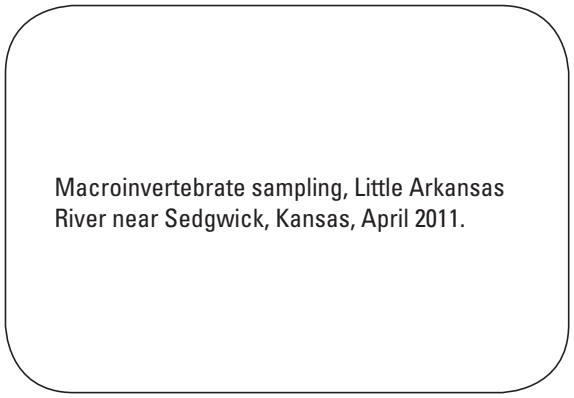
USGS
science for a changing world

Prepared in cooperation with the city of Wichita, Kansas

Protocols for Collection of Streamflow, Water-Quality, Streambed-Sediment, Periphyton, Macroinvertebrate, Fish, and Habitat Data to Describe Stream Quality for the Hydrobiological Monitoring Program, *Equus* Beds Aquifer Storage and Recovery Program, City of Wichita, Kansas

Open-File Report 2012–1055

U.S. Department of the Interior
U.S. Geological Survey



Macroinvertebrate sampling, Little Arkansas River near Sedgwick, Kansas, April 2011.

Bridge water-quality sampling, Little Arkansas River near Sedgwick, Kansas, June 2010.

Cover photograph index

Streambed-sediment sampling Little Arkansas River near Sedgwick, Kansas, April 2011.

Cover background photograph;
Little Arkansas River near Sedgwick, Kansas, August 2011.

Protocols for Collection of Streamflow, Water-Quality, Streambed-Sediment, Periphyton, Macroinvertebrate, Fish, and Habitat Data to Describe Stream Quality for the Hydrobiological Monitoring Program, *Equus* Beds Aquifer Storage and Recovery Program, City of Wichita, Kansas

By Mandy L. Stone, Teresa J. Rasmussen, Trudy J. Bennett, Barry C. Poulton, and Andrew C. Ziegler

Prepared in cooperation with the city of Wichita, Kansas

Open-File Report 2012–1055

U.S. Department of the Interior
U.S. Geological Survey

U.S. Department of the Interior
KEN SALAZAR, Secretary

U.S. Geological Survey
Marcia K. McNutt, Director

U.S. Geological Survey, Reston, Virginia: 2012

Suggested citation:
Stone, M.L., Rasmussen, T.J., Bennett, T.J., Poulton, B.C., and Ziegler, A.C., 2012, Protocols for collection of stream-flow, water-quality, streambed-sediment, periphyton, macroinvertebrate, fish, and habitat data to describe stream quality for the Hydrobiological Monitoring Program, *Equus* Beds Aquifer Storage and Recovery Program, city of Wichita, Kansas: U.S. Geological Survey Open-File Report 2012–1055, 55 p.

Contents

Figures

Tables

Conversion Factors, Abbreviations, and Datum

Multiply	By	To obtain
	Length	
meter (m)	3.281	foot (ft)
meter (m)	1.094	yard (yd)
centimeter (cm)	0.3937	inch (in.)
millimeter (mm)	0.03937	inch (in.)
micrometer (µm)	0.001	millimeter (mm)
nanometer (nm)	0.001	micrometer (µm)
inch (in.)	25.4	millimeter (mm)
foot (ft)	0.3048	meter (m)
mile (mi)	1.609	kilometer (km)
	Area	
square centimeter (cm^2)	0.00108	square foot (ft^2)
square foot (ft^2)	0.09290	square meter (m^2)
	Volume	
liter (L)	0.2642	gallon (gal)
liter (L)	61.02	cubic inch (in^3)
million gallon (Mgal)	3,785	cubic meter (m^3)
cubic foot (ft^3)	0.02832	cubic meter (m^3)
	Flow rate	
foot per second (ft/s)	0.3048	meter per second (m/s)
cubic foot per second (ft^3/s)	0.02832	cubic meter per second (m^3/s)
million gallon per day (Mgal/d)	0.04381	cubic meter per second (m^3/s)
	Concentration	
milligram per liter (mg/L)	1	part per million (ppm)
	Pressure	
pound per square inch (psi)	6.895	kilopascal (kPa)
	Mass	
gram (g)	0.03527	ounce, avoirdupois (oz)

Temperature in degrees Celsius (°C) may be converted to degrees Fahrenheit (°F) as follows:
°F=(1.8×°C)+32

Horizontal coordinate information is referenced to the North American Datum of 1983 (NAD 83).

Specific conductance is given in microsiemens per centimeter at 25 degrees Celsius (µS/cm at 25°C).

Concentrations of chemical constituents in water are given either in milligrams per liter (mg/L) or micrograms per liter (µg/L).

Acronyms and Other Abbreviations Used in Report

ACS	American Chemical Society
ADAPS	automated data and processing system
ADCP	acoustic Doppler current profiler
ADV	acoustic Doppler velocimeter
ASR	aquifer storage and recovery
CDOM	colored dissolved organic matter
CH	clean hands designee
CHIMP	continuous hydrologic instrumentation monitoring program
CPR	cardiopulmonary resuscitation
DCP	data collection platform
DELT	deformities, eroded fins, lesions, and tumors
DH	dirty hands designee
DIW	deionized water
DOI	Department of the Interior
EWI	equal width increment
FDOM	fluorescent fraction of dissolved organic matter
FNU	formazin nephelometric unit
GIS	geographic information system
GPS	global positioning system
GRSAT	graphical rating and shift application tool
HBMP	hydrobiological monitoring program
HDPE	high-density polyethylene
ILWS	integrated local water supply
JHA	job hazard analysis
KDHE	Kansas Department of Health and Environment
KDWPT	Kansas Department of Wildlife, Parks and Tourism
MF	membrane filtration
mFC	membrane fecal coliform
MPN	most probable number
mTEC	membrane-thermotolerant *Escherichia coli*
NAWQA	National Water-Quality Assessment, U.S. Geological Survey
NWISWeb	National Water Information System
NWQL	National Water Quality Laboratory, U.S. Geological Survey
OSW	Office of Surface Water
PDA	personal digital assistant
PFD	personal flotation device
PPE	personal protective equipment
PTSA	1,3,6,8-pyrenetetrasulfonic acid tetrasodium salt
RBP	rapid bioassessment protocols
SDI	serial data interface
USEPA	U.S. Environmental Protection Agency
USGS	U.S. Geological Survey
VOC	volatile organic compound
WFO	Wichita field office

Acknowledgments

The authors thank the U.S. Geological Survey (USGS) Wichita Field Office (WFO) field staff, including Beatrice Ritchie, Barbara Dague, Carlen Collins, and James Beilman. The authors greatly appreciate the efforts of Billy Justus from the USGS Arkansas Water Science Center in assistance with fish sampling. The following USGS personnel provided helpful information and comment during the preparation of this report: James Petersen (Arkansas Water Science Center), Brian Pellerin (California Water Science Center), Brian Loving (Kansas Water Science Center), James Putnam (Kansas Water Science Center), and Charles Perry (Kansas Water Science Center).

The authors also thank Debra Ary of the city of Wichita, Kansas for providing information and assistance with the coordination of sampling activities. Vernon Strasser of the city of Wichita Water and Wastewater Laboratory provided support in water sample analysis. Jeff Conley, Aaron Austin, and Ryan Waters from the Kansas Department of Wildlife, Parks and Tourism (KDWPT) also provided valuable assistance with fish sampling.

Protocols for Collection of Streamflow, Water-Quality, Streambed-Sediment, Periphyton, Macroinvertebrate, Fish, and Habitat Data to Describe Stream Quality for the Hydrobiological Monitoring Program, *Equus* Beds Aquifer Storage and Recovery Program, City of Wichita, Kansas

By Mandy L. Stone, Teresa J. Rasmussen, Trudy J. Bennett, Barry C. Poulton, and Andrew C. Ziegler

Abstract

The city of Wichita, Kansas uses the *Equus* Beds aquifer, one of two sources, for municipal water supply. To meet future water needs, plans for artificial recharge of the aquifer have been implemented in several phases. Phase I of the *Equus* Beds Aquifer Storage and Recovery (ASR) Program began with injection of water from the Little Arkansas River into the aquifer for storage and subsequent recovery in 2006. Construction of a river intake structure and surface-water treatment plant began as implementation of Phase II of the *Equus* Beds ASR Program in 2010.

An important aspect of the ASR Program is the monitoring of water quality and the effects of recharge activities on stream conditions. Physical, chemical, and biological data provide the basis for an integrated assessment of stream quality. This report describes protocols for collecting streamflow, water-quality, streambed-sediment, periphyton, macroinvertebrate, fish, and habitat data as part of the city of Wichita's hydrobiological monitoring program (HBMP). Following consistent and reliable methods for data collection and processing is imperative for the long-term success of the monitoring program.

Introduction

The city of Wichita's water supply currently (2012) comes from two primary sources: Cheney Reservoir and the Wichita *Equus* Beds aquifer well field (fig. 1). Because future water demands will exceed the capacity of the current water supply, the city of Wichita's Water Utilities Department developed an Integrated Local Water Supply (ILWS) Plan. One of the principal components of the ILWS Plan to increase the city of Wichita's available water supply for their future water demands through 2050 is the artificial recharge of the *Equus* Beds aquifer (City of Wichita, 1993). The *Equus* Beds Aquifer Storage and Recovery (ASR) Program involves pumping water out of the Little Arkansas River during higher streamflow conditions, treating it using the National Primary Drinking Water Regulations (U.S. Environmental Protection Agency, 2009) as a guide to water quality and to establish a treatment goal, and injecting it into the *Equus* Beds aquifer. The water will then be stored in the aquifer until needed by the city of Wichita.

The city of Wichita committed to develop and implement a hydrobiological monitoring program (HBMP) as part of mitigation described in the 2003 environmental report (Burns and McDonnell Engineering Company, Incorporated, 2003) for the ILWS Plan. A primary recommendation of a HBMP year five report by Burns and McDonnell Engineering Company, Incorporated (2010) was to implement a sampling protocol to ensure that specific sampling methods are followed. This document provides these sampling protocols.

Overview of the *Equus* Beds Aquifer Storage and Recovery Program

The *Equus* Beds aquifer covers parts of Sedgwick, Harvey, McPherson, and Reno Counties (fig. 1) and has been a primary source of water for the city of Wichita since 1940. The volume of water that historically has been pumped out of the easternmost extent of the *Equus* Beds aquifer has exceeded the aquifer's natural recharge rate in this area (Hansen and Aucott, 2010). Therefore, water levels in the easternmost area of the aquifer have decreased substantially. The easternmost area of the aquifer is susceptible to saltwater contamination from the Arkansas River and saltwater intrusion from the saltwater contamination plumes that already exist in the aquifer upgradient of the easternmost extent that were caused by oil field evaporation pits used in the 1930s (Whittemore, 2007). The *Equus* Beds ASR Program, along with greater use of Cheney Reservoir, will help the city of Wichita meet increasing future water demands. The ASR Program also will inhibit the encroachment of saltwater into the easternmost part of the aquifer (Ziegler and others, 2010).

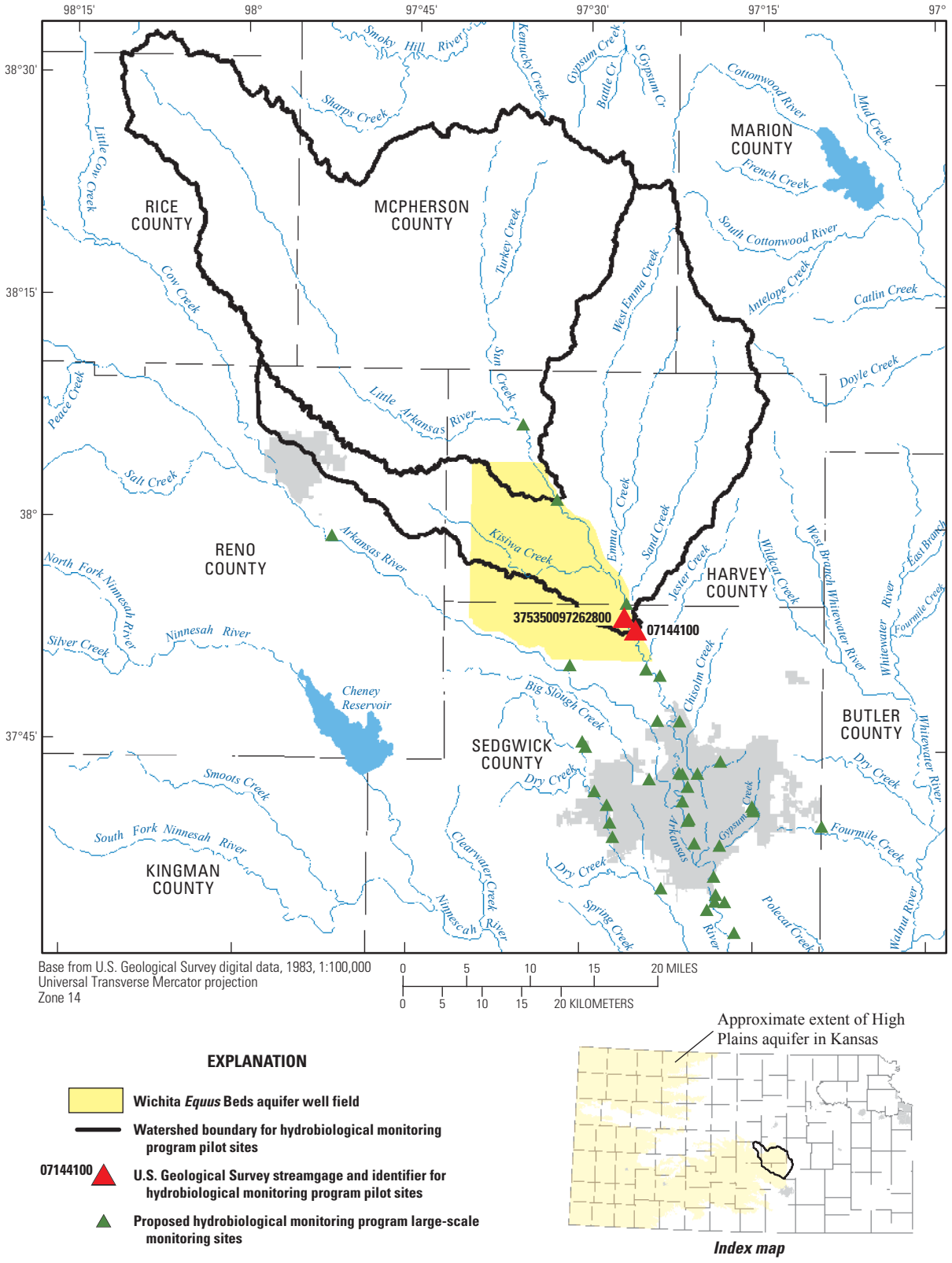


Figure 1. Map showing locations of monitoring sites and the watershed for the city of Wichita, Kansas' *Equus* Beds Aquifer Storage and Recovery (ASR) Program pilot project and monitoring sites for the proposed large-scale hydrobiological monitoring program (HBMP).

Figure 2. *Equus* Beds Aquifer Storage and Recovery (ASR) Program Phase II water intake facility and in situ water-quality monitor near Sedgwick, Kansas at site 375350097262800, Little Arkansas River upstream from ASR Facility (*A*) and in situ water-quality monitor and Solitax connected to a data logger in a water resistant shelter on a bridge at site 07144100, Little Arkansas River near Sedgwick, Kansas (*B*).

The *Equus* Beds ASR Program consists of four phases of construction. Phase I was completed in September 2006 and has the capacity to capture a maximum of 10 million gallons per day (Mgal/d) of above base-flow water from the Little Arkansas River and water temporarily stored in the aquifer immediately adjacent to the river for recharge. The stored water also serves as a hydraulic barrier to saltwater contamination. Primary components built during Phase I included three collection and diversion wells adjacent to the Little Arkansas River, a water intake structure that allows floodwater to be diverted into the system, a water treatment facility to clean the water, four recharge wells, and two recharge basins to inject the water into the *Equus* Beds aquifer. Phase II was initiated in 2008 with a preliminary design. Construction activities commenced in 2009 with construction of a 30 Mgal/d surface-water treatment plant and a 60 Mgal/d river intake structure equipped currently (2012) to divert 30 Mgal/d, as well as drilling of recharge recovery wells. This part of the ASR Program includes a larger water intake facility (fig. 2*A*) and a greater capacity water treatment facility than those constructed for Phase I. Phase II consists of 13 construction projects: surface-water treatment plant and river intake, recharge/recovery well completion, well-field maintenance facility, overhead power lines, substation, recharge/recovery well support facilities, surge tanks, and 6 pipeline projects. It is anticipated that Phase II will be completed by the end of 2012. More information about the city of Wichita ASR Program can be found at *http://www.wichitawaterproject.org/*.

Purpose and Scope

The purpose of this report is to provide an integrated set of protocols for collecting streamflow, water-quality, streambed-sediment, periphyton, macroinvertebrate, fish, and habitat data. This report describes protocols for collecting stream-quality data for the city of Wichita, Kansas' HBMP, including the *Equus* Beds Aquifer Storage and Recovery Program and other water-quality monitoring activities. Streamflow, water-quality, streambed-sediment, periphyton, macroinvertebrate, fish, and habitat data provide important information for an integrated assessment of stream quality. Following consistent and reliable methods for data collection is imperative for the long-term success of the monitoring program. Standard published methods for each component of data collection serve as the basis for these protocols. This report combines those methods into a single document that contains guidance on preparation, safety, protocols, and equipment for collection of stream-quality data. The data obtained from following these protocols will be used to establish baseline conditions before full implementation of aquifer recharge and to evaluate changes that may be related to the recharge program. In addition, data will be used to document stream quality, evaluate changing conditions, identify environmental factors affecting streams, provide science-based information for decision-making, and help meet regulatory monitoring requirements.

Protocols

Overview of Approach

The protocols described in this report were developed for the proposed large-scale HBMP. This HBMP encompasses and consolidates required water-quality assessment activities for the city of Wichita to meet regulatory and operational needs for the *Equus* groundwater recharge program, National Pollutant Discharge Elimination System (NPDES) wastewater and stormwater permits, and provide for public recreation safety in the Arkansas River. As a pilot study to a larger-scale HBMP, initial focus is on monitoring the effects of the Phase II ASR intake facility on the Little Arkansas River beginning in the spring of 2011.

To quantify and characterize the effects of the return line of the ASR facility discharging into the Little Arkansas River, upstream and downstream sampling locations were selected. The existing streamgaging station, Little Arkansas River near Sedgwick, Kansas (site 07144100; figs. 1 and 2) located about 1.7 river miles (mi; 2.7 kilometers) downstream from the ASR facility, was selected as the downstream sampling site. The upstream site (site 375350097262800; figs. 1 and 2) was installed at the ASR structure (fig. 2*A*) immediately upstream from the river intake. Data collection includes: gage height (river stage), streamflow (at the downstream site), physical water characteristics, ions, nutrients, suspended sediment and solids, copper, arsenic speciation, and pesticides. Streamflow and water chemistry constituents are measured for all flow conditions. To avoid scouring from high flows and to facilitate stream access and safety issues associated with higher flows, sediment, periphyton, macroinvertebrate, and fish, samples are collected during periods of low flow.

Safety and Landowner Permission

Field safety measures follow the chapter entitled "Safety in Field Activities" (chapter A9) from the USGS National Field Manual for the Collection of Water-Quality Data (variously dated). Personal protective equipment (PPE) is safety equipment for skin, eyes, ears, face, head, extremities, and respiration. Proper PPE selection depends on the hazards likely to be encountered and the compliance with regulatory safety requirements.

Many injuries and accidents can be prevented by analyzing and communicating potential hazards. A job hazard analysis (JHA) identifies hazards and details actions for mishap avoidance (appendix 1). JHA program requirements are described in the Department of the Interior (DOI) Departmental Manual 485, chapter 14, and in the USGS Handbook 445-2-H, chapter 2. JHA components include the sequence of steps that are associated with a field activity, identification of hazards that are involved with each of these steps, and determination of any controls needed to eliminate or reduce the hazards.

Many precautionary measures and certifications are required for USGS personnel. Operators of Government-owned or leased vehicles are certified and periodically recertified in defensive driving. USGS employees driving Government vehicles must have a valid U.S. State driver's license, take a course in driver safety every 3 years, tie down or otherwise secure all cargo, wear a seatbelt, obey all traffic regulations, and operate the vehicle safely. A list of personal, USGS, and local emergency contacts and health information centers will be kept in the field vehicle. Cardiopulmonary resuscitation (CPR) and other first-aid certification and recertification are mandatory for field personnel.

Safety precautions are exercised at all times during field activities, travelling to and from sites, and in the laboratory. Wading should not be attempted in streams that have values of depth multiplied by velocity equaling or exceeding 10 square feet per second (ft^2/s). A personal flotation device (PFD) must always be worn when working on bridges along with a high visibility vest with reflective tape. Gloves and safety glasses are to be worn at all times in the laboratory and when handling chemicals. Latex gloves are worn while collecting water and sediment samples because pathogens are found in nearly all surface-water systems. Chemicals are secured away from the passenger compartment and behind a safety screen or barrier for the safe containment and transport of chemicals in field vehicles.

Landowner permission will be obtained before any sampling activities for sites that are located on privately owned land or if access to a site requires crossing private property. The landowner will be provided with information regarding the purpose of sampling, the dates and times of entry and departure, and the anticipated duration of sampling if recurring visits are to be made. Special care will be taken to minimize any disturbances and damage to property will be strictly avoided.

Streamflow Protocols

Streamflow, or discharge, is defined as the volumetric rate of flow of water, or volume per unit time, in an open channel. Stream stage is the height or elevation of the water surface above an established datum plane and is a critical value in the computation of streamflow. The objective of operating a streamgaging station is to obtain a continuous record of stage and streamflow at a site. It requires installing and maintaining a streamgage that continuously measures stream stage, obtaining streamflow measurements during the range of flow conditions that occur at the site, developing a stage-discharge relation (rating curve) to compute continuous streamflow from stage data, and maintaining the rating curve by making necessary adjustments based on ongoing streamflow measurements. These streamflow protocols are integrated from two documents: stage measurement is described in detail in Sauer and Turnipseed (2010) and streamflow measurements at streamgaging stations are described in detail in Turnipseed and Sauer (2010).

Equipment and Supplies

- Personal flotation device (PFD)
- High visibility clothing
- Personal digital assistant (PDA)
- Pencils and clipboard
- Hip or chest waders
- Measuring tape
- Tag line
- Top-setting wading rod
- Acoustic Doppler velocimeter (ADV)
- Acoustic Doppler current profilers (ADCPs)

Streamgage Installation and Maintenance

Stream stage, or gage height, is the independent variable in a stage-discharge relation (fig. 3*B*) to compute streamflow. It is usually expressed in feet (ft) and hundredths of a foot. A reference, or outside, gage is used to set data loggers from which the primary gage height record is obtained. The reference gage is independent of the recording streamgage and is typically mounted on the most solid, stable structure available and is considered "truth" (Sauer and Turnipseed, 2010). This reference gage can be a wire-weight gage that is mounted on a bridge or a tape-down location for those sites that cannot have wire-weight gages installed.

A stage sensor is a device that automatically senses the vertical height of the water surface and includes nonsubmersible or submersible pressure transducers or radars. A data logger is a device that automatically records and stores gage-height readings sensed by a stage sensor. Stage recorders generally store gage heights at predetermined time intervals, such as every 15 minutes or every hour. A telemetry system is the means by which gage height (and other) data are transmitted from a data collection platform (DCP) to another location, such as a satellite. The accuracy standard set by the USGS Office of Surface Water (OSW) for the measurement of stage for most applications is plus or minus (±) 0.01 ft or 0.2 percent of the effective stage.

When selecting the streamgage site, controls, rating curves, backwater, streamflow variables, and acquisition of stage data are considered. Criteria for selecting the streamgage site include unchanging natural controls that promote a stable stage-streamflow relation, an appropriate reach for measuring streamflow throughout the stage range, and efficient access to the streamgage and measuring location. If a section control is present, the site should be located where the stage sensing point is in a low-velocity pool; on channel control streamgages, the site should be located in a clear, straight reach. The streamgage orifice or submersible transducer is placed low enough in the water to record the lowest possible stage. When possible, the instrument shelter should be high enough to be above the 200-year (0.5-percent probability) flood elevation. Appropriate reference marks and points are located on and off the streamgaging structure to maintain timely and accurate level surveys of the streamgage.

A list of key steps for a streamgage inspection is shown in table 1. When arriving at a streamgaging station, the clock in the DCP is checked and reset if necessary. Solar-charging panels are then inspected at each site visit and cleaned periodically. The reference gage is read at the same time as the recorder reading before the gage-height record is retrieved from the recorder; this reading is recorded, along with the time and date. The reference gage reading is then compared to the recorder reading to determine if there are any discrepancies. If a difference of greater than ± 0.02 ft is recorded, then the cause is determined and corrected. Inspection of the streamgage record before visiting the streamgage can indicate recorded stage data problems. The orifice is inspected to make sure it has not been buried by sediment. The water surface area is inspected to make sure there are no obstructions, such as logs, sand, or other debris. The radar signal should not be impinging off a pier or other obstruction that would affect the stage measurement. After other work is done at the streamgage, such as making a streamflow measurement or

Table 1. Steps for a streamgage inspection and streamflow measurement.

Step order number	Action
1	Arrive on site, begin SiteVisit in personal digital assistant (PDA).
2	Inspect stream channel and control.
3	Read reference gage height and record with time.
4	Read recorder gage height and record with time.
5	If discrepancy found with steps 3 and 4, determine the cause and make correction.
6	Inspect orifice.
7	Determine if making a wading or bridge measurement and select instrument.
8	Select streamflow measurement cross section.
9	Determine stream width if using an acoustic Doppler velocimeter (ADV) or mechanical current meter.
10	Determine measurement sections if using an ADV.
11	Take streamflow measurements in each section if using an ADV.
12	Make streamflow measurement if using an acoustic Doppler current profiler (ADCP).
13	Read reference gage height and record with time.
14	Read recorder gage height and record with time.
15	Input pertinent streamflow measurement information and SiteVisit notes into PDA.

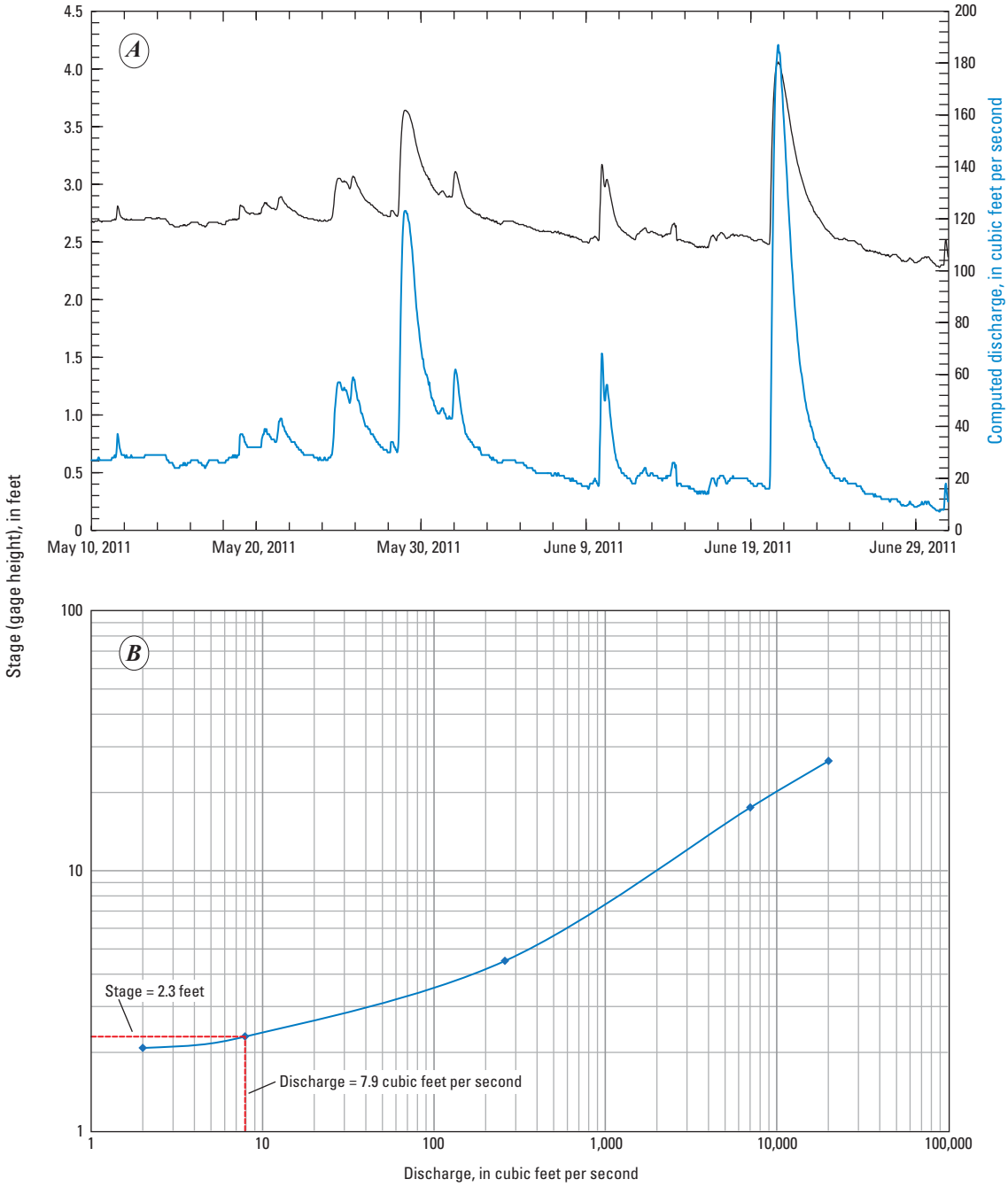


Figure 3. Example of real-time continuous data for gage height and computed discharge for site 07144100, Little Arkansas River near Sedgwick, Kansas (*A*) and example of a stage-discharge relation used to convert the stage (gage height) record to a discharge record (*B*).

other maintenance, the streamgage has a final check. Another complete set of streamgage readings is then made.

Documents maintained at each streamgage house include: the most recent expanded rating table to determine shift of measurement, the most recent station description, a log updated by servicing personnel upon each site visit describing measuring section location and gage readings, the most recent levels with all reference locations and elevations, a programming setup sheet for the data collection platform, a traffic control plan for the site, and a job hazard analysis. Bridges and orifice systems tend to settle or rise because of such factors as earth movement and battering by floodwaters. As such, leveling, a procedure by which surveying instruments are used to determine the differences in elevations between points, is used to set the streamgages and to check them from time to time for vertical movement. Levels are run at newly

installed streamgages at the time of construction and once every year for the first 3 years. Levels are run at established streamgages every 3 years if the outside reference is a wire-weight gage or yearly if it is a tape-down measuring point as long as all points are stable, otherwise levels are run annually. Reference and recorder gages are reset to agree with levels when the levels indicate an error of 0.015 ft or greater. Levels are run using field methods and documentation described in Kenney (2010).

Site Documentation

A station description is prepared for each monitoring site and becomes part of the permanent record for each site. The station description includes the location of the water-quality monitor and streamgage, a historical record of ongoing activities at the site, alternate routes to take during high-flow conditions, a description and history of the streamgage instrumentation, and maps, sketches, and photographs pertaining to monitoring site operations. JHAs, required for each site, provide safety guidelines to prevent unnecessary exposure to job-related hazards (appendix 1).

Streamflow Measurements

Streamflow is expressed in cubic feet per second (ft^3/s). A streamflow measurement is made by subdividing a stream cross section into segments and then by measuring the depth, width, and average velocity within each segment. The total streamflow is the summation of the flow in each of the segments of the stream cross section. Low-flow streamflow measurements can be made with a variety of instruments, but are made primarily by wading in the stream with an acoustic Doppler velocity meter (ADV). If acoustic instruments are not available, mechanical Price-AA or Price-pygmy current meters can be used. High-flow streamflow measurements are made with acoustic Doppler current profilers (ADCPs), and Price-AA mechanical current meters can be used if an ADCP is not available or will not work because of conditions in the stream.

A list of key steps for taking a streamflow measurement is shown in table 1 (steps 7–15). The first steps in making a streamflow measurement are to determine if a wading or a bridge measurement will be made and to select an appropriate measurement cross section. If the stream cannot be waded, the measurement is made from a bridge or cableway if possible. For details regarding site selection using an ADCP, refer to Mueller and Wagner (2009). If the stream can be waded, the field personnel determine the appropriate measurement cross section with these characteristics: has a reasonably straight channel; has a stable streambed free of large rocks, vegetation, or obstructions; has a smooth water surface; has steady flow conditions; has velocities that are greater than 0.3 ft/s; and is close to the streamgage.

When performing mid-section measurements, after the cross section is selected, the width of the stream is determined with a tag line or measuring tape, or bridge marks. The tag line or tape is placed at right angles to the direction of flow. If the bridge markings, tag line, or tape are not at right angles to the flow, the angle that it deviates should be noted and included in the section discharge calculation. There should be about 20 to 35 sections. The sections should not have greater than 10 percent of the total streamflow in them. An ideal measurement is one in which there are no sections that have greater than 5 percent of the total streamflow. Generally, the width of the sections should be shorter as depths and velocities become greater; equal widths of sections across the cross section should not be used unless the streamflow is evenly distributed. The minimum section width is 0.3 ft.

Wading measurement depths are measured using a top-setting wading rod, onto which the ADV or mechanical current meter attaches. When using a wading rod in greater velocities, there will be a velocity-head buildup of water on the wading rod. In this instance, stream depth is where the surface of the stream intersects the wading rod, not on the top of the velocity-head buildup. Wading rods are graduated in tenths of a foot, and stream depths are measured and recorded to the nearest 0.01 ft. ADV or mechanical current meter velocities are measured with either the 0.6-depth method or the two-point method. These methods are designed to be representative of the average vertical velocity. Generally, sections with water depths less than 1.5 ft are measured with the 0.6-depth method. The 0.6-depth method also is used when large amounts of slush ice or debris make it impossible to observe the velocity accurately at the 0.2 depth and when the stage in a stream is changing rapidly and a measurement must be made quickly. Velocity is measured at 0.6 of the depth below the water surface in the 0.6-depth method. Sections with water depths of 1.5 ft or greater are measured with the two-point method. Velocity is measured at 0.2 and 0.8 of the depth below the water surface in the two-point method. The average of these two velocities is the mean velocity in the vertical. When the velocity at the 0.8 depth is greater than that at the 0.2 depth, a third velocity is obtained at the 0.6 depth and then the average of the 0.8 and the 0.2 depth velocities is averaged with the 0.6 depth velocity.

Data Processing and Review

Real-time data on USGS Web pages are reviewed frequently by automated and manual procedures to ensure quality and prevent distributing erroneous information. Automated procedures include the setting of minimum and maximum threshold values for stage and elevation and their ranges of change. If these settings are exceeded, the erroneous data are masked from public viewing. Erroneous data are edited in Automated Data Processing System (ADAPS), the system used nationally by USGS to process, store, and retrieve water data. Frequent and ongoing screening and review of Web data is required, including at least a daily review of hydrographs during normal hours of operation.

Surface-water gage-height data are collected as a continuous record (for example, hourly or 15-minute values) by electronic storage on data loggers and transmitted electronically by satellite or other means. Streamflow records are computed by converting the gage-height record (fig. 3*A*) to a streamflow record by using a stage-discharge relation, or rating (fig. 3*B*). As such, the accuracy of the gage-height record is crucial for ensuring the accuracy of computed streamflow. Gage-height data are plotted to verify that there are no erroneous or missing data. Missing periods of gage-height data can be estimated or augmented by backup data stored in electronic form in the data logger. Streamflow for days of missing gage-height data normally is estimated. If the missing gage-height data does not affect the mean gage height for the day, the mean of the available partial-day streamflow can be entered into ADAPS as a write-protected, non-estimated daily mean streamflow.

A correction applied to gage-height data to compensate for the effect of settlement or uplift of the streamgage is measured by levels and is called a datum correction. A correction applied to gage-height data to compensate for differences between the recording gage and the reference gage is called a data correction. A correction applied to the stage-streamflow relation, or rating, to compensate for variations in the rating is called a shift.

To maintain accurate real-time streamflow records, it is critical that rating analysis and shift application to the record be completed as soon as possible after a streamflow measurement has been made. Shifts and datum corrections are applied within one working day following return from a site visit. Final streamflow records are completed within 30 days after a site visit. Malfunctioning streamgages are repaired as soon as possible, but no later than 3 days from the day the malfunction occurred. The USGS Kansas Web site for real-time surface-water data is located at *http://waterdata.usgs.gov/ks/nwis/rt*.

The Graphical Rating and Shift Application Tool (GRSAT) program is used for rating development and shift analysis (Aquatic Informatics, 2008). If a streamflow measurement plots within the accuracy of that measurement from the active shifted stage-discharge rating, the current shift-adjusted rating is presumed to be valid and a new shift may not be necessary. Otherwise, a new shifted rating will be developed and applied, and additional measurements may be made to confirm and define the rating change. Procedures for the development, modification, and application of ratings are described in detail in Kennedy (1984). Guidelines pertaining to rating and records computation presented in Rantz and others (1982) and Kennedy (1983) also are followed.

Continuous Water-Quality Monitor Protocols

Collecting water-quality data is a primary component in the ongoing operation of water-resource studies. Chemical concentrations in water commonly are determined by manual sampling procedures and laboratory analysis. These methods, however, leave gaps in data between sample collection. Because water quality frequently varies in short time periods, frequent and repeated measurements to adequately characterize variability are necessary. When the time interval between repeated water-quality measurements is small (hourly to every 15 minutes), the resulting record is considered continuous. An in situ device that measures water quality continuously is called a continuous water-quality monitor, or monitor. When the water-quality monitor is connected to a DCP, continuous data can be transmitted in near real time. One objective of operating a water-quality monitoring station is to provide a continuous record of water quality (specific conductance, pH, water temperature, dissolved oxygen, turbidity, and other indicators of water quality). When a continuous record is available, it can serve as the basis for the computation of constituent loads at that particular location. Additionally, if a relation can be established between continuously measured and discretely measured constituents, monitor data can serve as a surrogate for estimating other constituent concentrations and loads. The real-time data help identify temporal changes in selected water-quality constituents, thereby enhancing the existing streamgaging network to provide an alert system for regulators, water users, and the public. These continuous water-quality monitor protocols are adapted from those described in Wagner and others (2006).

Equipment and Supplies

- Personal flotation device (PFD)
- High visibility clothing
- Hip or chest waders
- Personal digital assistant (PDA)
- Pencils and clipboard
- Cleaning brush
- Deionized water
- Calibration standard solutions
- Cooler
- Calibrated field water-quality monitor

Equipment Installation and Maintenance

The main objective in water-quality monitor placement is the selection of a stable and secure location that is representative of the stream. Cross-sectional measurements of water-quality constituents are made to determine the most representative location for monitor placement, taking into consideration degree of mixing under varying streamflow and seasonal conditions. Additional cross-sectional measurements are made after installation to ensure that monitor installation is representative of the stream during all seasons and flow

conditions. The best location for a monitoring site is often the one that is best for measuring streamflow, as described previously in the streamflow protocols. A site should meet minimum stream depth requirements for sensors, be safely accessible under all conditions, and be located to avoid vandalism. In addition to monitor installation, equipment must be installed to support electronic storage on a DCP with transmissions of data by satellite.

A list of key steps for routine servicing of water-quality monitors is shown in table 2. Thorough field notes are maintained for all site visits. The Continuous Hydrologic Instrumentation Monitoring Program (CHIMP) software program, loaded onto a field Personal Digital Assistant (PDA), is used to enter and store site hydrologic data (Prewitt and others, 2011). After arriving onsite and beginning the visit in a PDA, the next step for operation and maintenance of a continuous water-quality monitor is to conduct a site inspection by recording monitor readings, time, and monitor conditions. An independently calibrated field monitor is used to corroborate data from the continuous water-quality monitor. With the independent field monitor placed near the sensors, readings (and time) are observed and recorded. The monitor is then removed from the monitoring location and the sensors are cleaned. The monitor is returned to the monitoring location and the monitor readings and time are recorded. Using the independent field monitor, the readings near the sensors are recorded. The monitor is removed once more, rinsed thoroughly, and the calibration is checked. The calibration-check values are recorded and the monitor should be recalibrated if necessary. The monitor is then returned to the monitoring location and the monitor readings and time are recorded. Lastly, the readings near the sensors are recorded using the independent field monitor.

General maintenance functions for a water-quality monitoring station include daily maintenance functions performed in the office and maintenance functions performed during field visits. Daily maintenance functions are completed by viewing real-time data in National Water Information System (NWISWeb) and include review of sensor function; a battery check; deletion of spurious data; and, if needed, transferring data downloaded from the monitor during a field inspection into the ADAPS database. Maintenance functions for field visits include calibrating the field meters in the office before going to the monitor station and at the site; inspecting the site for signs of physical disruption; inspecting and cleaning of the monitor and sensors for fouling, corrosion, or damage; cleaning of the deployment tube; checking the battery, date, and time; routine cleaning and servicing of sensors; checking calibration and recalibrating if necessary; and downloading data.

Inspection of the sensors and comparison against known standards or buffers is performed onsite at least every 3 months and cleanings are done at least every 4 weeks or more frequently if necessary. If readings exceed the calibration criteria, the sensors on the continuous water-quality monitor may need to be recalibrated. Water-quality monitors are recalibrated when it is determined that the sensor is out of calibration beyond the calibration criterion. Failure of the sensor to check properly against known standards or buffers can be an indication of calibration drift, sensor malfunction, or mislabeled or contaminated standards or buffers.

Table 2. Steps for routine servicing of water-quality monitors.

Step order number	Action
1	Arrive on site, begin visit in personal digital assistant (PDA).
2	Record monitor readings, time, and monitor conditions.
3	Record independent field monitor readings and time.
4	Clean sensors.
5	Return monitor to monitoring location and record readings and time.
6	Remove and rinse monitor and check calibration.
7	Recalibrate monitor if necessary.
8	Return monitor to monitoring location and record readings and time.
9	Record independent field monitor readings and time.

Site Documentation

A station description is prepared for each monitoring site and becomes part of the permanent record for each site. The station description includes the location of the water-quality monitor and streamgage, a historical record of ongoing activities at the site, alternate routes to take during high-flow conditions, a description and history of the streamgage instrumentation, and maps, sketches, and photographs pertaining to monitoring site operations. JHAs, required for each site, provide safety guidelines to prevent unnecessary exposure to job-related hazards (appendix 1).

Water-Quality Monitor

In situ water-quality monitors are monitors whose sensors are placed at a measuring point and communication cables are run to a data logger and power system that are located in a water resistant shelter (fig. 2*A*, *B*). The monitors are YSI 6600 V2 water-quality monitors. Direct current 12-volt batteries in conjunction with solar panels meet the power requirements of the sensors and recording equipment.

Temperature and Specific Conductance

Temperature affects water density, constituent solubility, pH, specific conductance, chemical reaction rates, and biological activity (Wilde, 2006). Continuous water-quality monitor sensors measure temperature with a thermistor, which is a semiconductor with resistance that changes with temperature. Thermistors are accurate, durable, require little maintenance, inexpensive, and can measure temperature to ± 0.1 degree Celsius (°C). The calibration criterion for temperature sensors

is ± 0.2°C. When measurements do not meet the calibration criterion the sensor is replaced. The sensor model used is the YSI 6550 temperature/conductivity sensor.

Electrical conductivity is a measure of the capacity of water to conduct an electrical current and is a function of the types and quantities of dissolved substances in water (Radtke and others, 2005). Conductivity of water increases as concentrations of dissolved ions increase. Specific conductance is the conductivity expressed in units of microsiemens per centimeter (µS/cm) at 25°C. Specific conductance sensors are designed to measure specific conductance in the range of 0 to 2,000 µS/cm or larger. The calibration criterion for specific conductance sensors is ± 5.0 µS/cm or ± 3.0 percent of the measured value, whichever is larger.

Specific conductance sensors are cleaned with a soft brush. Calibration of specific conductance sensors is minimally checked with two calibration standard solutions that bracket the range of conductivity readings recorded since the last time the sensor was checked with standards. A third calibration standard solution is used to check the linearity of the sensor between the high and low standard solutions. Zero response of the dry sensor in air is checked and recorded to ensure linearity of sensor response at low values. If sensor cleaning does not bring the sensor within the calibration criteria the sensor must be calibrated.

Dissolved Oxygen

Diffusion from surrounding air, atmospheric aeration, and photosynthesis are sources of dissolved oxygen in water (Hem, 1992). Dissolved oxygen is removed from water by respiration and decomposition of organic material. Dissolved oxygen plays a role in chemical reactions in water and is important for the survival of aquatic organisms. Dissolved oxygen is measured with a luminescent sensor that is based on dynamic fluorescence quenching. The dissolved oxygen sensor has a light-emitting diode to illuminate an oxygen-sensitive substrate that, when excited, emits a luminescent light with a lifetime that is directly proportional to the ambient oxygen concentration. The sensor model is the YSI 6150 optical dissolved oxygen sensor. Another method of characterizing dissolved oxygen is the percentage of dissolved oxygen saturation.

Calibration of the dissolved oxygen sensor is checked using the air saturated water method. The water-quality monitor with the attached dissolved oxygen sensor is placed in a container of water that has been saturated with air to 100 percent saturation. The calibration criterion for dissolved oxygen sensors is ± 0.3 milligrams per liter (mg/L). When measurements do not meet the calibration criterion the sensor is replaced.

pH

pH is a measure that represents the negative base-10 logarithm of hydrogen-ion activity of a solution in moles per liter (Hem, 1992). Water that has a pH less than 7 is acidic and water with a pH greater than 7 is basic or alkaline. Continuous pH sensors use the electrometric pH-measurement method using a hydrogen-ion electrode. Combination electrodes have a proton-selective glass bulb reservoir that is filled with a buffer. A silver wire coated with silver chloride is immersed in the internal reference electrode buffer reservoir. Protons on both sides of the glass pH electrode selectively interact with the glass and set up an external potential gradient across the outer glass membrane. This external potential difference across the glass membrane is proportional to the pH of the medium. The pH sensor used is the YSI 6561 pH sensor. Calibration of the pH sensor is minimally checked with pH buffer 7 and pH buffer 10. If pH readings are recorded below 7.0 units, then pH buffer 4 solution also may be needed to check the sensor. The calibration criterion for pH is ± 0.2 pH units. If the sensor does not meet the calibration criterion the sensor is recalibrated or replaced.

Colored Dissolved Organic Matter

Colored dissolved organic matter (CDOM) occurs naturally, primarily as a result of tannins released from decaying material in the water. However, activities from agriculture and wastewater discharge also can affect CDOM in streams (Vidon and others, 2008; Wilson and Xenopolous, 2009; Welch and Lindell, 1992). A Turner Designs Cyclops-7 CDOM sensor is used to measure the fluorescent fraction of dissolved organic matter (FDOM). The CDOM sensor, obtained from Turner Designs, is attached to the YSI water-quality monitor. Output voltage can be transmitted in real-time to the DCP using a serial data interface (SDI). A Zebra-Tech Cyclops-7 self-controlled Hydro-wiper will be installed, or a Turner Designs wiper in another YSI, Incorporated optical port will be modified, to keep the optical window as clean as possible. The CDOM sensor is calibrated with either the standard 1,3 6,8–pyrenetetrasulfonic acid tetrasodium salt (PTSA) or quinine sulfate solution and a blank of organic-free deionized water. The PTSA or quinine sulfate solution can be diluted to different concentrations to check the calibration range and linearity of the sensor.

Chlorophyll

Chlorophyll is an algal pigment that is used to gather light for photosynthesis (Wehr and Sheath, 2003). The YSI 6025 is the chlorophyll sensor used. The chlorophyll sensor gives an indication of total algal abundance. Chlorophyll fluoresces when irradiated with light of a particular wavelength. The chlorophyll sensor emits a wavelength of 470 nanometers (nm) and detects wavelengths of 650 to 600 nm. The natural variability of fluorescence in streams requires additional calibration and record-working steps when using the fluorescence sensors for measurements of chlorophyll. Chlorophyll sensors are calibrated using a two-point calibration. The first point is a zero standard using turbidity-free deionized water and the second point is a standard made from red fluorescent tracer (rhodamine b) dye. Instructions on how dye standards are

made can be found in the manufacturer's instruction manual (YSI Environmental, 2005). Because the standards do not directly measure chlorophyll, multiple regression analysis is needed to develop a relation between the readings from the fluorescence sensor and results from laboratory analyses of samples collected during multiple collection periods throughout the range of sensor readings. The continuous record from the fluorescence sensor may then be used to estimate the chlorophyll record.

Turbidity

Turbidity is the cloudiness of water caused by suspended and dissolved material such as clay, silt, finely divided organic material, plankton, other microscopic organisms, organic acids, and dyes (ATSM International, 2007). The sensor used is the YSI 6136 turbidity sensor. Calibration of the turbidity sensor is checked with two Hach StablCal, or manufacturer's turbidity standard solutions, of 100 and 1,000 formazin nephelometric units (FNUs). When necessary, a three-point calibration is done using 0, 100, and 1,000 FNU standards and the calibration is checked at higher FNUs if stream turbidity is expected to exceed 1,000 FNUs. The calibration criterion for turbidity is ± 0.5 FNUs or ± 5 percent of the measured value, whichever is larger.

Solitax

The YSI 6136 turbidity sensor reads a range from 0 to 1,000 FNUs, but the sensor has a maximum reporting limitation and may truncate values in highly turbid waters. Hach Solitax ts-line SC turbidity sensors are capable of recording turbidity values for as many as 4,000 FNUs or an estimated 50,000 mg/L of suspended solids. The Solitax sensor has a backscatter photoreceptor that measures suspended solids. There are four primary components of the Solitax sensors: power, the SC controller unit, data collection and output, and calibration. The controller unit controls the operations of the Solitax sensor and can be programmed for a variety of tasks. The most frequently used tasks are setting the sensor's wiper interval, setting the data output to either turbidity or suspended solids mode, and calibration or setting the controller's date and time as well as the logging intervals.

The SC controller and Solitax sensor require a 24-volt power system instead of the standard 12 volts that most field equipment uses. The 24 volts needed for the sensor can be obtained in two ways, either by using two 12-volt batteries wired in series or by using one 12-volt battery with a 12 volt to 24 volt inverter. During the spring through fall when the weather is warm and sunlight is at a maximum, batteries are charged by solar panels. However, during the colder months or when sunlight is limited, keeping batteries charged for a longer period of time can be difficult. Therefore, battery swaps are done routinely to minimize the amount of lost data.

Solitax sensors are used to augment turbidity information when the YSI 6136 sensor has reached its maximum reporting limits. Solitax sensors are installed in the stream alongside the water-quality monitor. The sensor is checked in 0, 100, 1,000, and possibly 1,800 or 4,000 FNU Hach standards, at the beginning of deployment and during any subsequent calibration checks. The Solitax sensor must be calibration checked and run in total suspended solids mode. The analog output is converted to SDI mode using an analog-to-SDI-12 converter to allow communication from the sensor to a DCP.

Nitrate

Nitrate (NO_3^-) is a naturally occurring water-soluble molecule made up of nitrogen and oxygen. Common sources of nitrate in surface water include fertilizers, animal waste, septic tanks, municipal sewage treatment systems, and decaying plant debris (Hem, 1992). The Hach Nitratax sensor with a 5-millimeter (mm) path length is an ultraviolet nitrate sensor that uses two-beam technology. This Nitratax sensor has a range of 0.1 to 25 mg/L, an accuracy of the greater of ± 3 percent of the measured value or 0.5 mg/L, and a resolution of 0.1 mg/L. The sensor is equipped with a self-cleaning wiper to keep the detection window free of sediment particles or algal buildup for more accurate readings.

The Nitratax sensor analog output is converted to SDI mode using an analog-to-SDI-12 converter to collect data and transfer it to the DCP. Only analog readings from the Nitratax sensor can be transmitted through the DCP. The 0.1 to 25 mg/L concentration range of the Nitratax sensor is scaled down to a 0 to 5 volt scale. The controller can be set to internally log with data routinely downloaded into ADAPS to obtain actual concentration values. After sufficient data have been collected for a large range of concentration readings, a regression equation is developed to convert the transmitted voltage reading into a concentration value. Calibration checks are made with nitrate standards of known concentrations.

Data Processing and Review

Real-time water-quality data are inspected on a daily basis using NWISWeb and ADAPS to immediately rectify any problems caused by sensor malfunction. Invalid data are deleted as soon as possible and missing data are augmented by backup data stored in electronic form in the data logger. Minimum and maximum thresholds and rates-of-change are used to automatically block potentially erroneous data from being viewed publicly.

Processing water-quality monitoring records involves initial evaluation of raw data; application of corrections for sensor calibration, drift, or fouling; final data evaluation; record processing; and final record review. The procedure requires analysis of water-quality monitoring data such as water temperature, specific conductance, pH, dissolved oxygen, and turbidity and other onsite observations that were recorded before, during, and after the cleaning, calibration checks, and recalibration procedures for the onsite water-quality monitor.

Table 3. Equipment and supplies for water-quality field sampling and laboratory processing.

[L, liter; VOC, volatile organic compound; mL, milliliter; HCl, hydrochloric; µm, micrometer; mm, millimeter]

Field sampling	Laboratory processing for water-quality constituents	Laboratory processing for bacterial analyses
Personal flotation device (PFD)	0.45-µm disposable capsule filter	Autoclave.
High visibility clothing	Tubing	Sterile phosphate-buffered water with magnesium chloride.
Hip or chest waders	Peristaltic pump	Sterile deionized water, unbuffered, for diluting samples.
Powderless latex gloves	Precleaned, prebaked amber glass sample bottles (125 mL and 1 L)	Incubators.
Pencils and clipboard	Acid-rinsed polypropylene sample bottle (250 mL)	Sterile glass pipets.
Water-quality sampling field and laboratory sheet (appendix 2)	Plain polypropylene sample bottles (125 mL, 250 mL, 500 mL, and 1 L)	Pi pet bulb.
US DH-81 sampler	Amber (brown) polypropylene sample bottles (125 mL)	Thermometer.
US DH-95 sampler	142-mm stainless steel plate filter assembly	Autoclavable plastic bags.
US D-95 sampler	Organic grade blank water	Autoclave tape.
1-L fluorocarbon polymer bottle	1-L fluorocarbon polymer squeeze bottle	Alcohol burner.
D-77 fluorocarbon polymer cap and nozzle assembly	6.35-mm fluorocarbon polymer tubing	Alcohol bottle containing 100 mL of ethanol.
Teflon churn splitters	Ceramic piston pump	Cultivation media.
Churn carrier with lid	Stainless steel rounded forceps	Counter.
Measuring tape	Refrigerator	Filter funnel and base.
Tag line	Coolers and ice	Sterile plastic filter holder.
Deionized water	2-mL nitric acid ampules	Stainless steel rounded forceps.
Tap water	1-mL 1:7 (4.5 normal) sulfuric acid ampules	Membrane filters.
Plastic bags (food storage and heavy duty)		Culture plates.
Coolers and ice		Vacuum source.
Autoclaved 1-L Nalgene sample bottles		Refrigerator.
VOC sampler		
40-mL amber glass vials		
1:1 HCl acid drops for preserving VOC samples		

Corrections to the data are not made unless the cause or error is validated or explained by onsite observations, field notes and measurements, and comparison to nearby monitors. If raw data differs from corrected data by more than the established allowable limits then the corrected data are not reported. Field notes and calibration accuracy are a necessary component of ensuring the computed record accuracy. Further details regarding record computation are described in Wagner and others (2006).

Water-Quality Sampling Protocols

The primary objective in collecting a water-quality sample is to obtain environmental data that are representative of the study system. Sampling and processing techniques may vary according to the type of compound, such as inorganic or organic constituents. Collecting surface-water samples that accurately represent the physical and chemical characteristics of the study system requires the appropriate use of sampling equipment and methods to describe environmental variability and to prevent contamination or bias in the sampling process. This section describes standard methods for the routine collection of representative water samples for basic water-quality constituents, fecal indicator bacteria, and volatile organic compounds. These water-quality sampling protocols are adapted from the National Field Manual for the Collection of Water-Quality Data (U.S. Geological Survey, variously dated), and the USGS National Water-Quality Assessment Program (NAWQA) Protocol for Collecting and Processing Stream Water Samples (Shelton, 1994).

Sample Collection and Processing Procedures

Equipment and supplies for water-quality field sampling and laboratory processing are shown in table 3. Completion of detailed field sheets (appendix 2) are required for all sampling activities. A list of key steps for water-quality sample collection is shown in table 4.

Basic Water-Quality Constituents

Sampling Equipment

The sampler used for wadeable water sample collection is the US DH-81 with an acid-washed 1-liter (L) polytetrafluoroethylene bottle, and a D-77 polytetrafluoroethylene cap and nozzle assembly. The DH-81 sampler consists of a polypropylene collar screwed onto a plastic-coated wading rod. The samplers used for nonwadeable water sample collection are suspended, usually from a bridge, and lowered into the stream by a rope or cable. The nonwadeable samplers are US DH-95 or US D-95 with an acid-washed 1-L polytetrafluoroethylene bottle and a D-77 polytetrafluoroethylene cap and nozzle assembly. Regardless of sampler type, the water samples are composited in an acid-washed 14-L polytetrafluoroethylene churn splitter. The churn splitter, after being washed in a liquinox solution and rinsed in tap or deionized water, is placed in a 5-percent hydrochloric acid rinse solution. The churn splitter is rinsed with copious amounts of deionized water and air dried in the laboratory and placed inside two large clear plastic bags and then into a churn carrier before being transported into the field.

Cleaning Equipment

Many steps should be used to avoid sample contamination. The most common causes of sample contamination during sample collection include poor handling techniques, input from atmospheric sources, dirty equipment, and the use of equipment made of materials that are inappropriate for the study target analytes. Good field practices are implemented and clean hands/dirty hands sampling techniques are used to prevent or minimize sample contamination from these sources. The clean hands designee (CH) has the only contact with the sample bottle, transfers the sample from sampler to splitter, and preserves the sample. The dirty hands designee (DH) operates sampling equipment and manages any contact with potential sources of contamination. Both designees must wear disposable powderless latex gloves. Hands are kept gloved and away from potential sources of contamination during sample processing. Equipment is field rinsed and a prescribed sampling order is followed. Atmospheric contamination is minimized and a sufficient number of blanks and replicates are collected to monitor for inadvertent contamination.

Field personnel are responsible for selecting sampling sites, including the specific points and transect(s) at which samples will be collected. Optimal sampling site locations are located: at or near a streamgage, in straight reaches with

Table 4. Steps for water-quality sample collection.

[ASR FAC, aquifer storage and recovery facility]

Step order number	Action
1	Select sampling equipment based on selected sampling site.
2	Sedgwick River site—Select cross section, set the tag line if taking a wading measurement.
3a	Sedgwick River site—Select equal width increments (EWIs).
3b	Upstream ASR FAC site—Select equal depth increments.
3c	At ASR FAC site—Select weighted basket.
4	Put on latex gloves and keep hands away from contamination.
5	Take volatile organic compound (VOC) sample.
6	Put on new pair of latex gloves and keep hands away from contamination.
7	Field rinse sampling equipment with environmental water.
8a	Sedgwick River site—Sample cross section using equal width increment sampling methods.
8b	Upstream ASR FAC site—Water sample collected using weighted basket or depth integrated sampler (determined by flow conditions).
8c	At ASR FAC site—Sample collected using weighted basket.
9	While sampling, composite water samples into churn splitter.
10	When finished sampling, re-close bags around the churn splitter and place lid on churn carrier.
11	Put on new pair of latex gloves and keep hands away from contamination.
12	Take indicator bacteria grab samples.
13	Record dates and times on all paperwork (field sheets, labels, and analytical service requests).
14	Place sample labels on appropriate sample bottle.
15	Put on new pair of latex gloves and keep hands away from contamination.
16	Except for bacteria samples, process water-quality samples in field vehicles according to parts per billion sampling protocols.
17	Rinse sampling equipment with deionized or tap water.
18	At field office, select bacteria filtration equipment, filters, agar plates.
19	Put on new pair of latex gloves and keep hands away from contamination.
20	Process bacteria samples.

uniform flow, far enough above and below confluences to avoid poorly mixed cross sections, in reaches upstream from bridges or other structures, in unidirectional flow that does not have eddies, and at a cross section where samples can be collected at any stage throughout the study period.

Water samples are collected into a 14-L polytetrafluoroethylene churn splitter. CH is responsible for field rinsing the equipment before sampling begins. Powderless latex gloves are put on and 2 to 4 L of stream rinse water are poured from the sampler into the churn splitter through the top of the churn. The churn splitter is removed from the churn carrier, leaving the outer plastic bag inside the carrier. The churn disk is moved up and down several times with the stream rinse water to ensure the inside is thoroughly wetted and the rinse water is then swirled vigorously in the churn. A hole is pierced through the inner plastic bag to expose the churn spigot and the rinse water is drained through the spigot. After draining the rinse water from the churn splitter, the churn splitter in the plastic bag is rotated so that the spigot is not exposed. The inner plastic bag containing the churn splitter is placed into the outer plastic bag in the churn carrier.

Collecting Stream-Water Samples

The standard procedure for stream sampling is to collect the sample through the entire depth of the water column at multiple vertical transects by the equal-width increment (EWI) method. This method generates a representative cross-sectional sample where each unit of streamflow is equally represented. When EWI sampling is not possible, an abbreviated sampling method, such as a weighted-bottle or dip (grab) sample, is collected.

The stream cross section is divided into a number of EWIs. Five to 10 increments are used for streams less than 5 ft [1.52 meters (m)] wide and a minimum of 10 increments are used in streams 5 ft (1.52 m) wide or greater. A maximum of 20 increments should be used in extremely wide and shallow cross sections. The sample verticals should be spaced at least 6 inches [in.; 15.24 centimeters (cm)] apart. Samples are collected by lowering and raising a sampler through the water column at the center of each increment by DH. Samples from several verticals can be collected in the same bottle. The bottle should not overfill because secondary circulation and enrichment of heavy particles can occur and bias the sample. The same transit rate is used for all verticals. When additional verticals cannot be sampled without overfilling the bottle, the bottle is emptied directly into the churn splitter by CH until all of the verticals have been sampled.

Sample processing procedure depends on the targeted analytes and the intended use of the data. As with sample collection, the clean hands/dirty hands protocols are used during processing steps. Sample processing equipment is kept covered with a clean, noncontaminating material when not in use and sample bottles are kept capped and covered or bagged.

Laboratory Processing

Clean hands/dirty hands protocols are followed throughout laboratory processing. The churn splitter and inner bag are removed from the churn carrier in the laboratory. Prelabeled bottles are placed within easy reach of the churn spigot. The sample is churned at a uniform rate by raising and lowering the disk inside the churn splitter with smooth, even strokes. When churning, the disk should touch the bottom on every stroke and the stroke length should be as long as possible without breaking the water surface. If the disk breaks the surface of the water, excessive air is introduced into the sample and could affect dissolved gases, bicarbonate, pH, and other characteristics of the sample. The churning rate should be about 9 inches per second (in./s). The sample is pre-mixed by churning for about 10 strokes to uniformly disperse suspended material before subsampling.

General sample processing order for water composited in a churn splitter is as follows: suspended sediment, suspended solids, turbidity, nonfiltered nutrients, total organic carbon, trace elements, major ions, filtered nutrients, dissolved organic carbon, and organics. Specific information on sample processing details related directly to the pilot HBMP is listed in table 5. Unfiltered samples are collected first and placed directly into the appropriate type of sample bottle (table 5) from the churn splitter. An adequate volume of sample water for rinsing is withdrawn while continuously churning. The sample bottles are rinsed three times before the subsamples are withdrawn. The churning/subsampling process should not be interrupted. Unfiltered samples are preserved if necessary (table 5), then refrigerated until they are shipped to the analyzing laboratory.

Clean hands/dirty hands protocols are used in the plate-filter procedure for organic sample analysis and gloves are changed after setting up the equipment. A clean 142-millimeter (mm) stainless steel plate filter assembly is removed from the protective plastic bag by CH. CH places a 0.7-micrometer (µm) glass microfiber filter on top of the screen in the filter assembly with stainless steel forceps and wets it with a few drops of organic-grade blank water from a 1-L polytetrafluoroethylene squeeze bottle. CH closes the plate filter assembly by aligning the top and bottom plates and lightly tightening the locking ring. CH then attaches two 6.35-mm polytetrafluoroethylene tubes to a ceramic piston pump, attaching one tube to the inlet connector of the filter assembly and the other into the 1-L polytetrafluoroethylene squeeze bottle filled with organic-grade blank water. One L of organic-grade blank water is passed through the filter. The blank water is passed through the filter with the air-vent valve open and CH tilts the assembly slightly to the side to allow all trapped air to escape. CH closes the air-vent valve when water begins to discharge through the valve. DH turns the pump on and off. CH removes the intake tube from the squeeze bottle and places it in the churn splitter sample water. Sample water is then passed through the filter into the appropriate sample bottles (table 5). If the filter begins to clog it is discarded, a new filter is placed into the assembly, and the above procedures are repeated. Samples are preserved

Table 5. Water-quality constituents and processing and shipping information for the *Equus* Beds Aquifer Storage and Recovery (ASR) Program hydrobiological monitoring program (HBMP).

[USGS, U.S. Geological Survey; NWQL, National Water Quality Laboratory; ASR, analytical services request; GCV, glass chilled vial; VOCs, volatile organic compounds; mL, milliliter; HCl, hydrochloric; <, less than; °, degrees; C, Celsius; ±, plus or minus; --, not applicable; WFO, Wichita field office; L, liter; WMWWL Wichita Municipal Water and Wastewater Laboratory; OWML, Ohio Water Microbiology Laboratory; ISL, Iowa Sediment Laboratory; EWI, equal width increment; TBY, turbidity; RU, raw unacidified; SC, specific conductance; ANC, acid neutralizing capacity; RCB, raw chilled bottle; WCA, whole water chilled acidified; µm, micrometer; FA, filtered acidified; SAS, arsenic speciation; µL, microliter; EDTA, ethylenediaminetetraacetic acid; DI, deionized; FCC, filtered chilled container; FU, filtered unacidified; TOC, total organic carbon; GCC, glass chilled container; DOC, dissolved organic carbon; IMA, immunoassay; OGRL, Organic Geochemistry Research Laboratory; UAS, ultraviolet absorbing substances]

USGS NWQL schedule (SCH) or lab code (LC)	Water-quality constituent ASR code and category	Analyzing laboratory	Container information	Collection method	Processing notes	Shipping information
Grab samples (not collected into, or processed from, churn splitters)						
SCH 1380	GCV—VOCs	USGS NWQL	three 40-mL VOC vials	Centroid dip, grab, weighted basket, or VOC sampler	Do not rinse vials; there must not be any air bubbles in vials; vials are acidifed immediately after collection with HCl acid drops to pH < 2 (generally 4 drops); sample processed at analyzing laboratory	Chilled, must arrive at 4°C ± 2°C.
--	Bacteria (fecal coliform and *Escherichia coli*), Bacteria Reaction Tests (BARTS): iron-related, sulfate-reducing, slime-forming	USGS WFO	1-L autoclaved Nalgene bottle	Centroid dip or grab into autoclaved 1-L Nalgene bottle	Do not rinse bottle; sample processed at analyzing laboratory	Chilled, must arrive at 4°C ± 2°C before processing.
--	Bacteria	WMWWL	two 250-mL autoclaved Nalgene bottles	Centroid dip or grab into autoclaved 250-mL Nalgene bottle	Do not rinse bottle; sample processed at analyzing laboratory	Chilled, must arrive at 4°C ± 2°C before processing.
--	Coliphage	USGS OWML	1-L autoclaved Nalgene bottle	Centroid dip or grab into autoclaved 1-L Nalgene bottle	Do not rinse bottle; sample processed at analyzing laboratory	Chilled, must arrive at 4°C ± 2°C.
First churn splitter - total (whole water) inorganic samples						
--	Suspended sediment, loss on ignition, 5-point particle size	USGS ISL	1-L wide-mouth polypropylene bottle	EWI into churn splitter	Do not rinse bottle	--
--	Total suspended solids	WMWWL	500-mL plain polypropylene bottle	EWI into churn splitter	Use unfiltered sample to rinse bottle	--
--	Turbidity, total residual chlorine, specific conductance, and pH	WMWWL	500-mL plain polypropylene bottle	EWI into churn splitter	Use unfiltered sample to rinse bottle	Chilled, must arrive at 4°C ± 2°C.
LC 2187	TBY—Turbidity	USGS NWQL	500-mL plain polypropylene bottle	EWI into churn splitter	Use unfiltered sample to rinse bottle	--

Table 5. Water-quality constituents and processing and shipping information for the *Equus* Beds Aquifer Storage and Recovery (ASR) Program hydrobiological monitoring program (HBMP).—Continued

[USGS, U.S. Geological Survey; NWQL, National Water Quality Laboratory; ASR, analytical services request; GCV, glass chilled vial; VOCs, volatile organic compounds; mL, milliliter; HCl, hydrochloric; <, less than; °, degrees; C, Celsius; ±, plus or minus; --, not applicable; WFO, Wichita field office; L, liter; WMWWL Wichita Municipal Water and Wastewater Laboratory; OWML, Ohio Water Microbiology Laboratory; ISL, Iowa Sediment Laboratory; EWI, equal width increment; TBY, turbidity; RU, raw unacidified; SC, specific conductance; ANC, acid neutralizing capacity; RCB, raw chilled bottle; WCA, whole water chilled acidified; µm, micrometer; FA, filtered acidified; SAS, arsenic speciation; µL, microliter; EDTA, ethylenediaminetetraacetic acid; DI, deionized; FCC, filtered chilled container; FU, filtered unacidified; TOC, total organic carbon; GCC, glass chilled container; DOC, dissolved organic carbon; IMA, immunoassay; OGRL, Organic Geochemistry Research Laboratory; UAS, ultraviolet absorbing substances]

USGS NWQL schedule (SCH) or lab code (LC)	Water-quality constituent ASR code and category	Analyzing laboratory	Container information	Collection method	Processing notes	Shipping information
First churn splitter - total (whole water) inorganic samples—Continued						
--	Total recoverable copper	WMWWL	250-mL acid-rinsed polypropylene bottle	EWI into churn splitter	Use unfiltered sample to rinse bottle; acidify sample with nitric acid to pH < 2	
SCH 1050, [1]SCH 13	RU—SC, pH, ANC	USGS NWQL	250-mL plain polypropylene bottle	EWI into churn splitter	Use unfiltered sample to rinse bottle	--
LC 20	RCB - Color	USGS NWQL	250-mL plain polypropylene bottle	EWI into churn splitter	Use unfiltered sample to rinse bottle	Chilled, must arrive at 4°C ± 2°C.
[1]SCH 535	WCA—Total Nutrients, microkjeldahl phosphorus and nitrogen	USGS NWQL	125-mL plain polypropylene bottle	EWI into churn splitter	Use unfiltered sample to rinse bottle; acidify sample with 1 mL of 4.5 normal sulfuric acid	Chilled, must arrive at 4°C ± 2°C.
First churn splitter - filtered inorganic samples						
--	Filtered metals	WMWWL	250-mL acid-rinsed polypropylene bottle	EWI into churn splitter	Filtered through a 0.45-µm capsule filter; use filtered sample to rinse bottle; acidify sample with nitric acid to pH < 2	--
SCH 1050, SCH 13, LC 3122, [1]LC 3132	FA—Filtered metals, arsenic, selenium	USGS NWQL	250-mL acid-rinsed polypropylene bottle	EWI into churn splitter	Filtered through a 0.45-µm capsule filter; use filtered sample to rinse bottle; Acidify sample with nitric acid to pH < 2	--
--	Filtered mercury	WMWWL	250-mL acid-rinsed glass bottle	EWI into churn splitter	Filtered through a 0.45-µm capsule filter; use filtered sample to rinse bottle; acidify sample with nitric acid to pH < 2	--
LC 3142	SAS—Filtered arsenic speciation	USGS NWQL	8-mL brown polypropylene bottle prerinsed with DI water and 100 µL EDTA preservative added to bottle	EWI into churn splitter	Do not rinse bottle in field (bottle is pre-rinsed with DI water and treated with 100 µL EDTA); filtered through a 0.45-µm capsule filter	Chilled, must arrive at 4°C ± 2°C.

Table 5. Water-quality constituents and processing and shipping information for the *Equus* Beds Aquifer Storage and Recovery (ASR) Program hydrobiological monitoring program (HBMP).—Continued

[USGS, U.S. Geological Survey; NWQL, National Water Quality Laboratory; ASR, analytical services request; GCV, glass chilled vial; VOCs, volatile organic compounds; mL, milliliter; HCl, hydrochloric; <, less than; °, degrees; C, Celsius; ±, plus or minus; --, not applicable; WFO, Wichita field office; L, liter; WMWWL Wichita Municipal Water and Wastewater Laboratory; OWML, Ohio Water Microbiology Laboratory; ISL, Iowa Sediment Laboratory; EWI, equal width increment; TBY, turbidity; RU, raw unacidified; SC, specific conductance; ANC, acid neutralizing capacity; RCB, raw chilled bottle; WCA, whole water chilled acidified; µm, micrometer; FA, filtered acidified; SAS, arsenic speciation; µL, microliter; EDTA, ethylenediaminetetraacetic acid; DI, deionized; FCC, filtered chilled container; FU, filtered unacidified; TOC, total organic carbon; GCC, glass chilled container; DOC, dissolved organic carbon; IMA, immunoassay; OGRL, Organic Geochemistry Research Laboratory; UAS, ultraviolet absorbing substances]

USGS NWQL schedule (SCH) or lab code (LC)	Water-quality constituent ASR code and category	Analyzing laboratory	Container information	Collection method	Processing notes	Shipping information
First churn splitter - filtered inorganic samples—Continued						
--	Filtered nitrate and orthophosphorus	WMWWL	125-mL brown polypropylene bottle	EWI into churn splitter	Filtered through a 0.45-µm capsule filter; use filtered sample to rinse containers	--
SCH 535	FCC—Filtered nitrate, nitrite, orthophosphorus, ammonia, phosphorus	USGS NWQL	125-mL brown polypropylene bottle	EWI into churn splitter	Filtered through a 0.45-µm capsule filter; bottle rinsed with filtered sample	Chilled, must arrive at 4°C ± 2°C.
--	Filtered ammonia	WMWWL	250-mL plain polypropylene bottle	EWI into churn splitter	Filtered through a 0.45-µm capsule filter; bottle rinsed with filtered sample; acidify sample with 2 mL 1:1 sulfuric acid	Chilled, must arrive at 4°C ± 2°C.
--	Filtered phosphorus	WMWWL	250-mL plain polypropylene bottle	EWI into churn splitter	Filtered through a 0.45-µm capsule filter; bottle rinsed with filtered sample; acidify sample with 2 mL 1:1 sulfuric acid	Chilled, must arrive at 4°C ± 2°C.
--	Filtered anions and alkalinity	WMWWL	500-mL plain polypropylene bottle	EWI into churn splitter	Filtered through a 0.45-µm capsule filter; bottle rinsed with filtered sample	--
[1]SCH 13	FU—Filtered anions	USGS NWQL	250-mL plain polypropylene bottle	EWI into churn splitter	Filtered through a 0.45-µm capsule filter; bottle rinsed with filtered sample	--
--	Filtered cyanide	WMWWL	500-mL plain polypropylene bottle	EWI into churn splitter	Filtered through a 0.45-µm capsule filter; bottle rinsed with filtered sample; add sodium hydroxide to pH > 12	Chilled, must arrive at 4°C ± 2°C.
Second churn splitter - total (whole water) organic samples						
--	TOC—Total organic carbon	WMWWL	125-mL amber glass bottle baked at 450°C by laboratory	EWI into churn splitter	Do not rinse bottle; acidify sample to pH < 2 with 1 mL of 4.5 normal sulfuric acid	Chilled, must arrive at 4°C ± 2°C.

Table 5. Water-quality constituents and processing and shipping information for the *Equus* Beds Aquifer Storage and Recovery (ASR) Program hydrobiological monitoring program (HBMP).—Continued

[USGS, U.S. Geological Survey; NWQL, National Water Quality Laboratory; ASR, analytical services request; GCV, glass chilled vial; VOCs, volatile organic compounds; mL, milliliter; HCl, hydrochloric; <, less than; °, degrees; C, Celsius; ±, plus or minus; --, not applicable; WFO, Wichita field office; L, liter; WMWWL Wichita Municipal Water and Wastewater Laboratory; OWML, Ohio Water Microbiology Laboratory; ISL, Iowa Sediment Laboratory; EWI, equal width increment; TBY, turbidity; RU, raw unacidified; SC, specific conductance; ANC, acid neutralizing capacity; RCB, raw chilled bottle; WCA, whole water chilled acidified; µm, micrometer; FA, filtered acidified; SAS, arsenic speciation; µL, microliter; EDTA, ethylenediaminetetraacetic acid; DI, deionized; FCC, filtered chilled container; FU, filtered unacidified; TOC, total organic carbon; GCC, glass chilled container; DOC, dissolved organic carbon; IMA, immunoassay; OGRL, Organic Geochemistry Research Laboratory; UAS, ultraviolet absorbing substances]

USGS NWQL schedule (SCH) or lab code (LC)	Water-quality constituent ASR code and category	Analyzing laboratory	Container information	Collection method	Processing notes	Shipping information
Second churn splitter - total (whole water) organic samples—Continued						
LC 3211	TOC—Total organic carbon	USGS NWQL	125-mL amber glass bottle baked at 450°C by laboratory	EWI into churn splitter	Do not rinse bottle; do not acidify TOC sample sent to NWQL	Chilled, must arrive at 4°C ± 2°C.
SCH 1608	GCC—Total organochlorine pesticides and alachlors	USGS NWQL	1-L baked amber glass bottle baked at 450°C by laboratory	EWI into churn splitter	Do not rinse bottle	Chilled, must arrive at 4°C ± 2°C.
Second churn splitter - filtered organic samples						
--	DOC—Dissolved organic carbon	WMWWL	125-mL amber glass bottle baked at 450°C by laboratory	EWI into churn splitter	Filtered through a 0.7-µm glass fiber filter; do not rinse bottle; acidify sample to pH < 2 with 1 mL of 4.5 normal sulfuric acid	Chilled, must arrive at 4°C ± 2°C.
LC 2613	DOC—Dissolved organic carbon	USGS NWQL	125-mL amber glass bottles baked at 450°C by laboratory	EWI into churn splitter	Filtered through a 0.7-µm glass fiber filter; do not rinse bottle; acidify sample to pH < 2 with 1 mL of 4.5 normal sulfuric acid	Chilled, must arrive at 4°C ± 2°C.
--	IMA—Atrazine	USGS OGRL	three 125-mL amber glass bottles baked at 450°C by laboratory	EWI into churn splitter	Filtered through a 0.7-µm glass fiber filter; do not rinse bottle	Chilled, must arrive at 4°C ± 2°C.
LC 2614 and 2615	UAS—Ultraviolet absorbing organic constituents	USGS NWQL	125-mL amber glass bottles baked at 450°C by laboratory	EWI into churn splitter	Filtered through a 0.7-µm glass fiber filter; do not rinse bottle	Chilled, must arrive at 4°C ± 2°C.
SCH 2003	GCC—Filtered herbicides	USGS NWQL	1-L baked amber glass bottle baked at 450°C by laboratory	EWI into churn splitter	Filtered through a 0.7-µm glass fiber filter; do not rinse bottle	Chilled, must arrive at 4°C ± 2°C.

[1]When sample processed for NWQL replicate.

if necessary (table 5), then refrigerated before being shipped to the analyzing laboratory.

Filtered inorganic constituent samples (trace elements, major ions, and filtered nutrients) are filtered through a Pall Corporation 0.45-µm pore size disposable capsule filter. The capsule filter is a disposable self-contained unit composed of a pleated filter medium encased in a plastic housing that can be connected in-line to a peristaltic pump that generates sufficient pressure to force water through the filter. Capsule filters are cleaned before leaving for field sampling and only CH touches those portions of tubing that will be in direct contact with the deionized water (DIW) or capsule filter. The capsule filter is removed from protective packaging, the pump tubing is attached to the inlet connector of the capsule filter, and 2 L of DIW are pumped through the filter by CH. DH operates the pump while CH inverts the filter so the arrow on the housing is pointing upward to expel trapped air from the capsule during initial filling. DH removes tubing from the DIW reservoir and continues to operate the pump to drain as much of the DIW that remains in the capsule filter as possible. CH detaches the capsule filter from the pump tubing and puts it into a clean, sealable plastic bag.

The tubing is first rinsed by CH by placing the intake end of the pump tubing into the composite sample in the churn splitter to filter the sample water. DH starts the pump to rinse the tubing. The filter should be turned so that the outlet is pointing upward so the flow of the sample forces trapped air out of the capsule filter while it is filling. The sample bottles (excluding the prebaked amber glass bottles) should be rinsed three times with sample water before filling. Samples should be preserved if necessary (table 5), then refrigerated.

Samples should be kept below 4°C, but not frozen, until they are shipped to the analyzing laboratory (table 5). Samples should be packaged and shipped to the laboratory for analysis as soon as possible. Generally, the shorter the time between sample collection/processing and sample analysis, the more reliable the analytical results will be. Sample bottle caps are tightened to prevent leakage. Glass bottles are placed in foam sleeves. Sample bottles should be shipped in sealed plastic bags on ice in a cooler lined with doubled heavy duty plastic bags.

Fecal Indicator Bacteria

Fecal indicator bacteria are used to assess the microbiological quality of water. Though these bacteria do not usually cause disease, they are associated with fecal contamination and the presence of waterborne pathogens. The presence of *Escherichia coli* (*E. coli*) bacteria is direct evidence of fecal contamination from homeothermic organisms. Densities of fecal coliform bacteria can be associated with fecal contamination. Sterile techniques must be followed when collecting and processing samples for fecal indicator bacteria.

A list of equipment and supplies for field sampling and laboratory processing for bacterial analyses is shown in table 3. Powderless latex gloves should be worn at all times during sample collection. A hand-dipped centroid sample is collected. A 1-L autoclaved Nalgene sample bottle is grasped near the base with the sampler's hand and arm on the downstream side of the bottle. The open bottle mouth is plunged quickly downward below the water surface and the bottle is lowered in a manner that avoids contact with or disturbance of the streambed. The bottle is allowed to fill with the opening pointed slightly upward into the current. The bottle is removed with the opening pointed upward toward the water surface and tightly capped with about 2.5 to 5 cm (1 to 2 in.) of headspace for proper mixing.

After collection, the samples are immediately chilled in an ice-filled cooler at 1 to 4°C, but not frozen. Samples should be processed as quickly as possible. Membrane filtration (MF) is used for identification and enumeration of *E. coli* and fecal coliform bacteria. Enumeration is based on observation of reactions typical of the target bacteria on the test medium.

Powderless latex gloves should always be worn when processing bacteria samples and should be changed after each sample set. Sterile phosphate-buffered water amended with magnesium chloride is used to dilute samples and to rinse the filtration unit and utensils. Before sample processing in the laboratory, the appropriate sample volumes are selected and plates are labeled with the site number, the volume of the sample filtered, and the date and time of sample collection. Several sample volumes that are anticipated to yield one or two filters with counts in the ideal range are selected. The ideal colony count range (colonies per membrane filter) is 20 to 80 colonies for *E. coli* samples and 20 to 60 colonies for fecal coliform samples.

Laboratory countertops are cleaned with an antibacterial cleaning solution, such as 70-percent isopropyl or ethyl alcohol, before samples are processed. Incubators are preheated for at least 2 hours at specified temperatures. *E. coli* samples require temperatures of 35.0°C for 2 hours of resuscitation and 44.5°C for 22 to 24 hours of incubation. Fecal coliform samples require an incubation time of 44.5°C for 24 plus or minus 2 hours. Several sample volumes are selected [see chapter 7.1 of the USGS National Water National Field Manual for the Collection of Water-Quality Data (U.S. Geological Survey, variously dated) for details on sample size selection details]; if the sample volume to be plated is less than 1 milliliter (mL), dilutions with sterile buffered water in 99-mL dilution bottles are prepared. Transferring 11 mL of sample to a 99-mL dilution bottle creates a 1 to 10 dilution. Transferring 1 mL of sample to a 99-mL dilution bottle creates a 1 to 100 dilution. The dilution solutions also can be diluted in series as needed. For example, transferring 1 mL of a 1:10 dilution to a 99-mL dilution bottle creates a 1:1,000 dilution or transferring 1 mL of a 1:100 dilution solution to a 99-mL dilution bottle creates a 1:10,000 dilution. When preparing a dilution series, a sterile pipet is used to measure each sample volume. The dilution bottle is closed and shaken vigorously at least 25 times after each sample volume transfer.

The filtration unit is assembled by inserting the base of the sterile filter holder assembly into a side-arm flask. The

filtration unit is connected to a vacuum pump and the inside of the filtration unit is rinsed with sterile buffered water. Stainless steel rounded-tip forceps are sterilized by immersing the forceps tips in a small bottle or flask containing 70 to 90 percent ethanol. The forceps are then passed through an open flame of an alcohol burner. The alcohol is allowed to burn out and the forceps are allowed to cool. The filter is removed from its sleeve with the sterilized forceps and the sterilized funnel is removed from the base with one hand. While holding the funnel in one hand the membrane filter is placed on top of the filter base with the gridded side facing upward. Modified membrane-thermotolerant *Escherichia coli* (mTEC) filters are used for *E. coli* samples and membrane fecal coliform (mFC) filters are used for fecal coliform samples. The filter funnel is carefully replaced on the filter base. Care should be used to avoid tearing or creasing the membrane filter. Forceps are returned to the alcohol container. The samples are filtered in order of smallest to largest sample volume and the forceps are resterilized before each use. The sample should be shaken vigorously at least 25 times before each sample volume is withdrawn in order to break up particles and to ensure an even distribution of indicator bacteria in the sample container.

The required sample volume is removed by a sterile pipet by placing the tip in the center of the sample volume. A pipet bulb with a valve is used for volume control and the sample is pipetted onto the filter funnel. About 20 mL of sterile buffered water is poured into the funnel before pipetting the sample to allow even distribution of bacteria on the membrane filter. The pipet is allowed to drain and should be touched to the inside of the funnel to remove any remaining sample. A vacuum is applied—a pressure of about 5 pounds per square inch (psi) should not be exceeded to avoid damage to bacteria. The inside of the funnel is rinsed three times with 20 to 30 mL of sterile buffered water while a vacuum is applied. The funnel is removed and held in one hand while the membrane filter is removed using sterile forceps. The funnel is replaced on the filter base and the vacuum is released. A labeled plate is opened and the membrane filter is placed on the medium with the grid side facing upward using a rolling action. Air bubbles should not be trapped under the membrane filter. The plate is closed by pressing the top firmly onto the bottom. The plate is inverted and incubated within 20 minutes to avoid growth of interfering microorganisms. The other sample volumes are then filtered from smallest to largest volume order. The volumes filtered and the time of processing is recorded on the field and laboratory sheet (appendix 2). The countertop should be washed between each sample and the filter apparatus is washed and sterilized before the next use.

The plates are removed from the incubator after incubation. The number of target colonies are counted and recorded on the field forms for each sample volume filtered. The colonies are recounted until the results agree within 5 percent and the results are recorded on the field and laboratory sheet. Recounting is done by turning the plate 90 degrees to obtain a different view. The red to magenta colonies are counted under natural light for *E. coli* colonies. Fecal coliform colonies are wholly or partly light to dark blue, sometimes having brown or cream-colored centers. Whole numbers are reported for results less than 10 and 2 significant figures are reported for results greater than or equal to 10.

Volatile Organic Compounds

Collecting samples for volatile organic compounds (VOCs) is a special case of dip sampling that requires special equipment. Powderless latex gloves should be worn at all times. Samples for analysis of VOCs are collected as a single vertical point sample. The VOC sampler should be deployed where the stream velocity represents the average flow, which is typically near mid-channel in the cross section. When collecting VOC samples, special care must be used to avoid contamination from any oily film and debris floating on the stream surface. VOC samples are collected directly into USGS National Water Quality Laboratory (NWQL) laboratory-supplied prebaked 40-mL amber glass vials (table 5). The glass VOC vials are not to be cleaned or rinsed as they are supplied by NWQL ready to use. The VOC sampler must be cleaned after each use and field rinsed before use. The sampler is field rinsed by either submerging it in the stream for several minutes or dousing it three times with native water before inserting the VOC vials.

Four 40-mL unlabeled VOC vials are uncapped and placed into the VOC sampler in an area protected from any direct source of contamination after gloves are changed. The sampler top is secured and locked in position and the vial caps are stored in a clean, protected area. The sampler is lowered into the stream near mid-channel to about one-half of the total depth at that vertical. The sampler is held in one position until it is full. Air bubbles will rise to the surface while the sampler is being filled but may be difficult to see. It takes about 3 to 4 minutes for the sampler to fill and the sample is retained in the vials during the last 15 to 20 seconds of sampling. The sampler is removed when bubbles are no longer present, or after about 5 minutes, and returned to a protected area. The sampler is opened carefully and each vial is lifted slowly from the sampler reservoir using metal tongs—this should be done carefully to avoid losing the convex meniscus on each vial and should be preserved with 1:1 hydrochloric acid to pH less than 2.

The vial is quickly capped, shaken, and inverted to check carefully for air bubbles. If bubbles are present, the vial must be discarded. Three vials from the sampler set are required for one complete sample. A fourth vial may be needed if the water is extremely turbid. If two or more of the vials have air bubbles, resampling is necessary. The vials are dried, labeled, placed into a foam sleeve, and stored on ice for transport to the laboratory. The sampler is cleaned and stored properly.

Collection of Quality-Control Samples

The sources of variability and bias introduced by sample collection and processing affect the interpretation of water-quality data. Collection of quality-control samples ensures that

the data collected are compatible and of sufficient quality to meet program objectives. The quality of data collected must be documented by collecting quality-control samples. A series of quality-control samples (field blanks, replicates, and field-matrix-spike samples) is collected because the quality of the data collected and the validity of any interpretation cannot be evaluated without quality-control data.

Field blanks demonstrate that equipment-cleaning protocols adequately remove residual contamination from previous use, sampling and sample-processing procedures do not result in contamination, and equipment handling and transport between periods of sample collection do not introduce contamination. Field blanks for pesticides are collected immediately before processing native water through the sample processing sequence for field samples. Preparation of field blanks requires passing a volume of organic-free DIW through all sample equipment contacted by the actual sample. Field blanks for major ions and nutrients should be collected by the same approach, but using inorganic-free DIW after preparation of the organic blank.

Sample replicates provide information needed to estimate the precision of concentration values determined from sample processing. Each replicate sample is an aliquot of native sample water from a churn splitter and is processed immediately after the primary churn split sample using the same equipment, placed into the same type of bottle, prepared in the same way, and stored and shipped in the same way.

Field-matrix spikes assess recoveries from field matrices and assist in evaluating the precision of results for the range of target analytes in different matrices. A field-matrix spike is prepared by adding a standard spike solution provided by the USGS NWQL to a split of sample water processed in the same way as the regular sample. Matrix-spike kits with instructions are available from NWQL.

Quality assurance for *E. coli* analysis requires blanks for testing equipment and processing and regular maintenance of bacteria incubators. Each *E. coli* sample set should have an equipment blank and procedural blank. The equipment blank should be processed before the water sample is processed by rinsing the funnel with about 100 mL of sterile buffer water through a sterile membrane filter. The procedural blank should be processed after the equipment blank and water sample is processed by rinsing the funnel with about 100 mL of sterile buffer water through a sterile membrane filter. Quality-control blanks should be checked for colony growth and results are reported on the field and laboratory form (appendix 2). The presence of colonies on blanks indicates that results of the bacterial analyses are suspect and should not be reported or the results should be clearly qualified. One or more colonies on the equipment blank indicate inadequate sterilization of either the equipment or the buffered water, or a contamination during the sampling and analysis process. One or more colonies on the procedural blank indicate either inadequate rinsing or contamination of the equipment or the buffered water during sample processing. Bacteria incubator temperatures should be verified on a regular schedule.

Equipment Cleaning Procedures

Basic Water-Quality Constituents

Equipment is rinsed with DIW directly after use while it is still wet. Cleaned equipment is placed in doubled storage bags. Gloves should be worn throughout the cleaning process and changed after each cleaning step. A 2-percent detergent solution is prepared using a nonphosphate, laboratory-grade detergent. An acid solution is prepared using a 5-percent volume-to-volume dilution of American Chemical Society (ACS) trace-element-grade hydrochloric acid in DIW. Equipment and tubing should be in the detergent solution for 30 minutes and then scrubbed with a soft brush. Equipment is rinsed thoroughly with tap water. All nonmetal equipment and tubing is placed into the acid solution for 30 minutes. The exterior and interior surfaces of each piece of equipment is rinsed thoroughly with DIW and placed on a clean surface to dry. Cleaning equipment with metal parts is the same as non-metal, except the acid wash step is omitted. Dry, clean equipment is placed inside doubled plastic bags. Small equipment, parts, and tubing are placed in sealable plastic bags.

Fecal Indicator Bacteria

Equipment for the collection and analysis of bacterial samples must first be cleaned and then sterilized by autoclaving. Equipment is washed thoroughly with a dilute, nonphosphate laboratory-grade detergent. Equipment is rinsed three times with tap water and then three to five times with DIW. Autoclavable equipment is placed into autoclave plastic bags and sealed with autoclave tape. Equipment is autoclaved at 121°C at 15 psi for 15 minutes.

Volatile Organic Compounds

All equipment that will come in contact with the VOC sample should be soaked in a dilute, nonphosphate laboratory-grade detergent, rinsed with tap water, rinsed with VOC grade blank water, then air dried before sampling. The top and base of the VOC sampler should be submerged in a dilute solution of nonphosphate laboratory-grade detergent and scrubbed thoroughly with a nylon brush. A small squeeze bottle filled with the detergent solution should be used to flush the copper tubing. The sampler is rinsed thoroughly with warm tap water to remove all soap residue, rinsed thoroughly with VOC grade organic blank water, and allowed to air dry.

Streambed-Sediment Sampling Protocols

Streambed-sediment studies assess the occurrence and spatial distribution of trace elements and hydrophobic organic contaminants. Fine-grained particles and organic material are natural accumulators of trace elements and hydrophobic organic contaminants in streams, most of which are highly sorptive and associated with particulate material (Shelton and Capel, 1994). Though the water itself may contain only small

concentrations of these constituents, sediment may contain relatively large concentrations (Horowitz, 1991). Nonpoint source contributions of many of these contaminants may be intermittent or storm-related. As such, the contaminants may not be detected in single or periodic water samples. Bed sediments in depositional environments of streams provide a time-integrated sample of particulate matter transported by a stream. The objective for this section is to describe methods for collecting and processing streambed-sediment samples. These streambed-sediment protocols are adapted from the USGS NAWQA Protocol for Collecting and Processing Stream Bed Sediment Samples (Shelton and Capel, 1994) and the National Field Manual for the Collection of Water-Quality Data (U.S. Geological Survey, variously dated).

Equipment and Supplies

- Personal flotation device (PFD)
- High visibility clothing
- Hip or chest waders
- Latex gloves
- Polytetrafluoroethylene scoop
- Polytetrafluoroethylene spatulas
- 63-micrometer stainless steel sieve
- Glass compositing bowl
- Polytetrafluoroethylene wash bottle
- Plastic tarpaulin
- 500-milliliter glass jars
- First aid kit
- Camera
- Global Positioning System (GPS) unit

Sample Collection and Processing Procedures

The appropriate season and hydrologic conditions for sampling streambed sediment are determined by current and antecedent streamflow conditions. Unusually high flows can wash out, redistribute, or bury substantial parts of sediment deposits. Sampling should be delayed following high-flow events to allow deposition of fresh sediment. When sampling for bed sediment during summer or autumn, low-flow conditions are best to provide maximum direct access to the streambed and to minimize seasonal streamflow variability.

Locations in the stream where the energy regime is low and fine-grained particles accumulate in the streambed are depositional zones. Depositional zones can cover large areas at some sites and small pockets at other sites. The stream velocities at these zones have decreased and the fine-grained particles have deposited in the streambed. Depositional zones include areas on the inside bend of a stream or areas downstream from obstacles such as boulders, islands, sand bars, or shallow waters near the shore. Wadeable depositional zones are preferred because they are easy to identify and sample.

A list of key steps in sampling and processing streambed-sediment samples is shown in table 6. A streambed-sampling site is the reach of the stream approximately 100 m (328 ft) in length upstream from a water column sampling or streamflow measurement site. Five to 10 wadeable depositional zones containing fine-grained particulate material at each site should be identified. Depositional zones should be selected that represent upstream effects and various flow regimes, such as: left bank, right bank, center channel, and different depths of water. This will ensure that the sediment sample represents depositional patterns from various flow regimes and sources within the reach. Each depositional zone at a sampling site will be subsampled several times, and the subsamples will be composited with samples from other depositional zones sampled at the same site. Compositing will smooth the local scale variability and represent the average contaminant levels present at the site. All sampled zones should be underwater from the time of deposition until collection.

Table 6. Steps for streambed-sediment sample collection and processing.

Step order number	Action
1	Establish sampling reach.
2	Identify optimal sampling sites within established reach.
3	Put on latex gloves.
4	Establish sample processing area and rinse equipment with native water.
5	Take subamples from downstream sampling site.
6	Sieve sample into glass bowl.
7	Transfer sieved sediment into glass jars.
8	Repeat steps 5, 6, and 7 for all identified sampling sites.
9	Rinse all equipment with deionized water.

Before sampling depositional zones at each site, an area on the shore near the sampling zones is prepared to accumulate the sample and for later processing. While wearing latex gloves, the precleaned equipment is unwrapped on a clean plastic tarpaulin. The precleaned equipment is rinsed with native water.

The selected depositional zone is approached by moving upstream to avoid disturbing the area to be sampled and the sampling point is approached from downstream. The top 2 to 3 centimeters (0.8 to 1.2 in) of surficial fine material from the

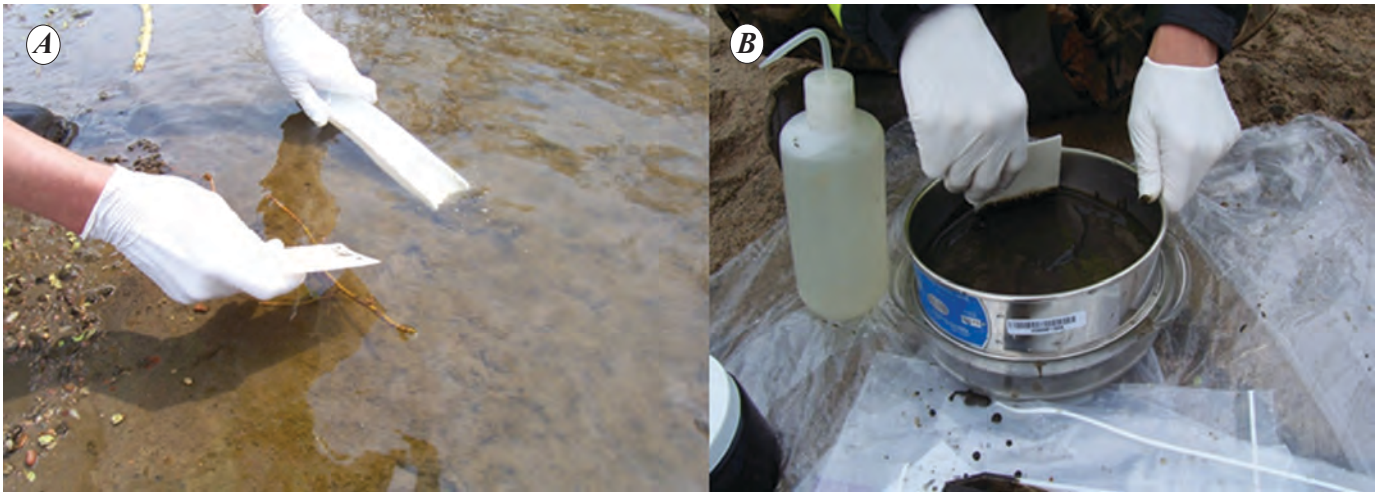


Figure 4. Streambed-sediment sampling using a polytetrafluoroethylene scoop and spatula (*A*) and a sediment sample being sieved through a 63-micrometer stainless steel sieve into a glass bowl (*B*).

streambed is removed with a polytetrafluoroethylene scoop (fig. 4*A*). The sample is brought to the stream surface and the fine sediments are protected from being washed away by the stream by placing a polytetrafluoroethylene spatula on top of the sample to hold it in place on the scoop. The material is deposited in a glass container for compositing with other subsamples from the same site. A site is sampled at 5 to 10 different depositional zones to complete the areal coverage and produce the necessary volume of material, which is approximately 1.5 L of wet sediment.

Latex gloves are worn while processing the sample. The composite sample is thoroughly mixed in the glass container with the polytetrafluoroethylene scoop. Laboratory analysis is done on the fine materials (less than 63 µm) and the bulk samples can be sieved in the field before shipping, or by the laboratory. The 500-mL glass jars are filled to be shipped to appropriate laboratories for sieving and analysis from the composited bulk sample. Bulk material that is field-sieved before shipping is removed from the composited glass container and small amounts are placed onto a 63-µm mesh sieve with the polytetrafluoroethylene scoop (fig. 4*B*). A stainless steel or polytetrafluoroethylene sieve is used for material analyzed for organic compounds. A plastic or polytetrafluoroethylene sieve is used for material analyzed for trace elements. Native water that has been collected directly from the stream into a 500-mL polytetrafluoroethylene wash bottle is used to elutriate the sediment sample. The fine sediments pass through the sieve with a stream of water delivered by the wash bottle. Small amounts of sediment material are worked through the sieve and the material remaining on the sieve is discarded. It is not necessary to sieve all of the material that is less than 63 µm in each aliquot. Sieved sediments are then transferred into 500-mL glass jars.

Collection of Quality-Control Samples

Quality data are assured through a sampling and analytical approach designed to minimize or compensate for potential sources of contamination and variability. Quality assurance is verified through independent sampling and analyses. The awareness and avoidance of chemical contamination are necessary in each step of sample collection and processing: sampling, subsampling, field processing, shipping, and laboratory processing. Because sediments are natural accumulators of analytes, there is less concern of gross sample contamination than in the water column. Glass and polytetrafluoroethylene are the materials of choice to make contact with the bed sediments.

The primary potential sources of variability in streambed-sediment composition at a site are temporal variability, areal variability among depositional zones, areal variability within depositional zones, and depth variability. Temporal variability is managed by collecting all samples during low flow conditions when changes with time are expected to be minimal. Areal variability is minimized by compositing samples within and among zones to yield an average for the reach. Variability in depth is managed through a consistent sampling approach of surficial sediment, visual inspection, and sampling depth management.

The quality-assurance steps designed into the sampling strategy and methods will be verified during the initial phase of sampling by a comparative study of duplicate sample collection and analyses that aggregates all potential sources of variability. If the verification indicates quality-control problems, more specific tests will be designed as required. About 10 percent of samples collected per year should be collected as quality-control samples and should include replicates and blanks. Replicates are two concurrent samples that are collected using identical methodology as closely together in time

and space as possible. Concurrent sample data are intended to provide the user with a measure of sampling precision and are intended to indicate inhomogeneities in the system being sampled.

Sample Handling, Storage, Delivery, and Shipping

Streambed-sediment samples are refrigerated at a temperature that is less than 4°C but above freezing. Samples for nutrient and trace-element analyses should be packed without ice for shipping to the laboratory that processes nutrient and trace elements. Samples for organic-contaminant analyses are placed into a protective sleeve, packed in ice (but not frozen), and shipped to the USGS NWQL.

Equipment Cleaning Procedures

All equipment is rinsed thoroughly with DIW in the field after sampling. All equipment should be cleaned before field activities and between sites. Cleaning procedures are designed to control contamination by removing paper, glue, plasticizers, oils, and metals from the sampling and processing equipment. The equipment should be stored in a plastic food storage container after cleaning. Equipment is soaked in a 0.2-percent phosphate-free detergent solution for 30 minutes and scrubbed with a soft brush. Equipment is then rinsed with copious amounts of tap water and then with copious amounts of DIW. The equipment is acid washed for trace element analysis. Finally, the equipment is rinsed with DIW and allowed to air dry. Cleaned and dried equipment is stored inside sealable polyfluorocarbon or other uncolored plastic bags.

Selection of Biological Sampling Reaches

Biological sampling includes collection of periphyton, macroinvertebrate, and fish data. A reach should be selected that includes most of the in-stream habitat types that are representative of the stream during biological sampling. The same reach is sampled for periphyton and macroinvertebrates. The fish sampling reach should coincide with the periphyton and macroinvertebrate reach, but is determined in a more strict empirical manner.

The periphyton and macroinvertebrate sampling reach should include a repeating series of the main habitat types to accommodate replicate sampling. The reach length generally is determined based on the number of replicates needed, the availability of habitat types, and the longitudinal distance in which the habitats repeat in occurrence (Barbour and others, 1999). Reach length for each site is the distance that is needed to capture a repeating series of the typical habitats in the stream and this can be based on: riffle-pool or glide-pool series, meander bend-channel crossover series, or a combination of these. In general, reach length will be greater in larger streams and in cases where multiple replicate samples are needed. This approach also allows addition of study sites in different stream types or those differing in discharge or watershed area.

A representative fish sampling reach length is specified as 20 times the average wetted width, which should ensure that all habitat types are represented within the reach (Fitzpatrick and others, 1998). Reach length should range from 150 to 300 m [164 to 328 yards (yd)] for wadeable streams and 500 to 1,000 m (547 to 1,094 yd) for nonwadeable streams (Fitzpatrick and others, 1998; Meador and others, 1993).

Periphyton Sampling Protocols

Periphyton consists of the attached algae that grow on submerged surfaces in water bodies, such as rocks, woody debris, and sand (Allan, 1995). Periphyton are primary producers and serve as a primary link between abiotic factors and higher trophic levels. Algae have short life cycles and respond rapidly to changes in environmental conditions. Physical, chemical, and pollution tolerances and growth optima have been described for many periphytic algal species, allowing periphytic communities to be used as indicators of ecological conditions. Because periphyton is an intermediate between the physical, chemical, and biotic factors in aquatic systems, periphyton is an important indicator of stream health (Lowe and Pan, 1996). These periphyton protocols are adapted from those used by the Kansas Biological Survey (Bouchard and Anderson, 2001), the U.S. Environmental Protection Agency's (USEPA) rapid bioassessment protocols (RBP; Stevenson and Bahls, 1999), and the USGS revised protocols for sampling algal, invertebrate, and fish communities as part of NAWQA (Moulton and others, 2002).

Equipment and Supplies

- Personal flotation devices (PFDs)
- High visibility clothing
- Hip or chest waders
- Bar clamp sampler
- Petri dishes
- Spatula
- Beakers
- Plastic pipettes
- Test tube brushes
- Meter stick
- Squirt bottles

- Filter base and funnel
- Erlenmeyer flask
- Tubing
- Hand pump
- Filters
- Stapler
- Periphyton sampling field data sheet
- Copies of field protocols
- Pencils and clipboard
- First aid kit
- Global Positioning System (GPS) unit

Sample Collection Procedures

A list of key steps for periphyton collection is shown in table 7. Periphyton samples are collected during normal or low-flow periods and sample collection is delayed at least 2 weeks after high-flow events that have disturbed and removed stream periphyton communities. Sampling begins at the downstream boundary of the sampling reach and progresses upstream to minimize disturbing the stream bottom. All sampling equipment is rinsed three times with distilled water before use, between each sample collected between each site, and before the equipment has had time to dry.

Table 7. Steps for periphyton sample collection.

Step order number	Action
1	Determine available habitat types.
2	Select necessary samplers.
3	Collect 15 cobbles or substrate samples.
4a	If collecting cobbles, take cobbles to onsite processing station and transfer periphyton to beaker using bar clamp sampler.
4b	If collecting substrate samples, use petri dish and spatula to place samples into a beaker.
5	Measure volume of composited sample and pour into amber bottle.
6	Vigorously shake sample.
7	Filter two aliquots for chlorophyll analysis.

Periphyton is collected from unattached hard substrates (cobble or woody debris) in riffles and runs when present. A single habitat sampling approach for periphyton helps to minimize variability among sites because of differences in habitat (Stevenson and Bahls, 1999; Moulton and others, 2002). Five unattached hard substrates approximately 25.6 to 64 cm (10 to 25 in.) in diameter (Wentworth, 1922) are collected randomly from three adjacent riffles, when present, at each site. If three riffles are not present, five randomly collected unattached hard substrates from three run habitats are sampled. If no substrate is available within the stream reach, five loose sediment (sand, silt, fine-particulate organic material, or clay) samples are collected randomly in three areas of the reach. At each sampling area the left bank location is recorded using a GPS unit. A meter or yard stick is used to measure and record water depth at each sampling location.

For sites with unattached hard substrates available, the substrates are placed in a plastic bucket and transported to an onsite processing station. A bar clamp sampler is clamped onto a smooth part of each substrate and a new test tube brush is used to scrub periphyton from the known area of the surface of the substrate (fig. 5). The periphytic material is wetted with 0.7-µm filtered streamwater by a squirt bottle and transferred to a 250-mL beaker using a fresh plastic pipette. This process is repeated several times until all of the visible periphyton is removed from the sampling area, which is typically approximately 8.6 square centimeters (cm^2; 1.33 square inches) of each cobble.

Figure 5. Periphyton sampling with the bar clamp sampler and test tube brush.

At sites where unattached hard substrates are not present, an inverted petri dish is used to sample the surface of loose sediments. Five randomly selected samples from each of three sampling locations for a total of 15 samples per reach are collected. At each sampling location, the lid of a small plastic petri dish (about 17 cm^2 or 2.6 square inches) is held upside down in the water and the inside of the lid is rubbed to remove air bubbles. The inside of the lid is turned toward the substrate to be sampled without disturbing the sediments. The petri dish lid is carefully and slowly pressed into the streambed sediment. The lid is slid onto a spatula to enclose the discrete collection. While the petri dish is held tight against the spatula, extraneous sediment from the spatula is carefully washed

away, and the petri dish is then lifted out of the water. The lid is inverted and the spatula is removed. The sediment is rinsed from the lid with a squeeze bottle filled with filtered stream water into a 500-mL beaker. This procedure is repeated at each additional sampling location in the reach, yielding a composite sample of 15 discrete collections in the 500-mL beaker.

The composited sample in the beaker is made up of 15 discrete collections from 15 cobbles or sediment locations. After all samples are composited, periphytic material is rinsed from the beaker into a graduated cylinder. Sample volume is recorded and the sample is poured into a 1-L high-density polyethylene (HDPE) amber bottle (Stevenson and Bahls, 1999; Moulton and others, 2002; Hambrook-Berkman and Canova, 2007). The sample is vigorously shaken and split into three aliquots as described below. Two aliquots are processed for chlorophyll and one for taxonomic identification and enumeration.

Chlorophyll samples are processed as described in Hambrook-Berkman and Canova (2007). The chlorophyll filtration apparatus is assembled by attaching a filter base with a rubber stopper to the filtering Erlenmeyer flask. The flask is joined to a hand-operated vacuum pump (with gauge) with a section of tubing. Using clean forceps, a 47-mm glass-fiber filter is placed on the filter unit base and the funnel is clamped onto the filter unit. The filter is wetted with deionized water. After the sample is homogenized by being shaken vigorously for at least 30 seconds, a 10-mL (generally) subsample is collected using a pipette and dispensed into a graduated cylinder. The volume of the subsample is recorded. The subsample is poured through the filter using additional deionized water if necessary while the hand pump is pumped to create a vacuum that does not exceed 15 psi. The graduated cylinder is rinsed with deionized water several times onto the glass-fiber filter. The filter is examined—an adequate amount of algal biomass for analysis is indicated by the green or brown color of material retained on the filter. Additional 10-mL subsamples are filtered until an appropriate amount of biomass is retained on the filter. The number of 10-mL aliquots filtered is recorded. The funnel sides are rinsed with deionized water and the filter is removed from the funnel base with forceps. The filter is folded in half with the filtered biomass inside. Each filter is wrapped in a larger labeled (sample site, date, and volume filtered) filter paper house and the edges of the larger filter are stapled closed. The filter house is labeled and housed in a container with dessicant. This process is repeated once more for a duplicate chlorophyll sample. The container with chlorophyll samples and dessicant is stored in a cooler filled with ice and placed immediately in the freezer upon return to the laboratory. After the duplicate chlorophyll samples have been collected, the sample volume remaining in the sample bottle is determined by subtracting the total volume of sample used for chlorophyll samples from the total volume of sample recorded previously and recorded.

Samples for taxonomic identification and enumeration are preserved with a 9:1 Lugol's iodine:acetic acid solution. Approximately 1 mL of the Lugol's iodine solution is used to preserve each 100 mL of sample. A sample label is placed on the sample bottle (sample site, date, sampling area, and sample volume). After preservation, the sample is stored in a cooler filled with ice and placed in the refrigerator immediately upon return to the laboratory. The known areas for all 15 unattached hard surfaces or petri dishes in the samples are summed to determine total surface area sampled. After sample collection is completed at each site, all field sampling equipment is rinsed three times with stream water, then three times with deionized water.

Chlorophyll is analyzed at the USGS Kansas Water Science Center in Lawrence, Kansas. Total chlorophyll (uncorrected for degradation products) is extracted in heated ethanol (Sartory and Grobbelar, 1986) and analyzed fluorometrically using the USEPA method 445.0 (Knowlton, 1984; Arar and Collins, 1997). BSA Environmental Services, Incorporated, Beachwood, Ohio, analyzes periphyton samples for taxonomic identification, enumeration, and biovolume of diatoms and soft algae. The soft algae in the periphyton samples are first enumerated to the lowest possible taxonomic level using membrane-filtered slides (McNabb, 1960). A minimum of 400 natural counting units are counted. A natural counting unit is a natural grouping of algae such as each individual filament, colony, or isolated cell. Diatoms are counted by natural counting unit as a general category, and then examined more closely in permanent diatom mounts. Diatom slides are made using the traditional nitric acid digestion method (Patrick Center for Environmental Research, 1988). A minimum of 400 valves (cells) are identified to the lowest possible taxonomic level. Biovolume, calculated using mean measured cell dimensions, is an estimate of algal biomass. Biovolume factors for soft algae and diatoms are calculated using the methods described in Hillebrand and others (1999). Diatom biovolumes are calculated from the permanent slides. A mean biovolume measurement per cell is calculated for each sample, and that value is used as the biovolume measurement in the general diatom category.

Macroinvertebrate Sampling Protocols

Macroinvertebrates have been used extensively in water-quality monitoring (Cairns and Pratt, 1993; Merritt and others, 2008). Benthic macroinvertebrate assemblages have several advantages for evaluating stream quality; they are good indicators of localized conditions, they integrate the effects of short-term environmental variations, they are made up of species that constitute a broad range of trophic levels and pollution tolerances, and sampling them is relatively easy and inexpensive (Barbour and others, 1999). The objective of this section is to provide a description of the methodology used for collecting a representative sample of the macroinvertebrate community. Macroinvertebrate sampling protocols vary among state agencies and across other federal agencies. Many protocols are designed for screening-level assessments and tend to be qualitative, such as those outlined in the USEPA RBP (Barbour and

others, 1999). These protocols will use the Kansas Department of Health and Environment's (KDHE) qualitative macroinvertebrate protocols for assessing biological quality of streams and rivers (Kansas Department of Health and Environment, 2000), which have been used for stream evaluations in water-quality monitoring studies within the state (Poulton and others, 2007; Rasmussen and others, 2009; Graham and others, 2010).

Equipment and Supplies

- Personal flotation devices (PFDs)
- High visibility clothing
- Appropriate collection permits
- Hip or chest waders
- 9-inch by 18-inch rectangular frame kicknets with a 500-micrometer mesh size
- Large white sorting trays mounted on a portable stand
- Forceps
- 125-milliliter polyethylene sample bottles containing 80-percent ethanol
- Hand counters
- Stopwatch
- Macroinvertebrate sampling field data sheet
- Copies of field protocols
- Pencils and clipboard
- First aid kit
- Camera
- Global Positioning System (GPS) unit

Sample Collection Procedures

A list of key steps for macroinvertebrate sample collection is shown in table 8. Before invertebrate sampling begins, the sampling reach and types of in-stream habitats present in the reach must be identified and documented. The KDHE macroinvertebrate protocol calls for two 100-organism field-sorted samples to be collected with a standard rectangular frame kicknet (9 in. by 18 in. with 500-µm mesh, fig. 6*A*) simultaneously by each of two individual biologists working independently of one another. For each of the two biologists, sampling ends after 1 hour, even if the 100-organism count has not been reached. Both 100-organism samples are later pooled into one 200-organism sample. For replicate sampling at a site, the reach is divided into three subequal parts, and the process is repeated for each of the two additional replicates. The KDHE protocol does not provide a specific definition on how to partition the sampling among habitats, but does include the following guidelines: no more than one-third of the organisms should come from any one habitat alone, the sampling should maximize the diversity of organisms collected at a site, habitats should be sampled in a systematic fashion so that the source of the organisms collected is spread out over all of the habitats in proportion to their availability, and the relative abundance of organisms collected should best represent their relative abundance in the stream across all habitats (Kansas Department of Health and Environment, 2000). To provide more consistency and efficiency during macroinvertebrate sampling, these protocols will use two additional tools: a streamside sorting tray elevated with a portable stand to improve visibility of sorting organisms from debris (fig. 6*B*, *C*) and a checklist of stream habitat types as a guideline for systematic sampling to reduce bias and provide thorough coverage of the habitats present.

During sampling, all available macrohabitats (riffles, pools, runs) and microhabitats (various water depths, velocities, or substrata within a macrohabitat) are sampled according to their relative availability. For each replicate, both biologists sample different parts of the same habitats, excluding areas that they previously disturbed. Macroinvertebrate specimens are collected by disturbing riffles and leaf packs and allowing the current to carry dislodged organisms (and debris on which organisms may occur) into the kicknet. The net also is swept through submerged or floating aquatic vegetation, submersed terrestrial vegetation and tree roots, accumulations of woody debris, and growths of filamentous algae. Fine sediments (silt and fine sand) are sieved by rinsing stream water through the nets. Samples are emptied into the sorting tray (fig. 6*B*, *C*) and a small amount of water is placed in the sorting tray along with the sample debris to enhance the visibility of the organisms. A hand counter is used to count the organisms as they are removed from the tray with forceps (fig. 6*C*). To obtain additional macroinvertebrate taxa during the 1-hour period, attached organisms are also hand-picked

Table 8. Steps for macroinvertebrate sample collection.

Step order number	Action
1	Establish sampling reach.
2	Identify and document instream habitat types.
3	Set up and inspect equipment.
4	Ensure sampling bottles are properly labeled.
5	Begin stopwatch.
6	Collect kicknet sample.
7	Process sample in sorting tray.
8	Repeat steps 6 and 7 in all habitat types until nearly 100 organisms are collected or 60 minutes has passed.
9	Hand-pick additional taxa from stationary substrates until 100 organisms are collected or 60 minutes has passed.

Figure 6. Macroinvertebrate sampling. Using a rectangular frame kicknet (*A*), streamside macroinvertebrate collection using white sorting trays (*B*), and macroinvertebrate collection using white sorting tray, forceps, polyethylene sample bottle containing 80-percent ethanol, and hand counter (*C*).

from stationary substrates in the stream such as large woody debris, large cobble, or artificial objects. As specimens are separated from debris they are placed directly into 125-mL polyethylene sample bottles containing 80 percent ethanol (fig. 6*C*). Bottles are identified using a waterproof marker and external waterproof labels with site number and collection date.

Quality-Assurance and Quality-Control

Sample labels must be properly completed, including site identification, date, and collector's name. After sampling has been completed at a given site, all equipment that has come in contact with the sample should be rinsed thoroughly, examined carefully, and picked free of organisms and debris. The equipment should be examined again before use at the next sampling site.

Quality-assurance and quality-control measures for macroinvertebrate identification, enumeration, and data entry follow those outlined in Moulton and others (2000) and include within-laboratory cross checking of specimen identification. Updated taxonomic keys and voucher specimens are kept on file at the NWQL. Additional quality-assurance measures include repeats of identification and enumeration procedures on the same sample by different laboratory technicians and a full comparison of bench sheets for a minimum of 10 percent of the samples.

Sample Handling, Storage, and Shipping

For each replicate collected at a site, the two independent samples are combined into one 200–organism sample and the sample bottle preservative is decanted and refilled with 80-percent ethanol to reduce the chance of spoilage. Sample bottles are sealed with electrical tape. Samples are shipped to the USGS NWQL for identification and enumeration.

Fish Sampling Protocols

In addition to macroinvertebrates, fish assemblages commonly are used in evaluating water quality (Barbour and others, 1999). Because most fishes are mobile and tend to live longer than most macroinvertebrates, they may be better indicators of stressors (Karr, 1981; Barbour and others, 1999). The objective of this section is to describe methods for collecting a representative sample of the fish community from the study stream. These fish protocols are adapted from the USEPA RBP (Barbour and others, 1999) and the USGS revised protocols for sampling algal, invertebrate, and fish communities as part of NAWQA (Moulton and others, 2002).

Equipment and Supplies

- Personal flotation devices (PFDs)
- High visibility clothing
- Hip or chest waders
- Appropriate collection permits
- Backpack or tote barge-mounted electrofisher
- Dip nets
- Seines
- Elbow-length lineman's gloves
- Chest waders
- Polarized sunglasses
- Buckets/livewells
- Jars for voucher/reference specimens
- Waterproof jar labels
- 10-percent buffered formalin
- Measuring board
- Balance
- Tape measure
- Fish sampling field data sheet
- Applicable topographic maps
- Copies of field protocols
- Pencils and clipboard
- First aid kit
- Global Positioning System (GPS) unit

Sample Collection Procedures

Key steps for fish sampling are shown in table 9. Fish collection procedures focus on a multihabitat approach–habitats are sampled in relative proportion to their local representation. Each sample reach should contain riffle, run, and pool habitats, where available. Whenever possible, the reach should be sampled sufficiently upstream from any bridge or road crossing to minimize the hydrological effects on overall habitat quality. Wadeability and accessibility may ultimately govern the exact placement of the sample reach. The location of the downstream and upstream limit of the reach is recorded on each field data sheet.

Table 9. Steps for fish sample collection.

Step order number	Action
1	Establish a sampling reach that is representative of the stream and record the downstream and upstream limits of reach.
2	Prepare sampling equipment and put on waders, gloves, and polarized glasses.
3	Determine electrofisher output voltage for effective sampling.
4	Electrofish in a downstream to upstream direction.
5	Process fish.
6	Electrofish in a downstream to upstream direction.
7	Process fish.
8	Seine for fish in an upstream to downstream direction.
9	Process fish.

Each reach has various in-stream habitats consisting of different geomorphic channel units, substrates, and hydrologic conditions. Fish species are distributed in the stream reach according to these in-stream habitats. No single fish collection equipment or method is adequate to sample all habitats. Therefore, two complementary methods should be used for sampling fish: electrofishing and seining. Electrofishing is done in two separate passes of the reach and the fish collected from the first pass are processed before the second pass. Seining is done after electrofishing. Three seine collections are taken and combined before fish processing.

The conductivity of the stream is measured before electrofishing to determine the appropriate output voltage for effective sampling. A backpack or tote barge electrofisher can be used in wadeable streams. Backpack electrofishing with a single anode is usually most effective in shallow (less than 1 m or 3.3 ft deep) and narrow (less than 5 m or 16.4 ft wide) streams. Towed electrofishing gear (a tote barge) with multiple anodes is usually more effective in wide (greater than 5 m or 16.4 ft) wadeable streams with pools deeper than 1 m (3.3 ft). Channel width, depth, and access should be considered before choosing between backpack and towed electrofishing methods. As a general rule, the output voltage from the electrofisher should be about 3,000 watts.

Electrofishing techniques for wadeable streams require an electrofishing crew consisting of three to six individuals. Each crew member must be insulated from the water and the electrodes; therefore, chest waders and lineman's gloves are required. Electrode and dip net handles must be constructed of insulating materials (such as wood or fiberglass). Electrofishers/electrodes must be equipped with functional safety switches. Field crew members must not reach into the water unless the electrodes have been removed from the water or the electrofisher has been disengaged. When using backpack electrofishing gear, one crew member is designated as the operator. With towed gear, three crewmembers are designated as

operators. Two crew members are assigned to collect stunned fish with dip nets. One additional crew member is sometimes needed to transfer netted fish into a flow-through holding bottle or live cage. All crew members must wear polarized glasses to enhance their ability to see fish that have been stunned by the electrical field. Regardless of the electrofishing gear used, two separate electrofishing passes are made in the reach. The fish collected in the first pass are processed before the second electrofishing pass begins. Fish from the first pass are released so they will not be caught in subsequent sampling efforts. Fish community data for each pass are kept separate.

Sampling begins at the downstream end of the designated sampling reach and proceeds upstream. Collection begins at a shallow riffle, or other physical barrier at the downstream limit of the sample reach, and terminates at a similar barrier at the upstream end of the reach. The distance of the dip netters from the anode increases with current velocity and turbidity. All geomorphic channel units and in-stream habitat features, such as woody snags, undercut banks, macrophyte beds, or large boulders within the wadeable sampling reach are sampled using pulsed direct current. This technique may require electrofishing from one shoreline to the other in a "zigzag" pattern, while consistently sampling all areas within the reach.

Although electrofishing is the single most effective method for sampling stream fish communities, it is biased toward collection of larger fish (Wiley and Tsai, 1983). Therefore, seining is used to complement electrofishing collections. Seining is an effective method for sampling small-sized individuals (less than 10 cm or 3.9 in. total length). Seining is always conducted following electrofishing, except for instances when seines are used as barriers to fish escaping an electrical current. Seines are sampling devices that trap fish by enclosing or encircling them and are manufactured in a variety of dimensions and mesh sizes. The USGS NAWQA program uses 6.4 mm (0.25 in.) as a standard mesh size for seines. Three sizes of seines are commonly used to sample fish communities: the "common sense" seine, the bag seine, and the beach seine. The common sense ("minnow" or "standard ichthyological collection") seine is 3 m by 1.2 m (3.3 yd by 1.3 yd) and is attached to two brails. The bag seine is 7.6 m (8.3 yd) or 9.1 m (10 yd) and typically has a bag or pocket in the center of the seine. As the bag seine is pulled through the water, fish are herded toward the center of the net and into the bag. A beach seine is typically used along the shorelines of large bodies of water and is usually greater than 30 m (33 yd) long. Regardless of the seine used, three seine collections are collected and combined before processing fish. Seining is generally more effective moving in a downstream direction at about the current velocity (Moulton and others, 2002).

The goal of processing collections of fish in the field is to collect information on taxonomic identification, length, weight, abundance, and the presence of external anomalies with minimal harm to specimens that will be released alive back into the stream. Fish are stored into identifiable and unidentifiable groups. Threatened and endangered species, species of concern, and game fish are processed first. Unidentifiable species are preserved and identified in the field office. Fish are held and anesthetized in a manner consistent with minimizing stress and death. Fish are identified and enumerated. The total length and weight of at least 30 specimens of each species (excluding threatened and endangered species) are measured. As many as 30 specimens of each species (excluding threatened and endangered species) are examined for external anomalies. Data are recorded on a data sheet. Selected specimens for identification in the laboratory are preserved and all other specimens are released alive back into the stream. Because the collection methods used are not consistently effective for young-of-the-year fish and because their inclusion may seasonally skew bioassessment results, fish less than 20 mm total length will not be identified or included. Fish should be released downstream from the sampling reach to minimize the potential for resampling and sample bias.

Identifications are made by a crew member who is familiar with the fish species commonly found in the study area. An attempt is made in the field to identify all fish to the species level. Uncertain identifications require that those specimens be vouchered for later identification in the laboratory. Length measurements are determined by using a measuring board consisting of a linear metric scale on a flat wooden or plastic base with a stop at the zero point. Total length measurements are recorded individually for as many as 30 individuals of each species collected from the reach. These 30 individuals are selected from the total number of individuals to represent the range of lengths present in the sample for a particular species. The fish are positioned with their body on its right side, the head facing the observer's left, and the mouth closed. The snout of the fish is pushed against the measuring board stop. The total length is measured as the distance from the closed mouth to the extreme tip of the caudal fin when the lobes of the caudal fin are squeezed together. Total length is recorded to the nearest millimeter on the data sheet.

Weight measurements are obtained by using portable electronic and hanging scales. Weight is determined for each fish that is measured for total length. However, for individuals of a species weighing less than 1 g, a batch weight is recorded; an average individual weight for a given species can be calculated by dividing the batch weight by the total number of individuals weighed in the batch. The scale is leveled, calibrated, and tared. The fish are placed on the scale and the weight is recorded to the nearest gram on the data sheet.

External anomalies are externally visible skin or subcutaneous disorders or parasites. External anomalies might indicate the presence of sublethal environmental stresses, intermittent stresses, behavioral stresses, or chemically contaminated substrates. External anomalies include deformities, eroded fins, lesions, and tumors collectively referred to as "DELT anomalies" (Sanders and others, 1999). All fish that are individually measured and weighted also are examined for the presence of DELT anomalies. Caution should be used not to include injuries that might have resulted from the collection methods. Deformities are skeletal anomalies that affect the head, spinal vertebrae, and fins. Eroded fins are reductions in

fin surface area and hemorrhaging along fin rays, which can be caused by chronic disease, parasite infestation, or poor water quality. Lesions are defined as tissue alterations that include ulcerated, reddened tissue, open sores, or exposed tissue. Tumors are circumscribed growths of tissue growing independently of the structural development of the fish and serving no physiological function.

Specimens that are not positively identified in the field are preserved, labeled, and returned to the laboratory for later identification. Fish are euthanized with an overdose of anesthetic (for example, carbon dioxide). All specimens of a particular species or similar looking unknown species are placed in a sample bottle. There should be sufficient space in the jar for adding a necessary volume of 10-percent buffered formalin to preserve tissues. A small incision along the right side of the body is made to allow buffered formalin to penetrate the body of fish specimens longer than 100 mm (3.9 in.) total length. A completed sample label is placed inside each bottle. Enough 10-percent buffered formalin is added to fill the sample bottle and the bottle is tightly sealed. The fish are left in the buffered formalin for 2 to 7 days to preserve tissues. Fish may be prepared for long-term preservation and storage after being returned to the laboratory and properly preserved according to Walsh and Meador (1998).

Stream Habitat Assessment Protocols

Evaluating stream habitat is crucial to any assessment of ecological integrity and the presence of an altered habitat structure is considered one of the primary stressors of aquatic systems (Karr and others, 1986). The goal of stream habitat characterization is to relate habitat to other physical, chemical, and biological factors that describe water-quality conditions. Most habitat assessment protocols contain a synthesis of specific ecological variables that can be scored for a range of conditions of relative levels of quality. The objective of this section is to provide a concise set of protocols to evaluate stream habitat conditions. These habitat assessment protocols are adapted from the USEPA RBP (Barbour and others, 1999) and the protocols described in Rasmussen and others (2009). Some variables are suitable for assessing either high-gradient or low-gradient streams. When appropriate, the proper variable must be selected depending on the stream gradient.

This assessment protocol integrates data for three habitat categories: channel, stream bank/riparian, and in-stream aquatic. Variables measured in the channel category include indicators of overall channel morphology such as channel slope and sinuosity. Variables included in the stream bank/riparian category provide information on organic material sources, bank conditions, and the degree of disturbance in the riparian zone. Variables in the in-stream aquatic category provide information on the availability of cover and substrate materials and the stream's capacity for meeting basic physical requirements for support of a diverse and well-balanced aquatic community. There are a total of 14 habitat variables (table 10) that are scored on a scale of 1 (poor conditions) to 12 (optimal conditions, appendix 3).

Table 10. Variables included in habitat assessment (Rasmussen and others, 2009; Barbour and others, 1999).

Habitat assessment variable
Flow status.
Channel slope and morphological status.
Sinuosity and riffle frequency.
Bank stability.
Canopy cover.
Bank and riparian protection.
Length and extent of buffers.
Mean buffer width.
Percentage of altered banks.
Substrate fouling.
Velocity/depth combinations and pool variability.
Riffle substrate embeddedness and pool substrate composition.
Sediment deposition.
Diversity of epifaunal substrate and cover.

Habitat data are evaluated at two hierarchical scales [small (stream segment) and large (stream reach)] using a classification system proposed by Frissell and others (1986) and slightly modified by the NAWQA program (Fitzpatrick and others, 1998). Segment-scale data also are obtained from geographic information system (GIS) coverages, topographic maps, and aerial photographs. A stream segment is defined as a section of stream that is relatively homogeneous with respect to physical, chemical, and biological properties and generally bounded by tributary junctions, point-source discharges, or other features that might be expected to change stream properties (Fitzpatrick and others, 1998). The upstream boundary of the segment is defined by a change in stream order or the presence of a point source discharge. The downstream boundary of the segment is defined as 50 m (54.7 yd) downstream from the downgradient boundary of the reach. Reach-scale data are collected in the field. The reach is a section of the stream where a streamgage is located and where biological sampling occurs. To capture habitat diversity that is representative of the segment, the reach should include at least two riffle-pool sequences or at least two runs or glides where current velocity is greater than in pools. If two riffle-pool or glide-pool sequences are not present, the stream reach should be 40 times the mean wetted width at base flow or 20 times the narrowest wetted width. Another method of determining the reach is to include at least two complete riffle-pool sequences.

Equipment

- Personal flotation device (PFD)
- High visibility clothing
- Hip or chest waders
- Clinometer
- Aerial photographs
- Topographic maps
- Densiometer
- Stream Habitat Assessment Sheet (appendix 3)
- Clipboard and pencils
- Camera
- Global Positioning System (GPS) unit

Channel Characteristics

Channel characteristics are indicators of channel condition that may have direct or indirect effects on aquatic biota and are related to stream morphology and hydrology.

Flow Status

Flow status (Barbour and others, 1999) is a reach-scale variable that indicates the extent of streambanks and substrate materials exposed during base-flow conditions. When water does not cover much of the streambed, the amount of suitable substrate for aquatic organisms is reduced. The flow status changes as the channel changes (during aggradation of the streambed, for example) or as flow decreases or increases (as a result of irrigation diversion, drought, or municipal discharge, for example). Flow status is most useful for interpreting biological condition under abnormal or minimal flow conditions. Optimal flow-status conditions for biota exist when water reaches the base of both streambanks and a minimal amount of channel substrate is exposed. Conditions are poor when minimal water is present in the channel and water occurs mostly as standing pools.

Channel Slope and Morphological Status

Channel slope and morphological status (Rasmussen and others, 2009) is a reach-scale measurement of the slope of streambanks in relation to the channel and channel shape. This variable is an indicator of the degree of incision, downcutting, or headcutting that has occurred in the channel. Downcutting and lateral cutting can impair function because of increased scour and downstream sediment transport. Downcutting channels frequently have changes in the elevation of the stream bottom and steeper angles between the bottom of the channel and the top of the streambanks. Bank slope is measured using clinometer readings and visual estimation at 10 evenly spaced points along the reach length for right and left (determined by looking downstream) streambanks. Percentage bank slope measurements are made at each transect from the middle of the channel.

Morphological status is one of the more difficult variables to interpret because the degree of channel incision that is present in a stream may be dictated by the stream size, type, geology, and ecoregional characteristics (Harrelson and others, 1994). Incision may have occurred recently or gradually throughout many years or decades. In some instances, bank and riparian conditions are more protected from erosion, and the process is slowed. Morphological status might usually be scored based on an assumption that a steeper bank-slope angle is an indicator that channel incision is more active or recent. The percentage difference in elevation between opposing banks also is considered in the score rather than relying on the degree of bank slope itself. The difference, expressed as the mean percentage difference in slope between right and left banks for the reach, is an indicator of the potential for flood-plain interaction during flooding. Flood-plain interaction may provide more protection for aquatic organisms during floods and an increase in organic material inputs. The difference in elevation between opposing streambanks may indicate a greater likelihood for flood-plain interaction. Therefore, this variable is scored based on the assumption that when channel slopes are nearly the same on both sides of the stream, flood-plain interactions are less likely to occur, may require floods of larger magnitude, or may occur with less frequency.

The site score for percentage difference in elevation of opposing streambanks takes into account the slope values for right and left banks (in degrees), percentage difference in bank slope, and the predominant cross-sectional shape of the stream bottom. Conditions are considered optimal when bank elevations are near the elevation of the active flood plain, the channel cross section is V- or U-shaped, there is little evidence of lateral or downcutting, the mean bank slope is less than 15 percent, and the mean difference between right and left bank slopes is greater than 5 percent. Poor conditions exist when banks are much higher than the elevation of the active flood plain, the channel is trapezoid-shaped, mean bank slope is greater than 35 percent, and the mean difference between right and left bank slopes is less than 2 percent.

Sinuosity and Riffle Frequency

Sinuosity is a segment-scale measure that describes the meandering of the stream (Barbour and others, 1999) and is used to assess high- and low-gradient streams. It is the ratio of the channel length to the valley centerline length (Schumm, 1963) and can be obtained from aerial photographs and topographic maps. Streams that are more sinuous provide diverse habitat and fauna, and a stream is better able to handle flow surges when streamflow fluctuates as a result of runoff. The absorption of this energy by bends and repeated channel

cross-over and bend sequences protects the stream from excessive erosion and flooding and provides a refuge for benthic invertebrates and fish during periods of runoff. Conditions for sinuosity are considered optimal when the bends in the stream increase the stream length three to four times compared to a straight line. Conditions are poorest if the channel is straight as a result of channelization.

For high-gradient streams, in addition to sinuosity, riffle frequency is quantified. Riffle frequency (Barbour and others, 1999) is a measure of the number of riffles in the stream segment and is obtained from aerial photographs or topographic maps. Riffles are a source of high-quality habitat and a diverse fauna; therefore, an increased frequency of riffles greatly enhances the diversity of the stream community. Streams with more frequent, longer riffles tend to provide more available surface area of epifaunal substrate in comparison to streams dominated by long pools. In certain types of streams riffle occurrence may not be readily apparent because channel constrictions, exposed gravel bars, bluffs, or other channel features that may indicate riffle presence are not visible from maps or aerial photographs. Riffle frequency also is related to a decline in surface-water elevation, and this may provide an indication of riffle frequency for types of streams where other riffle/pool sequence indicators cannot be determined from maps and aerial photographs. Streams with infrequent riffles usually have less channel slope.

Riffle frequency is scored based on a combination of the number of riffles observed in the reach and the longitudinal decline in water elevation throughout the segment. Elevations are determined from a GPS unit. Conditions are considered optimal when elevation declines at least 26 feet per mile [ft/mi, 5 meters per kilometer (m/km)] and at least four riffles occur within the reach. Conditions are poor if the elevation decrease is less than about 5 ft/mi (1 m/km) and only one shallow riffle occurs within the reach.

Streambank and Riparian Characteristics

Bank and riparian characteristics provide information on stream energy sources, degree of disturbance in the riparian zone, and the potential for streambank erosion. Bank and riparian characteristics measured include bank stability; canopy cover; bank and riparian protection; length, extent, and width of buffers; and percentage of altered banks.

Bank Stability

Bank stability (Barbour and others, 1999) is a reach-scale measure of whether the streambanks are eroded or have the potential for erosion during periods of increased streamflow. It is a visual estimation of the percentage of the bank area that is stable (not eroding or sloughing) and includes vegetation, natural bedrock outcroppings, and the roots of woody vegetation that stabilize the bank soils or deflect high flows during periods of runoff. The right bank and left bank are evaluated separately. Steep unvegetated banks are generally more likely to collapse and suffer from erosion than are gently sloping banks. Signs of erosion include crumbling, unvegetated banks, exposed tree roots, and exposed soil. Eroded banks may indicate a problem of sediment movement and deposition, and also can indicate a scarcity of cover and organic particulate material input to streams.

Bank stability is determined by averaging a series of visual estimations made at 10 evenly spaced points in the stream throughout the reach. Each bank is evaluated separately and the mean (right and left banks) is calculated. Bank conditions are considered optimal when banks appear stable throughout the reach, less than 5 percent of the banks show evidence of erosion, and more than one-third of the erodible banks on outside bends is protected by roots or vegetation. Conditions are poor when 60 to 100 percent of banks have erosional scars.

Canopy Cover

Canopy cover (Natural Resources Conservation Service, 1998) is a measure of the percentage of the reach that is shaded by overhanging vegetation and other features in the stream channel. Stream shading is important because it decreases light availability and helps to keep water temperatures cool, which limits excessive algae and vegetation growth. However, fully shaded streams may limit primary production to the extent that it may affect the presence of grazing macroinvertebrates and limit the stream's ability to attenuate levels of excess nutrients. Canopy cover is scored based on the assumption that streams support healthier and more diverse aquatic biota when there is partial shade as compared to those exposed to full shade or full sunlight.

Canopy cover is visually estimated from the center of the stream at 10 evenly spaced points along the reach and then averaged. The relative amount of shade is estimated by assuming that the sun is directly overhead and the vegetation is in full leaf. Conditions are considered optimal when 50 to 80 percent of the reach is shaded and poor when less than 10 or more than 90 percent is shaded.

Bank and Riparian Protection

Bank and riparian protection (Barbour and others, 2009) is a measure of the percentage of the bank surface area within the reach that is covered with natural materials such as vegetation, rock, or bedrock outcroppings. Percentage of coverage is estimated visually for the left bank and right bank from 10 evenly spaced points and then averaged. Artificial materials such as riprap or concrete are not included in the estimate. This measure provides an indication of how well the streambank and the near stream part of the riparian zone resist erosion, uptake nutrients, and control in-stream scouring.

Length and Extent of Buffers

Length and extent of buffers (Rasmussen and others, 2009) provide an estimate of the extent of buffers and the number of gaps in longitudinal continuity. Buffers are defined as land covered with natural vegetation that could include forest, shrubs, or grasses. This variable takes into account the buffers within the reach and segment and is obtained from onsite observations and aerial photographs. The longitudinal continuity of buffers is related to the number of bridge crossings and stormwater drains entering the stream and the extent of areas cleared for construction and development. In areas where these activities are common, there are more frequent opportunities for stormwater to enter the stream directly without passing through vegetated soils. An increase in direct stormwater drainage connections also can affect the intensity and magnitude of flooding. Conditions are considered optimal when the mean longitudinal length of buffers that are at least 20 ft (6 m) wide is larger than 2,500 ft (750 m) and when greater than 90 percent of the stream segment length is buffered. Conditions are poor when the mean longitudinal length of buffers is less than 820 ft (250 m) and encompasses less than 70 percent along both banks.

Average Buffer Width

Average buffer width (Barbour and others, 1999) is a reach-scale measurement of the mean width of natural vegetation (including forest, shrubs, or grasses) from the edge of the streambank out through the riparian zone. The vegetative zone serves as a buffer to pollutants entering a stream from runoff, as a control of erosion, and as inputs of nutrients and organic material into the stream. A wider buffer allows runoff more time to percolate into soils or be filtered by vegetation before entering the stream. Wider, more vegetated, and less-disturbed riparian zones also produce more organic material that provides a constant supply of energy to the stream. Buffer width is estimated visually for the left and right banks separately at 10 evenly spaced points in the stream over the length of the reach. Conditions are considered optimal when the mean buffer width is larger than about 60 ft (18 m) on both banks. Conditions are poor when the mean buffer width is less than 20 ft (6 m).

Percentage of Altered Banks

The percentage of bank and above-bank riparian zones that have been physically altered (Rasmussen and others, 2009) can provide an indication of large-scale changes in the shape of the stream channel. Alterations along the banks may reduce organic material inputs or hydrologic diversity. Alterations include channelization, concrete, levees, dikes, piers, riprap, impoundments, bridges, and in-stream activities such as clearing, operation of heavy equipment, and bridge construction. Streams that have been straightened, deepened, or converted to concrete channels have fewer natural habitats for fish, macroinvertebrates, and plants than do naturally meandering streams. Some older modifications that have become overgrown with native vegetation may not score as poorly as recently altered areas. Percentage of altered banks is estimated at 10 points along each bank of the reach. Conditions are optimal when none of the alteration activities are occurring in the reach and past human activities affect less than 10 percent of the total bank and buffer area. Conditions are poor when more than three activities or features are present or more than 70 percent of the bank and buffer area is affected by human activities.

In-stream Habitat Characteristics

Habitat characteristics located within the stream provide information about in-stream cover and aquatic habitat that are directly available as living space for aquatic organisms. These features, all measured at the reach scale, relate to the ability of the stream to meet basic physical requirements for supporting diverse and well-balanced aquatic communities.

Substrate Fouling

Substrate fouling (Rasmussen and others, 2009) is an estimate of the amount of periphyton growth and accumulation of fine materials that are covering riffle substrate. It is visually estimated for the length of the reach by examining several locations where the bottom substrate is visible. Excessive amounts of periphyton growth trap fine particulates and can cause the clogging of interstitial spaces in substrates, often leading to greater substrate embeddedness and a decline in overall living space for macroinvertebrates and fishes. Substrate fouling also is related directly to larger sediment loads during rainfall, extent of bank erosion, and the turnover of periphyton growth, because these characteristics represent the direct sources for finer substrate particles that may be deposited. Conditions are optimal when visible periphyton and fine materials affect less than 10 percent of the substrate and little sloughing occurs when substrate is physically disturbed. Poor conditions exist when more than 60 percent of the substrate is covered with periphyton and fine materials and extensive cloudiness occurs when substrate is disturbed.

Velocity/Depth Combinations and Pool Variability

For high-gradient streams, velocity/depth combinations are quantified. Patterns of velocity and depth (Barbour and others, 2009) are related to habitat diversity. Streams with at least four patterns of velocity and depth (slow-shallow, slow-deep, fast-shallow, and fast-deep) generally have the most diversity. This is a reach-scale measurement that is visually estimated. Optimal conditions exist when all four combinations are present and poor conditions exist when only one is present.

For low-gradient streams, pool variability (Barbour and others, 1999) is assessed. Pool variability rates the overall mixture of pool types found in streams according to size and depth. The four basic types of pools are large-shallow, large-deep, small-shallow, and small-deep. A stream with many pool types will support a wide variety of aquatic species. Rivers with low sinuosity and monotonous pool characteristics do not have sufficient quantities and types of habitat to support a diverse aquatic community. General guidelines are any pool dimension (that is, length, width, oblique) greater than one-half the cross section of the stream for separating large from small and 1-m (1.1 yd) depth separating shallow and deep.

Riffle Substrate Embeddedness and Pool Substrate Composition

Riffle substrate embeddedness is assessed for high-gradient streams. Riffle substrate embeddedness (Barbour and others, 1999) is a measure of rock and snag substrates in riffles that are surrounded by or sunken into finer materials. Generally, as rocks become embedded, the surface area and living space available to macroinvertebrates and fish decrease. Riffle substrate embeddedness is evaluated by hand removal of 20 randomly chosen cobblestones across riffle transects within the reach, estimating the depth of the cobble in fine material as a percentage of total depth, and averaging the 20 values. Conditions are optimal when mean cobble depth in fine materials is less than 20 percent of total fine material depth and poor when cobble depth is more than 75 percent of total depth.

Low-gradient streams are assessed using pool substrate composition (Barbour and others, 1999). Pool substrate characterization evaluates the type and condition of bottom substrates found in pools. Firmer sediment types (for example, gravel, sand) and rooted aquatic plants support a wider variety of organisms than a pool substrate dominated by mud or bedrock and no plants. In addition, a stream that has a uniform substrate in its pools will support far fewer types of organisms than a stream that has a variety of substrate types.

Sediment Deposition

Sediment deposition (Barbour and others, 1999) provides an estimate of the amount of sediment that has accumulated in pools and other changes that have occurred to the stream bottom as a result of deposition. Sediment deposition may form islands and point bars and fill runs and pools. Usually deposition occurs in areas that are obstructed by natural or manmade debris and areas where the streamflow decreases, such as the inside part of meander bends or along the edges of small backwater inlets. Large amounts of sediment deposition may indicate a continually changing environment unsuitable for many organisms. Sediment deposition is visually estimated for the whole reach. Conditions are considered optimal when less than 5 percent of the stream bottom (less than 20 percent for low-gradient streams) is affected by deposition and little or no island or point-bar deposition is visible. Conditions are poor when thick sediment deposits are visible, more than 50 percent of the stream bottom (more than 80 percent for low-gradient streams) changes frequently, and fresh deposits occur along primary parts of the overbank areas.

Diversity of Epifaunal Substrate and Cover

Diversity of epifaunal substrate and cover (Barbour and others, 1999) is a measure of the number and variety of in-stream habitat and cover types. This includes natural structures in the stream such as leaf packs, anchored woody debris, root mats, overhanging or inundated vegetation, organic debris accumulation, undercut banks, submerged macrophyte beds, and isolated backwater. These features provide protection, feeding sites, sites available for colonization by grazers and clingers, emergence sites, and sites for spawning. For optimum conditions, these features are fairly stable. A wide variety and abundance of good habitat increase overall biotic diversity in the reach. As variety and abundance of habitat decrease, diversity decreases, and the potential for recovery following disturbance declines. Snags and submerged logs are among the most productive habitat structures for macroinvertebrate colonization, particularly if they have been submerged for a long period of time.

Diversity of epifaunal substrate and cover is visually estimated for the stream reach. Optimal conditions exist when at least seven of the habitat/cover types (leaf packs, anchored woody debris/logs/trees, root mats, overhanging and inundated vegetation, organic debris accumulation, undercut banks, submerged macrophyte beds, and isolated backwaters or inlets) are present and at least 70 percent are stable and available for aquatic colonization. Poor conditions exist when one or none of the cover types are present and less than 20 percent are stable or available for colonization.

Summary

The city of Wichita, Kansas uses the *Equus* Beds aquifer for municipal water supply and plans for artificial recharge of the aquifer have been implemented in several phases to meet future water needs. Phase I of the *Equus* Beds Aquifer Storage and Recovery (ASR) Program began with injection of water from the Little Arkansas River into the aquifer for storage and subsequent recovery in 2006. Construction of a river intake structure and surface-water treatment plant began as implementation of Phase II of the *Equus* Beds ASR Program in 2010. A substantial part of the ASR Program is the monitoring of water quality and the effects that recharge activities may have on stream conditions. An integrated assessment of stream quality is provided by physical, chemical, and biological data for the city of Wichita's Hydrobiological Monitoring Program (HBMP). Following established and reliable methods for stream data collection is crucial for long-term success of the HBMP.

Protocols for measuring streamflow (discharge), operating continuous water-quality monitors, collecting water-quality samples, collecting streambed-sediment samples, sampling periphyton, macroinvertebrates, and fish, and assessing stream habitat are described and consolidated into one document. Protocols were collected from established and approved scientific and data collection procedures and standard methods for planning and executing studies and laboratory analyses. These protocols were developed to be used for the city of Wichita, Kansas' HBMP for the *Equus* Beds ASR Program, but can easily serve as a framework for, or be adapted to, other stream studies.

Protocols for continuous streamflow and water-quality data are described. Streamflow protocols characterize methods for establishing and maintaining streamgaging stations and taking accurate streamflow measurements. Streamflow is measured for all flow conditions. Continuous streamflow is computed from the gage height record by application of a stage-streamflow relation. Streamflow and stage measurement data processing steps are specified. Methods for continuous water-quality monitor installation and maintenance are detailed, including protocols for operating temperature and specific conductance, dissolved oxygen, pH, colored dissolved organic matter, chlorophyll, turbidity, Solitax, and nitrate sensors.

Field and laboratory protocols for water and streambed-sediment sampling are described in detail. Procedures for obtaining water and streambed-sediment samples that are representative of the stream system being studied are presented. Water-quality constituents are sampled for all flow conditions; to avoid scouring from high flows, sediment samples are collected during low-flow periods. Sample processing methods for water and streambed sediment are explained, with emphasis on sample contamination avoidance by using clean hands/dirty hands techniques. Laboratory water sample processing techniques are described for inorganic and organic constituents. Additionally, water and streambed-sediment sampling equipment cleaning procedures are detailed.

Protocols for biological samples that are representative of the stream system being studied are described. Field methods for periphyton, macroinvertebrate, and fish collection are detailed. To avoid scouring from higher flows, periphyton, macroinvertebrate, and fish samples are collected during low-flow periods. There are many advantages of using benthic periphytic, macroinvertebrate, and fish communities for evaluating stream quality. Appropriate periphyton, macroinvertebrate, and fish sampling techniques and equipment are explained. Details on sampling all available periphyton, macroinvertebrate, and fish habitats are provided. Periphyton collection for chlorophyll and taxonomic identification in two substrate types are described. Macroinvertebrate collection using a qualitative method is described. Two complementary fish sampling methods are documented as well as fish processing techniques.

Habitat assessment protocols describe methods for quantifying 14 habitat characteristics. These characteristics are classified into channel, streambank and riparian, and in-stream habitat categories. Habitat characteristics are evaluated on either a stream-segment or a stream-reach scale. The habitat assessment characteristics can be used for either high-gradient or low-gradient streams.

References Cited

Allan, J.D., 1995, Stream ecology—Structure and function of running waters: Boston, Mass., Kluwer Academic Publishers, 388 p.

Aquatic Informatics, 2008, GRSAT R2.0 help manual: Vancouver, Canada, Aquatic Informatics.

Arar, E.J., and Collins, G.B., 1997, U.S. Environmental Protection Agency Method 445.0, In vitro determination of chlorophyll-*a* and pheophytin-*a* in marine and freshwater algae by fluorescence, Revision 1.2: Office of Research and Development, 22 p.

ASTM International, 2007, D4410, Terminology for fluvial sediment, *in* ASTM International, Annual book of standards, water and environmental technology: West Conshohocken, Pa., American Society for Testing and Materials International, 7 p.

Barbour, M.T., Gerritsen, J., Snyder, B.D., and Stribling, J.B., 1999, Rapid bioassessment protocols for use in streams and wadeable rivers—periphyton, benthic macroinvertebrates, and fish (2d ed.): U.S. Environmental Protection Agency Report, EPA 841/B-99/002, 18 p.

Bouchard, R.W., Jr., and Anderson, J.A., 2001, Description and protocol for two quantitative periphyton samplers used for multihabitat stream sampling: Report for the Central Plains Center for Bioassessment, 12 p.

Burns and McDonnell Engineering Co., Inc., 2003, Final environmental impact statement for the Integrated Local Water Supply Plan for the city of Wichita.

Burns and McDonnell Engineering Co., Inc., 2010, Hydro-Biological Monitoring Plan Year Five Report.

Cairns, J., Jr., and Pratt, J.R., 1993, A history of biological monitoring using benthic macroinvertebrates, *in* Rosenberg, D.M., and Resh, V.H., eds., Freshwater biomonitoring and benthic macroinvertebrates: New York, N.Y., Chapman and Hall, p. 10–27.

City of Wichita, 1993, Integrated Local Water Supply Plan: Wichita, Kans.

Fitzpatrick, F.A., Waite, I.R., D'Arconte, P.J., Meador, M.R., Maputin, M.A., and Gurta, M.E., 1998, Revised methods for characterizing stream habitat in the National Water Quality Assessment Program: U.S. Geological Survey Water-Resources Investigations Report 09–4052, 67 p.

Frissell, C.A., Liss, W.J., Warren, C.E., and Hurley, M.D., 1986, A hierarchical framework for stream habitat classification—viewing streams in a watershed context: Environmental Management, v. 10, p. 199–214.

Graham, J.L., Stone, M.L., Rasmussen, T.J., and Poulton, B.C., 2010, Effects of wastewater effluent discharge and treatment facility upgrades on environmental and biological conditions of the upper Blue River, Johnson County, Kansas and Jackson County, Missouri, January 2003 through March 2009: U.S. Geological Survey Scientific Investigations Report 2010–5248, 85 p.

Hambrook-Berkman, J.A., and Canova, M.G., 2007, Algal biomass indicators: U.S. Geological Survey Techniques of Water-Resources Investigations, book 9, chap. A7, sec. 7.4, accessed September, 2011, at *http://pubs.water.usgs.gov/twri9A7*.

Hansen, C.V., and Aucott, W.R., 2010, Status of groundwater levels and storage volume in the *Equus* Beds aquifer near Wichita, Kansas, January 2006 to January 2010: U.S. Geological Survey Scientific Investigations Report 2010–5231, 47 p.

Harrelson, C.C., Rawlins, C.L., and Potyondy, J.P., 1994, Stream channel reference sites—an illustrated guide to field technique: Fort Collins, Colorado, U.S. Department of Agriculture, U.S. Forest Service, Rocky Mountain Forest and Range Experiment Station, General Technical Report RM-245, 61 p.

Hem, J.D., 1992, Study and interpretation of chemical characteristics of natural water (3d ed.): U.S. Geological Survey Water-Supply Paper 2254, 263 p.

Hillebrand, H., Duerselen, C.D., Kirschtel, D.B., Pollingher, U., and Zohary, T., 1999, Biovolume calculation for pelagic and benthic microalgae: Journal of Phycology, v. 35, p. 403–424.

Horowitz, A.J., 1991, A primer on trace-metal sediment chemistry (2d ed.): Ann Arbor, Mich., Lewis Publishing Company, 136 p.

Kansas Department of Health and Environment, 2000, Division of Environment, quality management plan, part III—Stream biological monitoring program, quality assurance management plan: Topeka, Kans., Bureau of Environmental Field Services, Technical Services section, 42 p.

Karr, J.R., 1981, Assessment of biotic integrity using fish communities: Fisheries, v. 6, p. 91–107.

Karr, J.R., Fausch, K.D., Angermeier, P.L., Yant, P.R., and Schlosser, L.J., 1986, Assessing biological integrity in running waters—a method and its rationale: Illinois Natural History Survey, Special publication 5.

Kennedy, E.J., 1983, Computation of continuous records of streamflow: U.S. Geological Survey Techniques of Water-Resources Investigations, book 3, chap. A13, 53 p.

Kennedy, E.J., 1984, Discharge ratings at gaging stations: U.S. Geological Survey Techniques of Water-Resources Investigations, book 3, chap. A10, 59 p.

Kenney, T.A., 2010, Levels at gaging stations: U.S. Geological Survey Techniques and Methods 3–A19, 60 p.

Knowlton, M.F., 1984, Flow-through microcuvette for fluorometric determination of chlorophyll: Water Resources Bulletin, v. 20, p. 1,198–1,205.

Lowe, R.L, and Pan, Y., 1996, Benthic algal communities as biological monitors, *in* Stevenson, R.J., Bothwell, M.L., and Lowe, R.L., eds., Algal ecology—freshwater benthic ecosystems: San Diego, Calif., Academic Press, 753 p.

McNabb, C.D., 1960, Enumeration of freshwater phytoplankton concentrated on the membrane filter: Limnology and Oceanography, v. 5, no. 1, p. 57–61.

Meador, M.R., Cuffney, T.F., and Gurtz, M.E., 1993, Methods for sampling fish communities as a part of the National Water-Quality Assessment Program: U.S. Geological Survey Open-File Report 93–104, 40 p.

Merritt, R.W., Cummins, K.W., and Berg, M.B., 2008, Aquatic insects of North America (4th ed.): Kendall/Hunt Publishing, 1,158 p.

Moulton, S.R., II, Kennen, J.G., Goldstein, R.M., and Hambrook, J.A., 2002, Revised protocols for sampling algal, invertebrate, and fish communities as part of the National Water-Quality Assessment Program: U.S. Geological Survey Open-File Report 02–150, 75 p.

Mueller, D.S., and Wagner, C.R., 2009, Measuring discharge with acoustic Doppler current profilers from a moving boat: U.S. Geological Survey Techniques and Methods 3–A22, 72 p. (Also available at *http://pubs.usgs.gov/sir/2006/5079*.)

Natural Resources Conservation Service, 1998, Stream visual assessment protocol: U.S. Department of Agriculture, National Water and Climate Center Technical Note 99–1, 36 p.

Patrick Center for Environmental Research, 1988, Diatom cleaning by nitric acid digestion: The Academy of Natural Sciences of Philadelphia, Protocol No. P–13–02, 39 p.

Poulton, B.C., Rasmussen, T.J., and Lee, C.J., 2007, Assessment of biological conditions at selected stream sites in Johnson County, Kansas and Cass and Jackson Counties, Missouri, 2003 and 2004: U.S. Geological Survey Scientific Investigations Report 2007–5108, 68 p.

Prewitt, B., Toth, A., King, A., Grimsley, K., and Rasmussen, P., 2011, USGS CHIMP User Manual, 54 p., accessed February, 2012 at *http://wwwrustla.er.usgs.gov/FCIS/CHIMP/documentation/CHIMP_4.10_user_manual.pdf*.

Radtke, D.B., Davis, J.V., and Wilde, F.D., 2005, Specific electrical conductance: U.S. Geological Survey Techniques of Water-Resources Investigations, book 9, chap. A6, section 6.3, accessed August 2011, at *http://water.usgs.gov/owq/FieldManual/Chapter6/Final508Chapter6.3.pdf*.

Rantz, S.E., and others, 1982, Measurements and computation of streamflow, v. 1 and 2: U.S. Geological Survey Water-Supply Paper 2175, 631 p.

Rasmussen, T.J., Poulton, B.C., and Graham, J.L., 2009, Quality of streams in Johnson County, Kansas, and relations to environmental variables, 2003–07: U.S. Geological Survey Scientific Investigations Report 2009–5235, 84 p. with appendixes.

Sanders, R.E., Miltner, R.J., Yoder, C.O., and Rankin, E.T., 1999, The use of external deformities, erosion, lesions, and tumors (DELT anomalies) in fish assemblages for characterizing aquatic resources—A case study of seven Ohio streams, *in* Simon, T.P., Assessing the sustainability and biological integrity of water resources using fish communities: New York, CRC Press, 671 p.

Sartory, D.P., and Grobbelar, J.U., 1986, Extraction of chlorophyll-*a* from freshwater phytoplankton for spectrophotometric analysis: Hydrobiologia, v. 114, p. 117–187.

Sauer, B.B., and Turnipseed, D.P., 2010, Stage measurement at gaging stations: U.S. Geological Survey Techniques and Methods, book 3, chap. A7, 45 p.

Schumm, S.A., 1963, Sinuosity of alluvial rivers on the Great Plains: Geological Society of America Bulletin, v. 74, p. 1,089–1,100.

Shelton, L.R., 1994, Field guide for collecting and processing stream-water samples for the national water-quality assessment program: U.S. Geological Survey Open-File Report 94–455, 42 p.

Shelton, L.R., and Capel, P.D., 1994, Guidelines for collecting and processing samples of stream bed sediment for analysis of trace elements and organic contaminants for the national water-quality assessment program: U.S. Geological Survey Open-File Report 94–458, 20 p.

Stevenson, R.J., and Bahls, L.L., 1999, Periphyton protocols, *in* Barbour, M.T., Gerritson, J., Snyder, B.D., and Stribling, J.B., eds., Rapid bioassessment protocols for use in streams and wadeable rivers—Periphyton, benthic macroinvertebrates, and fish (2d ed.): Washington, D.C., U.S. Environmental Protection Agency, Office of Water, 841–B–99–002, p. 6/1–6/23.

Turnipseed, D.P., and Sauer, V.B., 2010, Discharge measurements at gaging stations: U.S. Geological Survey Techniques and Methods, book 3, chap. A8, 87 p.

U.S. Environmental Protection Agency, 2009, National primary drinking water regulations: U.S. Environmental Protection Agency Report, EPA–816–F–09–004, accessed March, 2011 at *http://www.epa.gov/ogwdw/consumer/pdf/mcl.pdf*.

U.S. Geological Survey, variously dated, National field manual for the collection of water-quality data: U.S. Geological Survey Techniques of Water-Resources Investigations, book 9, chaps. A1–A9, (Also available at *http://pubs.water.usgs.gov/twri9A*).

Vidon, P., Wagner, L.E., and Soyeux, E., 2008, Changes in the character of DOC in streams during storms in two Midwestern watersheds with contrasting land uses: Biogeochemistry, v. 88, p. 257–270.

Wagner, R.J., Boulger, R.W., Jr., Oblinger, C.J., and Smith, B.A., 2006, Guidelines and standard procedures for continuous water-quality monitors—Station operation, record computation, and data reporting: U.S. Geological Survey Techniques and Methods 1–D3, 96 p.

Walsh, S.J., and Meador, M.R., 1998, Guidelines for quality assurance and quality control of fish taxonomic data collected as part of the National Water-Quality Assessment Program: U.S. Geological Survey Water-Resources Investigations Report 98–4239, 33 p.

Wehr, J.D., and Sheath, R.G., eds., 2003, Freshwater algae of North America—Ecology and classification: San Diego, Calif., Academic Press, 918 p.

Welch, E.B., and Lindell, T., 1992, Ecological effects of wastewater (2d ed.): London, Spon Press, 425 p.

Wentworth, C.K., 1922, A scale and grade and class terms for clastic sediments: Journal of Geology, v. 30, p. 377–392.

Whittemore, D.O., 2007, Fate and identification of oil-brine contamination in different hydrogeologic settings: Applied Geochemistry, v. 22, no. 10, p. 2,009–2,114.

Wilde, F.D., 2006, Temperature: U.S. Geological Survey Techniques of Water-Resources Investigations, book 9, chap. A6, section 6.1, accessed August, 2011 at *http://water.usgs.gov/owq/FieldManual/Chapter6/6.1_ver2.pdf*.

Wiley, M.L., and Tsai, Chu-Fa, 1983, The relative efficiencies of electrofishing vs. seines in Piedmont streams of Maryland: North American Journal of Fisheries Management, v. 3, p. 243–253.

Wilson, H.F., and Xenopolous, M.A., 2009, Effects of agricultural land use on the composition of fluvial dissolved organic carbon: Nature Geoscience, v. 2, p. 37–41.

YSI Environmental, 2005, Alternate calibration methods for the YSI 6025 chlorophyll sensor: YSI Environmental Tech Note 0401 T612, 2 p.

Ziegler, A.C., Hansen, C.V., and Finn, D.A., 2010, Water quality in the *Equus* Beds aquifer and the Little Arkansas River before implementation of large-scale artificial recharge, south-central Kansas, 1995–2005: U.S. Geological Survey Scientific Investigations Report 2010–5023, 143 p.

Appendix 1

Site Specific Job Hazard Analyses
Surface Water Discharge, Groundwater Elevations, and Water-Quality Site

Station Name: Little Arkansas River Upstream of ASR FAC **Station Number:** 375350097262800

Maximum Wading Stage: **Undetermined**.

Most Used Cross-Section: If needed, wading can be done 50 ft upstream of ASR building. PFD's are required at all stages or when near gravel or rip-rap bank.

Streambed Characteristics Soft shifting sand.

QW activity: QW sampling and field monitor measurements are done from the northeast corner of the ASR building near the river.

Potential Unsafe Condition

The following is a list of Potential Hazards relative to this specific site:

Construction: Personnel shall wear high visibility vests, hard hats, and safety glasses at all times while construction continues at the ASR Facility.

QW Processing: Keep electrical connections from getting wet. Wear gloves and eye protection as required for sampling and using acids/bases for treatment of samples.

Wading across stream: The bottom of the channel across from the ASR building has been dug out and is extremely deep. If needed, personnel should wade the channel upstream of the building where construction has not taken place.

Scour of the channel: Test the bottom with the wading rod as sand can get quite soft. Proceed across slowly. Use the same path once a good section is established.

Insects: Not expected to be an issue at this site unless field personnel walk through the trees.

Gage Cleaning: Follow NE Hantavirus gage cleaning protocols for personal protection associated with rodents.

If rescue or self rescue is not realistic, wading the channel should not be attempted.

Sheriff Sedgwick County
141 West Elm St.
Wichita, KS 67203
316-383-7264

Wesley West Emergency Center
8714 West 13th St. N.
Wichita, KS 67214
316-962-9900

Hospital Directions: Travel south on S Emma Creek Road to 109th St. N, then 1 mile east to Ridge Road. From intersection of 109th St. N and Ridge Road, travel south to 13th St. in Wichita. Go west on 13th St. to Tyler St., then north on Tyler about ½ block to the emergency entrance of Wesley Medical Center. Map attached to printed JHA.

Prepared By: T.J. Bennett Reviewed By:
Date: March 21, 2011 Date:

Appendix 2

THINK TWICE. WORK SAFE.

USGS science for a changing world

U. S. GEOLOGICAL SURVEY SURFACE-WATER QUALITY NOTES

NWIS RECORD NO ___ ___ ___ ___ ___ ___ ___ ___ / ___ ___ ___ ___ ___ ___ ___ ___

STATION NO. _____ SAMPLE DATE _____/_____/_____ SAMPLE TIME _____

STATION NAME _____ PROJECT NO. __ **GC11SE00 19440 00** __ SAMPLE PURPOSE (71999) _ 10 _

PROJECT NAME ____ **HBMP Project** ____ Med. Code _____ Sample Type _____ Status_ U _ Hydro. Cond. _____ Event _____ Source _ 5 _

SAMPLED BY _____ Samples Received by_____ DATE _____/_____/_____

FIELD MEASUREMENTS

GAGE HT (00065) _____ ft | COND (00095) _____ µS/cm@25 °C | F. COLIFORM (31625) _____ col/100mL

Q, INST. (00061) _____ cfs MEAS. RATING EST. | TEMP, AIR (00020) _____ °C | FC: Rmk _____ FC: Qual _____

DIS. OXYGEN (00300) _____mg/L | TEMP, WATER (00010) _____ °C | E. COLI (90902) _____ col/100mL

DIS. OXYGEN (00301) _____ % Sat | TURBIDITY (63680) _____ FNU TS087, YSI 6136 | EC: Rmk _____ EC: Qual _____

BAROMETRIC PRES. (00025) _____ mm Hg | CHLOROPHYLL (62361) _____ µg/L | OTHER ( ) _____

pH (00400) _____ UNITS | OTHER ( ) _____

SAMPLING INFORMATION

Sampler Type (84164): _____ Sampler ID: _____ Sample Compositor/Splitter: PLASTIC TEFLON CHURN CONE OTHER _____

Sampler Bottle/Bag Material: PLASTIC TEFLON OTHER _____ Nozzle Material: PLASTIC TEFLON OTHER _____ ___ Nozzle Size: 3/16" 1/4" 5/16"

Stream Width: _____ ft mi Left Bank: _____ Right Bank: _____ Mean Depth: _____ ft Ice Cover _____% Ave. Ice Thickness _____ in.

Sampling Points: _____ _____ _____ _____ _____ _____ _____ _____ _____ _____ _____ _____ _____ _____ _____ _____ _____

Sampling Location: WADING CABLEWAY BOAT BRIDGE UPSTREAM DOWNSTREAM SIDE OF BRIDGE _____ ft mi above below gage _____

Sampling Site: POOL RIFFLE OPEN CHANNEL BRAIDED BACKWATER Bottom: BEDROCK ROCK COBBLE GRAVEL SAND SILT CONCRETE OTHER _____

Stream Color: BROWN GREEN BLUE GRAY CLEAR OTHER _____ Stream Mixing: WELL-MIXED STRATIFIED POORLY-MIXED UNKNOWN OTHER _____

Weather: SKY- CLEAR CLOUDY _____% PRECIP- LIGHT MEDIUM HEAVY SNOW RAIN MIST WIND: CALM LIGHT BREEZE GUSTY WINDY EST: WIND SPEED_____

Sampling Method (82398): EWI [10] EDI [20] SINGLE VERTICAL [30] MULT VERTICAL [40] OTHER _____ STAGE: RISING FALLING PEAK STABLE- NORMAL HIGH LOW

LABORATORY INFORMATION

LAB	Schedule/Lab	NO. OF	BOTTLE SIZE	BOTTLE TYPE	BOTTLE CODE		LAB	SCHEDULE/	NO. OF	BOTTLE	BOTTLE TYPE	BOTTLE CODE
NWQL	SC 2003	1	1 Liter	Amber glass	GCC		Wichita	Table1	2	250 ml	Autoclave	BACTI
	SC 1608	1	1 Liter	Amber glass	GCC				1	125 ml	Amber glass	TOC
	SC 1380	3	40 ml	Amber glass	VOC				1	250 ml	AR poly	RA
	LC 1984/1986	1	125 ml	Plain poly	WCA				2	500 ml	Plain poly	RU
	LC 2187	1	500 ml	Plain poly	TBY				1	250 ml	AR poly	FA
	LC 3211	1	125 ml	Amber glass	TOC				1	250 ml	AR glass	FAM
	LC 2613	1	125 ml	Amber glass	DOC				1	125 ml	Plain poly	FCC
	LC 3142	1	8 ml	Brown poly	SAS				2	250 ml	Plain poly	FCA
	LC 20	1	125 ml	Plain poly	RCB				1	500 ml	Plain poly	FU
	LC 2614/2615	1	125 ml	Amber glass	UAS				1	500 ml	Plain poly	FCN
	NWQL Rep	8	Assorted	Assorted	SEE PAGE 4				1	125 ml	Amber glass	DOC
Lawrence	IMA	3	125 ml	Amber glass	IMA							
Iowa	Sediment	1	1 or 3 Liter	Plain poly	SED		WFO	FC EC	1	1 Liter	Autoclave	BACTI
Ohio	Coliphage	1	1 liter	Autoclave	BACTI			BARTS	3	25 ml	IRB SRB SLYM	

COMPILED BY: _____ **CHECKED BY:** _____ **DATE:** _____

Version 3:rev.03/12/2012 2:08 PM

STN NO_____

CROSS SECTION NOTES

MONITOR # _____

STATION	ft FROM LEFT BANK (00009) OR ft FROM RIGHT BANK (72103)	TIME	DEPTH ft (81903)	LOGIN NUMBER	pH units (00400)	TEMP °C (00010)	SC µS/cm (00095)	DO mg/L (00300) LUMIN YSI 6150	DO % Sat (00301) LUMIN YSI 6150	TURBIDITY FNU (63680) YSI 6136	CHLORO-PHYLL µg/L (62361)

QUALITY-CONTROL INFORMATION

LOT NUMBERS

PRESERVATIVES 7.5N HNO_3 (FOR METALS & HG) _____ 4.5N 1:7 H_2SO_4 (FOR WCA, TOC, DOC) _____

1:1 H_2SO_4 (FOR AMMONIA, PHOSPHORUS) _____ NaOH (FOR CYANIDE) _____

1:1 HCL (For VOC vials) _____

COMMENTS _____

_____

BLANK WATER INORGANIC (99200)_____ ORGAINC (99202)_____

FILTERS _____ CAPSULE (99206) BRAND _____ LOT NUMBER _____ SIZE_____

_____ PLATE BRAND _____ LOT NUMBER _____ SIZE_____

STN NO_____

MICROBIOLOGY

FECAL COLIFORM

Date collected: _____ Time collected: _____

Time in: _____ Date: _____/_____/_____

Time out: _____ Date: _____/_____/_____

VOLUME mL	COUNT COL/100mL	USED IN CALC?	REMARKS*
BLANK			
BLANK			

INCUBATE 22–26 hrs @44.5ºC FILTER SIZE=0.65 µM

IDEAL COUNT= 20-60 COL/100 mL PO_4-$MgCl_2$ BUFFER

RESULT (31625) _____ COL/100 mL FILTER BRAND_____

*REMARK ___ ___ **QUALIFIER ___ ___ FILTER LOT #_____

*REMARKS E=ESTIMATED
<=LESS THAN >=GREATER THAN
**SEE BACK PAGE

E. COLI

Date collected: _____ Time collected: _____

Time in at 35ºC / 44.5ºC: _____/_____ Date: _____/_____/_____

Time out: _____ Date: _____/_____/_____

VOLUME mL	COUNT COL/100mL	USED IN CALC?	REMARKS*
BLANK			
BLANK			

INCUBATE 2 HRS @ 35.0ºC, THEN 22-24 HRS @ 44.5ºC FILTER SIZE=0.45 µM

IDEAL COUNT= 20-80 COL/100 mL PO_4-$MgCl_2$ BUFFER

RESULT (90902) _____ COL/100 mL FILTER BRAND_____

*REMARK ___ ___ **QUALIFIER ___ ___ FILTER LOT #_____

*REMARKS E=ESTIMATED
<=LESS THAN >=GREATER THAN
**SEE BACK PAGE

QA INFORMATION FOR BARTS

TYPE	LOT #
IRB	_____
SRB	_____
SLYM	_____

QA INFORMATION FOR BACTERIA PLATES

TYPE	BRAND	LOT #
FECAL COLIFORM	Hach	_____
E. COLI	Hach	_____

SAMPLING COMMENTS: _____

_____

_____

_____

_____

_____

_____

_____

_____

Sample Medium Codes
WG Ground water
WS Surface water
WGQ Quality-control sample (GW)
WSQ Quality-control sample (SW)
OAQ Blank

Sample Type Code
9 Regular
7 Replicate
6 Reference
2 Blank
1 Spike

Hydrologic Event Code
B Under ice cover
J Storm
K Backwater
7 Flood
9 Routine

Hydrologic Conditions Codes
A Not Determined
4 Stable, low stage
5 Falling stage
6 Stable, high stage
7 Peak stage
8 Rising stage
9 Stable, normal stage

Analysis Source Codes
1 USGS lab and non-USGS field
2 USGS lab and non-USGS lab
3 USGS lab and non-USGS lab and field
4 USGS lab and field and non-USGS field
5 USGS lab and field and non-USGS lab
6 USGS lab and field and non-USGS lab and field
8 USGS lab
9 USGS lab and field
• USGS field and non-USGS lab

Value qualifier codes
d Diluted sample: hi range exceeded
g Count < 0.5 percent
j Count >= 15 percent (dominant)
K Counts outside acceptable range
Q insufficient sample received
& Biological organism est as dominant

Remark Codes
> Greater than reported value
< Less than reported value
E Estimated

Null-value Qualifiers
e required equipment not functional or available
f sample discarded; improper filter used
o insufficient amount of water

82398 SAMPLING METHOD
10 Equal Width Increment (EWI)
20 Equal Discharge Increment (EDI)
30 Single Vertical
40 Multiple Verticals
50 Point Sample
70 Grab Sample (Dip)
4020 Open-top bailer
4025 Double-valve bailer
4030 Suction
8010 Other

84164 SAMPLER TYPE
125 Kemmerer Bottle
3044 US DH-81
3045 US DH-81 With Teflon Cap And Nozzle
3053 US D-95 Teflon Bottle
3054 US D-95 Plastic Bottle
3055 US D-96 Bag Sampler
3060 Weighted-Bottle Sampler
3070 Grab Sample
3080 VOC Hand Sampler
4020 Open-top bailer

ADDITIONAL SAMPLING COMMENTS: _____

NWQL Replicates	Bottle Code	Bottle Type
LC 3211	TOC	1 - 125 ml Glass Amber
SC 535	FCC	1 - 125 ml Brown Poly
SC 535	WCA	1 - 125 ml Plain Poly
SC 13	FU	1 - 250 ml Plain Poly
SC 1050, LC 3122, LC 3132	FA, RU	1 - 250 ml AR Poly, 1 - 250 ml Plain Poly
LC 2187	TBY	1 - 500 ml Plain Poly
LC 2613	DOC	1 - 125 ml Glass Amber

A COMPLETE SET OF FIXED-VALUE CODES CAN BE FOUND ON-LINE AT: http://wwwnwis.er.usgs.gov/conversion/nwisdocs4_1/qw/QW.user.book.html

STN NO_____

ALKALINITY/ANC CALCULATIONS

BEGINNING H_2O TEMP. _____ **°C**

pH	ΔpH	VOL ACID DC	pH	ΔpH	VOL ACID DC

BEGINNING H_2O TEMP. _____ **°C**

pH	ΔpH	VOL ACID DC	pH	ΔpH	VOL ACID DC

CALCULATIONS

ALKALINITY (mg/L AS $CaCO_3$) = B x F_1 / V_s x CF

BICARBONATE (mg/L) = [B-2A] x F_2 / V_s x CF

CARBONATE (meq/L) = A x F_3 / V_s x CF

where:

A= DC or mLs acid from initial pH to endpoint near 8.3= _____

B= DC or mLs acid from initial pH to endpoint near 4.5 = _____

V_s = volume of sample, in milliliters

CF=correction factor (obtain from OWQRL for Hach acid cartridges of certain lot numbers — default value is 1.00)

Factor	0.16 Normal	1.60 Normal
F1	10.0	100
F2	12.2	122
F3	12.0	120

NOTE: For samples with pH > 9.2, these equations for bicarbonate and carbonate will fail to give accurate results. Use the Alkalinity Calculator at http://oregon.usgs.gov/alk. Attach output copy if alkalinity values are calculated using

HACH CARTRIDGE CORRECTION FACTOR _____

[SEE OWQRL WEB PAGE (NEWS) FOR INFO]
http://owqrl.er.usgs.gov/owqrlnews.shtml

COMMENTS/CALCULATIONS:

FIRST TITRATION RESULTS

DATE ___ ___/___ ___/___ ___ ___ ___

TIME ___ ___ ___ ___ TEMP _____

ALKALINITY/ANC _____mg/L AS $CaCO_3$

BICARBONATE _____mg/L AS HCO_3^-

CARBONATE _____mg/L AS CO_3^{2-}

ACID: 1.6N 0.16N 0.01639N

OTHER:_____

ACID LOT NO. _____

ACID EXPIRATION DATE __ __/__ __/__ __ __ __

SAMPLE VOLUME: _____mL

___ FILTERED ___ UNFILTERED
___ INCRIMENTAL EQUIVELANCE
___ INFLECTION POINT TITRATION
___ GRAN METHOD

STIRRING METHOD: ___ MAGNETIC ___ MANUAL

SECOND TITRATION RESULTS

DATE ___ ___/___ ___/___ ___ ___ ___

TIME ___ ___ ___ ___ TEMP _____

ALKALINITY/ANC _____ mg/L AS $CaCO_3$

BICARBONATE _____ mg/L AS HCO_3^-

CARBONATE _____ mg/L AS CO_3^{2-}

ACID: 1.6N 0.16N 0.01639N

OTHER:_____

ACID LOT NO. _____

ACID EXPIRATION DATE __ __/__ __/__ __ __ __

SAMPLE VOLUME: _____mL

___ FILTERED ___ UNFILTERED
___ INCRIMENTAL EQUIVELANCE
___ INFLECTION POINT TITRATION
___ GRAN METHOD

STIRRING METHOD: ___ MAGNETIC ___ MANUAL

STN NO _____

SPIKE VIALS (99104)_____ _____ _____ **SURROGATE VIALS** _____ _____ _____

BLANK WATER

INORGANIC (99200) _____ 2nd INORGANIC (99201) _____

PESTICIDE (99202) _____ 2nd PESTICIDE (99203) _____

VOC (99204) _____ 2nd VOC (99205) _____

DATE RECEIVED BLANK FROM NWQL(99109) _____ **DATE TRIP BLANK SHIPPED TO NWQL**(99110) _____

(Circle appropriate selections)

99100 Blank-solution type
- 10 Inorganic grade (distilled/deionized)
- 40 Pesticide grade (OK for DOC)
- 50 Volatile-organic grade (OK for pesticides and DOC)
- 200 Other

99101 Source of blank water
- 10 NWQL
- 80 Ocala Water Quality & Research Lab

99102 Blank-sample type
- 1 Solution
- 30 Trip
- 80 Equipment (done in non-field environment)
- 100 Field

99105 Replicate-sample type
- 20 Sequential

99106 Spike-sample type
- 10 Field

99107 Spike-solution source
- 10 NWQL

99108 Spike-solution volume, mL _____

99111 QC sample associated with this environmental sample
- 1 No associated QA data
- 10 Blank
- 30 Replicate sample
- 40 Spike sample
- 100 More than one type of QA sample
- 200 Other

NOTES _____

Appendix 3

U. S. GEOLOGICAL SURVEY STREAM HABITAT ASSESSMENT

USGS science for a changing world

STATION ID_____ STATION NO_____ DATE _____ TIME_____

STATION NAME _____

PROJECT NAME _____ PROJECT NO. _____

EVALUATED BY _____

UPSTREAM LAT/LONG _____ _____ ELEV (m) _____

DOWNSTREAM LAT/LONG _____ _____ ELEV (m) _____

WEATHER _____ clear _____ partly cloudy _____overcast _____ fog/haze _____drizzle _____intermittent rain _____ rain _____ snow

ESTIMATED RAINFALL IN LAST 5 DAYS _____ in PHOTOS TAKEN _____

GENERAL STREAM REACH INFORMATION

Channel dimensions:

Wetted channel width _____m Bed width _____ m

Bank full width _____ m Reach length _____ m

High water mark _____ m

Bank angle:

Right _____flat (<5°) _____ gradual (3-30°) _____ steep (30-75°)

_____ very steep (75-90°) _____ overhung (>90°)

Left _____ flat (<5°) _____ gradual (3-30°) _____ steep (30-75°)

_____ very steep (75-90°) _____ overhung (>90°)

Proportion (%) of reach that is riffle _____ pool _____

run_____ stagnant _____

Number of riffle/pool sequences _____

Length of riffles (range _____

Streambank composition (%) trees_____ grasses/weeds _____

bare ground _____ bedrock_____ rip rap _____ other (specify)_____

% channelized _____

Source of streamflow (check all that apply) runoff_____ spring _____

WWTF _____ culvert (describe)_____ other (specify)_____

Riparian land use (%) industrial_____ commercial_____ residential_____

pasture_____ row crop_____ woods_____ construction _____

other (specify)_____

Riparian cover (%) trees _____ grasses/weeds _____ bare ground _____

impervious surface _____ buildings _____ other (specify) _____

Water color and appearance _____ brown _____ green _____ gray _____ clear

_____ foam _____ livestock waste _____trash _____other

Odor _____normal _____sewage _____ petroleum _____ chemical

Bottom deposits (%) sewage sludge_____ lime sludge _____ trash _____

iron precipitate_____ other (specify) _____

Algae (%) stream bottom covered by algae_____

filamentous _____

Submerged macrophytes none_____ sparse _____ large areas (%) _____

Emergent macrophytes none_____ sparse _____ large areas (%) _____

Non-native species absent _____ sparse _____ isolated clumps _____

frequent (25-33%) _____ extensive (>33%)_____ Species_____

ADDITIONAL OBSERVATIONS

Category 1 — Channel Conditions and Characteristics

	Optimal	Suboptimal	Marginal	Poor
A. Flow Status (reach)	Water reaches base of both lower banks, and minimal amount of channel substrate is exposed. 12 11 10	Water fills >75% of the available channel; or <25% of channel is substrate exposed. 9 8 7	Water fills 25-75% of the available channel, and/or riffle substrates are mostly exposed. 6 5 4	Very little water in channel and mostly present as standing pools. 3 2 1
B. Channel Slope and Morphological Status (reach)	Average Percent Left Bank Right Bank Bank shape Banks are low at elevation of active flood plain (slope < 20%). Channel cross sectional shape is a V or U, and there is no evidence of lateral or downcutting. Mean slope of reach (both banks considered) is <15% and average % difference in slopes between right and left banks is > 5.0%. 12 11 10	1 2 3 4 Banks are a moderate height at elevation of active flood plain (slope 20—45%). Channel cross sectional shape is a U, and there is some evidence of lateral or downcutting. Mean slope of reach (both banks considered) is 15-24.9% and average % difference in slopes between right and left banks is 3.5-5.0%. 9 8 7	5 6 7 Banks are high at elevation of active flood plain (slope 45-60%). Channel cross sectional shape is a U or trapezoid with steeper sides, and there is some evidence of lateral or downcutting. Mean slope of reach (both banks considered) is 25-34.9% and average % difference in slopes between right and left banks is 2.0-3.49%. 6 5 4	8 9 10 Banks are high at elevation of active flood plain (slope >60%). Channel cross sectional shape is a trapezoid with steep sides, and there is considerable evidence of lateral or downcutting. Mean slope of reach (both banks considered) is > 35% and average % difference in slopes between right and left banks is < 2.0%. 3 2 1
C1. Sinousity and Riffle Frequency (high gradient, segment)	The bends in the stream increase the stream length 3 to 4 times longer than if it was in a straight line. (note– channel braiding is considered normal in coastal plains and other low-lying areas. This parameter is not easily rated in these areas. > 2.50	The bends in the stream increase the stream length 2 to 3 times longer than if it was in a straight line. 1.75 — 2.49	The bends in the stream increase the stream length 1 to 2 times longer than if it was in a straight line. 1.25 — 1.74	Channel straight; waterway has been channelized for a long distance. < 1.25
(choose either C1 or C2)	Occurrence of riffles relatively frequent; variety of habitat is key. In streams where riffles are continuous, placement of boulders or other large natural obstruction is important. > 5m drop per km and at least 4 riffles visible within the reach 12 11 10	Occurrence of riffles infrequent. 2.5-4.9 m drop per km and 3 riffles visible in reach 9 8 7	Occasional riffle or bend; bottom contours provide some habitat. 1.0-2.49 m drop per km and 2 riffles visible in reach 6 5 4	Generally all flat water or shallow riffles; poor habitat. < 1.0 m drop per km and 1 riffle visible in reach 3 2 1
C2. Sinuosity (low gradient, segment)	The bends in the stream increase the stream length 3 to 4 times longer than if it was in a straight line. (note– channel braiding is considered normal in coastal plains and other low-lying areas. This parameter is not easily rated in these areas. > 2.50 12 11 10	The bends in the stream increase the stream length 2 to 3 times longer than if it was in a straight line. 1.75 — 2.49 9 8 7	The bends in the stream increase the stream length 1 to 2 times longer than if it was in a straight line. 1.25 — 1.74 6 5 4	Channel straight; waterway has been channelized for a long distance. < 1.25 3 2 1

Category 2—Bank and Riparian Conditions

	Optimal	Suboptimal	Marginal	Poor
A. Bank Stability (reach)	Average 1 2 3 4 Left bank Right bank Banks are stable throughout reach. Evidence of erosion/sloughing or bank failure absent or minimal (<5% affected). 33% or more of the eroding surface area of banks on outside bends is protected by roots that extend to the base flow elevation. 12 11 10	5 6 7 Banks are moderately stable throughout reach. Infrequent, small areas of erosion/sloughing mostly healed over. 5-30% of bank in reach has erosion areas. Less than 33% of the eroding surface area of banks on outside bends is protected by roots that extend to the base flow elevation. 9 8 7	8 9 10 Banks are moderately unstable throughout reach. Evidence of erosion/sloughing or bank failure obvious; 30-60% of bank in reach has areas of erosion. High erosion potential during floods. 6 5 4	Banks are unstable, with many eroded (raw) areas frequent along straight sections and bends. 60-100% of banks have erosional scars. High erosion potential during floods. 3 2 1

Category 2—Bank and Riparian Conditions (cont.)

B. Canopy Cover (reach)

Average _____

1	2	3	4	5	6	7	8	9	10

50-80% shaded			30-50% shaded			80-90% or 10-30%			<10% shaded or >90% shaded		
12	11	10	9	8	7	6	5	4	3	2	1

C. Bank/ Riparian Protection (reach)

Average _____

	1	2	3	4	5	6	7	8	9	10
Left bank										
Right bank										
% native										

More than 90% of the streambank surfaces and immediate riparian zones covered by native vegetation, including trees, understory shrubs, or nonwoody macrophytes; vegetative disruption through grazing or mowing minimal or not evident; almost all plants allowed to grow naturally			70-90% of the streambank surfaces covered by native vegetation, but one class of plants is not well-represented; disruption evident but not affecting full plant growth potential to any great extent; more than one-half of the potential plant stubble height remaining.			50-70% of the streambank surfaces covered by native vegetation; disruption obvious; patches of bare soil or closely cropped vegetation common; less than one -half of the potential plant stubble height remaining.			Less than 50% of the streambank surfaces covered by native vegetation; disruption of streambank vegetation is very high; vegetation has been removed to 5 centimeters or less in average stubble height.		
12	11	10	9	8	7	6	5	4	3	2	1

D. Length and Extent of Buffers (segment/reach)

Average _____

	1	2	3	4	5	6	7	8	9	10
Left bank										
Right bank										

Mean longitudinal lengths of buffers that are at least 6m in width (both banks considered) is > 750m, with 90-100% of the stream segment length is buffered.			Mean longitudinal lengths of buffers that are 6m in width (both banks considered) is 500-749m, with 80-89.9% of the stream segment length buffered.			Mean longitudinal lengths of buffers that are 6m in width (both banks considered) is 250-499m, with 70-79.9% of the stream segment length buffered.			Mean longitudinal lengths of buffers that are 6m in width (both banks considered) is <250m, with less than 70% of the stream segment length buffered.		
12	11	10	9	8	7	6	5	4	3	2	1

E. Average Buffer Width (reach)

Average _____

	1	2	3	4	5	6	7	8	9	10
Left bank										
Right bank										

Average width of riparian zone >18 m along both banks, encompassing 90-100% of the reach.			Average width of riparian zone 12-18 m along both banks, 70-90% of reach buffered.			Average width of riparian zone 6-12 m along both banks, with 30-70% of reach buffered.			Average width of riparian <6 m along both banks, with 30% of the reach buffered.		
12	11	10	9	8	7	6	5	4	3	2	1

F. Percent (%) Altered Banks (reach)

1. Concrete as part of channel base or stream bank
2. Channelization or channel straightening
3. Presence of impoundments or dams
4. Presence of grade control structures
5. Presence of levees
6. Presence of in-stream activities: (such as bulldozing, heavy equipment), snag removal, bridge construction/ maintenance
7. Riparian clearing (active, adjacent to stream bank
8. Presence of dikes, artificial deflectors, or wiers
9. Bridge(s)

Average _____

	1	2	3	4	5	6	7	8	9	10
Left bank										
Right bank										

Stream normal with none of these activities occurring in the reach upstream or adjacent to the site. Evidence of past human activities in the reach affect less than 10% of the total bank and riparian area.			One of these activities or features are present upstream or adjacent to the site. Evidence of past and/ or present human activities in the reach affect 10-30% of the total bank and riparian area.			1-3 of these activities or features are present upstream or adjacent to the site. Evidence of past and/ or present human activities in the reach affect 40-70% of the total bank and riparian area.			More than 3 of these activities or features are present upstream or adjacent to the site. If 3 or less of these activities are present, then human activities (past and/or present) in the reach affect >70% of the total bank and riparian area.		
12	11	10	9	8	7	6	5	4	3	2	1

Category 3—Aquatic Habitat Availability

	Optimal	Suboptimal	Marginal	Poor
A. Substrate Fouling (reach)	Substrate fouling level 10% or less with visible periphyton growth at normal levels. When substrate is moved (slightly disturbed physically), very little turbidity or periphyton sloughing results. 12 11 10	Substrate fouling level 10-30% with visible peripyton growth at above normal levels. When substrate is moved (slightly disturbed physically), very little turbidity or periphyton sloughing results. 9 8 7	Substrate fouling 30-60% with visible peripyton growth at above normal levels. When substrate is moved (slightly disturbed physically), moderate turbidity, water cloudiness, and periphyton sloughing is observed. 6 5 4	Substrate fouling level >60% with visible peripyton growth covering a majority of the substrate. When substrate is moved (slightly disturbed physically), extensive turbidity, water cloudiness, and periphyton sloughing is observed. 3 2 1
B1. Velocity/Depth Combinations (high gradient, reach) **(choose B1 or B2)**	All 4 velocity/depth regimes present (slow-deep, slow-shallow, fast-deep, fast-shallow). (slow is <0.3 m/s, deep is >0.5 m). 12 11 10	Only 3 of the 4 regimes present (if fast-shallow is missing, score lower than if missing other regimes). 9 8 7	Only 2 of the 4 habitat regimes present (if fast-shallow or slow-shallow are missing, score low). 6 5 4	Dominated by 1 velocity/ depth regime (usually slow-deep). 3 2 1
B2. Pool Variability (low gradient, reach)	Even mix of large-shallow, large-deep, small-shallow, small-deep pools present. 12 11 10	Majority of pools large-deep; very few shallow. 9 8 7	Shallow pools much more prevalent than deep pools. 6 5 4	Majority of pools small-shallow or pools absent. 3 2 1
C1. Riffle substrate Embeddedness (high gradient, reach) **(choose C1 or C2)**	Average _____ Cobble Est % 1 2 3 4 5 6 7 8 9 10 Cobble Est % 11 12 13 14 15 16 17 18 19 20 Cobble and boulder particle are 0-25% (depth) covered with fine sediment. Some obvious layering of cobble observed in many area. 12 11 10	Cobble and boulder particles are 25-50% (depth) covered with fine sediment. Layering of cobble may be present, but rare. 9 8 7	Cobble and boulder particles are 50-75% (depth) covered with fine sediment. 6 5 4	Cobble and boulder particles are more than 75% (depth) covered with fine sediment. 3 2 1
C2. Pool Substrate Composition (low gradient, Reach)	Mixture of substrate materials, with gravel and firm sand prevalent; root mats and submerged vegetation. 12 11 10	Mixture of soft sand, mud, or clay; mud may be dominant; some root mats and submerged vegetation present. 9 8 7	All mud or clay or sand bottom; little or no root mat; submerged vegetation. 6 5 4	Hard-pan clay or bedrock; no root mat or submerged vegetation. 3 2 1
D. Sediment Deposition (reach)	Little or no enlargement of islands or point bars and less than 5% (<20% for low gradient streams) of the bottom affected by sediment deposition. Large sand/silt deposits in channel absent and no evidence of fresh sediment deposition on overbank. 12 11 10	Some new increase in bar formation, mostly from gravel; sand or fine sediment; 5-30% (20-50% for low-gradient) of the bottom affected; slight deposition in pools. Large sand/silt deposits in channel uncommon, with small localized areas of fresh sand/ silt deposits along top of low banks. 9 8 7	Moderate deposition of new gravel. Sand or fine sediment on old and new bars; 30-50% (50-80% for low-gradient) of the bottom affected; sediment deposits at obstructions, constriction, and bends; moderate deposition of pools prevalent. Large sand/silt deposits in channel common, with numerous small localized areas of fresh sand/silt deposits along top of low banks. 6 5 4	Heavy deposits of fine material, increased bar development; more than 50% (80% for low-gradient) of the bottom changing frequently; pools almost absent due to substantial sediment deposition. Large sand/silt deposits very common in channel, with moderate to heavy sand/silt areas freshly deposited along major portion of overbank areas. 3 2 1
E. Diversity of Epifaunal Substrate and Cover Types (reach) Cover and substrate types: Leaf packs Anchored woody debris/logs/trees Root mats Overhanging and/or inundated vegetation Organic debris accumulation Undercut banks Submerged macrophyte beds Isolated backwaters or inlets	Good mix of favorable aquatic habitats and substrate, at least 70% is stable and present at a stage at allow full colonization potential. At least 7 of the types are present in the reach. Some organic substrates (logs, tress, accumulations) should not be new fall or transient. 12 11 10	Good mix of favorable aquatic habitats and substrates, about 40-70% of which is stable and present at a stage to allow full colonization potential. 5-6 of these types are present in the reach. Some organic substrates (logs, trees, accumulations) may be new fall or transient. 9 8 7	Aquatic habitats and substrates exist but are less than desirable, about 20-40% of which may be stable, but some cover types poorly represented or absent. Only 2-3 of these types are present in the reach. Most organic substrates (logs, trees, accumulations), if present, represent new fall, transient, or above water line. 6 5 4	Less than 20% of the habitats available are stable and available for aquatic colonization. None to 1 of these cover types are present in the reach. Substrates are frequently disturbed, recently moved, or lacking. 3 2 1

Publishing support provided by:
Rolla Publishing Service Center

F**or additional information concerning this publication, contact:**
Director, USGS Kansas Water Science Center
4821 Quail Crest Place, Lawrence, KS (785) 842–9909

Or visit the Kansas Water Science Center Web Site at:
http://ks.water.usgs.gov