.6	142.1	146.7	151.5	156.5	161.6	167.0	172.5	178.2	184.2	190.4	196.8	203.5	210.5	217.8	225.4	233.3
47	114.3	118.3	122.4	126.6	130.9	135.4	140.1	145.0	150.0	155.3	160.7	166.4	172.3	178.5	185.0	191.8	198.9	206.4	214.2	222.4	231.0	240.0	249.5	259.5	270.0
48	123.9	128.4	133.1	137.9	143.0	148.3	153.9	159.7	165.8	172.2	178.9	186.0	193.5	201.4	209.8	218.7	228.2	238.2	248.9	260.3	272.3	285.1	298.7	313.0	328.2
49	135.5	140.8	146.4	152.3	158.5	165.0	172.0	179.3	187.2	195.6	204.6	214.3	224.7	235.9	248.1	261.3	275.5	290.9	307.6	325.5	344.8	365.4	387.3	410.6	435.2

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IDEXX Quanti-Tray®/2000 MPN Table *(per 100ml)*

# Large Wells Positive	# Small Wells Positive																							
	25	26	27	28	29	30	31	32	33	34	35	36	37	38	39	40	41	42	43	44	45	46	47	48
0	25.3	26.4	27.4	28.4	29.5	30.5	31.5	32.6	33.7	34.7	35.7	36.8	37.8	38.9	40.0	41.0	42.1	43.1	44.2	45.3	46.3	47.4	48.5	49.5
1	26.6	27.7	28.7	29.8	30.8	31.9	32.9	34.0	35.0	36.1	37.2	38.2	39.3	40.4	41.4	42.5	43.6	44.7	45.7	46.8	47.9	49.0	50.1	51.2
2	27.9	29.0	30.0	31.1	32.2	33.2	34.3	35.4	36.5	37.5	38.6	39.7	40.8	41.9	43.0	44.0	45.1	46.2	47.3	48.4	49.5	50.6	51.7	52.8
3	29.3	30.4	31.4	32.5	33.6	34.7	35.8	36.8	37.9	39.0	40.1	41.2	42.3	43.4	44.5	45.6	46.7	47.8	48.9	50.0	51.2	52.3	53.4	54.5
4	30.7	31.8	32.8	33.9	35.0	36.1	37.2	38.3	39.4	40.5	41.6	42.8	43.9	45.0	46.1	47.2	48.3	49.5	50.6	51.7	52.9	54.0	55.1	56.3
5	32.1	33.2	34.3	35.4	36.5	37.6	38.7	39.9	41.0	42.1	43.2	44.4	45.5	46.5	47.7	48.9	50.0	51.2	52.3	53.5	54.6	55.8	56.9	58.1
6	33.5	34.7	35.8	36.9	38.0	39.2	40.3	41.4	42.6	43.7	44.8	46.0	47.1	48.3	49.4	50.6	51.7	52.9	54.1	55.2	56.4	57.6	58.7	59.9
7	35.0	36.2	37.3	38.4	39.6	40.7	41.9	43.0	44.2	45.3	46.5	47.7	48.8	50.0	51.2	52.3	53.5	54.7	55.9	57.1	58.3	59.4	60.6	61.8
8	36.6	37.7	38.9	40.0	41.2	42.3	43.5	44.7	45.9	47.0	48.2	49.4	50.6	51.8	53.0	54.1	55.3	56.5	57.7	59.0	60.2	61.4	62.6	63.8
9	38.1	39.3	40.5	41.6	42.8	44.0	45.2	46.4	47.6	48.8	50.0	51.2	52.4	53.6	54.8	56.0	57.2	58.4	59.7	60.9	62.1	63.4	64.6	65.8
10	39.7	40.9	42.1	43.3	44.5	45.7	46.9	48.1	49.3	50.6	51.8	53.0	54.2	55.5	56.7	57.9	59.2	60.4	61.7	62.9	64.2	65.4	66.7	67.9
11	41.4	42.6	43.8	45.0	46.3	47.5	48.7	49.9	51.2	52.4	53.7	54.9	56.1	57.4	58.6	59.9	61.2	62.4	63.7	65.0	66.3	67.5	68.8	70.1
12	43.1	44.3	45.6	46.8	48.1	49.3	50.6	51.8	53.1	54.3	55.6	56.8	58.1	59.4	60.7	62.0	63.2	64.5	65.8	67.1	68.4	69.7	71.0	72.4
13	44.9	46.1	47.4	48.6	49.9	51.2	52.5	53.7	55.0	56.3	57.6	58.9	60.2	61.5	62.8	64.1	65.4	66.7	68.0	69.3	70.7	72.0	73.3	74.7
14	46.7	48.0	49.3	50.5	51.8	53.1	54.4	55.7	57.0	58.3	59.6	60.9	62.3	63.6	64.9	66.3	67.6	68.9	70.3	71.6	73.0	74.4	75.7	77.1
15	48.6	49.9	51.2	52.5	53.8	55.1	56.4	57.8	59.1	60.4	61.8	63.1	64.5	65.8	67.2	68.5	69.9	71.3	72.6	74.0	75.4	76.8	78.2	79.6
16	50.5	51.8	53.2	54.5	55.8	57.2	58.5	59.9	61.2	62.6	64.0	65.3	66.7	68.1	69.5	70.9	72.3	73.7	75.1	76.5	77.9	79.3	80.8	82.2
17	52.5	53.9	55.2	56.6	58.0	59.3	60.7	62.1	63.5	64.9	66.3	67.7	69.1	70.5	71.9	73.3	74.8	76.2	77.6	79.1	80.5	82.0	83.5	84.9
18	54.6	56.0	57.4	58.8	60.2	61.6	63.0	64.4	65.8	67.2	68.6	70.1	71.5	73.0	74.4	75.9	77.3	78.8	80.3	81.8	83.3	84.8	86.3	87.8
19	56.8	58.2	59.6	61.0	62.4	63.9	65.3	66.8	68.2	69.7	71.1	72.6	74.1	75.5	77.0	78.5	80.0	81.5	83.1	84.6	86.1	87.6	89.2	90.7
20	59.0	60.4	61.9	63.3	64.8	66.3	67.7	69.2	70.7	72.2	73.7	75.2	76.7	78.2	79.8	81.3	82.8	84.4	85.9	87.5	89.1	90.7	92.2	93.8
21	61.3	62.8	64.3	65.8	67.3	68.8	70.3	71.8	73.3	74.9	76.4	77.9	79.5	81.1	82.6	84.2	85.8	87.4	89.0	90.6	92.2	93.8	95.4	97.1
22	63.8	65.3	66.8	68.3	69.8	71.4	72.9	74.5	76.1	77.6	79.2	80.8	82.4	84.0	85.6	87.2	88.9	90.5	92.1	93.8	95.5	97.1	98.8	100.5
23	66.3	67.8	69.4	71.0	72.5	74.1	75.7	77.3	78.9	80.5	82.2	83.8	85.4	87.1	88.7	90.4	92.1	93.8	95.5	97.2	98.9	100.6	102.4	104.1
24	68.9	70.5	72.1	73.7	75.3	77.0	78.6	80.3	81.9	83.6	85.2	86.9	88.6	90.3	92.0	93.8	95.6	97.2	99.0	100.7	102.5	104.3	106.1	107.9
25	71.7	73.3	75.0	76.6	78.3	80.0	81.7	83.3	85.1	86.8	88.5	90.2	92.0	93.7	95.5	97.3	99.1	100.9	102.7	104.5	106.3	108.2	110.0	111.9
26	74.6	76.3	78.0	79.7	81.4	83.1	84.8	86.6	88.4	90.1	91.9	93.7	95.5	97.3	99.2	101.0	102.9	104.7	106.6	108.5	110.4	112.3	114.2	116.2
27	77.6	79.4	81.1	82.9	84.6	86.4	88.2	90.0	91.9	93.7	95.5	97.4	99.3	101.2	103.1	105.0	106.9	108.8	110.8	112.7	114.7	116.7	118.7	120.7
28	80.8	82.6	84.4	86.3	88.1	89.9	91.8	93.7	95.6	97.5	99.4	101.3	103.3	105.2	107.2	109.2	111.2	113.2	115.2	117.3	119.3	121.4	123.5	125.6
29	84.2	86.1	87.9	89.8	91.7	93.7	95.6	97.5	99.5	101.5	103.5	105.5	107.5	109.5	111.6	113.7	115.7	117.8	120.0	122.1	124.2	126.4	128.6	130.8
30	87.8	89.7	91.7	93.6	95.6	97.6	99.6	101.6	103.7	105.7	107.8	109.9	112.0	114.2	116.3	118.5	120.6	122.8	125.1	127.3	129.5	131.8	134.1	136.4
31	91.6	93.6	95.6	97.7	99.7	101.8	103.9	106.0	108.2	110.3	112.5	114.7	116.9	119.1	121.4	123.6	125.9	128.2	130.5	132.9	135.3	137.7	140.1	142.5
32	95.7	97.8	99.9	102.0	104.2	106.3	108.5	110.7	113.0	115.2	117.5	119.8	122.1	124.5	126.8	129.2	131.6	134.0	136.5	139.0	141.5	144.0	146.6	149.1
33	100.0	102.2	104.4	106.6	108.9	111.2	113.5	115.8	118.2	120.5	122.9	125.4	127.8	130.3	132.8	135.3	137.8	140.4	143.0	145.6	148.3	150.9	153.7	156.4
34	104.7	107.0	109.3	111.7	114.0	116.4	118.9	121.3	123.8	126.3	128.8	131.4	134.0	136.6	139.2	141.9	144.6	147.4	150.1	152.9	155.7	158.6	161.5	164.4
35	109.7	112.2	114.6	117.1	119.6	122.2	124.7	127.3	129.8	132.5	135.3	138.0	140.8	143.6	146.4	149.2	152.1	155.0	158.0	161.0	164.0	167.1	170.2	173.3
36	115.2	117.8	120.4	123.0	125.7	128.4	131.1	133.9	136.7	139.5	142.4	145.3	148.3	151.3	154.3	157.3	160.5	163.6	166.8	170.0	173.3	176.6	179.9	183.3
37	121.3	124.0	126.8	129.6	132.4	135.3	138.2	141.2	144.2	147.3	150.3	153.5	156.7	159.9	163.1	166.5	169.8	173.2	176.7	180.2	183.7	187.3	191.0	194.7
38	127.9	130.8	133.8	136.8	139.9	143.0	146.2	149.4	152.6	155.9	159.2	162.6	166.1	169.6	173.2	176.8	180.4	184.2	188.0	191.8	195.7	199.7	203.7	207.7
39	135.3	138.5	141.7	145.0	148.3	151.7	155.1	158.6	162.1	165.7	169.4	173.1	176.9	180.7	184.7	188.7	192.7	196.8	201.0	205.3	209.6	214.0	218.5	223.0
40	143.7	147.1	150.6	154.2	157.8	161.5	165.3	169.1	173.0	177.0	181.1	185.2	189.4	193.7	198.1	202.5	207.1	211.7	216.4	221.1	226.0	231.0	236.0	241.1
41	153.2	157.0	160.9	164.8	168.9	173.0	177.2	181.5	185.8	190.3	194.8	199.5	204.2	209.1	214.0	219.1	224.2	229.4	234.8	240.2	245.8	251.5	257.2	263.1
42	164.3	168.6	172.9	177.3	181.9	186.5	191.3	196.1	201.1	206.2	211.4	216.7	222.2	227.7	233.4	239.2	245.2	251.3	257.5	263.8	270.3	276.9	283.6	290.5
43	177.5	182.3	187.3	192.4	197.6	202.9	208.4	214.0	219.8	225.8	231.8	238.1	244.5	251.0	257.7	264.6	271.7	278.9	286.3	293.8	301.5	309.4	317.4	325.7
44	193.6	199.3	205.1	211.0	217.2	223.5	230.0	236.7	243.6	250.8	258.1	265.6	273.3	281.2	289.4	297.8	306.3	315.1	324.1	333.3	342.8	352.4	362.3	372.4
45	214.1	220.9	227.9	235.2	242.7	250.4	258.4	266.7	275.3	284.1	293.3	302.6	312.3	322.3	332.5	343.0	353.8	364.9	376.2	387.9	399.8	412.0	424.5	437.4
46	241.5	250.0	258.9	268.2	277.8	287.8	298.1	308.8	319.9	331.4	343.3	355.5	368.1	381.1	394.5	408.3	422.5	437.1	452.0	467.4	483.3	499.6	516.3	533.5
47	280.9	292.4	304.4	316.9	330.0	343.6	357.8	372.5	387.7	403.4	419.8	436.6	454.1	472.1	490.7	509.9	529.8	550.4	571.7	593.8	616.7	640.5	665.3	691.0
48	344.1	360.9	378.4	396.8	436.0	456.9	478.6	501.2	524.7	549.3	574.8	601.5	629.4	658.6	689.3	721.5	755.6	791.5	829.7	870.4	913.9	960.6	1011.2	
49	461.1	488.4	517.2	547.5	579.4	613.1	648.8	686.7	727.0	770.1	816.4	866.4	920.8	980.4	1046.2	1119.9	1203.3	1299.7	1413.6	1553.1	1732.9	1986.3	2419.6	>2419.6

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3.14 EPA Method 1669 - Clean Sampling of Natural Waters for Trace Metals

Justification for and details about EPA Method 1669 can be found in Section 6.12.

The procedure presented has been approved by the EPA. However, the current procedure followed by the Surface Water Monitoring and Standards Unit may be slightly different. Each contract laboratory have lab specific procedures based on the equipment they provide and the analysis methods followed. An equipment list will not be presented because of the differences in contract laboratory procedures.

3.14.1 Clean Hands/Dirty Hands Pre-Sampling Precautions

The SOP describes field sampling procedures commonly referred to as "clean hands/dirty hands" techniques. Upon arrival at the sampling site, one member of the two-person sampling team is designated as "dirty hands" and a second member is designated as "clean hands." All operations involving contact with the sample bottle and transfer of the sample from the sample collection device to the sample bottle are handled by the individual designated as clean hands. Dirty hands is responsible for preparation of the sampler (except the sample container itself), operation of any machinery; e.g., pump, and for all other activities that do not involve direct contact with the sample.

When sampling for mercury, a fiberglass boat with an electric motor and wooden or fiberglass oars is preferred. The boat should be washed and stored in an area that minimizes exposure to dust and atmospheric particles. If an internal combustion engine is required, it should be shut off at a distance far enough from the sampling point to avoid contamination, then the sampling team should manually propel the boat to the sampling point. Personnel may wear an unlined, long-sleeved wind suit consisting of pants and jacket and constructed of nylon or other synthetic fiber to prevent mercury adsorbed onto cotton or other clothing materials from contaminating samples.

3.14.2 Manual Grab Sampling

All sampling personnel must don clean gloves before commencing sample collection activity.

1. Dirty hands opens the cooler or storage container, removes the double-bagged sample bottle from storage, and unzips the outer bag.

2. Clean hands opens the inside bag containing the sample collection bottle, removes the bottle, and reseals the inside bag. Dirty hands then reseals the outer bag.

3. Clean hands moves to the collection location and submerges the sample bottle, allowing the bottle to partially fill with sample water. Clean hands screws the cap on the bottle, shakes the bottle several times, and empties the rinseate away from the sampling

location. After two more rinsings, clean hands holds the bottle under water and allows the bottle to fill with sample water. After the bottle has filled and while the bottle is still underwater, clean hands replaces the cap on the bottle. Clean hands must insure that the sample water does not come into contact with the air.

4. After the bottle cap has been replaced, dirty hands reopens the outer plastic bag, and clean hands opens the inside bag, places the bottle inside, and seals the inner bag.

5. Dirty hands seals the outer bag.

6. If the sample is to be analyzed for dissolved metals, it is filtered in accordance with the procedure described in Section 3.14.5.

7. Record the bottle number or other information on the Field Data Sheets or field notes.

3.14.3 Grab Sampling with a Sampling Device

The following details the sampling technique with a suspended grab sampling device; e.g., DH-81 grab sampler or Beta bottle (Lakes).

All sampling personnel must don clean gloves before commencing sample collection activity. If it is necessary to attach a bottle to the device in the field, clean hands performs this operation inside the field-portable clean box.

1. Dirty hands remove the sampling device from its storage container and opens the outer polyethylene bag.

2. Clean hands open the second polyethylene bag and removes the sampling device. On those occasions where it may be possible to pre-attach a sample bottle to the sampling device in the laboratory, then the entire assemble, bottle and device, is handled as the bottle alone is in the instructions below.

3. Dirty hands open the cooler or storage container, removes the double-bagged sample bottle from storage, and unzips the outer bag.

4. Clean hands open the inside bag containing the sample bottle, removes the bottle, and reseals the inside bag. Dirty hands then reseals the outer bag.

5. Clean hands change gloves.

6. Dirty hands submerge the sampling device to the desired depth.

7. When the bottle is full, dirty hands remove the sampling device from the water.

8. Dirty hands returns the sampling device to its large inner plastic bag; clean hands pulls the bottle out of the collar, unscrews the bottle from the sampling device, and caps the bottle. Clean hands and dirty hands then return the bottle to its double-bagged storage. If the sampling device is to be re-used, it must be decontaminated in accordance with Section 3.14.6.

3.14.4 Sampling with a Continuous Flow Sampling Device

Before putting on wind suits or gloves, the sampling team removes the bags containing the pump, tubing, batteries, gloves, plastic wrap, wind suits, and, if samples are to be filtered, the filtration apparatus from the coolers or storage containers in which they are packed.

All sampling personnel must don clean gloves before commencing sample collection activity.

1. Dirty hands remove the pump from its storage bag, and opens the bag containing the new tubing.

2. Clean hands install the tubing while dirty hands holds the pump. Clean hands immerses the inlet end of the tubing into the sample stream.

3. Both clean hands and dirty hands change gloves.

4. Dirty hands turns the pump on and allows the pump to run for 5-10 minutes or longer to purge the pump and tubing.

5. If the sample is to be filtered, clean hands installs a new cartridge filter at the end of the tubing.

6. The sample is collected by rinsing the sample bottle (with the water from the pump) as dictated by the tier being followed and collecting the sample from the pump. The pump tubing and the filter are discarded after use.

3.14.5 Field Filtering

The sampling procedures described in Sections 3.14.2 and 3.14.3 are used for samples collected using manual collection systems which require a separate filtering step. Some laboratories include filtering apparatus and instructions for its use with the bottles. If such is the case, follow the laboratory's instructions for filtering. The procedures below are for those instances when the laboratory doesn't provide apparatus or instructions.

This procedure describes in-line filtration using the peristaltic pump in a similar manner to the continuous-flow approach. Tubing and filters must be changed between samples.

1. Set up the filtration system inside the clean box using a length of pump tubing that is practical for the setup. Place the peristaltic pump immediately outside of the clean box and poke a small hole in the bag around the clean box for passage of the tubing.

2. Dirty hands remove the pump from its storage bag, and opens the bag containing the new tubing.

3. Clean hands install the tubing while dirty hands holds the pump.

4. Both clean hands and dirty hands change gloves.

5. Clean hands install a new cartridge filter at the end of the tubing.

6. Clean hands remove the bottle containing the water sample from the inner storage bag and places the sample inside the clean box. Clean hands also places two clean empty sample bottles, a bottle containing reagent water, and a bottle for waste, in the clean box.

7. Clean hands remove the cap from the reagent water bottle and places the end of the pump tubing in the bottle.

8. Dirty hands start the pump and passes approximately 200 ml of reagent water through the tubing and filter into the waste bottle. Clean hands then moves the outlet tubing to a clean bottle and collects the remaining reagent water as an equipment blank. Dirty hands stops the pump and clean hands caps the blank bottle.

9. Clean hands replace the cap on the bottle, returns the bottle to the inside bag, and closes the bag.

10. Clean hands remove the cap from the sample bottle and places the intake end of the tubing in the bottle.

11. Dirty hands start the pump and passes approximately 50 ml through the tubing and filter into the remaining clean sample bottle and then stops the pump. Clean hands uses the filtrate to rinse the bottle, discards the waste sample, and returns the outlet tube to the sample bottle. Repeat as dictated by the tier being followed.

12. Dirty hands start the pump and the remaining sample is processed through the filter and collected in the sample bottle. If preservation is required, acid is added to the sample by clean hands at this point.

13. Clean hands replace the cap on the bottle, returns the bottle to the inside bag, and closes the bag.

14. Clean hands then places the both the sample and blank closed bags into their respective outer bags held by dirty hands.

15. Dirty hands closes the outer bags, and places the double-bagged sample bottles into a clean, ice-filled cooler for immediate shipment to the laboratory.

3.14.6 Field Decontamination of Equipment

Sampling activity can be planned such that sufficient equipment is brought to the field that field decontamination of the sampling equipment between samples in unnecessary. If it is not possible to plan sampling activity in this manner, dedicated sampling equipment should be provided for each sampling event.

If samples are collected from adjacent sites of the same source water (e.g., immediately upstream or downstream), rinsing of the sampling apparatus with water that is to be sampled should be sufficient.

If it is necessary to cross a gradient (i.e., going from a high-concentration sample to a low-concentration sample), such as might occur when collecting at a second site, the following procedure may be used to clean the sampling equipment between samples.

1. Inside the clean box, use the "clean hands/dirty hands" procedure to process the dilute nitric acid solution through the apparatus. Dispose of the spent dilute acid in accordance with the project plan.

2. Process 1 L of reagent water through the apparatus to rinse the equipment and discard the spent water.

3. Collect a field blank as described in Section 3.14.9.

4. Rinse the apparatus with copious amounts of the ambient water sample and proceed with sample collection.

3.14.7 Quality Assurance/Quality Control

The sampling team shall employ a strict quality assurance / quality control (QA/QC) program. The team must collect equipment blanks, field splits, field blanks, QC samples and others as dictated by the project plan.

The sampling team is permitted to modify the sampling techniques described in this method to improve performance or reduce sampling costs, provided that reliable analyses of samples are obtained and that samples and blanks are not contaminated. Each time a modification is made to the procedures, the sampling team is required to demonstrate that the modification does not result in contamination of field and equipment blanks.

3.14.8 Equipment Blanks

Before using any sampling equipment at a given site, the field team is required to generate equipment blanks to demonstrate that the equipment is free from contamination. Equipment blanks must be run on all equipment that will contact the sample. Two types of equipment blanks are required: reagent water blanks and sampling equipment blanks.

A reagent water blank is prepared by the laboratory at the time they fill the reagent water bottle for the field team. This sample is not shipped to the team, but is analyzed and the results compared to the equipment blank generated by the field team.

A reagent-water-filled container should be shipped to the field site and handled the same as all other sample containers and sampling equipment.

Equipment blanks are generated by processing reagent water through the equipment using the same procedures to collect the sample. Sampling personnel must collect an equipment blank before collecting the sample.

If any metal(s) of interest or any potentially interfering substance is detected in either the equipment or reagent water blank at the minimum level specified in the referenced method, the source of contamination/interference must be identified and removed.

3.14.9 Field Blanks

To demonstrate that sample contamination has not occurred during field sampling and sample processing, at least one field blank must be generated for every 10 samples that are collected, but no less than one per day. Field blanks are collected before sample collection.

Field blanks are generated by using the reagent water supplied by the laboratory, processing the water through each of the sample processing steps and equipment (e.g., tubing, sampling devices, filters, etc.) that will be used in the field, collecting the field blank in one of the sample bottles, and shipping the bottle to the laboratory for analysis in accordance with the method(s) referenced in Table 5. However, manual grab sampling field blanks are represented by the equipment blank. Subsurface sampler field blanks are collected by immersing the tubing into the water and pumping water into a sample container. Filtering is accomplished using the procedures described in Section 3.14.5. If it is necessary to decontaminate the sampling equipment between samples, a field blank should be collected after the cleaning procedures and before the next sample is collected.

3.14.10 Field Splits

To assess the precision of the field sampling and analytical processes, at least one field split sample must be collected for every 10 samples that are collected during a given event. The field split is collected by splitting a larger volume (a single container which is filled following clean

hands/dirty hands procedures) into two aliquots in the clean box. Using a churn splitter inside a clean box is difficult, but until an alternative method is found, must be used. Field splits for dissolved metals determinations must be filtered using the procedures in Section 3.14.5.

3.14.11 Health and Safety

When working with potentially hazardous materials, follow U.S. EPA, OSHA and specific health and safety procedures.

Decontamination can pose hazards under certain circumstances even though performed to protect health and safety. Hazardous substances may be incompatible with decontamination methods. For example, the decontamination solution or solvent may react with contaminants to produce heat, explosion, or toxic products. Decontamination methods may be incompatible with clothing or equipment; some solvents can permeate or degrade protective clothing. Decontamination solutions and solvents may pose a direct health hazard to workers through inhalation or skin contact, or if they combust.

3.14.12 Method Sources

EPA Method 1669, Sampling Ambient Water for Determinations of Metals at EPA Water Quality Criteria Levels (undated).

USEPA Region 9 Laboratory; Richmond, California; Standard Operating Procedure #1229; (undated).

3.15 Biological Sampling of Wadeable Streams

3.15.1 Macroinvertebrates

Macroinvertebrate sampling is conducted to assess the biological integrity of perennial, wadeable streams. ADEQ has developed bioassessment tools in the form of Indexes of Biological Integrity (IBI) along with habitat evaluations for this purpose. There are two IBIs, the Warm water IBI and Cold water IBI. The procedure for calculating the IBIs is found in Section 3.15.2 and in the Biocriteria Program QAPP (ADEQ, 2001). Guidance for performing habitat evaluations, needed to determine causes and sources of impacts, is presented in Section 3.17. This procedure presents the field methods used for collecting macroinvertebrates, the initial step in conducting a bioassessment.

Equipment required: D-frame net fixed with a 500 micron mesh net (Figure 15), forceps, a large bucket, a metal sieve having a 500 μm mesh, a white dissecting tray, 37% formaldehyde, and 99% isopropyl alcohol.

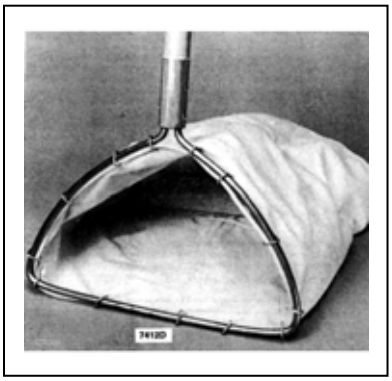

Figure 15 D-frame net with 500 micron mesh net.

3.15.1.1 Site Selection

The study reach length should be one of the following: 1) 25 times the bankfull width of the stream in wadeable streams, or a minimum of 100 meters in larger streams, or 2) long enough to encompass 2 meander lengths of the stream and multiple riffles in which to produce a composite sample, but not less than 100 feet in length. For a definition of bankfull, see the Rosgen stream type identification section of the habitat assessment protocol, Section 3.17.3. The study reach should be selected to represent typical habitat conditions found in the larger stream segment. The study reach length should begin at the top of a riffle or run and end at the bottom of a riffle or run.

If the reach is to be used as a reference or background reach, the following general criteria must be met:

- The site must be accessible within a 2-hour walk or 3-4 miles from the nearest four wheel drive road
- No known discharges upstream
- No major impoundments upstream

- ❖ No human caused channel alterations at the site; e.g. diversions, dredge and fill projects
- ❖ At least 0.5 miles downstream of road crossings
- ❖ The site should be perennial. The indicators for perennial condition are likely to be the presence of fish, univoltine insects, and healthy unstressed riparian plants
- ❖ The site should be free of local land use impacts
- ❖ There should be no recorded violations of pH or dissolved oxygen water quality standards
- ❖ The Habitat Assessment Index score should be greater than 14 (see Section 3.17.2.6).

3.15.1.2 Sample Collection Information

Biocriteria research efforts have focused on perennial, wadeable streams to date. Sample collection methods have been developed and refined only for this waterbody type at this point. The following sampling conditions and time frames must be met in order to collect macroinvertebrates for ADEQ bioassessment purposes. A stream reach must be:

- ❖ Wadeable
- ❖ Perennial
- ❖ Contain fast-flowing riffle or run habitat
- ❖ Contain heterogeneous substrates
- ❖ Sampled during the spring index period (April-May for warm water streams and May-June for cold water streams).

Wadeable means no deeper than can be safely waded across when collecting samples. **Perennial** refers to stream segments which flow continuously throughout the year (excluding effluent dependent waterbodies). **Riffle habitat** refers to the portions of streams where moderate velocities and substrate roughness produce moderately turbulent conditions which break the surface tension of the water and may produce whitewater (Bain and Stevenson, eds. 1999). **Run habitat** refers to segments of streams where there is moderate velocity water, but non-turbulent conditions which do not break the surface tension of the water and do not produce whitewater (Bain and Stevenson, eds.1999).

Heterogeneous substrate means a mixture of particle sizes comprising the stream bottom material that is less than 50% composed by travertine, bedrock or sand. Streams with *homogeneous* substrates, such as bedrock or sand have aquatic communities which exhibit limited taxa richness and loss of structure and function when compared with reference conditions.

The **spring index period** is defined as a period of time following winter runoff in which baseflow conditions will be found in most streams. Baseflow conditions generally are achieved post winter runoff in the desert streams in April-May and in mountain streams in May-June. A period of 4 weeks post-bankfull flood condition is generally required prior to macroinvertebrate sampling, even during the spring index sampling period. Hydrologic conditions are checked in the office prior to a site visit and field conditions are documented on the SEM Field Form for

Macroinvertebrate Sample Collection in the field prior to sampling to confirm that sampling is occurring during the correct sample collection conditions.

Macroinvertebrate samples are not collected when the following conditions occur:

- A bankfull or greater magnitude flow event has occurred within 4 weeks of site visit or when extreme high flow events have occurred, resulting in deep scouring of the streambed and benthic community such that the macroinvertebrate community will not recover within the spring index period
- Extended drought conditions have reduced flow from previously perennial condition to pools only or stagnant wetland habitat
- Stream substrates are dominated (consisting of >50% of that substrate type) by bedrock, travertine, or sand are considered non-target conditions.

Macroinvertebrate samples should be collected before pebble counts and before any disturbance to the stream channel by investigators. The collection begins at the downstream end of the assessment reach and proceeds upstream.

A macroinvertebrate sample consists of a three-minute timed composite sample from kick samples collected from three riffle habitats within the study reach. The target sampling area is approximately one square meter per each one minute sample. Select 3 or more riffles which represent the variety of substrate sizes, velocities, depths, and habitats found within the reach. Collect one-minute timed samples from each of three habitats or divide the time as needed among the variety of habitats. If three good-sized riffles are not available to be sampled, spread the three minute sample time over whatever riffle/run areas are available.

3.15.1.3 Sample Collection

Once the sampling sites within the reach have been selected, the first sample should be at the lowermost riffle.

1. Fill a bucket half full with stream water.

2. Place the D-frame net on the stream bed in the path of flowing water, and agitate a one square meter area of substrate vigorously for one minute by kicking or hand turning rocks to collect dislodged material. Sample as much variation of the flow and substrate as possible including large and small substrates.

3. Deposit the contents of the net into the bucket. At this point there is no need to pick the net clean.

4. Repeat the sampling procedure for the second and third riffles. After the last riffle, use forceps to remove organisms attached to the D-frame net. Before leaving the site, the D-frame dip net, bucket, and sieve should be rinsed and scrubbed with a brush to dislodge small invertebrates, egg masses, and organic material, so that it is not

transferred to the next site.

5. Swirl the contents of the bucket and pour the non-sediment portion into a 500 µm mesh sieve.

6. Add water again to the bucket, swirl and pour the contents into the sieve. Repeat this procedure several times until all insects and organic debris are emptied and only sediment remains.

7. Dump the remaining sediment into a dissecting tray and search the sediment for any remaining organisms, especially cased Trichoptera, snails, and freshwater clams and then discard the remaining sediment.

8. Gently, squeeze the sample to remove excess water from algae laden samples. Using a plastic spoon or hands, gently dispense the sample from the sieve into a wide mouth, one-liter sample jar. Fill the jar half to three-quarters full. If additional sample remains in the sieve, use an extra jar to contain it. Rinse any leftover material in the sieve into a corner and spoon out as much as possible. Check the sieve for any remaining animals. If the entire sample does not fit into one jar, then add the remainder to a second jar. If the sample will not fit into two jars, then field split the sample.

9. To perform a one-half field split, evenly spread the entire sample in a white dissecting tray and divide the sample with your hands into two equal portions. Place one half of the sample into the two sample jars and discard the other half into the stream. Note on the field form that the sample was "field split 2". A quarter split can be performed if a half-split still provides too much sample material to fit in two jars.

10. Place label(s) (see Section 3.15.3.2) in the jar(s), add enough 99% isopropyl alcohol to cover the sample material by about 1 inch.

11. Seal the jar(s), and affix a second label to outside of the jar(s).

3.15.1.4 Sample Labeling

Each macroinvertebrate sample should have two identification labels penciled on "write-in-the-rain" paper: one placed inside the jar, visible from the outside, and one affixed to the outside of the jar, attached with clear plastic tape. If more than one jar is used for a sample, put jar numbers on all labels (e.g., 1 of 2, 2 of 2). Each tag should have the following information at a minimum:

Waterbody name
Site code number
Habitat sampled (riffle)
Date
ADEQ and collectors' initials

3.15.1.5 Preservation and Storage

After samples have been preserved with 99% isopropyl alcohol, samples should be placed in an ice chest with ice to cool the sample. This prevents overheating and degradation of the sample, and prevents fumes from developing inside truck camper shells.

Samples should be stored in a cool environment and within flammable storage areas in the laboratory prior to shipping to the laboratory.

3.15.1.6 Chain of Custody

To complete the Chain of Custody, samples shall be locked in field trucks when sampling personnel are away from the truck. Sample jars shall be placed in the large, locked flammable cabinet in the equipment storage area of the ADEQ laboratory for storage, prior to shipping. The use of tamper-evident tape on shipping boxes to prevent tampering with samples during shipping is required. A Chain of Custody form will accompany the samples during shipment.

3.15.1.7 Sample Preparation for Shipping to Taxonomy Laboratory

1. Drain sample over a 500 micron sieve and large funnel into a waste container (e.g. 5-gallon isopropanol carboys) under a fume hood and place the sample back into the sample jar along with the correct interior label. Insure that all specimens have been picked off the sieve and placed in sample container, then cap tightly. Rinse and scrub the sieve with a brush prior to draining the next sample.

2. Wrap jars in bubble wrap and place in a heavy duty garbage bag with an absorbent sheet, inside a shipping box. ADEQ uses ADOT approved boxes that are 14x10x10", single-wall construction, edge Crush Test of 32 lbs/inch, gross weight limit of 65 lbs. The bubble wrap prevents shock and deterioration of the sample specimens. Close and seal the garbage bag. Place the Laboratory address on a sheet of paper inside the box, along with an inventory of samples. Seal the box with tamper-proof tape to continue chain of custody while samples are in transit. Mark boxes with “this side up” arrows to further prevent leakage and protect samples.

3. Ship boxes via commercial carrier. No hazardous materials labeling is required. ADEQ has received verbal approval from DOT and UPS to ship decanted macroinvertebrate samples without having to meet hazardous materials shipping requirements because of the very limited quantities of isopropanol alcohol present in the decanted samples. ADEQ marks boxes with Athis side up@ arrows to further prevent leakage and protect samples. Normal address labels are used for shipping. Enclose Chain of custody form with the samples.

All macroinvertebrate samples from a spring sample event are shipped to the laboratory in July of each year or as soon as practical after sampling.

The taxonomy laboratory verifies receipt of samples listed on the chain of custody form. The laboratory also refreshes the alcohol preservative.

3.15.1.8 Literature Cited

ADEQ, 2005. Biocriteria Program Quality Assurance Program Plan, revision D. ADEQ, Phoenix, AZ.

5 Macroinvertebrate Taxonomy Laboratory Procedures

The procedures followed at the taxonomic laboratory are not part of the field sampling procedures, however they are presented here for reference purposes. These procedures include sample processing, sorting, taxonomic identification levels, voucher specimens, and general quality control procedures.

Sample processing - Upon receipt of the samples, the laboratory will check and adjust the preservation in each sample, catalog the samples, check the attached inventory for accuracy, and sign the chain of custody papers. The consultant will then notify ADEQ of the receipt of samples, any damaged samples, or discrepancies between the inventory and actual sample labels.

Sample sorting - Samples must be sorted to separate the invertebrates from the sample matrix. The entire sample should be floated in water in a white plastic tray. Large debris is rinsed and removed from the sample until all organic matter and invertebrates are floated off the mineral residue. The mineral residue is then searched for stone-cased caddisflies and molluscs.

Sub-sampling and sorting - Arizona samples typically contain thousands of invertebrates and must be sub-sampled for results to meet a minimum count of 500-600 organisms. A Caton Tray is be used to randomly obtain fractions of the total sample from which all the invertebrates are removed and counted. Additional fractions are selected until the 500-600 target level is reached after which the number of squares subsampled are recorded. Terrestrial insects and non-benthic insects (e.g. corixidae, other swimmers, mosquitoes, or surface tension dwellers) should not be included in the count. Additional fractions are examined if one fraction is dominated by a single species. After the target number of specimens has been achieved, the entire unsorted sample is scanned for large or rare taxa, which may aid in identification of smaller instars or may expand the taxa list for that sample. The remaining unsorted sample is re-preserved with 70% isopropanol in individual containers and archived at the laboratory for one year from the date of sample receipt, after which time the laboratory will contact ADEQ prior to disposal.

Sorting - The sorting of invertebrates from the sample matrix shall be performed by trained technicians, using dissecting scopes with a minimum magnification of 6X. After identifications have been made, the sorted specimens, including the separated Chironomidae, should be archived for one year or incorporated into the reference or voucher specimen set. The laboratory shall keep logs for each sample sorted, the fraction sorted, sample matrix problems, etc. in addition to bench sheets of the taxa identified in each sample.

Sorting efficacy - The laboratory shall check the sample residues to check for a sorting efficacy of 95% or better. A statement of sorting efficacy for the ADEQ batch of samples should be presented in the laboratory report.

Taxonomic identification - Invertebrate identifications shall be performed by a trained and experienced taxonomist. The taxonomy contractor is responsible for obtaining the most accurate, consistently achievable identifications for ADEQ samples, using specialists as needed to obtain identifications to the general taxonomic levels listed in Table 5.

Reference specimens - A set of reference or voucher specimens shall be prepared from the batch of samples each year for incorporation into the reference specimen collection. Several specimens shall be preserved for each new taxon and the best or largest larval instars of other taxa shall be preserved to represent the taxa found that year and to update the historic reference collection at ADEQ. The taxonomist shall make recommendations for archiving any important specimens, if verification of identification by national specialists is required.

Lab Data Reports - Laboratory reports containing taxonomic identifications and counts for all samples for that year shall be submitted to ADEQ in electronic format. The electronic data shall be submitted in ACCESS database format or Excel spreadsheets formatted for database uploading. The Taxonomy Contractor shall perform quality control checks on the electronic data prior to submittal to ADEQ. The data set should contain at a minimum the Station ID, waterbody name and location, habitat, collection date, complete taxa ID from phylum to lowest level ID, raw number of individuals, the portion of sample analyzed including field splits where applicable, and adjusted final counts, which are corrected for sub-sample size and field splits. A copy of the bench sheets used by the taxonomist to develop the raw counts per sample should also be submitted.

Table 5. ADEQ Taxonomic levels of identification for macroinvertebrates.

Invertebrate Group	Level of taxonomy required
Aquatic insects (except the family Chironomidae)	Genus (or species where consistently identifiable)
Chironomidae	Family
Semi-aquatic insects	Family
Arachnida (Mites)	Class
Cladocera, Copepoda, Ostracoda	Class
Amphipoda, Decapoda, Isopoda	Class
Nematoda, Nematomorpha	Phylum
Turbellaria	Class
Annelida	Class
Mollusca	Family or Genus

3.15.2 Arizona Indexes of Biological Integrity

3.15.2.1 Calculating the Arizona Indexes of Biological Integrity

The Arizona Indexes of Biological Integrity can be applied to macroinvertebrate taxonomic data generated by the sample collection procedures provided in this document. All the appropriate sample collection conditions must be met in order to calculate the IBIs for bioassessment purposes (i.e. application of the narrative biocriteria standard). There are currently two Indexes; a cold and a warm water IBI. The following narrative provides the steps needed to calculate these Indexes from taxonomic lists and abundance data generated by taxonomy laboratories from the field collected macroinvertebrate samples.

1. Identify the appropriate reference community using the site elevation.

   - ❖ The warm water community is defined as being located below the 5000 foot elevation.
   - ❖ The cold water community is defined as being located above the 5000 foot elevation.

2. Calculate the macroinvertebrate metric values for the study sample following metric calculation procedures listed Figure 16. This table lists all the metrics used in both indexes and their definitions. Metrics required for each index are listed in Table 6.

3. Calculate the metric percent of reference score using either the warm or cold water reference metric threshold values associated with that community type (Tables 7 and 8.

4. Calculate an average of the percent of reference scores for all metrics to produce the IBI score. Table 9 provides an example of the scoring system for a warm water stream.

5. Determine assessment category for the IBI score from Table 10.

Figure 16. Formulas for calculating macroinvertebrate metrics used in the cold water and warm water Indexes of Biological Integrity.

Use the following formula to calculate the metric score (percentage of reference) for sensitive metrics whose values decrease with disturbance. Apply this formula to the following metrics.

Metric Score = (Sample value / metric threshold value) * 100

1. Total taxa richness
2. Number of Ephemeroptera taxa
3. Number of Tricoptera taxa
4. Number of Diptera taxa
5. Number of intolerant taxa
6. Percent Ephemeroptera
7. Percent Plecoptera
8. Percent scrapers
9. Number of scraper taxa

Apply the following formulas to calculate the metric score (percentage of reference) for tolerant metrics whose values increase with disturbance.

1. Hilsenhoff Biotic Index

   Metric score = (10 – Sample value) / (10 – Metric threshold value) * 100

2. Percent dominant taxon

   Metric score = (100 – Sample value) / (100 – Metric threshold value) * 100

Table 6. Descriptions for the warm water and cold water metrics used in Arizona=s IBIs.

Category	Metric	Definition	Expected Response to increasing disturbance
Richness measures	Total number of taxa	Number of different macroinvertebrate taxa	Decrease
	# Ephemeroptera taxa	Number of mayfly taxa	Decrease
	# Trichoptera taxa	Number of caddisfly taxa	Decrease
	# Diptera taxa	Number of true fly larvae.	Decrease
	# Intolerant taxa	Number of taxa having a tolerance value ≤3	Decrease
Composition measures	% Dominant taxon	Percent abundance of the single most abundant taxon.	Increase
	% Ephemeroptera	Percent abundance of mayflies, compared to total abundance of the sample	Decrease
	% Plecoptera	Percent abundance of stoneflies, compared to total abundance of the sample	Decrease
Tolerance measure	Hilsenhoff Biotic Index	Abundance-weighted average tolerance of assemblage	Increase
Trophic measures	% Scrapers	Percent abundance of the scraper functional feeding group, compared to total abundance of the sample	Decrease
	# Scraper taxa	Number of taxa in the scraper functional feeding group	Decrease

Table 7. Reference scoring thresholds for Warm Water metrics, used in the Arizona Warm Water Index of Biological Integrity.

Metric	Metric threshold value
Total taxa	37
Trichoptera taxa	9.0
Ephemeroptera taxa	9.0
Diptera taxa*	10.0
Scraper taxa	7.0
Percent scraper	23.7
Percent Ephemeroptera	70.0
Percent Dominant Taxon	19.1
Hilsenhoff Biotic Index	4.89

* Appropriate taxonomic effort is to genus for insects and to family for midges.

Table 8. Reference scoring thresholds for Cold Water metrics, used in the Arizona Cold Water Index of Biological Integrity.

Metric	Scoring threshold
Total taxa	38
Diptera taxa*	11.0
Intolerant taxa	6.0
Scraper taxa	11.0
Percent scraper	45.1
Percent Plecoptera	19.1
Hilsenhoff Biotic Index	4.23

* Appropriate taxonomic effort is to genus for insects and to family for midges.

Table 9. Example of the ADEQ Warm Water Index of Biological Integrity scoring system; Sycamore Creek near Round Valley bridge (Hwy 87) collected during spring 1995.

Metric	Metric Value	Metric Score (compared to warm water reference scoring threshold)
Total taxa	24	65
Trichoptera taxa	6	67
Ephemeroptera taxa	5	56
Diptera taxa	7	70
Scraper taxa	3	43
Percent scraper	20.3	86
Percent Ephemeroptera	26	37
Percent Dominant Taxon	41	73
Hilsenhoff Biotic Index	5.73	84
	Index Score (Metric Score means)	65 Attaining

Table 10. Assessments based on ADEQ macroinvertebrate IBI scores.

Macroinvertebrate bioassessment result	**Index of Biological Integrity Score**		**Assessment**
	Cold water	**Warm water**	
Greater than the 25th percentile of reference condition	≥ 52	≥ 50	Attaining
Between the 10th and 25th percentile of reference condition	46-51	40-49	Inconclusive
Less than the 10th percentile of reference condition	≤ 45	≤ 39	Impaired

3.15.3 Diatoms from Natural Substrates

Diatom samples are collected from riffle or run habitats of small to medium sized wadeable perennial streams during the spring index period; April-May for warm water streams and May-June for cold water streams.

The sample is collected from rocks having a flat and somewhat smooth surface. Care must be taken to not disturb the substrate in the area where the sample is to be taken; thus, the sample should be collected after the water sample, but prior to the pebble count and the macro-invertebrate sample. A single sample consists of a 6 cobble composite collection from a variety of riffle/run habitats throughout the study reach. The number of days past the last precipitation or flood event should be documented. Samples should not be taken within 2 weeks of a scouring flood event.

Equipment Required: A nine centimeter square vinyl template, Exacto knife, clean toothbrush, squeeze bottle, 250ml or 500ml sample jars, labels, and isopropanol preservative.

3.15.3.1 Sample Collection and Field Processing

1. Select 3 riffles in the assessment reach. From each riffle, select two relatively flat cobbles which have an obvious biofilm, for a total of six rocks. If there are fewer than 3 riffles in the reach, randomly select six rocks from the available riffle(s). The selected rocks should be in water of less than 1 meter (3 feet) deep. Begin sample collection at the bottom of the reach and move upstream. Try to avoid dislodging substrate when stepping onto riffles.

2. Outline a nine centimeter square area on each rock with the template and Exacto knife. Scrape the outlined area with 30 strokes of an Exacto knife. Make single direction strokes in one direction. Put the scrapings from the knife blade into the sample jar. Rotate the rock 90° and again scrape with 30 strokes in the same direction. Repeat until the rock has been scraped perpendicular to each side of the sample area. Rinse the scraped area of the rock into the sample jar with a squeeze bottle of water. Rinse the knife in the sample jar. Scrub the scraped area of the rock with a toothbrush for 10-15 seconds and rinse the scraped area again with water from the squeeze bottle. Rinse the toothbrush in the sample jar with water from the squeeze bottle. Use a new toothbrush and blade at each site.

3. Repeat the above process for each of the remaining 5 rocks. Composite all rock scrapings into the sample jar.

3.15.3.2 Labeling

Each diatom sample should have two identification labels penciled on "write-in-the-rain" paper: one placed inside the jar, visible from the outside, and one affixed to the outside of the jar, attached with clear plastic tape. If more than one jar is used for a sample, put jar numbers on all labels (e.g., 1 of 2, 2 of 2). Each tag should have the following information at a minimum.

Waterbody name
Site code number
Habitat sampled (riffle)
Date
ADEQ and collectors' initials

3.15.3.3 Preservation and Storage

1. Preserve the sample with about 10 ml of isopropanol and place in an ice chest with ice to prevent degradation of the sample while in the field. Care must be taken to not entirely submerge the collection jar in ice water.

2. Store the samples in a dark place until shipment to the contract laboratory.

3.15.3.4 Chain of Custody

1. Samples shall be locked in field trucks while sampling personnel are in the field.

2. Sample jars shall be placed in the large, locked flammable proof cabinet in the equipment storage area of the ADEQ laboratory for storage, prior to delivery.

3. A chain-of-custody form will be submitted with the samples at the time of delivery of samples to the laboratory.

SECTION 3

PART B

GEOMORPHOLOGY PROCEDURES

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3.16 Geomorphology

A geomorphic assessment of stream channels provides a process-based framework for understanding past and present watershed dynamics. A fluvial geomorphic assessment generally includes data collection, field investigations, and channel stability assessments (USDA, 1998). Much of ADEQ's field methodology is based on or derived from the work of Dr. Dave Rosgen, a consulting hydrologist and author with extensive work and expertise in fluvial geomorphology, with references detailed in Rosgen's text Applied River Morphology (1996). This section details those methods.

3.16.1 Stream Channel Condition

Stream channel condition or state is determined from field inspection and measurement of stream channel characteristics which include: amount, types, and age classes of riparian vegetation, sediment deposition patterns, frequency and amount of debris occurrence, meander patterns, stream size, flow regime, and altered states due to direct disturbance. The depositional pattern is particularly important as a screening level evaluation of excess sediment conditions. Depositional patterns, defined as bar features in alluvial channels, are evaluated and scored for purposes of the Rosgen channel stability index. The field form for determining depositional patterns present in the study reach is presented in Figure 1 (Rosgen, 1996).

3.16.1.1 Depositional Pattern – Field Procedure

1. Conduct a "toothpick survey" by walking the entire study reach, noting presence of point bars on the inside of river bends, or excess bar features in the form of side or mid-channel bars, islands, a braided river form, or delta bars downstream of tributary confluences. Bar features will typically be found in alluvial "Bc", "C", "D" and "E" channel types, but not in "A" and "B" type channels.

2. Conduct a cross-section survey at a riffle cross-section for purposes of identifying the proper channel type. It is important to know the channel type to evaluate whether the depositional features present are typical or not.

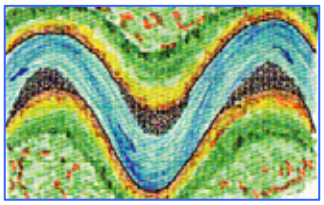
3. Presence of unvegetated and unconsolidated mid-channel or diagonal bars, or a branched channel or delta bars are all indicators of channel aggradation. The Rosgen scoring criteria in Table 1 are used to determine the stability rating and sediment supply score. The sediment supply score is then used in the final 6-parameter stability assessment score.

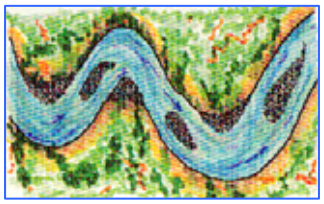
A bar is defined as a submerged or exposed ridge-like accumulation of sand, gravel, or other alluvial material formed within an active channel, along the banks, or at the terminus of a stream where a decrease in velocity induces deposition. A point bar is found on the inside of meander bends. Diagonal bars form diagonally to a stream channel, and may extend completely across the channel. Mid-channel bars form in the mid-channel zone and do not extend completely

across the channel. Islands are exposed bars or land segments within the stream channel that are relatively stable and normally surrounded by water. A side bar (or lateral bar) is located at the side of a stream channel, usually associated with the inside of slight curves. Delta bars are formed immediately downstream of the confluence of a tributary and the main stream (Armantrout, 1998).

Stream:	Reach:				
Date:	Observers:				
LIST ALL CATEGORIES THAT APPLY					

B1 Point bars

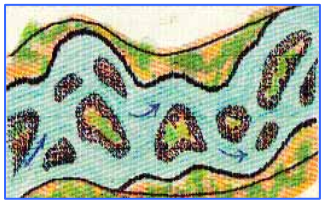




B2 Point bars with few mid-channel bars

B3 Numerous mid-channel bars

B4 Side bars



B6 Main channel branching with numerous mid-channel bars and islands

B8 Delta bars

B5 Diagonal bars

B7 Side bars and mid-channel bars with length exceeding 2 to 3 channel widths

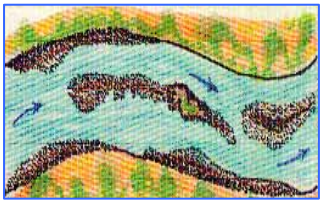


Figure 1. Field form for evaluation of depositional features (bars) in natural channels (after Rosgen, 1996).

Table 1. Scoring criteria for stability rating and sediment supply.

Depositional features present	Fig. 1 Illustration No.	Sediment supply category	Sediment supply score
Point bars	1	Low	2
Side bars	4		
Many mid-channel bars	3	Moderate	5
Diagonal bars	5	High	10
Point bars & few mid-channel bars	2		
Main channel branched with many mid-channel bars & islands	6		
Mixed side bar & mid-channel bars exceeding 2-3X width	7		
Delta bars	8		

3.16.1.2 Literature Cited

Armantrout, N.B., compiler. 1998. Glossary of Aquatic Habitat Inventory Terminology. American Fisheries Society, Bethesda, MD.

Rosgen, D. and H.L. Silvey. 1996. Applied River Morphology. Wildland Hydrology. Pagosa Springs, CO.

USDA. 1998. Stream Corridor Restoration, principles, processes, and practices. www.USDA.gov/stream_restoration/newtofc/ .

3.16.2 Cross-Section Surveys

Cross section surveys are conducted primarily for purposes of identifying Rosgen stream channel type, to represent riffle and pool habitats present in a reach, and to document bank erosion. A riffle cross-section survey is required for identifying stream channel type. Other cross-section surveys may be needed to conduct a Stream Stability Assessment. The riffle cross-section should be located where bankfull and floodprone indicators are present. Stream Type Classification and Bankfull identification procedures are presented in Sections 3B, 3.16.10 and 3.16.16, respectively. An illustrated guide to field surveying techniques is provided in Harrelson et al (1994). The following is an abbreviated version of the Harrelson procedure.

Equipment Required: Topcon Rotating Laser Level, stadia or telescoping rod, two pieces of rebar, flagging, camera, GPS unit, 100 foot measuring tape, calculator, laptop computer with spreadsheet or RiverMorph software installed, and cross-section field data sheets.

3.16.2.1 Cross-Section Surveying Procedure Using the Topcon Rotating Laser Level

1. Position the Topcon Rotating Laser Level on a promontory near the cross-section to be measured and, if possible, at an elevation above the floodprone depth. The Topcon unit should have an unobstructed view of the cross-section.

2. The first and last measurement should be made at a monumented benchmark, such as found at a USGS gauging station. If a monumented benchmark is not available, as will often be the case, you must create a monumented benchmark having a relative elevation of 100 feet. To construct a monumented benchmark, either pound a piece of rebar into the ground or mark a boulder or a rock outcropping. The rebar should extend out of the earth approximately two inches. These benchmarks need to be exactly noted on the field sketch with measurements from notable stationary and permanent objects so that they can be found at the next visit. A photograph of the monumented benchmark and the surrounding area, as well as a GPS reading is useful for re-locating the benchmark.

3. Anchor the measuring tape to the rebar on the left bank, with zero as the starting point at the rebar pin. Using the left bank as zero allows for conventional survey graphing of the cross-section. Run the tape across and perpendicular to the stream channel. Find a place on the opposite (right) bank that is near the same elevation as the first rebar and perpendicular to the direction of flow in the channel. Place a second piece of rebar here. Pull the tape as taut as possible and anchor the tape to the second in-ground rebar. Insure that the tape is not deflected by vegetation along the cross-section. To prevent the tape from whipping in the wind, tie several pieces of flagging to the tape.

4. If a USGS or other benchmark is available, shoot its elevation. Use the elevation of the benchmark above mean sea level as the reference elevation for the graphic. If no benchmark is available, start the cross-section at the constructed monumented benchmark.

5. Shoot the elevation of the constructed monumented benchmark on the left bank which is the starting point. The stadia rod should be placed on top of the rebar for this measurement. Also collect a measurement at the base of the left bank pin. The station number at this point is 0+00.

6. Moving downslope and along the cross-section, record the elevations of all relevant features in a field book or form (Figure 2). Relevant features might include changes in slope, edges of water, thalweg, bankfull, floodprone elevation, change in vegetation, etc. The person recording the elevations on the field data sheet must provide a description notation for each measurement in the comments column for each station (e.g., bankfull, LEW, REW, thalweg, top of terrace, bottom of bank). This information is very important when constructing the cross-section graphic

7. At each elevation measurement, record the cross-sectional distance from station 0+00 to the nearest tenth of a foot. Use a bubble level to ensure that the stadia rod is perpendicular to the earth before taking the elevation reading. If the cross section is longer than 100 feet, station numbers should follow surveyor's procedure for numbering stations (e.g., 112.4 feet is 1+12.4).

8. Shoot the rebar on the right bank at the top and base of the right bank rebar pin.

9. To check the accuracy of the survey, go back to the starting point, either the benchmark or the constructed monumented benchmark, and shoot that elevation again. The difference between the original elevation and the new calculated elevation is the error. Very small errors may result from rounding and are acceptable. Typically a closure of 0.02 feet is acceptable. Large errors may result from mistakes in calculation, so check the arithmetic first. Other errors may be due to inaccurately reading the rod, or note taking. If the error is too large, the line must be resurveyed to locate and correct the error. To estimate the allowable error, use the following equation.

$$0.007\sqrt{(\text{total distance}/100)}$$

10. Plot the cross-section in the field in a spreadsheet program or Rivermorph software if possible. The plotting of the cross-section in the field is a QC measure. Review the plot to determine if the survey is complete and that the bankfull elevation is reasonable. Resurvey near slope breaks if needed to verify the bankfull elevation or to eliminate possible errors before leaving the site. An example of a completed cross-section diagram is presented in Figure 3.

3.16.2.2 Literature Cited

C.C. Harrelson et al. 1994. Stream Channel reference Sites: An Illustrated Guide to Field Technique. USFS, General Technical Report RM-245.

page ____ of ____

CROSS-SECTION SURVEY

Site ID:_____ Site Description:_____

Date:_____ Watershed:_____ Stream Type:_____ Observers:_____

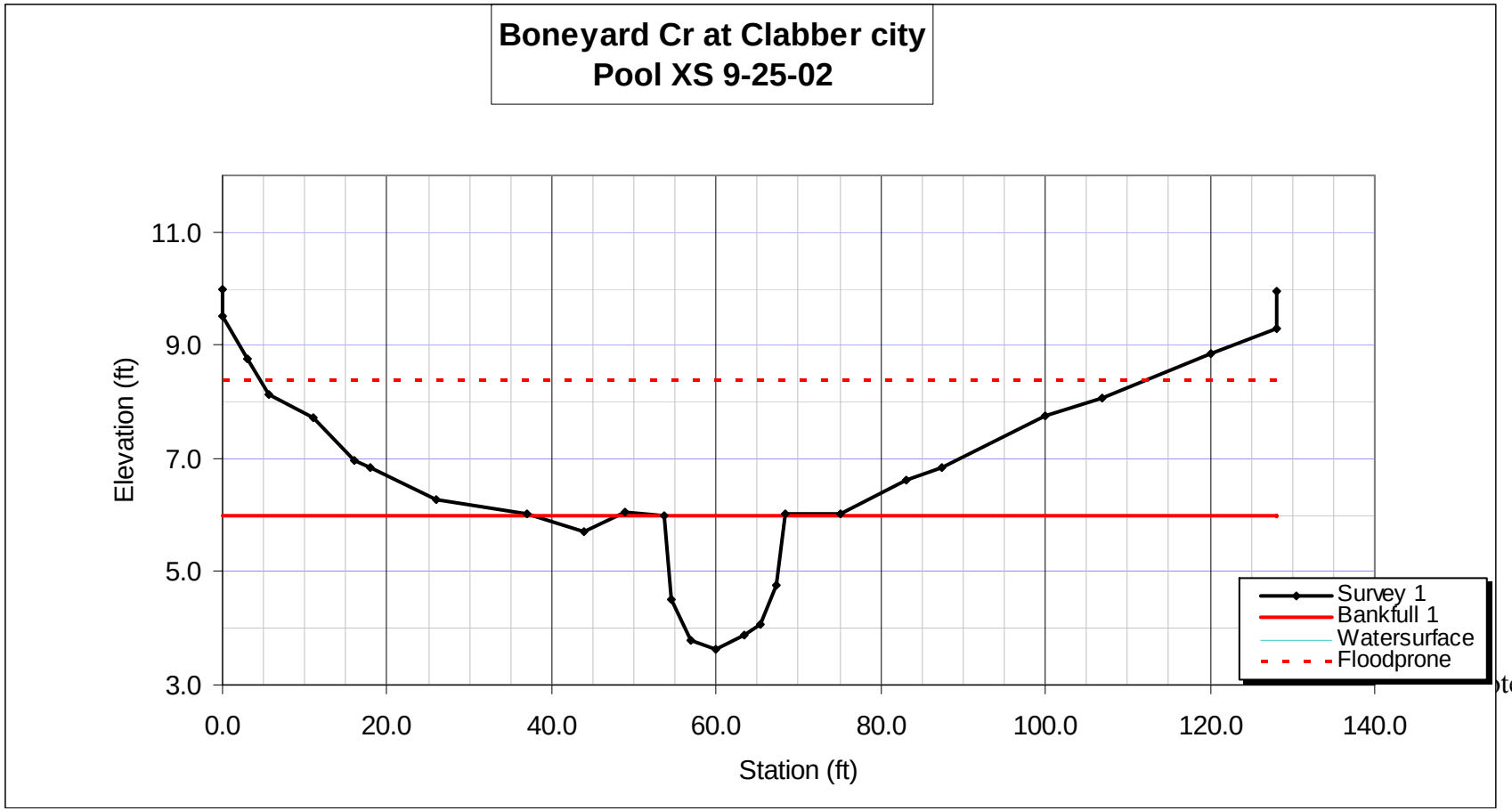
Notes:_____

Be sure to collect near bank slope, distance and water depth for near bank stress calculation.

Distance; Point; or	Backsight; rod reading @reference point	Height of Instrument; Ref. Pt + backsight	Fore-Sight	Elevation of Stream Bottom; HI - FS	Water depth; read from rod	Water surface elevation	NOTES, COMMENTS, OR REMARKS
STATION ft.	BS ft.	HI ft.	FS ft.	ELEV. ft.	WATER DEPTH ft.	WATER ELV. ft.	
							Near Bank Stress - top of _____(habitat)
							Near Bank Stress - bottom of _____

Figure 2. ADEQ cross-section survey field form.

Figure 3 Example of Cross-section survey.



3.16.3 Longitudinal Profile Procedure

The procedure described here is an extract of that described in Stream Channel Reference Sites: An Illustrated Guide to Field Technique (Harrelson, et al, 1994). For a detailed description of the procedure, see that publication.

The placement of the laser level and tripod should be planned in advance such that as many points along the channel can be taken without using a turning point. When a turning point is required there is the possibility of introducing error to the measured profile. The profile is best measured with two observers; one to operate the rod and one record the measurements on the field form (Figure 4). If a total station is used, the recorder also operates the total station.

The ideal location for measuring a profile is one that has unobstructed views from the laser level to the extendable rod; a condition that may only exist on meadow streams and channels that have been denuded of vegetation. Since unobstructed views along stream channels are rare, a survey of the selected reach should be conducted to ascertain the best placement of the laser level, usually at a high point (e.g., high bank or hill), and one that the extendable rod can reach for interception of the laser beam. Where high points are unavailable or dense vegetation exists, some clearing of tree limbs and brush may be required, but kept to a minimum.

The length of the profile reach is usually determined by the need for the measurements. At a minimum, the profile should not be less than two times the bankfull width or two channel meander wavelengths. A long profile reach will better represent the slope of the profile and minimize local disturbances in the channel.

If a benchmark of known elevation is available, then the benchmark is the elevation reference point (backsight; BS) from which all feature elevations (foresight; FS) are determined. The "…height of instrument (HI) is the elevation of the line of sight projected by the instrument. It can be found by adding the backsight rod elevation to the known (or assumed) elevation of the benchmark or the point on which the backsight was taken" (Harrelson et al, 1994). Where benchmarks are available, the elevation of the first feature is the elevation of the reference point. Typically, this is given an elevation of 100 feet. If the laser beam is 4.5 feet above the reference elevation, the height of instrument is 104.5 feet. All foresight rod readings are subtracted from the height of instrument to obtain feature elevations.

As the observers move downstream and move out of range of the laser level, a turning point must be made to continue the survey. A turning point (TP) "…is a reliable point upon which a foresight is taken to establish elevation. A backsight is then made to establish a new HI (Height of Instrument) and to continue a line of levels. The turning point retains the same elevation while the instrument is moved. Set the rod on a turning point (the last measured elevation) and record the foresight. Move the instrument as the rod stays in place. Make a backsight and record it" (Harrelson et al, 1994). It is critical when employing a turning point to continue a survey that the rod stay in place at the last measured elevation as the instrument is repositioned; otherwise, the chin of instrument set-ups is interrupted and all measurements and relative elevations below the last rod position will be in error.

The survey begins at the top of the reach where the end of fiberglass tape is anchored and the tape laid along the path of the thalweg. Measured elevation points, recorded as rod height, are determined by channel morphology. The distance from the end of the tape to the measured features is recorded as stations. When the reel end of the tape has been reached, the tape is again repositioned along the thalweg for the next downstream leg of the survey. This procedure is repeated until the entire reach has been surveyed. Water surface, banks, channel and/or water edges, and terraces are typically measured at the same stations as the bed features. It is important this process not be rushed and that enough time be allocated to complete the entire profile. Care should be taken to locate, as accurately as possible, the location of a new feature and the maximum depth of a pool. The latter measurement is especially important if pool facet slopes (Section 3B, 3.16.14) are to be calculated off the profile.

An alternative method of measuring a profile is a "leapfrogging" method where the length of each feature is measured separately. This method is best employed with three observers and two extendable rods. The first observer measures the elevation of point one (e.g., top of pool) and holds one end of the tape. The second observer moves to the second feature point (e.g., maximum pool depth) with the other end of the tape. The elevation of the second point is measured as well as the distance between the two features. The third observer records the feature callout, length, and rod height measurements. The first observer then moves to the third point (e.g., top of run) bringing the end of the tape to the next feature, where the elevation and feature length is measured. The "leapfrogging" continues to the end of the reach. Water surface, low banks, bankfull, and terraces are typically measured at the same stations as the features or wherever good indicators exist along the profile.

If a benchmark of known elevation is available, then the benchmark is the elevation reference point (backsight; BS) from which all feature elevations (foresight; FS) are determined. The "…height of Instrument (HI) is the elevation of the line of sight projected by the instrument. Find it by adding the backsight rod reading to the known (or assumed) elevation of the benchmark or the point on which the backsight was taken"
(Harrelson et al, 1994). Where benchmarks are unavailable, the elevation of the first feature is the elevation reference point. Typically, this is given an elevation of 100 feet. If the laser beam is 4.5 feet above the reference elevation, the height of instrument is 104.5 feet. All foresight rod readings are subtracted from the height of instrument to obtain feature elevations.

From the field profile survey form (Figure 4), elevations of geomorphic features are calculated. Plots of the profile are made with either RiverMorph or Excel software from which geomorphic analyses are made.

3.16.3.1 Literature Cited

Harrelson, C.C., C.L. Rawlins, and J.P. Potyondy. 1994. Stream channel reference sites: an illustrated guide to field technique. Gen. Tech. Rep. RM-245. U.S. Department of Agriculture, Forest Service, Rocky Mountain Forest and Range Experiment Station. 61pp.

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Longitudinal Profile

Site ID:_____ Site Description:_____

Date:_____ Watershed:_____ Stream Type:_____ Observers:_____

Notes:_____

	Backsight; rod reading @ reference point	Height of Instrument; Ref. Pt + backsight	Foresight	Elevation of Stream Bottom; HI - FS	Water depth; read from rod		
STA. ft.	BS ft.	HI ft.	FS ft.	ELEV. ft.	WATER DEPTH, ft.	BANK HEIGHT, ft.	Remarks

Figure 4 Longitudinal Profile Survey Field Form.

3.16.4 Channel Vertical Stability Assessment using the Bank Height Ratio

A channel that is entrenched or vertically contained cannot access its floodplain and will experience bank erosion at bankfull or greater flows. The bank height ratio (BHR) is a measure of the degree of incision of the study channel. Bank height ratio is defined as the ratio of the low bank height divided by the maximum bankfull depth at that cross-section (Rosgen, 2001a). Low bank height is any substantial bank below bankfull maximum height. The ratio can be calculated from a single riffle or pool cross-section survey. Ideally, a longitudinal profile delineating numerous low bank and maximum bankfull heights provides an accurate bank condition assessment. Use the maximum value of all available BHR data in the study reach for the final scoring criterion as shown in Table 2. The scoring criterion for BHR is used to evaluate the range of conditions from vertically stable to unstable, as per Rosgen (2001b).

Table 2. Bank height ratio scoring criteria.

Bank height ratio	Stability rating	Sediment supply category	Sediment supply score
1.0 – 1.1	Stable	Low	2
1.11 – 1.3	Moderately unstable	Moderate	5
>1.3	Unstable	High	10

3.16.4.1 Bank Height Ratio Procedure

1. Conduct cross-section surveys at both riffle and pool cross-sections, identifying the low bank height and bankfull elevations. Then divide the low bank height reading by the bankfull maximum depth at the cross-section to obtain the bank height ratio for each respective location.

2. Conduct a longitudinal profile, incorporating measurements of low bank heights and bankfull elevations. Plot the profile data. Then calculate bank height ratio wherever low bank height data points were collected, using the bankfull elevation line plotted on the graph.

3. Use Rosgen's scoring criteria in Table 2 to determine the stability rating, sediment supply category and sediment supply score. The sediment supply score is then used in the final 6-parameter stability assessment score.

3.16.4.2 Literature Cited

Rosgen, D.L. 2001a. A stream channel stability assessment methodology. Wildland Hydrology. Pagosa Springs, CO.

Rosgen, D.L. 2001b. Stream stability and sediment supply assessment protocols. Level III Short Course materials. Wildland Hydrology. Pagosa Springs, CO.

3.16.5 Channel Lateral Stability Using the Bank Erodibility Hazard Index and Near Bank Stress Ratios

Bank erosion is congruent with natural river processes; however, lateral migration rates of stream channels can be accelerated when variables controlling bank erosion processes are altered. The Bank Erosion Hazard Index (BEHI) provides an estimate of bank stability or stream-bank erosion potential through measurement of eight parameters. Rosgen's BEHI index (Rosgen, 1996) addresses the ability of stream-banks to resist erosion through measurement of the following variables:

1. Streambank height to bankfull height ratio
2. Riparian vegetation rooting depth to streambank height ratio
3. Rooting density
4. Stream bank angle or slope
5. Bank surface protection by debris and vegetation
6. Composition of bank materials
7. Stratification of bank materials and presence of soil lenses
8. Shear stress in the near bank region.

The first seven variables are scored according to Rosgen's BEHI method at riffle and pool cross-sections. As a final step, near bank stress is (NBS) calculated. A reach BEHI assessment is then performed. Additional BEHI /NBS surveys are performed if different bank types are encountered. The number of linear feet in each BEHI category is multiplied by the predicted erosion rate for that bank type and summed for an estimate of soil loss for the whole study reach. A predicted bank erosion rate is obtained by locating the BEHI and NBS categories on a graph of empirical bank erosion data. Graphs developed by Rosgen integrate the potential for bank erosion with the sheer stress directed at the bank by stream flow (Near Bank Stress) to predict an average annual erosion rate. The dominant BEHI and NBS categories determine which sediment supply category applies, as shown in the scoring criteria in Table 3 (Rosgen, 2001c).

Table 3. Bank Erosion Hazard Index and Near Bank Stress scoring criteria.

Dominant categories		Stability rating	Sediment supply category	Sediment supply score
BEHI	**NBS**			
Low	Low	Stable	Low	5
Low	Moderate			
Low	High	Moderately Unstable	Moderate	10
Low	Extreme			
Moderate	Low			
Moderate	Moderate			
Moderate	High	Unstable	High	20
Moderate	Extreme			
High	Moderate			
High	High			
High	Extreme			
Extreme	Extreme			

Required equipment: Laser level, two surveying rods, receiver, bubble levels, and Bank angle indicator/compass.

3.16.5.1 Bank Erosion Hazard Index (BEHI) Procedure

This procedure provides a description for each parameter in the BEHI as well as for the Near Bank Stress (NBS). The BEHI model uses several physical attributes of a bank to determine Bank Erosion Potential (BEP). Bankfull stage is used as a common point for this characterization. Bankfull stage is used as a common reference point because it is the primary channel-shaping or channel flow in the Rosgen system and can be identified in the field. The study bank, for purposes of the BEHI evaluation, is defined as a three foot wide section of bank that is intersected by bankfull stage. The field form used for assessing bank erosion potential is shown in Figure 5. Bank Erosion Potential is determined using the following field protocols.

1. Total bank height divided by bankfull height (ft/ft)

Total bank height is the vertical measurement from base of bank to top of bank. All bank height measurements begin where the slope of the bank meets the bed. Measure the height of the continuous bank intersected by bankfull stage. If a second bank is set back in a terrace away from the bankfull bank, it is not included in measuring height. Bank height is most accurately measured using a laser level and surveying rod and receiver, as part of a cross-section survey. Bank height can alternatively be measured with two rods and bubble levels as part of a bank profile, though this method is less accurate.

To calculate this metric, total bank height is divided by the bankfull stage at that bank profile location. Both values plus the metric value must be recorded on the BEHI form.

2. Root depth divided by bank height (ft/ft)

Root depth is defined as the depth of root mass providing stability to the bank. Long individual roots are not considered in root depth because these are not thought to add significant strength to the bank. On sloping or vegetated banks, overall root depth is generally assumed to be equivalent to the bank height, but is verified by surficial digging and exposing the roots of the dominant vegetation. Root depth is most accurately measured from the top of bank to the bottom root depth using a laser level and surveying rod and receiver, as part of a cross-section survey. Rooting depth can alternatively be measured with two rods and bubble levels as part of a bank profile.

To calculate this metric, rooting depth is divided by the total bank height at that cross-section. The root depth plus the metric value should be recorded on the BEHI form.

3. Root Density (%)

The root density is defined as the ratio of the root mass surface area to the total bank surface area. Root density on sloping, well-vegetated banks (native grasses, sedges and riparian trees) is typically near 100%. The root density value will be similar to the surface protection value when banks are well protected by vegetation. In cut banks with high erosion potential, there may be some root density above the bankfull elevation, but none below it. In such cases, make a weighted estimate of the whole bank root density by considering both the density in the upper soil layer and also the rooting depth. Scrape away the loose soil to expose roots, in order to properly evaluate root density. Make a visual estimate of root density, using rooting depth to assist the evaluation in cut banks

4. Bank Slope (ft/ft)

Bank slope or angle is measured in degrees from the horizontal. Bank slope is defined as rise over run from the toe of the bank to the top of the continuous bank intersected by bankfull stage. A vertical bank is 90 degrees. Bank slope can be measured with a clinometer, a compass, or a bank angle indicator. It can also be calculated from a cross-section graph, using the formula of Δ rise/Δ run. If there are two distinctly different bank angles on a bank, use the steepest slope to evaluate this parameter.

5. Surface Protection (%)

Surface protection is defined as the percentage of the bank surface protected by vegetation, root wads, large boulders or other resistant materials which protects the bank from eroding due to freeze and thaw action. Materials such as car bodies or uprooted tree trunks, which may contribute to bank erosion by redirecting high velocity toward the bank are not included as surface protection. Surface protection on sloping, well-vegetated banks is generally considered to be near 100%. In alluvial streams, surface protection will be similar to root density values.

6. Bank Materials

The BEHI score is adjusted for composition of bank materials. Where banks are composed of bedrock and boulders, no adjustment is made. Where banks are dominated by cobble, subtract 10 points from the BEHI score. Where cobble banks consist of a mixture of sand and gravel, no adjustment in BEHI score is made. For gravel dominated banks, add 5 points to the BEHI score. For banks dominated by sand, add 10 points. For silt/clay dominated banks no adjustment to BEHI score is made.

7. Bank Stratification

The BEHI score is adjusted for soil stratification layers which contribute to bank erosion potential. In a cut bank, unstable layers of sand and gravel, especially at or below the bankfull level, contribute to bank erosion potential. Add 5-10 points depending on the number of layers and position of the unstable layers in relation to the bankfull stage. Typically, five points are added when there is one unstable layer, and ten points for two or more unstable layers.

8. Calculation of BEHI score

Rosgen (1996) developed a 1-10 scoring system for each of the seven metrics, with a sum total BEHI score of 5 - 50 (Table 4). This scoring system is included in the specialized BEHI field form (Rosgen, 2001a) and is shown in Figure 5. The total BEHI scores give the following rating categories.

Table 4. Bank Erosion Hazard Index rating categories.

BEHI Total Score	BEHI Category
5 – 9.5	Very Low
10 – 19.5	Low
20 – 29.5	Moderate
30 – 39.5	High
40 – 45	Very High
46 - 50	Extreme

Bank Erodibility Hazard Index - Rating Guide

Stream | Reach | Date | Crew

Bank (LB or RB) | Bank Description

Bank Erosion Potential		Bank Height (ft): / Bankfull Height (ft):	Bank Height/ Bankfull Ht	Root Depth/ Bank Height	Root Density %	Bank Angle (Degrees)	Surface Protection%
VERY LOW	Value		1.0-1.1	1.0-0.9	100-80	0-20	100-80
	Index		1.0-1.9	1.0-1.9	1.0-1.9	1.0-1.9	1.0-1.9
	Choice		V: I:	V: I:	V: I:	V: I:	V: I:
LOW	Value		1.11-1.19	0.89-0.5	79-55	21-60	79-55
	Index		2.0-3.9	2.0-3.9	2.0-3.9	2.0-3.9	2.0-3.9
	Choice		V: I:	V: I:	V: I:	V: I:	V: I:
MODERATE	Value		1.2-1.5	0.49-0.3	54-30	61-80	54-30
	Index		4.0-5.9	4.0-5.9	4.0-5.9	4.0-5.9	4.0-5.9
	Choice		V: I:	V: I:	V: I:	V: I:	V: I:
HIGH	Value		1.6-2.0	0.29-0.15	29-15	81-90	29-15
	Index		6.0-7.9	6.0-7.9	6.0-7.9	6.0-7.9	6.0-7.9
	Choice		V: I:	V: I:	V: I:	V: I:	V: I:
VERY HIGH	Value		2.1-2.8	0.14-0.05	14-5.0	91-119	14-10
	Index		8.0-9.0	8.0-9.0	8.0-9.0	8.0-9.0	8.0-9.0
	Choice		V: I:	V: I:	V: I:	V: I:	V: I:
EXTREME	Value		>2.8	<0.05	<5	>119	<10
	Index		10	10	10	10	10
	Choice		V: I:	V: I:	V: I:	V: I:	V: I:
V = value, I = index					**SUB-TOTAL** (Sum one index from each column)		

Bank Material Description:

Bank Materials

Bedrock (Bedrock banks have very low bank erosion potential)
Boulders (Banks composed of boulders have low bank erosion potential)
Cobble (Subtract 10 points. If sand/gravel matrix greater than 50% of bank material, then do not adjust)
Gravel (Add 5-10 points depending percentage of bank material that is composed of sand)
Sand (Add 10 points)
Silt Clay (+ 0: no adjustment)

BANK MATERIAL ADJUSTMENT

Stratification Comments:

Stratification

Add 5-10 points depending on position of unstable layers in relation to bankfull stage

STRATIFICATION ADJUSTMENT

VERY LOW	LOW	MODERATE	HIGH	VERY HIGH	EXTREME
5-9.5	10-19.5	20-29.5	30-39.5	40-45	46-50

Bank location description (circle one)
Straight Reach Outside of Bend

GRAND TOTAL
BEHI RATING

Figure 5. Bank Erodibility Hazard Index (BEHI) field form by Rosgen (2001a).

3.16.5.2 Near Bank Stress Procedure

The erosive force against a bank is actually the shear stress contributed by the stream flow. Shear Stress in a stream is defined as the specific weight of water multiplied by the hydraulic radius and water surface slope. Slope should refer to the water surface at bankfull stage. Hydraulic radius is defined as the cross-sectional area divided by the wetted perimeter. Bank erosion potential increases directly with increases in the ratio of near bank stress to channel shear stress.

There are several methods for estimating or calculating near bank stress for any given bank in the study reach. Rosgen (2003) identified seven methods, associated with Level I through Level IV field assessment procedures. The Near Bank Stress procedure adopted for ADEQ use from the Rosgen suite of methods is given below. In the procedure, near bank stress is characterized by a ratio of the shear stress in the 1/3 of the channel nearest the eroding bank to the entire cross-sectional shear stress. The following measurements are required to calculate the shear stress ratios:

1. mean depth at a cross-section
2. average water surface slope for reach
3. water surface slope in near bank region for that habitat type (e.g. pool), and
4. near bank maximum depth.

This ratio of near bank to mean channel shear stress is then categorized similarly as the BEHI scores, as very low to extreme, as shown in Table 5.

Table 5. Near Bank Stress rating categories.

NBS Ratio	NBS Category
<0.8	Very Low
0.8 – 1.05	Low
1.06 – 1.14	Moderate
1.15 – 1.19	High
1.2 – 1.6	Very High
>1.6	Extreme

1. Field Measurements

A cross-section survey is needed to calculate cross-sectional area, wetted perimeter and hydraulic radius at bankfull elevation. A longitudinal profile is also needed to calculate the water surface slope of the reach and the water surface slope in the near bank region. Average water surface slope is measured along the thalweg of the whole study reach, beginning and ending at the top of riffle feature. The water surface slope in the near bank region is measured at the top and bottom of the bed feature in which the cross-section lies (e.g. Riffle, run or pool

habitat feature). Elevation measurements are collected at the thalweg on the stream bottom and at the water surface at the upstream end of the feature, such as top of pool and also at the downstream end of the feature, below the cross-section. If the stream is dry, take thalweg measurements one bankfull width upstream and downstream of the cross-section location.

2. Calculations

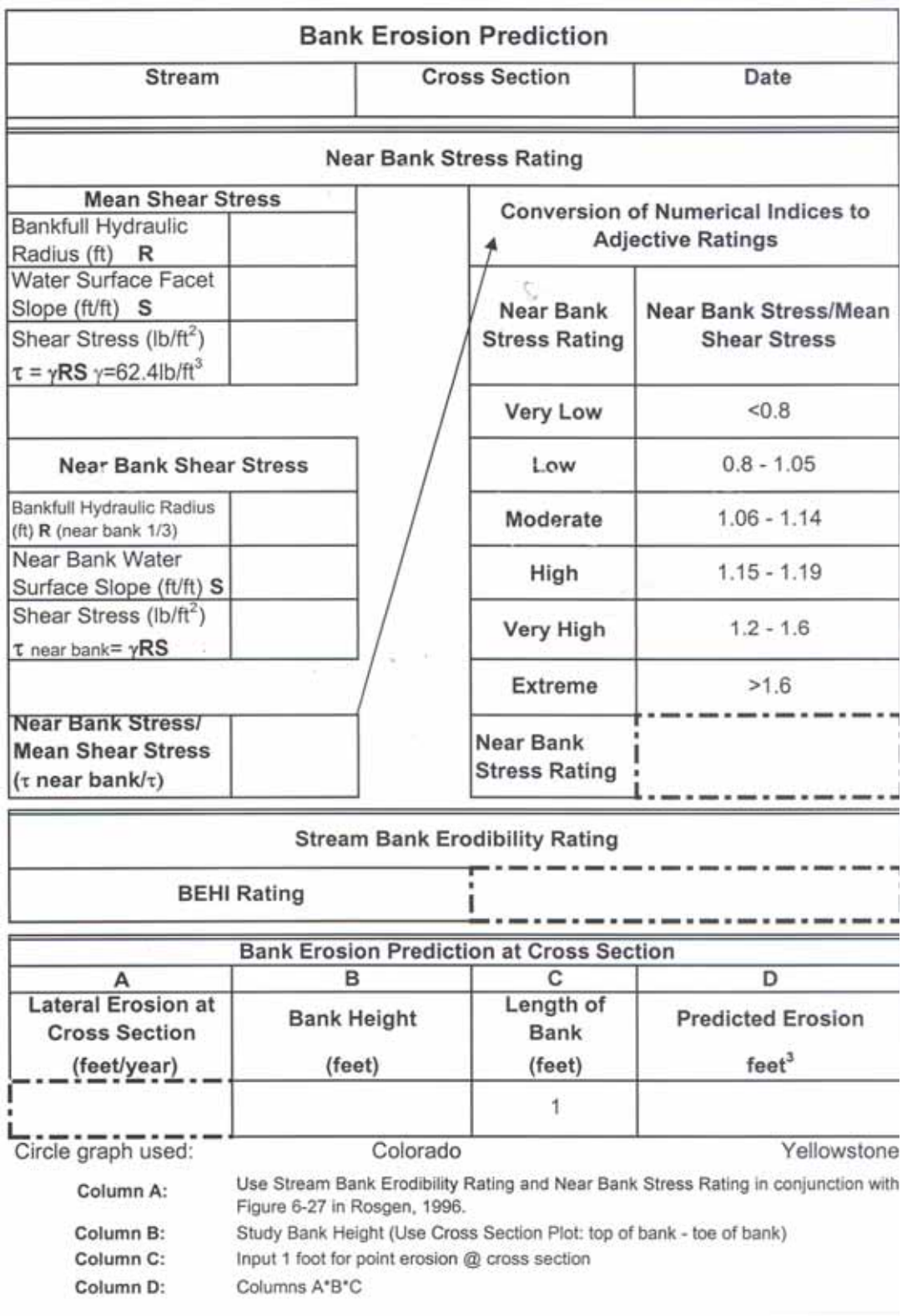
After plotting the cross-section profile, extract the necessary variables to calculate mean shear stress and near bank stress. Cross-sectional area and mean depth are first calculated. Bankfull mean depth is the ratio of bankfull cross-sectional area to bankfull width. Then, hydraulic radius of both the channel and the near bank region is calculated by dividing cross-sectional area by wetted perimeter (width plus 2 times mean depth). Values for the near bank shear stress and average reach shear stress are calculated by multiplying water surface slope, hydraulic radius, and the weight of water (Rosgen, 2001c). The ratio of near bank shear stress to average shear stress is calculated and the NBS category determined from the ratio indicated in Table 6. Alternately, measurements can be entered into "RiverMorph" software, which performs the shear stress calculations (RiverMorph LLC, 2002).

3. Predicted bank erosion

Rosgen (1996) produced two different graphs to integrate the BEHI and NBS scores and categories into a predicted bank erosion rate (Figure 7). The predicted bank erosion rate (ft/year) is plotted against both the NBS category and BEHI regression line in these graphs. This predicted lateral erosion rate is then multiplied by the bank height and length in the reach for a bank soil loss estimate (ft3/year) as shown in the form in Figure 6. This predicted soil loss estimate is then determined for each bank type in the reach and summed for a total soil loss estimate for the entire reach (Figure 8). Moody et al. (2003) determined that the Colorado USFS 1989 graph (Figure 7) worked best in Arizona for predicting bank erosion rates, which were verified in a BEHI validation study of over 40 sites in the Verde and San Pedro River basins.

4. Measured bank erosion

Actual bank erosion rates are measured with the use of multiple bank pins placed along eroding banks. Pin lengths protruding from the bank are remeasured after major storm and flow events. One or more bank pins (4' x 3/8" smooth rebar) are installed horizontally into bank material at bankfull elevation or above and below bankfull at each cross-section. Each cross-section represents a different common bank type occurring in the reach. In addition, bank pins are placed at upstream and downstream locations from the cross-section location in order to obtain an average bank loss from a minimum of three bank pins in that bank type. The length of the bank pin exposed after each flood event indicates the amount of bank material lost at that cross-section. The average bank loss is then multiplied by the length of that bank type for the total erosion rate (ft3/yr).



Bank Erosion Prediction

Stream	Cross Section	Date

Near Bank Stress Rating

Mean Shear Stress	
Bankfull Hydraulic Radius (ft) **R**	
Water Surface Facet Slope (ft/ft) **S**	
Shear Stress (lb/ft^2) $\tau = \gamma RS$ γ=62.4lb/ft^3	

Near Bank Shear Stress	
Bankfull Hydraulic Radius (ft) **R** (near bank 1/3)	
Near Bank Water Surface Slope (ft/ft) **S**	
Shear Stress (lb/ft^2) τ near bank= γRS	
Near Bank Stress/ Mean Shear Stress (τ near bank/τ)	

Conversion of Numerical Indices to Adjective Ratings

Near Bank Stress Rating	Near Bank Stress/Mean Shear Stress
Very Low	<0.8
Low	0.8 - 1.05
Moderate	1.06 - 1.14
High	1.15 - 1.19
Very High	1.2 - 1.6
Extreme	>1.6
Near Bank Stress Rating	

Stream Bank Erodibility Rating

BEHI Rating	

Bank Erosion Prediction at Cross Section

A	B	C	D
Lateral Erosion at Cross Section (feet/year)	Bank Height (feet)	Length of Bank (feet)	Predicted Erosion feet^3
		1	

Circle graph used: Colorado Yellowstone

Column A: Use Stream Bank Erodibility Rating and Near Bank Stress Rating in conjunction with Figure 6-27 in Rosgen, 1996.

Column B: Study Bank Height (Use Cross Section Plot: top of bank - toe of bank)

Column C: Input 1 foot for point erosion @ cross section

Column D: Columns A*B*C

Figure 6. Near Bank Stress (NBS) rating and Bank erosion prediction form by Rosgen (2001a).

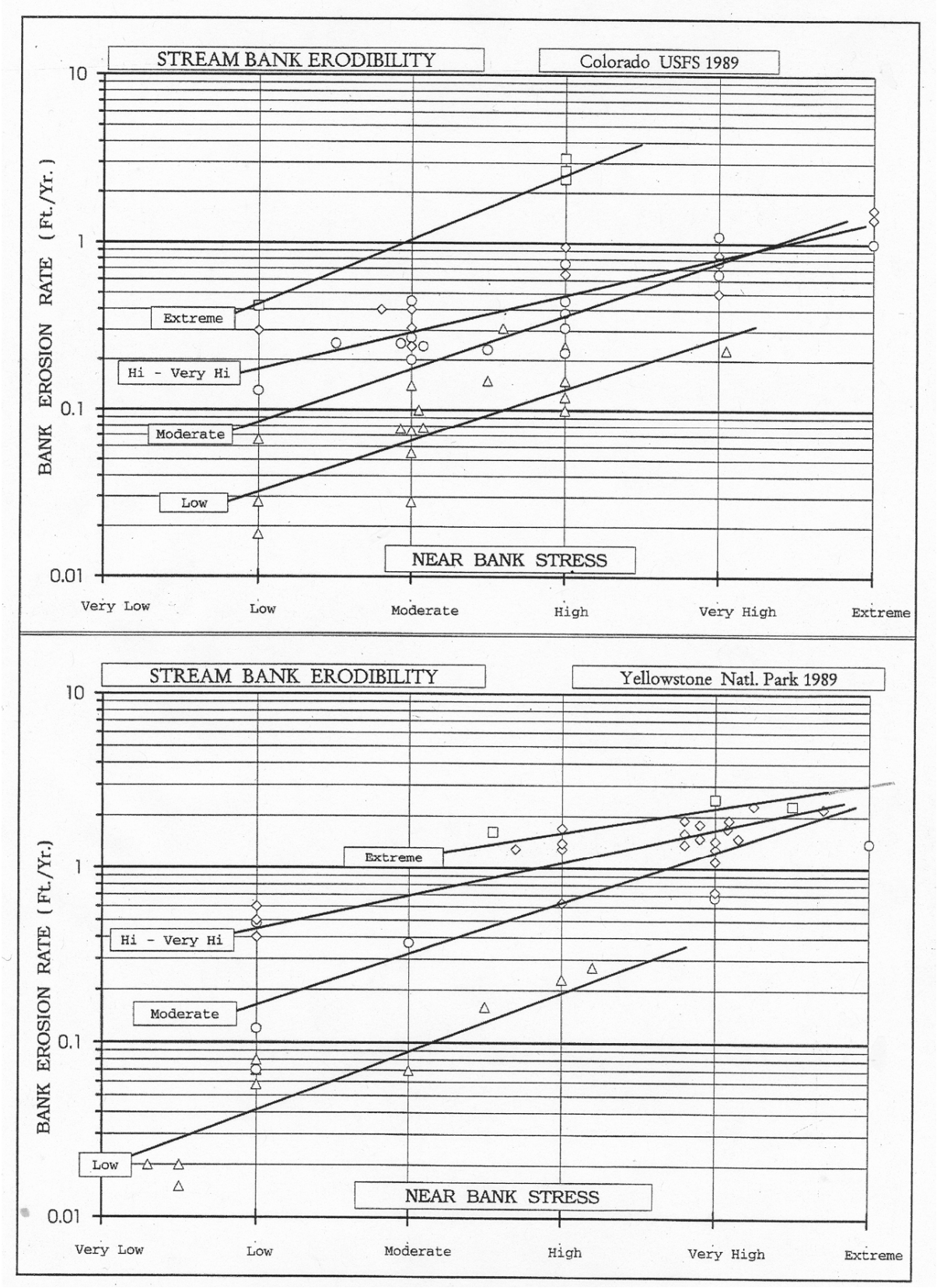


Figure 7. Bank erosion rate prediction graphs by Rosgen (1996).

Total Bank Erosion Calculation

Stream:			Total Bank Length:		Stream Type:	
Observers:			Date:		Graph Used:	
	BEHI (adjective)	**Near Bank Stress (adjective)**	**Erosion Rate (ft/yr)**	**Length of Bank (ft)**	**Bank Height (ft)**	**Erosion Sub-Total (ft^3/yr)**
1						
2						
3						
4						
5						
6						
7						
8						
9						
10						
11						
12						
13						
14						
15						
I. Sum erosion sub-totals for each BEHI/NBS combination					**Total Erosion (ft^3/yr)**	
II. Divide total erosion ($feet^3$) by 27 $feet^3/yard^3$					**Total Erosion (yd^3/yr)**	
III. Multiply Total Erosion ($yard^3$) by 1.3 (conversion of yd^3 to tons for average material type)					**Total Erosion (tons/year)**	

Figure 8. Reach assessment form for predicted bank erosion rates and quantities (Rosgen, 2001a).

3.16.5.3 Literature Cited

Moody, T., M. Wirtanen, and S.N. Yard. 2003. Validating the Bank Erodibility Hazard Index in Central and Southern Arizona. Prepared for the Arizona Department of Environmental Quality. Phoenix, AZ.

RiverMorph, 2002. RiverMorph User's Manual v2.0. Louisville, KY (www.RiverMorph.com)

Rosgen, D. and H.L. Silvey. 1996. Applied River Morphology. Wildland Hydrology. Pagosa Springs, CO.

Rosgen, D. 2001a. Abridged River assessment and morphology (RAM) forms. Wildland Hydrology. Pagosa Springs, CO.

Rosgen, D.L. 2001b. A stream channel stability assessment methodology. Wildland Hydrology. Pagosa Springs, CO.

Rosgen, D.L. 2001c. Stream stability and sediment supply assessment protocols. Level III Short Course materials. Wildland Hydrology. Pagosa Springs, CO.

Rosgen, D.L. 2003. Seven methods for estimating Near-Bank Stress in natural channels. Wildland Hydrology. Pagosa Springs, CO.

3.16.6 Channel Pattern

A well developed and appropriate meander pattern is important for maintenance of stream channel stability. Channel pattern measurements include: meander length, belt width, radius of curvature, meander width ratio, arc length, arc angle, ratio of radius of curvature to bankfull width, ratio of meander length to bankfull width, and sinuosity. These data are determined from measurements of stream channel characteristics obtained from ortho-photo quad maps or aerial photos. Measurements can alternately be collected in the field, but only in locations where the lack of vegetation allows.

Channel pattern parameters are important for comparison of study sites to reference sites and for restoration design; however, they are not scored and used in Rosgen's channel stability index. When the reference reach data for the same valley and stream type differ in watershed size, values for channel pattern parameters can be converted to dimensionless ratios by dividing by bankfull width. A large departure from meander reference values indicates instability as the stream channel evolves to a more stable form (e.g. lateral instability results from inadequate meander width or small radius of curvature ratios). Down-valley meander migration and excessive near-bank stress due to ratios of radius of curvature/bankfull width values of less than 2.0 indicate channel adjustment due to instability.

3.16.6.1 Channel Pattern Procedures

1. A narrative description of meander pattern is selected from Rosgen's (1996) Level III form "Meander Patterns" (Figure 9). Identification of meander pattern is best performed by inspection of recent aerial photographs or ortho-photo quads. Topographic maps can also be used but they typically provide only an historic perspective.

2. Sinuosity is defined as the ratio of meandering channel length to straight-line valley length and indicates how the stream has adjusted its slope to that of its valley. Sinuosity is best measured from recent aerial photographs or ortho-photo quads. Use a stream length that is at least two meander wavelengths for the measurement (Figure 10). A visual representation of different sinuosities is presented in Figure 11. The greatest sinuosity is found in a highly meandering "E" type alluvial channel, whereas the least sinuosity is common in "A" type high relief, entrenched streams.

3. Meander wavelength (L_m) is the linear downstream distance of one river meander as shown in Figure 12. Collect several field measurements or a minimum of three measurements from recent aerial photographs or ortho-photo quads. Calculate minimum, maximum and average values for ratio of meander wavelength / bankfull width. Distance values ranging from 10-14 bankfull widths are common for an individual meander wavelength.

4. Meander belt width (W_{blt}) is the width of a meander from outside bend to outside bend. Collect several measurements or a minimum of three measurements from recent aerial photographs or ortho-photo quads. Meander width ratios can then be calculated by dividing the belt width / bankfull width. Obtain minimum, maximum and average values.

5. The radius of curvature is used to evaluate channel resistance to erosion and meander migration rates. Collect several field measurements or a minimum of three measurements from recent aerial photographs or ortho-photo quads. Radius of curvature is defined by the following formula:

$$R_c = \frac{L_m K^{-1.5}}{13(K-1)^{0.5}}$$

where: R_c = radius of curvature in feet
L_m = meander length in feet
K = sinuosity

Another technique for field measurement is establishing the Chord length/mid-ordinate ratio as shown in Figure 12. Chord length is the measured distance between two points along the river meander. To determine the mid-ordinate, locate the mid-point of the chord, then

measure the length of the perpendicular middle ordinate. The formula for calculating radius of curvature from these measurements is as follows:

$$R_c = \frac{C2}{8M} + \frac{M}{2}$$

where: R_c = radius of curvature in feet
C = Chord length in feet
M = Mid-ordinate distance in feet

3.16.6.2 Literature Cited

Rosgen, D. and H.L. Silvey. 1996. Applied River Morphology. Wildland Hydrology. Pagosa Springs, CO.

Meander Patterns

Stream:	Reach:				
Date:	Observers:				
LIST ALL CATEGORIES THAT APPLY					

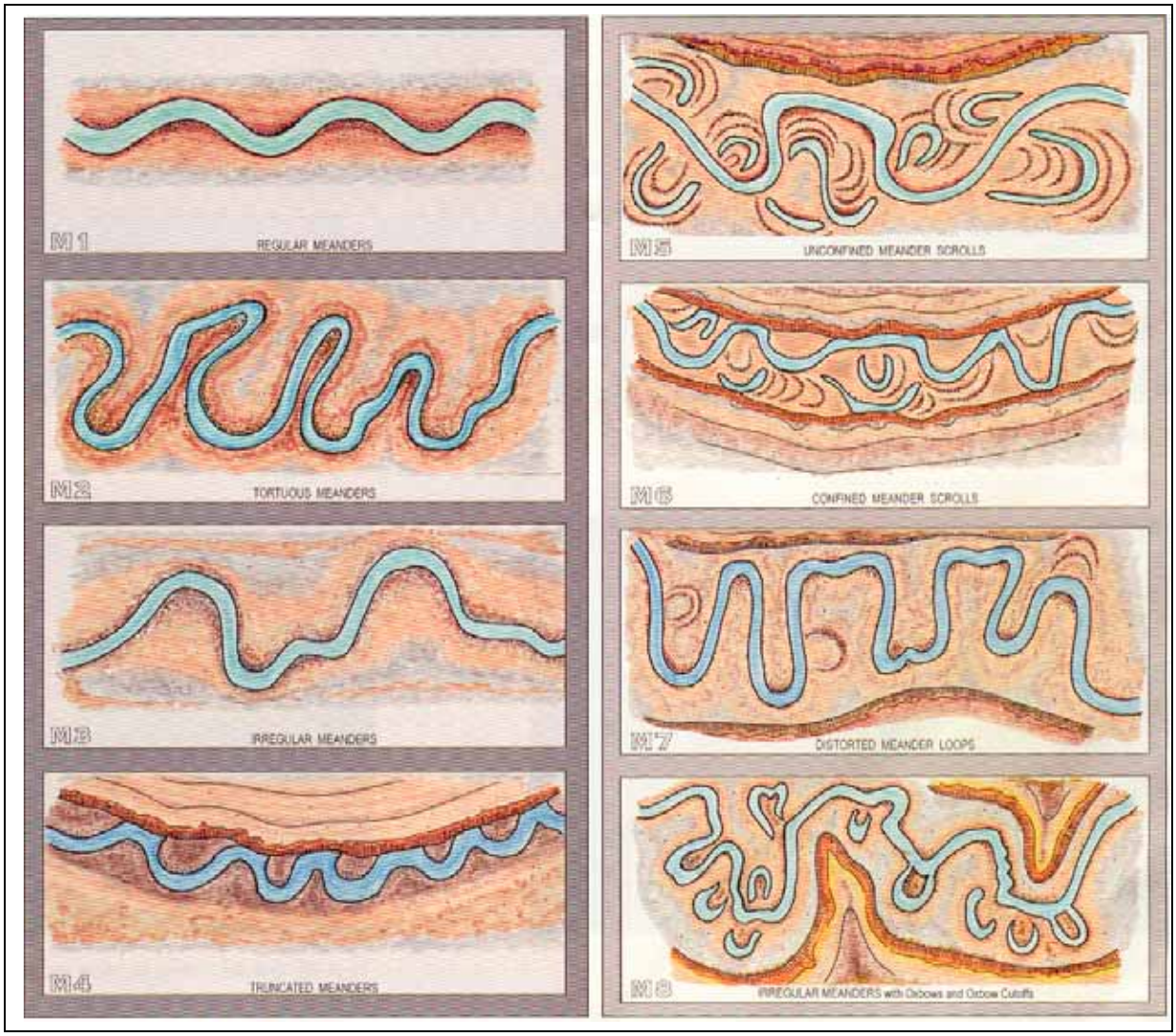


Figure 9. Narrative meander patterns of stream channels (Rosgen, 1996).

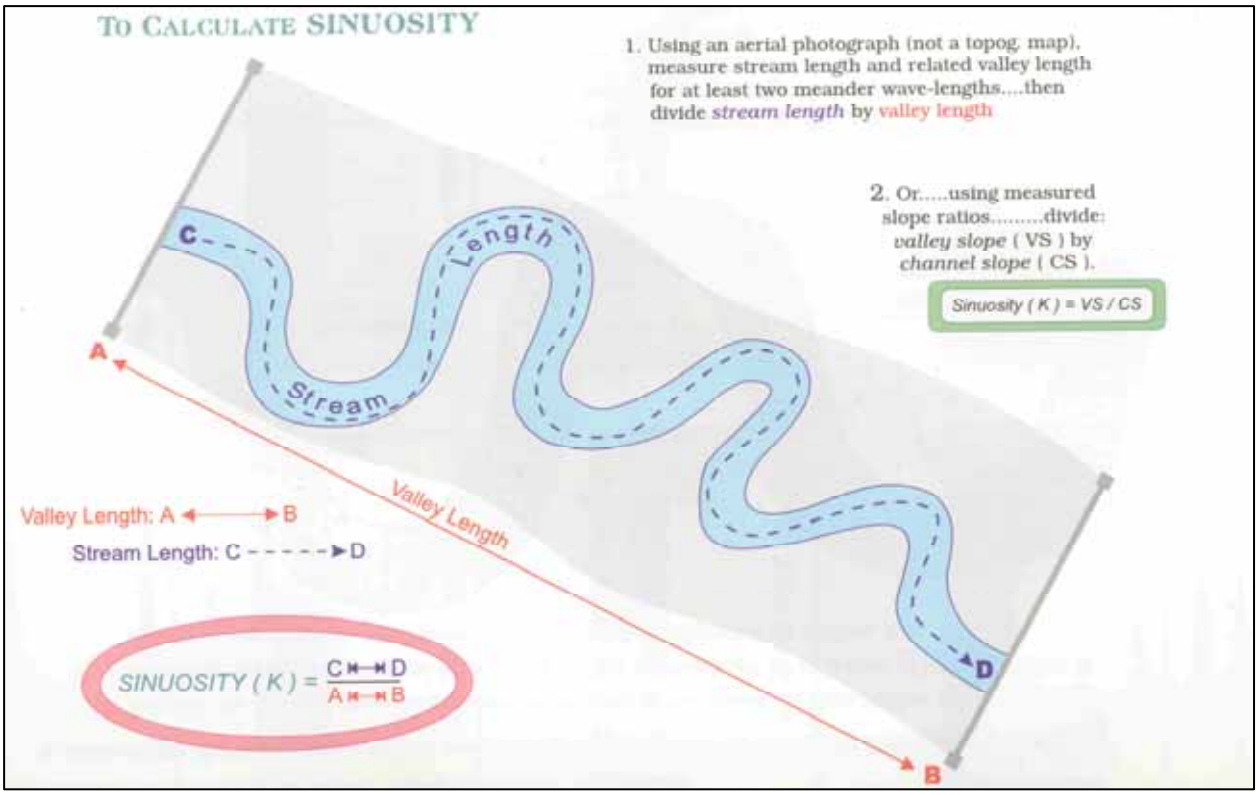


Figure 10. Measurements needed to calculate river sinuosity (after Rosgen and Silvey, 1998).

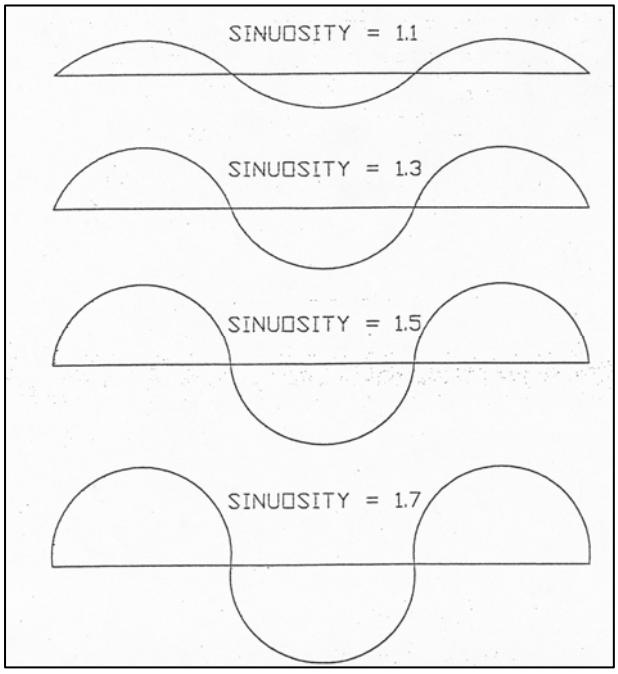
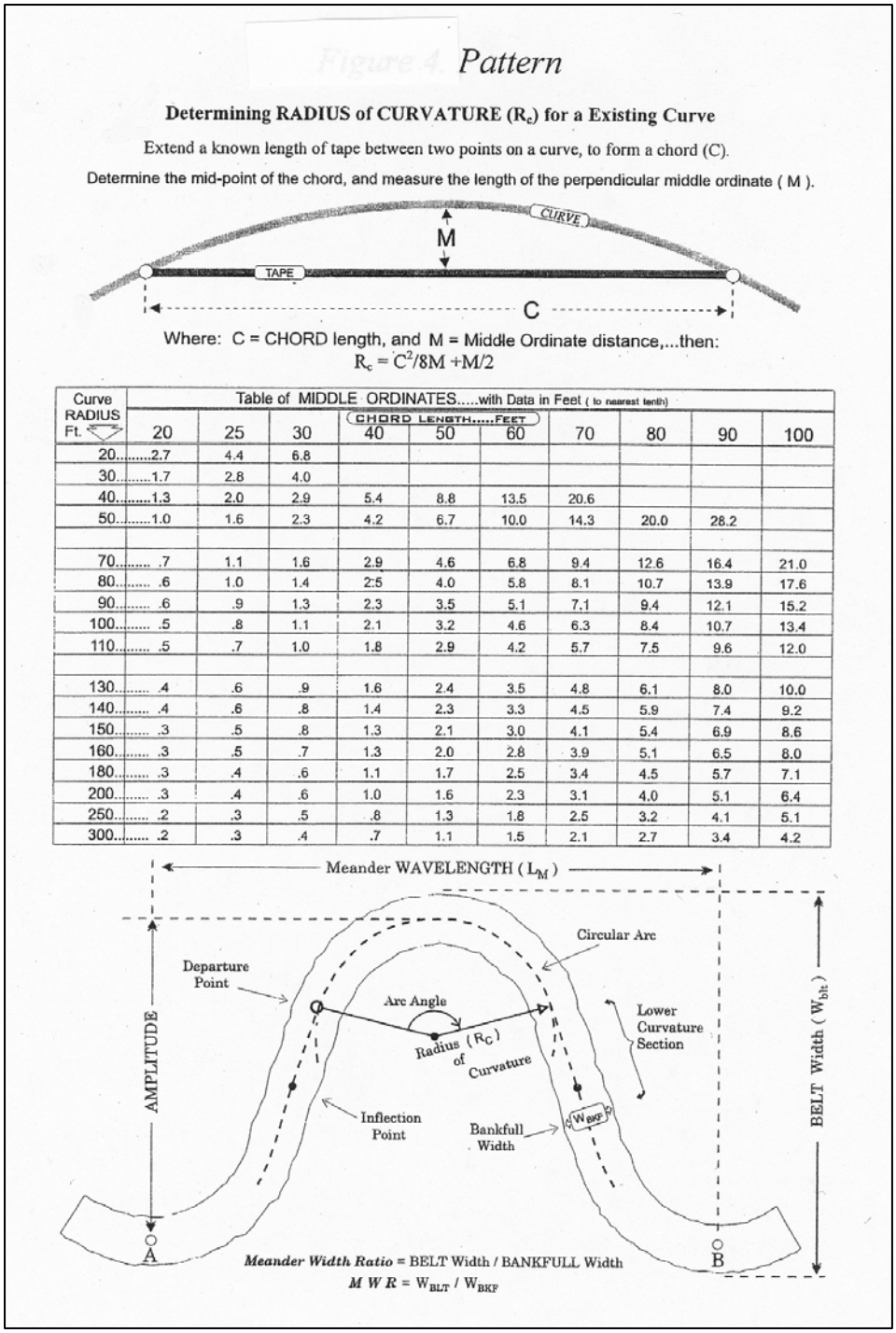


Figure 11. Examples of different channel sinuosities.

Figure 12. Measurements needed to calculate river meander geometry (Rosgen, D.L. 2001. Stream stability and sediment supply assessment protocols. Level III Short Course materials. Wildland Hydrology. Pagosa Springs, CO.).



Curve RADIUS Ft.	Table of MIDDLE ORDINATES.....with Data in Feet (to nearest tenth) CHORD LENGTH.....FEET									
	20	25	30	40	50	60	70	80	90	100
20	2.7	4.4	6.8							
30	1.7	2.8	4.0							
40	1.3	2.0	2.9	5.4	8.8	13.5	20.6			
50	1.0	1.6	2.3	4.2	6.7	10.0	14.3	20.0	28.2	
70	.7	1.1	1.6	2.9	4.6	6.8	9.4	12.6	16.4	21.0
80	.6	1.0	1.4	2.5	4.0	5.8	8.1	10.7	13.9	17.6
90	.6	.9	1.3	2.3	3.5	5.1	7.1	9.4	12.1	15.2
100	.5	.8	1.1	2.1	3.2	4.6	6.3	8.4	10.7	13.4
110	.5	.7	1.0	1.8	2.9	4.2	5.7	7.5	9.6	12.0
130	.4	.6	.9	1.6	2.4	3.5	4.8	6.1	8.0	10.0
140	.4	.6	.8	1.4	2.3	3.3	4.5	5.9	7.4	9.2
150	.3	.5	.8	1.3	2.1	3.0	4.1	5.4	6.9	8.6
160	.3	.5	.7	1.3	2.0	2.8	3.9	5.1	6.5	8.0
180	.3	.4	.6	1.1	1.7	2.5	3.4	4.5	5.7	7.1
200	.3	.4	.6	1.0	1.6	2.3	3.1	4.0	5.1	6.4
250	.2	.3	.5	.8	1.3	1.8	2.5	3.2	4.1	5.1
300	.2	.3	.4	.7	1.1	1.5	2.1	2.7	3.4	4.2

3.16.7 Channel Dimension Relations Using Width/Depth Ratios

The ratio of the riffle cross-section width/depth of a study reach to the width/depth ratio of a reference reach provides a measure of vertical stability. A reference reach is one that is either unimpaired or least impaired. A least impaired reach is one that is as close to being unimpaired as can be found in the area of the study reach. The bankfull width/mean bankfull depth ratio is obtained from riffle cross-section data. Increases in width/depth ratio are often associated with channel aggradation due to accelerated streambank erosion, excess sediment deposition and channel widening, which commonly occur in "C" type alluvial channels (Table 6). Decreases in width/depth ratio can indicate channel incision, as for example when a "B" type channel degrades into a "G" type channel (Figure 13). The width/depth ratio is one of the six variables used and scored for purposes of the Rosgen channel stability index and the following table presents scoring criteria for evaluating stability using this variable (Rosgen, 2001).

Table 6. Scoring criteria for evaluating channel stability.

Width/depth ratio / Reference w/d ratio	Stability rating	Sediment supply category	Sediment supply score
< 1.4	Stable	Low	2
1.4 – 1.7	Moderately unstable	Moderate	5
> 1.7	Unstable	High	10

3.16.7.1 Field and Analytical Procedures

1. Conduct a cross-section survey at the study reach as indicated in Harrelson et al. (1994), identifying bankfull features.

2. Using a spreadsheet program, calculate bankfull cross-sectional area, and divide it by bankfull width to determine bankfull mean depth.

3. Calculate width/depth ratio by dividing bankfull width by bankfull mean depth.

4. For the reference reach width/depth ratio, use either a local reference site on a similar stream type, the Rosgen guidance provided in Figure 6, or use regional mean ratios by channel type (Moody et al., 2003).

5. Calculate the ratio of the study reach width/depth ratio to the reference reach width/depth ratio and apply stability ratings given in the table above for purposes of the Rosgen stability index.

3.16.7.2 Literature Cited

Harrelson, C.C., C.L. Rawlins, and J.P. Potyondy. 1994. Stream Channel Reference Sites: an illustrated guide to field technique. USDA, Forest Service, Rocky Mountain Forest and Range Experiment Station, General Technical Report RM-245. Fort Collins, CO.

Moody, T., M. Wirtanen, and S.N. Yard. 2003. Channel Stability Assessment for Biocriteria Sites in the Verde River Watershed. Prepared for the Arizona Department of Environmental Quality, Phoenix, AZ.

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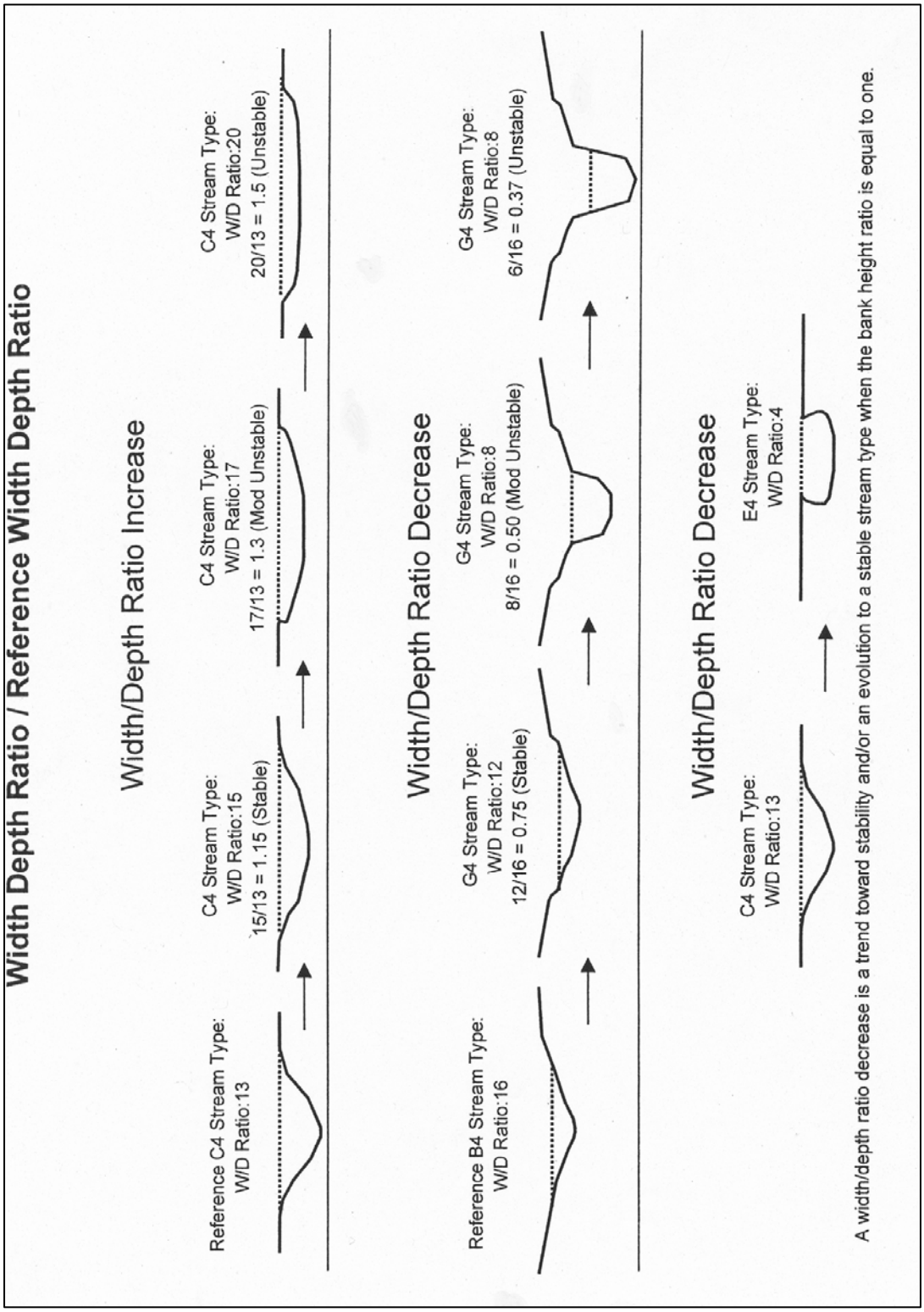


Figure 13. Examples of increases and decreases in width/depth ratios to reference condition that indicate impairment of stream stability (Rosgen, 2001).

3.16.8 Stream Channel Scour and Deposition Potential (Sediment Competence)

Adequate sediment transport is an essential component for maintenance of a stable channel. If either the channel depth or slope is less than needed to move the largest particle size of a bar sample, then aggradation or excessive deposition and a high width/depth ratio will occur. If the depth or slope exceeds that required to move the largest particle size, then degradation or excess scour may lead to channel incision. Sediment competence is determined by measuring the ability of a riffle cross-section to transport the largest particle in motion at bankfull stage. Though sediment competence is not one of the 6 variables in Rosgen's Stability Index, it is an important measure of vertical stability for alluvial stream channels, and can be used separately to evaluate channel stability. Implicit in this procedure is the assumption that bankfull discharge is responsible for transporting the greatest volume of sediment over time; that the bankfull discharge is the "effective discharge".

3.16.8.1 Sediment Competence Procedure

Field methods require collection of a pebble count from the riffle cross-section, sieve analysis of a bar sample collected from a bankfull bar feature, and a longitudinal profile. The bar sample must be collected from the lower third of a bankfull bar at an elevation halfway between bankfull stage and the channel thalweg as indicated in Figure 14 (Rosgen, 2001a). Critical dimensionless shear stress is calculated from the median particle sizes from both the riffle pebble count and the bar sample, using the following formula as described by Rosgen (2001b) and as shown in Figure 15.

$$\tau_{ci} = 0.0834(d_i/d_{50})^{-0.872}$$

where: τ_{ci} = critical dimensionless shear stress
d_i = median diameter of riffle bed material in mm.
d_{50} = median diameter of bar sample in mm.

The Shield's formula is used to calculate the bankfull mean depth needed to entrain the largest particle size in the bar sample, utilizing the largest particle size from the bar sample sieve test and the water surface slope of the study reach. The following formula is used to calculate the required bankfull mean depth (Rosgen, 2001b). Scores in the following table indicate when a channel is in stable, aggrading or degrading condition.

$$D_r = (\tau_{ci} * 1.65 * d_i)S_e$$

where: D_r = bankfull mean depth in feet
τ_{ci} = critical dimensionless shear stress
d_i = median diameter of riffle bed material
S_e = existing water surface slope of study reach

Slope evaluation	Depth evaluation	Stability category
$S_e/S_r = 1$	$D_e/D_r = 1$	Stable
$S_e/S_r < 1$	$D_e/D_r < 1$	Aggrading
$S_e/S_r > 1$	$D_e/D_r > 1$	Degrading

Where: S_e = existing water surface slope of study reach
S_r = bankfull water surface slope required
D_e = existing bankfull mean depth (from riffle cross-section)
D_r = bankfull mean depth required

3.16.8.2 Literature Cited

Rosgen, D. 2001a. Abridged River assessment and morphology (RAM) forms. Wildland Hydrology. Pagosa Springs, CO.

Rosgen, D.L. 2001b. A stream channel stability assessment methodology. Wildland Hydrology. Pagosa Springs, CO.

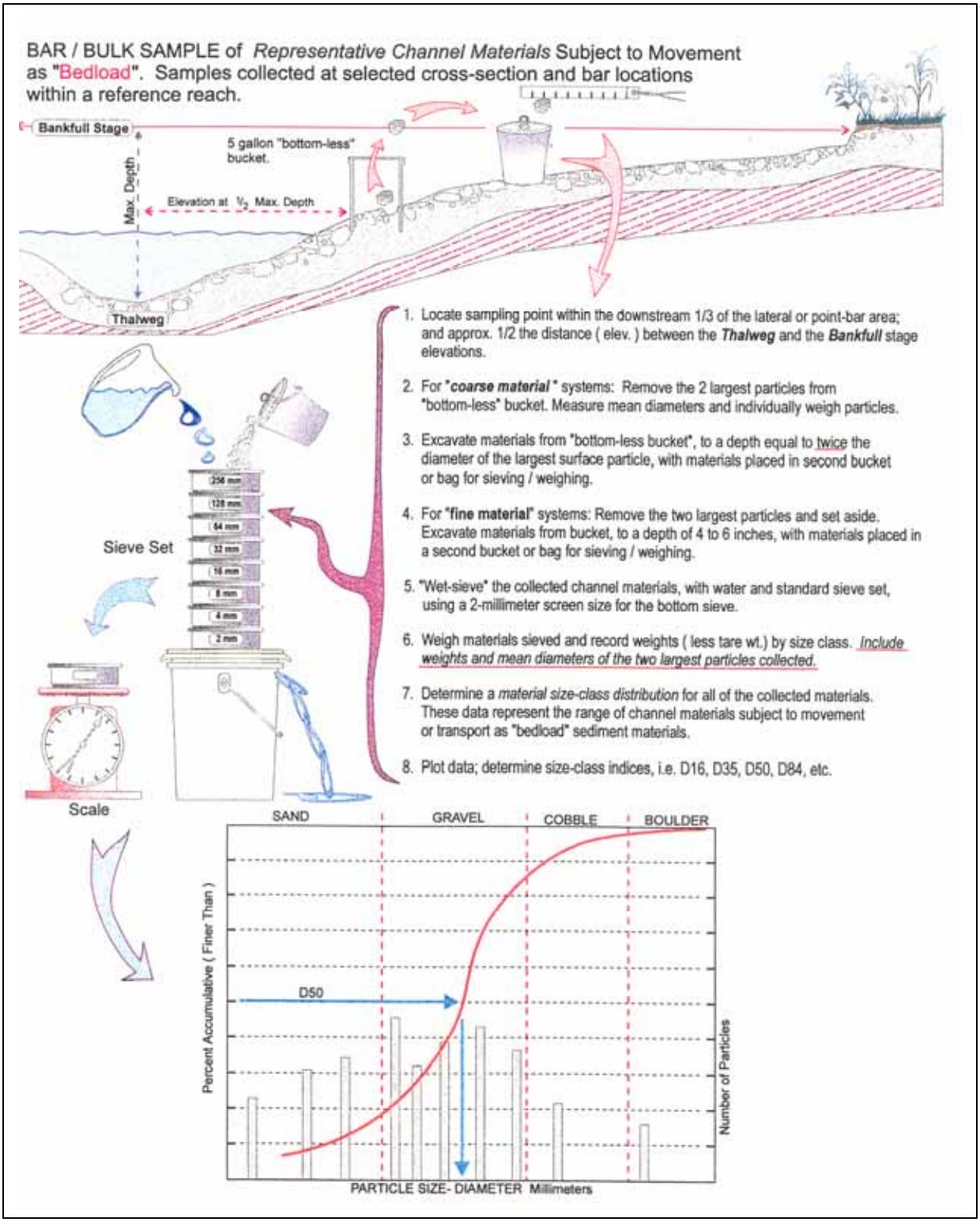


Figure 14. Bar sample procedure (Rosgen, 2001a).

Entrainment Calculation Form

Stream:	Reach:
Date:	Observers:

Critical Dimensionless Shear Stress:

$$\tau_{ci} = 0.0834(d_i/d_{50})^{-0.872}$$

Value	Variable	Definition
	d_i (mm)	D50 Bed Material (D50 from riffle pebble count)
	d_{50} (mm)	Bar Sample D50 or Sub-pavement D50
	τ_{ci}	**Critical Dimensionless Shear Stress**

Bankfull Mean Depth Required for Entrainment of Largest Particle in Bar Sample:

$$d_r = (\tau_{ci}*1.65*D_i)/S_e$$ 1.65 = submerged specific weight of sediment

Value	Variable	Definition
	τ_{ci}	Critical Dimensionless Shear Stress
	D_i (feet)	Largest particle from bar sample
	S_e (ft/ft)	Existing Bankfull Water Surface Slope
	d_r (ft)	**Bankfull Mean Depth Required**
	d_e (ft)	**Existing Bankfull Mean Depth (from riffle cross section)**

Circle: Stable (d_e/d_r =1) Aggrading (d_e/d_r <1) Degrading (d_e/d_r >1)

Bankfull Water Surface Slope Required for Entrainment of Largest Particle in Bar Sample:

$$S_r = (\tau_{ci}*1.65*D_i)/d_e$$ 1.65 = submerged specific weight of sediment

Value	Variable	Definition
	τ_{ci}	Critical Dimensionless Shear Stress
	D_i (feet)	Largest particle from bar sample
	d_e (ft)	Existing Bankfull Mean Depth (from riffle cross section)
	S_r (ft/ft)	Bankfull Water Surface Slope Required

Circle: Stable (S_e/S_r =1) Aggrading (S_e/S_r <1) Degrading (S_e/S_r >1)

Sediment Transport Validation

	Largest Particle in Bar Sample D_i (mm)
	Bankfull Shear Stress $\tau_c=\gamma RS$ (lb/ft^2)
	Moveable particle size (mm) at bankfull shear stress (predicted by the Shields Diagram: Blue field book:p238, Red field book: p190)
	Predicted shear stress required to initiate movement of D_i (mm) (see Shields Diagram: Blue field book:p238, Red field book: p190)

Figure 15. Sediment transport calculation form provided by Rosgen (2001c).

3.16.9 Pfankuch Channel Stability Evaluation

The Pfankuch Channel Stability Evaluation (Pfankuch, 1975) is an assessment of stream condition. It is used to qualitatively describe the potential for sediment material detachment and changes in sediment supply due to changes in streamflow and/or changes in watershed condition. The evaluation is segregated into three categories: upper banks, lower banks, and channel bottom. Each category has three to four ratings. The sum of the category ratings is transformed into an adjective rating of stream stability (e.g., Excellent, Good, Fair, and Poor).

Pfankuch's original evaluation was designed for use in western U.S.A. mountain stream channels, but did not provide for the varieties of stream type. Rosgen (1996) incorporated the evaluation into his river classification system, but dropped the "excellent" rating. After extensive use of Rosgen's modified version, ADEQ determined that some portions of the evaluation did not adequately evaluate arid land stream channels. Therefore, ADEQ staff made the appropriate modifications to the rating categories that evaluate Arizona streams (Figure 16). Under the "Lower Banks" category, two changes were made; "Bank Rock Content" was deleted and replaced by "Surface Protection," and "Deposition" was deleted. Pfankuch's entire category of six ratings for channel "Bottom" was removed and replaced by three ratings that more accurately describe arid land stream channel bottoms. Therefore, a correction factor of 1.226 is applied to convert the ADEQ scores upward for use with the original Pfankuch scoring criteria for each stream type.

3.16.9.1 Completing the Channel Stability Evaluation Form

Upon arriving at streamside and after selecting the study reach, monitoring staff walk the reach and carefully observe channel conditions. The length of the study reach is dependant upon the objectives of the study being conducted. A minimum of two trained observers is required for completing the evaluation form. The rating categories on the form do not require additional documentation since each parameter is self-explanatory. Once the ratings have been assigned and the scores tallied, the stability rating of Good, Fair or Poor is selected by stream type from the conversion table located at the end of Figure 16. This stability rating is then converted to a Sediment supply score for purposes of the Rosgen stability index, as indicated in the following table.

Table 7. Rosgen Stability Index.

Pfankuch condition class	Sediment supply category	Sediment supply score
Good	Low	5
Fair	Moderate	10
Poor	High	15

3.16.9.2 Literature Cited

Rosgen, David. 1996. Applied River Morphology. Wildland Land Hydrology, Pagosa Springs, CO.

Pfankuch, D.J. 1975. Stream reach inventory and channel stability evaluation. USDA Forest Service, R1-75-002. Government Printing Office #696-260/200, Washington, D.C.: 26 pp.

Figure 16. Arizona Modified Pfankuch Channel Stability Evaluation Field Form.

Site ID:_____ Site Description:_____

Date:_____ Observers:_____

MODIFIED PFANKUCH CHANNEL STABILITY EVALUATION (LEVEL III)

The Pfankuch evaluation is segregated into three categories; upper banks, lower banks, and channel bottom. Each category has three or four ratings to be scored. Rating scores are given as a range. The range has been provided as an option if there is some uncertainty about a rating item. If you choose to ignore the range then use the lowest score provided for the chosen item. The low score is the optimum condition.

	Rating
A. **"Upper Banks"**, or first terrace, is the flood plain area and is above bankfull. This landform comes into play only during floods. This category is designed to aid in rating the relative resistance to detachment and transport of particles (large and small, organic and inorganic) by floods.	
1. "*Landform Slope*" is the angle of slope of the flood plain. This can be estimated or measured with an Abney level (or similar device). Always choose the worst condition for the rating. If you have one flood plain at 30% and the other at >60%, rate the steeper slope because that is the area where erosion will be occurring at flood.	
a. Excellent: Side slopes to the channel are generally less than 30% on both banks.	2
b. Good: Side slopes up to 40% on one or occasionally both banks.	4
c. Fair: Side slopes to 60% common on one or both banks.	6
d. Poor: Steep slopes, over 60%, provide larger volumes of soil for downstream sedimentation from lateral bank cutting.	8
2. "*Mass Wasting*" involves existing or potential detachment of relatively large pieces of earth. Mass movement of banks by slumping or sliding introduces large volumes of soil and debris into the channel. This condition is common at meanders or on incised channels where high banks exist at great angles, especially over 60%.	
a. Excellent: There is no evidence of mass wasting that has or could reach the stream channel in recent times.	3
b. Good: There is evidence of infrequent and / or very small slumps. Those that exist may occasionally be "raw" but predominantly the areas are revegetated and relatively stable.	6
c. Fair: Frequency and / or magnitude of the mass wasting situation increases to the point where normal high water (bankfull or a little less) aggravates the problem of channel changes and subsequent undercutting or unstable areas with increased sedimentation.	9
d. Poor: Mass wasting is not difficult to detect.	12
3. "*Debris Jam*" includes those floatable objects that have been deposited on stream banks, in the flood plain, by man or by natural processes. It usually consists of tree trunks, limbs, twigs, and leaves. It forms obstructions, flow deflectors, and sediment traps. This inventory item assesses the potential for increasing these impediments to the natural direction and force of flow where they now lay. The Pfankuch evaluation considers debris jams to be a negative influence on the stream channel except when it is protecting the flood plain banks.	
a. Excellent: Some small debris may be present on the flood plain banks, but is essentially absent.	2
b. Good: Some debris present but it is small enough to be floated away in time. Only small jams could be formed with this material alone.	4
c. Fair: There is a noticeable accumulation of all sizes and the stream is large enough to float it away.	6
d. Poor: Moderate to heavy accumulations are present due to fires, insect damage to trees, disease mortality, windthrow, and logging slash. High flows will float some debris away and the remainder will cause channel changes.	8

4. "*Vegetative Bank Protection*" concerns the vegetative component in the flood plain. Factors to consider for this rating are the density of plant stems, varieties of vegetation, plant vigor, and recruitment.	
a. Excellent: Trees, shrubs, grass and forbs combined cover is more than 90% of the ground. Openings in the ground covers are small and evenly dispersed. A variety of age classes and species are represented. Growth is vigorous and reproduction of species in both the under- and over-story is proceeding at a rate to insure continued ground cover conditions. A deep dense root mat is inferred.	3
b. Good: Plants cover 70 to 90 percent of the ground. Shrub species may be more prevalent than trees. Openings in the tree canopy are large. While the growth vigor is generally good for all species, recruitment of new individuals may be sparse or lacking entirely. A deep root mat is not continuous and more serious erosive incursions are possible in the openings.	6
c. Fair: Plant cover ranges from 50 to 70 percent. Lack of vigor is evident in some individuals and / or species. Seedling reproduction is nil. Most of the flood plain does not have a deep root mat.	9
d. Poor: Less than 50 percent of the ground is covered. Trees are essentially absent. Shrubs largely exist in scattered clumps. Growth and reproduction vigor is generally poor. Root mats discontinuous and shallow.	12
UPPER BANK TOTAL	
B. "**Lower Banks**" is the area between bankfull and base flow. Aquatic, semi-aquatic, and terrestrial plants may grow here.	
1. "Channel Capacity" is the inventory of the channel width, depth, gradient and roughness determined by the volume of water carried downstream. This is essentially a measure of channel stability and departure from the ideal condition.	
a. Excellent: The ideal condition. The stream is balance with its watershed, neither aggrading nor degrading. Width / depth ratios are in the range of the desired stream type for that ecosystem.	1
b. Good: Adequate cross sectional area contains most peak flows. Not the ideal condition. Possibly a small amount of aggrading or entrenchment occurring, but nothing serious.	2
c. Fair: Channel barely contains the peak runoff in return interval or less and may be widening and becoming shallower (stream is aggrading). Or, channel is degrading and is noticeably incised.	3
d. Poor: Channel may be so aggraded that its capacity is generally inadequate to contain bankfull flows and overbank floods common. Or, the channel is deeply entrenched and bank erosion is occurring.	4
2. "Surface Protection" refers to the composition of bank materials which prevent erosion due to freeze/thaw activity and the near bank stress of bankfull or high flows. Look at root density and cobble armouring for this evaluation.	
a. Excellent: Greater than 75% of bank surface area is protected by dense root mats or cobble armouring.	2
b. Good: Between 50-75% of bank surface area is protected by dense root mats or cobble armouring.	4
c. Fair: Between 25-50% of bank surface area is protected by dense root mats or cobble armouring.	6
d. Poor: Less than 25% of bank surface area is protected by dense root mats or cobble armouring.	8
3. "*Obstructions to Flow*" is an inventory of objects within the stream channel, like rocks, embedded logs, bridge pilings, etc., change of flow and sometimes the velocity. Obstructions may produce adverse stability effects when they increase the velocity and deflect the flow into unstable and unprotected banks and across unstable bottom materials. They also may produce favorable impacts when velocity is decreased by turbulence and pools are formed.	
a. Excellent: Logs, rocks, and other obstructions to flow are firmly embedded and produce a pattern of flow which does not erode the banks and bottom or cause sediment buildup. Pool / riffle relationship is stable.	2
b. Good: Obstructions to flow and sediment traps are present, causing cross currents which create some minor bank and bottom erosion. Some of the obstructions are newer, not firmly embedded and move to new locations during high flows. Some sediment is trapped in pools decreasing their capacity.	4
c. Fair: Moderately frequent and quite often unstable obstructions, cause noticeable seasonal erosion of the channel. Considerable sediment accumulations behind obstructions.	6
d. Poor: Obstructions and traps are common, are often unstable to movement and cause a continual shift of sediments at all seasons. Since traps are filled as soon as formed, the channel migrates and widens.	8

	LB	RB
4. *"Cutting"* or downcutting of the channel is preempted first by scouring, uprooting, and loss of vegetation. In channels devoid naturally of vegetation, the first stages would be an increase in the steepness of the channel banks. If plant roots bind the surface horizon of the adjacent upper bank into a cohesive mass, undercutting will follow. Eventually the weight of the overhang will slump into the channel. Unconsolidated banks with or without vegetation will be nibbled away and never develop an overhang.		
Notice that you must evaluate both the left bank and the right bank of the rated reach.	LB	RB
a. Excellent: Very little or no cutting is evident. Raw eroding banks are infrequent, short and predominately less than 6" high.	2	2
b. Good: Some intermittent cutting along channel outcurves and at prominent constrictions. Eroded areas are equivalent in length to one channel width or less and the vertical cuts are predominately less than 12".	3	3
c. Fair: Significant bank cutting occurs frequently in the reach. Raw vertical banks 12" to 24" high are prevalent as are root mat overhangs and sloughing.	6	6
d. Poor: Nearly continuous bank cutting. Some reaches have vertical cut faces over 2 feet high. Undercutting, sod-root overhangs and vertical side failures may also be frequent in the rated reach.	8	8
LOWER BANK TOTAL		

C. Channel "**Bottom**" condition is the evaluation of sediment deposition within the bankfull channel.	
1. *Bottom Deposits*" is the evaluation of how well the channel is moving its sediment downstream. Ideally, there should be a defined thalweg, riffles, and pools. However, you have to be the judge whether or not this is the naturally occurring condition for that ecosystem. Some low gradient desert streams may be the exception.	
a. Excellent: A stream channel that is in balance with its watershed. Less than 20% of the stream bed is affected by sediment deposition.	6
b. Good: Some deposition occurring in the pools. For the whole reach, 20-50% of the bottom is experiencing some deposition.	12
c. Fair: Deposition is quite noticeable. 50-80% of the channel is affected. Sediment deposits at obstructions, constrictions, and bends; moderate deposition of pools and riffles, and stream braiding may be occurring.	18
d. Poor: Very noticeable deposition occurring over more than 80% of the reach. Pools and riffles mostly or entirely absent .	24
2. "*Bar Development and Deposition*" plays an vital role in most stream ecosystems. Exceptions are "A" type streams and geologically constrained streams. The appearance of sand and gravel bars where they did not previously exist may be one of the first signs of upstream erosion.	
a. Excellent: Little or no enlargement of point bars; sand bars are stable and completely vegetated.	4
b. Good: Some new increase in bar formation, mostly from gravel, sand or fine sediment; sand bars stable but not completely vegetated.	8
c. Fair: Moderate deposition on new gravel, sand or fine sediment on old and new bars; sand bars unstable with sparse vegetation.	12
d. Poor: Heavy deposits of fine material; increased bar development; sand bars unstable with no vegetation; transverse bars may be present.	16
3. "*Embeddedness*" is an inventory of the degree of sedimentation in riffles and pools.	
a. Excellent: Gravel and cobble particles are 0 - 25% surrounded by fine sediment.	2
b. Good: Gravel and cobble particles are 25 - 50% surrounded by fine sediment.	4
c. Fair: Gravel and cobble particles are 50 - 75% surrounded by fine sediment.	6
d. Poor: Gravel and cobble particles are more than 75% surrounded by fine sediment, or there is an absence of riffles and pools.	8
CHANNEL BOTTOM TOTAL	

Sum of "Upper Banks", "Lower Banks", and "Channel Bottom"	
Final Pfankuch Score (Sum of all categories X 1.226)	
Rosgen Stream Type	
Pfankuch Rating category (Good, Fair, Poor)	

Sediment Supply Condition	**Stream Bed Stability**	**Width/Depth Ratio**
Extreme_____	Aggrading_____	Normal_____
Very High_____	Degrading_____	High_____
High_____	Stable_____	Very High_____
Low_____	Total Score for Reach - UB_____ + LB_____ + BOT_____ = _____	

Use Rosgen Sediment Supply and stability summary worksheet to determine sediment supply and stream bed stability categories above. Use average W/D ratio for that basin or region to assess W/D ratio condition.

Remarks

CONVERSION OF STABILITY RATING TO REACH CONDITION BY STREAM TYPE (after Rosgen, 1996)

Stream Type	A1	A2	A3	A4	A5	A6	B1	B2	B3	B4	B5	B6
GOOD	38-43	38-43	54-90	60-95	60-95	50-80	38-45	38-45	40-60	40-64	48-68	40-60
FAIR	44-47	44-47	91-129	96-132	96-132	81-110	46-58	46-58	61-78	65-84	69-88	61-78
POOR	48+	48+	130+	133+	133+	111+	59+	59+	79+	85+	89+	79+
Stream Type	**C1**	**C2**	**C3**	**C4**	**C5**	**C6**	**D3**	**D4**	**D5**	**D6**		
GOOD	38-35	38-35	60-85	70-90	70-90	60-85	85-107	85-107	85-107	67-98		
FAIR	51-61	51-61	86-105	91-110	91-110	86-105	108-132	108-132	108-132	99-125		
POOR	62+	62+	106+	111+	111+	106+	133+	133+	133+	126+		
Stream Type	**DA3**	**DA4**	**DA5**	**DA6**	**E3**	**E4**	**E5**	**E6**				
GOOD	40-63	40-63	40-63	40-63	40-63	50-75	50-75	40-63				
FAIR	64-86	64-86	64-86	64-86	64-86	76-96	76-96	64-86				
POOR	87+	87+	87+	87+	87+	97+	97+	87+				
Stream Type	**F1**	**F2**	**F3**	**F4**	**F5**	**F6**	**G1**	**G2**	**G3**	**G4**	**G5**	**G6**
GOOD	60-85	60-85	85-110	85-110	90-115	80-95	40-60	40-60	85-107	85-107	90-112	85-107
FAIR	86-105	86-105	111-125	111-125	116-130	96-110	61-78	61-78	108-120	108-120	113-125	108-120
POOR	106+	106+	126+	126+	131+	111+	79+	79+	121+	121+	126+	121+

3.16.10 Stream Type Classification

Rosgen (1994) developed a stream type classification system which allows for comparisons of reference and study reaches and provides a basic understanding of channel processes. The stream type must be identified prior to collecting other measurements for the stream stability assessment, such as depositional pattern (Section 3.16.1), Pfankuch channel stability rating (Section 3.16.9), and stream type evolutionary scenario (Section 3.16.11). This classification system is based on channel measurements collected from a riffle cross-section survey, reach pebble count, longitudinal profile, and the following five elements:

- Entrenchment ratio (floodprone width/bankfull width)
- Bankfull width/bankfull mean depth ratio
- Sinuosity
- Reach slope
- Channel material, median particle diameter (D50).

The classification system scheme sorts fluvial streams into broad stream types A through G, representing the following categories:

A – Headwater
B – Intermediate
C – Meandering alluvial
E – Meandering alluvial with high sinuosity and low w/d ratio
D – Braided
F – Entrenched
G – Gully

These broad categories are further refined by the addition of slope ranges and median particle size to produce 41 categories of stream types which are described and photographed in detail in Rosgen (1996). After all field measurements are collected and ratios calculated, the key in Figure 17 is used to determine stream type. Figure 18 provides cross-section diagrams for each stream type for an easy visual representation.

Rosgen delineated eleven valley types and identified their associations with stream types (Table 8); however, Valley Types V, VII, X, and XI are unlikely to be found in Arizona and are not listed. For a discussion of valley types, see Rosgen (1996), Chapter 4.

Table 8. Valley types and their associated stream types.

Valley Type	Environment	Geomorphic Characteristics	Stream Type	
			Dominant	Other
I	Rugged mountains	Narrow valleys	A,G	
II	Less rugged mountains	Narrow valleys	B	G
III	Broad valleys in mountains	Incised upland rivers with alluvial fans at confluence	A,B,G,D	
IV	Gorges & canyons		F	C
VI	Faultline valleys		B	C,F
VIII	Developed floodplains	Alluvial terraces and floodplains	C,E	D,F,G
IX	Dune plains		C,D	

3.16.10.1 Literature Cited

Rosgen, D.L. 1994. A classification of natural rivers. Catena 22:169-199.

Rosgen, D.L. 1996. Applied River Morphology. Wildland Hydrology. Pagosa Springs, CO.

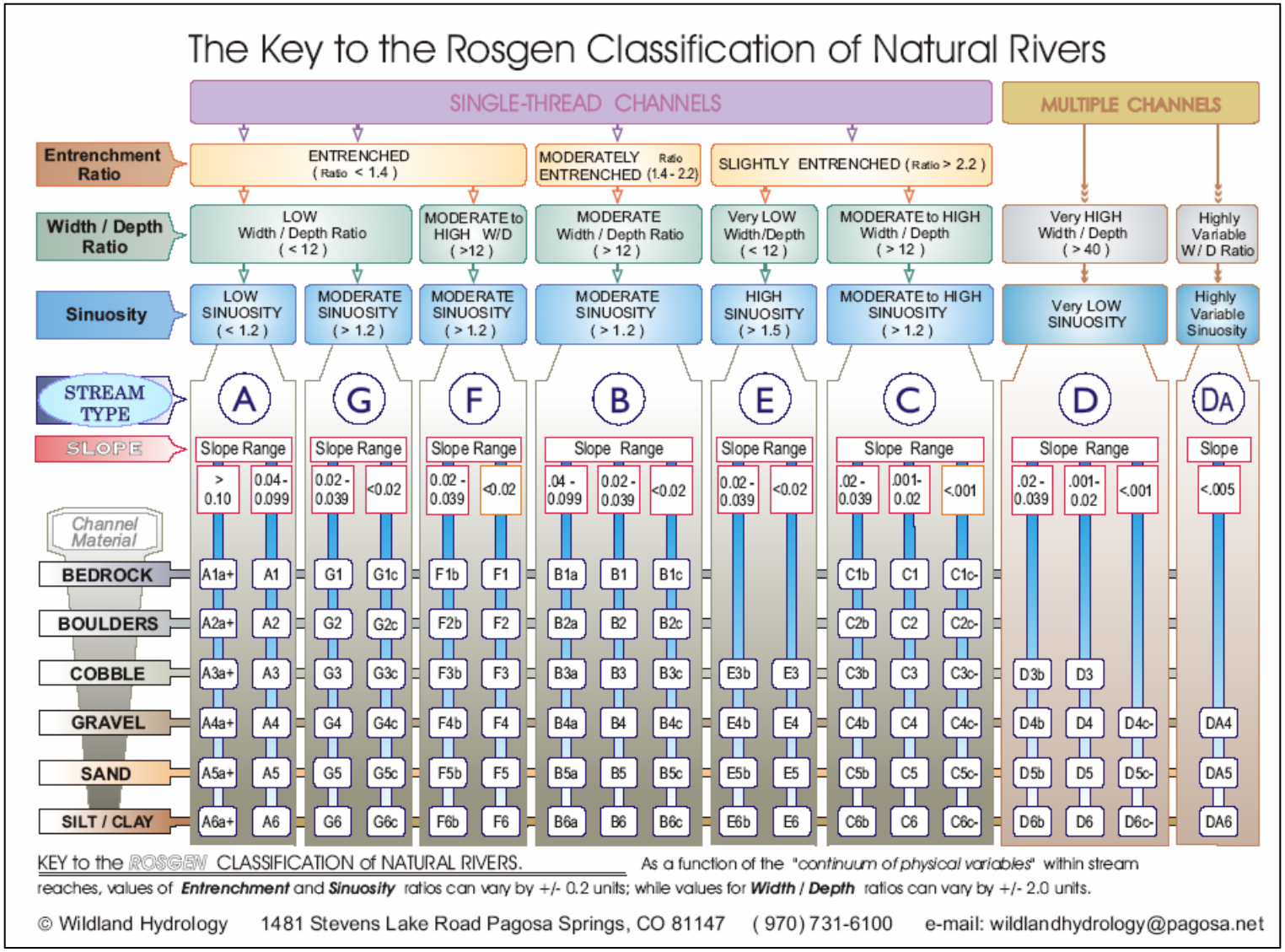


Figure 17. A Key for Rosgen Stream Type Classification (Rosgen, 1996).

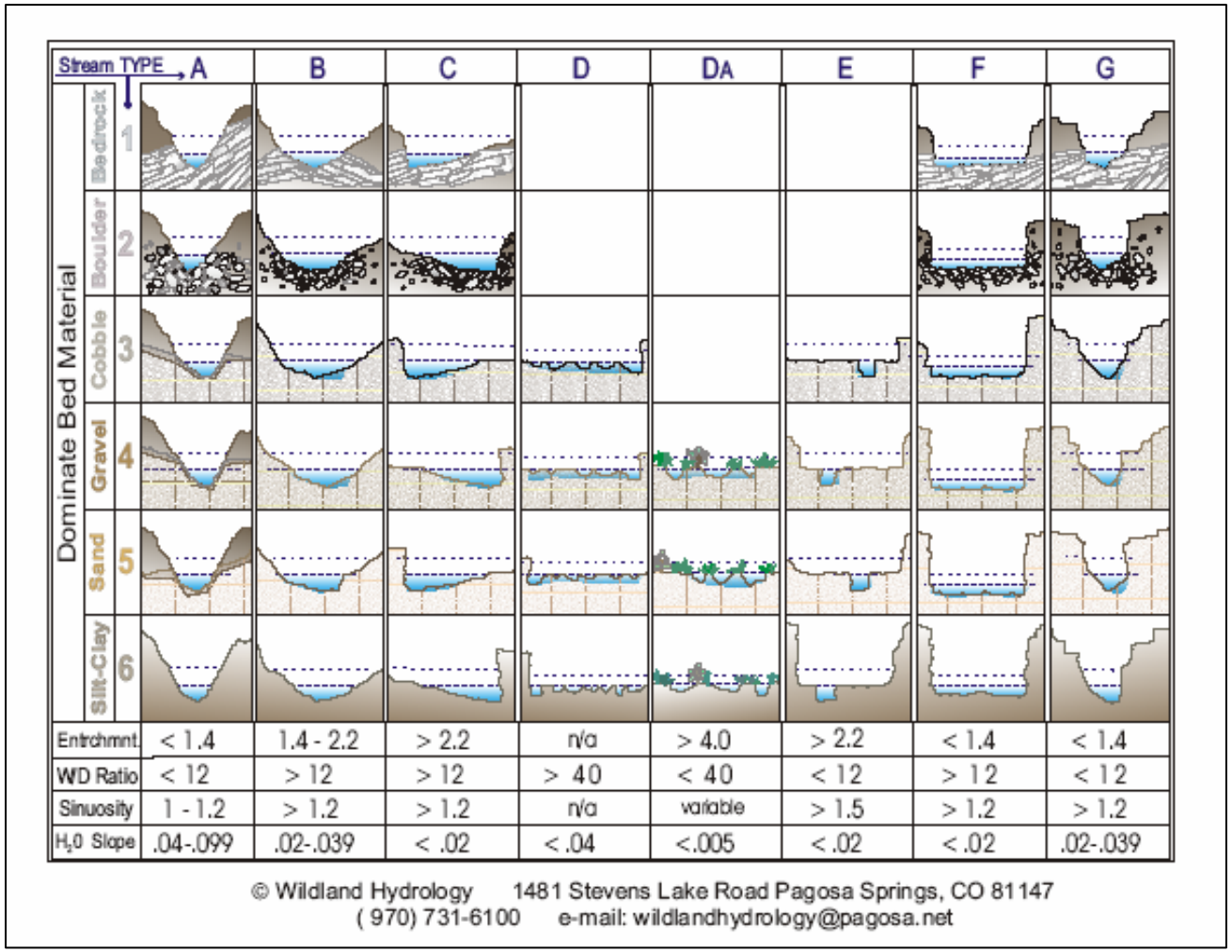


Figure 18. Visual representations of each major stream type in the Rosgen Classification system (Rosgen, 1996).

3.16.11 Stream Type Evolutionary Scenarios

Natural channels are dynamic by nature, responding to changes in flows and sediment loads by vertical and lateral adjustments and changes in bed material. Channel type evolution occurs as a result of vertical or lateral instability created as a stream system exceeds a geomorphic threshold. The formation of gullies, an unstable channel type, followed by a succession of channel types toward a more stable form is the best known of the channel evolution sequences (Figure 19). Various stream type evolutionary scenarios, which commonly occur in alluvial channels, have been described (Rosgen, 1996) and are used in the Rosgen Stability Index.

3.16.11.1 Field Protocols

Field protocols consist of performing a riffle cross-section survey and identifying other channel types that may occur within the two meander study reach. The existing stream type(s) occurring in the study reach are determined from the riffle cross-section data (entrenchment ratio, width/depth ratio), sinuosity, slope and median particle size of the bed material using Rosgen's stream classification system (Rosgen, 1994). The field observer must select not only the existing stream type, but also the location in one of the sequences of evolution. The field observer should also determine the potential stable stream type in addition to the existing stream type. This requires additional information about the valley type. For instance, which stream types are naturally associated with that valley type; what other "background" or "reference" stream channel types are common in the local area? The table below indicates channel type sequences indicative of low, moderate and high sediment supply or channel stability categories, and the associated Rosgen Stability Index score (Rosgen, 2001).

Table 9. Evolutionary sequences that estimate sediment supply by stream type.

Evolutionary sequence	Stability rating	Sediment supply category	Sediment supply score
(C → E), (Fb → B), (G → B), (F →Bc)	Stable	Low	5
(F → C), (B → G)	Moderately Unstable	Moderate	10
(E or C → G), (E or C → D) (G → F), (D → G)	Unstable	High	15

3.16.11.2 Literature Cited

Rosgen, D.L. 1994. A classification of natural rivers. Catena 22:169-199.

Rosgen, D.L. 1996. Applied River Morphology. Wildland Hydrology. Pagosa Springs, CO.

Rosgen, D.L. 2001. Stream stability and sediment supply assessment protocols. Level III Short Course materials. Wildland Hydrology. Pagosa Springs, CO.

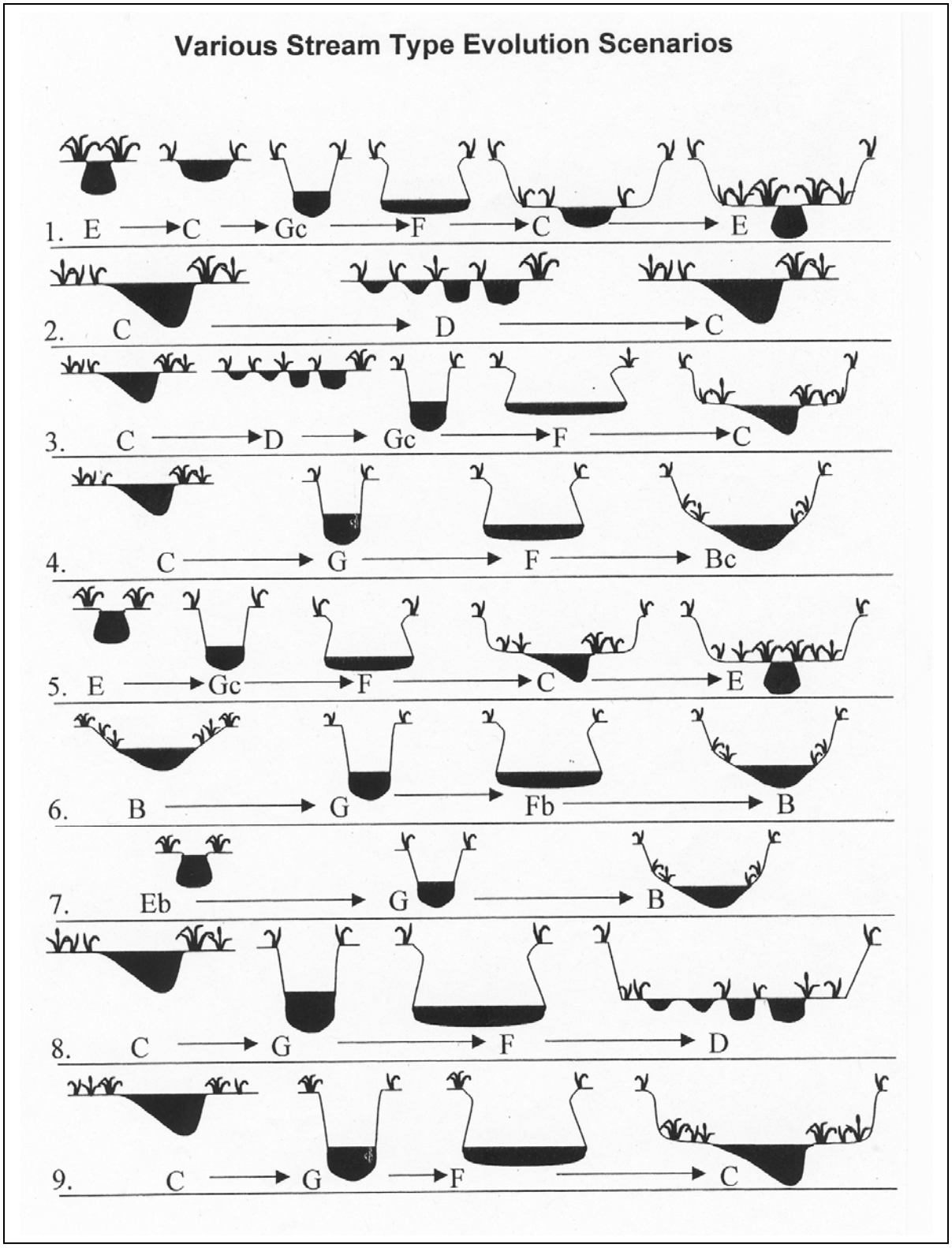


Figure 19. Stream type evolution scenarios from Rosgen (2001).

3.16.12 Rosgen Stability Assessment

Variables that describe vertical and lateral stability, excess sediment conditions, a change in stream type, and overall channel, bank and floodplain condition are included in the Rosgen Stability Index for a holistic geomorphic stability assessment (Figure 20). The bank height ratio measures vertical stability and the dominant BEHI/NBS category; width/depth ratio measures lateral stability. The depositional pattern is an indicator of excess sediment conditions, while the Pfankuch stability rating is an assessment of the overall channel, bank and floodplain conditions. Lastly, the evolutionary stage documents instability associated with channel type changes. These six variables are transformed into qualitative rating and then quantitative ratings of sediment supply, using guidance from Rosgen (2001). The sediment supply ratings are given in the following tables.

Sediment Supply Summary						
Sediment Supply	Dominant BEHI/NBS	(Width/depth ratio) / (Reference width/ depth ratio)	Bank Height Ratio	Pfankuch Channel Stability Rating	Evolutionary Stage	Depositional Pattern
Low	L/L, L/M	< 1.4	1.0 – 1.1	Good	(C → E), (Fb → B), (G → B), (F → Bc)	1,4
Moderate	M/M, M/L, L/H, L/Ex	1.4 – 1.7	1.11 – 1.3	Fair	(F → C), (B → G)	3
High	M/H, H/M, H/H, M/Ex, H/Ex, Ex/Ex	> 1.7	> 1.3	Poor	(E or C → G), (E or C → D) (G → F), (D → G)	2,5,6,7,8

Variables	Stability Categories		
	Low	**Moderate**	**High**
Dominant BEHI/NBS	5	10	20
(Width/depth ratio) / (Reference width/ depth ratio)	2	5	10
Bank Height Ratio	2	5	10
Pfankuch Channel Stability Rating	5	10	15
Evolutionary Stage	5	10	15
Depositional Pattern	2	5	10
Total Stability Index Score	21	45	80

Conversion of stability index score to overall channel stability rating

Channel Stability Rating	Stability Index Score
Low	21 – 33
Moderate	34 – 45
High	46 - 80

3.16.12.1 Literature Cited

Rosgen, D.L. 2001. Stream stability and sediment supply assessment protocols. Level III Short Course materials. Wildland Hydrology. Pagosa Springs, CO.

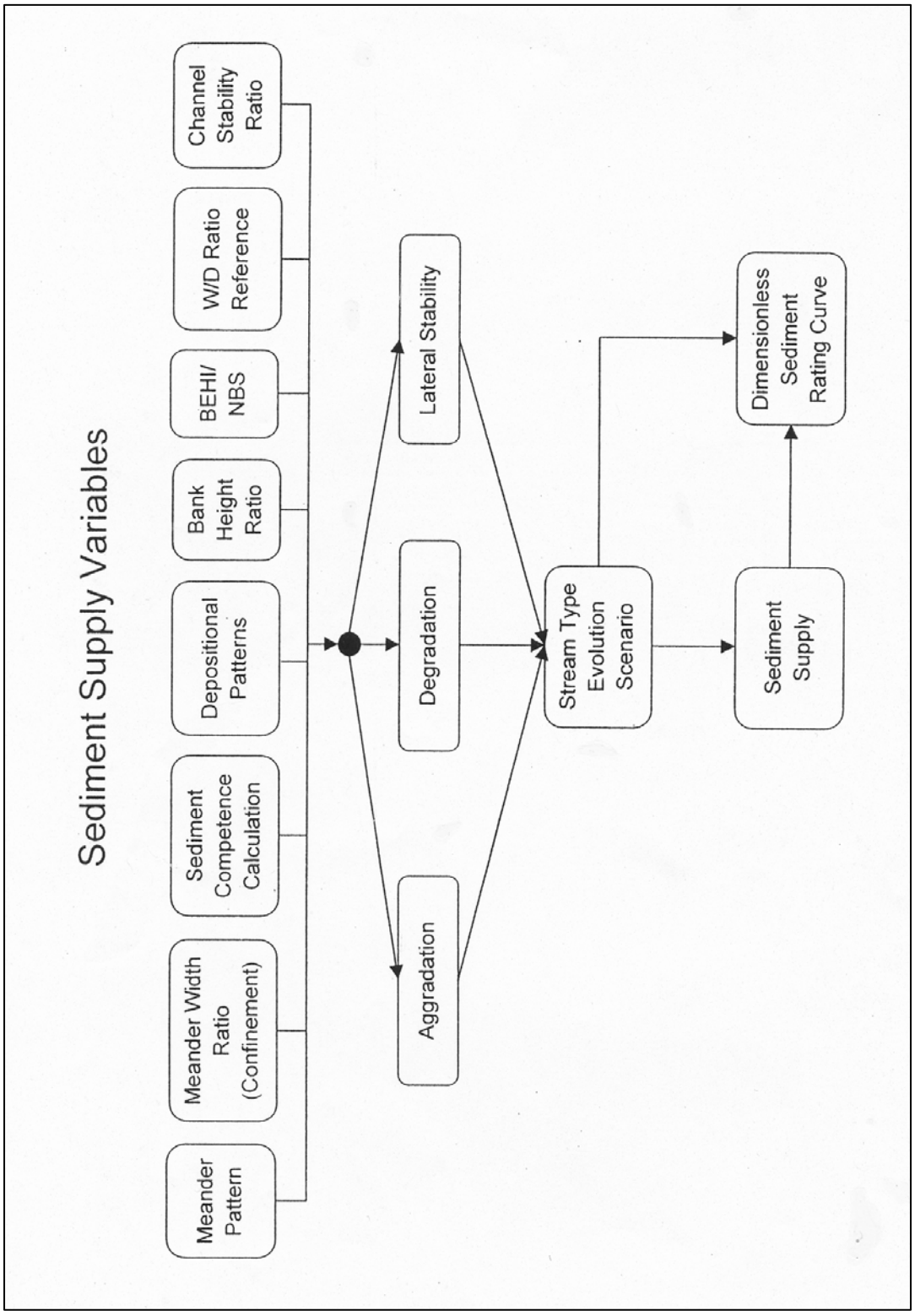


Figure 20. Rosgen Stability Assessment Variables (Rosgen, 2001).

3.16.13 Linear Habitat Complexity Index

The Linear Habitat Complexity Index (LHCI) identifies the degree of morphological complexity within a stream channel. The Index is based on run, riffle, and pool lengths. For the Index, glides are considered to be runs. It was inferred that reaches of stream channel with extensive runs were less physically heterogeneous (i.e. lacking complexity and habitat diversity for aquatic organisms) than complex channels having short runs frequently intermixed with pools and riffles. A heterogeneous channel is considered to be indicative of stability.

3.16.13.1 Linear Habitat Complexity Index Calculation Procedure

The Index is calculated from feature identification and feature lengths on a longitudinal profile. A run reference norm is determined from a reference reach using the LHCI calculation. When applied to a study reach, the index identifies runs exceeding the reference run norm. The Index is structured such that as LHCI values increase, habitat complexity trends toward homogeneity; thus, increasing index values indicate lower physical integrity than the reference reach.

The reference reach mean run length and standard deviation are calculated from the profile data. For the index, a reference run length norm is determined to be the mean run length plus one standard deviation.

The Index is a moving average of ten contiguous features. After the Index is calculated for the first ten features, the first feature is dropped and the eleventh feature added for the next reach calculation. This process continues to the end of the profile.

Linear Habitat Complexity Index = 1 – ((total length of ten features – the sum of the portions of runs exceeding reference run norm) / total length of ten features).

Plots of the results of the LHCI are composed for the reference and study reaches. Significant departures of the study reach from the reference reach are determined. Significant departures indicate that the study reach has less habitat complexity than the reference reach.

3.16.13.2 Examples

Two LHCI plots are provided as examples of a reference reach and a sediment impaired reach. The examples are taken from Lawson and Huth (2003). The two figures demonstrate the differences between a stable channel having a good mix of riffles, runs, and pools (Figure 21) and a less stable channel that is dominated by runs (Figure 22). Index scores greater than 0.2 likely portray a channel that is sediment impaired; however, this analysis method is currently being developed and plots from other streams will be necessary to confirm the 0.2 threshold value.

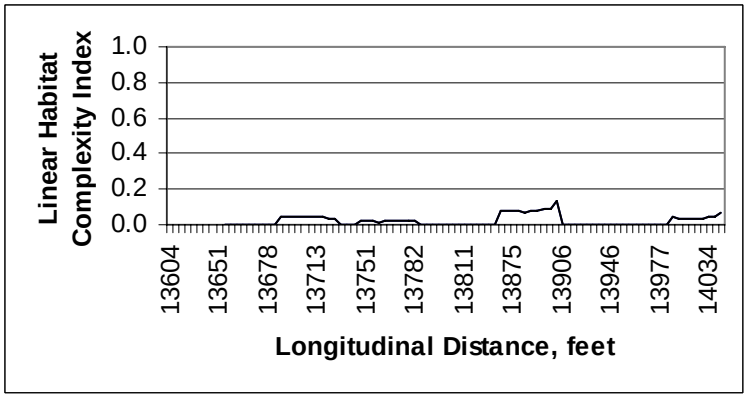


Figure 21. Plot of LHCI for a reference reach.

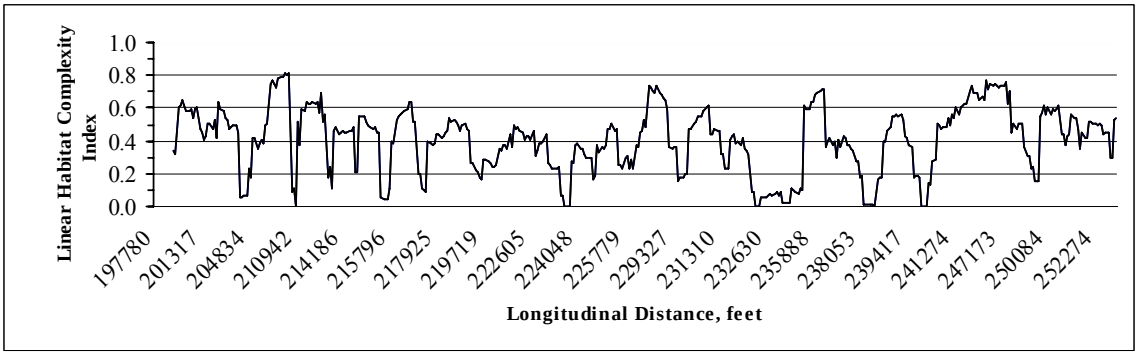


Figure 22. Plot of LHCI for a sediment impaired reach.

3.16.13.3 Literature Cited

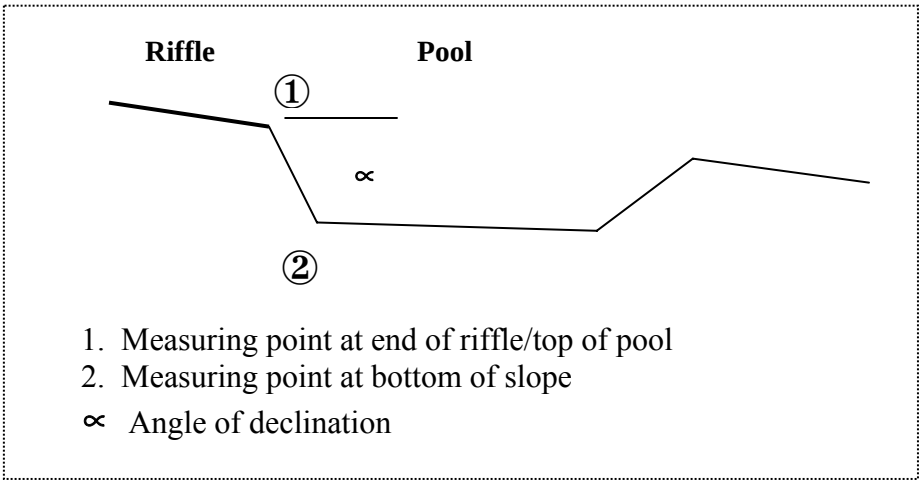
Lawson, L. and H. Huth, 2003. Lower Cienega Creek Restoration Evaluation Project: An Investigation into Developing Quantitative Methods for Assessing Stream Channel Physical Condition. ADEQ Report #EQR0303, Arizona Department of Environmental Quality, Phoenix, AZ.

3.16.14 Pool Facet Slope Analysis

The intent of this metric analysis is to quantitatively measure the degree of sedimentation in stream channels, especially in "C" and "E" type channels. Pools fill with sediment under conditions of sediment deposition. When deposition is severe, the number of pools become fewer and the pools that are present loose depth as they fill with sediment. In the longitudinal profile, pools follow riffles and runs. The pool facet slope is the angle of declination from the end of the upstream feature (riffle or run) to the lowest point in the pool (Figure23). This angle will trend towards the horizontal as the pool fills with sediment; therefore, the pool facet slope is an indicator of sediment conditions in a stream channel that is expected to contain pools.

When measuring the facet slope, care must be taken to measure, as accurately as possible, both ends of the facet. A procedure for measuring a more accurate longitudinal profile is described in the longitudinal profile procedure documentation. Examples of this analysis are detailed in Lawson and Huth (2003).

Figure 23. Pool facet slope points of measurement.



3.16.14.1 Pool Facet Slope Analysis Procedure

The facet angles are measured from longitudinal profiles where feature types and feature depths are known. This calculation is the rise over run and expressed as the mathematical ratio or a degree angle of declination (arctan ∝ = rise/run).

A profile of sufficient length for the analysis is determined by localized conditions. The length of the profile could be as much as 2,000 feet or several meanders. The profile length should give enough facet angles for statistical analysis when a study reach is compared to a reference reach. It is suggested that both the study and reference reaches contain a minimum of twenty-five pools. It is unknown, at this time, how different the angles of the two reaches must be to be significantly different. The calculations of mean, minimum, maximum, and standard deviation of reach pools are recommended for comparisons.

3.16.14.2 Literature Cited

Lawson, L. and H. Huth, 2003. Lower Cienega Creek Restoration Evaluation Project: An Investigation into Developing Quantitative Methods for Assessing Stream Channel Physical Condition. ADEQ Report #EQR0303, Arizona Department of Environmental Quality, Phoenix, AZ.

3.16.15 Particle Size Distributions

The composition of a stream bed and its banks influence the channel form, erosion rates, sediment supply and other parameters. The particle size distribution, as determined with a pebble count, provides a measurement of the median particle size found within the study reach. The median particle size is one of the components needed to determine Rosgen Stream Type (Rosgen, 1996). In addition, these distributions can be used for comparative purposes to determine, with statistical reliability, if there has been a shift over time in substrate size in the study reach.

There are multiple ways to perform a pebble count for a particle size distribution depending upon the observer's objective. A distilled version of the Zig-Zag Pebble Count Method (Bevenger and King, 1995) is presented. This count is a direct measure of the median particle size for the entire reach. Additional detail on the Wolman pebble count procedure is provided in Harrelson et al. (1994).

3.16.15.1 Zig-Zag Pebble Count Procedure

1. Once the study reach has been delineated, choose a random location on one bank at bankfull stage at either the top or bottom of the reach. For this discussion, the starting point is at the bottom of the reach. The top, mid-point and bottom points of the reach should have already been marked with flagging. The flagging points will help the observer pace the reach in equal segments.

2. From the downstream starting point, identify an upstream target point (not the mid-point) on the opposite bank that can be used to hold a straight line while sampling. Pace off seven feet (2 to 4 steps depending on observers stride), reach over the toe of the wader with the forefinger without looking down, pick up the first pebble touched, and measure the intermediate axis (neither the longest nor shortest of the three mutually perpendicular sides) in millimeters (Figure 24). The seven foot distance can be adjusted for the length of the reach. Determine the Size Range from the Wentworth size classes provided on the Pebble Count Field Data Sheet (Figure 25) and record the tally. Tallies for bank and bed material can be listed separately on the field form. Measure embedded rocks in place on the smaller of the two exposed axes.

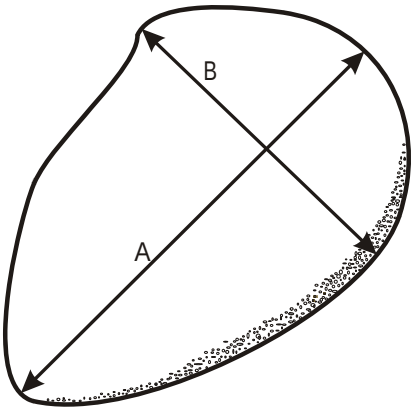


Figure 24. Axes of pebble

A = Longest Axis (length)
B = Intermediate Axis (width)
Thickness = Shortest Axis

Caution - there is a tendency to look down and select a pebble, but this should be avoided or the results will be biased toward larger particle sizes.

3. Discard the measured pebble downstream, focus on the target across the stream and pace another 7 feet for the next pebble.

4. Continue working across and up the stream until the far bankfull stage is reached. Then locate another target, this time on the bank on which the survey began, and work across the creek. Continue this technique until the survey is complete. The result should be a zig-zag line that traverses through the stream reach. The objective is to angle the target lines so that 50 counts have been made by mid-point of the reach and 100 counts at the top of the reach. One hundred counts is the ideal number for this procedure. Complete the pebble count survey at the end of the last transect and at the top of bankfull stage.

   Once the count has been completed, and before leaving the stream, sum the counts to insure that the goal of 100 pebbles have been measured.

6. The pebble count tallies must be graphed to determine the D15, D50 (median particle size), and D84 classes (Figure 26). D50 means that 50% of the particles measured have a mean diameter equal to or smaller than 50 millimeters.

Figure 25. ADEQ pebble count field form.

BED MATERIAL ANALYSIS FORM

Site Name:_____Date:_____

Size Class	Size Range(mm)	Tally	Count	Percent	Cum. %
Silt/Clay*	<0.062				
Sand**	0.063-2				
Very Fine Gravel	3-4				
Fine Gravel	5-8				
Medium Gravel	9-16				
Coarse Gravel	17-32				
Very Coarse Gravel	33-64				
Small Cobble	65-96				
Medium Cobble	97-128				
Large Cobble	129-180				
Very Large Cobble	181-256				
Small Boulder	257-512				
Medium Boulder	513-1024				
Large Boulder	1025-2048				
Very Large Boulder	2049-4096				
Bedrock					
	Totals				

Person Sampling:_____
PersonRecording:_____
Type of Transect:_____
Reach Location:_____
Particle Size Measurements: Template in ___ ϕ gradation;
Calipers: (yes/no); Ruler: (yes/no)
Stream Morphology:_____
_____
Bed Material Structure & Packing:_____
_____
Remarks:_____
_____
_____

%Fines (<2mm)

Size Classes

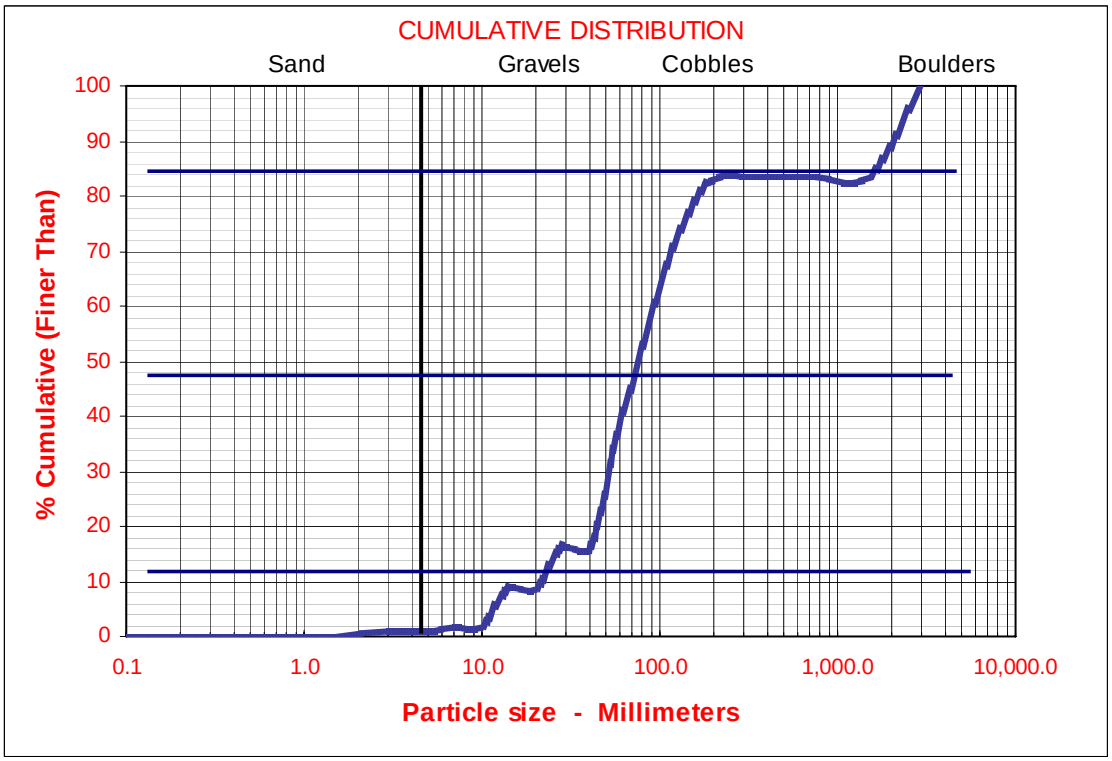
D15 =

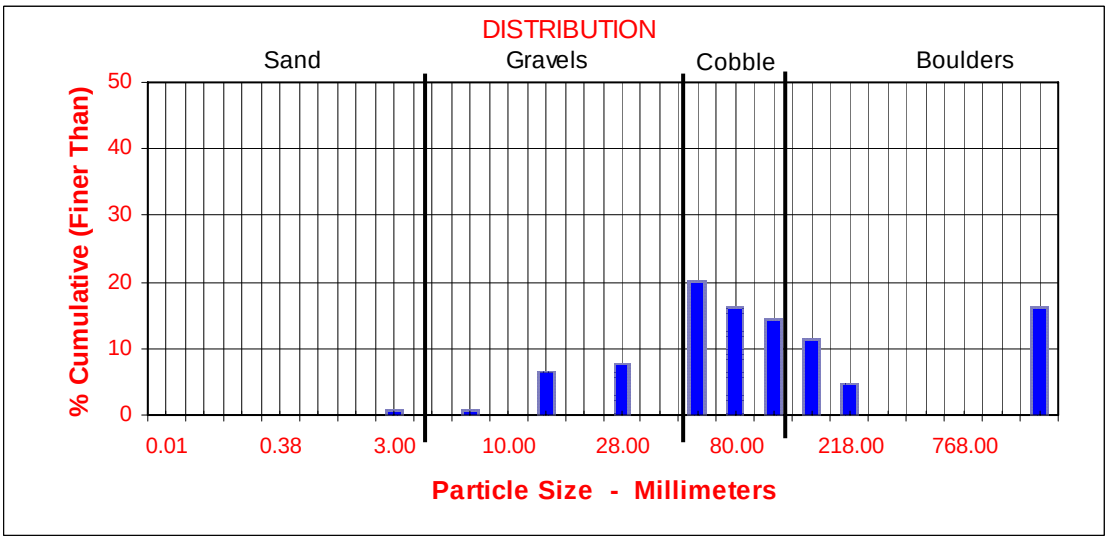
D50 =

D84 =

* Particles feel slick when rubbing between thumb and forefinger
** Particles feel gritty when rubbing between thumb and forefinger

Figure 26. Particle size cumulative distribution graphs, indicating values for D16, D50, D84 and D100 compiled in a bimodal bar chart distribution. The example shows the results of a zig-zag pebble count.





3.16.15.1.1 Literature Cited

Bevenger, G.S. and R.M. King. 1995. A Pebble Count Procedure for Assessing Watershed Cumulative Effects. USDA Forest Service Research Paper RM-RO-319, Fort Collins, CO.

Harrelson, C.C., C.L. Rawlins, and J.P. Potyondy. 1994. Stream Channel reference Sites: An Illustrated Guide to Field Technique. USDA, Forest Service, Rocky Mountain Forest and Range Experiment Station, General Technical Report RM-245. Fort Collins, CO.

Rosgen, D. and H.L. Silvey. 1996. Applied River Morphology. Wildland Hydrology. Pagosa Springs, CO.

3.16.15.2 Riffle Pebble Count Procedure

The riffle pebble count method is a modification of the Wolman pebble count procedure (Leopold, et al. 1964). While the Zig-Zag pebble count is used to identify the Rosgen Stream Type, the riffle pebble count is collected to identify the percent fines within the riffle habitat where macroinvertebrate collections are made. In other words it is a procedure for characterizing particle size distributions of riffle habitats of a study reach. This data is then used to evaluate whether a bimodal particle size distribution exists and to determine the amount of fine sediment in the substrate, affecting colonization space for aquatic life. Secondarily the data is used to determine whether changes in the substrate are occurring over time.

The ADEQ riffle pebble count consists of measuring particles at equal increments across multiple straight transects within the wetted width of riffle habitats where the macroinvertebrates were collected to achieve an approximate 100 count of particles.

1. A study reach of two meander lengths or a minimum of one hundred meters is first established and marked with flagging tape at the top and bottom of the reach. Usually three riffles are selected within the reach for macroinvertebrate sampling. Pebble count measurements will be collected along transects within these same riffles, working from the most downstream transect to upstream transect.

2. Set up a tape with chaining pins across each transect where the macroinvertebrates were previously collected. If three riffles are to be worked, divide the stream width by thirty-three to obtain the increment needed to collect thirty-three particles across the transect in a single pass. Do not collect particles closer than 0.3 tenths of a foot apart. If thirty-three particles cannot collected in one pass along the transect, make a second or third pass as close as possible to the transect tape, and working in an upstream direction without working the same area.

3. Use a marker system to ensure collection of a randomly selected particle. The tip of the pebble count ruler or off the front of a boot, placed at the appropriate station along the transect tape. To take particle readings, reach over the toe of the boot or at the tip of the ruler. Extend the forefinger, and without looking down, pick up the first pebble

touched, and measure the intermediate axis (A) in millimeters (Figure 24). The intermediate axis is neither the longer nor shorter of the three mutually perpendicular sides.

Determine the Size Range from the Field Data Sheet (Figure25) and record the tally. Measure embedded rocks in place by measuring the smaller of the two exposed axes.

Caution - there is a tendency to look down and select a pebble, but this should be avoided or the results will be biased toward larger particle sizes.

4. Discard the measured pebble downstream, move to the next station, and repeat step 3.

5. Continue working across the transect from wetted edge to wetted edge of the streambed. After completing the first thirty-three measurements at this transect, move upstream to the next transect, and repeat the process.

6. Once the count has been completed, and before leaving the stream, sum the tallies to insure that the goal of 100 particles have been counted. If the count is within a count of ten, it is an acceptable assessment.

3.16.15.2.1 Literature Cited

Leopold, L.B., M.G. Wolman, and J.P. Miller. 1964. Fluvial processes in geomorphology. Freeman, San Francisco, CA.

3.16.15.3 Pool Pebble Count

The pool pebble count method is a modification of the Wolman pebble count procedure (Leopold, et al. 1964) and similar to the riffle pebble count. The purpose of collecting pool pebble count data are threefold: 1) to provide a D50 for the associated cross-section and stream type; 2) to combine it with riffle data for a reach pebble count; and 3) to determine whether changes in the substrate are occurring over time.

The pool pebble count consists of measuring particles at equal increments across a transect from bankfull on one side to bankfull on the other. Tallies from the bank and bed are recorded separately and combined to achieve an approximate 100 count of particles. If more than one pool is surveyed, the work is performed from downstream to upstream.

1. A study reach of two meander lengths or a minimum of one hundred meters is first established and marked with flagging tape at the top and bottom of the reach. Usually three pools are selected within the reach for macroinvertebrate sampling.

2. Set up a tape with chaining pins across a transect. If three pools are to be worked, divide the stream width by thirty-three to obtain the increment needed to collect thirty-three particles across the transect in a single pass. Do not collect particles closer than 0.3 tenths of a foot apart. If thirty-three particles cannot collected in one pass through a transect, make additional passes through the transect as close to the first as possible, working from downstream to upstream.

3. Use a marker system to ensure collection of a randomly selected particle. The tip of the pebble count ruler or off the front of a boot, placed at the appropriate station along the transect tape. To take particle readings, reach over the toe of the boot or at the tip of the ruler. Extend the forefinger, and without looking down, pick up the first pebble touched, and measure the intermediate axis (A) in millimeters (Figure 24). Caution - there is a tendency to look down and select a pebble, but this should be avoided or the results will be biased toward larger particle sizes.

   Determine the Size Range from the Field Data Sheet (Figure25) and record the tally.

   For large rocks that are embedded in the streambed, measure the smaller of the two exposed axes.

4. Discard the measured pebble downstream, move to the next station, and repeat step 3.

5. After completing the first thirty-three measurements at this transect, move upstream to the next pool, and repeat the process.

6. Once the count has been completed, and before leaving the stream, sum the tallies from all the pools to insure that the goal of 100 particles have been achieved.

3.16.15.3.1 Literature Cited

Leopold, L.B., M.G. Wolman, and J.P. Miller. 1964. Fluvial processes in geomorphology. Freeman, San Francisco, CA.

3.16.16 Bankfull Identification

Rosgen's classification system for identifying different channel types is based on a common frame of reference among all streams; the bankfull elevation. The bankfull stage is described as the elevation at which incipient flooding occurs; that is the point at which stream flow overtops the natural channel banks and spreads across the floodplain. Evidence from a large number of rivers suggests that these flows are frequent, moderate sized flows with a typical return interval of 1-2 years and that they represent the channel forming or maintenance flows. Similar return intervals were empirically identified for Arizona streams, where the range of return intervals for over 30 gauged stations was determined to be 1.1 – 1.8 years (Moody and Odem, 1999).

The bankfull discharge is also equivalent to the "effective discharge"; the flow which transports the greatest volume of sediment over time. Though very high flows can move significant amounts of material, they occur infrequently and therefore transport only a small fraction of the total sediment volume over time. However, frequent moderate flood events typically carry the greatest amount of sediment; thus the bankfull flow is the most common channel shaping flow. The bankfull elevation must be consistently identified in the field in order to correctly identify the stream type. The stream type depends on measurements of bankfull width/depth ratio and entrenchment ratio which are dependant upon measurements of the bankfull stage.

The following procedure is a distillation of bankfull identification procedures found in Rosgen (1996), Harrelson et al (1994) and Moody et al (2000).

3.16.16.1 Field Procedure for Identifying Bankfull

1. Walk a stream reach of a minimum of two meander lengths to be assessed looking for bankfull indicators, such as:

   - topographic breaks in slope
   - tops of point bars
   - changes in vegetation
   - changes in size of bank or bar materials
   - evidence of an inundation feature such as small benches
   - the presence of a floodplain
   - exposed root hairs below an intact soil layer indicating exposure to erosive flow
   - bank undercuts.

   Vegetation is usually not a good bankfull indicator and must used with caution. At high elevations, an ash tree or willow tree line may at times be useful; however, a grass or seep willow line at any elevation is not.

2. Place stake flags, pieces of flagging, or other marking devices on the identified points along the reach where bankfull indicators are present. If bankfull stage has been properly identified, the stake flags should delineate a line identifying bankfull depth.

2. Conduct a longitudinal profile, surveying the identified bankfull features throughout the study reach.

3. Plot the longitudinal profile and draw a line through the alluvial features representing bankfull and floodplain elevation (Figure 27). The plot should verify bankfull depth. If it does not, then bankfull height must be re-evaluated and the bankfull identification procedure repeated.

4. A "regional curve" can also be used as another verification tool for bankfull identification. A regional curve is an empirically derived graph. A regression formula depicts the relationship of watershed area and channel characteristics. The most

commonly used Arizona statewide regional curve is the watershed area-channel cross-sectional area graph shown in Figure 28, derived by Moody et al (2000). The "Central and Southern Arizona" regression curve applies to streams in the Verde, Middle Gila, San Pedro, and Santa Cruz River Basins. The "Eastern Arizona and New Mexico" regression curve applies to streams in the Little Colorado River, Upper Gila River and Salt River Basins. Though a large amount of variability exists for these curves, they do provide a ballpark value of the bankfull elevation to screen for gross level errors in field bankfull identification. Local curves for sub-watersheds can be used for more accurate estimates of bankfull where available.

3.16.16.2 Literature Cited

Harrelson, C.C., C.L. Rawlins, and J.P. Potyondy. 1994. Stream Channel reference Sites: An Illustrated Guide to Field Technique. USDA, Forest Service, Rocky Mountain Forest and Range Experiment Station, General Technical Report RM-245. Fort Collins, CO.

Moody, T. and W. Odem. 1999. Regional relationships for bankfull stage in natural channels of Central and Southern Arizona. Prepared for USDA Forest Service and Arizona Department of Environmental Quality.

Moody, T, M. Wirtanen, K. Knight, and W. Odem. 2000. Integrating regional relationships for bankfull stage in natural channels of Arizona and New Mexico. Northern Arizona University, Flagstaff, AZ.

Rosgen, D. and H.L. Silvey. 1996. Applied River Morphology. Wildland Hydrology. Pagosa Springs, CO.

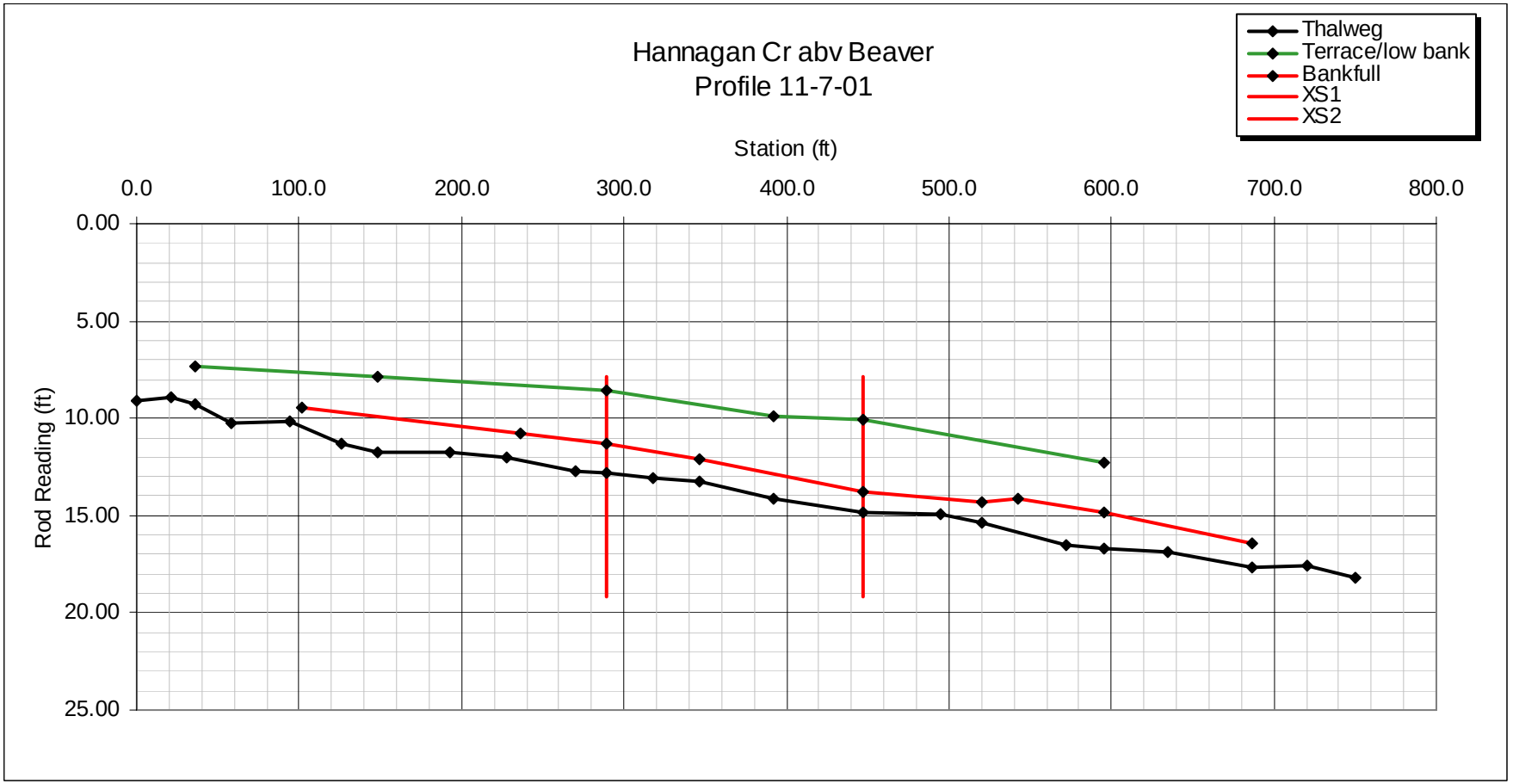
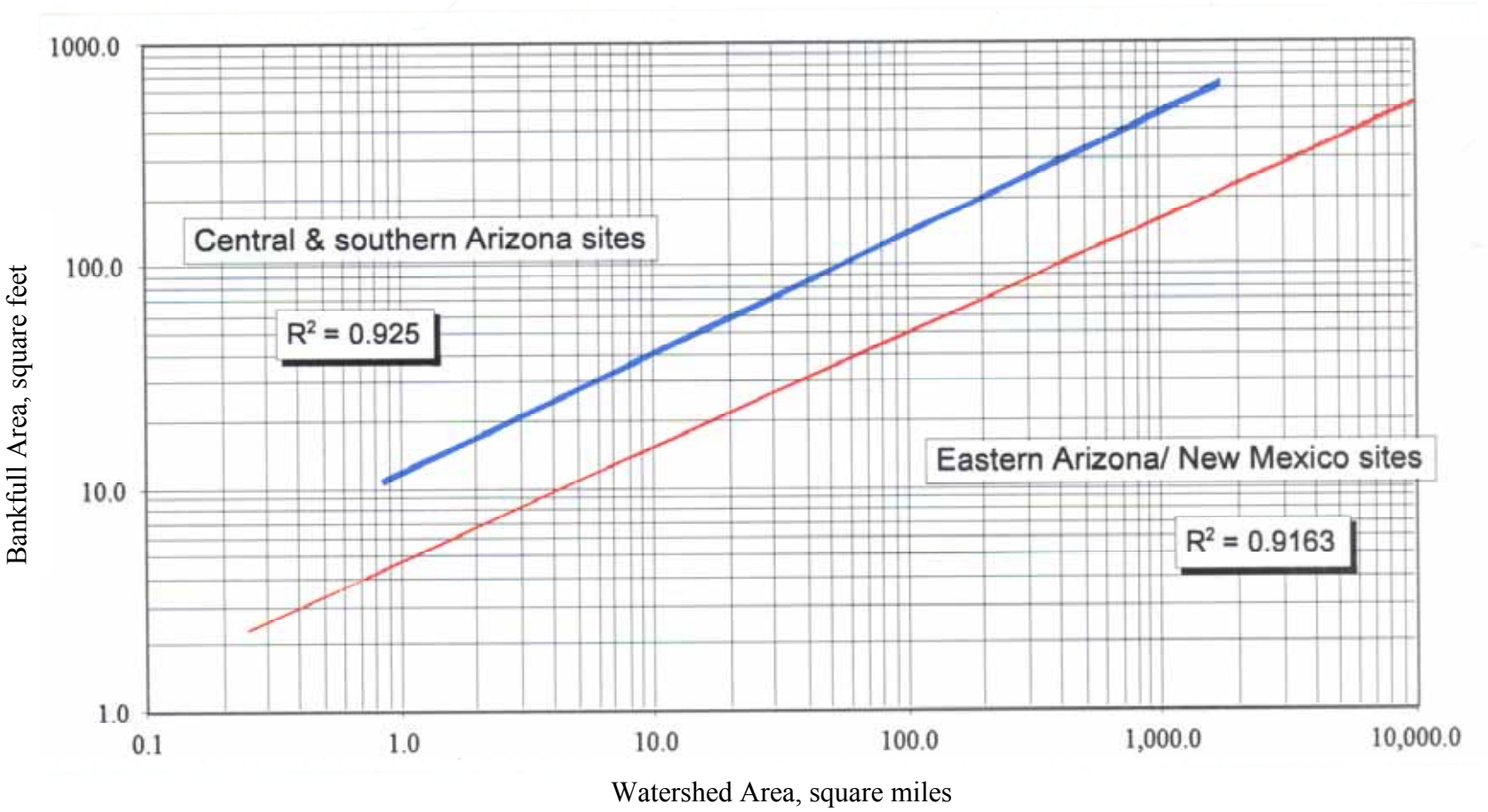


Figure 27. Example of bankfull identification using a longitudinal profile.

Figure 28. Arizona statewide regional curves for predicting channel cross-sectional area based upon watershed area (Moody et al. 2000).



SECTION 3

PART C

HABITAT ASSESSMENT PROCEDURES

TABLE OF CONTENTS

3.17 Habitat Assessments

The habitat assessment provides ecological information needed to interpret macroinvertebrate bioassessments. Habitat assessments are conducted by analyzing substrate, channel, riparian and other measures that are collected and recorded on ADEQ's Stream Ecosystem Monitoring (SEM) forms (Section 1.13.2) which are found at the end of this section. These field forms provide for the collection of water chemistry, discharge data, field observations about the hydrology, biology and general condition of the stream reach, non-point source observations, a site sketch, photos, the ADEQ Habitat Assessment Index, Rosgen stream type classification, and a riparian community assessment. The sum of all these data provide an ecological context in which to place the macroinvertebrate data. Causes and sources of biological impairment can be explained by the chemical physical, biological and land use information produced in the habitat assessment.

3.17.1 Site information

Page one of ADEQ's Stream Ecosystem Monitoring field data sheets (FDS) provides for the collection of locational information needed for data entry, mapping and general site information. Required information includes stream name and site description, date, time, site code, and field crew. Optional information includes watershed name, HUC-reach number, upland vegetation formation, designated uses, and site recommendations.

3.17.2 Field measurements, Observations and Sample Collection Information

Precipitation, cloud cover, air temperature, and field measurements of the water should be recorded. There is space on the field data sheet for more than one measurement of physicochemical parameters to allow for variations in measurements over space and time. For sample collection information, record the water collection method, which samples were collected, which analyses will be performed, sample time and what kind of QC sample was collected, if any.

Bacteria analyses (*E. coli*) are recorded. The procedures for collection and analysis are found in Section 3A, 3.13.

Stream habitat variables are selected as a decision tree to inform the sampler when it is an appropriate time to sample.

A discharge measurement is recorded using one of four methods: flow meter method (Section 3C, 3.18.1), float method (Section 3C, 3.18.2), USGS staff height (Section 3C, 3.18.3), and volumetric method (Section 3C, 3.18.4). Other sampling methods can be found in Harrelson, et al., 1994.

3.17.2.1 Non-Point Source Observations

Sources of potential impairment must be identified as part of the bioassessment process. A list of non-point sources (Figure 1) as used in the 305(b) water quality assessment process are tabled. Sources adjacent to the study reach as well as sources within the watershed are identified from visual observations in the field and from topographic maps or aerial photos.

Figure 1. Non-Point Source Codes.

NON-POINT SOURCE CODES

Circle sources directly impacting the site, asterisk sources located in the watershed. Source Group is bolded, Category Code is italicized, and Sub-category Code is regular style font.

Code	Source Category	Code	Source Category	Code	Source Category
1000	**Agriculture** *(Agriculture)*	7350	Upstream impoundment	6600	Hazardous waste storage/disposal
1050	Crop-related sources	7400	Flow regulation/Modification/Diversions	8000	Highway salt storage/use
1100	Non-irrigated crop production	7550	Habitat Modification	8200	Storage tank leaks
1200	Irrigated crop production	7555	Erosion materials from tributaries	8250	Underground storage tank leaks
1300	Specialty crop production	7600	Removal of riparian vegetation	8275	Above ground storage tank
1350	Grazing-related sources	7700	Streambank modification or destabilization	**0100**	**Wastewater** *(Industrial Point Source)*
1400	Pasture grazing - riparian and/or upland	7750	Highway/Road/Bridge-erosion or aggradation	0110	Major industrial point source
1410	Pasture grazing - riparian	7800	Drainage/Filling of wetlands	0120	Minor industrial point source
1420	Pasture grazing - upland	*7900*	*Marinas and recreational boating*	0200	Municipal point source
1500	Range grazing - riparian and/or upland	7910	Boating with in-water releases	0210	Major municipal point source
1510	Range grazing - riparian	7920	Boating with on-land releases	0220	Minor municipal point source
1520	Range grazing - upland	**5000**	**Mining** *(Resource extraction)*	0230	Package plants (small flows)
1600	*Intensive Animal feeding Operations*	5075	Active Mining operation	*0300*	*Other Wastewater*
1620	Concentrated Animal Feeding Operations point source/permitted)	5100	Surface Mining	0400	Combined system (sewage and stormwater)
1640	Confined animal feeding operations (non-point source	5150	Sand and gravel operations	0500	Collection system failure
1700	*Aquiculture/Fish Hatchery*	5200	Subsurface mining	0900	Sewage lagoons
2000	**Forestry** *(Silviculture)*	5300	Placer mining	0975	Reuse (Effluent to lakes, golf courses, artificial
2100	Harvesting, restoration (residue management)	5400	Dredge mining	6500	Septic systems
2200	Forest management (fertilization, pesticide use)	5500	Petroleum activities	6700	Septage disposal (e.g. from septic tank trucks)
2300	Logging roads	5600	Mill tailings	**8100**	**Other** (Atmospheric deposition)
2500	Clear cutting	5650	Mill or mine tailings	8400	Spills
8610	Wildfires or controlled burns	5700	Mine tailings	8500	Contaminated sediments
3000	**Hydro/Habitat Modification/Runoff** *(Construction)*	5800	Acid mine drainage	8510	
3100	Highway/Road/Bridge construction	5900	Abandoned mining operation	8530	
3200	Land development/Land clearing	5950	Inactive mining operation	8540	
4000	*Urban runoff/Stormwater sewers*	**8700**	**Recreation** (non-boating)	8600	
4100	Non-industrial (NPDES) stormwater runoff	8710	Golf courses	8910	
4200	Industrial (NPDES) stormwater runoff	8720	Camping/Campground recreation	Other Non-point Source Observations at the site or within the reach	
4300	Other urban runoff	8730	All terrain vehicles/Off road vehicles/Biking		
4400	Illicit connections to stormwater sewers (dry weather flows)	**6000**	**Storage and Disposal** (Land disposal/Storage)		
4500	Urban Highway/Road/Bridge runoff	6100	Sludge disposal/storage		
4600	*Non-urban runoff/Erosion and sedimentation*	6300	Landfills		
8300	Non-urban (highway/Road/Bridge Runoff/Maintenance)	6350	Inappropriate waste disposal/Wildcat dumping		
7000	*Hydrological modifications*	6400	Industrial land treatment		

3.17.2.2 Reach Observations

Narrative observations about the general stream condition can be helpful in diagnosing potential problems. The observations consist of general appearance of the stream reach and streambank, water appearance and odor, presence of fish, especially sunfish and crayfish as well as hydrological information about flood or drought evidence, flow regime and water source. Biotic interactions by exotic species such as crayfish and sunfish are an important source of impairment of the macroinvertebrate community. Hydrological information is important for identifying flood or drought impacts, and ensuring that the stream is perennial prior to macroinvertebrate sample collection.

3.17.2.3 Algae/Macrophytes

Observations about the percent cover by filamentous and diatomaceous algae and macrophytes can be very helpful for diagnosing nutrient problems and understanding the trophic structure of the macroinvertebrate community. Notes are collected on the percent cover of filamentous algae and macrophytes, the presence of floating algae mats and diatom cover, and the types of flora present in the study reach.

- Percent cover by filamentous algae is visually estimated for the whole study reach. Filamentous algae consists of green and blue-green algae that can form small tufts to large beards attached to substrates.
- Floating algae mats refers to detached clumps of green or blue green algae present in the stream. Large volumes of algae are usually an indication of nutrient enrichment. Sometimes floating mats will naturally occur in desert streams in summer or during drought conditions, when senescent communities develop.
- Algal slime refers to the abundance of diatom cover on substrates. The diatom cover can be detected visually as a brown staining on cobbles and can be felt as a slippery coating of material on rock surfaces.
- Macrophytes are generally found along the edge of water, but sometimes are found in shallow water or covering the stream bed (water grass). Percent cover is visually estimated for the study reach and is generally a low number unless there is nutrient enrichment.
- Identification of aquatic plants and algae species can be helpful in understanding nutrient cycling (e.g. abundance of nitrogen fixing blue-green algae indicates that nutrients are bound up in vegetation and are not abundant in the water) and whether nuisance species of macrophytes are present. Hydrilla and Eurasian milfoil infest waterbodies and are difficult to eradicate. Identification guides are available. We currently use the following identification references:

    - How to Know the Freshwater Algae (Prescott, 1978)
    - University of Florida macrophyte key (Ramey, 1995)
    - Western Wetland Flora (Mohlenbrock et al., not dated)

3.17.2.4 Site sketch

A sketch of the stream reach provides a visual representation of the general habitat available to the macroinvertebrate community. The site sketch demonstrates the relative proportions of macro-habitats, such as riffles, runs, and pools. It should also display micro-habitats such as woody debris, leaf packs, macrophyte and algae beds, undercut banks, and riparian vegetation. The sketch can also present potential sources of impairment such as excess sediment in the form of side and mid-channel bars from cut banks or degraded tributaries. The map should be scaled to include the entire study reach, displaying floodplains, terraces, features such as trees, rocks or flood debris, the stream name, date, direction of stream flow, a north arrow, benchmarks, point bars, abandoned channels, and sample locations.

3.17.2.5 Photo Monitoring

Photos are required to track changes in the channel over time and to demonstrate the substrate and channel conditions at the time of sampling. See Section 3C, 3.17.11 for the photo monitoring procedure.

3.17.2.6 Habitat Assessment Index

While all the notes in the field data sheets are considered part of the broad level habitat assessment, ADEQ provides a specific substrate and bank habitat assessment. The habitat condition parameters were extracted from USEPA's visual based habitat assessment protocols described in the Rapid Bioassessment Protocols (Barbour et al., 1999) and USEPA's Environmental Monitoring and Assessment Protocols (Lazorchak et al (eds.), 1998).

3.17.2.6.1 Riffle Habitat Quality

Habitat quality within riffles is evaluated through a survey of the variety of natural structures within the stream reach, such as cobble, large rocks, woody debris, and undercut banks available for colonization by macroinvertebrates. A wide variety and abundance of submerged structures provides benthic macroinvertebrates with a large number of habitat niches, thus increasing community diversity. As the habitat structure becomes less complex, the variety and abundance of cover decreases. Habitat loss leads to a decrease in community diversity, and the potential for community recovery lessens.

Complete the Habitat Assessment Field Data Sheet prior to conducting the habitat scoring. Walk a minimum distance of 100m or at least 25 times the bankfull width of the stream, identifying the relative abundance of each micro- and macro-habitat. For warm water streams, give an optimal score if there are 2-3 habitats in the common to abundant categories; suboptimal if there are 2+ habitats with 1 abundant; marginal if sand is common or abundant with 1 additional habitat; poor

if the habitat is dominated by abundant sand with possible algae or macrophytes present. For cold water streams, give an optimal score if there are 3+ habitats in the common to abundant categories; suboptimal if there are 2+ habitats with 1 abundant; marginal if there are 2+ habitats that are rare or common; poor if the habitat is dominated by abundant sand with possible algae or macrophytes present.

3.17.2.6.2 Extent of Riffle Habitat

In addition to habitat quality, the quantity of the riffle habitat is an important factor for the support of healthy biological stream communities. Good riffle habitat covers the width of the streambed, extends twice the width in riffle length, and is populated with an abundance of cobble. When present, these factors provide abundant habitat for maintenance of the macroinvertebrate community and support of the aquatic food web. Where cobble substrate is lacking, riffles may also be lacking. In streams with excess sediment, the interstitial spaces around the rocks fill with sand which converts the riffle to a sandy run. The lack of habitat in sandy runs prevent macroinvertebrate communities from developing.

Complete the Riffle Geometry portion of the field form prior to conducting the habitat scoring. Mark the widths and lengths of three or more riffles in the study reach. Calculate the length to width ratios for each and then calculate the average ratio. Use these data to score the Extent of Riffle Habitat.

3.17.2.6.3 Embeddedness in Riffles

Embeddedness refers to the extent to which rocks (gravel, cobble, and boulders) and woody debris are covered or sunken into the silt, sand, or mud in stream riffles. As rocks become more embedded, the surface area available as habitat for macroinvertebrates decrease. Embeddedness is the result of an infusion of fine sediments from upland and stream bank erosion into stream substrates. Embeddedness is one of the primary measures of excess bottom deposits.

Complete the Riffle Embeddedness portion of the field form prior to conducting the habitat scoring. Refer to the field data sheet for the Embeddedness calculation procedure. The sample transects should be traversed in the same location(s) as the macroinvertebrate samples. Collect particles at even increments across each transect. Visually estimate percent particle embeddedness. Begin and end transect at the edges of riffles. Count sand and fines as 100% embedded and bedrock, and hardpan as 0% embedded.

3.17.2.6.4 Sediment Deposition

This parameter measures the amount of sediment that has accumulated on the stream bottom and in pools throughout the reach, and for large-scale movement of sediment into a stream. Sediment deposition may cause the formation of side or mid-channel bars, enlargement of point bars, or may result in the filling of riffles and pools. Usually sediment deposition is evident in

areas that are obstructed by natural or manmade debris and in areas where stream flow decrease, such as at bends. Large amounts of fine sediment deposition throughout the reach creates a homogenous, unstable, sandy substrate that is unsuitable for macroinvertebrate colonization.

Complete the Depositional Features and riffle pebble count sections on the field form prior to conducting the habitat scoring. For the depositional features parameter, mark all categories that apply to the stream channel within the study reach of at least 25 times the bankfull width. Keep in mind that Rosgen A and B type streams are usually without depositional features.

The riffle pebble count is a modified version of the Wolman pebble count (Leopold, et al. 1964). The purpose of the riffle pebble count is to evaluate whether a bimodal particle size distribution exists and to determine the amount of fine sediment in the substrate. The ADEQ riffle pebble count consists of measuring particles at equal increments across multiple transects within the wetted width of riffle habitats where the macroinvertebrates were collected. The count objective is 100 particles. See Section 3B, 3.16.15 for pebble count procedures.

Field staff should be familiar with Rosgen stream types (Rosgen, 1996) and be able to identify the stream type of the study reach. The stream type is required to evaluate whether excess sediment is present for that stream type and to determine whether the channel features conform to the model for a stream type. There is additional discussion of bar features, bimodal particle size distribution, and excess sediment presented in each category of the sediment deposition parameter in Section 3B, 3.16.

3.17.2.6.5 Bank Stability

The bank stability parameter evaluates the active bankfull channel and is an indicator of the source and amount of sediment contributing to sediment deposition in the stream. Stable, well vegetated banks with little erosion will maintain a stable geomorphic profile and adequate cobble habitat. Unstable banks are characterized by steep walls, banks devoid of vegetation, exposed tree roots, and exposed soil. Unstable banks will erode during moderate flows, contributing large amounts of sediment to the stream bed.

Bank stability is evaluated by visual estimation or measurement of the percent of bank erosion for each bank. Both bank scores are summed for the total bank stability score. A visual estimate of bank erosion for each bank is determined from markings on the site sketch. To measure bank erosion, the length of eroding banks can be paced off or measured with a reel tape, as a percentage of the total bank length. Any bank height greater than bankfull height is considered to be eroding.

3.17.2.6.6 Habitat Assessment Index Scoring

Scores for the five habitat parameters are summed for either a warm water or cold water index score. The habitat parameters are the same but the field assessment criteria are slightly different in the cold and warm water Habitat Assessment Indexes, allowing for the increased habitat

diversity common in cold water streams. The five in-stream and bank habitat parameters are scored on a scale of 1 to 4, with higher scores indicating better condition. The habitat scores are summed for a total habitat score ranging from 6-24, with habitat improving with increasing scores. The Habitat Assessment Index score is then categorized as being good, impaired or very impaired, using the 25th percentile of ADEQ reference habitat assessment scores as the criterion. The 25th percentile of reference method was selected because it is a conservative scoring criteria and allows for the natural variance among reference site scores. The range of values for each Habitat Assessment Category are shown in Table 1. The scoring criteria are the same for both the cold and warm water Habitat Assessment Indexes. If both the IBI score and Habitat Assessment Index score are impaired, then the biological impairment is determined to be associated with sediment and habitat impairment.

Table 1. Habitat Assessment Index categories for perennial, wadeable streams in Arizona.

Habitat Assessment Category	Good	Impaired	Very Impaired
Habitat Assessment Index Scores	15 - 20	8 - 14	0 - 7

3.17.3 Rosgen Stream Type Identification

Six stream types, with associated letter codes A to G, were classified by Rosgen (1996). The stream type is determined from the following five variables:

1) entrenchment ratio
2) bankfull width/depth ratio
3) sinuosity
4) slope
5) median particle size of the channel material.

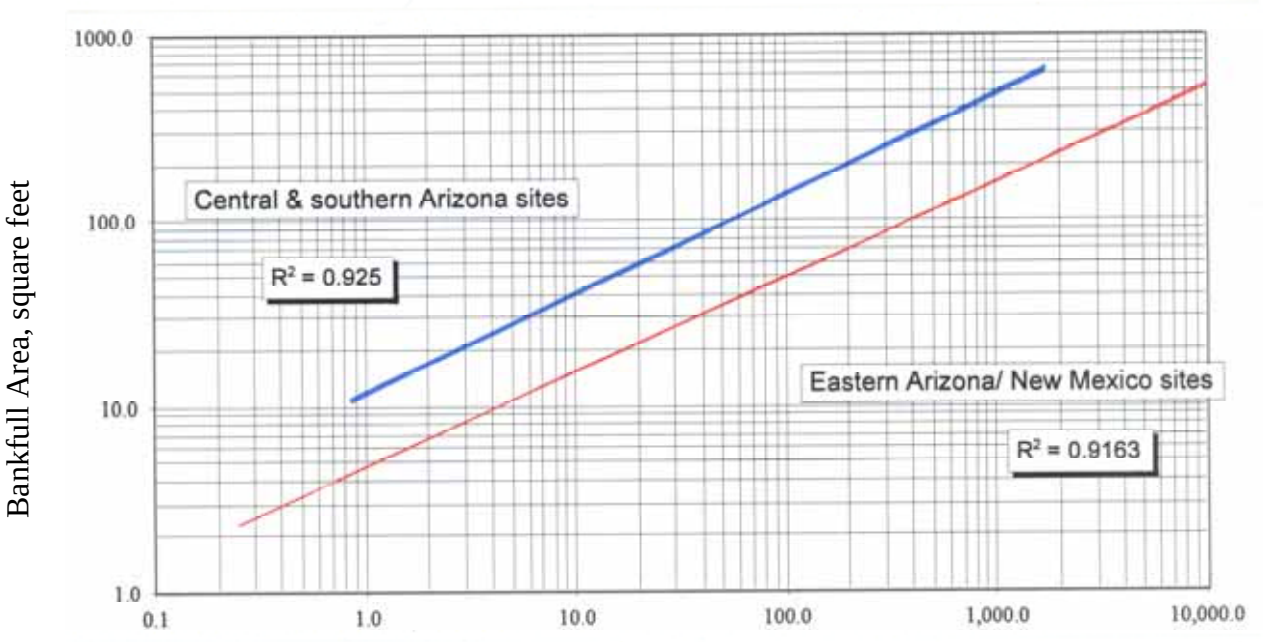
The following procedure describes methods for determining stream type without a rotating laser level, using the Arizona Regional Curves (Figure2) for determination of cross-sectional area.

The Regional Curve is a series of regression lines on a log-log graph of watershed area vs. cross-sectional area of channels at bankfull (Moody and Odem, 1999). The curve is based on empirical data from 66 gauged and ungaged statewide study sites, and is a good generic tool for predicting the cross-sectional area and bankfull elevation expected at a study site of known watershed area. There are two regression lines, one for central and southern Arizona, and one for eastern Arizona and New Mexico. Apply the central and southern Arizona curve for streams in the Verde, Santa Cruz, and San Pedro and middle/lower Gila Rivers. Apply the eastern

Arizona and New Mexico curve to streams in the upper Little Colorado River, upper Gila and Salt Rivers and their tributaries.

1. Identifying bankfull width and depth - The first task is to make a "toothpick" survey by walking the study reach, marking bankfull indicators with flag stakes for a stream reach length of at least 25 times bankfull width. Bankfull indicators include: top of point bars, changes in particle size, slope breaks, vegetation lines, presence of a floodplain at the point of incipient flooding, undercut banks, or rock stains. Vegetation lines should be used with caution and are not good indicators at most elevations, but will sometimes confirm another indicator. At a representative riffle cross-section, run the measuring tape across the stream channel, and to the width identified as bankfull during the tooth-pick survey. Position the stadia or extendable rod in the thalweg against the measuring tape. Ensure that the height of the extended tape is horizontal to the channel or water surface. Determine maximum depth at the junction of the tape and the stadia rod.

Figure 2. Regional Curves for Arizona and New Mexico.



2. Cross-sectional area (A_{bkf}) of the channel at bankfull elevation can be derived from bankfull width (W_{bkf}) and mean depth ($\overline{d}_{bkf}$). Mean depth is calculated by multiplying the bankfull maximum depth (D_{bkf}) by the coefficient, 0.6. The product is then multiplied by W_{bkf} to obtain cross-sectional area.

3. Compare this field calculated value against the appropriate regional curve value. The field value should be similar or close to the predicted value. The closeness is a best professional judgment. If the field value is very dissimilar from the predicted value,

there may be an explanation for the discrepancy. Typical explanations are an incorrect bankfull depth, a water diversion, or an impoundment in the watershed. If the issue cannot be resolved, use the field observed bankfull indicators to obtain cross-sectional area.

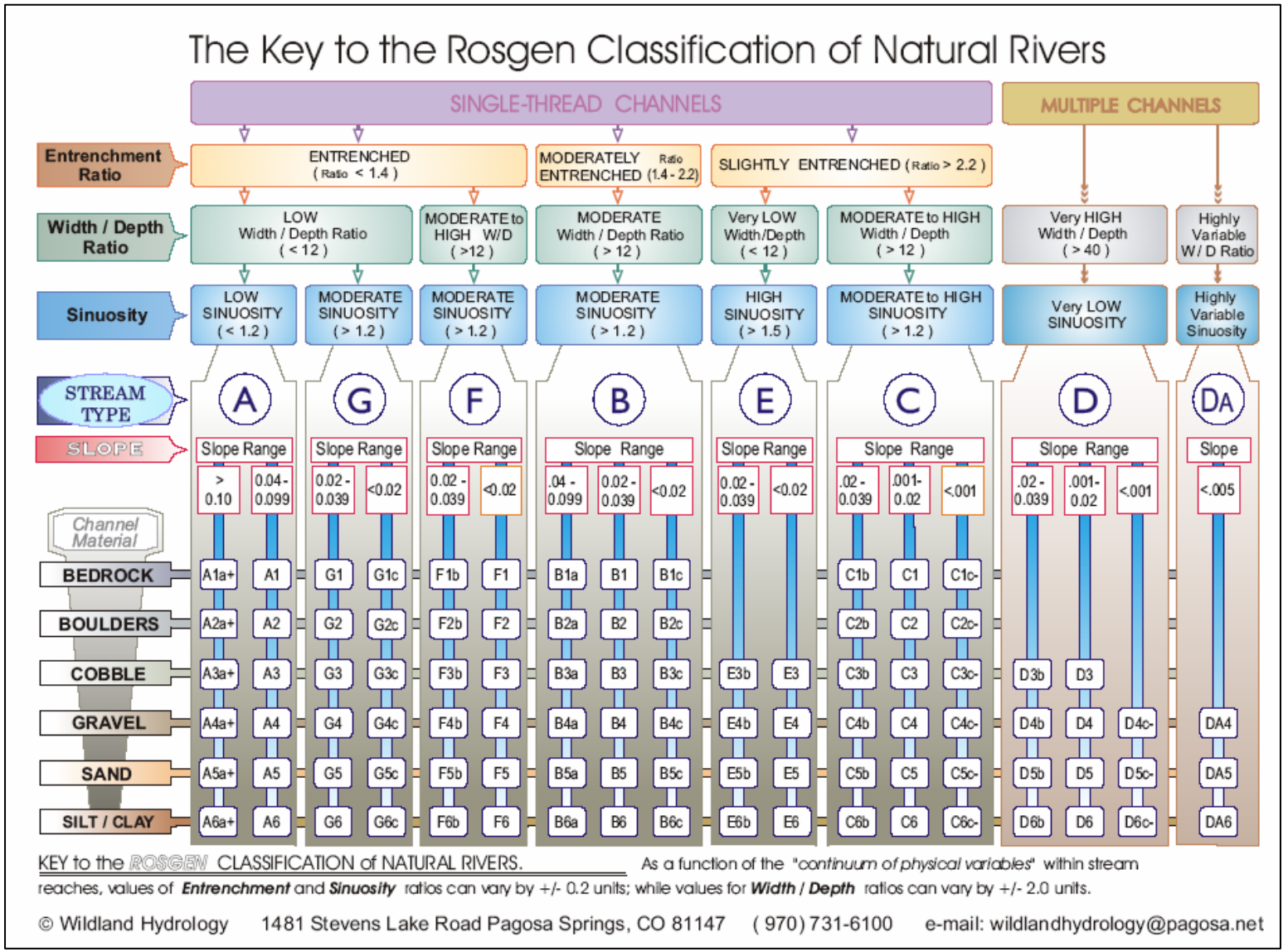
4. To identify floodprone width, take a width measurement at 2 times the maximum bankfull depth, measured in the thalweg. The width measured at this elevation should be close to the correct floodprone width. Other field observations of floodplain indicators may be used as well. The field protocol for doing this is to maintain the stadia or telescoping rod in the thalweg, then raise the tape to the Floodprone Depth and run the measuring tape out on both sides of the stream along the cross-section until earth is encountered. Ensure that the tape is horizontal to the water surface or streambed if ephemeral. The distance between the two ends is the measured Floodprone Width.

5. The five classification variables are calculated and Rosgen's classification chart is used to identify the stream type.

    - ❖ Entrenchment Ratio is calculated by dividing floodprone width by bankfull width.
    - ❖ Bankfull width/depth ratio is calculated by dividing bankfull width by bankfull mean depth ($D_{bkf} = A_{bkf} / W_{bkf}$; where D_{bkf} is bankfull mean depth, A_{bkf} is bankfull area, and W_{bkf} is bankfull width).
    - ❖ Sinuosity can be calculated from a topographical map for the study reach.
    - ❖ Slope can be calculated from a topographical map for the study reach,or if available, a longitudinal profile of the streambed.
    - ❖ The median particle size (D50) can be estimated from the riffle or reach pebble count cumulative percent data or from a graph of the cumulative percent by particle size class.

6. Determine stream type by using Rosgen's classification chart (Figure 3) and the five classification variables. The classification chart can also be found on page 5-6 in Applied River Morphology (Rosgen, 1996).

Figure 3. Rosgen stream type classification chart.



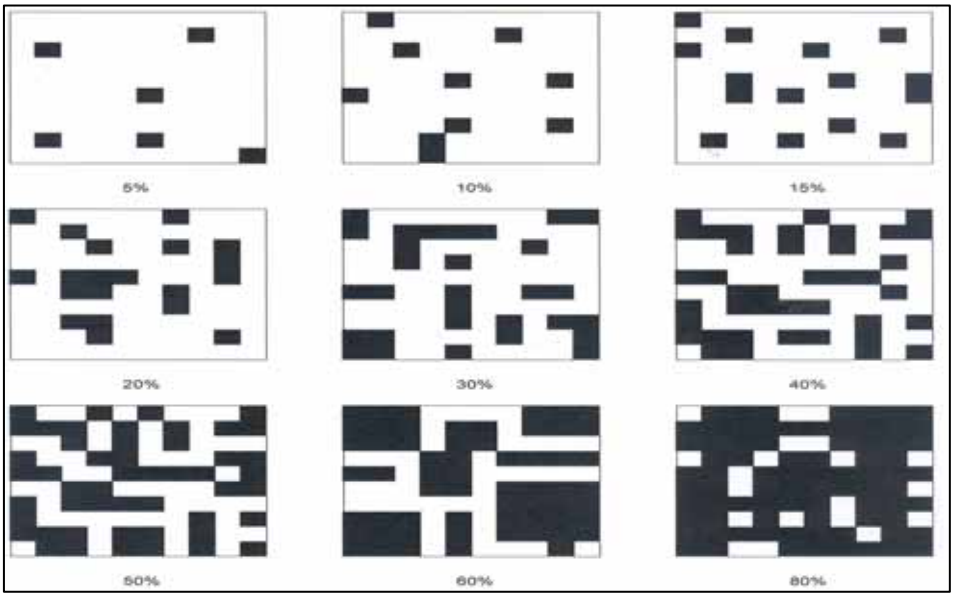
3.17.4 Riparian Community Assessment

The riparian community assessment is comprised of the estimate of vegetation cover and the observations of riparian associations and riparian species.

3.17.4.1 Vegetation Cover

Estimating percent cover of different canopy layers of the riparian community is a semi-quantitative measure of riparian condition. Record the estimated percent cover of the overstory of riparian trees, the understory of shrubs, ground cover and barren ground. Consider each vegetative layer separately with a score of 0-100 percent for each. Use the "Methods of measuring areal extent" provided on the field data sheet for visually estimating percent cover (Figure 4).

Figure 4. Figure used for estimating percent areal cover.



3.17.4.2 Riparian Associations and Riparian Species

The riparian association indicates the broad biotic community or zone in which the site occurs. This is identified primarily by presence of riparian tree species and secondarily by elevation in which it occurs. Individual riparian species may also be listed or circled on the field data sheet page. Some riparian species identification guides include:

- ❖ Shrubs and Trees of the southwest uplands (Elmore and Janish, 1976)

- Desert Plants: Riparian forest and scrubland community types of Arizona and New Mexico (Szaro, 1989)
- Trees of Arizona (Little, 1968; Brockman, 1968)
- Salicaceae: willow family key (Argus, 1995)
- Willow quick ID guide (ADEQa)
- Riparian tree quick ID guide (ADEQb)

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3.17.5 Measuring Percent Canopy Density with the Spherical Densiometer

Percent canopy density is measured with a Spherical Densiometer, manufactured by Forest Densiometers, Bartlesville, OK Section 3.17.10 details the procedure for measuring percent canopy density.

3.17.6 Regeneration Potential

Observations of regeneration capacity aid in evaluating the health of the riparian community. A stressed community will exhibit reduced age class diversity, changes in percent cover, loss of species diversity and increased abundance of exotic species. To complete the regeneration potential table, record the presence of the five most common trees in four age classes; mature trees, young trees, saplings, and seedlings. The community is considered in best condition if tree species are abundant in three age classes. Complete the assessment of riparian condition by filling out the scoring table (Table 2) given on the Stream Ecosystem Monitoring Field Data Sheets.

Table 2. Regeneration Potential of Riparian Trees.

Regeneration Potential of Riparian Trees					
Species		Mature Trees>16" @ 3 ft height	Young Trees~1 ¼" @ 3 ft. height	Saplings< 1 ¼"	Seedlings New growth
1					
2					
3					
4					
5					

Age Classes of the Dominant Riparian Tree Species							
☐	Species abundant in 3 age classes	☐	Abundant in 2 age classes	☐	One age class present	☐	No regeneration evident, few mature trees present, no saplings or seedlings, or if present, they are heavily grazed

3.17.7 Proper Functioning Condition (PFC) Assessment

Proper Functioning Condition (PFC) is a qualitative method for assessing the condition of riparian-wetland areas (Prichard et al,1993). The term PFC is used to describe both the assessment process, and a defined, on-the-ground condition of a riparian-wetland area.

The PFC assessment refers to a consistent approach for considering hydrology, vegetation, and erosion/deposition (soils) attributes and processes to assess the condition of riparian-wetland areas. A checklist is used for the PFC assessment which synthesizes information that is essential for determining the overall health of a riparian-wetland system. The on-the-ground condition termed PFC refers to how well the physical processes are functioning. PFC is a state of resiliency that will allow a riparian-wetland area to hold together during high-flow events with a high degree of reliability. This resiliency allows an area to produce desired values, such as fish habitat, bird habitat, or forage, over a period of time. Riparian-wetland areas that are not functioning properly cannot sustain these values.

PFC is a qualitative assessment based on quantitative science. The PFC assessment is intended to be performed by an interdisciplinary team with local, on-the-ground experience in the kind of quantitative sampling techniques that support the PFC checklist. These quantitative techniques are encouraged in conjunction with the PFC assessment for individual calibration, where answers are uncertain, or where experience is limited. PFC is also an appropriate starting point for determining and prioritizing the type and location of a quantitative inventory or monitoring.

The PFC form consists of a set of guidelines for filling out the checklist (Figure 5 and 6). The guidelines are from BLM training courses and training materials. The guidelines should accompany the checklist into the field and be referred to as the checklist is being filled out by the assessment team.

Several of the field data sheet habitat measurements should be used to assist the PFC evaluations, such as depositional features, pebble count, regeneration potential, and Rosgen stream type. If a "No" answer is given for any of the PFC items, a remark must be given that describes the condition. The number of yes and no answers on the checklist are used to summarize the overall condition into one of six categories: Proper functioning condition, Functional at risk/upward trend, Functional at risk/downward trend, Functional at risk/no apparent trend, Non-functional, and Unknown. There is no numeric scoring involved. Best professional judgment is used to determine the appropriate assessment category.

3.17.8 Field Observations

Field observations can be helpful in identifying field conditions which may have affected the sample. Note hydrologic conditions, channel features, streambed structure, sedimentation issues, possible predators, comments on the riparian vegetation, or other notes about potential non-point sources, relocating the site, and any future recommendations for the study site.

Figure 5. Guidelines for Completing the PFC Checklist.

GUIDELINES FOR COMPLETING THE PFC CHECKLIST

General guidance: If 75% or more of stream reach is PFC, classify entire reach as PFC. All "No" answers must have comments in notes section. Answers can go on the line between "Yes" and "No", but consider it a "No" and comment in notes section.

Q1. Instantaneous peak flows don't count. Inundation means to bankfull depth. Bankfull can be identified from top of the point bars, changes in vegetation, topographic break in slope, change in size of bank materials, evidence of an inundation feature such as small benches, exposed root hairs below an intact soil layer indicating exposure to erosive flow, and bank undercuts. "No" if channelization or entrenchment. "N/A" if a "V"-canyon without floodplain development.

Q2. Usually "N/A", but may be applicable at high altitude sites; also, consider the present environment (could they be present).

Q3. Based on the stream type expected & the regional curves, all three features must be present for a "YES". Use bankfull width, not wetted width. "NO" if straightness, excessive sediment, or entrenched channel.

Q4. Widening can mean encroaching on the channel as well as moving toward the terraces. The age of the vegetation in an indicator. "NO" if upland species encroaching on the floodplain or Kentucky bluegrass present. "YES" if recruitment of wetland/riparian species on new landforms. "N/A" if an A1 stream type.

Q5. Need to look at upland ground cover and erosion signs (e.g. plants on pedestals, debris dams around plants, rills, gullies). "NO" if side channel and mid-channel bars, gullies, fan shaped deposits from tributaries, braided channels, overloading of point bars, or cementing of streambed.

Q6. "YES" if 3 age classes (mature, young, saplings) present for a single species, or young and sapling classes if recruitment & replacement is occurring, or dense matting of herbaceous riparian/wetland plants. "NO" if individual plants. "N/A" if A1 Stream Type.

Q7. Maintenance means recruitment. Is it occurring? "YES" if several different species present (e.g. willows, rushes, sedges). It depends on the elevation and the potential natural community that might be present if all constraints are removed. In some environments, 2 species could be a "YES". Usually "NO" if 1 species present.

Q8. "YES" if sedges, rushes, willows, seep willows, alders, cottonwoods, etc. Don't consider quantity. Do you see any at all?

Q9. A high stream flow event is one that occurs once in 25-30 years. Q9 is similar to Q*, but you are now looking for quantity. "NO" if presence of upland species. "YES" if willows, alder, aspen, birch, cottonwood, sedge, rush, bulrush, and wetland grasses.

Q10. Are the plants healthy and dense? "NO" if yellow leaves, stunted plants, many dead stems and branches, a thin crown, infested with insects, diseased, or grazed down by browsers.

Q11. This is a quantity question. Use 80% cover as a guide. Look for riparian plants, herbaceous cover, salt cedar (tamarisk), seep willows, etc. "NO" if "NO" on Q9. If Q6-Q10 are "NO", this is probably a "NO".

Q12. "N/A" for meadows, desert streams, and probably intermediate elevation streams, or sedge/grass community streams. "YES" if fallen trees. For some locations consider living and dead trees and trees along banks out of the water.

Q13. "YES" if large boulders, roughness of the floodplain, large trees & dense vegetation along stream banks. "NO" if incision & no access of stream to floodplain.

Q14. "YES" if sedge/rush components. Consider potential, height and newness of the point bar. Sandy soils don't hold water well and there may be no potential for revegetation. A1 Stream Type is "N/A".

Q15. "NO" if straight channel, not confined geologically, and channel movement with every high flow event. "YES" if single channel, stable banks (especially on straight segments), & natural deposition.

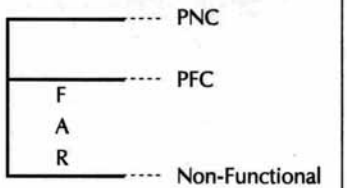
Q16. "NO" if entrenchment, down cutting (some is natural), excessive aggradation, unstable vertical banks. "YES" if streambed is armored with large rock, bedrock, heavy gravel. Don't consider old down cutting. If a bedrock stream then "N/A".

Q17. "NO" if excessive sediment from side drainages, excessive aggradation, mid-channel bars, braiding, or unstable banks. "NO" if Q5 is "NO".

Figure 6. PFC Field Worksheet.

Proper Functioning Condition Worksheet			
Yes	No	N/A	Hydrologic
			1) Flood plain inundated in "relatively frequent" events (1-3 years)
			2) Active/stable beaver dams
			3) Sinuosity, width/depth ratio, and gradient are in balance with the landscape setting (i.e., landform, geology, and bioclimatic region)
			4) Riparian zone is widening or has achieved potential extent.
			5) Upland watershed not contributing to riparian degradation
			Vegetative
			6) Diverse (3) age structure of vegetation (Recruitment for maintenance/recovery)
			7) Diverse composition of vegetation (For maintenance/recovery)
			8) Species present indicate maintenance of riparian soil moisture characteristics
			9) Streambank vegetation is comprised of those plants or plant communities that have root masses capable of withstanding high streamflow events
			10) Riparian plants exhibit high vigor
			11) Adequate vegetative cover present to protect banks and dissipate energy during high flows
			12) Plant communities in the riparian area are an adequate source of coarse and/or large woody debris
			Erosion Deposition
			13) Flood plain and channel characteristics (i.e., rocks, coarse and/or large woody debris) adequate to dissipate energy
			14) Point bars are revegetating
			15) Lateral stream movement is associated with natural sinuosity
			16) System is vertically stable
			17) Stream is in balance with the water and sediment being supplied by the watershed (i.e., no excessive erosion or deposition)

Functional Rating			
☐	Proper Functioning Condition	☐	Functional at risk, downward trend
☐	Functional at risk, upward trend	☐	Non-Functional
☐	Functional at risk, no apparent trend	☐	Unknown



PFC Remarks

Use reverse side for additional comments.

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Figure 7. Stream Ecosystem Monitoring Field Data Sheets.

WQDB Site Number:_____

ADEQ Arizona Department of Environmental Quality

Stream Ecosystem Monitoring Field Data Sheets

Site Code_____ Date_____ (MM/DD/YYYY) Water Sample Time _____

Site Name_____ Field Crew_____

GPS: Latitude_____ Longitude_____

				Entered:	Approved:
Meter Results					
E. coli		cfu/100 ml	TDS		mg/L
Air Temp		°C	Conductivity		µmos/cm
Water Temp		°C	pH		SU
Dissolved Oxygen		mg/L	Turbidity	Average =	NTU
				Signal Avg. =	
% D.O.		%			
Deviations from SOP					

Field Calibrations – Hydrolab and Turbidity Meter				
% D.O. →	Precal Reading =	Barometric Pressure: inches Hg =	X 25.4 =	mm Hg
	Postcal reading =			
Turbidity →	Standard =	Standard solution reading =	% Difference =	

Sample Collection Information				
Sample Method		Quality Control		Bottle Label Identification
Equal Width Increment (EWI)	☐	Equipment Churn Blank	☐	
Modified EWI	☐	Sample Split	☐	
Equal Discharge Increment	☐	Sample Duplicate	☐	
Grab	☐	DI Blank	☐	
If Grab Sample - distance (1/4, 1/3, ½, etc.) from REW = _____; Taken from – run ☐ pool ☐ riffle ☐				
DRY CHANNEL ☐		PONDED WATER – NO FLOW ☐		

Photo Reach Monitoring Log					Prints ☐ Digital ☐
Camera Make:		Model:		DEQ Name:	
☐	Upstream looking downstream	☐	Downstream looking upstream	☐	X-sec @ discharge location LDS ☐
☐	Upstream RB cross-section	☐	Downstream RB cross-section	☐	X-sec @ discharge location LUS ☐
☐	Upstream LB cross-section	☐	Downstream LB cross-section	☐	
☐	Upstream riffle substrate	☐	Downstream riffle substrate	☐	

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Flow Measurements					
Marsh-McBirney Flow Meter					
Measurement from: riffle ☐ run ☐ pool ☐					
Station	Distance from Initial Pt., ft.	Width, ft.	Depth, ft.	Velocity, ft/s	Discharge, cfs
1					
2					
3					
4					
5					
6					
7					
8					
9					
10					
11					
12					
13					
14					
15					
16					
17					
18					
19					
20					
21					
22					
23					
24					
25					
26					
27					
28					
29					
30					
	Total Width	QC width	Average	Average	Total Q

From USGS Gage	
USGS Gage Height =	USGS Discharge =

Float Method Discharge Measurement														
Timed Measurements, seconds														Avg. Time
Width, ft	X	Depth, ft	X	Dist., ft	X	Velocity, fps	X	0.85	=	Discharge, cfs				

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Albion Sample Documentation					
Metals	Composite ____ ; Grab ____ ; Blank ____ ; Duplicate ____ ; Split ____				
Hg	Composite ____ ; Grab ____ ; Blank ____ ; Duplicate ____ ; Split ____				
Deviations from 1669 protocol:	One person only processing the complete sample ☐	Re-used clean box ☐			
Delayed filtration/processing (not at site) ☐	Processed without a clean box, exposed to ambient atmosphere ☐				
No gloves or insufficient clean supplies (e.g. filter clogging, no replacement available) ☐	Other ☐				
Comments:					

E. Coli					
Collection Time ____	Distance (1/4, 1/3, ½, etc) from REW ____	From run ☐; riffle ☐; pool ☐			
Incubation Period is 24 hours for membrane filter technique and 18 hours for colilert technique					
Beginning Incubation Time ____ ; Enumeration Time ____					
Membrane Filter Results					
Dilution, ml	Number of colonies	Dilution Used in Calculation	Quality Control		
		☐		Pass	Fail
		☐	Equipment Blank	☐	☐
		☐	Technique Blank	☐	☐
Calculated Colony Forming Units/100 ml ⇨			Comments:		

Colilert Results			
Sample Number	Number Large Wells Positive	Number Small Wells Positive	Most Probable Number form Table
1			
2			
3			
4			
Average Most Probable Number =		cfu / 100 ml	
Comments			

Field Meter Documentation			
Meter Type	Model	Serial Number	DEQ Name
Hydrolab			
Turbidimeter			
Incubator			
Flow Meter			
Camera Make :			
Chlorine Colorimeter			
Laser Level			

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Field Conditions at Time of Visit			
Flood Evidence within Last Month			
1. None ☐	2. Fresh debris line in channel above bankfull elevation ☐	3. Grasses laid over ☐	
4. Fresh debris line in bushes/trees ☐	5. Other ☐	6. Drought conditions prevailing ☐	Flood width
7. Recent flood event greater than baseflow but less than bankfull ☐		8. Riparian vegetation scoured away ☐	
Weather Conditions :			
Precipitation at sample time : None ☐; Light ☐; Moderate ☐; Heavy ☐; Cloud Cover (%) =			
Previous Precipitation (w/in 24 hrs.) : None ☐; Light ☐; Moderate ☐; Heavy ☐			

Reach Observations	
General appearance in the channel (check all that apply) (GAS)	No refuse visible ☐; Small refuse visible ☐; Small volume refuse common ☐; large volume refuse (tires, carts) rare ☐; large volume refuse common ☐
General appearance along the banks (check all that apply) (GAB)	No refuse visible ☐; Small refuse visible ☐; Small volume refuse common ☐; large volume refuse (tires, carts) rare ☐; large volume refuse common ☐
Water Clarity (WAP)	Clear ☐; Milky ☐; Light brown ☐; Dark brown ☐; Oily sheen ☐; Greenish ☐; Other_____
Water odor (check all that apply) (WOD)	None ☐; Sewage ☐; Chlorine ☐; Fishy ☐; Rotten eggs ☐; Other_____
Appearance at water's edge (check one) (AWE)	No evidence of salt crusts ☐; White crusty deposits rare ☐; Numerous white crusty deposits ☐; banks covered with white crusty deposits ☐
Fish presence	Absent ☐; rare ☐; Common ☐
Crayfish presence	Absent ☐; rare ☐; Common ☐
Sunfish presence	Absent ☐; rare ☐; Common ☐
Leopard frog presence	Absent ☐; Number observed alive _____; Dead_____
Floating leaves or other organic mater (not algae)	Absent ☐; rare ☐; Common ☐
Leaves or other organic matter on streambed	Absent ☐; rare ☐; Common ☐

Organic Debris / Channel Blockages in Active Channel			
☐	1. No organic debris or channel blockages	☐	6. Extensive, large debris dams either continuous or influencing over 50% of channel area. Forces water onto flood plain even with moderate flows. Generally presents a fish migration blockage.
☐	2. Infrequent debris, what's present consists of small, floatable organic debris.	☐	7. Beaver dams. Few and/or infrequent. Spacing allows for normal stream/flow conditions between dams.
☐	3. Moderate frequency, mixture of small to medium size debris affects less than 10% of active channel area.	☐	8. Beaver dams - Frequent. Back water occurs between dams - stream flow velocities reduced between dams.
☐	4. Numerous debris mixture of medium to large sizes - affecting up to 30% of the area of the active channel.	☐	9 .Beaver dams - abandoned where numerous dams have filled in with sediment and are causing channel adjustments of lateral migration, evulsion, and degradation etc.
☐	5. Debris dams of predominantly large material affecting over 30% to 50% the channel area and often occupying the total width of the active channel.	☐	10. Man made structures - diversion dams, low dams, controlled by-pass channels, baffled bed configuration with gabions, etc.

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Flow Regime	
☐	Perennial stream channel. Surface water persists all year long
☐	Intermittent stream channel. One which flows only seasonally or occasionally. Surface source includes springs, snow melt, and flows that reappear along various locations of a reach, then run subterranean (interrupted)
☐	Subterranean stream channel. Flows parallel to and near the surface for various seasons
☐	Ephemeral stream channel. Flows only in response to precipitation
Category	
☐	Seasonal variation in stream flow dominated primarily by snowmelt runoff
☐	Seasonal variation in stream flow dominated primarily by stormflow runoff
☐	Uniform stage and associated stream flow due to spring fed conditions
☐	Regulated stream flow due to diversions, dam releases, dewatering, effluent dominated, etc.
☐	Altered flows due to development, such as urban streams, cut-over watersheds, vegetation conversions (e.g. forested to grassland) that changes flow response to precipitation events

Stream Type Identification						
Walk the reach and flag all likely bankfull indicators. Select the riffle with the best bankfull indicators to collect measurements. A measuring tape, stadia rod, and calculator are sufficient, although a laser level can also be used. Calculate the classification variables and use Rosgen stream type classification chart to identify stream type.						
Watershed Area:		Valley Type				
Predicted Cross-section Area:		☐ I	☐ III	☐ V	☐ VII	☐ IX
Which regional curve used?	☐ Central / Southern ☐ Eastern AZ / New Mexico	☐ II	☐ IV	☐ VI	☐ VIII	☐ X

Measurements for Determining Stream Type			
Measurement	Riffle Cross-section #1	Riffle Cross-section #2	Bankfull Indicators Used
Bankfull Width			☐ Top of point bars
Bankfull Max. Depth			☐ Change in particle size
Correction Factor			☐ Slope break
Bankfull Mean Depth			☐ Vegetation line
Cross-sectional Area			☐ Rock stains
Floodprone Width (2x BKF max depth)			☐ Undercut banks
STREAM TYPE =			☐ Presence of a floodplain at the elevation of incipient flooding

Notes/Comments

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Depositional Features				
Check off the feature that is most appropriate for the reach condition.				
☐ 1. Point Bars			☐ 5. Diagonal Bars	
☐ 2. Point Bars with Few Mid-Channel Bars			☐ 6. Main Channel Branching with Numerous Mid-Bars and Islands	
☐ 3. Numerous Mid-Channel Bars			☐ 7. Side Bars and Mid-Channel Bars with Length Exceeding 2 to 3 times Channel Width	
☐ 4. Side Bars			☐ 8. Delta Bars	☐ 9. NO bars
Illustrations from D. Rosgen, 1996. Applied River Morphology. Wildland Hydrology. Pagosa Springs, CO.				

Segment Habitat Quality				
Segment length equals 2 meander lengths or 20-30 times bankfull width of the stream. Use a minimum 300-feet reach to identify habitat types for large streams or rivers.				
Cobble	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Undercut banks	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Leaf packs	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Root masses	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Macrophyte beds	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Submerged logs / snares	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Sand dominated substrate	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐
Filamentous algae beds	Absent ☐;	rare ☐;	Common ☐;	Abundant ☐

Reach Channel / Habitat Complexity		
Reach length equals 2 meander lengths or 20-30 times bankfull width. Use minimum of 300 foot reach to identify habitat types for large streams		
Habitat	Number of paces	Total
Pool		
Riffle		
Run		
Riffle / Run Ratio =		

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Site Sketch
Include location of riffles, pools, snags, submerged logs, undercut banks, areas of stable cobble habitat, type of bar formations, location and types of riparian vegetation, and areas with cut or eroding banks. Pace off length of eroding banks, length and width of riffles.

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Decision Criteria for Sampling Macroinvertebrates

The target stream habitat for collecting macroinvertebrates must be wadeable, perennial, contain riffle or run habitat, must contain heterogenous substrates, and must be sampled during the spring index period. Spring index period is April - May for warm water streams and May - June for cold water streams. Use the following specific specific decision criteria to determine whether to collect a macroinvertebrate sample. Circle the action taken regarding whether a sample was collected. Where you have found the stream conditions to be inappropriate for macroinvertebrate sampling, record a comment indicating the rationale for not collecting.

Parameter	Condition	Action to Take
Hydrologic Conditions	Baseflow conditions are occurring and it is approximately 4 or more weeks after a bankfull flow event. *	Collect macroinvertebrates
	A bankfull or greater magnitude flow event has occurred within 4 weeks of site visit. Or extreme high flow events have occurred resulting in deep scouring of the streambed and benthic community such that the macroinvertebrate community will not recover within the spring index period.	Do not collect macroinvertebrates
	Extended drought conditions have reduced flow from previously perennial condition to pools only or stagnant wetland habitat.	Do not collect macroinvertebrates
Substrate Type	A substrate consisting of a mixture of some of the following particle sizes is the target condition: boulder, cobble, gravel, sand, clay, silt, bedrock.	Collect macroinvertebrates
	Streams which have substrates dominated (consisting of >50% of that substrate type) by bedrock, travertine, or sand are considered non-target conditions.	Do not collect
Waterbody Type	The target waterbody type is a flowing stream with riffle or run (erosional) habitats present.	Collect macroinvertebrates
	We do not have methods developed for the following waterbody types and are not sampling them at this time: Effluent dependent streams, wetlands, ephemeral streams, lakes, seasonally intermittent streams.	Do not collect

Comments: (Indicate rationale for not collecting macroinvertebrate sample, if different from the above descriptions)

* Identification of bankfull and high flow elevation in the field: Using known watershed area, use appropriate Regional Curve and field bankfull indicators to estimate bankfull elevation. Look for debris lines and other high flow markers as an indicator of the most recent high flow stage. This procedure is explained in more detail and a copy of the regional curves is provided in the ADEQ Habitat Assessment Procedures (2005)

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Biological Sampling and Observations			
Types of Biological Samples and Sample Locations			
Macroinvertebrates	Riffle ☐ and riffle field split ☐; Pool ☐ and pool field split ☐; Edge ☐ and edge field split ☐		
Algae	Diatoms – riffle ☐/ pool ☐; artificial substrate ☐; filamentous riffle ☐; filamentous pool ☐; filamentous composite ☐		
Observations			
Filamentous Algae Covering Streambed throughout the reach	1) <1% 2) 1-25% 3) 26-50% 4) 51-75% 5) 76-100%		
Floating algae (detached clumps/mats) floating downstream	1) <1% 2) 1-25% 3) 26-50% 4) 51-75% 5) 76-100%		
Algal slime on rocks, wood, etc. (not filamentous)	Absent ☐; rare-thin coating ☐; common thick coating ☐		
Comments			
Macrophytes			
Macrophytes covering streambed throughout the reach	1) <1% 2) 1-25% 3) 26-50% 4) 51-75% 5) 76-100%		
Comments			
Identification of Algae (A) and Macrophytes (M)			
☐ A	Cladophora (hairlike feel, long beards)	☐ M	Watercress (Rorippa)
☐ A	Spirogyra (slimy to touch, bright green)	☐ M	Monkey flower (Mimulus, yellow flower)
☐ A	Nostoc (looks like jelly beans or round black to blue colored nodules)	☐ M	Pondweed (Potomogeton, submerged water grass)
☐ A	Blue-greens (blue-green to black in colo, e.g. Oscillatoria, Anabena)	☐ M	Columbine (yellow fower)
☐ M	Stoneworts (feels gritty, looks like a vascular plant, found in upwelling zones)	☐ M	White water buttercup (Ranunculus, white flower)
☐ M	Vaucheria (dark green felt-like mats)	☐ M	Eurasian water milfoil (Myriophyllum)
☐ M	Hydrodictyon (bright green, net forming algae)	☐ M	Hydrilla
☐ M	Praesiola (cold water algae, looks like sea lettuce)		

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Riffle Pebble Count					
For transect method, tally 100-pebbles in riffle habitat only. Measure particles at equal increments across multiple transects within the wetted width of available riffle habitat throughout the reach.					
Size Class	Size Range	Tally	Count	Percent	Cum. %
Silt/Clay *	<0.062				
Sand **	0.063 – 2.0				
Very Fine Gravel	3 – 4				
Fine Gravel	5 – 8				
Medium Gravel	9 – 16				
Coarse Gravel	17 – 32				
Very Course Gravel	33 – 64				
Small Cobble	65 – 96				
Medium Cobble	97 – 128				
Large Cobble	129 – 180				
Very Large Cobble	181 – 256				
Small Boulder	257 – 512				
Medium Boulder	513 – 1024				
Large Boulder	1025 – 2048				
Very Large Boulder	2049 – 4096				
Bedrock	>4097				
	Totals				
Comments:				% fines <2 mm	
				# Size Classes	
				D15	
				D50	
Note: * Silt / clay particles feel slick when rubbing between thumb and forefinger.				D84	
** Sand Particles feel gritty when rubbing between thumb and forefinger.					

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Riffle Embeddedness

Embeddedness measurements are collected concurrently with particle sizes within wetted width of 3 riffle transects. Count sand & fines as 100% embedded, bedrock & travertine as 100% embedded and gravel from a gravel patch as 100% embedded. Embeddedness is taken as a visual estimate. Keep a tally of embeddedness counts within each embeddedness category. Sum the counts and calculate a percentage for each embeddedness category. Then calculate weighted percent embeddedness as indicated. Take a sum of the weighted percents and divide by 100 for an average embeddedness value.

Embeddedness Category	Embeddedness Range (percent)	Tally	Count	Percent	Weighted Percent
Low	0 - 33				(% embed * 17)
Moderate	34 - 66				(% embed * 17)
High	67 - 100				(% embed * 17)
				Avg. % Embed	(sum weighted percents/100

Riffle Geometry

Riffle #	Length	Width	Length / Width ratio
1			
2			
3			
4			
		Average length / width ratio	

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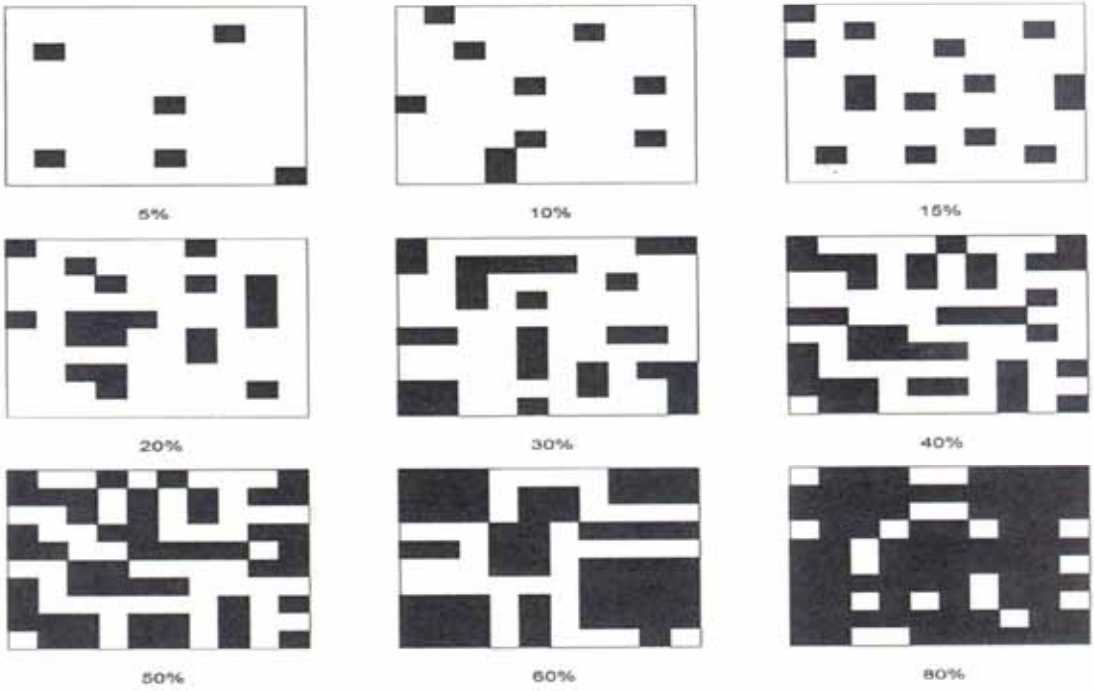
Riparian Vegetation Cover	
Record the percent cover of each vegetation type within the floodplain. Consider each vegetative layer separetly with a score of 0 – 100% for each. The object is to identify what vegetation type is holding the banks and floodplain together.	
Riparian Vegetation Cover	Estimated Percent Cover
Canopy of riparian trees > 15 feet high	
Understory of woody shrubs, saplings, herbs, grasses and forbs – 1.5 to 15 feet high	
Ground cover of woody shrubs, seedlings, herbs, and forbs - < 1.5 feet high	
Barren or bare dirt	



Riparian Association			
Place a check beside the most appropriate association using the riparian species list and elevation.			
☐	Sonoran riparian deciduous forest Cottonwood-Willow & Mesquite located at <3280' elevation	☐	Montane riparian deciduous forest mixed broadleaf species such as Big-tooth Maple; Narrowleaf Cottonwood; Box-elder; SW Choke Cherry; Arizona Alder; Pacific, Coyote, Red, or Bebb's Willow; located at 5740' – 8200' elevation
☐	Interior riparian deciduous forest Cottonwood-Willow & mixed broadleaf species such as Sycamore, Ash, Walnut, Alder, Soapberrry, and Hackberry located at 3280' – 5740' elevation	☐	Actic – Boreal forest Distinctive riparian communities are not present however there are some indicator species such as shrubby Scouler and Bebb's willows, Red Elderberry, Shrubby Cinquefoil, Goose-berry Currant, Raspberry, and Thin-leaf Alder located along streams of subalpine forests and meadows at >8200'.

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Riparian Species											
☐	Alder, Thinleaf	☐	Cinquefoil	☐	Hawthorn, River	☐	Tamarisk	☐	Willow, Peachleaf	☐	Equisetum
☐	Alder, Arizona	☐	Cottonwood, Fremont	☐	Maple, Big-tooth	☐	Walnut, Arizona	☐	Willow, Red	☐	Monkeyflower
☐	Ash, Lowell	☐	Cottonwood, Lanceleaf	☐	Maple, Rocky Mountain	☐	Willow, Arizona	☐	Willow, Scouler	☐	Phragmites
☐	Ash, velvet	☐	Cottonwood, Narrowleaf	☐	Mesquite	☐	Willow, Arroyo	☐	Willow, Seep	☐	Rushes
☐	Birch	☐	Elder, Blueberry	☐	Netleaf Hackberry	☐	Willow, Bebb	☐	Willow, Yewleaf	☐	Sedges
☐	Boxelder	☐	Elder, Mexican	☐	New Mexican Locust	☐	Willow, Bonpland	☐	Willow, Unknown	☐	Sacaton
☐	Buckthorn, Birchleaf	☐	Elderberry	☐	Raspberry	☐	Willow, Coyote	☐	Bamboo	☐	
☐	Buckthorn, California	☐	Elderberry, Desert	☐	Red-osier Dogwood	☐	Willow, Desert	☐	Carex	☐	
☐	Burro bush	☐	Gooseberry	☐	Soapberry	☐	Willow, Gooding	☐	Cattail	☐	
☐	Chokecherry	☐	Hawthorn, Cerro	☐	Sycamore, Arizona	☐	Willow, Pacific	☐	Deer Grass	☐	

Measuring Canopy Density			
	Number of Points Intercepted Along the Transect		
Position	Upper Reach	Mid-Reach	Lower Reach
Right Edge Water			
Middle – Looking Upstream			
Middle – Looking Downstream			
Left Edge Water			
Sum			

Mean Number of Points = Sum of the three columns_____ ÷ 3 =

If stream order <5	Percent Canopy Density = Mean Number of Points • 1.5 =	%
If stream order >5	Percent Canopy Density = Mean Number of Points • 0.75 =	%

Regeneration Potential of Riparian Trees					
	Species, in order of dominance	Mature Trees >16" @ 3 ft height	Young Trees –1 ¼" @ 3 ft. height	Saplings < 1 ¼"	Seedlings New growth
1					
2					
3					
4					
5					

Age Classes of Riparian Tree Species

(Classify according to species present, not just the dominant tree type of that plant association)

☐	Species abundant in 3 age classes	☐	Abundant in 2 age classes	☐	One age class present	☐	No regeneration evident, few mature trees present, no saplings or seedlings, or if present, they are heavily grazed

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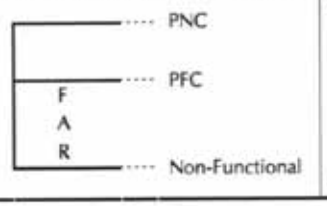
Proper Functioning Condition Worksheet			
Yes	No	N/A	Hydrologic
			1) Flood plain inundated in "relatively frequent" events (1-3 years)
			2) Active/stable beaver dams
			3) Sinuosity, width/depth ratio, and gradient are in balance with the landscape setting (i.e., landform, geology, and bioclimatic region)
			4) Riparian zone is widening or has achieved potential extent.
			5) Upland watershed not contributing to riparian degradation
			Vegetative
			6) Diverse (3) age structure of vegetation (Recruitment for maintenance/recovery)
			7) Diverse composition of vegetation (For maintenance/recovery)
			8) Species present indicate maintenance of riparian soil moisture characteristics
			9) Streambank vegetation is comprised of those plants or plant communities that have root masses capable of withstanding high streamflow events
			10) Riparian plants exhibit high vigor
			11) Adequate vegetative cover present to protect banks and dissipate energy during high flows
			12) Plant communities in the riparian area are an adequate source of coarse and/or large woody debris
			Erosion Deposition
			13) Flood plain and channel characteristics (i.e., rocks, coarse and/or large woody debris) adequate to dissipate energy
			14) Point bars are revegetating
			15) Lateral stream movement is associated with natural sinuosity
			16) System is vertically stable
			17) Stream is in balance with the water and sediment being supplied by the watershed (i.e., no excessive erosion or deposition)

Functional Rating

☐	Proper Functioning Condition	☐	Functional at risk, downward trend
☐	Functional at risk, upward trend	☐	Non-Functional
☐	Functional at risk, no apparent trend	☐	Unknown



PFC Remarks

Use reverse side for additional comments.

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GUIDELINES FOR COMPLETING THE PFC CHECKLIST

General guidance: If 75% or more of stream reach is PFC, classify entire reach as PFC. All "No" answers must have comments in notes section. Answers can go on the line between "Yes" and "No", but consider it a "No" and comment in notes section.

Q1. Instantaneous peak flows don't count. Inundation means to bankfull depth. Bankfull can be identified from top of the point bars, changes in vegetation, topographic break in slope, change in size of bank materials, evidence of an inundation feature such as small benches, exposed root hairs below an intact soil layer indicating exposure to erosive flow, and bank undercuts. "No" if channelization or entrenchment. "N/A" if a "V"-canyon without floodplain development.

Q2. Usually "N/A", but may be applicable at high altitude sites; also, consider the present environment (could they be present).

Q3. Based on the stream type expected & the regional curves, all three features must be present for a "YES". Use bankfull width, not wetted width. "NO" if straightness, excessive sediment, or entrenched channel.

Q4. Widening can mean encroaching on the channel as well as moving toward the terraces. The age of the vegetation in an indicator. "NO" if upland species encroaching on the floodplain or Kentucky bluegrass present. "YES" if recruitment of wetland/riparian species on new landforms. "N/A" if an A1 stream type.

Q5. Need to look at upland ground cover and erosion signs (e.g. plants on pedestals, debris dams around plants, rills, gullies). "NO" if side channel and mid-channel bars, gullies, fan shaped deposits from tributaries, braided channels, overloading of point bars, or cementing of streambed.

Q6. "YES" if 3 age classes (mature, young, saplings) present for a single species, or young and sapling classes if recruitment & replacement is occurring, or dense matting of herbaceous riparian/wetland plants. "NO" if individual plants. "N/A" if A1 Stream Type.

Q7. Maintenance means recruitment. Is it occurring? "YES" if several different species present (e.g. willows, rushes, sedges). It depends on the elevation and the potential natural community that might be present if all constraints are removed. In some environments, 2 species could be a "YES". Usually "NO" if 1 species present.

Q8. "YES" if sedges, rushes, willows, seep willows, alders, cottonwoods, etc. Don't consider quantity. Do you see any at all?

Q9. A high stream flow event is one that occurs once in 25-30 years. Q9 is similar to Q*, but you are now looking for quantity. "NO" if presence of upland species. "YES" if willows, alder, aspen, birch, cottonwood, sedge, rush, bulrush, and wetland grasses.

Q10. Are the plants healthy and dense? "NO" if yellow leaves, stunted plants, many dead stems and branches, a thin crown, infested with insects, diseased, or grazed down by browsers.

Q11. This is a quantity question. Use 80% cover as a guide. Look for riparian plants, herbaceous cover, salt cedar (tamarisk), seep willows, etc. "NO" if "NO" on Q9. If Q6-Q10 are "NO", this is probably a "NO".

Q12. "N/A" for meadows, desert streams, and probably intermediate elevation streams, or sedge/grass community streams. "YES" if fallen trees. For some locations consider living and dead trees and trees along banks out of the water.

Q13. "YES" if large boulders, roughness of the floodplain, large trees & dense vegetation along stream banks. "NO" if incision & no access of stream to floodplain.

Q14. "YES" if sedge/rush components. Consider potential, height and newness of the point bar. Sandy soils don't hold water well and there may be no potential for revegetation. A1 Stream Type is "N/A".

Q15. "NO" if straight channel, not confined geologically, and channel movement with every high flow event. "YES" if single channel, stable banks (especially on straight segments), & natural deposition.

Q16. "NO" if entrenchment, down cutting (some is natural), excessive aggradation, unstable vertical banks. "YES" if streambed is armored with large rock, bedrock, heavy gravel. Don't consider old down cutting. If a bedrock stream then "N/A".

Q17. "NO" if excessive sediment from side drainages, excessive aggradation, mid-channel bars, braiding, or unstable banks. "NO" if Q5 is "NO".

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PFC No Response Sheet – Comments and Notes

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Habitat Assessment Field Data Sheet				
Habitat Parameter	Condition Category			
	Optimal	Sub-optimal	Marginal	Poor
Habitat Quality	Large variety of habitats available for colonization which may include cobble, undercut banks, snags, submerged logs, leaf packs, root masses, macrophyte beds or other organic material.	Moderate variety of habitats which may include cobble, leaf packs, root masses, macrophyte beds or other organic material.	Habitat has minimal variety, substrate dominated by one particle size, may have some cobble, macrophyte beds, or algae beds.	Homogeneous substrate dominated by sand, shallow with uniform velocity, no shade on riffles, may have extensive filamentous algae beds.
Score ⇨	4	3	2	1
Extent of Riffle Habitat	Well developed riffle that is as wide as stream and its length extends 2x the wetted width of the stream.	Riffle is as wide as stream, but is less than 2x stream width; abundance of cobble; boulders and gravel are common.	Reduced riffle area does not extend across entire cross-section and is less than 2x width; gravel or large boulders and bedrock prevalent; cobble present.	Riffles virtually non-existent; sand, gravel, large boulders or bedrock prevalent; cobble lacking.
Score ⇨	4	3	2	1
Embeddedness of Riffles	Gravel, cobble, and boulder particles are 0-25% surrounded by fine sediment (bedrock is 0% embedded).	Gravel, cobble, and boulder particles are 25-50% surrounded by fine sediment.	Gravel, cobble, and boulder particles are 50-75% surrounded by fine sediment.	Gravel, cobble, and boulder particles are more than 75% surrounded by fine sediment (sand is 100% embedded).
Score ⇨	4	3	2	1
Sediment Depostion	Point bars in C type channel maintained, no mid-channel or side bars. No bimodal particle size distribution. No excess sediment in riffles and pools of A, B, or C type channels.	Point bars with few mid-channel bars or side bars in C type channels. No bimodal particle size distribution. Some filling in of pools in A, B, and C type channels.	Numerous mid-channel or diagonal bars in C type channels. Some loss of pool and riffle habitat in A, B, and C type channels. Bimodal distribution may be present with excess fines in the substrate.	Branched or braided C channel with numerous mid-channel bars and islands, some exceeding 2-3x channe width in length. Heavy deposites of finae material evident with bimodal particle distribution. Pools and riffles filled in, with run habitat dominating.
Score ⇨	4	3	2	1
Bank Stability within the active bankfull channel (score each bank)	Banks stable; no evidence of erosion or bank failure; <5% of bank affected.	Banks moderately stable; infrequent, small areas of erosion mostly healed over. 5-30% of bank in reach has areas of erosion.	Banks moderately unstable; 30-60% of bank in reach has areas of erosion; high erosion potential during floods.	Unstable; many eroded areas; "raw" areas frequent along straight sections and bends; 60-100% of bank has erosional scars.
Score_____ Left Bank ⇨	2	1.5	2	0.5
Score_____ Right Bank ⇨	2	1.5	2	.05
Sum of Habitat Category Scores_____ ⇨		Rating Category		
		0 – 7	8 – 14	15 - 20
		☐ Very Impaired	☐ Impaired	☐ Good Condition

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NON-POINT SOURCE CODES

Circle sources directly impacting the site, asterisk sources located in the watershed. Source Group is bolded, Category Code is italicized, and Sub-category Code is regular style font.

Code	Source Category	Code	Source Category	Code	Source Category
1000	**Agriculture** *(Agriculture)*	7350	Upstream impoundment	6600	Hazardous waste storage/disposal
1050	Crop-related sources	7400	Flow regulation/Modification/Diversions	8000	Highway salt storage/use
1100	Non-irrigated crop production	7550	Habitat Modification	8200	Storage tank leaks
1200	Irrigated crop production	7555	Erosion materials from tributaries	8250	Underground storage tank leaks
1300	Specialty crop production	7600	Removal of riparian vegetation	8275	Above ground storage tank
1350	Grazing-related sources	7700	Streambank modification or destabilization	**0100**	**Wastewater** *(Industrial Point Source)*
1400	Pasture grazing - riparian and/or upland	7750	Highway/Road/Bridge-erosion or aggradation	0110	Major industrial point source
1410	Pasture grazing - riparian	7800	Drainage/Filling of wetlands	0120	Minor industrial point source
1420	Pasture grazing - upland	7900	*Marinas and recreational boating*	0200	Municipal point source
1500	Range grazing - riparian and/or upland	7910	Boating with in-water releases	0210	Major municipal point source
1510	Range grazing - riparian	7920	Boating with on-land releases	0220	Minor municipal point source
1520	Range grazing - upland	**5000**	**Mining** *(Resource extraction)*	0230	Package plants (small flows)
1600	*Intensive Animal feeding Operations*	5075	Active Mining operation	*0300*	*Other Wastewater*
1620	Concentrated Animal Feeding Operations point source/permitted)	5100	Surface Mining	0400	Combined system (sewage and stormwater)
1640	Confined animal feeding operations (non-point source	5150	Sand and gravel operations	0500	Collection system failure
1700	*Aquiculture/Fish Hatchery*	5200	Subsurface mining	0900	Sewage lagoons
2000	**Forestry** *(Silviculture)*	5300	Placer mining	0975	Reuse (Effluent to lakes, golf courses, artificial
2100	Harvesting, restoration (residue management)	5400	Dredge mining	6500	Septic systems
2200	Forest management (fertilization, pesticide use)	5500	Petroleum activities	6700	Septage disposal (e.g. from septic tank trucks)
2300	Logging roads	5600	Mill tailings	**8100**	**Other** (Atmospheric deposition)
2500	Clear cutting	5650	Mill or mine tailings	8400	Spills
8610	Wildfires or controlled burns	5700	Mine tailings	8500	Contaminated sediments
3000	**Hydro/Habitat Modification/Runoff** *(Construction)*	5800	Acid mine drainage	8510	
3100	Highway/Road/Bridge construction	5900	Abandoned mining operation	8530	
3200	Land development/Land clearing	5950	Inactive mining operation	8540	
4000	*Urban runoff/Stormwater sewers*	**8700**	**Recreation** (non-boating)	8600	
4100	Non-industrial (NPDES) stormwater runoff	8710	Golf courses	8910	
4200	Industrial (NPDES) stormwater runoff	8720	Camping/Campground recreation	**Other Non-point Source Observations at the site or within the reach**	
4300	Other urban runoff	8730	All terrain vehicles/Off road vehicles/Biking		
4400	Illicit connections to stormwater sewers (dry weather flows)	**6000**	**Storage and Disposal** (Land disposal/Storage)		
4500	Urban Highway/Road/Bridge runoff	6100	Sludge disposal/storage		
4600	*Non-urban runoff/Erosion and sedimentation*	6300	Landfills		
8300	Non-urban (highway/Road/Bridge Runoff/Maintenance)	6350	Inappropriate waste disposal/Wildcat dumping		
7000	*Hydrological modifications*	6400	Industrial land treatment		

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3.17.10 Measuring Percent Canopy Density With the Spherical Densiometer

The Spherical Densiometer optically identifies a series of points in the canopy above the sampling location. The observer records the number of shaded points.

Equipment Required: One modified Model C concave Spherical Densiometer, Field Data Sheets, and writing instrument.

3.17.10.1 Measuring Percent Canopy for Streams Having a Strahler Stream Order of Less than Five

Within the stream reach being assessed, three cross-sections are visually established. Four measurements are taken at each cross-section.

The three cross-sections should represent the reach being evaluated. Typically, one cross-section is located at the uppermost end of the reach, one at mid-reach, and the last at the lowermost end of the reach. At each cross-section, four measurements are taken; 1) at right edge of water facing the right bank, 2) at mid-channel facing upstream, 3) at mid-channel facing downstream, and 4) at left edge of water facing the left bank.

1. At edge of water while standing in the stream, and facing the stream bank, hold the instrument level, away from the body, with the AV@ pointing toward the observer. Position the densitometer twelve inches above the water surface, and twelve inches from the edge of water as shown in Figure 8.

   The observers head reflection should be touching the top of the uppermost grid line. Center the bubble level which is located in the right corner. The densiometer mirror is scribed with interconnecting squares. Count all line intersecting points (recording points) that are surrounded or covered by vegetation (line intercept points) and record that number on the field data sheet in the appropriate location (see example below).

2. Move to the center of the stream and face upstream. Hold the instrument level, 12 inches above the water surface. Repeat as above.

Figure 8. Positioning the Densiometer at stream side.

3. Repeat this procedure facing downstream at mid-channel.

4. Repeat step 1 at the opposite bank.

5. Repeat steps 1 through 4 at the remaining two reach cross-sections.

6. On the field data sheet, sum the tallies for each column. Each column represents a cross-section. Sum the cross-section tallies and divide by 3 to obtain the mean number of points as shown on Table 3.

Table 3. Example of a completed field form.

	Number of points intercepted				
Along Transect	Upper Reach	Mid-Reach	Lower Reach		
Right Edge Water	*7*	*10*	*17*		
Middle - Looking Upstream	*1*	*2*	*0*		
Middle - Looking Downstream	*3*	*2*	*0*		
Left Edge Water	*11*	*9*	*15*	Grand Sum	Mean # of Points
SUM	*22* +	*23* +	*32*	*= 77*	÷ 3 *= 26*
If stream order < 5, Percent Canopy Density = Mean # Points X 1.5 = ____*38*_____%					
If stream order ≥5, Percent Canopy Density = Mean # Points X 0.75 = _____%					

7. If the Strahler stream order is less than 5, multiply the mean number of points by 1.5. This value is the percent canopy density.

3.17.10.2 Measuring Percent Canopy for Streams Having a Strahler Stream Order Equal to or Greater than Five

Use the same procedure described in Section 3C, 3.17.10.1, except eight readings are taken at each cross-section transect, since streams having a higher order number will be wider than those with a smaller order number. Take one reading at each bank and upstream and downstream readings at 1/4, 1/2, and 3/4 distances across the channel. Multiply the total recording points for all eight readings by 0.75. Then deduct one percent if the score is between 30 and 66. Deduct two percent for scores over 66. Make no deductions for scores below 30 (Cowley, 1992).

3.17.10.2.1 Modifying the Model C Concave Spherical Densiometer

Place a narrow strips of black tape at a right angles forming a "V" as shown in Figure 9. This will provide 17 line intersect recording points. The modification improves the measurement of canopy closure (Platts et al, 1987). To facilitate the reading of the mirror surface in the field, place black dots at the intersections of all lines with a Sharpie.

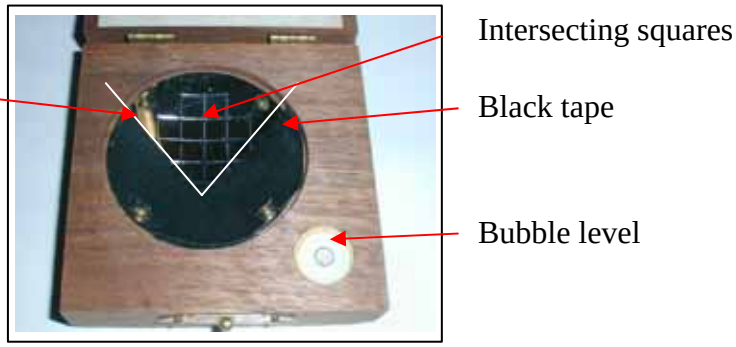

Figure 9. Modified Spherical Densiometer.

3.17.10.3 Literature Cited

Cowley, E.R. 1992. Protocols for classifying, monitoring, and evaluating stream/riparian vegetation on Idaho rangeland streams. Idaho Department of Health and Welfare, Div. Of Environmental Quality, Boise, Idaho.

Platts, W.S., C. Armour, G.D. Minshall, M. Bryant, J.L. Bufford, P. Cuplin, S. Jensen, G.W. Lienkaemper, G.W. Minshall, S.B. Monsen, R.L. Nelson, J.R. Sedell, and J.S. Tuhy. 1987. Methods for evaluating riparian habitats with applications to management. Gen. Tech. Report INT-221. U.S. Department of Agriculture, Forest Service, Intermountain Research Station. Ogden, Utah. 177 pp.

3.17.11 Photo Monitoring

The primary consideration in photo monitoring is the fulfilling of an objective. The objective may vary, depending on the reasons for establishing the site or the results expected over time. Determination of the objective will require some consideration. Generally speaking, monitoring implies the need to determine change. Ask yourself these questions when framing a shot: What will this picture demonstrate? What am I trying to show with this photo? Why is this photograph important? What is appealing about the shot? Does it capture a representative view of the site? (Hall, 2001).

Photos are taken at each visit to a FSN sampling site. Based on the desired objectives, the photo should provide a representative view of that site. The minimum number of photos is two: looking upstream from below the sample point and looking downstream from above the sample point. However, the taking of additional photos is encouraged. Document the sampling event, any changes from the last visit, outgrowths of filamentous algae on the stream bed, channel obstructions, man-made channel alterations or disturbances, floodplain debris, trash, sediment deposition features, point bars, bank erosion, head cuts, streambed particles, riparian community, wetlands community, bank particle composition, etc. The objective is to fully document the condition of the site and photos are ideal for this purpose.

Equipment Required: Either a digital camera or a 35 mm single lens reflex camera with color print film.

3.17.11.1 Procedure

Photographic documentation of sample sites is an essential first step in photo monitoring. At the first visit to a sample site, establish permanent photo points for the upstream/downstream pictures. The preferred type of photo points is a distinctive landmark such as a large tree or boulder. If such a naturally occurring landmark is unavailable at a given site, photo points should be marked in an unobtrusive manner (e.g. small pieces of flagging or rock cairn). Photo points should be described in detail on the field form at the first visit to the site and then recorded in the site files as a permanent part of the file. Photos of the photo points are recommended. If the sample site is not easily found, take photographs of the route to the site. The photos should feature points easily recognized and any changes in direction en route to the site that will enable a new visitor to navigate to the site.

Photos should be taken to include the sample point with a person framed within the photo to show scale. If the stream channel has been altered since the last site visit, additional photos

should be taken. Site alteration may include recent flood evidence, channel scour, sediment deposition, construction or man-made alterations in the floodplain or channel, or other biological or ecological changes that warrant documentation. All photos taken at a site should have the photo number recorded on the FSN Field Data Sheet form with the description of the photo (e.g. looking upstream, looking downstream).

Any photos taken at a site, whether print film or digital, should immediately be printed upon return to the office.

Print Film

On the back of each photograph, the scene should be fully identified with the site identification code, the date the photo was taken, and photo description (e.g. looking downstream, erosion along right bank, cottonwood-willow community), and any applicable notes. Photos should be placed in holders and placed into the respective site file.

Digital Photos

Digital photos should be printed on non-acidic picture quality paper. Color prints are preferable. It is suggested that the photos be inserted into a Word or Word Perfect document. Underneath each photo print the site description, site ID, photo description, and date of photograph. The file name attached to the digital photo should include the camera assigned number, site ID, photo description, and date. The suggested format for a digital file code is Camera Number (DSCN0134), Site ID (UGGLR205_38), Description Code (LDS), and Date (10 May 04). The JPEG file extension is preferred. Common photo description codes include LDS (looking downstream), LUS (looking upstream), and XSEC (cross-section). Each site file should contain a compact disk containing all digital photographs.

3.17.11.2 Literature Cited

Hall, F. 2001. Photo point monitoring handbook: part A – field procedures. Gen. Tech. Rep. PNW-GTR-526. Portland, OR: U.S. Department of Agriculture, Forest Service, Pacific Northwest Research Station.

3.18 Measuring Flow

Stream discharge (Q) is the volume of water passing through a cross-sectional area per unit of time. As such, discharge is expressed in terms of volume per unit of time; examples might include cubic feet per second, gallons per minute, millions of gallons per day, cubic meters per second, etc. Different types of discharge measurement methods may require the use or application of different units of measurement. Flows measured by gauging a cross-section are typically reported in cubic feet per second; flows measured volumetrically are recorded in units of gallons per minute or gallons per second. All such measurements, however, are converted to their CFS (cubic feet per second) equivalent for data repository storage.

3.18.1 Instantaneous Discharge with Flow Meter

Instantaneous discharge with a flow meter is calculated as the velocity (V) in feet per second multiplied by cross-sectional area (A) in square feet. For metered measurements, cross-sectional area is determined by stringing a graduated tape (1/10 ft. increments) across the channel to measure distance at cross-section stations where depth and velocity are measured. Depth of water is measured with a top setting rod (Figure 10) having 1/10 foot increments. Area is depth multiplied by width in small increments (Harrelson et al., 1994). A Marsh-McBirney Flow Meter is used to measure velocity and depth at a pre-determined position in the channel. Select a location in the stream channel that will provide a representative measurement of the entire flow. Do not select a location with a split channel, on a meander, or one with an obstruction immediately upstream from the measurement location.

3.18.1.1 Field Procedure

Equipment required: Marsh McBirney Model 2000 flow meter with sensor and cable, top-setting wading rod with one-tenth foot gradations, two tent pegs, a fiberglass measuring tape with one-tenth foot gradations, and an instantaneous discharge data sheet.

1. Extend the fiberglass tape across the channel from bank-to-bank and perpendicular to the flow. Each end of the tape should be tied to a tent peg firmly secured in the earth. If a bush or some other firmly anchored structure is available, it may also be used. After the tape has been tied to the tent pegs, the tape should be taut with as little sag as possible. If the channel is wide and the wind is blowing, tie strips of flagging on the tape to keep it from whipping.

2. Attach the meter sensor to the top-setting wading rod (Figure 10), place the sensor in the flow, turn the meter on, and check the reporting units. The meter should be set for reading flow in feet per second.

3. The meter can be set to average flows over a set period of time. To set the fixed point average, press the ⬆ and ⬇ keys simultaneously until the display shows the letters

Fixed Point Averaging (FPA). Press ⬆ or ⬇ keys until the FPA increment is set to 10 seconds. Wait until the display automatically switches back to velocity.

4. The observer taking the measurements should move to one edge of the channel, for example the right edge of water (REW), as determined by facing downstream. Position one eye directly above the tape at the exact location where the water and the bank interface, and call out the measurement to the Recorder, for example the reading is 0.8 feet. This figure should be recorded under DIST on the first line together with the abbreviation REW (right edge of water) (Table 4).

    - ❖ The convention of labeling banks relative to a downstream orientation is consistently applied to all bank determinations. There is no convention regarding which bank should serve as the beginning of the transect, as this may vary from site to site depending on access conditions. Neither is there any convention stating that the tape must have its lower end values starting from one or the other bank; calculation after the measurement is concluded proceeds in the same fashion whether values on the tape are increasing or decreasing. For this example, it is arbitrarily assigned that the measurement proceeds with increasing tape stations from the REW. Any of the three other permutations (decreasing from LEW, increasing from LEW, decreasing from REW) is also acceptable

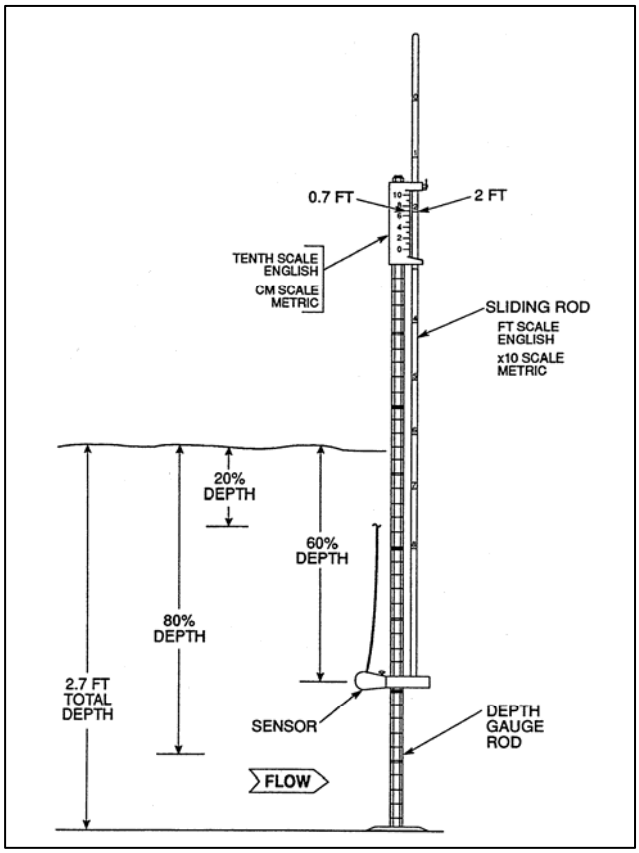

Figure 10. Top-setting wading rod.

5. The observer should move to the LEW, and read the measurement off the tape at the water-bank interface and calculate the width of channel. Divide the width by 20 and round to the nearest whole number. USGS recommends that no more than 5% of the stream discharge be represented in each sub-sectional area of the cross-section; in practice, this usually equates to 20 to 25 measurements across the width of the stream. For example, if the channel is 58 feet wide, 58 ÷ 20 = 2.5; round up to 3.0. Take flow measurements every 3 feet.

   For narrow channels the minimum spacing is 0.3 feet.

6. For our example, to begin the process of measuring discharge, the observer moves to the nearest foot mark beyond 0.8 feet and inserts the top-setting wading rod into the flow. Therefore, the first measurement station on the tape is at 1.0 foot and is recorded under DIST.

7. Observer positions the wading rod vertically with the sensor pointed upstream into the flow. Determine the depth of water from the rod depth gauge to the nearest 10th of a foot. The exception to this would be if the water level is at the half way mark between 0.4 feet and 0.5 feet on the depth gauge (hexagonal rod); in this case the reading is 0.45 feet. If the water level is between 0.4 and 0.45 or between 0.45 and 0.5, round off to the nearest 1/10 foot increment. The depth measurement is recorded under DEPTH.

The one-point method, as described in Step #7, is used when the water depth is less than 2.5 feet. For depths of 2.5 feet or greater, the two-point method for measuring flow is used. Whereas the one-point method measures flow at 0.6D (Depth), the two-point method requires an average of flow measurements taken at 0.2D and 0.8D (Corbett, 1962). If the depth is 2 feet, the single-point method requires a measurement at 1.2 feet (2 ft. x 0.6). If the depth is 3 feet, the two-point method requires readings at 0.7 feet. (3.5 ft. X 0.2) and 2.8 feet (3.5 ft. X 0.8). Record the average of the two velocities for that station.

- The top-setting wading rod consists of two rods; round and hexagonal. The depth gauge rod (hexagonal shaped) is 4.5 feet long, and the round rod is the sliding rod, 3 inches longer than the depth gauge rod. The sliding rod has grooves spaced about every 4.75 inches apart. At the top of the hexagonal rod is a lever called the "sliding rod lock" (Figure 10). The top of the depth gauge rod is graduated into 10 divisions numbered in even numbers.

8. The movable scale on the sliding rod represents the foot value of the water depth; the stationary scale on the hexagonal rod represents tenths of a foot. Any depth value setting must incorporate both of these scales by setting the station depth's value on the movable scale opposite the station depth's tenths value on the stationary scale. For our example, this would entail aligning the zero mark on the sliding rod between four and five on the stationary scale to represent 0.45 feet.

Table 4. Completed discharge measurement form.

INSTANTANEOUS DISCHARGE						
#	DIST	WIDTH	DEPTH	VEL	Area	Q
1	0.8	0.1	0	REW	0	0
2	1.0	1.6	0.45	0.22	0.72	0.16
3	4.0	3	0.59	0.33	1.77	0.58
4	7.0	3	0.98	0.56	2.94	1.65
5	10.0	3	1.21	0.89	3.63	3.23
6	13.0	3	1.53	1.25	4.59	5.74
7	16.0	3	1.65	1.66	4.95	8.22
8	19.0	3	1.48	2.11	4.44	9.34
9	22.0	3	1.5	2.46	4.50	11.07
10	25.0	3	1.5	2.35	4.50	10.58
11	28.0	3	1.53	2.56	4.59	11.75
12	31.0	3	1.53	2.54	4.59	11.66
13	34.0	3	1.65	2.12	4.59	10.49
14	37.0	3	1.68	2.29	5.04	11.54
15	40.0	3	1.78	2.37	5.34	12.66
16	43.0	3	1.67	2.18	5.01	10.92
17	46.0	3	1.23	2.56	3.69	9.45
18	49.0	3	1.04	1.89	3.12	5.90
19	52.0	3	0.85	1.01	2.55	2.58
20	55.0	3	0.3	0.89	0.90	0.80
21	58.0	1.5	0	LEW	0	0
22						
23						
Total Dist.		QC summed Total	Avg. Depth	Avg. Velocity	Total Area	Total Discharge
57.2		57.2	1.15	1.54	71.82	138

- ❖ The topmost groove on the sliding rod represents the depth distance from zero to one foot. The second groove represents the depth distance from 1 foot to 2 feet, and so on. If depth of water is less than or equal to 1 foot, use the topmost groove. If the depth is between 1 and 2 feet, use the second groove, etc.

9. With the sensor submerged, allow the sensor to stabilize, and then wait for the timing slide on the meter display to cycle one complete fixed point averaging cycle before reading the flow measurement. Make a mental note of the velocity reading. After the meter has cycled through a third cycle, record the reading if it is within 10% of the second reading. If the third reading is not within 10% of the second reading, continue to cycle through successive readings, comparing each one with the previous reading until two successive readings are within 10% of each other. The Recorder records the last observed reading under VEL on the field data sheet. If the readings are unstable and highly variable, take three successive readings and have the Recorder calculate the average.

10. In step 5, the measuring stations on the tape were determined to be at 3 foot intervals. After the first measurement at the one foot station on the tape, the next measurement is at the 4 foot station. Steps 7-9 are repeated until the left edge of water is reached.

11. At left edge of water, the Observer positions one eye directly above the tape at the exact location where the water and the ground interface. This distance is recorded under DIST and the abbreviation LEW next to it.

Refer to Section 5.13 for calculating discharge.

3.18.1.2 Meter Error Messages

The displaying of errors alerts the user of possible problems with either the meter or the process. Errors can be displayed as messages or numerical codes. There are three error messages and five numerical codes.

- ❖ With the exception of Err 2, error codes freeze the display. Turn the unit OFF, and then back ON to clear the display. If the error message persists, return the meter to the manufacturer for maintenance.

Low Bat - Indicates low battery voltage. Replace the batteries with two D cells. This operation will require a screwdriver or coin to open the battery compartment.

Noise - Indicates electrical noise is present in the flow. The noise flag usually comes on for a few seconds right after the sensor is placed in the water. This is normal. If the noise level is too high to get accurate readings, the screen will blank out.

Con Lost - Indicates sensor electrodes are out of the water or have become coated with oil or grease. After a few minutes, the unit will turn itself OFF. If the electrodes are coated, clean the sensor with a mild soap and a soft cloth.

Numbered Error Messages

Error #1 - Problem with sensor drive circuit. Check sensor disconnect.

Error #2 - Memory full error. Memory must be cleared before another reading can be stored.

Error #3 - Incorrect zero adjust start sequence. Reinitiate zero start sequence.

Error #4 - Zero offset is greater than the zero adjust range. Repeat the zero adjust procedure. If error is still displayed, unit needs servicing.

Error #5 - Electroconductivity lost or noises detected during zero adjust. Usually caused by the sensor being out of the water.

3.18.1.3 Key Summary

The function keys can be operated as single key functions or two-key functions.

One Key Function

ON/C - Turns Unit ON. Clears the display and restarts the meter.

OFF - Turns Unit OFF.

⬆ - Increments FPA (fixed point averaging), TC (Time Constant), and Memory Location.

⬇ - Decrements FPA, TC, and Memory Location.

RCL - Alternates between Recall and Real-Time Operating Modes.

STO - Stores Values in Memory.

Two Key Function

ON/C + OFF - Change Units, Turns Beeper ON/OFF.

⬆ + ⬇ - Alternates between FPA (fixed point averaging) and rC (Time Constant) Filtering.

ON/C + STO – Memory may be cleared from either the real-time or recall mode by pressing ON/C and STO simultaneously.

RCL + STO - Initiates zero adjust sequence. Zero stability is ± 0.05 ft/sec.

3.18.2 Float Method

The float method is a simple means of estimating discharge.

Equipment Required: A measuring tape, a timer (i.e. digital watch), and 5-10 floats. For floats, use orange peel, a water-soaked block of wood, or other natural material that sinks at least halfway into the water, is visible from shore, not influenced by the wind, and is expendable and non-polluting.

3.18.2.1 Float Method Procedure

1. Measure and mark two points along the length of the channel, at least two to three channel widths apart, at the channel cross-section.

2. Two observers are best. One tosses the float into the channel above the marker and calls out when it crosses the upstream point. Toss each float a different distance from the bank to obtain an average of velocities.

3. The downstream observer starts the timer, sighting across the stream from the lower point. When the float passes, stop the watch and record the time. Repeat the procedure 5 to 10 times. Determine the mean surface velocity. A coefficient of 0.85 is commonly used to convert the velocity of a surface float to mean velocity in the vertical (http://wwwrcamnl.wr.usgs.gov/sws/fieldmethods/).

4. Using the previously measured cross-sectional area, multiply velocity times area to find discharge (Q= VA). Record it on a data sheet with date, time, etc. If the cross-sectional area cannot be obtained because of unsafe wading conditions, record the velocity. If it is possible to return to the site under favorable conditions, measure the cross-sectional area and compute the estimated Q (Harrelson et al, 1994).

3.18.3 U.S.G.S. Staff Gage

At sites located near or next to a U.S.G.S. gauging station, a discharge measurement can be made by recording the time of day and the staff gauge height. On the U.S.G.S. web page http://waterdata.usgs.gov/nwis/ , find the appropriate gauging station and determine the discharge from the table provided and record on the field data sheet for that site.

3.18.4 Volumetric Measurement

The volumetric measurement of discharge is only applicable to small discharges, but it is the most accurate method of measuring such flows. In this method the hydrographer observes the time required to fill a container of known capacity, or the time required to partly fill a calibrated container to a known volume.

Volumetric measurements are usually made where the flow is concentrated in a narrow stream, or can be so concentrated, so that all the flow may be diverted into a container (Examples or possible locations include: V-notch weir, artificial control where all the flow is confined to a notch or to a narrow width of catenary-shaped weir crest, and a cross section of natural channel where a temporary earth dam can be built over a pipe of small diameter, through which the entire flow is diverted).

Volumetric measurements have also been made when no other type of measurement is feasible, as for example on small streams composed of a series of pools behind broad-crested weirs. At low flows the depth of water on the weir crest is too shallow to be measured by current meter, and the velocity in the pools is too slow for such measurement. Discharge is measured by taking timed samples of flow sufficient to fill a container of known volume held along the downstream face of a control.

3.18.5 Source Material

http://wwwrcamnl.wr.usgs.gov/sws/ fieldmethods/.

Corbett, D. M. 1962. Stream-gaging procedure. U.S. Geological Survey, Water-Supply Paper 888, Washington, DC.

Marsh-McBirney, Inc. 1990. MMI Model 2000 Flo-Mate Portable Water Flowmeter Instruction Manual. Frederick, MD.

3.18.6 Literature Cited

Harrelson, C.C., C.L. Rawlins, and J.P. Potyondy. 1994. Stream channel reference sites: an illustrated guide to field technique. U.S.D.A., Forest Service, Rocky Mountain Forest and Range Experiment Station. Gen. Tech. Rep. RM-245, Ft. Collins, CO.

3.19 Equipment and Personnel Decontamination Procedures

The purpose of this procedure is to provide a description of methods for preventing or reducing cross-contamination and description methods that will protect the health and safety of site personnel.

3.19.1 Field Equipment Decontamination

All reusable sampling equipment should be properly cleaned before going into the field. When sampling and field activities are completed, sampling equipment that has come in contact with contaminated or suspect water should be decontaminated before leaving the site. The purpose of the field decontamination procedures is to protect the field equipment from cross contamination. The field decontamination procedure should provide a quick method of removing most sample residues from the equipment.

3.19.1.1 Sampling Equipment and Containers

Surface water with known or suspected contamination by biological organisms should be decontaminated with a mixture of bleach and water. A specified technique is not specified, but two suggestions for decontaminating equipment are presented.

- The bleach/water mixture is contained in a bucket. Equipment is placed into the bucket and thoroughly brushed with a long handle bottle brush and then rinsed with either tap or distilled water. For equipment that is too large to fit into the bucket, hold the piece over the bucket and proceed as described above. For churn splitters, pour some of the bleach/water mixture into the container and proceed as described above.
- A bleach/water mixture is contained in a spray can. The piece of equipment is held over a bucket while it is being sprayed with the mixture. The bucket collects the drippings from the equipment and disposed of properly. The decontaminated equipment is then rinsed with either tap or distilled water. The outside and inside surface of churn splitters is sprayed and rinsed with either tap or distilled water. The bleach/water residue is collected in the disposal bucket for later disposal.

3.19.2 Sampling Safety

Proper safety precautions must always be observed when sampling. In all cases, the person collecting a sample must be aware of the potential dangers from the material to be sampled and from the site location. Background site information should be obtained about the sample stream. This information will be helpful in deciding the extent of sampling safety precautions to be observed. If background information does not exist, use the best available precautions.

The following general safety rules and practices should be implemented whenever sampling:

1. The best way to simplify decontamination is to minimize contact with contaminants.

2. Each sample should be handled with care to minimize the risk of personal exposure.

3. If special handling of the sample is appropriate, such as safety glasses/goggles, hard hats, boots, gloves, and respirators, must be worn in areas where hazardous conditions are suspected. In addition, eye and hand protection should be worn when handling acidic, caustic (e.g. bleach), or other hazardous liquids (including preservative chemicals, such as formaldehyde).

3.19.2.1 Sample Handling

The portion of sample to be analyzed is transferred to the appropriate containers. It may be necessary to transfer the sample into two or more containers if multiple analyses will be requested and they require different preservatives.

Preservatives should be added to water samples as soon as practical after collection (in general, within 15 minutes). As each portion of sample is transferred to the various containers, it may be necessary to stir or agitate the samples so that suspended matter remains evenly distributed.

The receiving laboratory must be notified of preservatives used in the samples and the extent of the potentially hazardous material in the water sample.

SECTION 4

POST-TRIP PROCEDURES

TABLE OF CONTENTS

4.00 Post-Trip Procedures

4.10 Maintenance of Hydrolab Units

4.10.1 Specific Electrical Conductivity

On older sonde units and on some older minisondes, the EC probe consists of a set of nickel electrodes covered by a hard plastic conductivity cell block. When working with this type of probe, first remove the conductivity cell block with a screwdriver and then remove the rubber o-rings from the electrodes.

Polish all exposed surfaces of the nickel electrodes including the ends with the emery strips (#400 wet/dry sandpaper) found in the maintenance kit.

Do not to touch the other probes with the sandpaper (especially the pH and redox); protect them with wet cotton.

Rinse with deionized water and then replace the o-rings and the conductivity cell block.

Newer units will probably be equipped with a four electrode graphite conductivity sensor. This type of sensor should never be cleaned with abrasive material of any type. When cleaning the electrodes, use cotton and a solvent such as methanol or isopropyl alcohol to remove any residue that may present. The sensor can also be cleaned using warm soapy water. Rinse with deionized water when finished.

4.10.2 pH

1. Clean the glass probe with a cottonball or Q-tip saturated with methanol or isopropyl alcohol. Rinse the probe thoroughly with deionized water.

2. If the response time of the pH probe does not produce 95% of the calibration reading within 90 seconds, then shock the probe with a weak HCl solution.

3. Wrap the probe with cotton saturated with 0.1M HCl and leave for 5 minutes. Remove the cotton and rinse with deionized water. Soak the probe in pH 7.0 buffer for 10 minutes before calibrating.

4. The reference electrolyte is replaced in the older sonde units by inverting the entire unit and gently pulling off the white cylindrical sleeve. Empty the remaining electrolyte and rinse the sleeve with a small amount of fresh pH reference electrolyte. After rinsing, refill the sleeve to the top with standard pH electrolyte and push sleeve back about half-way onto its mount. At this point, turn the sonde unit so the probes are pointing upwards, allowing any air bubbles in the electrolyte to move up towards the porous junction. Push the sleeve down the rest of the way, purging any air within the chamber and flushing the porous junction on the tip of the sleeve with fresh reference

solution. In most cases the fresh reference solution can be seen coming through the pores in the junction.

In the newer minisonde units, the porous Teflon® junction is located at the top of the probe next to the pH electrode. A screw driver is used to remove it so that the old reference solution can be dispensed and replaced with new solution. Use a syringe to replace the solution in the probe. When the porous junction is replaced, the fresh solution is flushed thru the junction.

5. The porous junction of the electrolyte sleeve can become clogged with impurities over time. A new junction may be required if air bubbles are obstructing the flow of electrolyte to pass through the junction during the process described in step #4.

4.10.3 Dissolved Oxygen

1. To change the DO membrane, remove the DO sensor guard (older sonde units) and o-ring securing the membrane. The membrane should be replaced each time the unit is serviced. The membrane should be replaced if air bubbles are under the membrane, the membrane is damaged, or if the membrane is wrinkled.

2. To replace the DO electrolyte solution, tip the probe until the old solution is dispensed. Slowly refill with the squeeze bottle of KCL electrolyte provided in the kit. Empty the probe a second time. Refill the probe again being careful not to induce any bubbles into the cylinder. Gently tap the cylinder to remove any bubbles and let the unit sit for 5-10 minutes to allow gases to escape. Add a few more drops of solution to form a meniscus if needed.

3. To replace the standard membrane, secure the membrane on one side of the top of the sleeve with the thumb, and with a smooth firm motion, stretch the other end of the membrane over the sensor surface and hold it in place with the index finger. Secure the membrane with the o-ring and trim the excess membrane below the o-ring. There should be no wrinkles in the membrane or bubbles in the electrolyte under the membrane. Avoid overstretching the membrane. Do not touch any of the internal parts of the DO sensor.

4. Test the unit for bubbles by gently tapping the sonde cylinder and by inverting the probe a few times.

5. The DO sensor should be allowed to sit overnight to allow the membrane to relax before calibrating.

4.10.4 Turbidity

1. When the lens in the sensor is coated with impurities, rinse with water directed at the lens to remove deposits.

2. Remove the retainer/guard by unscrewing the black nylon screw.

   Do not remove the lens. The lens can be cleaned in place by wiping with a clean, soft, non-abrasive cloth saturated with rubbing alcohol. After cleaning, rinse the sensor and retainer/guard with water, and reattach the retainer/guard.

3. Frequency of maintenance depends on the rate and type of fouling, and the deployment technique. These are factors which must be evaluated by the operator.

4.10.5 Redox

The platinum band on the redox sensor can be polished with toothpaste and a soft cloth or a fine polishing strip. Rinse and let soak overnight.

The redox probe does not need frequent maintenance. A toxic solution of quinhydrone is needed for calibration. It is recommended that the unit be sent to the Hydrolab® servicing department for this procedure periodically.

4.20 Hydrolab Post Trip Equipment Check

After returning from the field, field sampling staff will once again check, clean, and repair all field meters and related sampling equipment to ensure that they were in good working condition for the duration of the field trip. All post-trip checks, cleaning, and repairs made to field meters will be noted in the individual instrument log books and the calibration worksheets (checks only) (Figure 1) which are kept in instrument folders near the SWMSU offices. This recording will be made immediately upon completion of the task.

If the field meter post trip checks are out of the ranges identified below, the following steps must be done:

1. If the post-trip check results in comparisons that are within the limits identified below, data is acceptable for all purposes.

2. If the post-trip check results in readings that deviated outside of the ranges identified below, the data will not be acceptable for assessment or compliance purposes and should be excluded or discarded from the dataset. Be sure to make a note in the field notes that such data was rejected.

3. Determine when, if possible, any problems occurred in the field. If this can be determined, the readings taken before the problem occurred may be acceptable. Any values obtained after the problem occurred should be discarded.

4. If the problem resulted in large differences between meter readings and stated solution values, try to isolate the problem and correct it as outlined in the Pre-Trip Equipment Calibration Procedures (Section 2.10). If the problem cannot be corrected satisfactorily, tag the meter for repair.

A. pH readings for all buffer calibration endpoints shall be within 0.3 standard units of the post-calibration value achieved in the pre-trip equipment preparation without additional modification. Hydrolab instrument specifications specify an instrument tolerance of +/- 0.2 SU, and and additional 0.1 SU is permitted for minor environmental differences (temperature, etc.).

B. EC readings shall be within 10% of pre-trip post-calibration values without modification.

C. Dissolved oxygen saturation values will be evaluated site by site according to calibration performance in the field. Readings will be evaluated after site calibration by stabilization time (should be within 2 minutes) and adherence to a full 100% calibration.

A calibration worksheet has been developed to help record instrument performance based on these criteria. See the enclosed worksheet.

Figure 1. Field meter post-trip equipment check worksheet.

HYDROLAB CALIBRATION RECORDS			
Trip Name:		Date:	
PRE-TRIP MEASUREMENTS			
Conductivity Measurements			
EC PreCal Reading:	EC PostCal Reading:	Calibrate: Y ☐ N ☐	
pH Measurements			
pH 7.00 Buffer PreCal Reading:		pH 7.00 Buffer PostCal Reading:	
pH 10.00 Buffer PreCal Reading:		pH 10.00 Buffer PostCal Reading:	
pH Calibrate: Y ☐ N ☐			
Dissolved Oxygen Measurements			
D.O. % Saturation Before:		D.O. % Saturation After:	
Pre-Trip Battery Voltage:			
Reference Specifications/Tables for Evaluation of Post-Trip Readings			
pH Standard: Absolute Difference no Greater than 0.3 SU, Pre- to Post-Trip			Pass ☐ Fail ☐
EC Comparison Table: Assuming a Test Value of 1450 usm/cm			
Difference: Pre- to Post-Trip	% Difference	Difference: Pre- to Post-Trip	% Difference
15 ☐	1	60 ☐	4.1
30 ☐	2.1	75 ☐	5.2
45 ☐	3.1	145 ☐	10
If Greater than 145usm/cm, Record as >10% ☐			

CALIBRATION RECORDS				
Trip Name:			Date:	
Hydrolab & Surveyor Name/Model:				
Post-Trip				
EC Post-Trip Reading:		Difference from Pre-Trip Reading:		
Percent Difference:	Values Accepted: ☐	Values Rejected: ☐		
pH 7.00 Buffer Post-Trip Reading:		Diff. from Pre-Trip PostCal Reading:		
pH 10.00 Buffer Post-Trip Reading:		Diff. from Pre-Trip PostCal Reading:		
Values Accepted: ☐		Values Rejected: ☐		
D.O. Percent Saturation				
Site ID	Calibrated to 100%	Stabilized within 2 minutes	Value Accepted?	Value Rejected?

SECTION 5

DATA MANAGEMENT

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5.00 Data Management

5.10 Preparing Site Files

Information and data from Surface Water Monitoring and Standards Unit (SWMSU) monitoring sites are contained in individual files for each sampling site. Individual site files are housed on the 5th Floor at the central Phoenix office. Lateral file cabinets are dedicated to housing the site files.

5.10.1 Site File Contents

Each site file contains the following information starting with the inside cover of the file.

Site Information sheet

- The Site Information sheet can be copied from Water Quality Data Base. It will have information regarding the site, such as the name of the site, DEQ Database number, site location, and, if applicable, a signed written permission form from the property owner to enter the property and collect samples. Ownership of the land should not be assumed to be public land and should be investigate during the reconnaissance stage of site selection.

Road Log and Map

- A detail road log should be written during the reconnaissance stage of site selection. A photocopy of a USGS map with the site location highlighted is recommended. In addition, any special comments such as locked gates, 4x4 roads, or other access issues need to be attached to the log.

Correspondence/Exceedances

- Any miscellaneous correspondence with the property owner
- All correspondence on Surface Water Quality Standards exceedances that apply to the site
- Any contractual or sampling agreements pertaining to the site
- All internal and external correspondence regarding the site

Photographs

- Archive-quality slides, prints, or digital images of site taken at each site visit. At least 2 photographs taken at each site; one looking upstream and one looking downstream from the sampling site (see Section 3C, 3.23 for Photo Monitoring procedures).

Macroinvertebrate Data

- ❖ All field and taxonomy laboratory data sheets pertaining to macroinvertebrate collections (see Section 3A, 3.15.1 for macroinvertebrate procedures)

Stream Channel Physical Assessments

- ❖ Habitat Assessment Field Data Sheet for Cold and Warm Water Streams, Stream Ecosystem Monitoring Data Sheets, and any other data sheets describing the physical condition of the stream channel

Field Notes

- ❖ Field data sheets and field measurements from each site visit
- ❖ A flow calculation sheet
- ❖ Any historic copies of field notes from each site visit
- ❖ Sketches or diagrams representing the site

.

Water Quality Chemical Data

- ❖ Located on inside back cover of site file
- ❖ All analytical laboratory data from each site visit
- ❖ Water Quality Standards Exceedence and QA/QC checkoff sheet

5.10.2 Filing System

Site files are filed in the lateral file cabinets by hydrologic basin. There are ten major basins:

1. Colorado-Grand canyon (CG)
2. Colorado-Lower Gila (CL)
3. Little Colorado River Basin (LC)
4. Verde River Basin (VR)
5. Salt River Basin (SR)
6. Upper Gila River Basin (UG)
7. Middle Gila River Basin (MG)
8. San Pedro River Basin (SP)
9. Santa Cruz River Basin (SC)
10. Bill Williams River Basin (BW)

Site files are sorted within each file cabinet drawer in alphabetical order by a 2 character basin code (e.g. UG for Upper Gila River) and a 3-character stream identification code (e.g. ETK). See Section 1.10.2 for assignation of stream codes.

Example 1: In the Upper Gila River Basin cabinet the following would be found:

Name of Stream	Basin/Stream/& River Mile Codes
East Turkey Creek	UG ETK 007.70
Frye Canyon Creek	UG FRY 007.00
Gila River above Bonita Creek	UG GLR 190.39

Multiple sites on the same stream with identical 3-character stream codes are filed in numeric order by monitoring point (from mouth of stream to headwaters).

Example 2: Four sampling sites have been established on Eagle Creek in the Upper Gila River Basin. The four sites have the same basin and 3-digit stream codes (UG EAG). They are then sorted by the river mile code. The river mile code represents the location of the site in miles from its mouth to headwaters.

1. Eagle Creek above Gila River UG EAG 000.05
2. Eagle Creek below Gold Gulch UG EAG 006.05
3. Eagle Creek above Sheep Wash UG EAG 023.24
4. Eagle Creek above Honeymoon Campground UG EAG 035.99

In example 1 and 2, the site codes were presented with spaces between the basin code and stream code and between stream code and the river miles designation for ease of interpretation. Typically, when the code is written, there are no spaces (e.g. UGEAG035.99).

5.10.3 Site File Security

Site files contain unique and irreplaceable information, and thus are kept in locked lateral file cabinets to provide security against loss. The lateral file cabinets are located on the west wall on the 5th Floor. Access to the SWMSU site file cabinets is limited to SWMSU personnel and all others by special arrangement. SWMSU staff is responsible for site files that are removed to their work areas. They must be returned and properly filed in alphanumeric order after usage.

It is recommended that site files be returned to the file cabinet after one week. If files need to be used for a longer period of time, an "OUT CARD" should be placed in the file with the user's name and cubicle number.

The SWMSU manager is the primary contact for access to SWMSU's locked site file cabinets and has custody of the cabinet keys.

5.10.4 Signing Out SWMSU Site Files by Non-SWMSU Staff

Non-SWMUSU staff may check out site files to their work areas for an 8-hour work day, but the files must not be removed from the 5th Floor and must be returned by 5:00 p.m. Files may be signed out from the Hydrologic Support and Assessment Section (HSAS) secretary or the

SWMSU manager. An "OUT CARD" must be filled out with the date, name, telephone extension, and cubicle number where the file will be kept during the work day.

5.10.5 Public Review of Site Files

The data and information in SWMSU site files are public records and they may be viewed by members of the public upon request during regular business hours. Members of the public who wish to review a site file must make arrangements with the HSAS secretary or the SWMSU manager. Site files may only be reviewed on the 5th Floor in a supervised area.

5.10.6 Copying Contents of Site Files by the Public

Copies of the contents of site files will be made by the ADEQ Copy Center. Billing for copies is the responsibility of the person making the copy request. Charges for copies for members of the public shall be in accordance with current ADEQ copy policy.

5.11 Data Management Quality Control

The Surface Water Quality Database (SWQDB) Application - Surface Water Module is designed to meet environmental data and information needs of the department and major cooperators (e.g. Arizona Department of Water Resources, Arizona Department of Game and Fish, U.S. Geological Survey) engaged in water quality monitoring activities. The SWQDB provides a comprehensive repository for surface water site characteristics, measurements, observations, and water quality sampling results. Water quality results include the identity and concentration of chemicals detected, and the analytical methods used.

5.11.1 Data Entry and Data Checking

All manual data entry tasks shall be split between two roles: data entry and data checking. Data entry tasks are the responsibility of the field trip lead who subsequently assumes the role of the data processor. The processor is also responsible for giving final approval to the data entered in the database once the checker has looked over the data. Data checking tasks are ideally performed by the person who served as the assistant on the field trip to confirm accurate entry, population with sufficient and complete comments, accuracy of calculations (bacteria counts, flow calculations, etc.), and notation or consideration of QC checks.

Data must be entered and all QA/QC checks need to be done as soon as possible upon receipt of the lab results. Most labs only hold samples for 30 days. Staff should review reports for obvious errors, outliers, and exceedances upon receipt. If an analytical error is suspected and an analysis rerun is desired, this 30-day time limit must be met.

After the data has been entered for a particular sample date and time, the processor will review the QA/QC checks for data validity (Form 1). Problems will be noted and followed up on in a timely manner by the person managing the site.

5.11.1.1 Compilation of Laboratory Reports and Field Sheets

The typical procedure for handling laboratory and field data are:

- Confirm that lab data matches field data (date and time)
- Confirm that all lab data are received and date stamped (including QC samples)
- Confirm that lab data are complete and no tests are missing
- If data are missing, contact lab for missing data, note the discrepancy on the lab report, and place in a pending file. It is recommended that hard-copy results be held from filing in the site file until the updated report is received.
- Proceed with data entry while waiting for missing data

Form 1. Water Quality Sample QA/QC Checkoff Sheet.

Site ID:	WQDB Code:

WATER QUALITY SAMPLE QA/QC CHECKOFF SHEET

Site Name and Description:	
Date Sampled:	Date Data Entered into WQDB:

Quality Assurance / Quality Control Registry	
Has the Laboratory Report been date stamped upon arrival at DEQ?	Yes < No <
Has the Laboratory Report has been checked for water quality standards violations?	Yes < No <
Have all Report results been scrutinized for dubious or unusual data values?	Yes < No <
Have split or duplicate samples been compared for agreement?	Yes < No < N/A <
Were equipment blank samples in conformity with each other?	Yes < No < N/A <
Have water quality standards violation letters been sent out?	Yes < No < N/A <
Has a notice of exceedence been placed in the Site File?	Yes < No < N/A <
Data Validation	
Is the Field/Lab Specific Conductivity ratio between 0.9 and 1.1?	Yes < No <
Is the Field/Lab pH ratio between 0.9 and 1.1?	Yes < No <
Is the TDS/Specific Conductivity ratio between 0.55 and 0.75?	Yes < No <
Is the TDS/Calculated Sum ratio between 1.0 and 1.2?	Yes < No <
Is the Cation/Anion balance %difference in the acceptable range?	Yes < No <
Has the QA/QC been completed within 30-days of the receipt of the Laboratory Report?	Yes < No <
Remarks:	

Initials of person completing this form:	Date Completed:

5.11.2 Data Entry

All water quality data is entered into the SWQDB and is discussed in the Arizona Department of Environmental Quality Water Quality Database Training Manual (2001), which was generated by the Database Management and Analysis Group. For a complete discussion of section and program protocols for data entry and entry standards, see the Arizona Department of Environmental Quality Water Quality Surface Water Quality Database Data Entry Standards (2002). To view these documents go to http://intranet/training/manuals.html#water'.

5.11.3 Data Checking

All data manually entered into the SWQDB will be checked upon completion of entry by the designated data checker for the trip. This is usually the assistant on the sampling trip, but the checker may be any individual who has database editing rights chosen by the lead sampler. Any errors in data entry should be corrected by the checker. The data are then noted on the *Surface Water Events Per Visit* (SWEPV) screen [WQ751] as being checked with the initials of the checker and the date. The checker also checks the QA/QC measures to verify data validity or confirm problems with the data.

The checking of data entry should conform to the following procedure.

- Attention shall be paid to correct decimal placement, missing data, erroneous values, missing codes or flags, and correctly formatted sample times and dates
- Enter comments missed on first data entry. Where possible, apply standard codes available in a drop-down list with additional comments appended in the following field. If the missed comments are standardized, enter on the SWEPV screen [WQ751]. If the missed comments are general in nature, enter them via the "Comments" button on the *Surface Water Site Visits* screen [WQ762].
- After data has been checked, add the standard comment on the Events screen denoting that data has been checked and append the checker's initials and date in the following field
- Advise the data processor of items needing attention (preferably in writing)
- Return checked data to the data processor

The final data check by the data Processor should conform to the following procedure.

- Follow up on items needing attention
- Insure that the database entries are accurate and complete
- Processor shall click the database "Approval" button to lock the entries against change. Prior to approval, all records are open to editing and viewing by any individual granted SWQDB editing rights. After approval, records are available for read-only access by all parties, including the lead sampler. Changes to records after approval require the assistance of the Data Management Unit.

5.11.4 Surface Water Quality Data Base Laboratory Notation Codes

Table 1 is a list of laboratory results notation codes currently in use in the SWQDB that may qualify or flag results with adjunct information. The list is a hybrid from several sources, consisting of a combination of STORET remark codes, USGS codes, and Arizona Department of Health Services lab-qualifier codes. Codes most frequently used are indicated by an asterisk.

5.11.5 Identifying a QA/QC Problem

The ADEQ Fixed Station Network program calculates five QA/QC data validation measures on data values returned by the laboratory. These analyses duplicate a number of laboratory QC tests typically run by the reporting lab as a means of double-checking the validity of the numbers reported by the lab and familiarizing personnel with the data received. The five measures currently calculated include the following:

- Field/Lab pH ratio
- Field/Lab conductivity ratio
- Total Dissolved Solids/Conductivity ratio
- Total Dissolved Solids/Calculated sum of constituent's ratio
- Cation/Anion Balance

The field/lab ratios serve a limited function of identifying lab or field results for pH or conductivity worthy of closer scrutiny and possible further inquiry, either with the laboratory or in calibration logbooks for FSN equipment. These ratios might ultimately pinpoint a measurement problem that could result in an individual data result qualification in the database or possible exclusion of a problematic individual parameter measurement. However, corroborating evidence of a problem on other fronts must be present for data exclusion to take place. Holding times for both pH and conductivity are short and typically not met by laboratories after a normal sampling run; consequently, excluding data on the basis of the ratios alone cannot be defended as scientifically sound. Nevertheless, these ratios have served a useful function in catching numerous transcription and reporting errors over their years of usage.

Results of the other QC analyses are considered as additional data points associated with the sample event. These measures (TDS/Conductivity, TDS/Sum, Cation/Anion Balance) characterize the matrix being analyzed and do not address the validity or invalidity of any individual analyte result. As such, they are used for informational content and as investigative tools. Should one or more of any of these QC tests fall outside the normal range for natural waters, they may ultimately result in a sample reanalysis to confirm the result. However, in no case would any of the above listed QC measures be used as a means of wholesale acceptance or rejection neither of sample event data analyses, nor of individual analyte or parameter results except as outlined in the previous paragraph.

Table 1. Laboratory results notation codes.

Lab Notation	Description
LT	'Less than' value - unknown if below quantification level
ND*	Not detected
LT*	'Less than' value - detection was below quantification level
LT*	'Less than' value - no presence of compound detected
GT	Greater than quantification level.
TR	Trace
U	Compound was not detected above the concentration listed.
J*	Values are estimated, data is valid for limited purposes.
K	Compound is present, but below listed value
R	Values have been rejected, data is invalid for all purposes.
L	Result between contract quantitation & instrument detect limit
N	Presumptive evidence of the presence of the compound
UJ	Sample quantitation limit was adjusted-value is estimated
POS	Positive
PR	Compound is present
B*	Colony counts outside acceptable range (20-60 cfu)
C	Value calculated
D	Dilution factor used
E	Reported value estimated due to matrix interference
H	Values are estimated by field kit method
P*	Bacteria too numerous to count (TNTC)
Q*	Holding time exceeded
V	Significant rain during past 48 hours may affect results
X	Other (see comments from sample)
F	Analyte found in sample blank as well as sample
S	Spiked sample recovery outside control limits
M	Duplicate analysis outside control limits
Y	QC ratios outside acceptable range
O*	Sampled, but analysis lost or not performed
A	Value reported is the mean of two or more determinations
G	Value reported is the maximum of two or more determinations
T	Value reported is less than criteria of detection
W	Value observed is less than lowest value under "t"
Z	Too many colonies to count, value is filtration volume
D2	Sample required dilution due to high concentration

5.11.5.1 Dubious or Questionable Values

Dubious or questionable data values are more easily identified when the data reviewer has familiarity with past analytical results from the sample site. All results are scrutinized and any dubious or questionable values that are found are tracked back to the lab or field sheets for possible transcription errors or misreported values. Conversations with laboratory personnel may be necessary to resolve the problem.

5.11.5.2 Split/Duplicate Agreement

Any split/duplicate samples must be compared to assure reasonable agreement. The adopted standard for acceptable results from splits and duplicates is a +/- 10% difference from the mean of the two samples for each cation, anion, and nutrient. For total and dissolved metals, particularly of low concentrations, a difference of 10% to 50% may be allowed based on best professional judgment. Where one split is reported as a non-detect, and the other a value above the detection limit, ADEQ has adopted the ADHS protocol of acceptance of split results if the reported value in such a case does not exceed two times the detection limit.

Certain circumstances may preclude the application of these measures, including the analysis of less than complete sampling suites, sampling in heavily-polluted waters or floodwaters, and the necessity of analyzing for parameters not originally included in the sampling plan in order to run the test. Most sampling situations, though, will benefit from the application of these measures. The following recommendations are made:

Field/Lab pH ratio:	Should be calculated any time both a field pH measurement and a lab pH measurements are performed. Though pH holding time is only 15 minutes and labs rarely if ever meet this holding time, pH does not drift as much as formerly thought. An evaluation of FSN historic data shows that in a population of 3300 ratios existing for historic data, 93% met the acceptance criteria. Furthermore, several lab transcription errors have been caught over time due to the employment of this test.
Field/Lab EC Ratio:	Should be calculated any time both a field EC measurement and a lab EC measurement are performed. Holding time of 28 days on conductivity is not a factor.
TDS/EC Ratio:	Recommended for all samples. Routinely provided by ADHS.
TDS/ Calculated Sum:	Recommended for samples where a complete set of cations and anions analyses are ordered. A complete set consists of the following: for cations, analyses of Ca, Mg, K, and Na (NH_3 may be incorporated if present); for anions, analyses of CO_3, HCO_3, Cl, F, NO_2+NO_3, and SO_4. Alkalinity (hardness as $CaCO_3$) is also necessary. The constituents of the actual sum include Ca, Mg, K, Na, Cl, F, NO_2+NO_3, SO_4, and 0.6 x the hardness as $CaCO_3$.

Cation/Anion Balance: Recommended for samples where a complete set of cations and anions analyses are ordered. A complete set consists of the following: for cations, analyses of Ca, Mg, K, and Na (NH_3 may be incorporated if present); for anions, analyses of CO_3, HCO_3, Cl, F, NO_2+NO_3, and SO_4. This information is routinely provided by ADHS.

5.11.5.3 Resolving Problems with QA/QC Measures

If QA/QC measures are not within the range designated as acceptable, there are several possible reasons as to why they fall outside the designated range. Troubleshooting these problem values includes the following and are listed in order of common occurrence.

- Double check data entries in the database. This is the most common problem and by far the easiest to catch and correct.
- Check to see that the database entries contain a complete set of analytical results. A missing cation or anion will cause the QA/QC measures to be out of range.
- Double check the value reported by the lab. Laboratories are not without error. If an error is suspected, call the lab about it.
- Check any historic data at this site for obvious trends. For instance, if a particular QA/QC ratio is low and the historic data is also consistently low over a period of a year or so, then there may not be a problem. This may just be a unique chemical characteristic of the site and not indicative of a QA/QC problem with the data. Long-term sites have a compilation sheet showing historic QC values in the site file. Refer to the sheet for a determination of historic trend data.
- Check to see if the sample was collected under unusual conditions; i.e. flood, drought, incoming pollutants, high algae content, etc. This can potentially throw ion balances and field to lab ratios out of the acceptable range.
- In select cases, where suspicious values are present, or splits and/or duplicates have widely-differing results, have the test rerun. As the QA/QC measures are meta-data and indicate characteristics of the matrix rather than individual analytes, test reruns based solely on out-of-range QC data validation measures are not warranted. Instead, samples that have anomalous QC ratios shall be qualified by either a comment or a sample flag.

  Test reruns may cost extra money, but can be worthwhile if the new value is significantly different than the first. If there was an error on the lab's part, most labs won't charge for the test rerun. If the lab integrity is in question, have another lab do the rerun.

- Have the lab provide you with their QA/QC values. These measures should include ion balances as well as other internal checks that labs perform to assure the quality of their data. It is possible that the lab is performing ion balances that are somewhat different than ours and theirs may be more reliable. Most labs are willing to do this at no extra cost. If the lab has a problem with the ion balances, they will often state this

on the cover sheet or in the remarks area provided on the lab reports. It is typical that if the lab has problems, they will have already tried to resolve them and have found that they cannot be resolved.

Information regarding the QA/QC measures and various troubleshooting methods were collected from ADHS laboratory personnel, Standard Methods for the Examination of Water and Wastewater, and the U.S. Geological Survey Water-Supply Paper 2254 entitled Study and Interpretation of the Chemical Characteristics of Natural Water by John D. Hem, 1989.

5.11.5.4 QA/QC Ratios on Field and Laboratory EC and pH

These field to lab ratio checks are the ratio of the field value divided by the lab value.

Field/Laboratory pH Ratio

Normal Acceptable Range = 0.9 TO 1.1

For pH ratios: Unqualified Range = 0.95 to 1.05 ⇨ Acceptable - No further action is necessary.

Qualified Range = 0.90 to 0.95 or 1.05 to 1.10 ⇨ PH ratios falling into the Qualified Range may have masked problems, and it is encouraged that they be investigated for transcription errors or other problems.

Example: Field pH = 7.60 and Lab pH = 7.10

Field / Lab pH ratio = 7.60/7.10 = 1.07 (round to 1.1)

This value falls within the normal acceptable range for this QA/QC ratio and is deemed technically acceptable. However, it is encouraged that the values be investigated more closely, as a difference of 0.55 standard units represents a sizable shift in the acidic-basic character of the water. There may be a masked problems with such a result. Further investigation is advised.

Field/Laboratory EC Ratio

The EC ratio is calculated identical to the pH ratio but those ratios are not subjected to the advisory range designated for pH ratios. Any value falling between 0.9 and 1.10 for an EC ratio is acceptable without reservation.

CAUTION: Effluent dominated waters, industrial sewage, and other highly polluted samples will have more problems with the pH ratio than ambient water samples. The pH of a sample may change very rapidly if the sample contains large amounts of algae, microbes or air bubbles. Over-zealous churning of the churn splitter can also cause a change in pH by aerating a sample

that may have been anaerobic when field measurements were taken. Floods or drought have little negative impact on this QA/QC ratio.

Troubleshooting for pH and EC F/L ratios falling outside acceptable ranges with the most important items listed first.

- Check field value entries in the database against field values contained in field notes
- Check lab value entries in the database against lab values on lab report
- Call lab to check reported lab values. Many times, a simple transcription error is to blame
- Determine if there is a trend over time at this particular site that may indicate that the resultant ratio is a unique chemical characteristic of most samples collected
- Check, clean or replace batteries, KCl reference solution, or probes in field equipment
- For a chronic problem, the field meter may need to be returned to the manufacturer for servicing
- On occasion, effluent-dominated waters' pH or electroconductivity may change between the field measurement and the lab measurement. This is entirely normal. Sample holding times between collection and lab analysis can have a direct impact on the actual magnitude of change.

Note: A ratio outside of accepted limits with no corroborating evidence of a problem from another source (e.g., post-trip calibrations out of bounds, field notes indicating equipment problems, lab qualifiers, etc.) shall not be censored from the data set on the basis of the ratio alone.

5.11.5.5 QA/QC Measures on Total Dissolved Solids

There are two QA/QC checks related to total dissolved solids (TDS). They are:

1. **TDS/EC Ratio** - this is a ratio of TDS (lab evaporation in mg/l) to EC (lab EC in μmhos/cm or μsiemens/cm). According to Hem, this ratio should range from 0.55 to 0.75. The reason this ratio is not equal to 1.0 is that the TDS lab results do not reflect 100% of the measured lab EC. Some constituents that are reflected in the lab EC are not detected in the lab TDS.

   Example: TDS = 388 Lab EC = 439

   Normal Acceptable Range = 0.55 TO 0.75

   TDS/EC Ratio = 388/439 = 0.884 (round to 0.9)

This value falls outside of the normal acceptable range and is, therefore, not acceptable without some additional investigating.

CAUTION: Some ambient waters in the state will consistently have values that fall outside this ratio. This is acceptable if the trend is established and obvious as observed in historic sample results. If a sample is high in bicarbonate (HCO_3), the TDS/EC ratio will be low because bicarbonate volatilizes and escapes measurement in this lab test. If a sample is high in sulfate (SO_4), the TDS/EC ratio will be high as sulfate tends to retain water and cause the lab evaporation to weigh more. Ambient waters with extremely high or extremely low TDS can give variable results.

Troubleshooting with the most important items listed first.

- Check lab EC and TDS entries in the database against the lab report
- Check historic TDS/EC ratios for trend indicative of unique chemical characteristics of this site. Long-term sites have a compilation sheet showing historic QC values in the site file. Refer to the sheet for a determination of historic trend data.
- Check bicarbonate and sulfate values to see if they are exceedingly high or low compared to past data at the site
- Check field to lab EC ratio for acceptance. If it is acceptable then the problem may be TDS.
- Call lab to check on reported lab values or have tests rerun

2. **TDS/Sum** - this ratio relates TDS (lab evaporation reported in mg/l) to the sum of the major constituents (lab values for Ca, Mg, Na, K, SO_4, F, NO_3+NO_2 and Cl; all must be reported in or converted to mg/l). Hardness as $CaCO_3$ is also incorporated at 0.6 times its reported value.

   Example: TDS = 388

   Normal Acceptable Range = 1.0 to 1.2

   Sum of ions in mg/l [Ca+Mg+Na+K+F+(NO_2+NO_3) +SO_4+Cl]
   Hardness as $CaCO_3$: 350 mg/l
   Calculation = (24+15.1+35+12.3+0.5+0+22+23.4) + (0.6*350) = 318.54
   TDS/Sum = 388/318.54 = 1.218 (round to 1.2)
   This value falls at the limit of the range and is acceptable.

CAUTION: This ratio may have the same problems as the TDS/EC ratio because of the nature of sulfate during the TDS lab test. The sum of the major cations and anions may also be suspect if the sample contains large amounts of nutrients, silica, or trace metals (i.e. nitrates, iron, manganese) as a result of flooding or incoming pollution. The sum value may be low if large amounts of trace metals are present in sample because these significant values are not calculated into the sum total. Ambient waters with extremely high or extremely low TDS can give variable results.

Troubleshooting of TDS ratios falling outside of acceptable ranges with the most important listed first.

- ❖ Check data entries in the database for TDS, major cations, and anions against lab report
- ❖ Check nutrients and trace metals analyses for exceedingly large amounts of trace metals or nutrients. These are commonly elevated during flood events.
- ❖ Estimate the amount, if any, the sum may be increased by including these additional values (must be in mg/L) and recalculate the new ratio by hand if necessary
- ❖ Check TDS/EC ratio for acceptance - if it is not acceptable then problem may be TDS
- ❖ Call lab to check reported lab values or have tests rerun

Note: While it is desirable that complete sampling suites meet these criteria, TDS ratios are considered metadata points indicating characteristics of the matrix in which sampling occurs. Consequently, such ratios indicate little concerning the accuracy of any individual analyte and shall be used as investigatory tools only. They shall not be used to exclude data from entry in the DEQ repositories unless evidence of other problems with the data exist (e.g., field and lab TDS values do not agree, etc.).

5.11.5.6 QA/QC Check on Ion Balances

The units of Cations/Anions measure is in milliequivalent per liter (meq/L) - this check is a general ion balance check using the five major cation values and the five (5) major anion values reported by the lab.

The cations are: Ca, Mg, Na, K, NH_3
The anions are: F, CO_3, HCO_3, SO_4, Cl

The cation/anion values are converted from mg/L (milligrams per liter) to meq/L by using the following conversion factors obtained from Standard Methods for the Examination of Water and Wastewater - 20th Edition.

Ca (mg/L) = Ca x 0.0499 = Ca (meq/L)
Mg (mg/L) = Mg x 0.08229 = Mg (meq/L)
Na (mg/L) = Na x 0.0435 = Na (meq/L)
K (mg/L) = K x 0.02557 = K (meq/L)
NH_3 (mg/l) = NH_3 x 0.0713 = NH_3 (meq/l)
CO_3 (mg/L) = CO_3 x 0.033 = CO_3 (meq/L)
HCO_3 (mg/L) = HCO_3 x 0.01639 = HCO_3 (meq/L)
SO_4 (mg/L) = SO_4 x 0.02082 = SO_4 (meq/L)
Cl (mg/L) = Cl x 0.02821 = Cl (meq/L)
F (mg/l) = F x 0.0526 = F (meq/l)
NO_2+NO_3 as N (mg/l) = (NO_2+NO_3) x 0.0713 = NO_2+NO_3 as N (meq/l)

The converted cation values are summed to obtain a total cation balance value in meq/L. The converted anion values are also summed to obtain a total anion balance value in meq/L. See the following example for the step-by-step procedure.

Example:

Cation and Value, mg/L	Conversion Factor	Meq/L Value
Ca = 24	0.0499	1.20
Mg = 15.2	0.08229	1.25
Na = 35	0.0435	1.52
K = 12.3	0.02557	0.31
NH_3 = ND*	0.0713	0.00
	Total	4.28

Anion and Value, mg/L	Conversion Factor	Meq/L Value
F = ND*	0.0526	0.00
CO_3 = 0.5	0.033	0.02
HCO_3 = 215	0.01639	3.52
SO_4 = 22	0.02082	0.46
Cl = 23.4	0.02821	0.66
NO_2+NO_3 = ND*	0.0713	0.00
	Total	4.66

* Analytes that are reported as "ND" are generally assumed to be 0.00 meq/l in ion balance calculations. Rarely does this assumption by itself cause a balance to fall out of range. If it does so, ½ the detection limit may be substituted into the milliequivalents calculation.

Standard Methods outlines the following acceptance criteria for ion balance calculations:

Anion Sum (meq/l)	Acceptable Difference
0 - 3.0 meq	+/- 0.2 meq/l
3.0 - 10.0	+/- 2%
10.0 - 800	+/- 5%

Percentage difference is calculated as follows:

$$\%Diff. = 100 \times \frac{\sum cations - \sum anions}{\sum cations + \sum anions}$$

From the example above,

$$100 \times \frac{4.28 - 4.66}{4.28 + 4.66} = -4.25\%$$

Since the Anion Sum (-4.25%) falls in the range of 3.0-10.0 meq/l, the greatest difference acceptable is +/-2%. This would be an example of a sample that does not meet the ion balance acceptance criteria, and further investigation would be advised.

CAUTION: As with the TDS/Sum ratio, if a nutrient or trace metal has exceptionally high values, then the sum value for the respective cations or anions will not reflect a true summation of all ions available. For example; if iron is exceptionally high, then the summed cation value, which does not take iron into consideration in the calculation, will appear relatively low. A similar ratio problem appears when nutrients such as nitrate or phosphate are elevated. Since they are not included in the anion summation calculation, this may cause failure to meet the acceptance criteria.

Troubleshooting with the most important listed first.

- ❖ Check data entries in the database against major cations and anions on lab report
- ❖ Check additional analyses for exceptionally large amounts of trace metals or nutrients. These values, if large enough, may have an effect on the total cation or total anion values.
- ❖ Estimate the amount, if any, the sums may be increased by converting the additional values to their respective meq/l equivalent, adding these additional values and recalculate the new ratio by hand if necessary
- ❖ Call lab to check reported lab values

5.11.5.7 QA/QC Comparison for Split and Duplicate Samples

A split sample is a large composite sample that has been fully mixed and split between two (2) identical sets of sample bottles. Split samples are akin to "identical twins" - the same exact chemical make up. Duplicate samples are simply two (2) samples collected at the exact same location but not composited together and split apart. Thus, duplicate samples are more akin to "fraternal twins" - the similar gene pool, but slightly different chemical make up. Either way these check samples are collected, their chemical characteristics should be very close to their respective "twin" regular sample.

There is a Excel spreadsheet file available for section personnel that allows QA/QC checks to be performed on split and duplicate samples. This spreadsheet is identified as "Percent diff.xls." A copy of this file is kept available for use on the J: drive (currently J:/HSA/FSNDATA/SWMFORMS).

1. Enter the data for each sample in one of the two columns available.

2. Percent difference will be calculated in the far right column using the following formula:

$$\%Diff. = \frac{Sample1 - Sample2}{\frac{Sample1 + Sample2}{2}} \text{ x } 100, \text{ or } \quad \%Diff. = \frac{2(Sample1 - Sample2)}{Sample1 + Sample2} \text{ x } 100$$

Example: Lab EC (Sample1) = 591 and QC Lab EC (Sample) = 602

$$\%Diff. = \frac{2(591 - 602)}{591 + 602} x100 = 1.8$$

3. The percent difference value for all of the checks should be less or equal to ten percent (10%) with two exceptions: trace metals checks and analytes whose averages are less than two times the detection level. The trace metal values are reported in μg/L and are generally small. Therefore, a slight variation in numbers may cause a one-hundred percent difference which would be non-indicative of a real problem. A five-hundred percent difference may be more indicative of a true problem. A professional judgment must be made here by section personnel. Likewise, analytes close to the detection limit are more likely to show a large percentage difference when the absolute magnitude of the difference is small. In these cases, the 10% rule of thumb should be disregarded.

5.11.6 Literature Cited

ADEQ, 2002. Arizona Department of Environmental Quality Surface Water Quality Database Data Entry Standards. Phoenix, AZ.

ADEQ, 2001. Arizona Department of Environmental Quality Water Quality Database Training Manual. Phoenix, AZ.

American Public Health Association, 20th edition. 1999. Standard Methods for the Examination of Water and Wastewater. Washington, D.C.

Hem, J.D., 1989. U.S. Geological Survey Water-Supply Paper 2254. Study and Interpretation of the Chemical Characteristics of Natural Water. U.S. Gov't. Printing Office, Washington, D.C.

5.12 Determining and Reporting Exceedances of Surface Water Quality Standards

Water quality data should be reviewed and compared to surface water quality standards as soon as possible after receipt of analytical results from the laboratory to determine whether there are any exceedances of surface water quality standards.

Surface water quality standards are prescribed in Title 18, Chapter 11, Article 1 of the Arizona Administrative Code. Water quality standards for parameters for which water quality data are routinely collected may be found on the Surface Water Quality Standards (Figure1). This form is an only an example and will likely change after any Water Quality Standards Triennial Review process has been completed. The form is filled out in conjunction with form Water Quality Sample QA/QC Checkoff Sheet (Figure2). Completion of both forms are a convenient method for determining exceedances of surface water quality standards.

Instructions for using the Surface Water Quality Standards Checkoff Sheet and Water Quality Sample QA/QC Checkoff Sheet follow:

1. Complete front page of the Sample checkoff sheet.

   - ❖ Enter site code and unique ADEQ identifier from Surface Water Quality Database for the sampling site on the checkoff sheet
   - ❖ Enter the stream name and location
   - ❖ Determine applicable designated uses for the stream reach where the sample was collected from Appendix B of the Surface Water Quality Standards rules. Many surface waters are specifically listed in Appendix B by major river basin. Look to see if the surface water that you sampled is listed in Appendix B. If so, enter the listed designated uses for the surface water on the front of the checkoff sheet and highlight the same designated uses on the reverse side. If the surface water you sampled is not listed in Appendix B, consult the tributary rule at R18-11-105 to determine designated uses.
   - ❖ Compare water quality data from laboratory report to the applicable water quality standards. Highlight any water quality standards that are exceeded. Remember to compare water quality data to both acute and chronic aquatic life criteria. Note each exceedence in the remarks section on the front of the checkoff sheet.

2. Complete the QA/QC checks from the front of checkoff sheet. Are any of the water quality standards exceedances based on suspicious values or qualified data? Could any exceedence be related to natural background conditions? If so, note in the remarks section of the checkoff sheet.

3. Document exceedances on a Notice of Exceedence memorandum for internal routing within ADEQ. A copy of the Notice of Exceedence should be sent to the Unit Manager of the Surface Water Monitoring and Standards Unit, the §305(b) Water Quality

Assessment Coordinator, and the Section Manager of the Water Quality Compliance Section. A facsimile of the Notice of Exceedence memorandum is found on Figure 3.

4. If an exceedence is of a surface water quality standard intended to protect human health (i.e., the domestic water source (DWS), fish consumption (FC), full body contact recreation (FBC) or the partial body contact recreation (PBC) designated uses) a Notice of Exceedence letter (Figure3) should be prepared and sent to the Director of the County Health Department of the county where the sampling site is located. Do not send notices to the County Health Director when the exceedence is for a water quality standard to protect aquatic life.

5. File a copy of the Notice of Exceedence in the site file.

Figure 1. Surface Water Quality Standards (in µg / L) (Effective March 8, 2002).

Parameter	A&Wc acute	A&Wc chronic	A&Ww acute	A&Ww chronic	A&Wedw acute	A&Wedw chronic	DWS	FC	FBC	PBC	AgI	AgL
D.O mg/L	7.0[a]		6.0[a]		3.0[a,b] 1.0[a,c]							
Chlorine (T)	11	5	11	5	11	5	700		140,000	140,000		
E. coli									235 cfu	576 cfu		
pH	6.5 to 9.0	6.5 to 9.0	6.5 to 9.0	6.5 to 9.0	6.5 to 9.0	6.5 to 9.0	5.0 to 9.0	6.5 to 9.0	6.5 to 9.0	6.5 to 9.0	4.5 to 9.0	6.5 to 9.0
Total Ammonia	Total ammonia standards apply to surface waters with the A&Wc and A&Ww designated uses only. Criteria are pH- and temperature-dependent. See Table 24 for acute criteria and Table 25 for chronic criteria in Appendix A of the Surface Water Quality Standards rules.											
Total Phosphorus	Total phosphorus standards are site-specific. See R18-11-109(F).											
Total Nitrogen	Total nitrogen standards are site-specific. See R18-11-109(F).											
Nitrate							10,000		2,240,000	2,240,000		
Nitrite							1,000		140,000	140,000		
Antimony	88 D	30 D	88 D	30 D	1000 D	600 D	6 T	4,300 (T)	560 T	560T		
Arsenic	360 D	190 D	360 D	190 D	360 D	190 D	50 T	1,450 (T)	50 T	420 T	2,000 T	200 T
Beryllium	65 D	5.3 D	65 D	5.3 D	65 D	5.3 D	4 T	1,130 T	2,800 T	2,800 T		
Boron							630 T		126,000T		1,000 T	
Cadmium	HD	HD	HD	HD	HD	HD	5 T	84 T	700 T	700 T	50 T	50 T
Chromium							100 T		100 T	100 T	1,000 T	1,000 T
Copper	HD	HD	HD	HD	HD	HD	1,300 T		1,300 T	1,300 T	5,000 T	500 T
Lead	HD	HD	HD	HD	HD	HD	15 T		15 T	15 T	10,000 T	100 T
Mercury	2.4 D	0.01 D	2.4 D	0.01 D	2.6 D	0.2 D	2 T	0.6 T	420 T	420 T		10 T
Selenium	20 T	2.0 T	20 T	2.0 T	50 T	2.0 T	50 T	9,000 T	7,000 T	7,000 T	20 T	50 T
Zinc	HD	HD	HD	HD	HD	HD	2,100 T	69,000 T	420,000T	420,000T	10,000 T	25,000 T

Footnotes:

[a] If dissolved oxygen is less than water quality standard but percent saturation ≥ 90%, then no exceedence
[b] 3 hours after sunrise to sunset
[c] Sunset to 3 hours after sunrise
HD Hardness-dependent. Consult Appendix A of the surface water quality standards for water quality standards.
D Dissolved
T Total recoverable

Figure 2. Water quality sample QA / QC checkoff sheet.

Site ID:	SWQDB Code:

WATER QUALITY SAMPLE QA / QC CHECKOFF SHEET

Site Name and Description:	
Date Sampled:	Designated Uses:

Quality Assurance / Quality Control Registry	
Has the Laboratory Report been date stamped upon arrival at DEQ?	Yes < No <
Has the Lab Report has been checked for water quality standards exceedances?	Yes < No <
Has a Notice of Exceedence (NOE) memo been routed internally?	Yes < No <
Are any exceedances related to human health designated uses?	Yes < No <
If yes, has a NOE been sent to County Health Director?	Yes < No <
Have all Report results been scrutinized for suspicious data values?	Yes < No <
Have split or duplicate samples been compared for agreement?	Yes < No < N/A <
Did equipment blank samples come back clean?	Yes < No < N/A <
Have water quality standards violation letters been sent out?	Yes < No < N/A <
Has a Notice of Exceedence been placed in the site file?	Yes < No < N/A <
Data Validation Measures	
1) Is the Field/Lab Specific Conductivity ratio between 0.9 and 1.1?	Yes < No <
2) Is the Field/Lab pH ratio between 0.9 and 1.1?	Yes < No <
3) Is the TDS/Specific Conductivity ratio between 0.55 and 0.75?	Yes < No <
4) Is the TDS/Calculated Sum ratio between 1.0 and 1.2?	Yes < No <
5) Is the Cation/Anion balance %difference in the acceptable range?	Yes < No <
6) Has the QA/QC been completed within 30-days of the receipt of the Laboratory Report?	Yes < No <
Remarks:	

Initials of person completing this form:	Date Completed:

Figure 3. Notice of Exceedence.

ARIZONA DEPARTMENT
OF
ENVIRONMENTAL QUALITY

1110 West Washington Street • Phoenix, Arizona 85007
(602) 771-2300 • www.adeq.state.az.us

Janet Napolitano
Governor

Stephen A. Owens
Director

October 5, 2005

[Name of County Health Director]\
[Address]

RE: NOTICE OF EXCEEDANCE; SURFACE WATER QUALITY STANDARDS

[Salutation]:

This letter is to notify your office that the ADEQ Surface Water Monitoring and Standards Unit collected a water sample from [Monitoring Site Name] ([Site ID]) on [Sample date] as part of our routine surface water quality monitoring program. Analyses determined that Surface Water Quality Standards (A.A.C. R18-11) were exceeded as follows:

Water Quality Parameter	State Limit	Reported Value	Reporting Lab
[Parameter]	[State limit and units]	[Reported value and units]	[Reporting lab and lab #]

This information will be compared to previous data if available. If the exceedance is chronic or of a severe nature, further investigations will be performed to determine the cause. Feel free to call me should you have any questions.

Sincerely,

[Sampler Name], [Job Title]
Surface Water Monitoring and Standards Unit
Arizona Dept. of Environmental Quality
1-800-234-5677 X(Extension] or 602-(Full Phone)

cc:
Linda Taunt, Manager, Hydrologic Support and Assessment Section
Steve Pawlowski, Manager, Surface Water Monitoring and Standards Unit
Jason Sutter, Manager, TMDL and Assessment Unit
Mike Traubert, Manager, Water Quality Compliance Section
Susan Ward, Manager, Watershed Management and Assessment Unit

Printed on recycled paper

5.13 Calculating Discharge

After all flow measurements have been recorded on the field data sheet, discharge can be calculated. Figure 4 shows a field data sheet with recorded measurements.

In this example, 20 measurements were taken across the stream channel. Calculations are performed from left to right on each line across the form. Calculations are also performed for each column to obtain total distance (DIST), total WIDTH, average DEPTH, average velocity (VEL), total discharge (Q).

Using the information recorded on the example field data sheet, the values are calculated as shown in Figure 5. Round numbers as per convention; if the last number is 1, 2, 3, or 4, then round down; if the last number is 5, 6, 7, 8, or 9, then round up.

5.13.1 Discharge Calculation Procedure

1. Calculate the Total DIST by subtracting $DIST_{20}$ from $DIST_1$. In this example, the cross-section distance is 58.0 feet.

2. Calculate WIDTH from the DIST measurements as follows.

    a. For the WIDTH calculation on line 1, subtract $DIST_1$ from $DIST_2$ and calculate the mean
    b. For the WIDTH calculation on line 2, subtract $DIST_1$ from $DIST_3$ and record the mean
    c. For the WIDTH calculation on line 3, subtract $DIST_2$ from $DIST_4$ and record the mean
    d. This method of calculating WIDTH is continued until the last measurement on line 20. For the WIDTH calculation on line 20, subtract $DIST_{19}$ from $DIST_{20}$ and calculate the mean
    e. Sum the values in the WIDTH column. Compare it to the Total Distance value. If they are not the same, an error has been made which must be rectified before proceeding with the Q calculations.
    f. Sum the DEPTH column and record the mean
    g. Sum the VEL column and record the mean
    h. To obtain Q, multiply WIDTH $*$ DEPTH $*$ VEL for each line
    i. Sum the Q column to obtain discharge in cubic feet per second

5.13.2 Calculating Discharge with an Excel Template

An alternative to the manual calculations is an Excel Spreadsheet (Figure 6) where the calculations are machine calculated as the form is being populated with the flow data. This template can be found at J:HAS\Surface Water Monitoring\SWMFORMS\Discharge Calculator.xls. The completed form should be printed and included with the field data sheets in the site file.

5.13.3 Source Material

Harrelson, C.C., C.L. Rawlins, and J.P. Potyondy. 1994. Stream channel reference sites: an illustrated guide to field technique. U.S.D.A., Forest Service, Rocky Mountain Forest and Range Experiment Station. Gen. Tech. Rep. RM-245, Ft. Collins, CO.

Figure 4. Field recorded discharge measurements.

INSTANTANEOUS DISCHARGE					
	DIST	WIDTH	DEPTH	VEL	Q
1	0.8		0	REW	
2	1.0		0.45	.22	
3	4.0		0.59	0.33	
4	7.0		0.98	0.56	
5	10.0		1.21	0.89	
6	13.0		1.53	1.25	
7	16.0		1.65	1.66	
8	19.0		1.48	2.11	
9	22.0		1.50	2.46	
10	25.0		1.53	2.35	
11	28.0		1.53	2.56	
12	31.0		1.55	2.54	
13	34.0		1.65	2.12	
14	37.0		1.68	2.29	
15	40.0		1.78	2.37	
16	43.0		1.67	2.18	
17	46.0		1.23	2.56	
18	49.0		1.04	1.89	
19	52.0		0.85	1.01	
20	55.0		0.3	.89	
21	58.0		0	LEW	
22					
Total Dist.		QC summed Total	Avg. Depth	Avg. Velocity	Summed total Q

Where:

DIST = Distance; recorded in tenths of a foot (0.1 ft).

WIDTH = a calculated value in tenths of a foot (0.1 ft).

DEPTH = recorded in hundredths of a foot (0.01 ft).

VEL = Velocity; measured in hundredths of feet per second (fps).

Q = Discharge; a calculated value in cubic feet per second (cfs); to the nearest tenth or hundredth of a foot depending on total flow estimation

Figure 5. Completed discharge measurement form.

INSTANTANEOUS DISCHARGE						
	DIST	WIDTH	DEPTH	VEL	AREA	Q
1	0.8	0.1	0	REW	0	0
2	1.0	1.6	0.45	.22	0.72	0.16
3	4.0	3	0.59	0.33	1.77	0.58
4	7.0	3	0.98	0.56	2.94	1.65
5	10.0	3	1.21	0.89	3.63	3.23
6	13.0	3	1.53	1.25	4.59	5.74
7	16.0	3	1.65	1.66	4.95	8.22
8	19.0	3	1.48	2.11	4.44	9.34
9	22.0	3	1.50	2.46	4.50	11.07
10	25.0	3	1.53	2.35	4.50	10.58
11	28.0	3	1.53	2.56	4.59	11.75
12	31.0	3	1.55	2.54	4.59	11.66
13	34.0	3	1.65	2.12	4.59	10.49
14	37.0	3	1.68	2.29	5.04	11.54
15	40.0	3	1.78	2.37	5.34	12.66
16	43.0	3	1.67	2.18	5.01	10.92
17	46.0	3	1.23	2.56	3.69	9.45
18	49.0	3	1.04	1.89	3.12	5.90
19	52.0	3	0.85	1.01	2.55	2.58
20	55.0	3	0.3	.89	0.90	0.80
21	58.0	1.5	0	LEW	0	0
22						
Total Dist.		QC summed Total	Avg. Depth	Avg. Velocity	Total Area	Total Q
57.2		57.2	1.15	1.54	71.82	138

Figure 6. Discharge results from Excel template.

Site Name: South Fork Lost Keeper Creek
Date: 3/22/2005
Time: 1400 hours
Party: RW/KP
Weather: Clear sky and windy
Air Temp: 22 C
Water Temp: 15 C

Shaded columns contain formulas; do not enter values in these columns.

Distance From Initial Point	Width	Depth	Observation Depth Valid Entries: (0.2, 0.6, 0.8)	Velocity at point: Field Readings	Mean Velocity: Average of 2 Point Velocities	Area	Discharge
0.8	0.1	0		0			
1	1.6	0.45		0.22		0.72	0.1584
4	3	0.59		0.33		1.77	0.5841
7	3	0.98		0.56		2.94	1.6464
10	3	1.21		0.89		3.63	3.2307
13	3	1.53		1.25		4.59	5.7375
16	3	1.65		1.66		4.95	8.217
19	3	1.48		2.11		4.44	9.3684
22	3	1.5		2.46		4.5	11.07
25	3	1.5		2.35		4.5	10.575
28	3	1.53		2.56		4.59	11.7504
31	3	1.53		2.54		4.59	11.6586
34	3	1.65		2.12		4.95	10.494
37	3	1.68		2.29		5.04	11.5416
40	3	1.78		2.37		5.34	12.6558
43	3	1.67		2.18		5.01	10.9218
46	3	1.23		2.56		3.69	9.4464
49	3	1.04		1.89		3.12	5.8968
52	3	0.85		1.01		2.55	2.5755
55	3	0.3		0.89		0.9	0.801
58	1.5	0		0		0	0

Total Width	Sum of Widths	Average Depth	Average Velocity			
57.2	57.2	1.15	1.54	Total Area:	71.82	
				Total Discharge:		138

Width:
Method:
# of Sections:
Type of Meter: Flow mate
Meter #: 2000
Remarks:

No. of Sections: 20

Note: FSN Procedures include bank depths and velocities of 0 in the average velocity and average depth.

SECTION 6

SUPPORTING MATERIAL

TABLE OF CONTENTS

6.00 Documentation and Supporting Material

The information found in this section are not surface water sampling procedures, but are documentation and supporting material to processes related to procedures.

6.10 Macroinvertebrate Contract Taxonomy Laboratory Requirements for Processing Samples

The procedures followed at the consultant taxonomic laboratory are not part of the field procedures for collecting and preserving macroinvertebrate samples. However, this section is included to explain practices and requirements for documentation and future reference.

6.10.1 Sample Receipt

Upon receipt of the samples, the laboratory will check and adjust the preservation in each sample, catalog the samples, check the attached inventory for accuracy, and sign the chain of custody papers. The consultant will then notify ADEQ of the receipt of samples, any damaged samples, or discrepancies between the inventory and actual sample labels.

6.10.2 Sample Processing

Samples must be sorted to separate the invertebrates from the sample matrix. The entire sample should be floated in water in a white plastic tray. Large debris is rinsed and removed from the sample until all organic matter and invertebrates are floated off the mineral residue. The mineral residue is then searched for stone-cased caddisflies and mollusks.

6.10.3 Sub-Sampling

Arizona samples typically contain thousands of invertebrates and must be sub-sampled for results to meet a minimum count of 500-600 organisms. A Caton Tray is be used to randomly obtain fractions of the total sample from which all the invertebrates are removed and counted. Additional fractions are selected until the 500-600 target level is reached after which the number of squares subsampled are recorded. Terrestrial insects and non-benthic insects (e.g. corixidae, other swimmers, mosquitoes, or surface tension dwellers) should not be included in the count. Additional fractions are examined if one fraction is dominated by a single species. After the target number of specimens has been achieved, the entire unsorted sample is scanned for large or rare taxa, which may aid in identification of smaller instars or may expand the taxa list for that sample. The remaining unsorted sample is re-preserved with 70% isopropanol in individual containers and archived at the laboratory for one year from the date of sample receipt, after which time the laboratory will contact ADEQ prior to disposal.

6.10.4 Sorting

The sorting of invertebrates from the sample matrix shall be performed by trained technicians, using dissecting scopes with a minimum magnification of 6X. After identifications have been

made, the sorted specimens, including the separated Chironomidae, should be archived for one year or incorporated into the reference or voucher specimen set. The laboratory shall keep logs for each sample sorted, the fraction sorted, sample matrix problems, etc. in addition to bench sheets of the taxa identified in each sample.

6.10.5 Sorting Efficacy

The laboratory shall check the sample residues to check for a sorting efficacy of 95% or better. A statement of sorting efficacy for the ADEQ batch of samples should be presented in the laboratory report.

6.10.6 Taxonomic Identification

Invertebrate identifications shall be performed by a trained and experienced taxonomist. The taxonomy contractor is responsible for obtaining the most accurate, consistently achievable identifications for ADEQ samples. Specialists are used as needed to obtain identifications to the general taxonomic levels listed in Table 1. The specific identifications are presented on the ADEQ Macroinvertebrate Master Taxa List (Table 2).

Table 1. ADEQ Taxonomic levels of identification for macroinvertebrates.

Invertebrate Group	Level of taxonomy required
Aquatic insects (except the family Chironomidae)	Genus or species, where consistently identifiable
Chironomidae	Family level only
Semi-aquatic insects	Family
Arachnida (Mites)	Genus
Cladocera, Copepoda, Ostracoda	Class
Amphipoda, Decapoda, Isopoda	Class
Nematoda, Nematomorpha	Phylum
Turbellaria	Class
Annelida	Class
Mollusca	Family or Genus

Table 2. ADEQ Macroinvertebrate Master Taxa List.

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Annelida	Hirudinea	Arhynchobdellida	Erpobdellidae					
Annelida	Hirudinea	Pharyngodellida	Erpobdellidae	*Erpobdella*				
Annelida	Hirudinea	Rhynchobdellida	Glossiphoniidae	*Glossiphonia*				
Annelida	Hirudinea	Rhynchobdellida	Glossiphoniidae	*Helobdella*	*stagnalis*	9	PR	
Annelida	Hirudinea					10	PR	Predator
Annelida	Oligochaeta					8	CG	Collector
Arthropoda	Arachnida	Acari				5	PA	PA
Arthropoda	Branchiopoda	Cladocera				8	CF	Filterer
Arthropoda	Copepoda					8	CG	Collector
Arthropoda	Crustacea	Amphipoda						
Arthropoda	Crustacea	Amphipoda	Crangonyctidae	*Crangonyx*				
Arthropoda	Crustacea	Amphipoda	Gammaridae	*Gammarus*				
Arthropoda	Crustacea	Amphipoda	Talitridae	*Hyalella*				
Arthropoda	Crustacea	Decapoda	Astacidae					
Arthropoda	Crustacea	Decapoda	Cambaridae					
Arthropoda	Crustacea	Decapoda	Cambaridae			-99	UN	
Arthropoda	Insecta	Coleoptera	Dryopidae			5	SH	Shredder
Arthropoda	Insecta	Coleoptera	Dryopidae	*Helichus*		5	SH	Shredder
Arthropoda	Insecta	Coleoptera	Dryopidae	*Helichus*				
Arthropoda	Insecta	Coleoptera	Dryopidae	*Helichus*				
Arthropoda	Insecta	Coleoptera	Dryopidae	*Postelichus*		5	SH	Shredder
Arthropoda	Insecta	Coleoptera	Dytiscidae	*Agabus*				
Arthropoda	Insecta	Coleoptera	Dytiscidae	*Deronectes*				
Arthropoda	Insecta	Coleoptera	Dytiscidae					
Arthropoda	Insecta	Coleoptera	Dytiscidae			7	PR	Predator
Arthropoda	Insecta	Coleoptera	Dytiscidae	*Dytiscus*				
Arthropoda	Insecta	Coleoptera	Dytiscidae	*Hydroporinae*				
Arthropoda	Insecta	Coleoptera	Dytiscidae	*Hygrotus*				
Arthropoda	Insecta	Coleoptera	Dytiscidae	*Liodessus*				

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Arthropoda	Insecta	Coleoptera	Dytiscidae	*Neoclypeodytes*				
Arthropoda	Insecta	Coleoptera	Dytiscidae	*Oreodytes*				
Arthropoda	Insecta	Coleoptera	Dytiscidae	*Stictotarsus*				
Arthropoda	Insecta	Coleoptera	Elmidae	*Cleptelmis*		6	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Cleptelmis*				
Arthropoda	Insecta	Coleoptera	Elmidae	*Cylloepus*		5	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Dubiraphia*		8	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae			4	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Heterelmis*		4	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Heterlimnius*		3	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Hexacylloepus*				
Arthropoda	Insecta	Coleoptera	Elmidae	*Huleechius*			UN	
Arthropoda	Insecta	Coleoptera	Elmidae	*Macrelmis*		4	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Microcylloepus*		7	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Narpus*		4	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Neocylloepus*		4	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Neoelmis*		4	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Neoelmis*				
Arthropoda	Insecta	Coleoptera	Elmidae	*Optioservus*		5	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Optioservus*	*divergens*			
Arthropoda	Insecta	Coleoptera	Elmidae	*Xenelmis*		4	SC	Scraper
Arthropoda	Insecta	Coleoptera	Elmidae	*Zaitzevia*		6	SC	Scraper
Arthropoda	Insecta	Coleoptera	Gyrinidae	*Gyretes*		8	PR	Predator
Arthropoda	Insecta	Coleoptera	Gyrinidae	*Gyrinus*				
Arthropoda	Insecta	Coleoptera	Haliplidae					
Arthropoda	Insecta	Coleoptera	Haliplidae	*Haliplus*		8	MH	Shredder
Arthropoda	Insecta	Coleoptera	Haliplidae	*Peltodytes*		8	MH	Shredder
Arthropoda	Insecta	Coleoptera	Hydraenidae	*Hydraena*			UN	
Arthropoda	Insecta	Coleoptera	Hydraenidae				PR	Predator
Arthropoda	Insecta	Coleoptera	Hydraenidae	*Ochthebius*				
Arthropoda	Insecta	Coleoptera	Hydrophilidae	*Berosus*		8	PR	Predator

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Arthropoda	Insecta	Coleoptera	Hydrophilidae	*Enochrus*				
Arthropoda	Insecta	Coleoptera	Hydrophilidae			7	PR	Predator
Arthropoda	Insecta	Coleoptera	Hydrophilidae	*Laccobius*				
Arthropoda	Insecta	Coleoptera	Hydrophilidae	*Tropisternus*				
Arthropoda	Insecta	Coleoptera	Hydroscaphidae	*Hydroscapha*		-99	SC	Scraper
Arthropoda	Insecta	Coleoptera	Hydroscaphidae					
Arthropoda	Insecta	Coleoptera	Lutrochidae	*Lutrochus*		-99	CG	
Arthropoda	Insecta	Coleoptera	Noteridae			-99	PR	Predator
Arthropoda	Insecta	Coleoptera	Psephenidae					
Arthropoda	Insecta	Coleoptera	Psephenidae	*Psephenus*		7	SC	Scaper
Arthropoda	Insecta	Coleoptera	Scirtidae	*Elodes*		-99	OM	OM
Arthropoda	Insecta	Coleoptera	Scirtidae	*Prionocyphon*		-99	OM	OM
Arthropoda	Insecta	Coleoptera	Scirtidae			-99	OM	OM
Arthropoda	Insecta	Coleoptera				-99	UN	UN
Arthropoda	Insecta	Diptera	Athericidae					
Arthropoda	Insecta	Diptera	Athericidae	*Atherix*		7	PR	
Arthropoda	Insecta	Diptera	Blephariceridae	*Agathon*	*arizonica*	3	SC	Scraper
Arthropoda	Insecta	Diptera	Blephariceridae					
Arthropoda	Insecta	Diptera	Brachycera			6	PR	Predator
Arthropoda	Insecta	Diptera	Cecidomyiidae			-99	UN	UN
Arthropoda	Insecta	Diptera	Ceratopogonidae	*Atrichopogon*				
Arthropoda	Insecta	Diptera	Ceratopogonidae	*Bezzia/Palpomyia*				
Arthropoda	Insecta	Diptera	Ceratopogonidae			7	PR	Predator
Arthropoda	Insecta	Diptera	Ceratopogonidae	*Ceratopogoninae*		7	PR	Predator
Arthropoda	Insecta	Diptera	Ceratopogonidae	*Dasyhelea*		7	PR	Predator
Arthropoda	Insecta	Diptera	Ceratopogonidae	*Forcipomyiinae*		7	PR	Predator
Arthropoda	Insecta	Diptera	Ceratopogonidae	*Stilobezzia*				
Arthropoda	Insecta	Diptera	Chironomidae			6	PR	UN
Arthropoda	Insecta	Diptera	Dixidae	*Dixa*		3	CG	Collector
Arthropoda	Insecta	Diptera	Dixidae	*Dixella*		8	CG	Collector
Arthropoda	Insecta	Diptera	Dixidae					

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Arthropoda	Insecta	Diptera	Dixidae	*Meringodixa*		2	CG	Collector
Arthropoda	Insecta	Diptera	Dolichopodidae			6	PR	Predator
Arthropoda	Insecta	Diptera	Elmidae			-99	UN	
Arthropoda	Insecta	Diptera	Empididae	*Chelifera*		6	PR	Predator
Arthropoda	Insecta	Diptera	Empididae	*Clinocera*		6	PR	Predator
Arthropoda	Insecta	Diptera	Empididae			5	PR	Predator
Arthropoda	Insecta	Diptera	Empididae	*Hemerodromia*		6	PR	Predator
Arthropoda	Insecta	Diptera	Empididae	*Neoplasta*				
Arthropoda	Insecta	Diptera	Empididae	*Oreogeton*		1	PR	Predator
Arthropoda	Insecta	Diptera	Empididae	*Trichoclinocera*				
Arthropoda	Insecta	Diptera	Empididae	*Wiedemannia*			UN	
Arthropoda	Insecta	Diptera	Ephydridae			9	CG	Collector
Arthropoda	Insecta	Diptera	Muscidae	*Limnophora*		8	PR	Predator
Arthropoda	Insecta	Diptera	Muscidae			8	PR	Predator
Arthropoda	Insecta	Diptera	Mycetophilidae			-99	UN	UN
Arthropoda	Insecta	Diptera	Nematocera				UN	
Arthropoda	Insecta	Diptera	Psychodidae	*Maruina*		5	SC	Collector
Arthropoda	Insecta	Diptera	Psychodidae	*Pericoma*		5	CG	Collector
Arthropoda	Insecta	Diptera	Psychodidae	*Pericoma/Telmatoscopus*				
Arthropoda	Insecta	Diptera	Psychodidae	*Psychoda*		10	CG	Collector
Arthropoda	Insecta	Diptera	Psychodidae			5	CG	Collector
Arthropoda	Insecta	Diptera	Ptychopteridae	*Ptychoptera*		7	CG	Collector
Arthropoda	Insecta	Diptera	Ptychopteridae					
Arthropoda	Insecta	Diptera	Sciaridae			-99	UN	UN
Arthropoda	Insecta	Diptera	Sciomyzidae			-99	PR	Predator
Arthropoda	Insecta	Diptera	Simuliidae	*Prosimulium*		7	UN	
Arthropoda	Insecta	Diptera	Simuliidae			6	CF	Filterer
Arthropoda	Insecta	Diptera	Simuliidae	*Simulium*			UN	
Arthropoda	Insecta	Diptera	Stratiomyidae	*Caloparyphus*		7	CG	
Arthropoda	Insecta	Diptera	Stratiomyidae	*Euparyphus*		7	CG	
Arthropoda	Insecta	Diptera	Stratiomyiidae			8	CG	Collector

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Arthropoda	Insecta	Diptera	Syrphidae			10	CG	Collector
Arthropoda	Insecta	Diptera	Tabanidae	*Chrysops*				
Arthropoda	Insecta	Diptera	Tabanidae			7	PR	Predator
Arthropoda	Insecta	Diptera	Tabanidae	*Tabanus*				
Arthropoda	Insecta	Diptera	Tanyderidae			1	UN	UN
Arthropoda	Insecta	Diptera	Thaumaleidae			-99	UN	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Tipula*	*gothicana*			
Arthropoda	Insecta	Diptera	Tipulidae	*Antocha*		6	CG	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Cryptolabis*		4	UN	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Dicranota*		6	PR	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Erioptera*		4	CG	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Gonomyia*		4	UN	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Hesperoconopa*		1	UN	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Hexatoma*		5	PR	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Holorusia*		5	SH	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Limnophila*				
Arthropoda	Insecta	Diptera	Tipulidae	*Limonia*		7	MH	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Molophilus*		4	UN	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Paradelphomyia*		4	UN	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Pedicia*		6	PR	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Rhabdomastix*		2	UN	UN
Arthropoda	Insecta	Diptera	Tipulidae	*Tipula*		6	OM	UN
Arthropoda	Insecta	Diptera	Tipulidae			4	UN	UN
Arthropoda	Insecta	Ephemeroptera	Ameletidae	*Ameletus*		0	CG	
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Baetis*	*adonis*			
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Baetis*	*bicaudatus*	4	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Baetodes*	*edmundsi*			
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Diphetor*	*hageni*	5	UN	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Acentrella*	*insignificans*		UN	
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Baetis*	*magnus*			
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Fallceon*	*quilleri*	4	UN	Collector

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Baetis*	*tricaudatus*	4	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Acentrella*	*turbida*	6	UN	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Acentrella*		6	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae			4	UN	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Baetis*		5	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Baetodes*		4	SC	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Baetodes*		-99	UN	
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Callibaetis*		9	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Camelobaetidius*		4	UN	
Arthropoda	Insecta	Ephemeroptera	Baetidae			-99	CG	
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Centroptilum*		6	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Cloeodes*		4	UN	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Diphetor*		5	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Baetidae	*Fallceon*		4	UN	Collector
Arthropoda	Insecta	Ephemeroptera	Caenidae	*Caenis*	*bajaensis*			
Arthropoda	Insecta	Ephemeroptera	Caenidae					
Arthropoda	Insecta	Ephemeroptera	Caenidae	*Caenis*		7	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Ephemerellidae	*Drunella*	*coloradensis/flavilinea*	2	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Ephemerellidae	*Drunella*	*grandis*	2	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Ephemerellidae	*Ephemerella*	*inermis/infrequens*	3	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Ephemerellidae	*Serratella*	*micheneri*	2	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Ephemerellidae	*Serratella*	*tibialis*	2	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Ephemerellidae	*Drunella*		0	SC	
Arthropoda	Insecta	Ephemeroptera	Ephemerellidae	*Drunella*		1	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Ephemerellidae	*Ephemerella*		3	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Ephemerellidae			1	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Epeorus*	*grandis*	0	SC	Scraper
Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Cinygmula*		4	SC	Scraper
Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Epeorus*		1	SC	Scraper
Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Epeorus*				
Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Epeorus*		0	SC	

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Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Epeorus*		-99	SC	
Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Heptagenia*				
Arthropoda	Insecta	Ephemeroptera	Heptageniidae			4	SC	Scraper
Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Leucrocuta*				
Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Nixe*				
Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Rhithrogena*		2	SC	Scraper
Arthropoda	Insecta	Ephemeroptera	Heptageniidae	*Stenonema*		6	SC	Scraper
Arthropoda	Insecta	Ephemeroptera	Isonychiidae	*Isonychia*		2	CF	
Arthropoda	Insecta	Ephemeroptera	Leptohyphidae	*Vacupernius*	*packeri*			
Arthropoda	Insecta	Ephemeroptera	Leptohyphidae	*Asioplax*				
Arthropoda	Insecta	Ephemeroptera	Leptohyphidae	*Homoleptohyphes*				
Arthropoda	Insecta	Ephemeroptera	Leptohyphidae					
Arthropoda	Insecta	Ephemeroptera	Leptohyphidae	*Tricoryhyphes*				
Arthropoda	Insecta	Ephemeroptera	Leptophlebiidae	*Thraulodes*	*speciosus*			
Arthropoda	Insecta	Ephemeroptera	Leptophlebiidae	*Choroterpes*		7	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Leptophlebiidae	*Leptophlebia*		7	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Leptophlebiidae			6	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Leptophlebiidae	*Neochoroterpes*		-99	CG	
Arthropoda	Insecta	Ephemeroptera	Leptophlebiidae			-99	UN	
Arthropoda	Insecta	Ephemeroptera	Leptophlebiidae	*Paraleptophlebia*		4	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Leptophlebiidae	*Thraulodes*		6	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Leptophlebiidae	*Traverella*		6	CF	Collector
Arthropoda	Insecta	Ephemeroptera	Siphlonuridae			7	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Siphlonuridae	*Siphlonurus*		7	OM	Collector
Arthropoda	Insecta	Ephemeroptera	Tricorythidae	*Leptohyphes*		7	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Tricorythidae			7	CG	Collector
Arthropoda	Insecta	Ephemeroptera	Tricorythidae	*Tricorythodes*		7	CG	Collector
Arthropoda	Insecta	Hemiptera	Belostomatidae	*Abedus*		8	PR	Predator
Arthropoda	Insecta	Hemiptera	Belostomatidae					
Arthropoda	Insecta	Hemiptera	Belostomatidae	*Lethocerus*				
Arthropoda	Insecta	Hemiptera	Corixidae			8	PR	Predator

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Arthropoda	Insecta	Hemiptera	Corixidae	*Sigara*				
Arthropoda	Insecta	Hemiptera	Corixidae	*Trichocorixa*				
Arthropoda	Insecta	Hemiptera	Naucoridae	*Ambrysus*	*arizonus*			
Arthropoda	Insecta	Hemiptera	Naucoridae	*Ambrysus*	*circumcinctus*			
Arthropoda	Insecta	Hemiptera	Naucoridae	*Ambrysus*	*melanopterus*			
Arthropoda	Insecta	Hemiptera	Naucoridae	*Ambrysus*	*mormon*			
Arthropoda	Insecta	Hemiptera	Naucoridae	*Ambrysus*	*occidentalis*			
Arthropoda	Insecta	Hemiptera	Naucoridae	*Ambrysus*	*pulchellus*			
Arthropoda	Insecta	Hemiptera	Naucoridae	*Ambrysus*	*puncticollis*			
Arthropoda	Insecta	Hemiptera	Naucoridae	*Ambrysus*	*thermarum*			
Arthropoda	Insecta	Hemiptera	Naucoridae	*Ambrysus*	*woodburyi*			
Arthropoda	Insecta	Hemiptera	Naucoridae	*Ambrysus*		7	PR	Predator
Arthropoda	Insecta	Hemiptera	Naucoridae					
Arthropoda	Insecta	Hemiptera	Notonectidae	*Buenoa*			UN	
Arthropoda	Insecta	Hemiptera	Notonectidae	*Notonecta*		7	PR	
Arthropoda	Insecta	Hemiptera	Notonectidae					
Arthropoda	Insecta	Lepidoptera						
Arthropoda	Insecta	Lepidoptera	Pyralidae	*Petrophila*		6	SC	Shredder
Arthropoda	Insecta	Lepidoptera	Pyralidae					
Arthropoda	Insecta	Megaloptera	Corydalidae					
Arthropoda	Insecta	Megaloptera	Corydalidae	*Corydalus*				
Arthropoda	Insecta	Megaloptera	Corydalidae	*Corydalus*	*cornutus*	6	PR	Predator
Arthropoda	Insecta	Megaloptera	Corydalidae	*Neohermes*				
Arthropoda	Insecta	Megaloptera	Corydalidae	*Neohermes*	*filicornis*	4	PR	Predator
Arthropoda	Insecta	Megaloptera	Sialidae					
Arthropoda	Insecta	Megaloptera	Sialidae	*Sialis*		7	PR	Predator
Arthropoda	Insecta	Odonata	Aeshnidae			5	PR	Predator
Arthropoda	Insecta	Odonata	Aeshnidae	*Anax*		8	PR	Predator
Arthropoda	Insecta	Odonata	Aeshnidae	*Anax*				
Arthropoda	Insecta	Odonata	Aeshnidae	*Oplonaeschna*		5	PR	Predator
Arthropoda	Insecta	Odonata	Calopterygidae	*Hetaerina*	*vulnerata*			

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Arthropoda	Insecta	Odonata	Calopterygidae			5	PR	Predator
Arthropoda	Insecta	Odonata	Calopterygidae	*Hetaerina*		6	PR	Predator
Arthropoda	Insecta	Odonata	Coenagrionidae	*Argia*		7	PR	Predator
Arthropoda	Insecta	Odonata	Coenagrionidae	*Argia*				
Arthropoda	Insecta	Odonata	Coenagrionidae			9	PR	Predator
Arthropoda	Insecta	Odonata	Cordulegastridae	*Cordulegaster*		3	PR	Predator
Arthropoda	Insecta	Odonata	Cordulegastridae					
Arthropoda	Insecta	Odonata	Corduliidae	*Macromia*		2	PR	Predator
Arthropoda	Insecta	Odonata	Gomphidae	*Erpetogomphus*		5	PR	Predator
Arthropoda	Insecta	Odonata	Gomphidae			4	PR	Predator
Arthropoda	Insecta	Odonata	Gomphidae	*Ophiogomphus*		4	PR	Predator
Arthropoda	Insecta	Odonata	Gomphidae	*Progomphus*		4	PR	Predator
Arthropoda	Insecta	Odonata	Lestidae	*Archilestes*		9	PR	Predator
Arthropoda	Insecta	Odonata	Lestidae	*Lestes*		9	PR	Predator
Arthropoda	Insecta	Odonata	Lestidae			9	PR	Predator
Arthropoda	Insecta	Odonata	Libellulidae	*Paltothemis*	*lineatipes*	9	PR	Predator
Arthropoda	Insecta	Odonata	Libellulidae	*Brechmorhoga*	*mendax*	9	UN	UN
Arthropoda	Insecta	Odonata	Libellulidae	*Brechmorhoga*		9	PR	Predator
Arthropoda	Insecta	Odonata	Libellulidae	*Libellula*		8	UN	
Arthropoda	Insecta	Odonata	Libellulidae	*Libellula*				
Arthropoda	Insecta	Odonata	Libellulidae			9	PR	Predator
Arthropoda	Insecta	Odonata	Libellulidae	*Paltothemis*		9	PR	Predator
Arthropoda	Insecta	Odonata	Platystictidae					
Arthropoda	Insecta	Odonata	Zygoptera			-99	UN	
Arthropoda	Insecta	Plecoptera	Capniidae	*Capnia*				
Arthropoda	Insecta	Plecoptera	Capniidae			3	SH	Shredder
Arthropoda	Insecta	Plecoptera	Capniidae	*Mesocapnia*				
Arthropoda	Insecta	Plecoptera	Chloroperlidae			3	PR	Predator
Arthropoda	Insecta	Plecoptera	Chloroperlidae	*Suwallia*		1	PR	
Arthropoda	Insecta	Plecoptera	Chloroperlidae	*Sweltsa*		3	PR	Predator
Arthropoda	Insecta	Plecoptera	Nemouridae	*Zapada*	*cinctipes*	4	SH	Shredder

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Arthropoda	Insecta	Plecoptera	Nemouridae	*Zapada*	*columbiana*	1	SH	Shredder
Arthropoda	Insecta	Plecoptera	Nemouridae	*Zapada*	*frigida*	2	SH	Shredder
Arthropoda	Insecta	Plecoptera	Nemouridae	*Zapada*	*Oregonensis Group*	2	SH	Shredder
Arthropoda	Insecta	Plecoptera	Nemouridae	*Amphinemura*		4	SH	Shredder
Arthropoda	Insecta	Plecoptera	Nemouridae	*Eucapnopsis*				
Arthropoda	Insecta	Plecoptera	Nemouridae	*Malenka*			UN	
Arthropoda	Insecta	Plecoptera	Nemouridae			3	SH	Shredder
Arthropoda	Insecta	Plecoptera	Nemouridae	*Zapada*		2	SH	Shredder
Arthropoda	Insecta	Plecoptera	Perlidae	*Hesperoperla*	*pacifica*	4	PR	Predator
Arthropoda	Insecta	Plecoptera	Perlidae	*Claassenia*	*sabulosa*	4	PR	Predator
Arthropoda	Insecta	Plecoptera	Perlidae	*Anacroneuria*		4	PR	Predator
Arthropoda	Insecta	Plecoptera	Perlidae	*Doroneuria*		2	PR	Predator
Arthropoda	Insecta	Plecoptera	Perlidae	*Hesperoperla*				
Arthropoda	Insecta	Plecoptera	Perlidae			4	PR	Predator
Arthropoda	Insecta	Plecoptera	Perlidae					
Arthropoda	Insecta	Plecoptera	Perlodidae	*Cultus*		2	PR	Predator
Arthropoda	Insecta	Plecoptera	Perlodidae	*Diura*		2	PR	Predator
Arthropoda	Insecta	Plecoptera	Perlodidae	*Diura*				
Arthropoda	Insecta	Plecoptera	Perlodidae	*Isogenoides*				
Arthropoda	Insecta	Plecoptera	Perlodidae	*Isoperla*		2	PR	Predator
Arthropoda	Insecta	Plecoptera	Perlodidae			2	PR	Predator
Arthropoda	Insecta	Plecoptera	Perlodidae	*Skwala*		2	PR	Predator
Arthropoda	Insecta	Plecoptera	Taeniopterygidae	*Taenionema*			UN	
Arthropoda	Insecta	Plecoptera	Taeniopterygidae			2	SH	Shredder
Arthropoda	Insecta	Trichoptera	Apataniidae	*Apatania*		2	SC	UN
Arthropoda	Insecta	Trichoptera	Brachycentridae	*Brachycentrus*	*americanus*	4	SC	Filterer
Arthropoda	Insecta	Trichoptera	Brachycentridae	*Brachycentrus*	*occidentalis*	4	SC	Filterer
Arthropoda	Insecta	Trichoptera	Brachycentridae					
Arthropoda	Insecta	Trichoptera	Brachycentridae	*Brachycentrus*		4	SC	Filterer
Arthropoda	Insecta	Trichoptera	Brachycentridae	*Micrasema*		4	MH	Filterer
Arthropoda	Insecta	Trichoptera	Calamoceratidae					

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Arthropoda	Insecta	Trichoptera	Calamoceratidae	*Phylloicus*		3	SH	Shredder
Arthropoda	Insecta	Trichoptera	Glossosomatidae	*Agapetus*		4	SC	Scraper
Arthropoda	Insecta	Trichoptera	Glossosomatidae	*Culoptila*		6	SC	Scraper
Arthropoda	Insecta	Trichoptera	Glossosomatidae	*Glossosoma*		4	SC	Scraper
Arthropoda	Insecta	Trichoptera	Glossosomatidae					
Arthropoda	Insecta	Trichoptera	Glossosomatidae	*Protoptila*		6	SC	Scraper
Arthropoda	Insecta	Trichoptera	Helicopsychidae	*Helicopsyche*		7	SC	
Arthropoda	Insecta	Trichoptera	Helicopsychidae					
Arthropoda	Insecta	Trichoptera	Hydrobiosidae	*Atopsyche*		-99	PR	Predator
Arthropoda	Insecta	Trichoptera	Hydropsychidae	*Cheumatopsyche*		8	CF	Filterer
Arthropoda	Insecta	Trichoptera	Hydropsychidae	*Hydropsyche*		7	CF	Filterer
Arthropoda	Insecta	Trichoptera	Hydropsychidae			-99	CF	Filterer
Arthropoda	Insecta	Trichoptera	Hydropsychidae	*Leptonema*		4	CF	Filterer
Arthropoda	Insecta	Trichoptera	Hydropsychidae	*Smicridea*		-99	CF	Filterer
Arthropoda	Insecta	Trichoptera	Hydroptilidae			7	PH	PH
Arthropoda	Insecta	Trichoptera	Hydroptilidae	*Zumatrichia*	*notosa*			
Arthropoda	Insecta	Trichoptera	Hydroptilidae	*Alisotrichia*		-99	UN	PH
Arthropoda	Insecta	Trichoptera	Hydroptilidae	*Hydroptila*		7	PH	PH
Arthropoda	Insecta	Trichoptera	Hydroptilidae			6	PH	PH
Arthropoda	Insecta	Trichoptera	Hydroptilidae	*Ithytrichia*		8	SC	PH
Arthropoda	Insecta	Trichoptera	Hydroptilidae	*Leucotrichia*		7	SC	PH
Arthropoda	Insecta	Trichoptera	Hydroptilidae	*Mayatrichia*		7	SC	PH
Arthropoda	Insecta	Trichoptera	Hydroptilidae	*Neotrichia*		7	SC	PH
Arthropoda	Insecta	Trichoptera	Hydroptilidae	*Ochrotrichia*		6	PH	PH
Arthropoda	Insecta	Trichoptera	Hydroptilidae	*Oxyethira*		8	PH	PH
Arthropoda	Insecta	Trichoptera	Hydroptilidae	*Zumatrichia*		7	PH	PH
Arthropoda	Insecta	Trichoptera	Lepidostomatidae	*Lepidostoma*	*acarolum*	4	SH	Shredder
Arthropoda	Insecta	Trichoptera	Lepidostomatidae	*Lepidostoma*	*sp. 1 (sand case)*	4	SH	Shredder
Arthropoda	Insecta	Trichoptera	Lepidostomatidae	*Lepidostoma*	*sp. 2 (turret case)*	4	SH	Shredder
Arthropoda	Insecta	Trichoptera	Lepidostomatidae	*Lepidostoma*		4	SH	Shredder
Arthropoda	Insecta	Trichoptera	Lepidostomatidae					

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Arthropoda	Insecta	Trichoptera	Leptoceridae	*Oecetis*	*disjuncta*			
Arthropoda	Insecta	Trichoptera	Leptoceridae			4	OM	OM
Arthropoda	Insecta	Trichoptera	Leptoceridae	*Mystacides*		5	OM	OM
Arthropoda	Insecta	Trichoptera	Leptoceridae	*Nectopsyche*		7	OM	OM
Arthropoda	Insecta	Trichoptera	Leptoceridae	*Oecetis*		8	OM	OM
Arthropoda	Insecta	Trichoptera	Leptoceridae	*Oecetis*		5	PR	
Arthropoda	Insecta	Trichoptera	Leptoceridae	*Triaenodes*		7	OM	OM
Arthropoda	Insecta	Trichoptera	Limnephilidae	*Onocosmoecus*	*unicolor*	4	OM	UN
Arthropoda	Insecta	Trichoptera	Limnephilidae	*Dicosmoecinae*		3	UN	UN
Arthropoda	Insecta	Trichoptera	Limnephilidae	*Hesperophylax*		7	OM	UN
Arthropoda	Insecta	Trichoptera	Limnephilidae			4	UN	UN
Arthropoda	Insecta	Trichoptera	Limnephilidae	*Limnephilus*		6	SH	UN
Arthropoda	Insecta	Trichoptera	Limnephilidae	*Psychoglypha*		1	CG	UN
Arthropoda	Insecta	Trichoptera	Odontoceridae	*Marilia*		0	OM	OM
Arthropoda	Insecta	Trichoptera	Odontoceridae			0	OM	OM
Arthropoda	Insecta	Trichoptera	Philopotamidae	*Chimarra*		5	CF	Filterer
Arthropoda	Insecta	Trichoptera	Philopotamidae					
Arthropoda	Insecta	Trichoptera	Philopotamidae	*Wormaldia*		4	CF	Filterer
Arthropoda	Insecta	Trichoptera	Polycentropodidae					
Arthropoda	Insecta	Trichoptera	Polycentropodidae	*Polycentropus*		6	PR	Collector
Arthropoda	Insecta	Trichoptera	Polycentropodidae	*Polyplectropus*		6	PR	Collector
Arthropoda	Insecta	Zygoptera	Protoneuridae				UN	
Arthropoda	Insecta	Trichoptera	Psychomyiidae	*Psychomyia*		4	SC	Collector
Arthropoda	Insecta	Trichoptera	Psychomyiidae					
Arthropoda	Insecta	Trichoptera	Psychomyiidae	*Tinodes*		3	SC	Collector
Arthropoda	Insecta	Trichoptera	Rhyacophilidae	*Rhyacophila*	*angelita*	4	PR	Predator
Arthropoda	Insecta	Trichoptera	Rhyacophilidae	*Rhyacophila*	*coloradensis*	5	PR	Predator
Arthropoda	Insecta	Trichoptera	Rhyacophilidae	*Rhyacophila*	*rotunda*	0	PR	Predator
Arthropoda	Insecta	Trichoptera	Rhyacophilidae	*Rhyacophila*		2	PR	Predator
Arthropoda	Insecta	Trichoptera	Rhyacophilidae					
Arthropoda	Insecta	Trichoptera	Sericostomatidae	*Gumaga*		8	SH	Shredder

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Arthropoda	Insecta	Trichoptera	Sericostomatidae					
Arthropoda	Insecta	Trichoptera	Uenoidae	*Neophylax*		3	UN	
Arthropoda	Insecta	Trichoptera	Uenoidae	*Oligophlebodes*		1	SC	
Arthropoda	Insecta	Trichoptera	Uenoidae					
Arthropoda	Insecta	Trichoptera		*Metrichia*		-99	UN	
Arthropoda	Malacostraca	Amphipoda	Gammaridae			6	CG	Collector
Arthropoda	Malacostraca	Amphipoda	Gammaridae	*Gammarus*		6	CG	Collector
Arthropoda	Malacostraca	Amphipoda	Hyalellidae	*Hyalella*	*azteca*	8	CG	
Arthropoda	Malacostraca	Decapoda	Astacidae	*Orconectes*		6	OM	
Arthropoda	Malacostraca	Isopoda				8	CG	Collector
Arthropoda	Ostracoda	Ostracoda				8	CG	Collector
Cnidaria	Hydrozoa	Hydroida	Hydridae	*Hydra*		5	PR	
Mollusca	Bivalvia	Basommatophora	Lymnaeidae			8	CG	Collector
Mollusca	Bivalvia	Basommatophora	Planorbidae			8	SC	Scraper
Mollusca	Bivalvia	Bassommatophora	Physidae	*Physella*		8	CG	
Mollusca	Bivalvia	Mesogastropoda	Hydrobiidae			8	SC	Scraper
Mollusca	Bivalvia	Veneroida	Corbiculidae	*Corbicula*		8	CF	
Mollusca	Bivalvia	Veneroida	Corbiculidae	*Corbicula*				
Mollusca	Bivalvia	Veneroida	Corbiculidae					
Mollusca	Bivalvia	Veneroida	Pisidiidae	*Pisidium*		6	CF	
Mollusca	Bivalvia	Veneroida	Pisidiidae	*Pisidium*				
Mollusca	Bivalvia	Veneroida	Pisidiidae	*Pisidium*				
Mollusca	Bivalvia	Veneroida	Pisidiidae	*Pisidium*				
Mollusca	Bivalvia	Veneroida	Pisidiidae	*Pisidium*				
Mollusca	Bivalvia	Veneroida	Pisidiidae			8	CG	Collector
Mollusca	Gastropoda	Basommatophora	Ancylidae					
Mollusca	Gastropoda	Basommatophora	Lymnaeidae	*Fossaria*				
Mollusca	Gastropoda	Basommatophora	Lymnaeidae	*Pseudosuccinea*				
Mollusca	Gastropoda	Basommatophora	Lymnaeidae	*Radix*				
Mollusca	Gastropoda	Basommatophora	Lymnaeidae	*Stagnicola*				
Mollusca	Gastropoda	Basommatophora	Physidae					

Phylum	Class	Order	Family	Genus	Species	Tolerance Value	Specific Functional Feeding Group	Family Functional Feeding Group
Mollusca	Gastropoda	Basommatophora	Planorbidae	*Gyraulus*				
Mollusca	Gastropoda	Basommatophora	Planorbidae	*Menetus*				
Mollusca	Gastropoda	Bassommatophora	Ancylidae	*Ferrisla*		6	SC	Scraper
Mollusca	Gastropoda	Bassommatophora	Planorbidae	*Gyraulus*		6	SC	Scraper
Mollusca	Gastropoda	Mesogastropoda	Hydrobiidae	*Pyrgulopsis*				
Mollusca	Gastropoda	Mesogastropoda	Valvatidae	*Valvata*		8	SC	
Mollusca	Gastropoda	Mesogastropoda	Valvatidae	*Valvata*				
Mollusca	Gastropoda	Pharyngodellida						
Nematoda						5	UN	UN
Nematomorpha						-99	PA	PA
Nemertea			Hoplonemertea	*Prostoma*				
Platyhelminthes	Turbellaria					4	UN	UN
Porifera						-99	CF	Filterer

Notes to Table 2

1. Functional Feeding Groups are Collector Gatherer (CG), Collector Filterer (CF), Macrophyte Herbivore (MH), Omnivore (OM), Parasite (PA), Predator (PR), Scraper (SC), Shredder (SH), and Unknown (UN).
2. A Tolerance Value of -99 signifies that the value is unknown.

6.10.7 Reference Collection and Storage

A set of reference or voucher specimens shall be prepared from the batch of samples each year for incorporation into ADEQ=s reference specimen collection. Several specimens shall be preserved for each new taxon and the best or largest larval instars of other taxa shall be preserved to represent the taxa found that year and to update the historic reference collection at ADEQ.

The Contractor shall make recommendations for archiving any important specimens, if verification of identification by national specialists is required.

6.10.8 Reporting

Laboratory reports containing taxonomic identifications and counts for all samples for that year shall be submitted to ADEQ in electronic and printed copy. The electronic data shall be submitted in ACCESS database format or Excel spreadsheets formatted for database uploading. The Contractor shall perform quality control checks on the electronic data prior to submittal to ADEQ. The data set should contain at a minimum the Station Identification (ID), waterbody name and location, habitat, collection date, complete taxa ID from phylum to lowest level ID, raw number of individuals, and the portion of sample analyzed including field splits where applicable, and adjusted final counts, which are corrected for sub-sample size and field splits. The printed copy should include the same minimum data recorded in the electronic dataset and should also include a copy of the bench sheets used by the taxonomist to develop the raw counts per sample.

6.10.9 Quality Control Tasks

ADEQ may conduct Laboratory audits as needed. ADEQ may periodically request the laboratory to examine quality control reconstituted samples or voucher specimens from another lab and produce a short letter regarding the accuracy of identifications.

6.10.10 Quality Control Field and Laboratory Procedures

The D-frame dip net, bucket, and sieve should be rinsed and scrubbed with a brush to dislodge small invertebrates, egg masses, and organic material, prior to leaving the site.

Biological field and laboratory quality control procedures and performance characteristics are listed in Table 3.

Table 3. Biological field quality control procedures and performance characteristics.

Procedure	Performance Characteristic	Description
Sampling device	Precision - repeatability in a habitat	The D-frame dip net is a good choice for use in Arizona streams, as it can be used in riffle habitats with virtually all substrate sizes. The precision of sampling with this net is repeatable because a timed sampling effort is used which applies across different stream substrate types.
	Bias - exclusion of certain taxa (mesh size)	The D-frame sampler is outfitted with a 500μ mesh size net opening, which retains organisms of a consistent size for identification.
	Interferences - matrix/physical limitations	Excess filamentous algae can foul a sample, but it is considered part of the organic matter of a sample and is packaged with the biological sample.
Sampling method	Precision - variable metrics or measures among replicate samples at a site	Measurement error is quantified by replicate sampling at 10% of our sampling sites each year. Samples are processed and analyzed separately and their metrics and IBI score compared to obtain a measure of the method precision. This is an estimate of the precision of the entire method which includes variability due to small-scale spatial variability within a site, operator consistency and bias, and laboratory consistency.
	Bias - exclusion of certain taxa or habitats	Riffle only, 500μ mesh size
	Performance range - limitations in certain habitats or substrates	Riffle only, sample edge vegetation for sandy substrates riffles
	Interferences - high river flows, training of personnel	Sampling not performed during high flows for safety reasons. The method has only been tested on a limited basis for large river sampling.
Field Sample Processing	Bias - efficiency of locating small organisms in sample transfer	The sieve is carefully rinsed after straining a sample. The sieve is washed prior to leaving a sample site.
	Performance range - sample preservation and holding time	Sample preserved with isopropanol and capful of formalin for better preservation in Arizona heat. Formalin also allows longer holding time.
	Interferences - Weather conditions	Sample taking maybe performed during light rains and slightly elevated flows, but not during bankfull or greater flows.
	Accuracy - of sample transfer process and labeling	There is a standard format for sample labels which includes stream name, site id, date, habitat sampled, collector info, whether sample was field split and # of jars in sample.

6.11 Bioassessments as Applied to NPDES Permits

The application of bioassessments to NPDES permittees must adhere to the following requirements:

- A bioassessment should occur concurrently with ambient water monitoring
- A bioassessment survey plan should be completed and submitted to ADEQ by December 31st of each year. The plan should contain sample dates, locations of background and study sites, sampling personnel and qualifications, name and location of contract laboratory, biological and habitat sampling protocols and method of analysis.
- ADEQ sampling and analysis protocols should be followed as closely as possible while using the most updated Quality Assurance Program Plan
- Laboratory protocols should follow ADEQ recommendations in Table 4
- The bioassessment report should be submitted to ADEQ for review. The report should contain: an executive summary, introduction, study area description table, including maps and photos, methods, results and discussion, literature cited, and appendices with complete taxa lists and copies of completed field forms for each site. The results and discussion section should cover a physical characterization of the sites, a habitat assessment, water quality, fish and wildlife, macroinvertebrates, and long term trends at the study sites.
- Macroinvertebrate analyses should contain: a list of taxa and abundances, the calculated warm or cold water IBI score, the benthic habitat score, and graphs indicating a comparison of reference and study site IBI scores for the current year, changes in the reference and study IBI scores over a permit period and changes in the reference and study site habitat scores or habitat values over the permit period.
- The first bioassessment shall be subject to a quality assurance review to be conducted by ADEQ. The voucher specimens from the laboratory should be submitted to ADEQ for a quality control review of the taxonomic identifications by the ADEQ contract taxonomist. Major revisions should be incorporated into the final bioassessment report.

Table 4. Biological laboratory quality control procedures and performance characteristics.

Procedure	Performance Characteristic	Description
Laboratory sample processing	Precision - split samples	Duplicate samples are collected at the rate of 10% of the total # of samples during each year=s index period. This is a test of the labs ability to create consistent IDs.
	Bias - sorting certain taxonomic groups or organism size	Large specimens are removed first from the sample. All organisms, regardless of size are sorted for ID from each 1/32 section of the sample.
	Interferences - distractions, equipment	Field and lab equipment, such as sieves and nets, are thoroughly washed between sites and samples.
	Accuracy - sorting method, lab equipment	Caton Tray used for consistent method of sorting samples, especially where thousands of insects per sample are found.
Taxonomic enumeration	Precision - split samples	The similarity of duplicate samples is verified using the Arizona warm water and cold water IBI=s, rather than the individual taxonomic identifications.
	Bias - counts and identifications for certain taxonomic groups	Our taxonomist offers 500 counts of insects per sample, which exceeds the number of specimens counted by many other states. Where a particular taxa is dominant in the sample, that taxon is not included in the 500 count. Our laboratory has used a number of nationally recognized specialists to provide confirmed identifications of specimens for our reference/voucher collection.
	Interferences - appropriateness of taxonomic keys	List of taxonomic keys used by our laboratory is included in our SOP=s.
	Sensitivity - level of taxonomy related to type of stressor	Our standard taxonomic effort (identifications to genus in most cases, with midges at family level) is generally used for all samples. Identification of Chironomidae to genus can be done on an as-needed basis for samples/sites found to be impaired.
	Accuracy - identification and counts	Use of nationally recognized specialists to create the Arizona reference collection, by which all other samples are identified.

6.12 Detailed Discussion of the Clean Sampling Of Natural Waters for Trace Metals (EPA Method 1669

This Standard Operating Procedure is derived from EPA Method 1669. It provides detailed documentation of the procedure not germane to the field procedure which is found in Section 3A, 3.15. The working field procedure is an excerpt from the following text.

6.12.1 Scope and Application

Method 1669 is commonly referred to as "Clean Hands Sampling." This Standard Operating Procedure (SOP) describes collection, handling, filtration, preservation and quality control procedures for sampling natural waters under controlled conditions. The procedure includes the sample handling, analysis, and quality control instructions necessary for reliable determination of trace metals in aqueous samples. Samples will subsequently be analyzed for presence of total recoverable metals and/or dissolved (<0.45 μm) metals. The procedure is designed to prevent the contamination of ambient water samples with the metal(s) of interest and to yield accurate measurements as prescribed by EPA analytical methods listed in Table 5. Implementation of these sampling techniques supports water quality monitoring and permitting programs administered under the Federal Clean Water Act of 1972.

The procedure is applicable to the metals listed below and other metals, metals species, and elements amenable to determination at trace levels; i.e., ambient concentrations in the part-per-trillion (ppt) to low part-per-billion (ppb) range. It is not intended for the analysis of metals from untreated discharges from industrial facilities, in-process waters, landfill leachates, processed inorganic substances and other samples containing mid- to high-level (part-per- thousand to part-per-million) concentrations of the analytes listed below.

Antimony (Sb)	Arsenic (As)	Cadmium (Cd)
Trivalent Chromium (Cr^{+3})	Hexavalent Chromium (Cr^{+6})	Copper (Cu)
Lead (Pb)	Mercury (Hg)	Nickel (Ni)
Selenium (Se)	Silver (Ag)	Thallium (Tl)
Zinc (Zn)		

USEPA - Region 9 has provided ADEQ with three tiers for clean hands sampling. The laboratory providing the sampling equipment may provide a methodology for clean sampling which should be followed. If not provided by the laboratory or the laboratories instructions are incomplete, the procedures outlined in this document are to be followed.

Tier III - Follow Method 1669 protocol as written.

- Required for aqueous Hg, Cr^{+6}, and Cr^{+3}. Wind suits may be required for aqueous Hg, if field contamination exists
- Required for enforcement cases or where data collected will be used for enforcement purposes
- Acid-cleaned bottles constructed of the appropriate material must be stored and transported in double bags

- Sampling equipment must be acid-cleaned
- Must follow clean hands/dirty hands procedures (Section 6.12.8 – 6.12.12
- Water samples must be field-filtered unless laboratory directs otherwise. The laboratory used will give directions; e.g., if received by the lab within 48 hours of collection, laboratory filtering is acceptable. If field filtered, a deionized water (DIW) blank and a filter blank are required and all sample handling, filtering, etc. must be completed inside a clean box.
- Total metals and dissolved metal samples must be acidified at the same time, whether in the field or lab
- A field blank is required
- Rinse bottle with sample water at least three times prior to collection of sample unless directed otherwise by laboratory

Tier II - Less stringent procedure of Method 1669 - This procedure is recommended for sampling in ambient waters with aqueous concentration of target species $\leq$5 ppb or the initial visit to the waterbody.

- Acid-cleaned bottles constructed of the appropriate material must be stored and transported in double bags
- Sampling equipment must be acid-cleaned
- Must follow clean hands/dirty hands procedures (Section 6.12.8 – 6.12.12)
- Water samples should be field-filtered. The laboratory used will give directions; e.g., if received by the lab within 48 hours of collection, laboratory filtering is acceptable. If field filtered, also requires a deionized water (DIW) blank and a filter blank and all sample handling, filtering, etc. must be completed inside a clean box.
- A field blank is required.
- Total metals and dissolved metal samples must be acidified at the same time, whether in the field or lab
- Rinse bottle with sample water at least three times prior to collecting the sample unless directed otherwise by laboratory

Tier I - Less stringent than Tier II - It is recommended for sampling in ambient waters with aqueous concentration target species $\geq$5 ppb or the initial visit to the waterbody.

- Utilizes commercially pre-cleaned bottles (high density polyethylene is acceptable) and sampling equipment
- The clean hands/dirty hands protocol is suggested together with unpowdered dye-free gloves
- Water samples should be field-filtered. The laboratory used will give directions; e.g., if received by the lab within 48 hours of collection, laboratory filtering is acceptable. If field filtered, also requires a deionized water (DIW) blank and a filter blank and all sample handling, filtering, etc. must be completed inside a clean box.
- A field blank is required
- Total metals and dissolved metal samples must be acidified at the same time, either in the field or laboratory
- Rinse bottle or sample equipment with sample water 1 or 2 times prior to collecting the sample

Table 5 Analytical Methods, Metals, and Concentration Levels Applicable to U.S.E.P.A. Method 1669.

Method	Technique	Metal	MDL (µg/L) [1]	ML (µg/L) [2]
1631	Oxidation/Purge & Trap/CVAFS	Mercury	0.0002	0.0005
1632	Hydride AA	Arsenic	0.003	0.01
1636	Ion Chromatography	Chromium(VI)	0.23	0.5
1637	CC/STGFAA	Cadmium	0.0075	0.02
		Lead	0.036	0.1
1638	ICP/MS	Antimony	0.0097	0.02
		Cadmium	0.013	0.1
		Copper	0.087	0.2
		Lead	0.015	0.05
		Nickel	0.33	1
		Selenium	0.45	1
		Silver	0.029	0.1
		Thallium	0.0079	0.02
		Zinc	0.14	0.5
1639	STGFAA	Antimony	1.9	5
		Cadmium	0.023	0.05
		Chromium(III)	0.1	0.2
		Nickel	0.65	2
		Selenium	0.83	2
		Zinc	0.14	0.5
1640	CC/ICP/MS*	Antimony	0.01*	
		Arsenic	0.15	
		Beryllium	0.015	
		Cadmium	0.004	
		Chromium	0.03	
		Cobalt	0.004	
		Copper	0.02	
		Lead	0.005	
		Nickel	0.01	
		Selenium	0.015	
		Thallium	0.005	
		Vanadium	0.02	
		Zinc	0.2	

Table 1 Notes:

1. Method Detection Limit as determined by 40 CFR Part 136, Appendix B.
2. The Minimum Level (ML) is calculated by multiplying laboratory-determined MDL by 3.18 and rounding result to nearest multiple of 1, 2, 5, 10, 20, 50, etc. This is in accordance with procedures used by the USEPA Engineering & Analysis Division (EAD - authors/developers of this sampling method) and described in the EPA Draft National Guidance for the Permitting, Monitoring, and Enforcement of Water Quality-Based Effluent Limitations Set Below Analytical Detection/Quantitation Levels, March 22, 1994.

* MDL for Method 1640 is based upon laboratory work by environmental laboratories using (metal clean) seawater as reagent water. Values represent reasonable and achievable MDL levels.

6.12.2 Partial Equipment List of Materials Unique to Clean Hands Sampling

Some or all of the sampling equipment may be provided by the laboratory, the sampler needs to ensure that the necessary equipment is obtained prior to going to the field.

- Sample bottles, pre-cleaned, constructed of the appropriate material (Teflon or glass if Hg sampling)
- If used, sampling device (DH-81, etc.), acid-cleaned
- Gloves, powderless, dye-free, constructed of the appropriate material
- Storage Bags, zip-type, nonvented, colorless polyethylene (various sizes)
- Plastic sheeting or large bags, colorless polyethylene for constructing or shielding the clean box
- Double-bagged Ice packs to keep samples chilled in the cooler during shipment
- Nonmetallic Cooler with white (dye-less) interior
- Filters, 0.45 µm
- Pump for continuous flow sampling with tubing (Teflon or Teflon-lined, short length of virgin silicone OK for pump section) of sufficient length with connectors
- Preservative ampoules for field preservation (lab will either provide or inform sampler of required preservatives)
- Teflon wash bottles with DI water and high-purity 10% acids (HCl and HNO_3) for rinsing
- Clean Box - either a pre-fabricated or field-constructed work enclosure of sufficient size and construction to allow the necessary tasks (filtering, etc.) to be completed while preventing contamination of the sample
- "Trace-metal grade" reagents must be used in place of "reagent grade" chemicals because acids, bases and other materials labeled "reagent grade" contain concentrations of metals that will interfere in the determination of trace metals at levels listed in Table 6

Table 6. Analytes, preservation requirements, and containers

Metal	Preservation Requirements	Acceptable Containers
Sb, As, Cd, Cu, Pb, Ni, Se, Ag, Tl, Zn	Add 5 ml of 10% HNO_3 to 1-L sample; preserve on-site or immediately upon laboratory receipt.	500 ml or 1 L fluoropolymer, conventional or linear polyethylene, polycarbonate, or polypropylene containers with lid
Cr^{+6}	Add 50% NaOH; preserve immediately after sample collection.	500 ml or 1 L fluoropolymer, conventional or linear polyethylene, polycarbonate, or polypropylene containers with lid
Hg	Total: Add 0.5% high-purity HCl or 0.5% BrCl to pH < 2; Total & Methyl: Add 0.5% high-purity HCl; preserve on-site or immediately upon laboratory receipt	Fluoropolymer or borosilicate glass bottles with fluoropolymer or fluoropolymer-lined caps

Reagent Water - Water in which the analytes of interest and potentially interfering substances are not detected at the Method Detection Limit (MDL) of the analytical method is used for sample analysis. A large carboy or other appropriate container filled with reagent water must be available for the collection of field blanks.

Hydrochloric acid - Dilute (10%), trace-metal grade, shipped with sampling kit for cleaning equipment between samples.

Nitric acid - Dilute (10%), trace-metal grade, shipped with sampling kit for cleaning equipment between samples.

Before samples are collected, all sampling devices and sample containers are cleaned in a laboratory using detergent, mineral acids, and reagent water. Most sampling devices and containers must be constructed of nonmetallic material and free from material that contains metals. Acceptable materials are high- and low-density polyethylene, polycarbonate, or polypropylene, unless mercury is the target analyte when fluoropolymers (e.g., Teflon) are required.

Disposable materials such as gloves, storage bags and plastic wrap may be used new without additional cleaning unless the results of the equipment blank pinpoint any of these materials as a source of contamination. In this case, either a different supplier must be obtained or the materials must be cleaned.

Avoid use of Pyrex, Kimax, methacrylate, polyvinylchloride, nylon, and Vycor materials. In addition, avoid material such as colored plastics, paper cap liners, pigments used to mark increments on plastics and rubber, all of which may contain trace levels of metals.

Samples may easily be contaminated by airborne dust, dirt, particulate matter, or vapors from automobile exhaust; cigarette smoke, nearby corroded or rusted bridges, pipes, poles, or nearby

roads, and even human breath (mercury amalgam in dental fillings). Muddy rain gear has the potential to contaminate samples, so it must be free of dirt and particulate matter prior to sampling.

Flexible tubing for use with a peristaltic pump should be constructed of styrene/ethylene/ butylene/silicone (SEBS) resin. Only new, laboratory-cleaned tubing shall be used. Tubing connectors, used to connect multiple lengths of tubing, shall be constructed of clear polyethylene or fluoropolymer and also cleaned by the laboratory. The tubing and connectors are individually double-bagged in clear polyethylene bags and stored until use.

Hard case tubing for connection to peristaltic flexible pump tubing shall be constructed of fluoropolymer in lengths as required to reach the point of sampling.

It cannot be overemphasized as to the ease of contaminating ambient water samples with the metal(s) of interest and/or interfering substances. The only way to measure the performance of these clean techniques is through the collection and analysis of uncontaminated blank samples in accordance with this guidance and the methods in Table 5. Thus it is a performance based sampling method; it is highly recommended that any modifications be thoroughly evaluated and demonstrated to be effective before ambient water samples are collected.

The terms "clean" and "ultraclean" have been used in other Agency guidance to describe the techniques needed to reduce or eliminate contamination in trace metals determinations. These terms are not used in this sampling method due to a lack of exact definitions.

Procedures outlined below are for use only by personnel thoroughly trained in the collection of samples for determination of metals at ambient water quality concentrations.

6.12.3 Method Summary

This SOP describes field sampling procedures commonly referred to as "clean hands/dirty hands" techniques. Upon arrival at the sampling site, one member of the two-person sampling team is designated as "dirty hands"; a second member is designated as "clean hands." All operations involving contact with the sample bottle and transfer of the sample from the sample collection device to the sample bottle are handled by the individual designated as clean hands. Dirty hands is responsible for preparation of the sampler (except the sample container itself), operation of any machinery; e.g., pump, and for all other activities that do not involve direct contact with the sample.

The clean hands/dirty hands technique has been included and adapted for three methods to collect natural waters samples. Manual sampling is the simplest and requires the least equipment. Subsurface samples are collected by either grab immersion sampling or pumping the sample to the surface. Grab sampling with a specialized device can be used to collect discrete samples and can conveniently obtain samples at several depths. Continuous flow sampling via pump systems is used to collect numerous samples at various depths. Field filtering and decontaminating equipment in the field are also included in this SOP.

Trace metal clean sampling does not require sophisticated sampling equipment. It does require metal-free sampling apparatus and sufficient training of personnel to modify collection procedures

to maintain sample integrity; e.g., minimize contamination sources. To achieve low detection levels in ambient water samples, personnel must complete significant preparation in the laboratory prior to traveling to the sampling site. Also, they must evaluate the environmental conditions which may affect sampling free of contamination. For example, whenever possible, samples are collected facing upstream and upwind to minimize introduction of contamination. Another example is to manually fill a sample bottle by rapid immersion in water and capping underwater to minimize exposure to airborne particulate matter.

Where this protocol calls for field filtration to collect samples for dissolved metals, unpreserved samples are filtered through a 0.45 µm capsule filter at the field site. After filtering, preservative is added and the samples are double-bagged and iced immediately. Sample containers are shipped or delivered to the analytical laboratory as soon as possible.

Acid preservation of samples (unfiltered and filtered) can be performed either in the field or the laboratory. The sampling team may prefer to utilize laboratory preservation of samples to expedite field operations and to minimize the potential for sample contamination since it can be performed under HEPA filters inside a Class 100 clean bench. Samples can be field preserved inside a glove box designed to provide a particulate free environment.

The laboratory is responsible for generating a acceptable equipment blank to demonstrate the sampling equipment and most notably sample containers are free from trace metals contamination before used by the field sampling team. An acceptable blank is one that is free from contamination below the quantitation level specified in the applicable 1600-series trace metal analytical method.

Quality control samples must be collected while sampling in the field. This includes equipment blanks, field blanks and field splits. The laboratory or cleaning facility must prepare a large carboy or other appropriate clean container filled with reagent water for use with collection of field blanks during sampling activities. The reagent-water-filled container should be shipped to the field site and handled the same as all other sample containers and sampling equipment.

6.12.4 Sample Containers, Handling, Preservation and Storage

Throughout this method, the sample containers, sampling devices, instrumentation, and all other materials and devices used in sample collection, sample processing, and sample analysis activities will be referred to collectively as the apparatus.

Before samples are collected, all sampling devices and sample containers are cleaned in a laboratory using detergent, mineral acids, and reagent water. Most sampling devices and containers must be constructed of nonmetallic material and free from material that contains metals. Acceptable materials are high- and low-density polyethylene, polycarbonate, or polypropylene, unless mercury is the target analyte when fluoropolymers (FEP, PFA or PTFE) are required. Details on material construction can be found in section 5. Sample containers must be acid-cleaned and prepared for field use in a class 100 clean room. After cleaning, sample containers may be filled with weak acid solution, individually double-bagged, and shipped to the sampling site. All sampling equipment is also bagged for storage or shipment.

Samples collected for dissolved metal determinations are to be filtered in the field (Section 6.12.12). They can be preserved in the field or in the laboratory, in either case, at the same time as the total metals samples are preserved.

6.12.5 Contamination Problems and Interferences

Much of the historical metals data in samples from natural waters are inaccurate due to contamination from sampling methods or containers. In the mid-1970's, when analytical chemists devised new techniques and applied them to environmental sampling, then it was revealed much of the ambient waters trace metals data were erroneously high. Preventing contamination of aqueous samples during the sampling, handling and analytical process is the greatest challenge faced in accurate trace metals determinations. Samplers must use extreme care to reduce contamination and avoid compromising the integrity of ambient water samples.

Thorough presentation of requirements and suggestions for controlling sample contamination are given in EPA Sampling Method 1669 and EPA analytical methods (e.g., Method 1640) listed in Table 5.

6.12.5.1 Contamination Control

There are numerous routes by which samples may become contaminated. Potential sources of trace metals contamination during sampling include metallic or metal- containing sampling equipment, containers, lab ware (e.g. powdered gloves that contain high levels of zinc), reagents, and deionized water; improperly cleaned and stored equipment, lab ware, and reagents; and atmospheric inputs such as dirt and dust from automobile exhaust, cigarette smoke, nearby roads, bridges, wires, and poles. Even human contact (hair or dental work) can be a source of trace metal contamination.

When possible obtain and use equipment that is completely free of the metal(s) of interest. Only fluoropolymer (FEP, PFA), conventional or linear polyethylene, polycarbonate, polysulfone, polypropylene, or ultrapure quartz should come in contact with the samples. PFA is less desirable than FEP due to expense. Regardless of construction, all materials that will directly or indirectly contact the sample must be cleaned prior to use.

Avoid exposure of the sample and sampling apparatus by performing operations in an area known to be free from contamination. Two of the most important factors are: (1) awareness of potential sources of contamination and (2) strict attention to work being performed.

Contact or expose samples or blanks only in a clean room, under a clean bench, inside a glove box, or clean plastic bag, so that exposure to atmospheric inputs is minimized. When not being used, the apparatus should be covered with clean plastic wrap, stored in the clean bench or in a plastic box or glove box, or bagged in clean, colorless zip-type bags (most convenient).

Personnel must wear clean, powderless, dye-free gloves while handling the apparatus, samples and blanks. If another object or substance is touched, the glove(s) must be changed before handling the apparatus again.

At sites where more than one sample will be collected, the sample known or expected to contain the lowest concentration of metals should be collected first with the sample containing the highest levels collected last. This will help minimize carryover of metals from high- concentration samples

to low-concentration samples. Without prior knowledge of the waterbody, sampling team should rinse the collection equipment with dilute acid and reagent water between samples and then collect a field blank.

An apparatus that may not directly contact samples may still be a source of contamination. For example, clean tubing placed in a dirty plastic bag may pick up contamination from the bag and subsequently transfer the contamination to the sample. Therefore, it is imperative that every piece of the Apparatus that is directly or indirectly used in the collection of ambient water samples be cleaned as specified in the analytical method(s) referenced in Table 5.

Samples may easily be contaminated by airborne dust, dirt, particulate matter, or vapors from automobile exhaust; cigarette smoke; nearby corroded or rusted bridges, pipes, poles, or nearby roads; and even human breath (mercury amalgam in dental fillings). Muddy rain gear has the potential to contaminate samples, so it must be free of dirt and particulate matter prior to sampling. This is also true of dried sediment particles inside a boat, wash the boat prior to sampling whenever possible. Areas where nearby soil is bare and subject to wind erosion should be avoided.

This sampling method is not intended for application to samples containing high concentrations (part per thousand to part per million) of metals such as untreated effluents, landfill leachates or process waters. Such high level samples must NOT be collected, processed, or shipped with ambient samples being collected for trace metals determinations.

Interferences resulting from samples will vary considerably from source to source, depending on the diversity of the site being sampled. If a sample is suspected of containing substances that may interfere in the determination of trace metals, sufficient sample should be collected to allow the laboratory to identify and overcome interference problems.

While contamination control is essential, personnel health and safety remain the highest priority.

6.12.6 Equipment/Apparatus

All sampling equipment and sample containers must be precleaned in a laboratory or cleaning facility, before they are shipped to the field site. Clean equipment should be packaged and arranged to minimize field preparation.

Here is a sample partial checklist of sampling apparatus; additional comments for most items appear later in this section.

- Sample Bottles, pre-cleaned, constructed of the appropriate material
- Gloves, powderless, dye-free, constructed of the appropriate material; wrist and shoulder-length
- Storage Bags, zip-type, nonvented, colorless polyethylene (various sizes)
- Plastic Wrap, colorless polyethylene
- Double bagged ice packs to keep samples chilled in the cooler during shipment
- Cooler, nonmetallic, with white interior for shipping samples
- Filters, 0.45 µm

- Pump for continuous flow sampling
- Tubing of sufficient length with connectors
- Carboy for collection and storage of dilute waste acids
- Preservative ampoules (optional) for field preservation
- Fluoropolymer wash bottles with DI water and high-purity 10% acids (HCl and HNO3) for rinsing
- Metal-free sampling pole for extension of sampling tubing away from the boat gunwhales, bridge railings, etc.
- Field-Portable Glove Bag (polyethylene)

When possible obtain and use equipment that is completely free of the metal(s) of interest. Only fluoropolymer, conventional or linear polyethylene, polycarbonate, polysulfone, polypropylene, or ultrapure quartz should come in contact with the samples. Fluoropolymer (FEP, PFA, or PTFE) or glass containers should be used for samples that will be analyzed for mercury because mercury vapors can diffuse in or out of other materials, resulting either in contamination or low-biased results. Avoid use of Pyrex, Kimax, methacrylate, polyvinylchloride, nylon, and Vycor. In addition, avoid material such as colored plastics, paper cap liners, pigments used to mark increments on plastics and rubber, all of which may contain trace levels of metals. PTFE is less desirable than FEP because the sintered material in PTFE may contain contaminants and is susceptible to serious memory effects.

Disposable materials such as gloves, storage bags, and plastic wrap, may be used new without additional cleaning unless the results of the equipment blank pinpoint any of these materials as a source of contamination. In this case, either a different supplier must be obtained or the materials must be cleaned.

Sample bottles should be constructed of fluoropolymer (FEP, PFA), conventional or linear polyethylene, polycarbonate, or polypropylene. If mercury is a target analyte, FEP bottles must be used. Previously cleaned sample bottles should be filled with 0.1% HCl (v/v); individually stored in double plastic bags. In most cases, it may be possible to empty the weak acid solution from the sample bottle immediately prior to transport to the field site.

Surface samples are collected using a grab sampling technique. Samples may be collected manually by direct submersion of the bottle into the water or by using a grab sampling device.

Subsurface sample collection may be appropriate in lakes and sluggish deep river environments or where depth profiling is determined to be necessary. Subsurface samples are collected by continuous-flow pumping of water into a sample bottle. Peristaltic pumps do not require cleaning; however, touching the metal head and metal controls necessitates changing of gloves before touching the apparatus. If a submersible pump is used, a large volume of sample should be pumped to clean the stainless steel shaft (hidden behind the impeller) that comes in contact with the sample. Pumps with metal impellers should not be used.

Continuous-flow sampling consisting of a peristaltic or submersible pump and tubing and may have a filter added to the sampling train immediately prior to sampling for dissolved metals.

Flexible tubing for use with peristaltic pump should be constructed of styrene/ethylene/butylene/ silicone (SEBS) resin. Only new, laboratory-cleaned tubing shall be used. Field decontamination is

not possible. Tubing connectors, used to connect multiple lengths of tubing, shall be constructed of PVC, clear polyethylene, or fluoropolymer and also cleaned by the laboratory. The tubing and connectors are individually double-bagged in clear polyethylene bags and stored until use.

Hard case tubing for connection to peristaltic flexible pump tubing shall be constructed of fluoropolymer in lengths as required to reach the point of sampling. If sampling will be at some depth from the end of a boom extended from a boat, bridge or other object, sufficient tubing to extend to the end of the boom and to the depth will be required.

For most sampling situations; i.e., most metals under most conditions, the use of a boat is acceptable. Immediately before use, the boat should be washed to remove any dust or dirt accumulation. All samples should be collected upstream of boat movement and upwind of any fossil fuel exhaust.

When sampling for mercury, a fiberglass boat with battery-powered engine and wooden or fiberglass oars is preferred. The boat should be washed and stored in an area that minimizes exposure to dust and atmospheric particles. If a motor is required, it should be shut off at a distance far enough from the sampling point to avoid contamination, then the sampling team should manually propel the boat to the sampling point. Personnel may wear an unlined, long-sleeved wind suit consisting of pants and jacket and constructed of nylon or other synthetic fiber to prevent mercury adsorbed onto cotton or other clothing materials from contaminating samples.

6.12.7 Reagents

"Trace-metal grade" reagents must be used in place of "reagent grade" chemicals because acids, bases and other materials labeled "reagent grade" contain concentrations of metals that will interfere in the determination of trace metals at levels listed in Table 5.

- Reagent Water - Water in which the analytes of interest and potentially interfering substances are not detected at the Method Detection Limit (MDL) of the analytical method used for analysis of samples. A large carboy or other appropriate container filled with reagent water must be available for the collection of field blanks
- Hydrochloric acid - Dilute (10%), trace-metal grade, shipped with sampling kit for cleaning equipment between samples
- Nitric acid - Dilute (10%), trace-metal grade, shipped with sampling kit for cleaning equipment between samples

6.12.8 Procedures

6.12.8.1 Site Selection-- Overview of Sampling Considerations

Selection of a representative site for surface water sampling is based on many factors including: study objectives, water use, point source discharges, non- point source discharges, tributaries, changes in stream characteristics, types of stream bed, stream depth, turbulence, and the presence of structures (bridges, dams, etc.). When collecting samples to determine ambient levels of trace

metals, the presence of potential sources of metal contamination are of extreme importance in site selection.

Ideally, the selected sampling site will exhibit a high degree of cross-sectional homogeneity. It may be possible to use previously collected data to identify locations for samples that are well mixed or are vertically or horizontally stratified. Since mixing is principally governed by turbulence and water velocity, the selection of a site immediately downstream of a riffle area will ensure good vertical mixing. Horizontal mixing occurs in constrictions in the channel. In the absence of turbulent areas, the selection of a site that is clear of immediate point sources, such as industrial effluents, is preferred for the collection of ambient water samples.

To minimize contamination from trace metals in the atmosphere, ambient water samples should be collected from sites that are as far as possible (e.g., at least several hundred feet) from any metal supports, bridges, wires or poles. Similarly, samples should be collected as far as possible from regularly or heavily traveled roads. If it is not possible to avoid collection near roadways, it is advisable to study traffic patterns and plan sampling events during lowest traffic flow.

The sampling activity should be planned to collect samples known or suspected to contain the lowest concentrations of trace metals first, finishing with the samples known or suspected to contain the highest concentrations. For example, if samples are collected from a flowing river or stream near an industrial or municipal discharge, the upstream sample should be collected first, the downstream sample collected second, and the sample nearest the discharge collected last. If the concentrations of pollutants is not known and cannot be estimated, it is necessary to follow clean sampling protocols at each sampling location.

When sampling from a boat, the bow of the boat should be oriented into the current (the boat will be pointed upstream). Ideally, the team should approach the site from down current and downwind to prevent contamination of the sample by particles sloughing off the boat or equipment. All sampling activity should occur from the bow, preferably pointed upwind and upstream.

If the samples are being collected from a boat, it is recommended that the sampling team create a stable workstation by arranging the cooler or shipping container as a work table on the upwind side of the boat, covering this worktable and the upwind gunnel with plastic wrap or a plastic tablecloth, and draping the wrap or cloth over the gunnel. If necessary, duct tape is used to hold the wrap or cloth in place.

Although the duties of clean hands and dirty hands would appear to be a logical separation of responsibilities, in fact, the completion of the entire protocol may require a good deal of coordination and practice. For example, dirty hands must open the box or cooler containing the sample bottle and unzip the outer bag; clean hands must reach into the outer bag, open the inner bag, remove the bottle, collect the sample, replace the bottle lid, put the bottle back into the inner bag, and zip the inner bag dirty hands must close the outer bag and place it in a cooler.

All operations involving contact with the sample bottle and with transfer of the sample from the sample collection device to the sample bottle (if the sample is not directly collected in the bottle) are handled by the individual designated as "clean hands." Dirty hands is responsible for all activities that do not involve direct contact with the sample; e.g., sample documentation.

Extreme care must be taken during all sampling operations to minimize exposure of the sample to human, atmospheric, and other sources of contamination. For mercury sampling, samplers should avoid breathing directly on the sample.

Before heading off to the sampling site, the sampling team should consider the type of sample to be collected, the amount of sample needed, and the devices to be used (grab, surface, or subsurface samplers). Sufficient sample volume should be collected to allow for necessary quality control analyses and various blanks.

6.12.9 Manual Sampling

The collection of surface samples directly into the sample bottle. At the site, all sampling personnel must put on clean gloves before commencing sample collection activity. Note that clean hands should put on shoulder-length polyethylene gloves and dirty hands should put on wrist length PVC gloves.

If samples are to be analyzed for mercury, the sampling team must also put their precleaned wind suits on at this time.

1. Dirty hands must open the cooler or storage container, remove the double- bagged sample bottle from storage, and unzip the outer bag.
2. Next, clean hands opens the inside bag containing the sample bottle, removes the bottle, and reseals the inside bag. Dirty hands then reseals the outer bag.
3. Clean hands unscrews the cap and, while holding the cap upside down, discards the dilute acid solution from the bottle into a carboy for wastes or discards the reagent water directly into the water body.
4. Clean hands then submerges the sample bottle, and allows the bottle to partially fill with sample. Clean hands screws the cap on the bottle, shakes the bottle several times, and empties the rinseate away from the site. After two more rinsings, clean hands holds the bottle under water and allows bottle to fill with sample. After the bottle has filled (i.e., when no more bubbles appear), and while the bottle is still inverted so that the mouth of the bottle is underwater, clean hands replaces the cap of the bottle. In this way, the sample has never contacted the air.
5. Once the bottle lid has been replaced, dirty hands reopens the outer plastic bag, and clean hands opens the inside bag, places the bottle inside it, and zips the inner bag.
6. Dirty hands zips the outer bag.
7. If the sample is to be analyzed for dissolved metals, it is filtered in accordance with the procedure described in Section 6.12.12.
8. Documentation After each sample is collected, the sample number is documented in the sampling log, and any unusual observations concerning the sample and the sampling are documented.

6.12.10 Grab Sampling

The collection of surface water samples with a grab sampling device. The following steps detail "clean hands/dirty hands" sample collection technique using a grab sampling device.

The sampling team puts on gloves. Ideally, a sample bottle will have been pre-attached to the sampling device in the laboratory. If it is necessary to attach a bottle to the device in the field, clean hands performs this operation inside the field-portable glove bag.

1. Dirty hands removes the sampling device from its storage container and opens the outer polyethylene bag.

2. Clean hands opens the inside polyethylene bag and removes the sampling device.
3. Clean hands changes gloves.
4. Dirty hands submerges the sampling device to the desired depth.
5. When the bottle is full, dirty hands removes the sampling device from the water.
6. Dirty hands returns the sampling device to its large inner plastic bag, clean hands pulls the bottle out of the collar, unscrews the bottle from the sealing device, and caps the bottle; clean hands and dirty hands then return the bottle to its double-bagged storage.

6.12.11 Continuous Flow Sampling

The sampling of surface water using a pump for sampling through the pre-cleaned tubing is performed as follows.

1. Before putting on wind suits or gloves, the sampling team removes the bags containing the pump, tubing, batteries, gloves, plastic wrap, wind suits, and, if samples are to be filtered, the filtration apparatus from the coolers or storage containers in which they are packed.
2. Clean hands and dirty hands put on PVC gloves.
3. Dirty hands removes the pump from its storage bag, and opens the bag containing the tubing.
4. Clean hands installs the tubing while dirty hands holds the pump. Clean hands immerses the inlet end of the tubing in the sample stream.
5. Both clean hands and dirty hands change gloves. Clean hands also puts on shoulder length polyethylene gloves.
6. Dirty hands turns the pump on and allows the pump to run for 5-10 minutes or longer to purge the pump and tubing.
7. If the sample is to be filtered, clean hands installs the pre-cleaned cartridge filter at the end of the tubing.

   NOTE: The filtration apparatus is not attached while flushing the sample tubing with ambient water, rather wait until immediately before sampling to prevent buildup of particulates from clogging the filter.
8. The sample is collected by rinsing the sample bottle and cap three times and collecting the sample from the flowing stream.

6.12.12 Field Filtering

The filtration procedure described below is used for samples collected using the manual or grab collection systems. This setup describes in-line filtration using the peristaltic pump or continuous-flow approach because this setup is optimal for reducing contamination. Tubing and filters must be changed between samples.

1. Set up the filtration system inside the glove bag, using the shortest piece of pump tubing as is practicable. Place the peristaltic pump immediately outside of the glove bag and poke a small hole in the glove bag for passage of the tubing.
2. Clean hands removes the water sample from the inner storage bag and places the sample inside the glove bag. Clean hands also places two clean empty sample bottles, a bottle containing reagent water, and a bottle for waste in the glove bag.
3. Clean hands removes the lid of the reagent water bottle and places the end of the pump tubing in the bottle.
4. Dirty hands starts the pump and passes approximately 200 ml of reagent water through the tubing and filter into the waste bottle. Clean hands then moves the outlet tubing to a clean bottle and collects the remaining reagent water as a blank. Dirty hands stops the pump.
5. Clean hands removes the lid of the sample bottle and places the intake end of the tubing in the bottle.
6. Dirty hands starts the pump and passes approximately 50 ml through the tubing and filter into the remaining clean sample bottle and then stops the pump. Clean hands uses the filtrate to rinse the bottle, discards the waste sample, and returns the outlet tube to the sample bottle. Repeat at least twice.
7. Dirty hands starts the pump and the remaining sample is processed through the filter and collected in the sample bottle. If preservation is required, preservative; e.g., acid, is added to the sample by clean hands at this point.
8. Clean hands replaces the lid on the bottle, returns the bottle to the inside bag, and zips the bag. Clean hands then places the zipped bag into the outer bag held by "dirty hands."
9. Dirty hands zips the outer bag, and places the double-bagged sample bottle into a clean, ice-filled cooler for immediate shipment to the laboratory.

6.12.13 Decontaminating Equipment in the Field

Ideally sampling activity can be planned so that sufficient equipment is brought to the field that field decontamination of the sampling equipment between samples in unnecessary. If it is not possible to plan sampling activity in this manner, dedicated sampling equipment should be provided for each sampling event.

1. If samples are collected from adjacent sites (e.g., immediately upstream or downstream), rinsing of the sampling apparatus with water that is to be sampled should be sufficient.
2. If it is necessary to cross a gradient (i.e., going from a high-concentration sample to a low-concentration sample), such as might occur when collecting at a second site, the following procedure may be used to clean the sampling equipment between samples:
    a. Inside the glove bag, use the "clean hands/dirty hands" procedure to process the dilute nitric acid solution through the apparatus. Dump the spent dilute acid in the waste carboy or in the waterbody away from the sampling point.

b. Process 1 L of reagent water through the apparatus to rinse the equipment and discard the spent water.

c. Collect a field blank as described in Section 6.12.14.2.

d. Rinse the apparatus with copious amounts of the ambient water sample and proceed with sample collection.

6.12.14 Quality Assurance/Quality Control

The sampling team shall employ a strict quality assurance/ quality control (QA/QC) program. The team must collect equipment blanks, field splits, field blanks and QC samples.

The sampling team is permitted to modify the sampling techniques described in this method to improve performance or reduce sampling costs, provided that reliable analyses of samples are obtained and that samples and blanks are not contaminated. Each time a modification is made to the procedures, the sampling team is required to demonstrate that the modification does not result in contamination of field and equipment blanks.

6.12.14.1 Equipment Blank

Before using any sampling equipment at a given site, the laboratory is required to generate equipment blanks to demonstrate that the equipment is free from contamination. Equipment blanks must be run on all equipment that will be used in the field. If, for example, samples are to be collected using both a grab sampling device and the jar sampling device, then an equipment blank must be run on both pieces of equipment. Two types of equipment blanks are required: bottle blanks and sampling equipment blanks.

Equipment blanks are generated in the laboratory by processing reagent water through the equipment using the same procedures that are used in the field. Therefore, the "clean hands/dirty hands" technique used during field sampling should be followed when preparing equipment blanks at the laboratory. Sampling personnel must collect a clean equipment blank before performing on-site field activities.

Detailed procedures for collecting equipment blanks are given in the analytical methods referenced in Table 5. If any metal(s) of interest or any potentially interfering substance is detected in the equipment blank at the minimum level specified in the referenced method, the source of contamination/interference must be identified and removed.

6.12.14.2 Field Blank

To demonstrate that sample contamination has not occurred during field sampling and sample processing, at least one field blank must be generated for every 10 samples that are collected, but no less than one per day. Field blanks are collected before sample collection.

Field blanks are generated by filling a large carboy or other appropriate container with reagent water in the laboratory, transporting the filled container to the sampling site, processing the water through each of the sample processing steps and equipment (e.g., tubing, sampling devices, filters, etc.) that will be used in the field, collecting the field blank in one of the sample bottles, and shipping the bottle to the laboratory for analysis in accordance with the method(s) referenced in Table 5. However, manual grab sampler field blanks are represented by the field filtration blank which is more likely to introduce contamination. Subsurface sampler field blanks are collected by immersing the tubing into the water and pumping water into a sample container. Filter the field blanks using the procedures described in Section 6.12.12. If it is necessary to decontaminate the sampling equipment between samples, a field blank should be collected after the cleaning procedures but before the next sample is collected.

6.12.14.3 Field Split

To assess the precision of the field sampling and analytical processes, at least one field split sample must be collected for every 10 samples that are collected during a given event.

The field split is collected by splitting a larger volume (a single container which is filled following clean hands/dirty hands procedures) into two aliquots in the glove box. Using a churn splitter inside a glove bag is normally not practical and will be avoided. Field splits for dissolved metals determinations must be processed using the procedures in Section 6.12.12.

6.12.15 Health and Safety

When working with potentially hazardous materials, follow U.S. EPA, OSHA and specific health and safety procedures.

Decontamination can pose hazards under certain circumstances even though performed to protect health and safety. Hazardous substances may be incompatible with decontamination methods. For example, the decontamination solution or solvent may react with contaminants to produce heat, explosion, or toxic products. Decontamination methods may be incompatible with clothing or equipment; some solvents can permeate or degrade protective clothing. Also, decontamination solutions and solvents may pose a direct health hazard to workers through inhalation or skin contact, or if they combust.

6.12.16 Method Sources

USEPA Method 1669, Sampling Ambient Water for Determinations of Metals at EPA Water Quality Criteria Levels (undated).

USEPA Region 9 Laboratory; Richmond, California; Standard Operating Procedure #1229; (undated).

USEPA. Draft National Guidance for the Permitting, Monitoring, and Enforcement of Water Quality-Based Effluent Limitations Set Below Analytical Detection/Quantitation Levels, March 22, 1994.