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Methods For Evaluating Stream, Riparian, and Biotic Conditions

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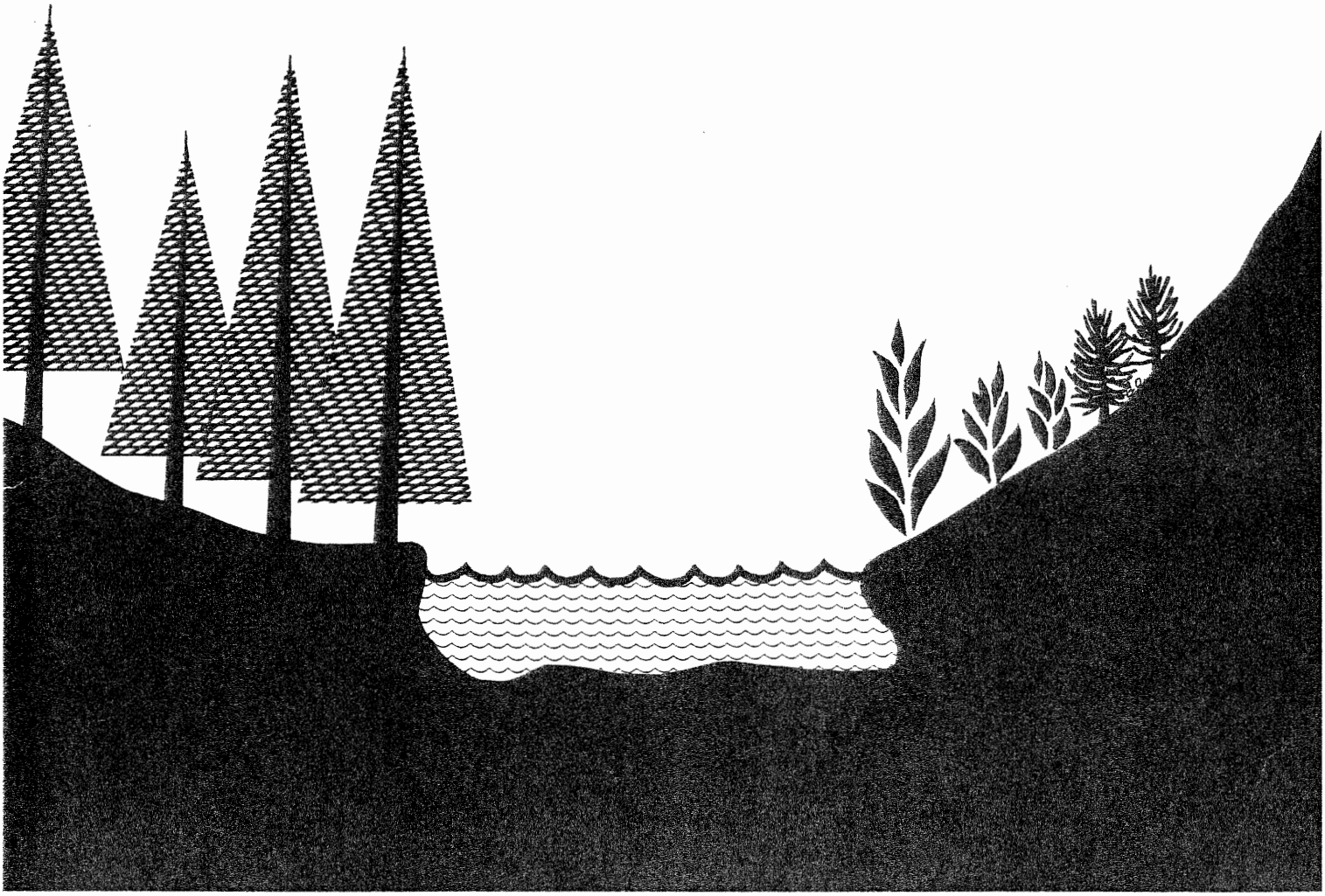

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ERRATA

Page 16 Figure 11, first line
A channel embeddedness of 4 instead of 2.

Page 23 Line 13, 1st column
Elevation instead of evaluation

Page 25 Figure 20
Photograph is upside down

Page 30 Equation 7
Eliminate the last parenthesis

Page 31 Equation 17
The first and last brackets in the denominator are backwards

Page 32 Line 52, 1st Column
28.3 instead of 20.3

Page 33 Table 12, line 2
6-10 instead of <6-10

Page 57 First equation should be

$$\overline{D} = \frac{A}{W} = \frac{\sum_{i=1}^{4}\left(\frac{D_i + D_{i+1}}{2}\right)\left(\frac{W}{4}\right)}{W} = \sum_{i=1}^{4} \frac{D_i + D_{i+1}}{8}$$

THE AUTHORS

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RESEARCH SUMMARY

Most stream habitat evaluation techniques currently in use today have not been tested to determine their validity in describing conditions and have been designed to optimize time rather than accuracy. The purpose of this report is to further standardize the way physical and biological attributes are measured and quantified and to shed light on the strengths and weaknesses of those attributes. This report discusses some of the environmental parameters that best measure and describe conditions existing in aquatic ecosystems. The precision and an estimation of the accuracy that can be expected when measuring many of these conditions are given.

ACKNOWLEDGMENT

We are grateful to the Library Executor of the late Sir Ronald A. Fisher, to Dr. Frank Yates, and the Longman Group Ltd., London, for permission to reprint our table 19 from their book *Statistical Tables for Biological, Agriculture, and Medical Research* (6th ed., 1974) and to the American Society of Civil Engineers for permission to reprint our table 4 from their publication *Sedimentation Engineering* edited by Vito A. Vanomi.

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INTRODUCTION

Background

The past decade has seen an increase in the number of studies evaluating the status and potential of streams as habitats for aquatic organisms. Stream inventories, monitoring, habitat research studies, assessments, channel and flow condition evaluations, and classification are used to evaluate this potential. The success or failure of these stream studies depends on the suitability, comprehensiveness, precision, and accuracy of measurements used to obtain the data upon which final interpretations are based. These interpretations have been used by planners and decision-makers on the assumption that they were derived from measurements that truly described stream habitat conditions and the resulting biotic community.

Within the past decade measurements of stream habitat conditions, such as velocity, depth, and cover, have been incorporated into models designed to indicate fish standing crops and to assist in evaluating impacts from land management activities. Binns (1979) developed a Habitat Quality Index to predict trout standing crops in Wyoming streams. The USDI Fish and Wildlife Service (Cooperative In-stream Flow Group) uses a cluster of aquatic habitat descriptors in a predictive model to quantify the effects of change in streamflow on fish survival. Their Aquatic Habitat Evaluation Team also has developed an Aquatic Habitat Evaluation Procedures model (HEP) and Habitat Suitability Index model (HSI) for obtaining data and interpretation for use in decisionmaking. Wesche (1974) developed a cover rating model that is used on Wyoming streams to determine aquatic habitat conditions and fish standing crops. Cooper (1976) employed an aquatic habitat survey model to measure stream channel conditions for information needed for land use planning. The success of these models depends on whether the model fits the situation, whether the correct combination of habitat descriptors is selected, and how precisely and accurately the habitat descriptors are measured.

Problem

Difficulties arise in developing accurate, complete methodologies because of problems encountered in attempting to quantitatively determine the true state of an aquatic system (Platts 1976). In addition, aquatic specialitists commonly collect their data during the warmer months of the year (from June through September), when access, streamflow, and water quality are optimum for aquatic observation. Aquatic habitats and their biotic communities are seldom evaluated during periods of floods, annual high flows, extreme low flows, anchor ice buildup, ice flow scouring, debris jam breakup, or sudden toxic flushes. Because some important limiting factors, inside or outside the system, usually exert their effects during periods of no data collection, true existing states, or the changes of these states over time in the stream have rarely been determined. A valid understanding of the mix of environmental conditions that control the fishery, therefore, eludes us.

Platts (1974, 1976) demonstrated that while masses of multivariable environmental data can be gathered during these warmer months, complete and reliable information still is lacking. His study also demonstrated that additional descriptive variables, not yet discovered, are needed if adequate quantification of stream condition is to be gained. In today's methodologies (where the "state-of-the-art" lacks refinement and the form often is directed by expediency and low cost), the observed physical, biological, and chemical conditions and variations used to predict fishery condition and reaction often are of low value for providing valid interpretations. These deficiencies must be taken into account by the user when designing procedures, collecting data, and making interpretations.

Most techniques used today to evaluate stream habitat are untested and were designed to optimize time rather than accuracy. Problems can arise if the stream methodology used is not suitable for the environmental situation and if the accuracy of interpretations is not known. Poor resource management decisions can result.

Purpose

The major purpose of this report is to help standardize the way that physical and biological attributes are measured and quantified and to shed light on the strengths and weaknesses of these attributes. Standardization of measurement techniques makes it possible to utilize information from area to area, compare study results, and evaluate information on a uniform basis. Only through constant refinement of present methods, incorporation of additional attributes, and standardization will we ever develop a practical means of obtaining information of use to resource managers. This report takes a step toward this goal and is presented in a format upon which future work can build and improve, thus continually upgrading the value and dependability of habitat and biomass assessment. With this improvement will come confidence in answering questions such as: (1) How much flow is needed in a specific stream for fish perpetuation? (2) How many cattle can be grazed in the riparian zone without excessive damage to the stream? (3) How much sediment can a stream take without losing productivity and will this timber sale exceed that amount? (4) Has the stream been altered from its natural condition? (5) Has the alteration depressed fish populations? (6) And, what needs to be done to rehabilitate the stream?

We hope to improve our methodology by providing an analysis of some of the attributes that are used in computer models or in methods to directly determine stream habitat and biotic conditions. The procedures identified in this report are intended for use by field personnel, such as biologists, hydrologists, aquatic specialists, watershed managers, entomologists, or others involved in providing information for resource management decisions. Our goal is to build a valid, objective, quantitative, repeatable procedure that will provide accurate evaluation of the stream and its biotic communities under any set of conditions. This report (1) presents standard techniques for measuring the aquatic, riparian, and biotic attributes, and (2) stresses the precision and accuracy that can be expected for each measurement. We acknowledge that this report is no panacea and that it provides no magic formula for answering all questions. Its purpose is to provide the field specialist with a method of building on and evaluating the methodology chosen to measure a particular aquatic habitat. The report is directed mainly toward ways of measuring the effects of land use practices, such as logging, road construction, livestock grazing, and mining. It does not address the hydrochemical environment or lower organisms, such as algae. Much refining and testing remains before a valid standard methodology will be available.

Solution

Identification of limiting or enhancing environmental factors is essential to the solution of any biological resource problem. Our inability to measure these factors often keeps us from determining the true dominant limiting factors. For the present, we need to use the best approaches or methods available and define their accuracy and precision.

This report discusses some of the environmental parameters that best measure and describe conditions existing in aquatic ecosystems. These parameters were based on the following criteria:

1. They describe as accurately as possible the physical or biotic portion of the aquatic habitat for which they are designed;
2. They singly or in concert provide the user with insight into what controls biotic communities;
3. They are useful in diagnosing deficiencies in stream habitats; and
4. They avoid duplication and overlap.

STUDY SITES

Aquatic Habitat

Much of the methodology presented in this section was tested on 51 streams in Idaho, 2 in Utah, and 2 in Nevada. The Idaho testing was done in four major areas. One area included 38 tributaries of the South Fork Salmon River where the methods were tested over a 2-year period. The second area included six streams scattered within the Salmon River, the Middle Fork Salmon River, and the South Fork Salmon River drainages where the methods were tested over a 6-year period. The third area included four major chinook salmon (*Onchorhyncus tshawytscha* [Walbaum]) spawning areas in the South Fork Salmon River to be tested over a 15-year period. The fourth area included seven streams in the Middle Fork Payette River drainage to be tested over a 7-year period.

The Utah-Nevada streams were representative of those found in the Basin-Range physiographic province and the Idaho streams were representative of those found in the Rocky Mountain physiographic province (Bailey 1980). The test streams ranged in elevation above mean sea level from 4,500 to 7,500 ft (1 372 to 2 286 m).

A complete description of the study streams is given in Platts (1968), Platts (1974), Platts and Megahan (1975), Platts (1978), and Megahan and others (1980).

Fisheries

The methods for analyzing fish populations are based on tests made over a 2-year period in 38 tributaries of the South Fork Salmon River where collections were obtained by the use of explosives and tests made over a 6-year period in five streams in the Salmon River, Middle Fork Salmon River, and South Fork Salmon River drainages in Idaho where electrofishing procedures were used. Two streams in Utah and two in Nevada were also studied for 2 years to test the reliability of electrofishing.

SAMPLING DESIGN

Usually it is physically, and almost always financially, impossible to make a 100 percent inventory of a condition of concern in the riparian or stream environment. As a consequence, it is necessary to devise a sampling system to provide as accurate a measure of the attribute as possible with acceptable cost and effort. Sampling does not always cause a reduction in reliability just because fewer measurements are taken; good data properly collected on 10 percent of a population can often provide more reliable information than poor data collected on 100 percent of the population.

A population is defined as the set of all possible measurements of the attribute being measured. For example, a fishery biologist might be concerned about the effects of accelerated sedimentation on fry survival in a salmon spawning area. The spawning area covers the entire 50-ft (15.2-m) width of a channel and extends along that stream for 200 ft (60.96 m) providing a total area of 10,000 ft^2 (929 m^2). Assuming a 1-ft^2 (0.09-m^2) core sampler is available, the population consists of 10,000 individual cores. Obviously, it would be impossible to collect 10,000 cores to describe the population. Sampling a portion of the population provides a means of estimating population characteristics, such as its mean and variability, and of defining the reliability of the estimates.

The purpose of this section (and, to a large extent, the entire manual) is to stress that any sample is an estimate of the characteristics of the population and, as such, is subject to error.

Anyone using sampling must be aware of the possibility of error and account for it or describe it. When possible, we have provided some measure of the reliability of the measurements described in this manual, using actual data collected over a number of years in our study streams.

In some cases, only very basic procedures are provided here. If necessary, additional guidance is available in handbooks, standard statistical texts, and from statisticians.

Bias, Accuracy, and Precision

Bias can be considered as any systematic error introduced into a sampling scheme. Bias often results from a lack of randomness in the selection of sample sites. Random selection simply means that every individual in the population has an equal chance of being selected. For example, bias could easily result if a stream surveyor were to sample stream depths by wading with hip boots in January — there would be a natural and understandable tendency to avoid deep sections where boots might be overtopped. In this case, the sample is not random because the greater depths were consciously, or perhaps unconsciously, avoided. Usually some mechanical system is used for site selection to avoid such bias. A table of random numbers or measurements from some arbitrary point is often used to accomplish this. Bias can also result from systematic errors in the measurement process. For example, the stream surveyor who measures water depths while leaning on the measurement rod to maintain balance could easily be introducing bias because the rod tends to sink into the bottom sediments. Investigators should do their best to avoid all known sources of bias in the site selection and measurement process.

Many kinds of errors, including unavoidable bias, exist to influence the accuracy of the data. Accuracy is the degree to which the measured value corresponds to the true value of the population. Unfortunately, the true population value, for example the population mean, is almost never known in natural systems. The best the investigator can do is to avoid bias and make the measurements as precise as possible. Precision can be defined as the repeatability of a series of measurements. Low precision is usually caused by poor or sloppy measurement techniques. Wide differfences between successive measurements or observers is a sure sign of low precision.

Target shooting provides an analogy for the terms "precision," "bias," and "accuracy." A wide grouping of hits all over the target indicates poor precision and poor accuracy. A close grouping indicates high precision, but not necessarily high accuracy. This apparent contradiction can occur when the group is not near the center of the target and is the direct result of bias. A very close group, randomly spaced at the center of the target, indicates unbiased, high precision, and high accuracy shooting. Unlike target shooting, it is almost always impossible to define accuracy in natural systems because the true population values are unknown.

Population Parameters

A parameter is a value used to describe a population. Oftentimes, the mean of the population is the parameter of interest. Means may have limited utility, however, because they give no measure of the dispersion of the values in the population. Accordingly, a second parameter, such as the variance or standard deviation, is often used to estimate population dispersion.

The sample mean, $\bar{X}$, is expressed as:

$$\bar{X} = \frac{\sum_{i=I}^{n} X_i}{n}$$

where X_i equals the individual sample values and n is the total number of samples.

The sample variance, S^2, is:

$$S^2 = \frac{\sum_{i=1}^{n} (X_i - \bar{X})^2}{n-1}.$$

An alternative method for computing variance is:

$$S^2 = \frac{\sum_{i=1}^{n} (X_i)^2 - \left(\sum_{i=1}^{n} X_i\right)^2}{n-1}.$$

The standard deviation, S, is simply the square root of the variance.

One other value provides a dimensionless measure of dispersion; the coefficient of variation (C.V.) is expressed by the ratio of the standard deviation to the mean:

$$C.V. = \frac{S}{\bar{X}}.$$

Some streambed sediment data collected with a McNeil core sampler on the South Fork of the Salmon River during 1971 provide an example of the use of these equations. Twenty samples were collected in the Poverty chinook salmon spawning area in the South Fork Salmon River, Idaho, using a random sampling technique to represent the percentage by weight of the upper 12 inches (30.5 mm) of streambed sediments that are less than 0.25 inch (6.35 mm) in size. The data are listed as follows:

Sample number	Percentage of sample less than 0.25 inch (6.35 mm) by weight
1	44
2	16
3	29
4	40
5	31
6	51
7	22
8	22
9	35
10	42
11	41
12	15
13	21
14	37
15	39
16	27
17	37
18	27
19	26
20	45
Total	647

The estimated population parameters for this sample of 20 cores are calculated as follows:

$$\text{Mean} = \overline{X} = \frac{\sum_{i=1}^{n} X_i}{n} = \frac{647}{20}$$

$$= 32.35 \text{ percent.}$$

$$\text{Variance} = S^2 \frac{\sum_{i=1}^{n} (X_i - \overline{X})^2}{n-1}$$

$$= \frac{1{,}986.55}{19} = 104.56 \text{ percent.}$$

$$\text{Alternately, } S^2 = \frac{\sum_{i=1}^{n} (X_i)^2 - \frac{(\sum_{i=1}^{n} X_i)^2}{n}}{n-1}$$

$$= \frac{22{,}917 - \frac{(647)^2}{20}}{19} = 104.56 \text{ percent.}$$

$$\text{Standard deviation} = S = \sqrt{S^2} = \sqrt{104.56} \text{ percent}$$

$$= 10.22 \text{ percent.}$$

$$\text{Coefficient variation} = \text{C.V.} = \frac{S}{\overline{X}} = \frac{10.22}{32.35}$$

$$= 0.36.$$

Standard Error

The equation for standard deviation presented above provides an estimate of the amount of variation occurring within a population based on a single sample from the population. Because there is variation within the population, the means for successive samples taken from that population also will vary. A measure of the variability between the various sample means is the standard error of the mean. The standard error of the mean is analogous to the standard deviation in that it provides a measure of the variability of individual sample means, just as the standard deviation provides a measure of the variability of individual population values. The standard error of the mean is very useful because it makes it possible to estimate the reliability of the sample mean. The standard error of the mean, $S\overline{x}$, is evaluated using the sample variance and number of observations as:

$$S\overline{x} = \sqrt{\frac{S^2}{n}}.$$

Confidence Limits

The reliability of a sample mean is expressed by the confidence limits for the sample. Sample means presented without some expression of their reliability are almost worthless. Freeze (1967) expresses it well:

> We have it on good authority that "you can fool all of the people some of the time." The oldest and simplest device for misleading folks is the barefaced lie. A method that is nearly as effective and far more subtle is to report a sample estimate without any indication of its reliability.

The confidence intervals (C.I.) for a sample mean are calculated by:[1]

$$\text{C.I.} = \text{mean} \pm (t)(\text{standard error}).$$

The value t is taken from the Student's distribution (appendix 1). In the table, the column headed "d.f." refers to degrees of freedom and is based on sample size. The d.f. selected for use in the table is equal to n-1 for the sample. The column labeled "Probability" determines the kinds of odds the investigator is willing to accept. For example, a probability of 0.05 means that there is only a 5 percent chance that the true mean will fall outside the confidence limits.

For the Poverty spawning area data presented earlier, the standard error of the mean is:

$$S\overline{x} = \sqrt{\frac{S^2}{n}} = \sqrt{\frac{104.56}{20}} = 2.29 \text{ percent.}$$

And the confidence interval, using d.f. equal to 19 (n-1 or 20-1) at the 0.05 probability level, is:

$$\text{C.I.} = \overline{x} \pm (t)(S\overline{x}) = 32.35 \pm (2.093)(2.29)$$

$$= 27.56 \text{ to } 37.14 \text{ percent.}$$

[1]The use of the t statistic assumes that the sample data follow the normal (Gaussian) distribution. Usually, the distribution of data is close enough to the normal distribution that use of the t statistic is warranted. However, tests for normality should be applied if there is any question. An example is provided by percentages, such as those used in the example data set. Percentages may not be normally distributed if many of the sample values fall above 80 or below 20 percent. Data transformations may be useful for assuring normality of a data set. Tests for normality and the necessary data transformations needed to assure normality are presented in standard statistical texts. No normality tests were used for the example data because all values, with one exception, were greater than 20 percent.

For this data set, the probability is 0.05 (or a 1 in 20 chance) that the population mean is outside the range of from 27.56 to 37.14. The more common way to look at this confidence interval is that there is a 95 percent chance that the population mean falls within the range from 27.56 to 37.14. Suppose we wanted to be even more sure that the range included the mean. The 0.01 probability level of t might be selected to accomplish this. In this case, there is a 99 percent chance that the population mean is between 25.80 to 38.90. Now there is only 1 chance in 100 that the confidence interval does not include the mean.

Sample Size

The larger the sample taken, the closer the sample mean and variance will be to the population mean and variance. Accordingly, the chances of making an error are reduced. However, samples cost money. Therefore, it is necessary to strike some balance between the cost of sampling and the cost of making an error. The confidence interval makes it possible to estimate the number of samples needed to obtain any given level of precision (E). As we saw above, the expression (t) ($S\bar{x}$) defined the spread of the confidence interval. If we think of either the plus (+) or minus (−) value of this spread as E, we can define the sample size in terms of any desired value of E as follows.

$$(t)\ (S\bar{x}) = E.$$

However, $S\bar{x}$ can be expressed in terms of S and n as:

$$S\bar{x} = \sqrt{\frac{S^2}{n}}.$$

Substituting this for $S\bar{x}$ above gives:

$$(t^2)\ \frac{S^2}{n} = E^2.$$

Solving for n gives the sample size needed to meet the defined level of precision E as:

$$n = \frac{t^2 S^2}{E^2}.$$

The streambed core data previously presented illustrate the use of this equation. Bjornn (1969 and 1973) did a study to evaluate the emergence of chinook salmon fry from sprawning gravels based on the percentage by weight of sediments less than 0.25 inch (6.35 mm) in size contained in the gravel. Fry mortality was directly proportional to the percentage of sediments smaller than 0.25 inch (6.35 mm). The sample of 20 cores collected in 1971 contained an average of 32.35 percent fines smaller than 0.25 inch (6.35 mm). Assuming Bjornn's relationship is applicable in the South Fork of the Salmon River, the fry mortality for the spawning area sampled would have been 66 percent based on an average of 32.35 percent fines less than 0.25 inch (6.35 mm) in diameter.

As we saw above, the average percent fines smaller than 0.25 inch (6.35 mm) can actually range between 27.6 to 37.1 percent based on the 95 percent confidence interval for the sample of 20 cores. The importance of this confidence interval range is better appreciated when expressed in terms of fry mortality. It is incorrect to say that mortality was equal to 66 percent based on a sample mean of 32.35 percent fines. All we can say is that we are 95 percent sure that the actual mortality falls somewhere between 53 and 79 percent based on the sample confidence interval of 27.6 to 37.1 percent sediments smaller than 0.25 inch (6.35 mm) and Bjornn's relationship. Such a range in mortality could have serious management implications. Suppose, for example, that a viable fish population cannot be maintained if fry mortality exceeds 75 percent. The fact that our sample confidence interval indicates there is a chance the 75 percent level can be exceeded would be a red flag for the land manager responsible for maintaining the fish population. The manager needs to know whether or not to institute an expensive spawning gravel cleaning program in order to protect the fish resource. A logical alternative to jumping into such a program would be to check the validity of the sample mean and confidence interval. This could easily be done by increasing the sample size. Although some additional sampling costs would be required, the potential for savings is substantial.

The sample size equation presented above provides a means to estimate the size of sample required for any desired level of precision, in this case, a level of fine sediments less than 0.25 inch (6.35 mm) in size that will result in a fry mortality level of less than 75 percent. Using Bjornn's relationship, 75 percent mortality will be obtained if the streambed contains 36.3 percent sediment smaller than 0.25 inch (6.35 mm). In this case, E is defined as the allowable value of 36.3 percent minus the sample mean of 32.35 percent or:

$$E = 36.3 - 32.35 = 3.95 \text{ percent.}$$

Knowing E and taking S^2 from the sample, we have all the components we need to solve the sample size equation, except t. Unfortunately, the value for t is based on n and we are trying to solve for n so it is necessary to use a method of successive approximations. The object is to select a value for n such that the corresponding value of t will produce the same calculated value for n when inserted into the sample size equation. This is illustrated with our example data where S^2 is calculated from the sample data and E = 3.95 was determined by the management decision defined above. The $t_{0.01}$ value will be used for the calculation to provide added assurance (at the 99 percent level) that the land manager will not make a mistake.

The first approximation for n might be 31. The $t_{0.01}$ value for n = 31 is 3.659 (using d.f. = n−1 = 30).

Substituting this value for t in the sample size equation:

$$n = \frac{(2.756)^2\ \ (104.6)}{(3.95)^2} = 51.$$

The selected n value of 31 is obviously too low.

A second n value is selected that is closer to 51, say 45. The $t_{0.01}$ for n = 45 is 2.693 using linear interpolation for t in the table. Substituting this value into the sample size equation gives an n of 49 indicating that the sample size is probably close to 50. The estimated sample size is only an approximation so continued refinement of estimated n is not called for — a total sample size of 50 would be the reasonable recommendation to meet the desired level of precision. A total of 20 cores have already been taken but an additional 30 cores should be randomly collected and analyzed to meet statistical requirements.

TRANSECT SYSTEM

The transect line intercept is a line determined by two points on opposite streambanks and is useful as the location reference for the measurement of habitat conditions. This line intercept allows for repeated measurements at exactly the same location at different times and yet allows the randomness in site selection needed to meet statistical requirements. The transect line intercept method has been used successfully in many studies that have documented aquatic conditions over space and time (Herrington and Dunham 1967); Platts 1974; Platts and Megahan 1975; Cooper 1976; Duff and Cooper 1978; Megahan and others 1980; Platts in press).

A reference location (point) the transect will pass through is determined in the middle of the channel. The transect intercept line runs from this point and traverses across the stream perpendicular to the main streamflow to establish reference points on the right and left bank. The right bank is determined by the observer facing downstream. To prevent stake movement from soil freezing and high water flows, steel stakes marking these points should be located above high water flows and driven into the ground at least 3 ft (0.91 m).

The next transect line intercept is determined by measuring along the middle of the channel the required spacing interval from the reference location (see appendix 2). This measurement determines the position of the second transect line intercept reference point on the right bank. In an equal-spaced transect group, the distances between points on the center of the channel that determines the transect line locations are equal. Because the line intercept must be perpendicular to the main streamflow, the distance between points on the banks will vary unless the stream channel form is a straight line. This approach is necessary to assure that transects are perpendicular to the flow which avoids introducing bias in measurement, especially stream width, and assists in delineating the boundaries of plots for electrofishing.

If the purpose is to determine or to monitor an environmental condition of the stream at a single point, then one transect is sufficient. For example, a single transect may be located below a point effluent discharge to determine localized changes in the water column over time. A single transect, however, does not allow determination of the environmental condition of an entire stream or a single reach within a stream, but only those conditions existing at a point within a stream.

If the data collected are to be used to describe the aquatic habitat condition of an entire stream or a reach of the stream receiving a point effluent discharge, then a sufficient number of properly spaced transects are required to determine the habitat conditions with acceptable confidence in the results (see appendix 2). The question often asked is how many transects at what intervals are required to insure reliable information with low confidence intervals so that significant change occurring in the stream will be detected. Even though the needed sample size may be known, money and manpower limitations often make it impossible to use the required number and spacing between transects. In this case, specialists should compensate for this by describing the reduction in accuracy in the data collected.

Transect Cluster

A transect cluster is a group of transects blanketing a stream or stream reach. Three main approaches are used in setting up the cluster. One approach (a multiple transect approach) is to determine the number of transects required to provide the desired sample size and then randomly to select this number of transects so that every point on the stream or reach being evaluated has an equal chance of being selected as a transect line intercept.

The second approach (the multiple station approach) is to randomly select stations throughout the stream or reach and then to group the desired number of transects around each station point. Five grouped transects commonly are used to form one station. Some statisticians favor the station approach because it allows close grouping of the transects. The disadvantage is that the reach between the station is not included in the analysis and can cause bias if the number of stations is inadequate. We have found either the multiple transect or multiple station approach to be adequate, provided the stations or transects are selected randomly and are of sufficient sample size to meet statistical requirements.

The third, and often best, approach is the straitified random station or transect design. This approach assumes that the user has good information on the stream, which then allows intensive sampling in the more complex areas and reduced sampling in the more homogeneous areas. If these requirements are met, better evaluations can then be made with less time and money. This method should not be used, however, unless high confidence exists in the reasons for stratifying the sample.

ACCURACY AND PRECISION OF MEASUREMENTS

Applying methods that will accurately determine environmental conditions is plagued by both bias, such as systematic observer error, and variability, such as that caused by high natural fluctuations in physical and biological conditions. Extreme fluctuations in the condition of the aquatic habitat and the resulting fish population play havoc with small sample sizes. The large variation caused by these fluctuations is further compounded by bias from observer error. To build confidence in results, the quality in the collection of data must be strictly controlled and the accuracy and precision of the measurements should be provided to the user of the data.

Most of the aquatic habitat attributes discussed in this section have been rated as to their ability to be measured accurately and precisely. The determination of accuracy was based on the ability of the measurement to mirror the expected true mean. The accuracy of the aquatic habitat measurements was estimated by graphing each attribute mean for each stream reach by year and analyzing the fluctuation between the annual means. By subjectively evaluating the time trend of the measurements in comparison to how the attribute was believed to have actually performed, accuracy was given a quality rating of poor, fair, good, and excellent. The subjective judgment was further guided by constant remeasurement of different observers' findings and closely watching how the environmental condition being measured performed over time.

Precision is a measure of the ability of an observer to repeatedly produce the same answer or the ability of different observers measuring the same condition to produce the same answer. For example, low precision results when an observer measures the streambank undercut and cannot always distinctly define the reference points to obtain the measurement. Thus, the methodology itself, regardless of the ability of the observer, can cause confusion in what to measure. An observer can come up with a different answer from year to year when measuring an undercut that has not changed during this period. The decrease in precision shows up especially in those measurements done subjectively. Precision was rated by evaluating the confidence intervals obtained in each habitat measurement. The precision of habitat

measurements having a confidence interval over ±21 percent was rated poor, ±11 to 20 percent was fair, ±5 to 10 percent was rated good, and anything less than ±5 percent was rated excellent.

An example of subjective measurements causing lowered precision is the evaluation of streambank instability that is subjectively determined from a narrative description. Many things can lower the precision of this meaurement, such as using different observers over time, observers changing their thinking from year to year, the ability of the procedure to measure accurately the attributes, weather conditions at time of measurement, size of stream, amount and type of experience and training, and degree of streambank instability. Some attributes, therefore, are almost impossible to measure with precision. Evaluations of precision were based on the confidence intervals around the mean over time. Most of the personnel collecting the data used in this study had advanced degrees in fisheries or closely related fields, were well trained, and had good-to-excellent equipment.

STREAM HABITAT EVALUATION

Water Column

The water column, the medium of support and movement for fish and other aquatic organisms, is controlled by the bank, channel gradient, channel form, stream bottom composition, and the volume of water in the channel. The water's constant three dimensional movement pattern, plus its often unpredictable fluctuations in flow rate, makes it difficult to measure and describe. Care must be taken in time trend studies of effects on fish summer standing crops to sample during base flow, which occurs during late summer in many areas. This will help minimize problems caused by fluctuating flow rates. In streams where flows are controlled by man (dams) and where flows vary day by day, care must be taken to sample uniform flow periods over time, or data collected will not be comparable. If the study is determining high flow effects, then timing of data collection must coincide. The data collection must fit uniform conditions as much as possible to be meaningful.

Stream Width

Stream width must be determined precisely to accurately measure fish standing crop and biomass per unit area. Platts (1974) found that as stream width increased, certain fish species decreased in number while others increased.

Stream width is the horizontal distance along the transect line from shore to shore along the existing water surface. Width is that length of the transect line intercept over the stream channel and bank that is covered by water. Stream width was recorded to the nearest 1 ft (0.3 m) in this report, but for more accuracy and precision it should be recorded to the nearest tenth of 1 ft (0.03 m).

To provide consistency in measurement, protruding logs, boulders, stumps, or debris surrounded by water are included in the measurement of the water surface. Islands are not included in the measurement. Any solid accumulation of inorganic sediment particles protruding above the water and more than 1 ft (0.3 m) in width is considered an island. The stream width measurement ends when, on approaching the shoreline, any material is not completely surrounded by water and water is only pocketing between the material (fig. 1). These guidelines are necessary to obtain measurement consistency from year to year on the same stream.

Figure 1. — Stream width boundaries where materials are no longer surrounded by water.

On our test streams, stream width exhibited good precision as determined by the 95 percent confidence interval about the mean of ±5.4 percent. In the stream reaches studied, precision and accuracy of the stream width received a year-to-year quality rating of good (see appendix 3).

Stream Depth

Stream depth is important in providing fish cover, determining stream velocities, and providing a measurement to determine fish standing crop and biomass per unit volume of water. Depth is an important element of the pool quality and fish environment ratings.

Stream depth is the vertical height of the water column from the existing water surface level to the channel bottom measured in tenths of feet (0.03 m). If a streamflow measurement is made, average depth is accurately calculated by dividing the streamflow rate by the product of width and average velocity. If flow data are not available, we determine stream depth as the average of the water depths taken at three locations: one-fourth, one-half, and three-fourths the stream width distance across the transect. The total of the three water measurements is divided by 4 to account for the zero depths at the stream shore where the water surface and the bank or the channel meet. The mathematical basis for this calculation is given in appendix 4.

In our test streams, because of the variation in stream depth and some observer error, the derived sample mean had a 95 percent confidence interval about the mean of ±8.2 percent; precision and accuracy rated good. We have not tested this method on streams averaging over 100 ft (30.5 m) in width, but believe that more than four measurements per transect should be taken when average widths exceed this, especially if transect spacing is wide.

Stream Shore Water Depth

The stream shore depth is critical for fish, especially young-of-the-year (fig. 2 and 3). Also, this measurement is effective in evaluating those land use activities that could modify the streambank or stream bottom morphology.

The water depth at the stream shore is measured at the shoreline or at the edge of a bank overhanging the shoreline (see fig. 2, angle A1). If the angle formed by the bank as it meets the stream bottom is over 90°, the stream shore water depth reading is always zero. If the angle is 90° or less, the water column goes under the streambank and the measurement of the stream shore water depth is greater than zero (see fig. 2, angles A2, A3, and

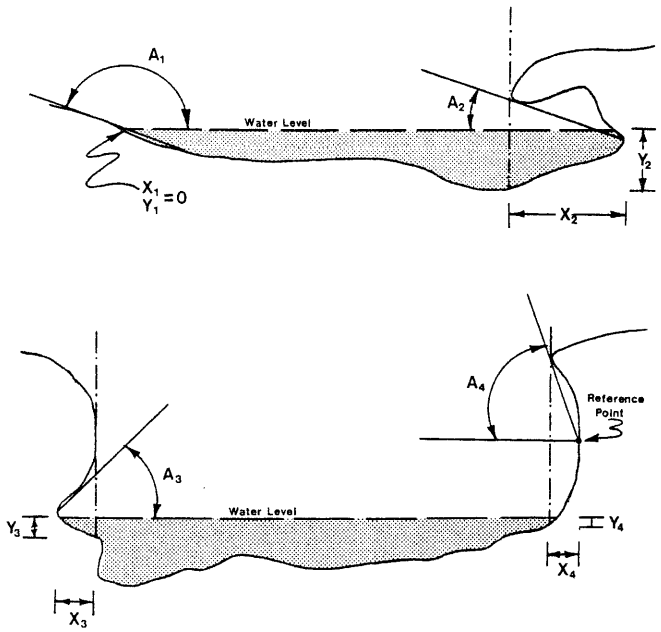


Figure 2. — Hypothetical channel cross sections illustrating bank angle (A), undercut (X), and water depth (Y) measurements.

Figure 3. — Measuring stream shore water depth.

A4). In this report the measurement was taken to tenths of feet and the measurements for both shores were totaled and averaged to get the overall rating for the transect.

Because of the variation in stream shore depth, the test sample mean had a 95 percent confidence interval about the mean of ±16.6 percent. This measurement has fairly wide confidence intervals, mainly because of the high variability and the difficulty in standardizing the technique. We did find, however, that the precision and accuracy rated good from year to year.

Pool

The pool-riffle ratio and pool quality have long been used to determine a stream's potential for rearing fish. A pool is that area of the water column that has slow water velocity and is usually deeper than a riffle or a run (fig. 4). The streambed gradient of the pool itself is often near zero and often concave in shape. The water surface gradient of pools at low flow is close to zero. Pools often contain large eddies with widely varying directions of flow compared to riffles and runs where flow is nearly exclusively downstream. Pools usually are formed around bends or around large-scale obstructions that laterally constrict the channel or cause a sharp drop in the water surface profile.

The measurements to determine the amount of pool, glide, run, and riffle are made directly along the transect line. In this report, pool width has been measured to the nearest foot (0.3); for better precision it should be measured to the nearest tenth of a foot, but only if pools can be defined to that detail. Problems arise in identifying these classes from each other because they are not separated by distinct boundaries.

Because of the variation in the amount of pool within an intercept line crossing the water column, the 95 percent confidence interval about the mean was ±10.3 percent and year-to-year precision and accuracy rated poor in our studies. Confidence intervals around the mean are fairly low because observers, even though they may have interpreted the pool boundaries incorrectly, did so in a uniform manner. The bias arises during the next rating, especially if a new observer is used, when a different interpretation enters into the rating criteria. As a result, the percent pool mean may fluctuate over time even though no changes occur in the water column.

Figure 4. — Determining the point on the transect line that separates the riffle from the pool.

Pool Quality

Pool quality (Platts 1974, and rating system in table 1) estimates the capability of the pool to provide fish survival and growth requirements. Platts (1974) found a good relationship between high quality pools and high fish standing crops. Small, shallow pools, needed by young-of-the-year fish to survive, rate low in quality, however, even though they are essential for their survival. The user should remember that this rating system was developed mainly from the habitat needs of fish of catchable size. In actuality, it takes a combination of pool classes to build a productive fishery.

This rating (table 1) requires that direct measurements of the greatest pool diameter and depth be combined with a cover analysis. Pool cover is any material or condition that provides protection to the fish from its predators or competitors, such as logs, organic debris, overhanging vegetation within 1 ft (0.3 m) of the water surface, rubble, boulders, undercut banks, or water depth.

As the transect line crosses the water column surface, it can intercept one pool, many pools, pools and riffles, or riffles only. If more than one pool is intercepted by the transect line, then pool widths times their respective quality ratings are summed and this

Table 1. — Rating of pool quality; designed for streams between 20 and 60 ft in width

	Description	Pool rating
1A	If the pool maximum diameter is within 10 percent of the average stream width of the study site	Go to 2A, 2B
1B	If the maximum pool diameter exceeds the average stream width of the study site by 10 percent or more	Go to 3A, 3B
1C	If the maximum pool diameter is less than the average stream width of the study site by 10 percent or more	Go to 4A, 4B, 4C
2A	If the pool is less than 2 ft in depth	Go to 5A, 5B
2B	If the pool is more than 2 ft in depth	Go to 3A, 3B
3A	If the pool is over 3 ft in depth or the pool is over 2 ft depth and has abundant fish cover[1]	Rate 5
3B	If the pool is less than 2 ft in depth, or if the pool is between 2 and 3 ft and the pool lacks fish cover	Rate 4
4A	If the pool is over 2 ft with intermediate[2] or better cover	Rate 3
4B	If the pool is less than 2 ft in depth but pool cover for fish is intermediate or better	Rate 2
4C	If the pool is less than 2 ft in depth and pool cover is classified as exposed[3]	Rate 1
5A	If the pool has intermediate to abundant cover	Rate 3
5B	If the pool has exposed cover conditions	Rate 2

[1]If cover is abundant, the pool has excellent instream cover and most of the perimeter of the pool has a fish cover.

[2]If cover is intermediate, the pool has moderate instream cover and one-half of the pool perimeter has fish cover.

[3]If cover is exposed, the pool has poor instream cover and less than one-fourth of the pool perimeter has any fish cover.

total divided by the total pool width to give the weighted average pool rating.

We had some difficulty in determining pool quality in our studies, but the 95 percent confidence interval about the mean was only ±8 percent; therefore, precision was good. Problems arise, however, in getting high accuracy, mainly because of observer error in discriminating pool from riffle, and the key (table 1) was designed for streams between 20 and 60 ft (6.1 and 18.3 m) in width, but was applied to streams from 1.5 to 150 ft (0.5 m to 45.7 m) in width. We found that table 1 should be modified for use on small or large streams.

Pool Feature

Pool feature is designed to classify the condition that formed or is maintaining the pool. Pool features by itself apparently does not have any influence on fish standing crop or species composition as it is the quality of the pool that counts and not the process that formed it (Platts 1974). The main use of this classification is to track changes in the stream caused by beaver or human activities, such as channelization, dams, ponds, or culverts, and to make sure this bias does not enter into the interpretation of a time trend analysis. No confidence levels are given for this measurement. Features forming pools are coded as follows:

Feature forming the pool	Code
Log, tree, root, stump, brush, or debris	1
Channel meander	2
Rubble or gravel	3
Boulder or bedrock	4
Stream channel[2]	5
Fine sediment	6
Streambank	7
Culvert, bridge, or other manmade object	8
Beaver dam or tunnel	9

Riffle

In many streams, riffles produce most of the fishes' aquatic food, form spawning areas, and provide some cover for rearing. Riffles are portions of the water column where water velocity is fast, stream depths are relatively shallow, and the water surface gradient is relatively steep. Channel profile is usually straight to convex. Fish expend high amounts of energy in riffles to maintain position.

Presently, we will record only the pool and riffle classes, because we have found it difficult to make all five (pool, riffle, run, glide, and pocket water) separate classifications. Glide and run are difficult to classify because they tend to fall into both the pool (glide) or riffle (run) classifications. In the results reported here, all glides and slower moving runs are considered pools. The faster moving runs are classified as riffles. The Blackfoot River in eastern Idaho was the only stream for which we felt we could accurately evaluate runs because they stand out, make up a large proportion of the water column, and are easily identified. As discussed in the section on pools, we had difficulties classifying riffles because on most streams there are no sudden breaks in the boundaries separating pools and riffles. Streams with high (more than 3 percent) or low (less than 0.5 percent) channel gradients are the easiest to classify.

In our studies, we had a 95 percent confidence interval about the percent riffle mean of ±17 percent. This is not good, but it is the best that we could expect without better guidelines to delineate the riffle areas. Precision and accuracy were poor.

Glide

A glide is that area of the water column that does not form distinguishable pools, riffles, or runs because it is usually too shallow to be a pool and too slow to be a run. This type of a water column resembles the flow that would be found in a shallow canal. Water surface gradient over the glide is nearly zero. We have not tested this variable sufficiently to draw any conclusions on its reliability for measurement other than it is difficult to classify.

Run

A run is that area of water column that does not form distinguishable pools, riffles, or glides, but has a rapid nonturbulent flow. A run is usually too deep to be a riffle and too fast to be a pool. Runs are like low incline planes where all water flows the

[2]Used when the pool-forming feature cannot be determined.

same fast pace, but at a pace not fast enough to cause much surface rippling. The channel form under a run is usually very uniform and the plane flat. As with the glide classification, we do not have enough data to interpret the precision and accuracy of measurement and we suggest caution in the use of this classification.

Pocket Water

Pocket water (alcoves) consist mainly of small pools behind boulders, rubble, or logs. They form small, shallow microniches where feeding trout and other fish species rest away from the faster waters surrounding the pocket. Pocket water usually supports a much lower fish standing crop than most other pools because of the small pool size and depth. They are usually rated in the pool quality analysis as class 1 or 2 pools. Seldom do they ever get wide enough or deep enough to be rated as class 4 or 5 pools. We have not tested this variable sufficiently to determine its usefulness.

Pool-Riffle Ratio

Pool-riffle ratio is the length or percent of riffle divided into the length or percent of pool. This ratio is a measurement used to predict the stream's capability of providing resting and feeding pools for fish and riffles to produce their food and support their spawning. The common interpretation is that a ratio of 1 to 1 is optimum. However, Platts (1974) found that the highest salmonid fish standing crops in the South Fork Salmon River drainage were in stream reaches with a ratio of 0.4:1. Some streams, however, having a high pool-riffle ratio are known to be high producers of salmonids. The precision and accuracy of the pool-riffle ratio can be no more accurate than can be obtained for pool and riffle individually.

Streamflow Measurement

The water and surrounding channel comprise a complex and dynamic hydraulic system where variable waterflows and associated changes in width, depth, and velocity interact with such factors as sediment transport, channel shape, bank cutting, and size of bottom materials. Fish can respond in a number of ways to variations in these factors, depending on species, age, and time of year. As an independent variable driving the system, flow is an important concern for any stream environment study.

The U.S. Geological Survey is the Federal agency responsible for the national streamflow measurement program. The Survey has developed a number of guides for making flow measurements in its publication series entitled "Techniques for Water-Resources Investigations of the United States Geological Survey" (Buchanan and Sommers 1969).

Flow (Q) is expressed as volume of water moving past a given stream cross section per unit of time and is determined by multiplying the cross sectional area of water (A) in square feet times flow velocity (V) in feet per second to give the traditional units of cubic feet per second. Unfortunately, flow velocity varies greatly within a channel, with both depth and width. Thus, it is not possible to measure streamflow with a single measurement of velocity. Rather, the channel must be broken into a number of sections (fig. 5) to account for variations in velocity with width.

The total flow calculation was based on the sum of the flows for individual sections as follows:

$$1.\quad Q = \sum_{i=1}^{n} (w_{i+1} - w_i)\left(\frac{d_i + d_{i+1}}{2}\right)\left(\frac{v_i + v_{i+1}}{2}\right)$$

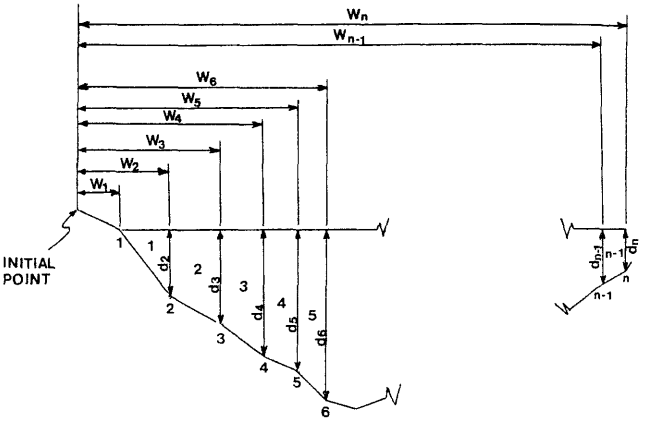


Figure 5. — Shown is the cross section design used for measurement of streamflow.

where:

n = the total number of individual sections

w_i = horizontal distance from the initial point

d_i = water depths for each section

v_i = measured velocity for each section.

The flow for each individual section is calculated and section flows are summed to get the total. For example, the flow for section 4 is:

$$2.\quad Q_4 = (w_5 - w_4)\left(\frac{d_5 + d_4}{2}\right)\left(\frac{v_5 + v_4}{2}\right).$$

At distances w_1 and w_n, the velocities are always 0. Values of D_i are also 0 at these distances, except when a vertical bank occurs as shown on the right bank in figure 5.

The number of subsections used in any flow measurement depends on the variability of velocities within the channel. Usually, at least 20 measurement points should be used unless the channel is extremely regular in both bottom elevation and velocity distribution. Measurement points are taken at all breaks in the gradient of the stream bottom and where any obvious changes in flow velocity occur within the channel. It is advisable to space the partial sections so that no partial section has more than 10 percent of the total flow contained in it. Equal widths of partial sections across the entire cross section are not recommended unless the channel cross section is extremely uniform.

Velocity variations with depth are accounted for by measuring flow at depths where velocity is equal to the average velocity for the total depth. Referring to figure 5, the proper measurement depths vary with water depth as follows:

a. If $d_i < 0.3$ ft (0.1 m), measure v_i at 0.5 d_i.

b. If 0.3 ft (0.1 m) $< d_i <$ 2.5 ft (0.76 m), measure v_i at 0.6 d_i.

c. If $d_i >$ 2.5 ft (0.76 m), measure v_i at 0.2 and 0.8 d and average.

All measurements are referenced to the water surface. For d_i values of less than 0.3 ft (0.1 m) or greater than 2.5 ft (0.76 m), the reference point makes no difference. However, for depths ranging from 0.3 to 2.5 ft (the most common range sampled in aquatic habitat studies) the velocity is taken at 0.6 d_i measured from the water surface. This is equivalent to measuring up 0.4 d_i from the bottom.

Velocity is measured with a current meter attached to a rod or cable for measuring depth. The rod is adjustable and can be set at the proper measurement depth. Many kinds of current meters are available, some of which require counting the number of revolutions of a rotor wheel for a specific period of time (usually, at least 30 seconds). The calculated number of revolutions per second is then used to determine velocity from a rating curve supplied with the meter. Current meters that provide direct measurements of flow velocity are also available. Current meters are precise instruments and should be treated as such. Operation and maintenance must be followed according to the manufacturer's directions in order to assure reliable data.

Solar Radiation

Total light incident on the stream and the resulting heat load are important factors regulating biological activity in the stream. Changes in stream heat load following timber harvest in the riparian zone have been a particular concern because of the potential of deleterious effects on fish caused by sharp increases in maximum water temperatures. Brown (1969) found temperature changes closely related to changes in solar radiation input to the channel following vegetation removal. He developed a procedure to estimate the change in annual maximum water temperature in tributary streams following clearcutting. Wooldridge and Stern (no date) tested Brown's procedure in Washington State and refined the procedure to account for partial removal of streamside vegetation. Their technique involves the use of a fisheye camera photo in conjunction with a polargraph overlay to determine incident shortwave radiation input.

We have used the angle of sun arc as an index of solar radiation input. This is defined in appendix 5.

Channel Morphology

The riparian zone is composed of two dominant features, the flood plain and the channel. The channel is further subdivided into banks and bottom. All of these features represent the interaction between the flow regime for the stream, the quantity and character of sediment movement past the channel section of interest, and the character of the materials making up the bed and banks of the stream. Channels are carved by the tractive forces created by flowing water; so it is logical that some relatively frequent flows dominate the channel-forming process. On the average, flows with a return interval of about 1.5 years or less can be contained within the stream channel, whereas greater flows spread out onto the flood plains. The flow that is just large enough to completely fill the channel — the so-called bank-full flow — is the dominant flow shaping stream channels. The morphology of the channel and flood plain are all referenced to this flow level. For purposes of aquatic environment inventory, the flood plain and components of the channel are defined as follows:

Channel — That cross section containing the stream that is distinct from the surrounding area due to breaks in the general slope of the land, lack of terrestrial vegetation, and changes in the composition of the substrate materials. The channel is made up of streambanks and stream bottoms.

Banks — The portion of the channel cross section that tends to restrict lateral movement of water. The bank often has a gradient steeper than 45^0 and exhibits a distinct break in slope from the stream bottom. Also, an obvious change in substrate materials may be a reliable delineation of the bank.

Stream bottom — The portion of the channel cross section not classified as bank. The bottom is usually composed of stream sediments or water-transported debris and may be covered by rooted or clinging aquatic vegetation. In some geologic situations, the stream bottom may consist of bedrock rather than sediments.

Flood plain — Area adjacent to the channel that is occasionally submerged under water. Usually the flood plain is a low gradient area well covered by various types of riparian vegetation.

Some actual channel cross sections collected on Frenchman Creek in the mountains of southern Idaho illustrate the nomenclature. The channel cross sections were surveyed using the generalized sag tape procedure (Ray and Megahan 1978). The cross sections are plotted using the same horizontal and vertical scales to avoid exaggeration of channel features. Figure 6 shows a well-defined channel with obvious breaks between the channel and the flood plain. The tops of both banks are usually close to the same elevation and are distinct from the flood plain because of breaks in slope gradient as shown in this example. The bottom of the left bank is very well defined compared to the right bank. There is a slight slope break at the water line on the right bank suggesting a possible change in the composition of the substrate material at this point. Field examination showed a very definite transition from bottom sediments to fine-textured, organic bank materials at this point.

It is almost impossible to conduct aquatic surveys under bank-

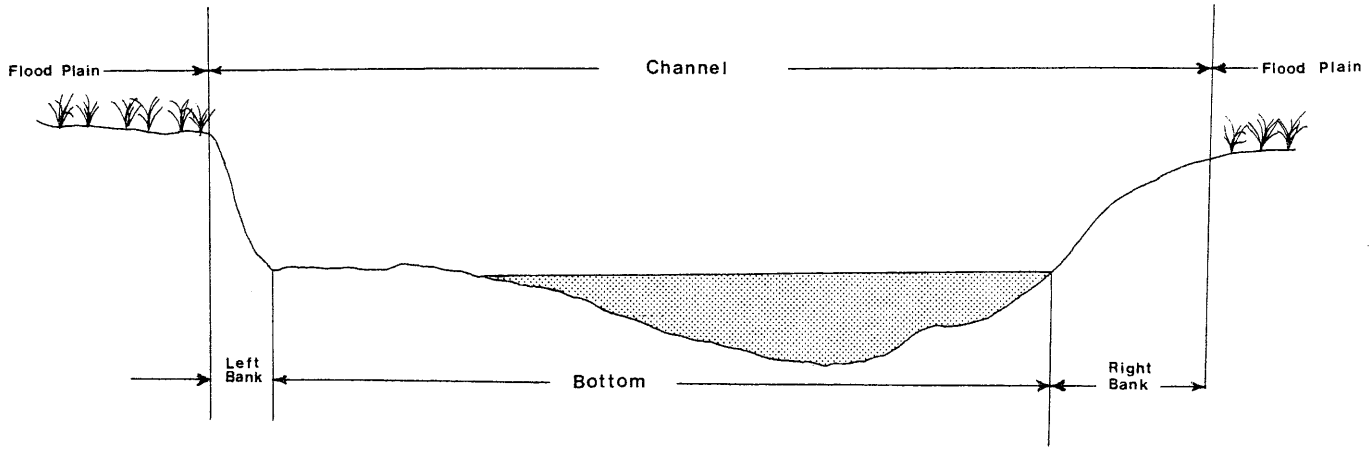


Figure 6. — A well-defined stream channel (downstream view).

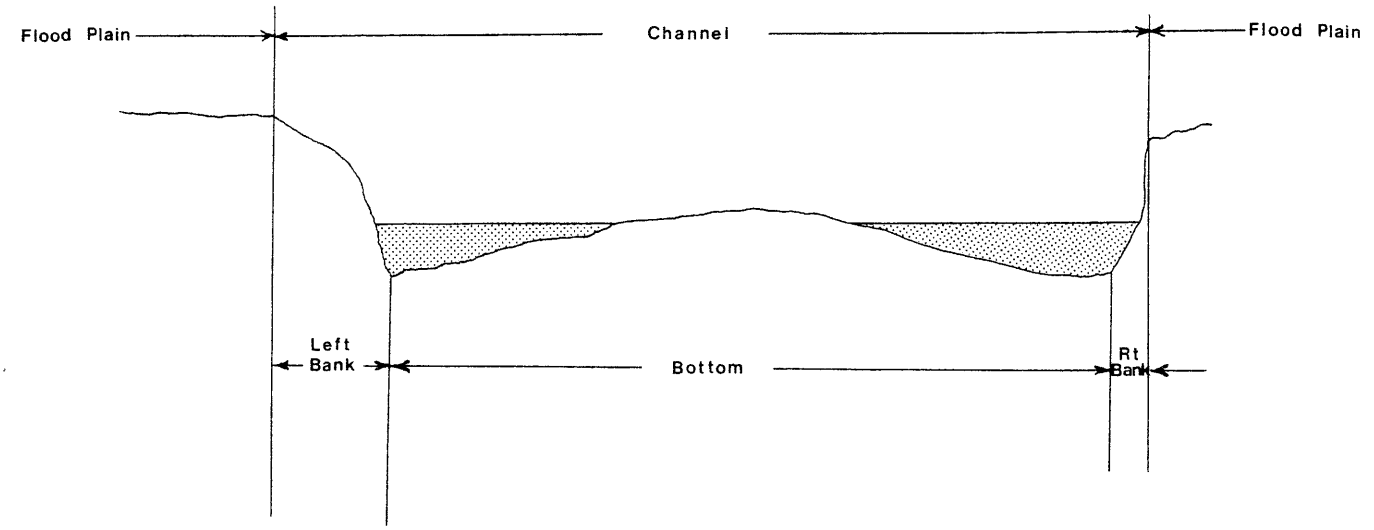


Figure 7. — A well-defined stream channel with concentrated low flows and exposed bottom (downstream view).

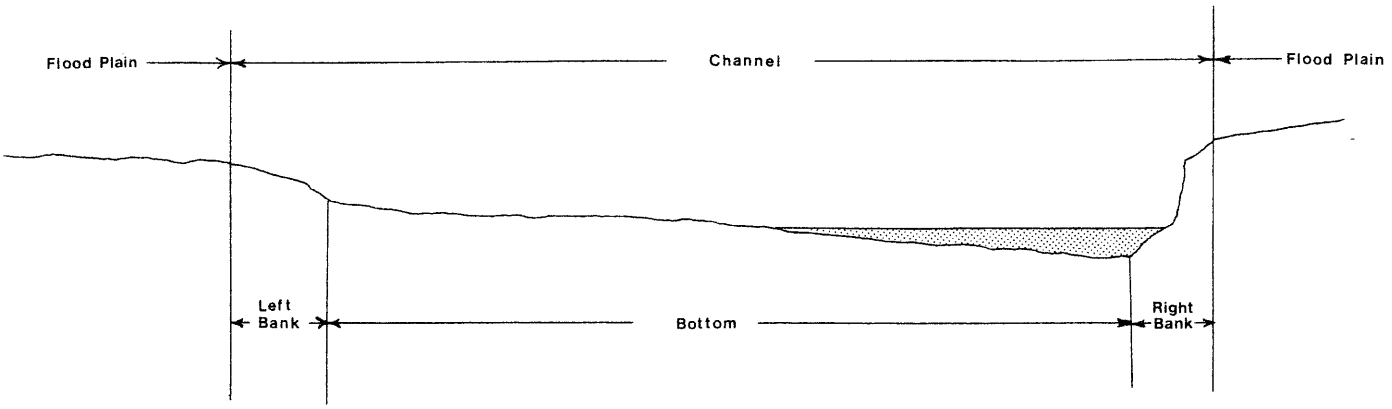


Figure 8. — Stream channel cross channel section on a bend in a stream.

full flow conditions for a variety of reasons, not the least of which is safety. Consequently, aquatic data are almost always collected during low flow periods when the channel is flowing far below bank-full capacity. Commonly, flow levels are so low at this time that part of the stream bottom is exposed, as was the case in all three of these example cross sections.

Figure 7 shows a well-defined channel and banks with low flows concentrated on both sides of the bottom and exposed bottom in the middle.

Figure 8 illustrates a common situation found when a cross section site falls on a bend in the stream. In these cases, the inside of the bend is a zone of sediment deposition and the outside is a zone of erosion for bottom and banks. The result is an asymmetrical cross section as shown in the figure. Oftentimes, it is difficult to delineate the bank, especially the bottom edge, on the inside of the bend because of sediment deposition. The left bank shown in the figure was delineated on the basis of a break in grade and vegetation growth at the top of the bank and a change from lithic sediments to organic materials, plus a small grade change at the bottom of the bank.

Figure 8 also illustrates another situation that occurs with some cross sections, especially asymmetrical cross sections, where tractive forces near the bank cause erosion. However, the top of the bank is usually stabilized by vegetation roots allowing the bank to undercut. Bank undercutting is most common along the outside of bends in the channel, but is not restricted to bends — it can, and commonly does, occur on straight channel reaches as well. Sometimes, the undercut bank collapses, causing a stairstep appearance at the bottom of the bank as shown on the bottom of right bank in figure 8.

Streambank

Well vegetated banks are usually stable regardless of bank undercutting, which provides excellent cover for fish. Valuable fish cover is ultimately lost when bank vegetation decreases, when banks erode too severely, or when banks undercut too quickly and slough off into the stream bottom.

Streambank Soil Alteration

Certain land uses, especially livestock grazing, can start the modification of a stream by causing instability of the bank (Platts in press). This streambank alteration rating, therefore, may provide a warning system for changes that will eventually affect fish populations.

The streambank alteration rating reflects the changes taking place in the bank from any force (table 2). The rating is separated into five classes. Each class, except the one with no alteration, has an evaluation spread of 25 percentage points. Once the class is determined, the observer must decide the actual percent of

Table 2. — Streambank soil alteration rating

Rating	Description
Percent	
0	Streambanks are stable and are not being altered by water flows or animals.
1 to 25	Streambanks are stable, but are being lightly altered along the transect line. Less than 25 percent of the streambank is receiving any kind of stress and if stress is being received, it is very light. Less than 25 percent of the streambank is false, broken down, or eroding.
26 to 50	Streambanks are receiving only moderate alteration along the transect line. At least 50 percent of the streambank is in a natural stable condition. Less than 50 percent of the streambank is false, broken down, or eroding. False banks are rated as altered. Alteration is rated as natural, artificial, or a combination of the two.
51 to 75	Streambanks have received major alteration along the transect line. Less than 50 percent of the streambank is in a stable condition. Over 50 percent of the streambank is false, broken down, or eroding. A false bank that may have gained some stability and cover is still rated as altered. Alteration is rated as natural, artificial, or a combination of the two.
76 to 100	Streambanks along the transect line are severely altered. Less than 25 percent of the streambank is in a stable condition. Over 75 percent of the streambank is false,[1] broken down, or eroding. A past damaged bank, now classified as a false bank, that has gained some stability and cover is still rated as altered. Alteration is rated as natural, artificial, or a combination of the two.

[1]False banks are those banks which have been cut back by cattle and are no longer immediately adjacent to the stream. They can become stabilized by vegetation, but base flows are usually too far removed from the stream to provide fish cover.

instability. Streambanks are evaluated on the basis of how far they have moved away from optimum conditions for the respective habitat type. Therefore, the observer must be able to visualize the streambank as it would appear under optimum conditions. Any natural or artificial alteration deviating from this optimum condition is included in the evaluation. This visualization makes uniformity in rating an alteration difficult, because it is difficult to train all observers to visualize the same optimum bank condition. Natural alteration is any change in the bank produced by natural events. Artificial alteration is any change obviously produced by exotic force. Trampling by man or livestock, disturbance by bulldozers, etc., are examples of artificial changes.

Natural and artificial alterations are reported individually, but together they cannot exceed 100 percent. Only that part of the streambank intercepted by the channel cross section transect line enters the evaluation in order to reduce the confidence intervals. Channel cross section transect lines have no end. The line crosses both streambanks as the channel transect line is extended. Rating the complete bank as a unit between groups of transects in our studies resulted in greater observer error.

It is commonly difficult to distinguish artificial from natural alterations; if there is any doubt, the alteration is classified as natural. It is possible to have artificial alterations cover already existing natural alterations and vice versa. Only the major type of alteration on a unit area enters the rating system in this case.

Table 3. — Streambank vegetative stability rating

Rating	Description
4 (Excellent)	Over 80 percent of the streambank surfaces are covered by vegetation in vigorous condition or by boulders and rubble. If the streambank is not covered by vegetation, it is protected by materials that do not allow bank erosion.
3 (Good)	Fifty to 79 percent of the streambank surfaces are covered by vegetation or by gravel or larger material. Those areas not covered by vegetation are protected by materials that allow only minor erosion.
2 (Fair)	Twenty-five to 49 percent of the streambank surfaces are covered by vegetation or by gravel or larger material. Those areas not covered by vegetation are covered by materials that give limited protection.
1 (Poor)	Less than 25 percent of the streambank surfaces are covered by vegetation or by gravel or larger material. That area not covered by vegetation provides little or no control over erosion and the banks are usually eroded each year by high water flows.

The cross sectional profile methods to be discussed later hopefully will replace this variable; however, the profiles do not determine whether changes in the streambank are caused by natural or artificial forces. Because the 95 percent confidence interval ($\pm$12.3 percent) around the mean and observer variation is quite wide, interpreting the data must be done carefully. Between the streams studied, there is a wide spread in the precision and accuracy of measurements. Precision was rated fair to good, but accuracy was rated mainly poor to fair, which means caution should be used in evaluating the data.

Streambank Vegetative Stability

The ability of vegetation and other materials on the streambank to resist erosion from flowing water was rated (table 3). The rating relates primarily to stability generated by vegetative cover, except in those cases where bedrock, boulder, or rubble stabilizes the streambanks. The rating takes all these protective coverings into account. The rated portion of the bank or flood plain includes only that area intercepted by the transect line within 5 ft (1.5 m) of the stream to the top of the bank. Surprisingly, the confidence intervals around the means from our study sites are quite low (about $\pm$3 percent); however, year-to-year precision and accuracy rated only fair. Therefore, the user should be cautious in its use.

Streambank Undercut

Streambank undercut provides cover for fish and often is considered a condition favorable to producing high fish biomass. Undercut is a good indicator of how successfully streambanks are protected under alternative land uses, such as livestock grazing and road building. The undercut, if it exists, is measured with a measuring rod directly under the transect line from the furthest point of protrusion of the bank to the furthest undercut of the bank (fig. 2); the water level does not influence this reading. In the studies reported here, the measurement was recorded to the nearest tenth of a foot. If more than one undercut occurs under

the transect, only the dominant undercut is recorded.

The 95 percent confidence intervals around the means in our studies (±18.5 percent) are wide; year-to-year precision and accuracy, however, are rated good. The major cause of the wide confidence interval is that the two points that define the undercut measurements are difficult to accurately determine. Then, too, there is naturally high variation in undercuts.

Stream Channel-Bank Angle

Fish often congregate near the streambank for the edge effect it provides. If the bank has been cut away and moved back from the water column, valuable rearing habitat is lost. This measurement is effective for monitoring land uses that can change the morphology and location of the streambank.

A clinometer is used to measure the angle formed by the downward sloping streambank as it meets the more horizontal stream bottom (fig. 9 and 10). If the streambank is undercut, the angle is always less than 90°. The angle is determined directly from the clinometer placed on the top of the rod as it forms the angle determined by the protruding edge of the bank to the midpoint of the undercut under the transect line (fig. 4).

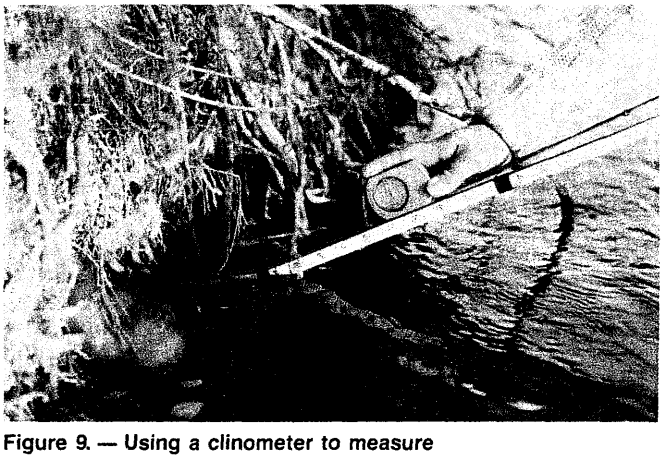


Figure 9. — Using a clinometer to measure a bank angle of 45°.

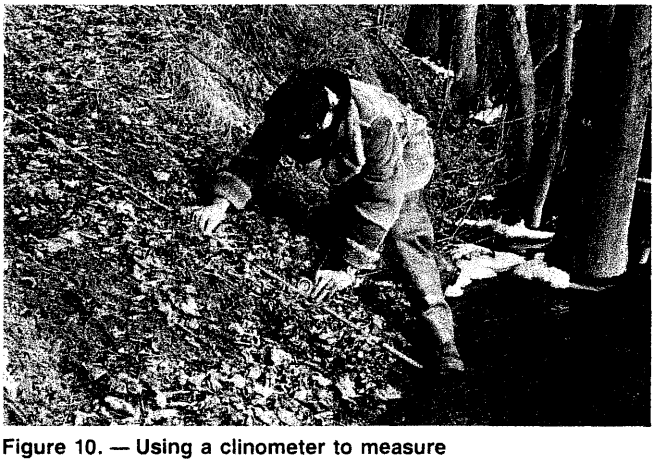


Figure 10. — Using a clinometer to measure a bank angle of 145°.

If the bank is not undercut, then the angle is 90° or more and is read from the bank side by placing the clinometer on the top of the measuring rod that is alined parallel to the streambank along the transect. The clinometer reading is subtracted from 180° to get the bank angle.

A streambank angle over 90° is easily read with precision and accuracy. An angle less than 90° is more difficult to read as multiple undercuts can complicate the bank profile making it difficult to determine the points delineating the angle. The key is to include the midpoint of the dominant undercut in the bank profile. The 95 percent confidence intervals around the means in our studies are quite narrow (±4.4 percent) and year-to-year precision and accuracy rate good.

Stream Bottom

The stream bottom is bounded by the streambanks and is the relatively level substrate plane over which the water column moves. The substrate is the mineral or organic material that forms the bed of the stream. During low flows the water column may recede from the streambank and not cover all of the stream bottom. During high flows the main channel bottom, overflow channels, and the streambanks are often completely covered with water. The stream bottom merges into the bank where the bottom rises to a steep angle toward the channel margin.

If a stream can only be sampled once, the low flow period is best since bed composition is relatively stable during this period. However, if percent fines in the redds is being used to determine their quality, it should be recognized that summer measurements of channel composition may not present a true picture of redd composition during winter high flow months when the fish eggs and alevins are in the gravel. Fine sediment measurements during summer conditions, at the time the major fish biomass is being produced, is important.

Channel Elevation

Channel elevation can be an indicator of certain conditions, such as the amount of channel icing or summer water temperature that can affect fish. Channel elevations can be determined within ±40 ft (12.0 m) from U.S. Department of the Interior Geological Survey topographic quadrangle maps with 40-ft (12.0 m) contours as long as the transect sites can be correctly located on the map. If maps are not available, altimeters can be used. These instruments are accurate if calibrated each morning at an official Geological Survey elevation marker and calibrated again during the day if barometric pressure changes significantly. The accuracy of "Thomen" hand-held altimeter measurements checked against a quadrangle map was within ±50 ft (15.3 m) of the map elevation.

Channel Gradient

Channel gradient is an important variable regulating stream velocity and as such is a concern for aquatic environment studies. Channel gradient is defined as the drop in water surface elevation per unit length of channel. Usually, channel gradients are taken in conjunction with channel cross section measurements. For our study streams, we measure the difference in water surface elevation between points located 100 ft (30.5 m) upstream and 100 ft downstream from each cross section. This assumes that both the channel and the water surface have the same gradient. Horizontal distance between upstream and downstream points is measured along the bank following the general longitudinal shape of the water surface. When measuring distances, care is taken to strike a

balance between measuring every minor fluctuation in the edge of the water surface, on one hand, to measuring across bends in the general shape of the channel, on the other. The channel gradient must be uniform for the 200-ft (31-m) long channel reach (100 ft upstream and downstream from a point) included in the measurement. If this is not the case, the distance should be reduced accordingly to wherever an obvious break in channel gradient occurs.

Elevations for determining gradient are read using an engineer's level and a stadia rod held at the water surface (normally at the water's edge). It may not be necessary to use an engineer's level for some applications. For example, hand level or clinometer measurements may provide acceptable gradient measurements in the design of channel improvement structures.

Channel Sinuosity

Channel sinuosity is defined as the ratio of channel length between two points on a channel to the straight line distance between the same two points. The ratio can vary from 1 for straight channels to 4 or more for strongly meandering channels. The value is useful for providing gross comparisons of aquatic habitat conditions between streams or reaches within the same stream. In general, low sinuosity suggests steeper channel gradient, fairly uniform cross section shapes, limited bank cutting, and limited pools. High sinuosity is associated with lower gradients, asymmetrical cross sections, overhanging banks, and bank pools on the outside of curves. The last situation is common on channel reaches in meadow areas.

Sinuosity should be determined over a channel reach long enough to make the value meaningful. This is based on the size of the channel and the nature of the reach. We use a distance of 20 times the bankful width to determine sinuosity.

Stream Channel Substrate

Surface visual analysis. — The composition of the channel substrate (table 4) is determined along the transect line from streamside to streamside. A measuring tape is stretched between the end points of each transect, and each 1-ft (0.3-m) division of the measuring tape is projected by eye vertically to the stream bottom and the materials assigned to the major sediment class observed for each 1-ft division of the bottom (table 4). For example, 1 ft of stream bottom containing 4 inches rubble, 6 inches gravel, and 2 inches fine sediment would be classified as 1 ft of gravel. With a large enough sample it is assumed that any bias in assigning the dominant sediment class would be compensated for. The individual 1 ft classifications are totaled to obtain the amount of bottom in each of the size classifications and these are totaled to equal the total transect width. We use reference sediment samples embedded

Table 4. — Classification of stream substrate channel materials by particle size from Lane (1947) based on sediment terminology of the American Geophysical Union[1]

Class name	Size range[2]				Approximate sieve mesh openings per inch	
	Millimeters		Microns	Inches	Tyler screens	United States standard
(1)	(2)	(3)	(4)	(5)	(6)	(7)
Very large boulders		4,096-2,048		160-80		
Large boulders		2,048-1,024		80-40		
Medium boulders		1,024-512		40-20		
Small boulders		* 512-256		20-10		
Large cobbles		256-128		10-5		
Small cobbles		* 128-64		5-2.5		
Very course gravel		64-32		2.5-1.3		
Course gravel		* 32-16		1.3-0.6		
Medium gravel		16-8		0.6-0.3	2-1/2	
Fine gravel		8-4		0.3-0.16	5	5
Very fine gravel		* 4-2		0.16-0.08	9	10
Very course sand	2-1	2.000-1.000	2,000-1,000		16	18
Course sand	1-1/2	*1.000-0.500	1,000-500		32	35
Medium sand	1/2-1/4	0.500-0.250	500-250		60	60
Fine sand	1/4-1/8	0.250-0.125	250-125		115	120
Very fine sand	1/8-1/16	*0.125-0.062	125-62		250	230
Course silt	1/16-1/32	0.062-0.031	62-31		270	
Medium silt	1/32-1/64	0.031-0.016	31-16			
Fine silt	1/64-1/128	0.016-0.008	16-8			
Very fine silt	1/128-1/256	0.008-0.004	8-4			
Coarse clay	1/256-1/512	0.004-0.0020	4-2			
Medium clay	1/512-1/1,024	0.0020-0.0010	2-1			
Fine clay	1/1,024-1/2,048	0.0010-0.0005	1-0.5			
Very fine clay	1/2,048-1/4,096	0.0005-0.00024	0.5-0.24			

[1]Reprinted with permission from *Sedimentation Engineering*, edited by Vito A. Vanomi and published by American Society of Civil Engineers, New York.

[2]Recommended sieve sizes are indicated by an asterisk (*).

Table 5. — Classification of stream substrate channel materials by particle size

Particle diameter size		Sediment classification
Millimeters	*Inches*	
610.0 or more	24.0 or more	Large boulder
305.0 to 609.0	12.0 to 23.9	Small boulder
76.1 to 304.0	3.0 to 11.9	Rubble (cobble)
4.81 to 76.0	.19 to 2.9	Gravel
.83 to 4.71	.033 to .18	Fine sediment — large
.83 or less	.033 and less	Fine sediment — fine

in plastic cubes placed on the bottom to help classify the smaller sediment class break sizes.

The classification in table 4 presents the accepted terminology and size classes for stream sediments and should be adapted and used by all specialists working with stream channel substrates. This is the only way all disciplines will use a single standardized procedure. The classification is well suited to the needs of biologists. Because our work was initiated in 1966, we used the classification shown in table 5, and the interpretations of boulder, rubble, gravel, and fine sediments in this study are based on this classification.

Boulder. — Boulder is stratified into two classes, large and small. Percent boulder in the channel can be determined fairly accurately if there is not a high amount of rubble between the 11- to 11.9-inch (279.4- to 302.3-mm) class. The 95 percent confidence interval around the mean of percent boulder (about ±40.9 percent) is high in our analysis in those streams containing boulder, because boulder makes up such a low percentage of the channel and is highly variable. In boulder-dominated channels these intervals can be expected to decrease greatly. Year-to-year precision was poor but accuracy in this measurement was rated good because this substrate class is easy to identify and measure. Also, there is little instream movement of boulder from year to year so time-trend analyses also have good accuracy and precision.

Rubble. — Rubble stabilizes the stream bottom, provides habitat for fish rearing, and is the substrate where much of the food for fish is produced. Measurement of the amount of rubble in the channel in our studies, like boulder, had high confidence intervals around the means (±35.9 percent) because of the high natural variation in the amount of rubble and the difficulty in accurate classification of those particle sizes between 2.5 to 3.5 inches (63.5 to 88.9 mm) in diameter. Year-to-year precision was low.

Gravel. — Gravel is important for spawning, incubation of embryos, and as substrate for some aquatic invertebrates. This particle size is a major sediment component in many small streams in our area. In our studies, the 95 percent confidence intervals around the means (about ±6 percent) is much lower than for rubble and boulder because gravel is more uniformally distributed in the channel. Year-to-year precision and accuracy ratings, however, were poor because the identification of gravel at both ends of the size spectrum is difficult. Particle sizes between 2.5 and 3.0 inches (63.5 and 76.1 mm) tend to be called rubble, whereas particles near the 0.19-inch (4.75-mm) range are often classified as fine sediment. Different sediment size classes embedded in plastic that can be laid on the channel for comparison help considerably in eliminating this bias.

Fine sediment. — Fine sediment is separated into two classes consisting of large fine sediment and small fine sediment. The reason for the separation is that the large fine particles can trap alevins in the redds, but the small fine particles decrease water permeability through spawning gravels. In our studies, the 95 percent confidence intervals around the means (±27.7 percent for large fine sediment and ±17.3 percent for small fine sediment) are wide. Year-to-year precision and accuracy rate fair; so some difficulty exists in collecting reliable data. The plasticized samples help considerably in defining the gray area between 0.19 inch (4.7 mm) and 0.3 inch (7.6 mm), which is gravel but often is classified as fine sediment.

Embeddedness. — Embeddedness rates the degree that the larger particles (boulder, rubble, or gravel) are surrounded or covered by fine sediment (table 6). The rating is a measurement of how much of the surface area of the larger size particles is covered by fine sediment (fig. 11). This should allow evaluation of the channel substrate's suitability for spawning, egg incubation, and habitats for aquatic invertebrates, and young overwintering fish. The rearing quality of the instream cover provided by the substrate can be evaluated also. As the percent of embeddedness decreases, the biotic productivity is also thought to decrease.

In our studies, the 95 percent confidence interval around the embeddedness mean was quite low (±5.4 percent), year-to-year precision was rated good, and accuracy was rated fair. Therefore, this is a fairly dependable measurement. The quantitative relationship between this variable and fish health and survival is not well known. Of the streams studied, some had a high fish biomass but low embeddedness rating.

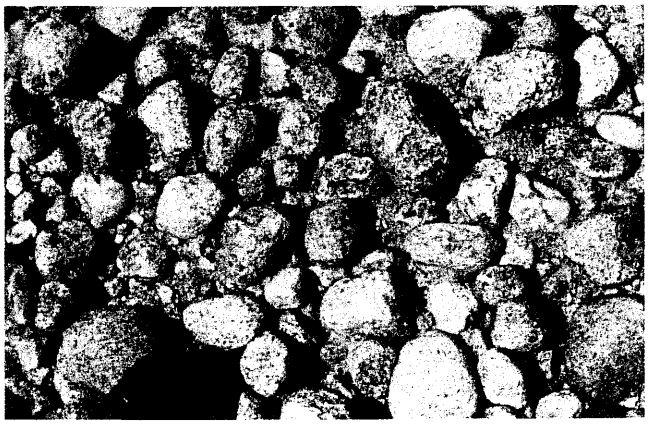


Figure 11. — A channel embeddedness of 2 because about 20 percent of the perimeter of the rubble-gravel particles are covered by fine sediment.

Table 6. — Embeddedness rating for channel materials (gravel, rubble, and boulder)

Rating	Rating description
5	Gravel, rubble, and boulder particles have less than 5 percent of their surface covered by fine sediment.
4	Gravel, rubble, and boulder particles have between 5 to 25 percent of their surface covered by fine sediment.
3	Gravel, rubble, and boulder particles have between 25 and 50 percent of their surface covered by fine sediment.
2	Gravel, rubble, and boulder particles have between 50 and 75 percent of their surface covered by fine sediment.
1	Gravel, rubble, and boulder particles have over 75 percent of their surface covered by fine sediment.

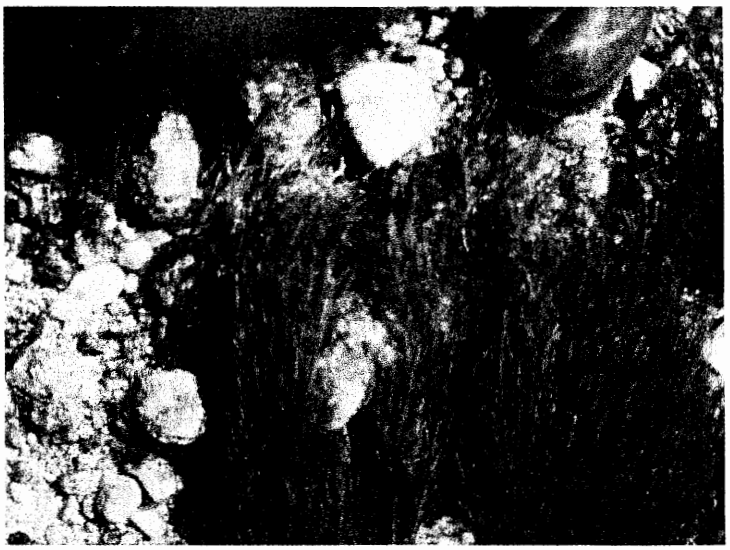


Figure 12. — Aquatic vegetation dense enough to be classified as instream cover.

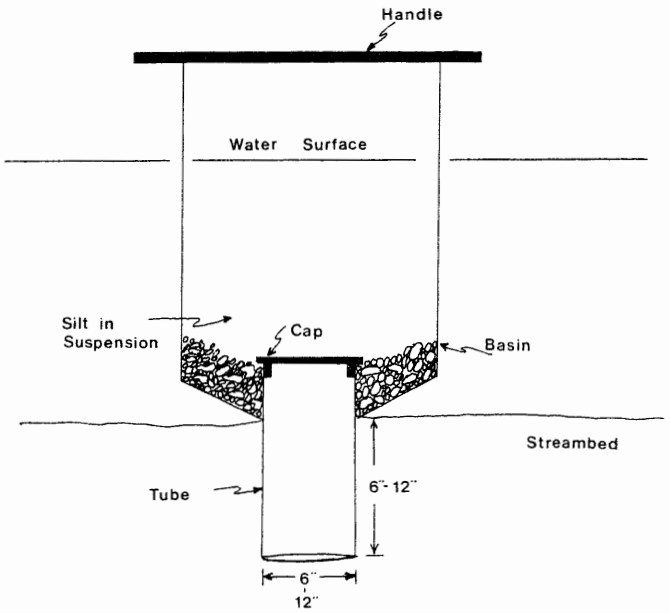


Figure 13. — Streambed depth material sampler (McNeil) type with completed sample.

Channel vegetative cover. Instream vegetative cover is measured directly along the transect line (fig. 12). Each 1-ft (0.3 m) division of the measuring tape across the transect is evaluated. If more than 50 percent of the foot contains cover, the complete foot is classified as cover. If not, it is ignored. Cover includes algal mats, mosses, rooted aquatic plants, organic debris, downed timber, and brush capable of providing protection for young-of-the-year fish. Thin films of algae on the channel substrate would not be included.

The 95 percent confidence interval around the mean in our studies were wide (±26.2 percent), mainly because of the large natural variation in the cover occurring in the channel. Year-to-year precision and accuracy were rated fair, which means that only major changes in cover condition will be detected by this method. The main problem with this measurement is that it is difficult to get agreement between what will and what will not provide adequate cover for young-of-the-year fish.

Subsurface analysis.[3] — Methods for sampling and analyzing the particle size distribution of gravels used by spawning salmonids have evolved slowly during the past 20 years. The first quantitative samplers to receive general use were metal tubes, open at both ends, that were forced into the substrate. Sediments encased by the tubes were removed by hand for analysis. A variety of samplers using this principle have been developed, but one described by McNeil (1964) and McNeil and Ahnell (1964) has become widely accepted for sampling streambed sediments.

More recently, scientists began experimenting with cryogenic devices to obtain sediment samples. These devices, generally referred to as "freeze-core" samplers, consist of a hollow probe driven into the streambed and cooled with a cryogenic medium. After a prescribed time of cooling, the probe and a frozen core of surrounding sediment adhering to it are extracted. Liquid nitrogen, liquid oxygen, solidified carbon dioxide ("dry ice"), liquid carbon dioxide (CO_2) and a mixture of acetone, dry ice, and alcohol have been used experimentally as freezing media. Several years of development have produced a reliable sampler (Walkotten 1976) that uses liquid CO_2. The freeze-core sampler, like the McNeil core sampler, has become widely accepted for sampling stream substrates.

[3]Contributed by Dr. Fred Everest, Research Fishery Biologist, U.S. Department of Agriculture, Forest Service, Corvallis, Oreg.

McNeil sampler. — The McNeil core sampler is usually constructed out of stainless steel and can be modified to fit most sampling situations (fig. 13). The sampler is worked into the channel substrate, and the encased sediment core is dug out by hand and deposited in a built-in basin. When all sediments have been removed to the level of the lip of the core tube, a cap is placed over the tube to prevent water and the collected sediments from escaping when the tube is lifted out of the water. Those suspended sediments in the tube below the cap are lost, but this loss is generally an insignificant percentage of the total sample.

The sediments and water collected are strained through a series of sieves to determine the particle size distribution, percent fines, or geometric mean diameter of the distribution. The sediments collected can be analyzed in the laboratory using the "dry" method or in the field using the "wet" method.

Disadvantages in using the McNeil sampler are that (1) it is limited in particle size diameter to the size the coring tube can trap; (2) it completely mixes the core materials so no interpretation can be made of vertical and horizontal differences in particle size distribution; (3) it is limited to the depth the core can enter the channel substrate, a factor controlled by the water depth, length of the collector's arm, and the depth the core sampler can be pushed into the channel; (4) it is biased if the core tube pushes larger particle sizes out of the collecting area; (5) it allows suspended sediments in the core to be lost; and (6) it cannot be used if the particle sizes are so big or the channel substrate so hard or so cemented that the core cannot be pushed to the required depth.

Regardless of the limitations of this method, when time and money are considered, this is probably the most economical method available to obtain estimates of channel substrate particle size distributions up to 12 inches (305 mm) in channel depth. We recommend the diameter of the McNeil tube to be at least 12 inches (305 mm).

Freeze-core samplers. — All of the freeze-core equipment presently available utilizes the same principles, but individual devices may use from one to many probes. The size of sample collected is directly related to the number of probes used and the amount of cryogenic medium used per probe.

Walkotten (1976), Lotspeich and Reid (1980), Everest and others (1980), and Platts and Penton (1980) give discussion on the construction, parts, operations of freeze-core samplers, and analysis of samples collected by the freeze-core method. Platts and Penton (1980) and Ringler (1970) believe that the single probe freeze-core sampler may be biased to the selection of larger size sediment particles.

The accuracy and precision of the single freeze-core and McNeil sampler have been compared in laboratory experiments. Samples collected by both devices were found to be representative of a known sediment mixture, but the freeze-core sampler was more accurate (Walkotten 1976). It is also more versatile, functioning under a wider variety of weather and water conditions, but it too has several disdvantages. It is difficult to drive probes into substrate containing many particles over 10 inches (25 cm) in diameter, and the freeze-core technique is equipment intensive, requiring CO_2 bottles, hoses, manifolds, probes, and sample extractors. Also, since it is necessary to subsample cores by depth for accurate interpretation of gravel quality (Everest and others 1980), it is often necessary to collect more massive cores than can be easily obtained by the single-core technique. For example, Adams (1980) used a single-probe device to extensively sample stream substrates in the Oregon coast range. He was able to extract up to six cores of sediment averaging about 3.5 lb per core (1.6 kg/core) per 20-lb tank (9.07-kg) of CO_2. Cores of such size are minimal for individual vertical subsampling. Skaugset (1980), on the other hand, was able to obtain cores exceeding 44.1 lb (20 kg) with a single probe device using 10 liters of liquid nitrogen per sample. Skaugset's cores were large enough for representative vertical subsampling, but liquid nitrogen is more expensive and more difficult to obtain, store, and use than liquid CO_2.

To alleviate problems caused by the small size of cores obtained by the single-probe sampler using CO_2, and to avoid use of liquid nitrogen as a cooling medium, Lotspeich and Reid (1980) and Everest and others (1980) modified the single-probe device. The modified freeze-core sampler uses a triangular array of three probes driven into the substrate through a template, that keeps the probes in a fixed relationship to each other. The "tri-tube" sampler (fig. 14) retains all of the advantages of the single freeze-core sampler, but it extracts larger cores—often more than 44.1 lb per 20-lb (20 kg per 9.1-kg) tank of CO_2 — which are probably more representative of substrate composition than small cores obtained by the single freeze-core, or cores obtained with McNeil samplers.

We recommend use of the multiprobe procedure if an analysis of horizontal and vertical stratification of sediments is required. We suggest use of the tri-tube sampler described by Lotspeich and Reid (1980) and Everest and others (1980) when numerous cores must be collected, and the sampler described by Platts and Penton (1980) when only a few large cores are needed.

The freeze methods allow collection of eggs and alevins in a redd at any stage of development; the methods will function at most air or water temperatures or stream depths, and will allow analysis of horizontal and vertical locations of eggs and alevins. But, because these techniques require several pieces of equipment, they are most conveniently used in accessible areas.

A major advantage of the freeze-core sampler is that it provides opportunity for vertical stratification of substrate cores. Everest and others (1980) have developed a subsampler that consists of a series of open-topped boxes made of 26-gage galvanized sheet metal (fig. 15). A core is laid horizontally on the boxes of the subsampler and thawed with a blowtorch. Sediments freed from the core drop directly into the boxes below.

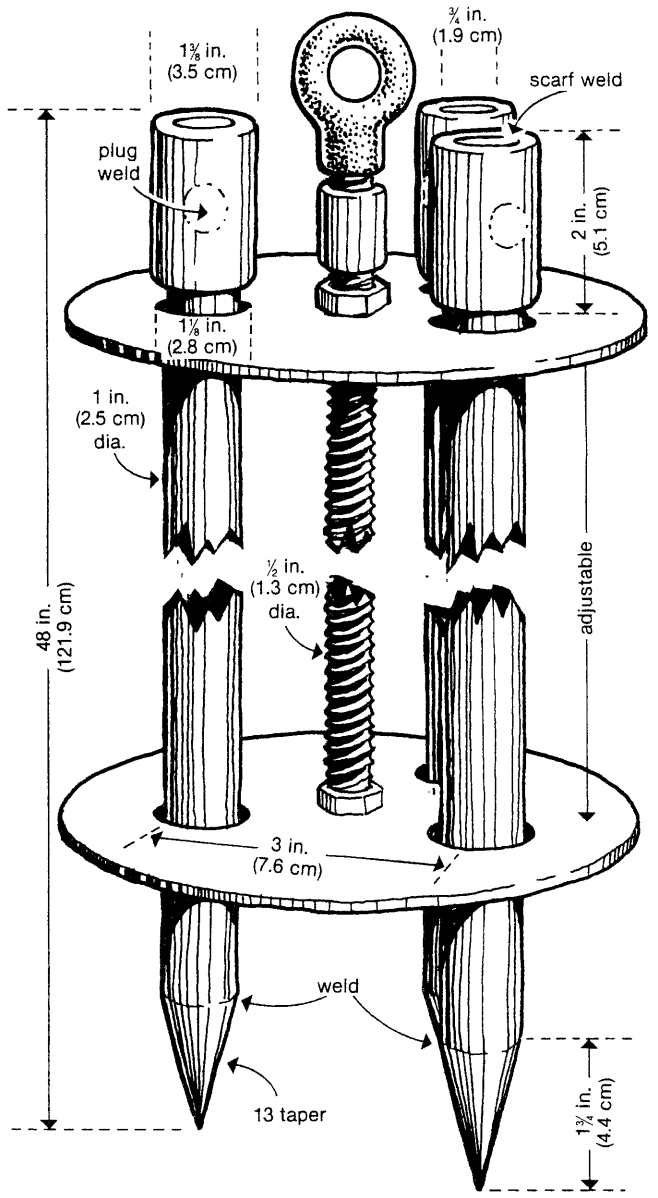


Figure 14. — Schematic diagram of tri-tube sampler.

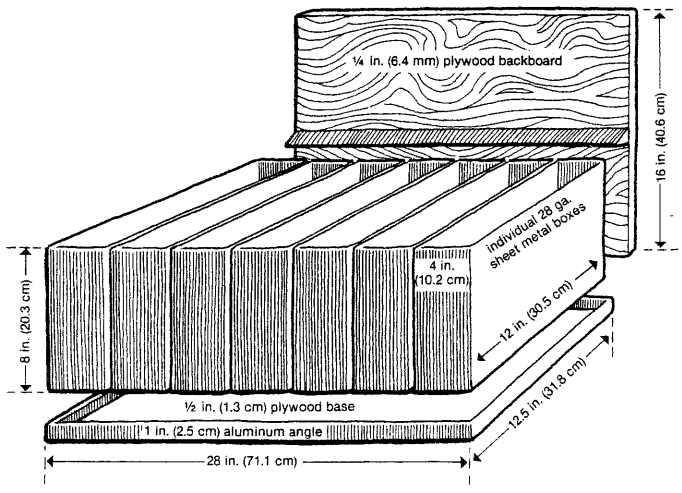


Figure 15. — Diagram of a freeze-core subsampler.

Sampling location and depth. — Selection of spawning sites by salmonids is a nonrandom activity. Adult salmonids selecting locations to spawn respond to such environmental variables as water depth and velocity, substrate composition, and proximity to cover. Because both sediment particle-size distribution and redd site selection are nonrandom events, the location from which samples are drawn to characterize spawning gravels should be identified by an experienced fishery biologist. Samples should only be drawn from locations that meet the known spawning requirements of a species. The suitability of each sampling site should be determined by quantitative measurements of water depth and velocity. The depth at which the sample is extracted is also critical to the analysis. Samples should be taken only as deep as the average depth of egg deposition for the species being studied. Since there is substantial stratification in stream gravels, sampling above or below the level of egg deposition might yield an inaccurate estimate of the size and distribution of particles within a redd. If prediction of survival to emergence of salmonid fry is desired, all samples should be collected from redds just prior to onset of emergence. Otherwise, temporal variations in gravel composition (Adams and Beschta 1980) might lead to inaccurate assessments of gravel quality at the onset of emergence.

Sample analysis. — Sediment samples can be analyzed either in the field or in the laboratory. The "wet method" can be done onsite and is the least expensive, but also the least accurate, method. The wet method usually uses a water-flushing technique with some hand shaking to sort sediments through a series of sieves. The trapped sediment on each sieve is allowed to drain and is poured into a water-filled graduated container. The amount of water displaced determines the volume of the sediment plus the volume of any water retained in pore spaces in the sediment. When the wet method is used, water retained in the sediment must be accounted for, since water retention per unit volume of fine sediments is higher than for coarse sediments. A conversion factor based on particle size and specific gravity can be used to convert wet volume to dry volume. Conversion factors for the normal range of particle sizes and specific gavities are listed in table 7.

For more exacting results, we recommend that the sediment samples be placed in containers and transported to the laboratory for analysis. Laboratory analysis of dry weights is the most accurate because all water in the sample can be evaporated, thus eliminating the need for conversion factors associated with the wet method. In the "laboratory method," the sediment sample is ovendried (24 hours at 221° F [105° C] or airdried, passed through a series of sieves, and that portion caught by each sieve is weighed. We recommend the Wentworth sieve series be adapted to the standard classification on table 4, this includes a progression of five size classes ranging from 0.002 inch to 3.94 inches (0.062 to 100 mm). The upper limit might seem arbitrary, but it approximates the largest size particles in which most salmonids will spawn. Consequently, few grains larger than 5 inches (128 mm) are present in preferred spawning areas. The sixth size class (10.1 to 20.2 inches [256 to 512 mm]) indicates the difficulty salmonids would have in moving the materials to deposit and cover their eggs.

Table 7. — Water gained in a wet sieving process and the factor for correcting volumetric data (Shirazi and Seim 1979)

Sieve size		Gram water gained gram dry gravel			Correction factor applied to wet sieved gravel		
		[1]$\rho = 2.2$	$\rho = 2.6$	$\rho = 2.9$	$\rho = 2.2$	$\rho = 2.6$	$\rho = 2.9$
Inches	*mm*						
3	76.2	0.02	0.01	0.01	0.97	0.96	0.96
	64	.02	.02	.01	.96	.96	.96
2	50.8	.02	.02	.02	.96	.96	.95
	32	.02	.02	.02	.95	.95	.94
1	25.4	.03	.02	.02	.94	.94	.94
	16	.03	.03	.03	.93	.93	.92
1/2	12.7	.04	.03	.03	.92	.92	.91
	8	.05	.04	.04	.91	.90	.89
1/4	6.35	.05	.05	.05	.89	.88	.88
	4	.07	.06	.06	.87	.86	.85
1/8	3.18	.08	.07	.07	.86	.85	.84
	2.0	.10	.09	.08	.83	.81	.81
1/16	1.59	.11	.10	.09	.81	.80	.79
	1.0	.13	.12	.12	.77	.76	.75
1/32	.79	.15	.14	.13	.75	.73	.72
	.50	.19	.18	.17	.70	.69	.67
1/64	.40	.21	.20	.19	.68	.66	.65
	.25	.27	.25	.23	.63	.61	.59
1/128	.20	.30	.28	.26	.60	.58	.57
	.125	.38	.35	.33	.54	.52	.51
1/512	.10	.43	.39	.37	.52	.50	.48
	.063	.54	.49	.47	.46	.44	.42

[1]ρ = gravel density.

Quality indexes. — The quality of gravels for salmonid reproduction has traditionally been estimated by determining the percentage of fine sediments (less than some specified diameter) in samples collected from spawning areas. The field data can be compared (Hall and Lantz 1969) to results of several laboratory studies (for example, Phillips and others 1975) to estimate survival to emergence of various species of salmonids. While an inverse relationship between percent fines and survival of salmonid fry has been demonstrated by several researchers, beginning with Harrison (1923), use of percent fines alone to estimate gravel quality has a major disadvantage; it ignores the textural composition of the remaining particles that can have a mitigating effect on survival. For example, imagine two samples each containing 20 percent fine sediment less than 1 mm diameter by weight, but the average diameter of larger particles is 10 mm in one sample and 25 mm in the other. Interstitial voids in the smaller diameter material would be more completely filled by a given quantity of fine sediment than voids in the larger material and the subsequent effect on survival of salmonid fry would be very different. Percent fines is a reasonable index to gravel quality, but has serious limitations because it ignores the textural composition of the remainder of the sample.

Other quality indexes have been developed recently in an attempt to improve upon the percent fines method. Platts and others (1979) used the geometric mean diameter (d_g) method for evaluating sediment effects on salmonid incubation success. This has advantages over the commonly used percent fines method in that it is a conventional statistical measure used by several disciplines to represent sediment composition; it relates to the permeability and porosity of channel sediments and to embryo survival as well or better than percent fines; and it is estimated from the total sediment composition. But despite these advantages, d_g has been shown by Beschta (in press) to be rather insensitive to charges in stream substrate composition caused by roading in a Washington watershed. Also, Lotspeich and Everest (1981) have shown that use of d_g alone can lead to erroneous conclusions concerning gravel quality. Because of these problems, Beschta (in press) has raised serious questions regarding the utility of the geometric mean diameter as a quality index.

Tappel (1981) offers another approach, which is a modification of the d_g method and uses a linear curve to depict particle size distribution by assigning the points 0.03 inch (0.8 mm) and 0.37 inch (9.5 mm) for determining a line. According to Tappel, the slope of this line gives a truer picture of fine sediment classes detrimental to incubation. A major drawback of this procedure, as with percent fines, is that it ignores the larger particles in a sample and consequently might suffer the same limitations.

A recent spawning substrate quality index that appears to overcome limitations of percent fines and geometric mean has been reported by Lotspeich and Everest (1981). Their procedure uses a measure of the central tendency of the distribution of sediment particle sizes in a sample and the dispersion of particles in relation to the central value to characterize the suitability of gravels for salmonid incubation and emergence. These two parameters are combined to derive a quality index called the "fredle index," which provides an indicator of sediment premeability and pore size. The measure of central tendency used is the geometric mean (d_g). Pore size is directly proportional to mean grain size and regulates intragravel water velocity and oxygen transport to incubating salmonid embryos and controls intragravel movement of alevins. These two substrate parameters are the primary legislators of salmonid embryo survival-to-emergence.

The fredle index (f) is calculated by the following method:

$$f = \frac{d_g}{S_o}$$

where:

$$d_g = (d_1^{w_1} \times d_2^{w_2} \times \ldots\ldots\ldots d_n^{w_n})$$

d_n = midpoint diameter of particles retained on the nth sieve

w_n = decimal fraction by weight of particles retained on the nth sieve

$$S_o = \frac{d_{75}}{d_{25}} = \text{sorting coefficient}$$

d_{75}, d_{25} = particle size diameters at which either 75 or 25 percent of the sample is finer on a weight basis.

Fredle numbers for sediment with a single grain size will be equal to the geometric mean because S_o is then 1. Sediments with the same d_g will have f numbers less than the geometric mean as S_o increases. The examples in figure 16 have a common d_g of 0.47 inch (12 mm) but yield fredle numbers of 12, 3.53, and 1.58, respectively. Sediments with small d_g values are less permeable than those with larger means because pores are small and intragravel flow and movement of alevins is impeded even through S_o might be 1. Also sediments with large d_g might be slowly permeable when S_o is large because pore spaces are occupied with smaller grains that impede interstitial flow and movement. Thus, the magnitude of the fredle index numbers is a measure of both pore size and relative permeability, both of which increase as the index number becomes larger.

The relationship between f values and survival-to-emergence of salmonid alevins has not been documented experimentally. The data of Phillips and others (1975), however, have been used to establish a preliminary relationship between these parameters. Phillips and others (1975) examined survival-to-emergence of coho salmon (*Oncorhynchus kisutch* [Walbaum]) and steelhead trout (*Salmo gairdneri* Richardson) embryos in gravel mixtures of known composition. Calculated fredle numbers for the mixtures of Phillips and others (1975) were plotted against survival (fig. 17). The preliminary relationship indicates that the fredle index is responsive to slight changes in gravel composition, survival, and variations in intragravel habitat requirements of individual species. For example, in Phillips and others (1975) artificial gravels with f of 2, 4, and 8, survival-to-emergence of 30, 60, and 88 percent, respectively, can be predicted for coho salmon, whereas survival of steelhead trout can be predicted at 45, 75, and 99 percent in the same mixtures. The difference between survival of coho salmon and steelhead trout at a given f is probably related to differences in the cranial diameter of alevins, which control their movement through pore spaces in gravel.

This method of calculating a quality index (f) for stream sediments allows biologists and land managers to identify the quality of gravel used for reproduction by anadromous salmonids. Also, comparisons can be made of gravel quality within and between streams, and temporal changes in texture and permeability can be monitored. The technique should be especially useful for measuring changes in gravel quality resulting from sedimentation from nonpoint sources in managed forest watersheds.

Channel Cross Section Surveys

Surveyed channel cross sections similar to those shown in figures 5 through 7 provide a permanent record of the channel

Top view

MIX 1		MIX 2		MIX 3	
GEOMETRIC MEAN	=12.00	GEOMETRIC MEAN	=12.00	GEOMETRIC MEAN	=12.00
SORTING COEFFICIENT	= 1.00	SORTING COEFFICIENT	= 3.40	SORTING COEFFICIENT	= 7.61
FREDLE INDEX	=12.00	FREDLE INDEX	= 3.53	FREDLE INDEX	= 1.58
% FINE SEDIMENT <0.04 INCHES DIAMETER	= 0	% FINE SEDIMENT <0.04 INCHES DIAMETER	= 15%	% FINE SEDIMENT <0.04 INCHES DIAMETER	= 30%
PREDICTED EMERGENCE OF COHO	= 98%	PREDICTED EMERGENCE OF COHO	= 51%	PREDICTED EMERGENCE OF COHO	= 22%

Side view

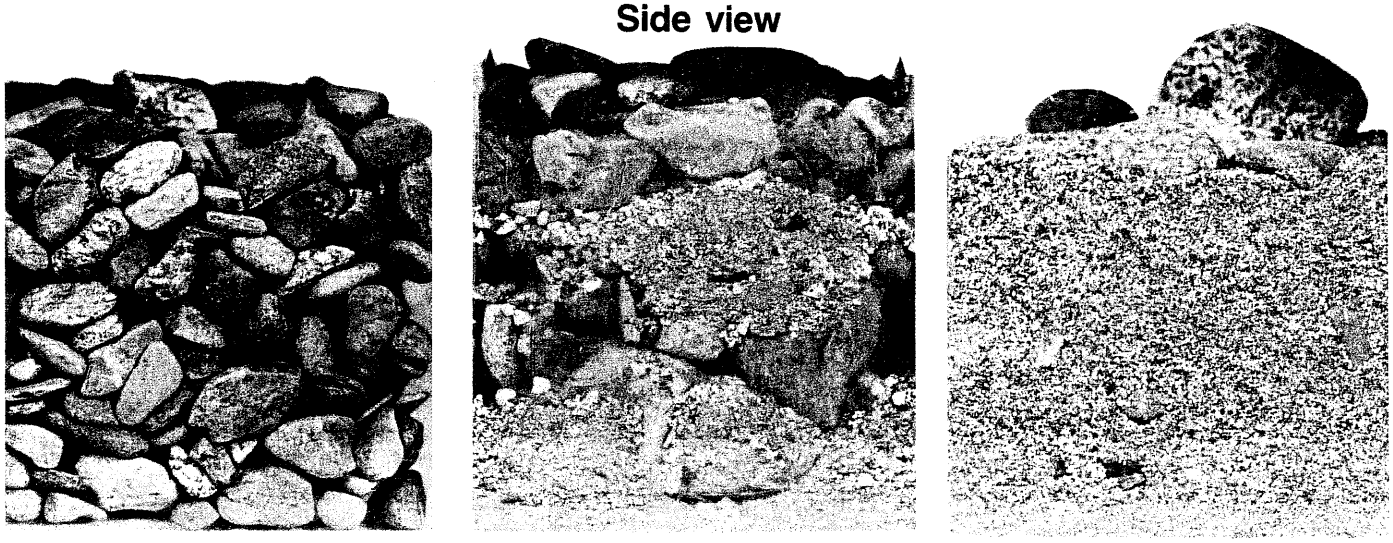


Figure 16. — Three gravel mixtures with a common geometric mean, but widely divergent distribution of particle sizes.

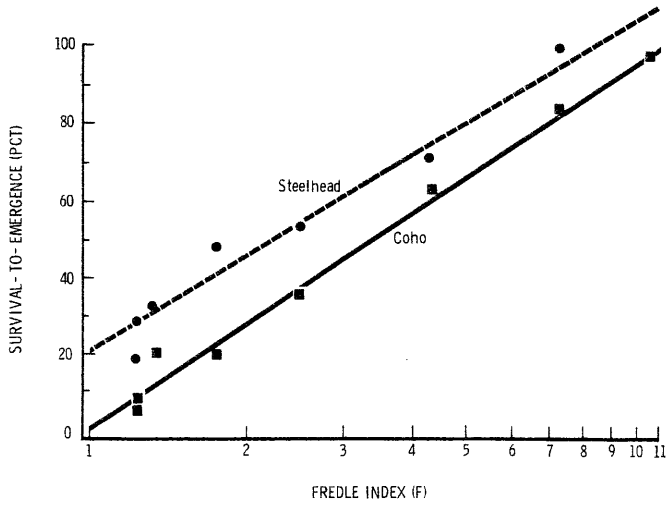


Figure 17. — Relationship between fredel index (f) numbers and survival-to-emergence of coho salmon and steelhead trout (semilog plot, lines fitted by eye; based on data of Phillips and others, 1975.)

morphology at any given point in time. Repeated surveys taken at the same locations over time can be used to evaluate time trends in channel bank and bed erosion and deposition. Plotted cross sections are also useful for estimating flow rates for water depths other than those found at the time flow is measured.

Surveys are conducted by stretching a measuring tape across the channel between permanent reference stakes located on the flood plain a safe distance of at least a few feet back from the top of the channel banks depending on bank erosion rates. The tops of the reference stakes, the ends of the tape, the bottom of the reference stakes and all obvious breaks in slope of the surface progressing across the tape are measured using a series of paired horizontal (actually taped distance) and vertical measurements. Care should be taken to measure all profile breaks because straight lines are assumed between measurement points when the data are plotted. Thus, any grade changes not measured will be erased in plotting. Usually, measurements are also made on the top and bottom of the reference stakes in order to simplify the comparison of successive cross sections made at the same location over time (fig. 18). We measure all horizontal and vertical distances to the nearest 0.1 and 0.01 ft (30.5 and 3.0 mm), respectively.

One common procedure for making vertical measurements is to use an engineer's level and level rod using conventional surveying techniques. This procedure requires at least two people, but is precise if done carefully. The channel cross section is plotted by referencing horizontal distances from the end of the tape (we use the right bank for convention) and vertical distance from a bench mark or the height of the instrument for each survey.

Figure 18. — Measuring from the top of the stake to the tension spring handle.

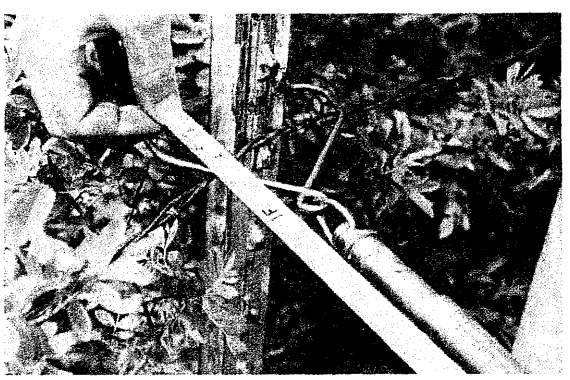


Figure 19. — Measuring from the fencepost to the end of the tension spring.

Another procedure for measuring vertical distances is to simply measure the distance from the tape to the ground surface using a measuring rod. This procedure utilizes the generalized sag tape procedure developed by Ray and Megahan (1979) and can be done by one person if necessary. This is the procedure that we use because it requires fewer person hours, is a little faster, and is precise if proper procedures are followed. Some additional data are required when the sag tape procedure is used including: (1) the difference in elevation between the two ends of the tape; (2) tension applied to the tape (fig. 19); and (3) the weight per foot of the tape.

The difference in elevation between the ends of the tape is determined with an engineer's level and level rod at the time of the first survey. The locations of the tape ends are marked on the reference stakes so that subsequent measurements for cross section surveys are not needed. Tape tension is measured at the time of each cross section survey using a small spring scale attached to the end of the tape. Tape weight is a constant for each tape and is determined by weighing the tape with all brackets or holding devices removed and dividing by the tape length.

Plotting the cross section using the sag tape procedure requires the solution of some ponderous equations. The job is simple, however, if programmed on a computer. The equations and a program flow chart for this generalized sag tape procedure were developed by Ray and Megahan (1979). A program is presently available for use at the USDA Forest Service Computer Center in Fort Collins, Colo., under the name R2-CROSS-81 (Weatherred and others 1981)

Sedimentation

Sedimentation is a broad term that encompasses two overlapping areas of interest: (1) sediment transport past a channel reference point; and (2) the deposition or erosion of material at a channel reference point. Both aspects of sedimentation are important to the aquatic community.

Sediment transport. — Sediment transport is a function of streamflow rate and the rate and size of sediment supply. Sediment transport usually increases logarithmically with streamflow. As a result, the increase in sediment transport for a given increment of flow is much higher for high streamflow rates than for low streamflow rates. Sediment moves either in suspension within the water column as suspended load or by bouncing or rolling along the bottom as bedload. Suspended sediment is most readily apparent to the casual observer and can be deleterious to fish if sediment concentrations are high enough for a long enough period. However, bedload may be more damaging because of loss of food supplies and spawning habitat and changes in channel morphology.

Evaluation of sediment transport goes beyond the scope of most aquatic environment studies and will not be discussed in detail here. This is primarily because determination of sediment transport must be made throughout the flow hydrograph. An isolated sediment transport measurement made during the low flow period required for most aquatic environment studies would be meaningless. Guidelines for collecting suspended sediment data are available in "Field Methods for Measurement of Fluvial Sediment" (Guy and Norman 1970) and by consultation with sedimentation specialists and hydrologists.

Techniques for bedload sediment measurement have not been standardized. The closest thing to a standard sampler is the Helley-Smith sampler (Helley and Smith 1971). Subsequent calibration by Emmett (1979) shows that the sampler has merit for most applications.

Laboratory analysis of sediment samples has also been standardized. Guy (1973) describes laboratory methods for analysis of both suspended and bedload samples for concentration, particle size distribution, and other properties of concern, such as percent organic matter.

Erosion and deposition. — Studies of erosion and deposition of materials at a given channel point are more relevant for aquatic environment studies than measurements of sediment transport past a given point. Evaluation of erosion or deposition of bottom materials requires successive (usually annual) measurements to be taken at low flow periods. Such measurements are easily included in aquatic habitat studies. Erosion or deposition are documented by changes in the evaluation of the bottom and in the particle size distribution of bottom materials. A number of techniques are available including surveyed cross sections, painted rocks, buried chains, streambed surface particle size evaluation, and particle size analysis of streambed cores. Megahan and others (1980) report on a study using many of these techniques to evaluate trends in channel conditions in the South Fork of the Salmon River over a 15-year period.

The best method for quantifying the volume of channel erosion and deposition is with the use of successive channel cross sections. A comparison of cross sections using the same data illustrates the amount and location of bed elevation changes. Sometimes no changes in bottom elevation are detected by successive cross sections taken during low flow periods, even though considerable erosion or deposition occurs during high flows. Channel cross sections taken at high and low flow levels are needed in such situations. However, frequent cross section surveys can be impractical and oftentimes downright dangerous, especially during high flows.

Painted rocks can be used to evaluate the amount of disturbance of surface bed materials during high flows. Various sized sediment particles are removed from the surface of the channel bottom and painted a brilliant color and then replaced at known locations on the streambed. Placement must be done carefully so that the painted rocks are fitted into the streambed surface similar to the undisturbed bed particles. A susequent comparison of the location of painted sediment particles provides an indication of the size of bedload particles moved during the intervening high flow period.

Painted rocks give an indication of the size of materials moved on the streambed surface but do not show the depth of erosion and subsequent deposition. The method of buried chains provides a means of doing this. A driving ring is attached to the end of a small gage chain. The ring is placed over the pointed end of a metal driving rod and the rod and chain are driven vertically to the desired depth. By twisting and tamping the driving rod during removal, the bed sediments are packed around the chain, leaving it suspended vertically in the bottom sediments. When bed scour occurs, the free upper end of the chain collapses or is bent from the vertical and swept downstream. Subsequent fill is deposited on top of the horizontal segment of chain. After high flow, the chain is relocated by survey and dug out. The position of the bend indicates the depth of scour and redeposition.

In some situations, the amount of scour and fill on the stream bottom is not as critical as is the change in particle size distribution of the bottom. This is especially true in salmon and steelhead spawning areas where increases in the percentages of fine sediment can severely reduce fry survival (Bjornn 1973; Phillips and others 1975).

Channel Debris and Sediment Storage

Debris in streams is often considered harmful because log jams can create physical blocks to migrating fish. In addition, excessive inputs of small debris, such as leaves and small branches, can reduce oxygen levels in the water under certain conditions (Narver 1971). However, it is well to bear in mind that organic debris, consisting of logs, branches, and leaves, is a natural and necessary component of forest aquatic ecosystems. The food base for the biological community of forest streams is mostly woody debris and leaves. Wood in streams also serves as a substrate for biological activity and creates other habitat niches by regulating the movement of water and sediment.

Debris loading can be influenced by forest management activities; loading may increase if logging debris is added to the channel or it may decrease if channel clearing takes place. Vegetation removal in the immediate vicinity of the channel also reduces debris loading in the long run by reducing the inflow of debris from natural mortality.

There has been increasing research in recent years to evaluate the role of debris in channels, including methods to inventory debris and its effects on channel sediment storage. Froelich (1973) describes a method for quantifying the volume of debris storage in channels. Swanson and Lienkaemper (1979) developed techniques to study the frequency of occurrence of wood and wood-created habitat in undisturbed forest streams in Oregon. They found that wood or wood-created habitat comprised 50 percent of the total stream area on first-order streams and 25 percent of the total stream area on third-order streams. Our research has been devoted to evaluating the volume of sediment storage behind channel obstructions because of biological implications and the need to develop monitoring techniques for accelerated sediment production from forest management activities. Megahan and Nowlin (1976) showed that, on the average, over 10 times more sediment was stored behind debris in seven study channels than was deposited in sediment basins at the mouth of the streams each year.

We use a sampling system to inventory sediment accumulations behind natural channel obstructions, including woody debris on headwater streams. Sample reaches 140 ft (42.7 m) in length are located at 360-ft (109.7-m) intervals starting at the mouth of the drainage and progressing upstream along the dominant channel until the point is reached where there are no obvious indications of flow. Obstructions are defined as any material in the channel causing sediment accumulations because of discontinuities in channel gradient and include: logs (more than 4 inches [10 cm] in diameter), rocks, roots, stumps; and other debris (includes branches, twigs, and leaves).

Sampling is restricted to obstructions causing sediment accumulations with the following minimum dimensions: height 0.66 ft (0.2 m); averge width 0.98 ft (0.3 m); and length 1.97 ft (0.6 m). Eliminating the smaller obstructions greatly reduces the work and causes a loss of only about 10 percent of the total volume of stored sediment. Height (H) is defined as the difference between a stadia rod reading taken on the bed at the downstream side of the obstruction (the rod is raised if necessary to correct for any scouring at this point) and a rod reading taken on the sediment deposit immediately upstream from the obstruction. Rod readings are taken to the nearest 0.01 ft (0.4 cm) using an abney level. Length (L) is the distance from the upstream end of the obstruction to the upstream end of the accumulated sediment. Width (W) of the sediment accumulation is the average of three widths taken normal to the length at distances of 0.16, 0.5, and 0.83 of the length from the obstruction. The upstream end and edges of sediment accumulations are defined by breaks in channel gradient, differences in the particle size distribution of bottom sediments, and differences in composition of bottom materials.

Total volume (V) of sediment stored behind the obstruction is calculated assuming a triangular wedge of sediment as:

$$V = \frac{H}{2} LW.$$

A third rod reading is taken at the upstream end of the obstruction to allow calculation of the slope of the accumulated sediments. The most apparent cause of the obstruction is defined by type as logs over 25 inches (63.5 cm) in diameter, rocks, roots, stumps, and organic debris (the last includes branches less than 25 inches diameter, twigs and leaves).

Stream Order

Stream order is defined by Horton (1945) and Langbein and Iseri (1960) by means of a method of numbering streams as part of a drainage basin network. Tributaries that have no branches are designated first-order streams; those that receive only first-order streams are second-order streams; larger branches that receive only first-order and second-order tributaries are designated third-order streams, and so on. Stream order provides a useful indicator of the physical and biological characteristics of streams (Lotrich 1973; Whiteside and McNatt 1972; Platts 1979).

We recommend that for stream order to provide high utility for interpretations, the first-order channels should be identified by direct inspection. In lieu of this, first-order streams are defined as the first channel formed in the headwaters that can be identified on USGS 7½-minute quadrangle maps. The largest available USGS map scale should be used if 7½-minute maps are not available for the area in question. Care should be used when comparing stream order in different geologic settings. The use of stream order, especially for planning purposes, can help compensate for lack of money or manpower by providing general information on fish species present, fish standing crops, stream width, stream depth, and channel substate composition (Platts 1979). However, we do not recommend using stream order alone if high resolution is needed.

RIPARIAN ZONE

The riparian ecosystem includes the streambank and the flood plain and is defined for this report as the vegetation portion of the streamside environment. Many land uses effect this part of the stream habitat. Riparian vegetation helps stabilize the streambanks, provides cover and food for fish, and intercepts solar radiation.

Streamside Cover

This rating considers all material (organic and inorganic) on or above the streambank that offers streambank protection from erosion and stream shading, and provides escape cover or resting security for fish:

Rating	Description
4	The dominant vegetation is shrub.
3	The dominant vegetation influencing the streamside and/or water environment is of tree form.
2	The dominant vegetation is grass or forbs.
1	Over 50 percent of the streambank transect line intercept has no vegetation and the dominant material is soil, rock, bridge materials, road materials, culverts, and mine tailings.

The area of streambank rated is that intercepted by the transect line that covers the exposed stream bottom, bank, and top of bank.

Initially in determining this rating, all vegetation along the stream that would reach the stream (if it were laid down towards the stream) was used in the analysis. This procedure caused high observer variation and increased confidence intervals. Therefore, we revised it to include only that cover intercepted by the transect line. This decreased the observer error and confidence intervals. The higher level offsite vegetation not considered must therefore be accounted for with some type of canopy rating.

In some rating systems (Forest Service Region 4 Methodology) used by fishery biologists, tree cover is given a higher environmental rating than shrubs. We found that streams bordered by brush had a higher fish standing crop than similar sized streams with tree borders (Platts 1974). Therefore, this manual rates brush cover higher than tree cover.

The cover rating is effective in evaluating the effects of such activities as channelization, logging, or cattle grazing on riparian habitat. This measurement in our studies had low confidence intervals about the mean (±4.1 percent) mainly because dominant cover tends to be uniform and observers evaluate the same conditions alike even though they may not rate it correctly. Year-to-year precision and accuracy were poor and demonstrate that problems can occur using this measurement.

Vegetation Use by Animals

Vegetation use under the transect line within 5 ft of the shoreline is rated visually. This evaluation considers vegetation disturbed during the present growing season and potential plant growth that does not exist because of past disturbance. An example of loss because of past use would be in areas where vegetation no longer exists because the streambank was dredged, trampled, or eliminated by a major cattle crossing. The rating, however, applies mainly to recent vegetation use. If use is determined on only one occasion or only one time a year, it should be done as soon as possible after the land use effect and before plant regrowth can occur.

The vegetation use rating is stratified into four classes:

Rating (percent)	Description
0 to 25 (Light)	Vegetation use is very light or none at all. Almost all the potential plant biomass at present stage of development remains. The vegetative cover is very close to that which would occur naturally without use. If bare areas exist, (i.e., bedrock) they are not because of loss of vegetation from past grazing use.
26 to 50 (Moderate)	Vegetative use is moderate and at least one-half of the potential plant biomass remains. Average plant stubble height is greater than half of its potential height at its present stage of development. Plant biomass no longer on site because of past grazing is considered as vegetation that has been used.
51 to 75 (High)	Vegetative use is high and less than half of the potential plant biomass remains. Plant stubble height averages over 2 inches. Plant biomass no longer on site because of past grazing is considered as vegetation that has been used.

76 to 100 (Very high)	Use of the streamside vegetation is very high. Vegetation has been removed to 2 inches or less in average stubble height. Almost all of the potential vegetative biomass has been used. Only the root system and part of the stem remain. That potential plant biomass that is now non-existent because of past elimination by grazing is considered as vegetation that has been used.

Once the observer has decided the class, then the actual percentage use is determined. For example, if the vegetation (grasses and forbs) has been reduced to less than 2 inches (50.8 mm) stubble standing height, the class rating is between 76 and 100 percent. If the vegetation is almost to ground level, the final intraclass rating would be 100 percent. If the vegetation is slightly less than 2 inches (50.8 mm) stubble height and there are no areas without vegetation from past livestock use, then the intraclass rating would be about 76 percent.

In our studies, the 95 percent confidence intervals about the means (±12 percent) are high, but still within acceptable limits for most streams studied. Precision and accuracy are good. The observer should be well trained and have ungrazed plots for constant comparison. Our visual estimates of vegetation use were quite close to use estimates gained with actual measurements with the Neal herbage meter (table 8).

Table 8. — Comparison of streamside herbage use using the Neal herbage meter versus the visual method

	1979			1980		
Study area	**Meter**	**Visual**	**Δ%**	**Meter**	**Visual**	**Δ%**
Idaho (10 streams)	45	44	1	58	60	2
Nevada (2 streams)	81	68	13	63	57	6
Utah (1 stream)	84	76	8	104	87	17

Herbage Production and Utilization

Herbage production and utilization were measured in a nondestructive method using a Neal Model 18-2000 electronic capacitance meter that measures the conductivity of materials within its field. The measurement generated by the meter is a unitless number that is linearly related to the mass of the measured material. As a result, clipped vegetation weights for selected plots can be graphed against their respective meter readings to generate a regression equation and curve from which further weights can be estimated directly from meter readings without the need to weigh each sample. To plot the regression line, at least 12 plots similar to the vegetation being sampled must be clipped and weighed. The regression equation also can be used to determine vegetative production for the study area, and a comparison of grazed and ungrazed sites provides a vegetation use estimate by simple mathematical manipulation as follows:

.1. 1 g per 2 ft² = 48 lb/acre

where meter reading estimates grams per 2 ft².

2. Production in the ungrazed area in pounds per acre is:

$$P_u = 48 \left[\frac{\sum_{u=1}^{n} (a + bx_u)}{n} \right]$$

where:

P_u = production in the ungrazed pasture

and

x_u = meter readings in the ungrazed pasture

a = y intercept

b = regression coefficient

n = number of primary sample plots

3. Production in the grazed area in pounds per acre is:

$$P_g = 48 \left[\frac{\sum_{g=1}^{n} (a + bx_g)}{n} \right]$$

where:

P_g = production in the grazed pasture in pounds per acre

and

x_g = meter readings in the grazed pasture.

4. Percent vegetative use $= \left[1 - \left(\frac{P_u}{P_g} \right) \right] (100).$

A brief description of operating procedures and field methodology is found in Neal and others (1976). Herbage meter measurements should be taken on ungrazed and grazed areas concurrently and immediately after the grazing season. We found the meter to be very accurate with regression curves that had R^2 values consistently greater than 0.85.

Vegetation Overhang

Vegetation overhang indirectly provides fish food and cover and shades the water from solar radiation (fig. 20). Overhang is a valuable variable to use in evaluating those land use effects such as logging and road construction that could alter the riparian habitat. Streamside cover rates all vegetation. Vegetation overhang rates only that vegetation overhanging the water column.

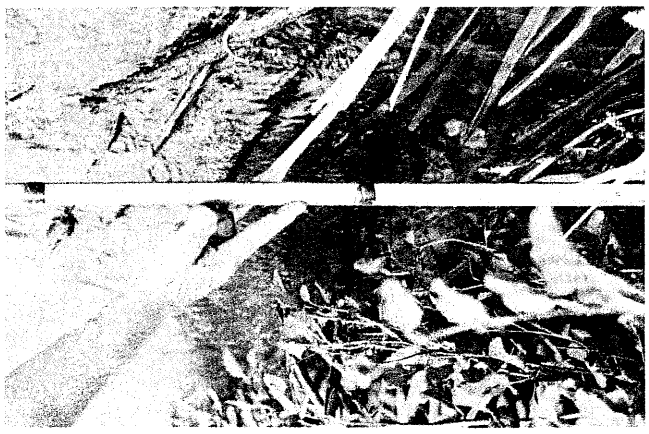


Figure 20. — Measuring overhanging streamside vegetation.

This is a direct measurement to the nearest 0.1 ft (0.03 m) of the vegetation (excluding tree trunks or downed logs) within 12 inches (304.8 mm) of the water surface and overhanging the water column (fig. 21).

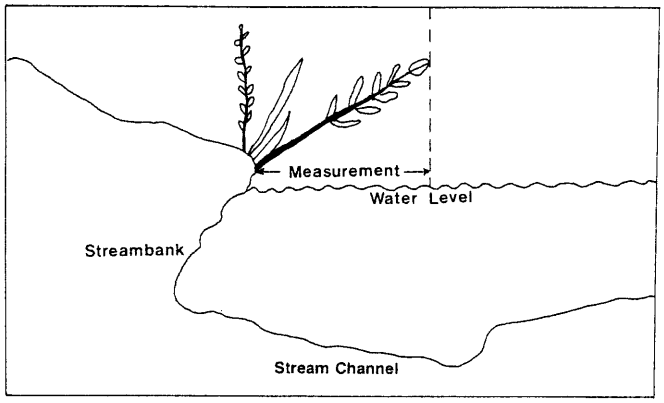


Figure 21. — Measurement of overhanging vegetation.

The measurement is taken along the transect line, beginning at the farthest protrusion of the streambank over the water surface, to the farthest point that vegetation covers the water column. This measurement does not include the undercut; therefore, the two measurements combined, give the total overhead cover.

In our studies the 95 percent confidence intervals around the means (±15.7 percent) are fairly wide, but year-to-year precision and accuracy rate fair.

Habitat Type

The streamside environment consists of many types of habitats and it is often theorized that the type and diversity can determine fish productivity. The habitat type evaluates only the channel-streambank area intercepted by the transect within 10 ft (3.05 m) of the stream shore. This rating is an evaluation of the dominant and subdominant material (organic or inorganic) composing the surface or overstory of the streamside environment. Combinations of plant and soil usually make up this environment, but occasionally a single material, such as sand or log, will describe the habitat type.

This rating was designed with the assumption that streamside environments composed of fine sediments (sand) have less value to fish than brush-sod environments, which have the highest value. All other habitat types fall between these two extremes (table 9). All existing cover is considered, but only the dominant and subdominant materials are selected for the final classification. The subdominant type would be the second most abundant material.

The year-to-year precision was excellent, but the accuracy ratings were only fair. Confidence intervals around the means in our studies (±4.9 percent) are low so the attribute has some possibilities. Over time, this measurement will determine changes in vegetation species as well as habitat type (such as a streamside environment that changes from brush-sod to fine-grass under an improper grazing situation).

FISH POPULATION EVALUATION

Fish populations are a result of the physical, biological, and chemical factors surrounding them and especially those biotic factors in the trophic levels below them. To sample all the trophic levels is not only expensive but often impossible; however, because fish are dependent on these lower levels, much understanding of ecosystem functioning can be gained from the fish themselves. The environmental tolerances and competitive interactions of fish are generally quite well known. The size, structure, and growth rates of the population allow determination of aquatic habitat conditions that existed in the past 2 to 10 years. Because year class strength is usually set during the early life-history stages, it allows us to follow several years of known conditions to determine reactions of fish to these conditions. Also, the results of the analysis can be related directly to the congressional mandate (Water Quality Act of 1972) of "fishable waters."

Sampling of fish populations must be done accurately because freshwater fish are notorious for wide fluctuations in year-class strengths. Use of electrofishing, explosive primacord, spot explosive concussion, toxicants, nets, scuba or snorkle, and redd counts are common field techniques to obtain data with which to estimate fish population numbers and biomass, fish species composition, and fish health and survival. Each technique has advantages and disadvantages that must be considered in the final selection of the method chosen to obtain the data.

Electrofishing

Electrofishing is an efficient capture method that can be used to obtain reliable population estimates, length-weight relationships, and age and growth on most streams of order 6 or less. Electrofishers tend to collect larger fish more easily than smaller fish, but the newer electrical transformers now available allow adjustable control of voltage, pulse, and electrical frequency thereby reducing size selectivity. Electrofishing efficiency can also be affected by stream conductivity, temperature, depth, and clarity of water. Each condition must be considered to ensure a reliable population estimate. Electrofishing can be more efficient than other methods of population estimates, such as seining and underwater observation. Boulder-rubble substrate, turbidity, aquatic vegetation, and undercut banks can bias other population estimation methods.

Using the newer electrofishers and successive removal-depletion techniques, we adequately sampled fish in streams up to stream order 5, even in infertile water (less than 35 mg/liter total dissolved solids). The removal-depletion method of population analysis (Zippin 1958) assumes that:

1. No animal can move in or out of the sample area;
2. Each animal has an equal chance of being captured;
3. The probability of capture is constant over all removal occasions.

These assumptions can be approached on small streams of order 5 or less if (1) pulse, frequency, and voltage are applied to reduce selectivity; (2) the sample area has fish passage blocks to keep fish from leaving the area; (3) a consistent proportion of the population is captured during each electrofishing pass; and (4) timing devices on the electrofishers are used to make sure capture effort is the same on all removals.

During electrofishing fish tend to swim or drift downstream; so it is imperative that the downstream blocking net be in place. Sometimes the upstream end of the sample area can be located at a fish passage restriction area. If this restriction is not available, then another blocking net is needed. We found that small salmonids less than 6 inches (152.4 mm) in length seldom tried to leave the area, but large salmonids would attempt to escape. A constant capture probability was difficult to obtain when sampling sculpin populations because of their tendency to remain in the substrate.

Two-Step Method

During 1975 and 1976, we used the two-step removal method (Seber and LeCren 1967) because it required only two passes with the electrofisher. Population estimates are easily derived with the simple formula:

Table 9. — Streamside habitat type rating

Rating	Streambank material		Rating	Streambank material	
	Dominant	Subdominant		Dominant	Subdominant
1		[1]All fines	13	Boulder	Root
2	Fines	Gravel	13	Boulder	Tree
2	Fines	Grass	13	Boulder	Sod
2	Fines	Rubble	13	Boulder	Brush
3	Fines	Boulder	12	Root	Fines
3	Fines	[2]Root	13	Root	Gravel
3	Fines	[3]Tree	12	Root	Grass
3	Fines	[4]Sod	13	Root	Rubble
3	Fines	Brush	13	Root	Boulder
4	Gravel	Fines	13		All root
5		All gravel	14	Root	Tree
6	Gravel	Grass	13	Root	Sod
6	Gravel	Rubble	14	Root	Brush
7	Gravel	Boulder	12	Tree	Fines
8	Gravel	Root	13	Tree	Gravel
8	Gravel	Tree	13	Tree	Grass
7	Gravel	Sod	13	Tree	Rubble
8	Gravel	Brush	13	Tree	Boulder
8	Grass	Fines	14	Tree	Root
9	Grass	Gravel	14		All tree
9		All grass	14	Tree	Sod
9	Grass	Rubble	14	Tree	Brush
9	Grass	Boulder	12	Sod	Fines
11	Grass	Root	13	Sod	Gravel
12	Grass	Tree	14	Sod	Grass
13	Grass	Sod	15	Sod	Rubble
17	Grass	Brush	16	Sod	Boulder
8	Rubble	Fines	18	Sod	Root
9	Rubble	Gravel	18	Sod	Tree
9	Rubble	Grass	17		All sod
10		All rubble	19	Sod	Brush
10	Rubble	Boulder	17	Brush	Fines
11	Rubble	Root	20	Brush	Gravel
11	Rubble	Tree	20	Brush	Grass
11	Rubble	Sod	21	Brush	Rubble
12	Rubble	Brush	22	Brush	Boulder
11	Boulder	Fines	23	Brush	Root
12	Boulder	Gravel	23	Brush	Tree
12	Boulder	Grass	24	Brush	Sod
12	Boulder	Rubble	23		All brush
12		All boulder			

[1]Fines include sands, silts, clays, and organic fine particle materials.
[2]Includes only roots from brush and trees.
[3]Downfall logs included.
[4]Sod has an extensive root mass and is more stable than grass or grass tufts.

$$\hat{N} = \frac{(U_1)^2}{(U_1 + U_2)} \quad (1)$$

where:

$\hat{N}$ = the fish population estimate

U_1 = the number of fish collected in first removal

U_2 = the number of fish collected in second removal.

The standard error of the estimate can be calculated using:

$$SE(\hat{N}) = \sqrt{\frac{(U_1)^2 \times (U_2)^2 \times T}{(U_1 - U_2)^4}} \quad (2)$$

where:

$SE(\hat{N})$ = standard error of the population estimate

T = the total number of fish collected $(U_1 + U_2)$.

To illustrate, assume that 400 fish were collected in the first removal and 350 in the second. The population estimate is:

$$\hat{N} = \frac{(400)^2}{(400-350)} = 3,200$$

and the standard error is:

$$SE(\hat{N}) = \sqrt{\frac{(400)^2 \times (350)^2 \times 750}{(50)^4}} = 1,533.62.$$

In this example, almost as many fish were collected in the second removal as in the first. The two-step method may not give estimates with narrow enough confidence intervals to determine whether fish standing crops were actually changing over time. Other depletion models are available that allow for two or more removals and provide better population estimates with narrower confidence intervals (table 10).

Zippin Method

From 1977 to 1981, we used two analyses with the multiple-step removal-depletion method: the Zippin 1958 method, based on Moran's (1951) work, and Burnham's maximum likelihood. After experimenting with two-, three-, four-, five-, and six-step removals, we felt, when time and money are considered, the four-step method is the most efficient. Using the Zippin approach with four removals, we narrowed the confidence intervals around the population estimate, and we could begin to determine whether small changes in the fish population over time were significant. The computer program (FPSP-AI) for calculating population estimates using this likelihood method is given in its entirety in appendix 6. The Zippin method is based on a maximum likelihood model (Moran 1951) which has the probabilities reduced to easily used graphs.

The first quantity required is:

$$T = \sum_{i=1}^{k} U_i \quad (T = U_1 + U_2 + \ldots + U_k) \qquad (3)$$

where:

T = total number of fish collected

U_i = number of fish collected in the ith removal

k = the number of removals.

In the previous example, 400 fish were removed in step 1 and 350 fish in step 2. Using the Zippin method with four passes (k=4), assume 100 fish were removed in step 3 and 50 fish in step 4. Then:

$$T = 400 + 350 + 100 + 50 = 900.$$

Next the ratio (R) must be determined from the following formula:

$$R = \frac{\sum_{i=1}^{k} (i-1)\, U_i}{T} = \frac{(1\text{-}1)U_1 + (2\text{-}1)U_2 + \ldots + (k\text{-}1)U_k}{T} \qquad (4)$$

In our example:

$$R = \frac{U_2 + 2U_3 + 3U_4}{T}$$

$$R = \frac{350 + 200 + 150}{900} = 0.78 .$$

Figure 22 must be used to find the proportion $(\hat{Q})^4$ of fish captured during all removals that correspond to the value for R.

The population estimate is then determined by:

$$N = \frac{T}{\hat{Q}}. \qquad (5)$$

where:

$\hat{Q}$ = the proportion of the fish captured during all removals and is determined from figure 22. The ratio R = (0.78) is used to find the point on the curve that corresponds to the $\hat{Q}$ value. In this case, $\hat{Q} = 0.92$.

Therefore in our example:

$$\hat{N} = \frac{T}{\hat{Q}} = \frac{900}{0.92} = 978 \text{ fish.}$$

[4]This proportion is deonated $(1\text{-}\hat{q}^k)$ in Zippin (1958) and its mathematical derivation is described in that publication.

Table 10. — An example of 95 percent population confidence intervals achieved with the two-step and multiple-step methods in the same stream reach on the South Fork Salmon River on a bull trout (*Salvelinus confluentus* Suckley) population

Year	Population estimate	Standard error	±95 percent confidence interval	Method
1975	405	31.1	61	two-step
1976	271	85.9	168	two-step
1977	808	8.4	17	four-step
1978	323	4.4	9	four-step
1979	1,511	17.2	34	four-step
1980	682	13.7	27	four-step
1981	386	11.9	23	four-step

The accuracy of a population estimate is largely determined by how closely the underlying assumptions of the removal-depletion method were followed. To measure the reliability of the population estimate, it may be useful to calculate confidence intervals. Confidence intervals enable one to state with given probability the population estimate within a certain range. Assuming that we have a normal frequency distribution, the chance that the true population differs from the population estimate by more than 1.96 standard errors above and below the population estimate is less than 1 in 20. For our work, we assume a normal frequency distribution, which may not be the case with small sample sizes.

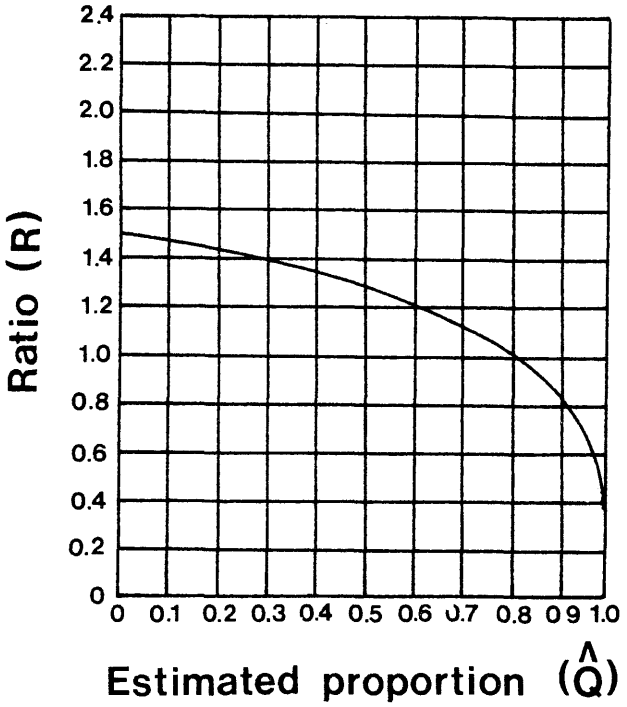


Figure 22. — Graph used to determine the estimated proportion ($\hat{Q}$) of fish captured during all removals from the ratio (R) of the sum of the products of the number of fish captured during each successive removal and the number of the preceeding removal to the total number of fish collected.

The formula for the standard error using the Zippin method is:

$$SE(\hat{N}) = \sqrt{\frac{\hat{N}(\hat{N}-T)T}{T^2-\hat{N}(\hat{N}-T)\frac{(k\hat{P})^2}{1-\hat{P}}}} \qquad (6)$$

where:

$\hat{P}$ = the estimated probability of capture during a single removal and is obtained from the graph in figure 23.

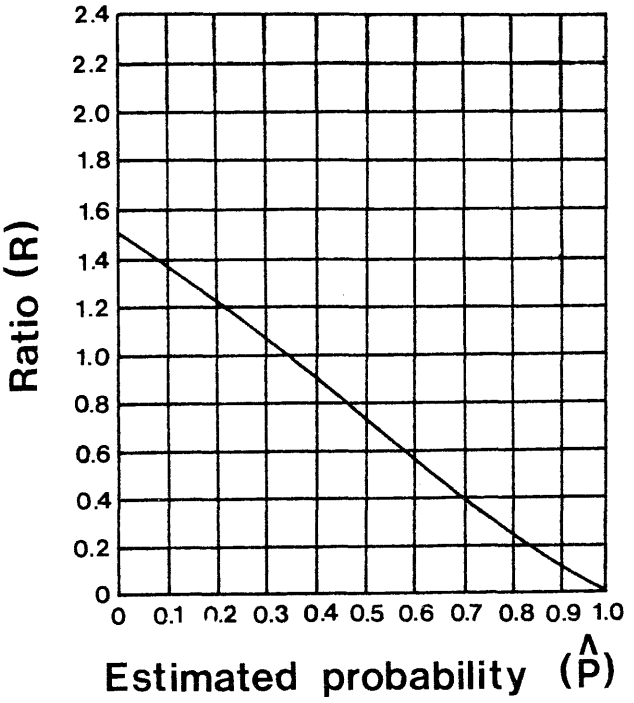


Figure 23. — Graph used to determine the estimated probability ($\hat{P}$) of capture during a single removal from the ratio (R) of the sum of the products of the number of fish captured during each successive removal and the number of the preceeding removal to the total number of fish collected.

The standard error from our example using four removals is:

$$SE(\hat{N}) = \sqrt{\frac{(978)\,(978-900)\,(900)}{(900)^2-(978)\,(978-900)\left[\frac{(4\times 0.47)^2}{(1-0.47)}\right]}}$$

$$= \sqrt{227.87} = 15.10.$$

The confidence interval is calculated by taking the population estimate plus and minus 1.96 times the standard error of the population estimate. The population estimate 978 $\pm$ 1.96 $\times$ 15.90 equals 978 $\pm$ 29.6, which equals 948 and 1,008. The third and fourth removals helped narrow the 95 percent confidence interval from 750 to 6,267 using the Seber-LeCren two-step method to 948 and 1,008 using the Zippin method with four removals. Generally speaking, the higher the number of removals, the narrower the confidence interval.

A chi-square test (χ^2) can be used to determine the goodness of fit between our actual removal pattern with its varying capture probabilities and a theoretical removal pattern that assumes a constant capture probability. The test tells us how closely we came to meeting the constant capture probability assumption.

The χ^2 test can be applied as follows:

$$\chi^2 = \sum_{i=1}^{k} \left[\frac{[U_i - \hat{E}(U_i)]^2}{\hat{E}(U)_i)} \right] \quad (7)$$

$$= \left[\frac{[U_1 - \hat{E}(U)_i)]^2}{\hat{E}(U_1)} + \frac{[U_2 - \hat{E}(U_2)]^2}{\hat{E}(U_2)} + \ldots + \frac{[U_k - \hat{E}(U_k)]^2}{\hat{E}(U_k)} \right]$$

where:

$\hat{E}(U_1)$ = the expected number of fish for the ith removal based on $\hat{P}$ (fig. 23) and the population estimate = $\hat{N}(1-\hat{P})^{i-1}(\hat{P})$

$\hat{P}$ = the estimated probability of capture during a single removal.

In our example:

$\hat{P} = 0.47$

$\hat{E}(U_1) = 978(1)(0.47) = 460$

$\hat{E}(U_2) = 978(0.53)(0.47) = 244$

$\hat{E}(U_3) = 978(0.53)^2(0.47) = 129$

$\hat{E}(U_4) = 978(0.53)^3(0.47) = 68.$

Therefore:

$$\chi^2 = \frac{(400-460)^2}{460} + \frac{(350-244)^2}{350} + \frac{(100-129)^2}{129} + \frac{(50-68)^2}{68}$$

$= 51.21$ with k-2 = 2 d.f.

The calculated χ^2 is compared to respective χ^2 table entry indicates that the removal pattern (400, 350, 100, and 50) did not give us a high goodness of fit, suggesting that a constant capture success was not achieved. On actual electrofishing, however, we have found that our field data allowed an adequate goodness of fit.

Maximum Likelihood Model

Computer estimation of fish population sizes is accomplished with a maximum likelihood model that was developed with the assistance of Dr. Ken Burnham from the U.S. Fish and Wildlife Service's Western Energy Land Use team. This model uses the successive depletion of catch sizes to estimate the actual population size by determining the likelihood of possible population sizes greater than or equal to the total catch. The population size with the highest likelihood is considered the best estimate of the actual population size.

The first quantities to be determined are the total catch (T), which is a summation of the number of fish caught in each of k removals (U):

$$T = \sum_{i=1}^{k} U_i = U_1 + U_2 + \ldots + U_k \quad (8)$$

and a function of the removals called C:

$$C = \sum_{i=1}^{k} iU_i = U_1 + 2U_2 + \ldots + kU_k. \quad (9)$$

These two values are then used to calculate the likelihoods of the possible population sizes ($\hat{N}_b$):

$$\hat{N}_b = T + b \quad (10)$$

where b is any arbitrary integer.

To determine $\hat{N}_b$ (the population estimate), we need to calculate the value of $\hat{N}_b$ with the greatest likelihood of occurring. This is accomplished by searching for the value of $\hat{N}_b$ associated with the highest probability. To do this, we define the likelihood function of b, called $\emptyset(b)$, which is essentially the natural logarithm of the population estimate probability when $\hat{N}_b = T+b$. We look at actual probabilities, but they are extremely small (between 0 and 1). It is more convenient to work with natural log probabilities.

$$\text{Let } h(b) = \sum_{b=1}^{j} \ln\left(1 + \frac{T}{b}\right) \quad (11)$$

$$\text{Then } \emptyset(b) = h(b) + T \ln\left[\hat{P}(\hat{N}_b)\right] + (C - T + kb) \ln\left[1 - \hat{P}(\hat{N}_b)\right] \quad (12)$$

where the capture probability ($\hat{P}[\hat{N}_b]$) is:

$$\hat{P}(\hat{N}_b) = \frac{T}{C + kb} \quad (13)$$

and:

j = the value of b at which the natural log-likelihood equation $\emptyset(b)$ is maximized.

Considering the possibility that the population estimate equals the total catch, the special case of b=0 needs to be defined so that division by zero is avoided.

From this:

$$\emptyset(0) = T \ln\left[\hat{P}(\hat{N}_o)\right] + (C - T + kb) \ln\left[1 - \hat{P}(\hat{N}_o)\right] \quad (14)$$

where:

$$\hat{P}(\hat{N}_o) = \frac{T}{C}.$$

Then $\emptyset(b)$ is calculated sequentially over the range of b=0, 1, 2, . . . j. When the function $\emptyset(b)$ is maximized at $\emptyset(j)$, the population equals

$$\hat{N}_i = T + j \quad (15)$$

and the capture probability equals

$$\hat{P}(\hat{N}_j) = \hat{P}(T+j) = \frac{T}{C+k(\hat{N}-T)} = \frac{T}{C+kj} \quad (16)$$

Calculation is too involved to illustrate here, but using the removal data from the previous example, the maximum likelihood population estimate is 973 compared to 978 from the Zippin approximation. Ninety-five percent confidence limits around $\hat{N}_j$ are easily determined by calculating the standard error of $\hat{N}$:

$$SE(\hat{N}_j) = \sqrt{\frac{\hat{N}[1-\hat{P}(N_i)]^k \quad \{1-[1-\hat{P}(\hat{N}_i)]^k\}}{\{1-]1-\hat{P}(\hat{N}_j)]^k\}^2 \quad - [k\hat{P}(\hat{N})]^2 \quad [1-\hat{P}(\hat{N})_j)[^{k-1}}} \quad (17)$$

$SE(\hat{N}_j) = 14.30.$

Therefore:

95 percent confidence limit lower $= \hat{N}_j - 1.96\ SE(\hat{N}_j)$

95 percent confidence limit upper $= \hat{N}_j + 1.96\ SE(\hat{N}_j)$

so 95 percent intervals equal:

$$\hat{N}_j \pm 1.96\ SE(\hat{N}). \quad (18)$$

Using our previous example and a population estimate of 973, we calculate;

$$\hat{N}_j = 973 \pm 28.02.$$

The χ^2 goodness of fit test for the Burnham maximum likelihood model is identical to that for the Zippin model except that it includes an extra term to account for the fish remaining in the stream after k removals.

$$\chi^2 = \sum_{i=1}^{k} \left[\frac{[U_i - \hat{E}(U_i)]^2}{E(U_i)}\right] + \left[\frac{[T - \hat{E}(T)]^2}{\hat{E}(T)}\right] \quad (19)$$

where:

U_i = the number of fish caught in removal i

$\hat{E}(U_i)$ = the expected catch from removal i

$= \hat{N}(1-\hat{P})^{i-1}(\hat{P})$

T = total catch

$\hat{E}(T)$ = expected total catch $= \sum_{i=1}^{k} E(U_i)$.

From our example, we calculate $X^2 = 66.02$ with k-2 = 2 degrees of freedom.

Use of the Burnham method in 1979 and 1980 resulted in narrower confidence intervals. Also, improved electrofishing techniques may be partly responsible for the narrowed confidence intervals.

Calculator Analysis

Hand calculators make the Seber-LeCren (1967) and Zippin (1958) methods simple to use. Also, calculators allow field checks of the collected data at the time of sampling to check electrofishing techniques and make sure that the required assumptions of capture are met. The successive catches can be graphed and if the plotted catches form along a linear regression line, constant capture and effort are usually indicated. If erratic catch data result, electrofishing methods must be reevaluated. If the erratic catch data are a function of nature, then nothing can be done.

Table 11. — Selected electrofishing population estimate results

Stream	Species	Population estimates per 1,800-ft reach	Confidence interval (± percent of estimate)
Horton	Brook trout	77	3
Gance	Cutthroat trout	1,135	2
Frenchman	Brook trout	716	5
Frenchman	Chinook salmon	60	[1]28
Frenchman	Sculpin	710	6
Johnson	Brook trout	346	5
South Fork Salmon River	Bull trout	682	4
Tabor	Rainbow trout	114	4
Bear Valley	Sculpin	6,577	2
Bear Valley	Chinook salmon	44	11
Bear Valley	Whitefish	121	[1]40

[1]Resulted from poor removal pattern.

Individual Fish Species

Estimates may have to be computed separately for individual fish species if they vary in their probability of capture. Species not having the same probability of capture can be evaluated separately and their probabilities added together to estimate total standing crop. We have found that rainbow trout (*Salmo gairdneri* Richardson), cutthroat trout (*Salmo clarki* Richardson), brook trout (*Salvelinus fontinalis* [Mitchill], bull trout (*Salvelinus confluentus* Suckley), and chinook salmon can be grouped together to determine total fish standing crops because their probability of capture is about the same. However, sculpin (*Cottus* sp.) and whitefish (*Prosopium* sp.) must be treated separately as their probabilities of capture are different.

Table 11 gives selected population estimates (at the 95 percent confidence level) using the Burnham maximum likelihood four-step removal method of determining fish population estimates.

Toxicants

Sodium Cyanide

Sodium cyanide (NaCn) used under strict safety precautions by trained fishery specialists is a cheap, fast, efficient, toxicant to use in collecting fish for determining fish standing crop, species composition, health, and survival rates. This compound can be purchased from chemical companies for about $1.00 per kilogram. The material is environmentally nonpersistent, but it is toxic to fish at all temperatures, and toxicity increases with temperature and is related to metabolic rates. The small amounts of compound needed to sample fish in small reaches of streams make it effective in hard-to-reach streams that are heavily vegetated, or in backcountry areas without access roads, where transporting electrofishers would be difficult. There is a need for a fish toxicant, such as sodium cyanide, that will facilitate fish removal and yet permit their return to the stream alive. The effects on fish from applied application rates of sodium cyanide over sufficient time for effects to take place are shown below:

Rate[5]	Effect
1.0 to 1.5	Trout can be collected and released unharmed, but whitefish die.
3.0	Trout will die; some more tolerant nongame fish can be collected.
5.0	All fish species can be collected, but high mortality occurs in most species.
6.0	All species die except possibly some carp and suckers.

Stream reaches selected for sampling need to be blocked off from fish escape using the same procedures discussed under electrofishing. It is suggested that these reaches be less than 300 ft (100 m) in length and less than 100 ft^3/s (2.8 m^3/s) in flow for a sufficient fish sample size with most of the population being affected by the toxicant.

Once the flow is determined the proper amount of (NaCN) is applied to the water column by placing the required number of Cyanobriks (each brick weighs about 1 ounce [20.3 g]) in a riffle at the upper end of the sample reach (Wiley and others 1975). This is an application rate and not a concentration rate. Dye is added so the flow of cyanide through the reach can be followed. Cyanobriks are manufactured by DuPont DeNemours and Company, Inc., and sold by the McKennon Chemical Company. Cyanegg, a pellet form, also can be used. The number of bricks required depends on the objectives of the sampling program and the species or group of species involved. The rate of application in the tabulation is based on water temperatures of 55° F (12.8° C) and pH of 7. Generally, 1 to 1.5 ounces (28.3 to 42.5 g) of NaCN per ft^3/s of flow and 3.0 to 3.5 ounces (84.9 to 99.1 g) of NaCN per ft^3/s of flow is effective in sampling fish in cold and warm water streams, respectively. Because of decreased metabolic rate (depressed effect of cyanide) on fish in cool water, it is recommended that NaCN not be used at water temperatures less than 50° F (10° C) (Wiley, personal communication).

If the user is working on habitats with mixed species and all fish must be returned to the stream unharmed, it means making more than one addition of the toxicant. For example, one might have to use 1.5 p/m of NaCN to collect the brown trout (*Salmo trutta Linnaeus*) and remove them from the sample area to an upstream site, and then make another run at 6.0 p/m to collect suckers (*Catastomus* sp.) and carp (*Cyprinus* sp.). Also, pools may wind up with heavier concentration of NaCN than riffles and must be watched carefully to make sure that fish can be quickly removed to eliminate mortality. Bridges (1958) found 1 p/m of NaCN in ponds produced complete kills on all species tested. However, if the fish were immediately collected upon showing stress and placed in fresh water, they survived. The size of the fish had no effect on the success of the toxicant. Recent work by Wiley in Wyoming on cutthroat trout showed the exposures for 10, 15, and 20 minutes to 1 p/m NaCN did not affect their growth or survival during the following 6-month period (Wiley, personal communication).

Sodium cyanide is dangerous to humans, so users are required to wear waders, raincoats, and rubber gloves when making contact with it (Wiley and others 1975). When transferring the chemical directly, a gas mask approved for cyanide gas or dust removal must be used. The compound should be used only in well ventilated areas. Avoid stagnant air pockets such as those that occur along streams in the early morning. Wiley and others (1975) list such safety rules as (1) cyanide must be stored in water tight containers under uniform temperatures and (2) fresh supplies of amyl nitrite inhalents must be on hand if needed to combat cyanide toxicity. At least two persons should be trained in cardiopulmonary resuscitation.

Sodium cyanide is not a registered fish toxicant. However, the Environmental Protection Agency has indicated that, when used in fishery research, it is not subject to the provisions of the Federal Insecticide, Fungicide, and Rodenticide Act. This statement should be checked prior to use as rules and regulations continually change. Sodium cyanide is an effective tool, but must be used with caution—never around domestic water supplies and always in well ventilated areas and under close supervision.

Rotenone

Over the years, rotenone has been the most widely used toxicant for fish collection or elimination. This chemical is used under much the same conditions as cyanide, but is usually in liquid form. A drip station is set up in a riffle to dispense the liquid at the concentration required. A drawback of rotenone over cyanide is that the carriers and solvents used to form the liquid repel fish more than cyanide does; therefore, blocking nets are required. Also, rotenone-toxified fish do not survive.

Toxicity of rotenone is greatest at water temperatures between 50° and 70° F (10° and 21° C) and drops as temperature

[5]This is an application rate per liter of streamflow and is not a concentration rate per liter.

decreases. In shallow streams, the toxicity decreases about 30 percent a day. This residual toxicity is another drawback because the chemical can travel long distances in flowing waters.

Potassium permanganate can be used to neutralize the rotenone effects. In standing waters, the potassium permanganate necessary to oxidize rotenone is equal to the amount of rotenone applied plus the chlorine demand of the water. In streams, this amount has been estimated as 2.5 mg/liter per cubic foot per second during the entire time the rotenone is passing through the neutralization point.

Species susceptibility ranges from 0.2 mg/liter for trout to 2.0 mg/liter for carp. At the recommended stream temperature of between 55° and 75° F (12.8° to 23.9° C) the application of 2 ounces (0.03 m^3) of 5 percent emulsified rotenone per cubic foot per second will obtain desired fish kills. For possible fish survival after collection, the fish must be netted immediately and placed in clean water. As with fish affected by cyanide, many fish never surface, but sink to the bottom. Although rotenone has been the most popular fish toxicant, we recommend that its use be avoided. The fish collection methods discussed previously do a better all-round job. The unpredictable nature of rotenone when applied to streams and the potential of "killing out" large stream areas has led many investigators to shun the use of this chemical for sampling purposes.

Explosives

Primacord

Explosion of primacord in small streams (up to stream order 4 or possibly 5) can assure an almost 100 percent collection of the fish population within the sample area. Primacard detonates at over 21,000 ft/s (6401 m/s), or essentially instantaneously. This explosive has a potential for a total kill of fish within 10 to 15 ft of the cord, provided that no major obstructions occur between the explosive and the fish (table 12).

Table 12. — Number of strands of standard size primacord to use in various stream widths and depths to assure complete fish mortality (Platts 1974)

Channel depth	Channel width				
	8	8-15	15-20	20-25	25-30
	Feet				
< 6	1	2	2	2	3
< 6-10	1	1	2	2	2
10.1-15	2	2	3	3	3

Primacord is not affected by air temperatures and can be stored for extended periods without deterioration. Water does not affect the cord for the short period of time it takes to set up the explosive grid and the cord will explode continuously even when the core is wet. Reinforced primacord is recommended because it has good flexibility, ties easily, holds well knotted, and has excellent resistance to water.

The stream area to be sampled should be blocked off with a net with mesh size small enough to keep young-of-the-year fish from leaving the area (0.125 inch and no larger than 0.225 inch [3.48 mm to 5.7 mm]). The nets must be placed at least 6 ft (2 m) above and below the sample area to keep the explosion from damaging the nets. Nets are needed for two reasons: (1) to keep fish from moving out of the area while the grid is being laid and (2) to stop dead fish from floating downstream out of the sample area after the explosion. If fish will not move out of the sample area during installation of the cord and all floating fish can be collected after the explosion, then nets are not needed.

The primacord is laid along the stream bottom, but the grid coverage must abide by the guidelines in table 12. If a major obstacle in the channel would shunt the force of the blast in the wrong direction, the cord is wrapped around the obstacle or placed on both sides of it. After the cord is laid out, it is detonated by using an electric blasting cap (from an approved electrical source) attached to one end of the trunkline. Primacord is relatively insensitive to heat, impact, friction electricity, or static electricity, so premature or accidental explosion is unlikely. Although relatively safe, it should be used only by qualified persons. No aquatic scientist should use this method until he or she has read "Primacord Detonating Fuse — What It Is And How To Use It" by the Ensign-Bickford Company, Simsbury, Conn., published in 1963. Also, each user should take a training course in explosives and be certified to handle primacord and blasting caps.

We found that electric blasting caps were the easiest and safest way of exploding the cord because the wires conducting the electrical current have safety shunts. Consequently, the cord will not detonate until this safety device is removed. The cap is simply attached to the primacord by electrician's tape, making sure that the "business end" of the cap is always pointed in the same direction as the primacord. The long electrical wires leading to the cap allow the users to get behind a protective block or far enough away from the blast for complete protection. Another reason for using electric blasting caps is that the users can detonate the cord at any desired instant. There is always the slim chance that the cap can be set off by static electricity that would not be stopped by the shunt, so the user should wear clothing of either wool or cotton, but not a mix of the two. Users should never remove or put on clothing while working with explosives. **(Blasting caps must not be brought close to the primacord until the cap is actually taped to the cord.)**

After each explosion, the dead fish are recovered by searching the stream channel; most will be on the bottom. The streambanks must also be inspected because occasionally a fish will be blown out of the channel. Usually, if they are blown above the water surface, they fall into the channel. The net should not be pulled until the water clears or until the water in the sampled area at time of explosion has passed through the downstream net. The net must be inspected closely as many of the fish will drift into it.

Abiding by the conditions for primacord use outlined in table 12, we sampled 2.75 miles (4.6 km) of stream in 39 tributaries in the South Fork Salmon River, Idaho. With constant checking, we determined that (to stream order 4) the fish sample collected was close to 100 percent of the true population. The streams were small enough for the blockage nets to be effective and the clear water allowed good observation of dead fish.

Direct Underwater Observation

Redd Counting[6]

The term "redd" is applied to salmonid nests containing embryos, but redd size varies according to the species and to female size. Salmon redd sizes vary from 18 to 137 ft^2 (1.7 to 12.7 m^2). Newly formed redds appear lighter in color than the undisturbed channel, except in gravel of basaltic origin, where the difference is much less apparent, making the redds more difficult to detect.

Training of redd-counting personnel should begin under the supervision of an experienced observer until counts are comparable. Redds should be closely examined by the trainee with parti-

[6]Contributed by Tom Welsh, Fishery Consultant, McCall, Idaho.

cular attention to the appearance of overlapping redds. If aerial (from an airplane or helicopter) counts are to be made, the trainee should have an intimate familiarity with the spawning areas. After the aerial count, he should reexamine the spawning riffles and compare the ground and aerial counts. "False redds," initial egg pockets that have been abandoned by the female before egg deposition, should not be counted.

Redd counting in streams is most easily accomplished in late summer when some salmonid species spawn and streams are low and clear. The counting of redds of spawning steelhead trout has had little success because of the higher, turbid flows in the spring. The smaller species of salmon habitually spawn in concentrated numbers, making detection of individual redds extremely difficult. In this case aerial fish counts are probably less subject to error. Aerial counts of adult salmon spawners can be used to detect differences in population size of $\pm$ 50 percent (Bevan 1961).

Newly constructed redds become progressively less discernible over time because periphyton is reestablished over the disturbed areas and, together with silt deposition, soon causes the lighter coloration of the redd to disappear. Watson (1970) found that Columbia River fall chinook salmon redds were detectable up to 6 weeks after their construction. The most accurate redd counts are made while the female is still protecting the redd. Earlier counts miss females that have not moved onto the riffles, whereas later counts miss some redds constructed earlier that have lost contrast with the surrounding substrate.

Redd counts should be used only as an index to determine large annual changes in population size. They are of limited value in determining population size for any given year, but can provide valuable time-series trends that assist in determining whether populations are stable, decreasing, or increasing. Redd counts can be biased by numerous variables, including streamflow, observer qualifications, water turbidity, light intensity, light reflection, and the changing of observers from year to year.

Ground counts — Ground counts are made while walking or using a boat and are usually more accurate and less costly than aerial counts because the observer has more time to examine each redd. Ground counts are best used on small, meandering streams with large amounts of overhanging vegetation or in steep-walled canyons where flying would be hazardous. If the spawning area is too extensive for complete counting, trend count areas can be established, preferably near the center of the spawning area, to develop yearly trend information.

Underwater redd counts. — Observations of deepwater redds are possible only with the use of scuba gear. Sockeye salmon (Oncorhynchus nerka [Walbaum]) have been detected spawning as deep as 80 ft (24.4 m) in lakes, and fall chinook as deep as 40 ft (12.2 m) in the Columbia River. Actual counts of redds are difficult with scuba gear; so the gear should be used only for determining the presence or absence of spawning redds. If divers can delineate the outer boundaries of the spawning area, then establish the average redd size, the number of redds can be crudely calculated. Underwater redd counts are slow and laborious, and counters must face the inherent danger of deep diving in rapidly flowing water.

Aerial redd counting. — Aerial trend counts have proven to be a fast, efficient method of providing an index of the spawning population. No valid comparison can be made between different observers' counts unless they have counted together and standardized their redd counting procedures.

In large rivers or in spawning areas with difficult access, aerial counts may be the only feasible method of providing population indexes. In areas of heavy redd concentrations, slower airspeeds (use of helicopters) permit the counting of individual redds, rather than multiples of 10 as required at faster airspeeds. Also, the observer can make nearly vertical observations, which increases the depth that redds can be detected, a considerable advantage on large, deep rivers. If an observer begins to suffer from motion sickness, the count should be terminated.

Aerial photographs provide a permanent record of spawning areas and can be used to estimate redd numbers. In areas of heavy redd concentration, the viewer can mark the individual redds and avoid either missing redds or duplicating counts.

Using color infrared, color positive, and color negative film in a camera equipped with a 153-mm lens in a fixed-wing airplane and photographing from a height of 1,200 ft (365.8 m) has proved successful for documenting redds for counting. The major disadvantage is cost, which averages about $3,000/mile ($1,865/km) on the Hanford reach of the Columbia river.

Snorkel and Scuba[7]

Under some circumstances, fish observations made while using snorkel and scuba gear may produce species composition and abundance data that are superior to those obtained by more conventional methods (Goldstein 1978; Griffith and Schill in press). The reasons are that: these methods may be used successfully in streams with low conductivity and substantial depth where the effectiveness of other methods, such as electrofishing, is reduced; data can be obtained with less time and money; heavy equipment may not be required, which makes the technique valuable in remote roadless areas; and fish are not handled and so are not injured. In addition, fish are observed in the habitat they have selected and not where they have been chased prior to capture. Therefore, better insight into their distribution and behavior can be gained.

Underwater observation procedures entail some limitations however, such as: water must be clear enough to allow identification of fish a minimum of about 5 ft (1.5 m) away; the observer must always keep the stream bottom in view for best results; the method does not work in areas that are too shallow or too swift; and the direct measurement of length and weight is not possible. Also, fish may escape past the diver and there is always the possibility of counting the same fish more than once. If necessary, however, individual fish may be killed with a "gun" that detonates electrical blasting caps (Everest 1978).

Two potential sources of bias must be accounted for to obtain reliable data when using snorkel or scuba methods. One potential source of bias lies in the fact that the reaction of fish to underwater observers varies greatly among fish species. Some species, such as the mountain whitefish, *Prosopium williamsoni* (Girard), that school or aggregate in large numbers may be difficult or impossible to census accurately. Other species, such as some darters (*Etheostoma* sp.) and sculpins, may be too secretive or evasive to be censused during the day, although night surveys may be effective. Trout and salmon often hold their territories in the presence of the observer and are relatively ideal to census. The second potential source of bias is the variability in performance among individual observers. Each observer, therefore, should compare his or her performance with that of others or individually check themselves on a stream section that holds a known number of fish. When fish censusing is repeated periodically in a stream, the same observer(s) should be used on each occasion.

Safety considerations cannot be overemphasized. All stream sections to be censused must be studied from the bank to determine

[7]Contributed by Dr. J. S. Griffith, Associate Professor, Idaho State University, Pocatello.

if there are any hazardous areas. Snorkel or scuba work should never be done alone, and ropes should never be attached to an observer's body while that person is in the water. A scuba course should be completed before using this technique. Snorkeling presents little risk if the correct equipment and safety procedures are used.

Procedures. — A neoprene suit of the wet or dry type, preferably ¼-inch (6.4-mm) thick, with boots, gloves, and hood is generally needed for warmth. The wet suit allows exposure to 60° F (16° C) for several hours, depending upon the individual. The dry suit, which is similar to a wet suit but has seals on ankles, wrists, and neck to exclude water, can be used in colder water or for longer exposure. The suit should be custom-fitted for each individual to maximize its effectiveness. Fins are needed in large rivers to increase maneuverability, but are a hindrance in smaller streams. At present, a complete wet suit costs about $250 from the manufacturer and a dry suit costs about $80 more.

Observers must move in the water with a minimum of disturbance and should look as far ahead as possible to locate the fish on the fringe of vision. Several practice sessions are needed to become effective in locating fish.

The underwater visibility should be measured before each day's observations. Use an object the same size as the fish to be observed (a flashlight, for example) and measure the maximum distance at which it can be seen. Record this measurement for comparison with subsequent observations in that reach.

Each census must be planned for a successful counting. Observers must determine the fish species to be censused and record the size groups or age groups of each species recorded, the time the census is to be taken, the habitat to be included in the sample, and the direction of observation routes in the stream.

If the fish community is diverse, it may not be possible for one observer to record the numbers of every species. In that case, the observer should select only key species to count, or several observers should be used and each should count different species. Fish counts can be recorded on hand-held tally counters or on underwater slates.

Young-of-the-year can usually be distinguished from older fish. If there is minimal overlap in size between successive age groups and the observer has prior knowledge of the relationship between age and size of fish in the population, it is feasible to keep separate counts for each age group (Griffith 1981). Direct estimation of fish length also may be feasible under some circumstances. Griffith and Fuller (1979) marked 45 trout 8.5 to 17.5 inches (216 to 445 mm) in length with color-coded tags and then had five observers estimate their length by sight only. Without advance preparation, 52 to 72 percent of the estimates by each observer fell within 1 inch (25 mm) of the actual fish length. After 1 hour of practice on objects of known length, the most experienced observer estimated lengths within 1 inch (25 mm) 90 percent of the time, with a mean of 62 percent for all observers. Each individual must train himself to compensate for the 1.33 underwater magnification factor by practicing and perhaps carrying a short ruler taped to the wrist or making length units on a glove.

The behavior of the fish should be considered when selecting the time of censusing. Daytime sampling is adequate or preferable for many fish species and is more convenient for the observer. Consistency is important. Cloudy days when visibility is reduced should be avoided, and shadows on sunny days should be minimized by diving around midday. If censusing is to be done at night, it should be done consistently on the same phase of the moon, as behavior and distribution of some fish species may vary between phases.

The only habitat that can be effectively snorkeled in small streams (usually second and third order) may be the pools. In larger streams, basic habitat types can be stratified and counts made separately for each, or all habitats can be grouped together, depending on the needs of the observer. If the habitat is uniform, the starting point for each census should be selected in a random manner. If an area is to be recensused in the future, it is critical that its boundaries be permanently marked with metal stakes and the reach photographed.

There are three possible directions to be used by the observer in conducting the census. Moving upstream is the most effective, if it is feasible. This can be done in small streams of low velocity where walking or crawling is possible. On larger streams, the observer must travel with the current. In some areas, the water may be shallow enough or slow enough to permit the running of transects from bank to bank perpendicular to the flow, but this is uncommon.

Most underwater counts are done to establish trends in species composition or species density to compare between areas, seasons, or years. Therefore such trend counts are designed as indexes of the relative status of a population rather than rigorous population estimates. With proper planning and careful execution, however, population estimates can be made under some circumstances.

Population estimation with single observer. — This technique should be used when the observer can scan the stream from bank to bank. Several passes should be made through the initial section to determine if such repeat counts are consistent. If not, the procedure must either be adjusted to gain the necessary accuracy or the technique should be abandoned. Another accuracy check is to have a second observer make the same count at the same time or immediately following the first observer's pass, if two observers would increase disturbance to fish.

The habitat types within the stream reach are counted separately or are combined, depending upon the design selected. If there is an indication that fish within the areas counted are not distributed randomly, the data should be tested for spatial distribution (Elliott 1977) by examining the relationship between the variance and mean of the population. If variance is significantly less than the mean, a uniform or underdispersed distribution is present; and if variance significantly exceeds the mean, the fish are clumped or overdispersed. If these conditions exist, data should be transformed as necessary. Confidence intervals around the mean are then calculated as described previously and expressed in terms of numbers of fish per unit of stream length or surface area.

Population estimation with several observers. — This approach is more complicated from the standpoint of logistics, but is necessary to obtain better data on large rivers. Observers in underwater gear drift with the current counting routes in lanes. Lane width is dictated by underwater visibility. To be effective, observers must stay in a line perpendicular to the current. Thin fiberglass or plastic poles about 16 ft (4.9 m) long are held by observers to maintain position in the current and to maintain correct width of counting lanes (Griffith and Schill in press). Each observer counts fish passed on one side of the observer's body only. Since shallow stream margins are likely to contain more juvenile fish (and perhaps some different species), fish should be counted separately. Confidence intervals can then be calculated as described above.

Population estimation using mark-recapture. — If it is possible to mark (by angling or another technique) a number of fish with color-coded tags that can be recognized by underwater observers (fin-clips are not adequate), population estimates can be made. Observers record the numbers of tagged and untagged fish

of the appropriate species that are seen. Using these data, the population can be estimated using the Petersen formula N=MC/R, where N is the estimated population size, M is the number of tagged fish released, C is the number of fish observed by the observers, and R is the number of tagged fish observed.

MACROINVERTEBRATE ANALYSIS

By convention, freshwater macroinvertebrates are those animals without backbones that are large enough to be seen without magnification. The main taxonomic groups of macroinvertebrates occupying freshwater environments are annelids, crustaceans, flatworms, mollusks, and insects (usually predominant). Their lower size limit has been variously defined by their retention on screens or nets with mesh openings of 0.023 inches (0.589 mm) (American Public Health Association 1976; Weber 1973), 0.011 inches (0.280 mm) (Winget and Mangum 1979), and 0.008 inches (0.210 mm) (Greeson and others 1977). The latter appears to be most suitable for obtaining representative collections of most macroinvertebrates in flowing waters (the principal exception is midge larvae) and has been adopted by the U.S. Geological Survey (Greeson and others 1977). A 0.210-mm mesh opening is equivalent to a U.S. Standard No. 70 sieve.

Macroinvertebrates are important intermediaries in the utilization of plant material, such as algae, vascular hydrophytes, leaves, and wood, and the recycling of nutrients in aquatic environments. They are a major food source for fish and serve to determine the well-being of those populations. In particular, the macroinvertebrates possess several characteristics that make them useful for detecting environmental perturbations: (1) most members of this community possess limited mobility so that their status reflects conditions in the immediate vicinity of the collection site, (2) most of the organisms (mussels are the main exception) have life spans of several months to a few years. Thus, their characteristics are a function of conditions during the relatively recent past, including sporadic influences that would be difficult to detect by periodic microbial or chemical analysis.

Some of the first things that a resource manager must consider in the utilization of macroinvertebrates as an investigative tool are whether the sampling should be qualitative or quantitative and whether to concentrate on selected "indicator species" or to include the entire community. Because of constraints of time and money, the temptation often is to employ qualitative collections and/or to examine selected groups. But this choice often proves most costly in the long run. It provides less information, thereby greatly reducing the reliability and usefulness of the data; yet the same or more specialized expertise may be required. Consequently, some form of quantitative or semiquantitative sampling of the full macroinvertebrate community is recommended for the situations most likely to be encountered by users of this manual.

The purpose of semiquantitative sampling is to determine the relative abundance of each species in a standardized manner so that spatial and temporal changes in numbers and/or biomass can be measured. Sampling methods include the use of uniform substrates (natural or arrificial) or collection with a dip net in a standardized manner or over an established time period. Values are reported per unit sample rather than per area. Expression of the results as a percentage of the total numbers collected at a site is to be avoided since the values obtained for each taxon are strongly influenced by the values of the other groups collected (Elliott 1977).

The purpose of quantitative sampling is to determine the absolute abundance of each species per unit area of habitat. The samples provide measures of population densities which may be used to detect variations in time and space and which are essential for the determination of biological production. In addition, quantitative samples may be more representative of actual conditions than semiquantitative ones. For example, introduced substrata may provide conditions considerably different from those actually found in the environment.

Sampling Strategy

Design of a proper sampling scheme must take into account the location of sample collections, when and how often the collections are to be made (sampling frequency), and the number of replicates to be obtained (sample size). In addition, sampling variability resulting from sampling device operations, physical features of the environment, laboratory sorting procedures, and biological features of the study populations may confound interpretations of the results. There are a number of sources of information for guidance in addressing these questions, including reviews by Elliott (1977), Greeson and others (1977), Hellawell (1978), Hynes (1970), Resh (1979), Southwood (1978), and Weber (1973).

Sample Location

Sampling location involves both selection of the collecting sites (stations) and determination of the specific location from which the samples are to be taken. Sample site selection is determined by the specific question being addressed. For example, a point-source of pollution or a localized problem area would require a minium of one site each above and below the affected area. Additional downstream stations would be necessary to assess the extent of influence of the disturbance and extra upstream stations would be useful to establish the variation between control sites (Hellawell 1978). Tracking the effect of a nonpoint souce disturbance might involve locating a number of stations along a length of stream or establishing collecting sites at control and disturbed locations in different watersheds (for example, grazed versus ungrazed, burned versus unburned).

When more than one site is being examined, one may choose to sample one or a few standard habitat types (especially appropriate in semiquantitative studies) or to obtain samples representative of the overall conditions at each site (as is usually required in quantitative programs). Riffles are commonly chosen as standard sample sites because of their relative uniformity in terms of substratum and current, their higher biotic diversity, and their greater accessibility except during flood. However, such erosional areas clearly are unsuitable or at least inadequate if one is interested in studying the effects of an agent, such as inorganic sediment, that would be apparent mainly in depositional areas. Likewise, if one is interested in comparing the productivity of one section of stream with another, then sampling all major habitats and expressing results as an area-weighted mean may be the most satisfactory approach. With this approach, when the area is divided into several strata (subhabitats), the sampling design is termed "stratified."

Regardless of which of the above strategies is used, a proper sampling scheme requires that replicate samples within a site be taken with conscious avoidance of bias. This may be done through either random or systematic sampling. Random sampling is done most easily by dividing the area into quadrants, each the size of a replicate sample, and then selecting the quadrants to be sampled by use of a random numbers table. The distribution of macroinvertebrates generally is heterogeneous (clumped), largely as a result of the nonrandom distribution of microenvironmental features, especially the substratum and the current. Consequently, sampling that is strictly random will have a relatively large error when applied to a natural population. For this reason stratified random sampling is often preferred.

In systematic sampling, the first unit in the sample is selected at random and the next units are established at fixed intervals from the first. Additional details and examples are given by Elliott (1977: 131-136), Greeson and others (1977: part 1, 10-19), and Weber (1973: 4-6). Users of this manual probably will find the systematic approach appropriate in most cases and easiest to apply. A common procedure for intermediate-sized, third- to fifth-order streams might be to mark off a length of stream or a riffle at established intervals, such as 3.3 ft (1 m), with each interval being the site of a potential sampling transect. The specific transects to be sampled could be selected at random from a container holding the numbers of all of the transects present. Upon reaching the selected transect, samples could be collected from the center of the stream, and from half of the way and one-fourth of the way between the center and each bank for a total of five replicates. In smaller, first- and second-order streams, the samples might be taken at fixed distances down the stream rather than across it.

Sample Frequency

The distribution and abundance of many macroinvertebrates and, consequently, their community composition are subject to wide seasonal variations. Thus, when conducting comparative studies, the investigator must avoid the confounding effects of these seasonal changes; collections made in different locations must be from the same time period (week or month) to minimize variations resulting from life cycle changes. If only one collection a year is possible, it should be taken in the spring when a majority of the insects present are well developed and easier to identify. The collections also should be made before spring runoff because high flows disturb the stream bottom and make working the stream difficult. If only two collections a year are made, the second set should be taken in late summer. All the same, monthly collections are desirable. However, in situations where the full community makeup and life cycle variations are not known, a minimum of one collection per season is recommended. Additional collections may be needed to pinpoint the effects of specific events and should be made just before and after an event, such as road construction.

Sample Size

The size of the mean, the degree of aggregation, and the desired precision of the mean estimate will influence the number of samples required to estimate densities of benthic populations (Resh 1979). A relatively large number of sample replicates, possibly several hundred, must be collected from each site if the goal of the sampling program is to describe the macroinvertebrates of an area with a high degree of accuracy. The number could increase many times if a stratified sampling scheme is called for. However, where most surveys are concerned, a high degree of accuracy may be counterproductive because extremely subtle, but statistically significant differences may be tolerated by the investigator or resource manager and reasonably rapid turnaround of results may be required; therefore, a compromise must be made between statistical accuracy and time and labor.

Three samples per habitat type is the absolute minimum required in any study and might be sufficient for a general faunal survey of a stream (Cairns and Dickson 1971). Five replicates per habitat would increase the statistical power of the samples with relatively little additional effort. For example, increasing the sample size (N) from 3 to 5 will (at $P<0.05$ and $N-1$ degrees of freedom) decrease the Student's t distribution (appendix 1) by 1.55 $\times$ t = 4.303 versus t = 2.776, whereas increasing the sample size from 5 to 60 will decrease t by less than half that much to t = 2.00 (appendix 1). Therefore, it is recommended that a minimum of five samples per habitat type be taken in the situations likely to be encountered by the users of this manual. In general, a larger number of replicates will be required to adequately represent the mean for a macroinvertebrate community consisting of a large number of species with a patchy distribution of individuals (the usual case in most unpolluted riffles) than will be required for a community represented by large numbers of a few species evenly distributed in the stream.

Elliott (1977: 129-131) describes two techniques for determining a suitable sample size. The first involves taking groups of five replicates at random, calculating the means for each 5, 10, 15, etc., units, and then plotting these against sample size. When the mean value ceases to fluctuate, a suitable sample size has been reached and this sample size can be used for that particular station or subhabitat. Since it is often impossible to calculate means at the time of sampling, this method is of limited application. In the second method, the ratio of standard error to arithmetic mean ($\bar{x}$) is taken as an index of precision (D). Therefore, sample size (N) can be calculated for a specified degree of precision by using the equation:

$$N = \left(\frac{ts}{Dx}\right)^2,$$

where D = relative error in terms of percentage confidence limits of the mean, s = standard deviation, and t = Student's t for the required probability. If, for example, in a preliminary survey or a previous set of samples, a mean number of individuals per sample was found to be 385 and the standard deviation 244, then for a relative error of ± 40 percent (equal to a standard error of about 20 percent, a reasonable level in most macroinvertebrate samples) with a probability of 95 percent ($t \tilde{=} 2$). Entering those numbers into the formula

$$N = \left(\frac{ts}{D\bar{x}}\right)^2,$$

we get

$$\left(\frac{2 \times 244}{0.4 \times 385}\right)^2,$$

which equals about 10 samples.

Sampling Methods

A number of possible sampling devices have been described for use in streams (American Public Health Association 1976; Greeson and others 1977; Hellawell 1978; Hynes 1970; Welch 1948). However, each device has its own sources of error and, since these are seldom known and are rarely identical between different types of samplers, it is well to limit the selection to a few relatively standard forms thereby facilitating comparison of results obtained by different workers. In the United States, of the semiquantitative samplers (fig. 24A, B, C, and D), the most common for use in streams are the multiplate (Hester and Dendy 1962) and basket (Mason and others 1967, 1973) samplers (fig. 24C, D, and E), whereas the most widely used quantitative devices are the Surber (Surber 1937) and modified Hess samplers (Waters and Knapp 1961) (fig. 24A and B). In streams that are too deep to wade, the semiquantitative collapsible basket developed by Bull

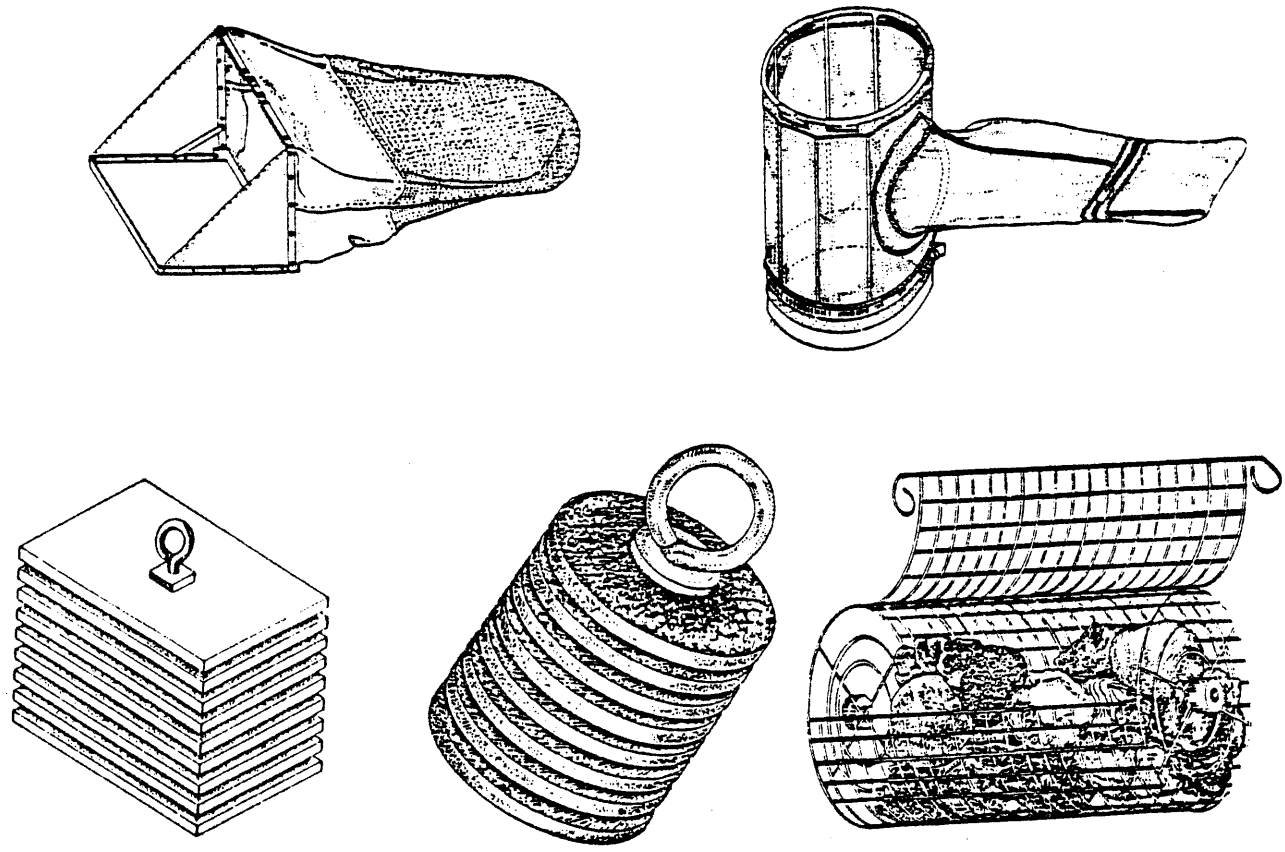


Figure 24. — Sampling devices for stream macroinvertebrates: (A) Surber sampler; (B) modified Hess net; (C) square; (D) circular versions of multiplate sampler; and (E) basket sampler. Illustrations A, B, and E are from Merritt and Cummins (1978) and are used with the authors' permission.

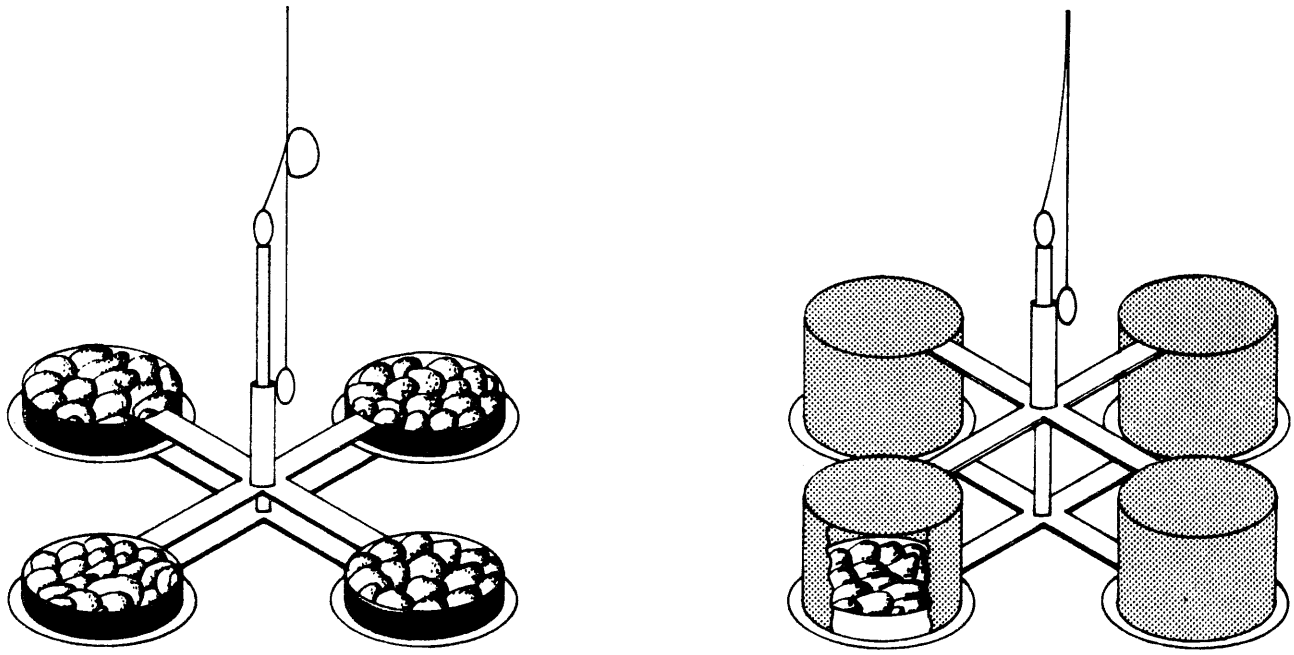


Figure 25. — Collapsible basket with substrate sampler (left) resting on streambed (right) being retrieved. (After B. Malmquist and L.M. Nilson, personal communication.)

(1968) and modified by Malmquist and Nilsson (personal communication) and the quantitative suction device described by Gale and Thompson (1975) are widely applicable (fig. 25 and 26). Procedures for the use of the various sampling devices are described in detail by Greeson and others (1977), Lind (1979), Weber (1973), and Welch (1948). Major items of consideration are described below

The specific sampling location should be approached from downstream and the collecting net placed into position as quickly as possible to reduce the potential for escape by the macroinvertebrates. For semiquantitative samplers, a hand-held dip net or specially fabricated net with a mesh of 0.008 inch (0.210 mm) is used to enclose the sampler, which is then carried to shore. The sampler and net contents may be placed directly into a container of preservative or the sampler may be disassembled at streamside, the plates or rocks placed in a tray of water and scrubbed clean with a brush, and the contents of the tray passed through the net before being placed in the container of preservative. If circular multiplate samplers having 3-inch (75 mm) diameter plates and 1-inch (25 mm) diameter spacers are used, the sampler can be placed directly into a widemouth quart jar. For quantitative samplers, the bottom frame of the Surber or Hess net should be pressed tightly against the stream bottom to avoid contamination from outside the sample area. On irregular bottoms, a more complete seal can be obtained by lining the bottom of the sampler with foam rubber, burlap, or other compressible material. The larger rocks should be lifted, scrubbed at the mouth of the net opening, and removed from the sampler. Thoroughly disturb the remaining sediment to a standard depth (usually 2.0 inches [50 mm] or 3.9 inches [100 mm]) by repeatedly digging and stirring (a railroad spike is useful for this). The invertebrates and lighter debris then will be carried into the net. The top of the net should be tipped downstream until a 45° angle is formed with the streambed and the sampler quickly removed from the water. The net should be dipped several times in the stream to wash the contents to the bottom, but workers must be careful not to submerge the net opening. Net contents should then be transferred to a sample container. A net or shallow pan should be placed beneath the container to catch any spillage. The net and its seams should be carefully checked for adhering specimens.

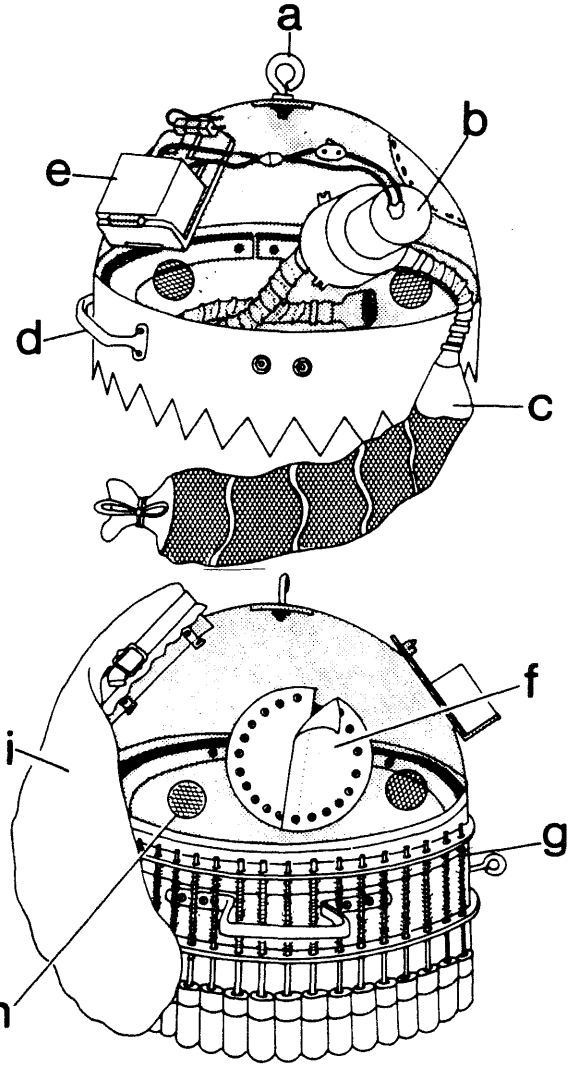


Figure 26. — Dome sampler with serrated band (rear view) and polyurethane cylinder band (side view): (a) eye bolt, (b) bilge pump, (c) net bag, (d) handle, (e) battery, (f) armhole cover, (g) self-adjusting contour rod, (h) screened port, and (i) rock bag. (From Gale and Thompson [1975] with the authors' permission.)

The samples should be preserved in 70 percent ethanol or 2 percent formaldehyde solution (5 percent formalin), and a volume of preservative at least equal to the volume of organic material added to insure adequate preservation. The containers should be filled to reduce damage to the macroinvertebrate specimens. Workers should use waterproof label paper or other material that will not deteriorate in water and a soft lead pencil or waterproof ink for identifying the collections. Label information should include location, habitat, and date of collection. Such additional information as sampling conditions, type of sampling device and mesh size, and name of the collector should be entered in a bound field notebook. The label should be placed inside the sample container; a duplicate label on the outside of the container provides added insurance that the information will not be lost and saves time in subsequent handling of samples.

Sample Processing

Preprocessing reduces weight and bulk and prevents destruction of invertebrates from grinding by sediment. The sample should be placed in a large bucket or tray. Add water and swirl or stir the contents of the container to suspend the organic material. The suspension should be poured through the collecting net so that the heavier inorganic sediments will be left behind. This process should be repeated until no additional organic debris enters the net. The inorganic residue should be spread in a white tray and flooded with water. Such specimens as stone-cased caddisflies, mollusks, or planarians that have withstood the washing process should be examined and removed with forceps. The sediments should be discarded and the remainder preserved.

Whether the preprocessing step is done in the field or in the laboratory, the next step is to process the sample through a series of steps that ultimately will yield the raw data of the macroinvertebrate phase of the study:

1. Remove the organisms from the organic debris.
2. Sort them into groups of look-alikes (coarse sorting).
3. Identify the individual specimens to the taxonomic level desired and sort the look-alike groups into these categories (fine sorting).
4. Count and/or weigh the contents of each category and enter the values onto data forms.

Trays with white background or light transmitted from below should be used for removing the macroinvertebrates from the remaining organic matter. A large, low power (3X), illuminated magnifier is helpful at this stage. Only very small amounts (approximately a heaping tablespoonful) of material should be placed into a 15.7- by 9.8- by 2.0-inch tray (400- by 250- by 50-mm) about one-third full of water. For samples containing large numbers of organisms, processing time can be substantially reduced if the samples are subdivided before sorting. Details of two possible subsampling procedures are given by Weber (1973) and Waters (1969). Separation of invertebrates from plant and inorganic debris may be facilitated by flotation (Anderson 1959), differential staining (Mason and Yevich 1967), or a combination of these procedures (Lackey and May 1971).

As organisms are picked from the debris, they should be coarse-sorted into major groups and placed into leakproof vials filled with preservative (16.9 ounces [500 ml] 70 percent ethanol plus 0.3 ounce [10 ml] formalin [40 percent formaldehyde solution] plus 0.2 ounce [5 ml] glycerin) and the vials labeled. All vials from a sample should be kept together in a suitable container until processing is completed. A record should be kept of which worker sorted the sample.

The taxonomic level to which macroinvertebrates are identified depends on project objectives and available resources. But, except in cases of severe environmental disturbance, most situations needing assessment require identification to genus or species. The taxonomic level to which identifications are carried in each taxon should be constant throughout a particular study. The accuracy of identification depends on the experience and skill of the investigator and the availability of taxonomic literature. Basic sources of information include books by Edmondson (1958), Edmunds and others (1976), Pennak (1978), Usinger (1956), and Wiggins (1977) and the literature cited in these publications. Most identifications to family and genus can be made with the aid of a 5 to 50X stereoscopic microscope; those identifications to species often require a compound microscope. Maximum counting efficiency is at 25X magnification with transmitted light (Frost 1971).

Biomass measurements can be obtained by drying the organisms at 221° F (105° C) for at least 4 hours and then weighing them. Ash-free dry mass can be obtained by incinerating the material at 1022° F (550° C) for 1 hour, cooling in a dessicator, and calculating the difference between initial (dry) and final (ash) weights.

Data Treatment and Interpretation

The treatment and interpretation of data obtained from macroinvertebrate collections is as much an art as it is a science and a detailed understanding of benthic invertebrate ecology is advisable. Basic information sources include books by Hellawell (1978), Hynes (1970, 1971), and Mackenthun (1969).

In the material that follows, we provide a synopsis of the principal methods used to analyze benthic macroinvertebrate data and information to guide interpretation of the results. However, the presentation is necessarily brief in keeping with the purposes of this manual. The nonspecialist should proceed with caution and should supplement the information provided by reference to the specific citations given or seek the aid of a competent professional.

Table 13. — Mean standing crops of benthic macroinvertebrates in some Rocky Mountain streams as ash-free dry mass

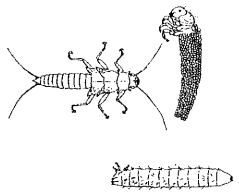
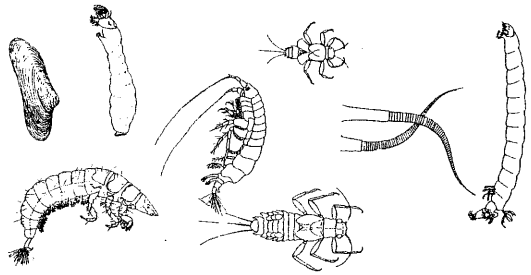
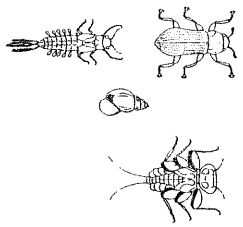
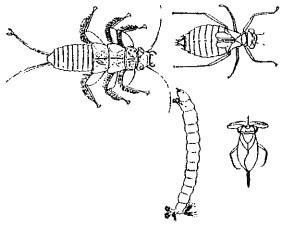
Stream	Location	$\bar{x}$ numbers/m²	$\bar{x}$ biomass/m²	Reference
			Grams	
Firehole River	Wyoming	940		Armitage 1958
Unnamed Springbrook	Colorado			
Station 5		1,700	34.7 (wet weight)	
Station 4		4,100	136.9 (wet weight)	Ward and Dufford 1979
Mink Creek (1968-69 study)	Idaho	6,900	10.8 (ash-free dry mass)	Minshall 1981
Strawberry River	Utah	8,800		Payne 1979
Mink Creek (1969-70 study)	Idaho	21,000	26.5 (ash-free dry mass)	Minshall 1981

Table 14. — Composition (percent of total numbers) of macroinvertebrate communities in some Rocky Mountain streams (Andrews and Minshall 1979)

Stream	Location	Ephemeroptera	Trichoptera	Diptera	Plecoptera	Others
Morrell Creek	Montana	65.0	4.4		14.5	[1]16.0
Deer Creek	Utah	63.9	17.9	12.3	2.2	3.4
Little Lost River	Idaho	61.5	14.4	10.3	12.4	1.4
Trail Creek	Idaho	52.0	6.0	16.0	6.0	20.0
Pine Creek	Idaho	48.0	18.0	9.0	7.0	18.0
Mink Creek	Idaho	46.5	9.9	13.0	12.2	18.4
Viviana Park Creek	Utah	43.5	5.7	14.4	26.8	9.6
Bridger Creek	Montana	9.2	71.9	12.2	1.7	5.0
Aspen Grove Creek	Utah	8.9	56.9	5.3	11.4	15.5
Madison River	Montana	22.5	35.4	23.8	1.8	16.5
Provo River	Utah	23.2	28.6	21.1	13.7	13.4
Portneuf River	Idaho	3.0	10.8	72.0	0.2	14.0

[1]Includes Diptera.

Table 15. — A general classification system for aquatic insect trophic categories (after Cummins 1973)

General category based on feeding mechanism	General particle size range of food	Subdivision based on feeding mechanisms	Subdivision based on dominant food	North American aquatic insect taxa containing predominant examples
	Microns			
SHREDDERS 	$< 10^3$	Chewers and miners	Herbivores, living vascular plant tissue	Trichoptera (Phryganeidae, Leptoceridae) Lepidoptera Coleoptera (Chrysomelidae) Diptera (Chironomidae, Ephydridae)
		Chewers and miners	Detritivores (large particle detritivores): decomposing vascular plant tissue	Plecoptera (Filipalpia) Trichoptera (Limnephilidae, Lepidostomatidae) Diptera (Tipuiidae, Chironomidae)
COLLECTORS 	$< 10^3$	Filter or suspension feeders	Herbivore-detritivores: living algal cells, decomposing organic matter	Ephemeroptera (Siphlonuridae) Trichoptera (Philopotamidae, Psychomyiidae, Hydropsychidae, Brachycentridae) Lepidoptera Diptera (Simuliidae, Chironomidae, Culicidae)
		Sediment or deposit (surface) feeders	Detritivores (fine particle detritivores): decomposing organic matter	Ephemeroptera (Caenidae, Ephemeridae, Leptophlebiidae) Trichoptera (Glossosomatidae, Helicopsychidae, Molannidae, Odontoceridae, Goerinae) Lepidoptera Coleoptera (Elmidae, Psephenidae) Diptera (Chironomidae, Tabanidae)
SCRAPERS 	$< 10^3$	Mineral scrapers	Herbivores: algae and associated material (periphyton)	Ephemeroptera (Heptageniidae, Baetidae, Ephemerellidae) Trichoptera (Glossosomatidae, Helicopsychidae, Molannidae, Odontoceridae, Goerinae) Lepidoptera Coleoptera (Elmidae, Psephenidae) Diptera (Chironomidae, Tabanidae)
		Organic scrapers	Herbivores: algae and associated material (periphyton)	Ephemeroptera (Caenidae, Leptophlebiidae, Heptageniidae, Baetidae) Hemiptera (Corixidae) Trichoptera (Leptoceridae) Diptera (Chironomidae)
PREDATORS 	$> 10^3$	Engulfers	Carnivores: whole animals (or parts)	Odonata Plecoptera (Setipalpia) Megaloptera Trichoptera (Rhyacophillidae, Polycentropidae, Hydropsychidae) Coleoptera (Dytiscidae, Gyrinnidae) Diptera (Chironomidae)
		Piercers	Carnivores: cell and tissue fluids	Hemiptera (Belastomatidae, Nepidae, Notonectidae, Naucoridae) Diptera (Rhagionidae)

Abundance

The raw data obtained from the processing of stream-collected macroinvertebrate samples can be analyzed in a variety of ways to enhance informational value to an aquatic specialist or resource manager. As a first step in data analysis, the values (numbers or biomass) for each taxon and for all taxa combined should be tabulated and the means and variances determined for each station. Expression of these results as amounts per sampler or amounts per unit area provides the basis for comparisons between stations, times, streams, and published works. Comparisons enable aquatic ecologists to determine such things as the biological condition of the stream, the extent to which the stream has been impacted by environmental disturbance, and the potential for stream improvement. Reliable published values for evaluations of this sort are few. But, it appears for example, that total numbers of organisms in most undisturbed Rocky Mountain streams can be expected to lie between 93 and 930/ft² (1 000 and 10 000/m² (table 13) depending on nutrient levels, current velocity, substratum type, and other factors controlling overall stream productivity.

In addition to evaluating the absolute quantities of organisms present, it is important to know the relative abundance of each taxon to establish the extent to which the macroinvertebrate community is considered to be in biological balance. For example, data for a number of Rocky Mountain streams (table 14) show that under normal circumstances mayflies (Ephemeroptera) or caddisflies (Trichoptera) would be expected to be numerically predominant. The predominance of true flies (Diptera) in the Portneuf River, Idaho, supports the contention (Minshall and Andrews 1973) that it is polluted. See appendix 7 for tolerance quotients of macroinvertibrates.

Richness

Another valuable indicator of macroinvertebrate community status is the total number of taxa (preferably species) present at a specific site on a given sampling date or on an annual basis. The number of taxa is termed richness and can be expected to decrease with either natural or man-caused environmental stress. In general, for unperturbed Idaho streams, it has been estimated that the number of persistent species of macroinvertebrates (exclusive of Chironomidae) occurring during the year will be between 50 and 65.

Functional Feeding Group Status

Cummins (1973, 1974) has advocated the organization of macroinvertebrate data into functional categories based on feeding behavior as a means of gaining insight into ecosystem status. A general scheme for placement into appropriate feeding categories is given in table 15 and additional information is summarized by Merritt and Cummins (1978). Although the approach shows considerable promise, relatively little use has been made of it to date (Hawkins and Sedell 1980; Minshall 1981). However, caution should be used in placing macroinvertebrates into functional categories using published information, such as table 13, as many of these are crude approximations and conditions may vary among streams and times of year.

Biological Indexes

The complexity of data on benthic macroinvertebrate communities has led to the use of various biological indexes in order to provide fuller undestanding of the data and/or to simplify their presentation and interpretation. However, whatever valuable adjunct these indexes serve, they should not be used as substitutes for the basic information on abundance and biomass described above. A particularly lucid explanation of the uses and limitations of biological indexes is given by Warren (1971).

Two approaches have been used. One involves mathematical manipulation of information on the number of individuals per taxon (abundance) and the number of taxa present in a community (richness) and is termed a diversity index. Since environmental stress frequently reduces community diversity, such indexes are potentially valuable devices, provided that the change in value of the index is related to the intensity of the disturbance. The second approach attempts to incorporate information on the environmental requirements of the species involved and is termed a biotic index.

Diversity Index

The most widely used community diversity index is that of Shannon-Wiener (Wilhm 1968; Wilhm and Dorris 1968) and is calculated as:

$$H' = -\sum_{i=1}^{s} (n_i/n)\log\,(n_i/n)$$

where n = the total number of individuals of all taxa, n_i = the number of individuals in the ith taxon, and s is the total number of taxa in the community. The base of the logarithm must be specified and usually is $\log_2$. The advantages of this index over other possible diversity indexes include: (a) relative abundances of the different taxa are taken into account; (b) it is relatively independent of sample size; and (c) the values are dimensionless and therefore are not dependent on the unit of measurement used.

In general, values ($\log_2$) of H' less than 3 are found for benthic invertebrates in areas of clean water, values from 1 to 3 in areas of moderate pollution, and values less than 1 in heavily polluted

Table 16. — Shannon-Weiner diversity (H′) and equitability (e) for some Rocky Mountain streams

Stream	Location	H′	e	Source
Unnamed Springbrook	Colorado	1.8-3.7	0.1-0.5	Ward and Dufford 1979
Mink Creek	Idaho	3.7	0.3	Minshall 1981
Horse Creek	Idaho	2.8-3.2		Newton and Rabe 1977
Upper Blackfoot River	Idaho	2.6-4.3	0.2-0.7	Platts and Andrews 1980
Portneuf River (Stations 2, 5, 8, 9b)	Idaho	1.3-2.6	0.1-0.4	Minshall and Andrews 1973

Table 17. — The hypothetical number of species (s*) for various values of H′ (Lloyd and Ghelardi 1964)

s*	H′	s*	H′	s*	H′	s*	H′
1	0.0000	51	5.0941	102	6.0792	205	7.0783
2	0.8113	52	5.1215	104	6.1069	210	7.1128
3	1.2997	53	5.1485	106	6.1341	215	7.1466
4	1.6556	54	5.1749	108	6.1608	220	7.1796
5	1.9374	55	5.2009	110	6.1870	225	7.2118
6	2.1712	56	5.2264	112	6.2128	230	7.2434
7	2.3714	57	5.2515	114	6.2380	235	7.2743
8	2.5465	58	5.2761	116	6.2629	240	7.3045
9	2.7022	59	5.3004	118	6.2873	245	7.3341
10	2.8425	60	5.3242	120	6.3113	250	7.3631
11	2.9701	61	5.3476	122	6.3350	255	7.3915
12	3.0872	62	5.3707	124	6.3582	260	7.4194
13	3.1954	63	5.3934	126	6.3811	265	7.4468
14	3.2960	64	5.4157	128	6.4036	270	7.4736
15	3.3899	65	5.4378	130	6.4258	275	7.5000
16	3.4780	66	5.4594	132	6.4476	280	7.5259
17	3.5611	67	5.4808	134	6.4691	285	7.5513
18	3.6395	68	5.5018	136	6.4903	290	7.5763
19	3.7139	69	5.5226	138	6.5112	295	7.6008
20	3.7846	70	5.5430	140	6.5318	300	7.6250
21	3.8520	71	5.5632	142	6.5521	310	7.6721
22	3.9163	72	5.5830	144	6.5721	320	7.7177
23	3.9779	73	5.6027	146	6.5919	330	7.7620
24	4.0369	74	5.6220	148	6.6114	340	7.8049
25	4.0937	75	5.6411	150	6.6306	350	7.8465
26	4.1482	76	5.6599	152	6.6495	360	7.8870
27	4.2008	77	5.6785	154	6.6683	370	7.9264
28	4.2515	78	5.6969	156	6.6867	380	7.9648
29	4.3004	79	5.7150	158	6.7050	390	8.0022
30	4.3478	80	5.7329	160	6.7230	400	8.0386
31	4.3936	81	5.7506	162	6.7408	410	8.0741
32	4.4381	82	5.7681	164	6.7584	420	8.1087
33	4.4812	83	5.7853	166	6.7757	430	8.1426
34	4.5230	84	5.8024	168	6.7929	440	8.1757
35	4.5637	85	5.8192	170	6.8099	450	8.2080
36	4.6032	86	5.8359	172	6.8266	460	8.2396
37	4.6417	87	5.8524	174	6.8432	470	8.2706
38	4.6792	88	5.8687	176	6.8596	480	8.3009
39	4.7157	89	5.8848	178	6.8758	490	8.3305
40	4.7513	90	5.9007	180	6.8918	500	8.3596
41	4.7861	91	5.9164	182	6.9076	550	8.4968
42	4.8200	92	5.9320	184	6.9233	600	8.6220
43	4.8532	93	5.9474	186	6.9388	650	8.7373
44	4.8856	94	5.9627	188	6.9541	700	8.8440
45	4.9173	95	5.9778	190	6.9693	750	8.9434
46	4.9483	96	5.9927	192	6.9843	800	9.0363
47	4.9787	97	6.0075	194	6.9992	850	9.1236
48	5.0084	98	6.0221	196	7.0139	900	9.2060
49	5.0375	99	6.0366	198	6.0284	950	9.2839
50	5.0661	100	6.0510	200	6.0429	1000	9.3578

waters (Mathis 1968; Wilhm and Dorris 1968; Wilhm 1970; Lloyd and Ghelardi 1964). Published values for Rocky Mountain streams (table 16) are sparse, but generally approach or exceed 3.

It also may be of interest to calculate the equitability or evenness of allotment of individuals among taxa. Equitability (e) can be calculated in several ways but a common method is as follows:

$e = \frac{s^*}{s}$ where s is the number of species actually collected and s* is a hypothetical number of species and may be obtained from table 16 for any given value of H′. Equitability is thought to be more sensitive than H′ to slight or moderate levels of degradation (Weber 1973). Values range between 0 and 1. Those values less than 0.5 are considered to characterize macroinvertebrate communities in relatively natural streams (Weber 1973). The few values published for Rocky Mountain streams range from 0.1 to 0.5 (table 17).

Redundancy (r) is a measure of the dominance of one or more taxa and is inversely proportional to the variety of species. It is calculated as:

$$r = \frac{H'_{max} - H'}{H'_{max} - H'_{min}}.$$

The theoretical maximum diversity and the minimum diversity, H'_{max} and H'_{min}, and are calculated as:

$$H'_{max} = \frac{\log_2 n! - s \log_2 \frac{n}{s}!}{n}$$

$$H'_{min} = \frac{\log_2 n! - \log_2 (n+s+1)!}{n}$$

The Shannon-Weiner Diversity Index and other measures derived from it have been widely criticized as inappropriate for detecting the impact of pollution and other types of environmental stress (for example, Peet 1974, 1975; Cook 1976; Zand 1976; Pielou 1975, 1977). Thus, the procedure should be used with caution if at all.

Biotic Indexes

Of the various biotic indexes that have been proposed for use with macroinvertebrates, two deserve attention here: the Biotic Condition Index and the Chandler Biotic Score. Each approach has its shortcomings. The BCI does not include a measure of relative abundance, while the CBS is based on subjective tolerance ratings. In practice, both systems are subject to user biases and previous experience, especially when taxa are encountered that were not included in the original system and when species within an order, family, or genus have quite different tolerances.

The Biotic Condition Index (Winget and Mangum 1979) currently is being advocated for use by Forest Service (Intermountain Region) personnel. Other than the data on which the Index was developed, no other results of its use have been published.

The BCI incorporates stream habitat (gradient, substrate composition), water quality (alkalinity, sulfate), and environmental tolerances of aquatic macroinvertebrate species. It is a function of a Predicted Community Tolerance Quotient (CTQ_p) divided by the Actual Community Tolerance Quotient (CTQ_a). The tolerance quotient (TQ) is the product of values derived from the taxon's tolerance to levels of alkalinity and sulfate plus its selectivity for or against fine substrate materials and low stream gradients. Values range from 2 to slightly greater than 100 with the larger values indicating greater tolerance. The TQ's have been determined for 54 taxa and values assigned to an additional 317 (appendix 7). The CTQ_p is the mean of the TQ's for a predicted macroinvertebrate community. To obtain a CTQ_p for a particular stream segment, the station is classified according to the criteria given above (appendix 8). A CTQ_a is simply a mean of the TQ's of the macroinvertebrates collected from any station on any given date. The Biotic Condition Index is calculated as:

$$BCI = \frac{CTQ_p}{CTQ_a} \times 100.$$

Values are expressed as percent of expected value.

In the Chandler Biotic Score system (Chandler 1970), the taxa are rated from intolerant to highly tolerant. The intolerant species have values near 100 and the highly tolerant species have values near 0. The score is adjusted over a 10-point range depending on relative abundance. In the original system, the numerical value for

Table 18. — Comparison of various indexes of pollution for selected stations on the Portneuf River (Minshall and Andrews 1973); Biotic Condition Index and Chandler Biotic Score values were obtained from Frazier and others (1980)

Indexes	Station 2	Station 5	Station 8	Station 9b
Abundance ($\bar{x}$ number/sampler)	373	2,691	63	32
Richness	28	26	13	10
Diversity (H′)	2.6	1.3	1.9	1.6
Equitability (e)	.3	.1	.4	.4
BCI $\bar{x}$ (n=5) SD	117.2 (±4.8)	107.0 (±8.1	61.2 (±1.1)	64.4 (±2.6)
CBS	58.6	53.6	48.5	38.6

each taxon was simply added to the summed values for all taxa, but this gave a wide range of scores from less than 100 to several thousand. Cook (1976) modified the system by dividing the score by the number of taxa. This produced a linear scale of values between 0 and 100, decreasing with an increase in environmental stress. The modified Chandler Biotic Score is obtained by assigning each taxon (s) in a sample a rating (R) based on its taxonomic status and relative abundance (appendix 9). These ratings are then summed and divided by the total number of taxa present:

$$CBS = \frac{\sum_{i=1}^{s} R_i}{s}.$$

Comparison of Indexes

The different indexes discussed above were calculated for several stations on the Portneuf River, Idaho, subjected to varying but generally increasing degrees of environmental stress. The results (table 18) are based on data given by Minshall and Andrews (1973). The indexes all show the same basic trend suggesting a progressive decrease in water quality proceeding downstream from stations 2 through 9b. The principal variant is H′, which was strongly influenced by a disproportionately large number of the dipteran *Simulium* (76 percent of total). The BCI values closely follow those of richness and suggest a much larger deterioration in water quality at the two downstream stations than is reflected by the CBS values. On the other hand, the BCI values indicate that the two upper stations are at or near their potential while the CBS values show that considerable deterioration has occurred even at those sites. To this extent, the H′ and CBS values are in accord. Based on the data currently available, it is not possible to conclusively determine which biotic index best reflects actual conditions. Stations 2 and 5 are known to be impacted upstream by various agricultural practices (xylene control of macrophytes, grazing, and irrigation uses). But, on the other hand, the Portneuf River in the vicinity of station 2 is considered a "blue-ribbon" trout stream. Neither index shows much of an impact of dewatering by irrigation diversion at station 5, yet the quality of that area as a summer fishery habitat clearly is degraded by this activity.

CONCLUSIONS

Much of the literature on evaluation of the stream environment lies hidden in unpublished reports. In addition, when a resouce manager attacks a stream problem to determine its solution, it is necessary to start from scratch. The attributes to be measured must be selected and procedures devised. Sometimes these are successful, but many times they are not. Progress has largely been by trial and error with no source of standard measures and procedures available for guidance.

This manual is an attempt to draw together and describe a comprehensive set of routine measurements for use by resource managers in evaluating and/or monitoring conditions in and adjacent to streams. In addition, we have tried wherever possible to evaluate and assess the reliability attainable with the various measurements. It has not been possible to do this in all cases, but we hope to move closer to that goal in subsequent versions of this manual. Other manuals on flowing water methods are available to evaluate: stream morphology (USDA Forest Service 1975); streamflow effects (Stalnaker and Arnette 1976); stream bank stability (Cooper 1976); and general stream conditions (Duff and Cooper 1978). But, these deal with isolated aspects of the stream-riparian *milieu* and exclude the biotic component of lotic ecosystems. In addition, they fail to provide alternative approaches from which to select the most appropriate measurements for a given situation and they do not indicate the limitations of the recommended procedures. Others (Rickert and others 1978; USDI Bureau of Land Management 1973) have suggested a subjective approach to stream evaluation largely for purposes of economy. These methods may work for the specific purpose they were designed, but they are often inadequate if the objectives change. The underlying problem with these types of intuitive methods is that from different perspectives, which cause different interpretations, the same stream can be evaluated differently.

Although this report places measures of reliability on many of the attributes, it does not give the reliability that can be expected from the complete family of attributes selected to characterize stream conditions. This can come with experience only and will depend on the objectives of the study. Much thought must be given to selecting the family of attributes to be measured for they must cover those states or changes in states that actually control the density and composition of the populations of interest. The biotic resource itself plays an important part in becoming a component in the family of attributes. Not only does it help to ascertain what the enviromental conditions are at the time of sampling but also what they were prior to sampling.

Stream evaluation methods are not perfect, nor will they be perfect in the near future. They will not do all things for all purposes. Therefore, such methods need constant refinement and new and better techniques must be developed. In addition, the reaction of biotic resources to environmental changes must be defined. These goals are some distance off, but we hope this manual hastens their accomplishment.

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APPENDIX 1

The t Distribution

Table 19. — Student's t distribution[1]

				p[3]				
v[2]	0.60	0.75	0.90	0.95	0.975	0.99	0.995	0.9995
1	0.325	1.000	3.078	6.314	12.706	31.821	63.657	636.619
2	.289	.816	1.886	2.920	4.303	6.965	9.925	31.598
3	.277	.765	1.638	2.353	3.182	4.541	5.841	12.941
4	.271	.741	1.533	2.132	2.776	3.747	4.604	8.610
5	.267	.727	1.476	2.015	2.571	3.365	4.032	6.859
6	.265	.718	1.440	1.943	2.447	3.143	3.707	5.959
7	.263	.711	1.415	1.895	2.365	2.998	3.499	5.405
8	.262	.706	1.397	1.860	2.306	2.896	3.355	5.041
9	.261	.703	1.383	1.833	2.262	2.821	3.250	4.781
10	.260	.700	1.372	1.812	2.228	2.764	3.169	4.587
11	.260	.697	1.363	1.796	2.201	2.718	3.106	4.437
12	.259	.695	1.356	1.782	2.179	2.681	3.055	4.318
13	.259	.694	1.350	1.771	2.160	2.650	3.012	4.221
14	.258	.692	1.345	1.761	2.145	2.624	2.977	4.140
15	.258	.691	1.341	1.753	2.131	2.602	2.947	4.073
16	.258	.690	1.337	1.746	2.120	2.583	2.921	4.015
17	.257	.689	1.333	1.740	2.110	2.567	2.898	3.965
18	.257	.688	1.330	1.734	2.101	2.552	2.878	3.922
19	.257	.688	1.328	1.729	2.093	2.539	2.861	3.883
20	.257	.687	1.325	1.725	2.086	2.528	2.845	3.850
21	.257	.686	1.323	1.721	2.080	2.518	2.831	3.819
22	.256	.686	1.321	1.717	2.074	2.508	2.819	3.792
23	.256	.685	1.319	1.714	2.069	2.500	2.807	3.767
24	.256	.685	1.318	1.711	2.064	2.492	2.797	3.745
25	.256	.684	1.316	1.708	2.060	2.485	2.787	3.725
26	.256	.684	1.315	1.706	2.056	2.479	2.779	3.707
27	.256	.684	1.314	1.703	2.052	2.473	2.771	3.690
28	.256	.683	1.313	1.701	2.048	2.467	2.763	3.674
29	.256	.683	1.311	1.699	2.045	2.462	2.756	3.659
30	.256	.683	1.310	1.697	2.042	2.457	2.750	3.646
40	.255	.681	1.303	1.684	2.021	2.423	2.704	3.551
60	.254	.679	1.296	1.671	2.000	2.390	2.660	3.460
120	.254	.677	1.289	1.658	1.980	2.358	2.617	3.373
∞	.253	.674	1.282	1.645	1.960	2.326	2.576	3.291

[1]Reprinted with permission from *Statistical Tables for Biological, Agricultural, and Medical Research* (6th ed., 1974) written by Sir Ronald A. Fisher and Dr. Frank Yates and published by Longman Group Ltd., London (previously published by Oliver & Boyd Ltd., Edinburgh).

[2]V = degrees of freedom.

[3]P = probability.

APPENDIX 2
Transect Spacing

For general, broad-base planning purposes or general studies that cover large land areas and do not need refined information, the 200-ft (61-m) transect spacing would be adequate for making general interpretations from the data collected. To determine habitat conditions for making project decisions at the project level (for example, the Ranger District level), the 50-ft (15.2 m) spacing is probably adequate. For research purposes or for intensive studies, such as determining influences from point source pollution or for answers needing high accuracy in the results, the transects should probably be no more than 10 ft (3.0 m) apart.

To determine the reliability of different transect spacings, four streams were selected for testing. Two were in the Blackfoot River drainage of eastern Idaho and two in the central Idaho Batholith. At selected sites on each stream, 181 transects were set in at 10-ft (3.0-m) intervals. The habitat attribute means, standard deviation (S) around the mean, and auto correlation coefficients (AC) were calculated at transect interval spacings from 10 to 200 ft (3.1 to 61 m) over the same reach of stream. Thus, at the 10-ft spacings, 181 data entries were used to determine the mean of each habitat variable. At 50-ft spacings, only 36 data entries were used. The means derived from the 10-ft spacing were assumed to be the true means.

Blackfoot River Drainage Tests

In the Blackfoot River drainage, those habitat measurements (table 20) having low values or those conditions that seldom occur in the study area, such as large boulder (>2 ft [>0.61 m]) in the channel, were often missed at spacings above 30 ft (9.1 m).

Table 20. — Means and standard deviations (S) for percent channel composed of large boulders for selected transect spacings in Diamond Creek, Idaho

	Spacing				
	10	20	30	40	50
			Feet		
Mean	0.08	0.07	0.01	0.0	0.0
S	0.06	0.06	0.09	0.0	0.0

The standard deviations in most habitat measurements increased as transect spacing increased, whereas the autocorrelation (correlation between individual values of a variable) coefficients decreased. The greatest differences occurred in stream width, with means about 100 percent greater at 50-ft (15.2-m) spacing than at the 10-ft (3.0-m) spacing (table 21).

Table 21. — Means, standard deviations (S), and autocorrelations (AC) for stream width for selected transect spacings in Diamond Creek

	Spacing				
	10	20	30	40	50
			Feet		
Mean	7.53	7.57	7.97	7.46	14.46
S	3.18	3.63	4.32	6.04	4.94
AC	0.28	0.18	0.24	0.15	0.20

Habitat measurement means tended to vary as spacing distance increased, especially after the 30-ft (9.1-m) spacing. A habitat variable mean derived from the largest (50-ft) (15.2-m) spacing, however, was often very close to the mean derived from the smallest (10-ft) (3.0-m) spacing (table 22).

Table 22. — Means, standard deviations (S), and autocorrelations (AC) for stream width for selected transect spacings in Angus Creek

	Spacing				
	10	20	30	40	50
			Feet		
Mean	16.56	16.33	16.28	16.33	16.86
S	5.90	6.27	5.83	5.88	5.65
AC	.48	.38	.21	.18	.18

In Angus Creek, mean stream width and its standard deviation remained about the same as spacing increased, whereas the autocorrelation decreased. This shows that the variation of stream width between the individual transects increased as spacing increased and sample size decreased.

Salmon River Drainage Tests

Stream reaches on Frenchman Creek and the South Fork Salmon River were studied each year from 1976 to 1980. Each reach covered 1,800 ft (548.6 m) of stream and was blanketed by 181 equal distance transects. The mean of each habitat variable was determined from transects spaced 10 ft (3.0 m), 20 ft (6.1 m), 30 ft (9.1 m), 40 ft (12.2 m), 50 ft (15.2 m), 100 ft (30.5 m) and 200 ft (61.0 m) apart. Again, as was the case in the Blackfoot River drainage, it was remarkable how often the mean derived from the 200-ft (61.0-m) spacing was about the same as the mean derived from the 10-ft (3.0-m) spacing. However, on certain habitat measurements the mean would be completely off target at the 200-ft (61.0-m) spacing.

As spacing between transects increased, the standard deviation and confidence intervals around the means increased. In measuring stream width over a 5-year period on the same reach in the South Fork Salmon River, confidence intervals around the means were about five times as wide at the 200-ft (61.0-m) spacing as at the 10-ft (3.0-m) spacing. In the Frenchman Creek reach, the confidence interval was two to three times as wide at the 200-ft (61.0-m) spacing. Stream depth also followed this pattern.

Percent riffle in the channel (table 23), because it varied considerably, was not determined with confidence when transect spacing exceeded 50 ft (15.2 m).

Table 23. — Means and percent confidence interval (CI) about the mean at the 95 percent confidence level for percent riffle in the South Fork Salmon River reach in 1980

	Spacing							
	10	20	30	40	50	60	100	200
				Feet				
Mean	27	27	30	27	31	33	36	38
CI	16	23	28	32	38	37	44	53

APPENDIX 2 (con.)

Some habitat variables, such as the pool quality rating (discussed later) were read quite accurately at the 200-ft (61.0-m) transect spacing (table 24). Confidence intervals, however, became much wider due mainly to the smaller sample size.

Table 24. — Means and percent confidence interval (CI) about the mean at the 95 percent confidence for pool quality in the Frenchman Creek reach in 1980

	Spacing							
	10	20	30	40	50	60	100	200
	Feet							
Mean	3.9	4.0	4.0	4.1	4.0	3.9	4.1	4.0
CI	4	4	5	4	8	8	11	10

APPENDIX 3

Examples of Accuracy, Precision, and Confidence Intervals

(Attribute Means from 1,800-ft [549-m] Study Reaches for One-Time Measurements)

Table 25. — Accuracy, precision, and confidence interval for stream width

Stream	Mean width	Confidence interval	Precision	Accuracy
	Feet	*± Percent*		
Horton Creek	4.2	6.6	Good	Excellent
Gance Creek	5.6	5.0	Excellent	Good
Frenchman Creek	11.5	5.8	Good	Good
Johnson Creek	9.5	5.1	Good	Good
South Fork Salmon River	15.6	4.7	Excellent	Good
Elk Creek	30.3	5.2	Good	Good

Table 26. — Accuracy, precision, and confidence intervals for stream depth

Stream	Mean depth	Confidence interval	Precision	Accuracy
	Feet	*± Percent*		
Horton Creek	0.4	6.8	Good	Excellent
Gance Creek	.2	9.2	Good	Good
Frenchman Creek	.8	7.8	Good	Good
Johnson Creek	.8	8.9	Good	Good
South Fork Salmon River	.8	8.0	Good	Good
Elk Creek	1.1	8.3	Good	Excellent

Table 27. — Accuracy, precision, and confidence intervals for stream shore water depth

Stream	Mean depth	Confidence interval	Precision	Accuracy
	Feet	*± Percent*		
Horton Creek	0.2	19.8	Fair	Good
Gance Creek	.3	26.6	Poor	Fair
Frenchman Creek	.5	13.2	Fair	Fair
Johnson Creek	.3	16.5	Fair	Fair
South Fork Salmon River	.5	10.6	Good	Poor
Elk Creek	.3	12.9	Fair	Good

Table 28. — Accuracy, precision, and confidence intervals for percent pool

Stream	Mean pool	Confidence interval	Precision	Accuracy
	Percent	*± Percent*		
Horton Creek	25.9	20.7	Poor	Fair
Gance Creek	34.4	13.5	Fair	Fair
Frenchman Creek	72.7	7.0	Good	Poor
Johnson Creek	76.3	6.1	Good	Poor
South Fork Salmon River	70.5	6.8	Good	Poor
Elk Creek	68.1	7.4	Good	Poor

Table 29. — Accuracy, precision, and confidence intervals for pool quality

	Mean pool quality	Confidence interval	Precision	Accuracy
		± Percent		
Horton Creek	2.5	11.6	Fair	Fair
Gance Creek	2.2	10.3	Fair	Poor
Frenchman Creek	3.7	6.2	Good	Poor
Johnson Creek	3.7	7.5	Good	Fair
South Fork Salmon River	4.0	6.8	Good	Fair
Elk Creek	4.0	5.4	Good	Good

Table 30. — Accuracy, precision, and confidence intervals for percent riffle

Stream	Mean riffle	Confidence interval	Precision	Accuracy
	Percent	*± Percent*		
Horton Creek	74.0	6.5	Good	Fair
Gance Creek	65.5	6.8	Good	Poor
Frenchman Creek	27.3	19.2	Fair	Poor
Johnson Creek	23.7	20.4	Fair	Poor
South Fork Salmon River	30.0	16.6	Fair	Poor
Elk Creek	30.3	5.2	Good	Poor

Table 31. — Accuracy, precision, and confidence intervals for sun angle

Stream	Sun arc angle	Confidence interval	Precision	Accuracy
	Degrees	*± Percent*		
Horton Creek	—	—	—	—
Gance Creek	—	—	—	—
Frenchman Creek	122.4	1.5	Excellent	Good
Johnson Creek	148.2	.4	Excellent	Poor
South Fork Salmon River	109.2	4.0	Excellent	Excellent
Elk Creek	163.0	.6	Excellent	Poor

APPENDIX 3 (con.)

Table 32. — Accuracy, precision, and confidence intervals for streambank soil alteration

Stream		Streambank alteration	Confidence interval	Precision	Accuracy
		Percent	*± Percent*		
Horton Creek	Natural	8.9	12.0	Fair	Good
	Artificial	22.7	8.8	Good	Good
Gance Creek	Natural	31.0	6.0	Good	Fair
	Artificial	13.3	13.5	Fair	Poor
Frenchman Creek	Natural	20.7	11.5	Fair	Fair
	Artificial	5.0	24.6	Poor	Poor
Johnson Creek	Natural	15.8	10.8	Fair	Fair
	Artificial	12.2	13.7	Fair	Poor
South Fork Salmon River	Naturai	21.2	12.4	Fair	Poor
	Artificial	7.2	15.3	Fair	—
Elk Creek	Natural	25.6	7.9	Good	Good
	Artificial	14.1	10.6	Fair	Poor

Table 33. — Accuracy, precision, and confidence intervals for streambank vegetative stability

Stream	Streambank vegetative stability	Confidence interval	Precision	Accuracy
	Units	*± Percent*		
Horton Creek	3.3	2.2	Excellent	Fair
Gance Creek	1.8	5.7	Good	Fair
Frenchman Creek	3.3	2.5	Excellent	Good
Johnson Creek	3.3	2.4	Excellent	Good
South Fork Salmon River	3.5	2.3	Excellent	Fair
Elk Creek	2.8	3.5	Excellent	Fair

Table 34. — Accuracy, precision, and confidence intervals for streambank undercut

Stream	Streambank undercut	Confidence interval	Precision	Accuracy
	Feet	*± Percent*		
Horton Creek	0.1	20.8	Poor	Good
Gance Creek	.08	30.5	Poor	Fair
Frenchman Creek	.5	15.2	Fair	Poor
Johnson Creek	.3	16.1	Fair	Poor
South Fork Salmon River	.4	14.2	Fair	Good
Elk Creek	.5	13.9	Fair	Good

Table 35. — Accuracy, precision, and confidence intervals for streambank angle

Stream	Channel bank angle	Confidence interval	Precision	Accuracy
	Degrees	*± Percent*		
Horton Creek	107.7	3.9	Excellent	Good
Gance Creek	118.5	3.7	Excellent	Good
Frenchman Creek	97.5	4.2	Excellent	Good
Johnson Creek	97.7	4.8	Excellent	Poor
South Fork Salmon River	103.9	6.6	Good	Good
Elk Creek	103.7	3.2	Excellent	Good

Table 36.—Accuracy, precision, and confidence intervals for streamside cover

Stream	Streamside cover	Confidence interval	Precision	Accuracy
	Units	*± Percent*		
Horton Creek	2.3	3.2	Excellent	Good
Gance Creek	2.2	5.8	Good	Poor
Frenchman Creek	2.1	3.5	Excellent	Poor
Johnson Creek	2.4	3.4	Excellent	Poor
South Fork Salmon River	2.3	4.1	Excellent	Poor
Elk Creek	2.0	4.4	Excellent	Poor

APPENDIX 3 (con.)

Table 37.—Accuracy, precision, and confidence intervals for vegetation use

Stream	Vegetation use	Confidence interval	Precision	Accuracy
	Percent	± *Percent*		
Horton Creek	29.8	5.8	Good	Excellent
Gance Creek	44.9	8.5	Good	Good
Frenchman Creek	11.1	32.5	Poor	Good
Johnson Creek	25.5	9.2	Good	Good
South Fork Salmon River	8.6	1.5	Excellent	Good
Elk Creek	31.7	14.7	Fair	Good

Table 38. — Accuracy, precision, and confidence intervals for vegetation overhang

Stream	Vegetation overhang	Confidence interval	Precision	Accuracy
	Feet	± *Percent*		
Horton Creek	0.5	8.3	Good	Poor
Gance Creek	.1	33.1	Poor	Poor
Frenchman Creek	.6	14.0	Fair	Good
Johnson Creek	.6	13.4	Fair	Poor
South Fork Salmon River	.8	13.5	Fair	Good
Elk Creek	.5	12.0	Fair	Good

Table 39. — Accuracy, precision, and confidence intervals for habitat type

Stream	Streamside habitat type	Confidence interval	Precision	Accuracy
	Units	± *Percent*		
Horton Creek	16.3	1.7	Excellent	Good
Gance Creek	9.4	6.9	Good	Good
Frenchman Creek	17.0	3.0	Excellent	Good
Johnson Creek	17.6	2.6	Excellent	Fair
South Fork Salmon River	14.9	3.3	Excellent	Poor
Elk Creek	13.5	4.2	Excellent	Fair

Table 40. — Accuracy, precision, and confidence intervals for fish environment

Stream	Fish environment rating	Confidence interval	Precision	Accuracy
	Units	± *Percent*		
Horton Creek	2.6	3.6	Excellent	Good
Gance Creek	1.5	7.4	Good	Poor
Frenchman Creek	3.3	4.7	Excellent	Good
Johnson Creek	3.5	4.7	Excellent	Good
South Fork Salmon River	3.2	5.3	Good	Poor
Elk Creek	3.5	3.8	Excellent	Poor

Table 41. — Accuracy, precision, and confidence intervals for boulder

Stream	Boulder	Confidence interval	Precision	Accuracy
	Percent	± *Percent*		
Horton Creek	0.0	0.0	Excellent	Excellent
Gance Creek	2.1	48.1	Poor	Good
Frenchman Creek	1.2	67.5	Poor	Good
Johnson Creek	.0	.0	Excellent	Excellent
South Fork Salmon River	1.5	30.1	Poor	Excellent
Elk Creek	.1	99.4	Poor	Good

Table 42. — Accuracy, precision, and confidence intervals for rubble

Stream	Rubble	Confidence interval	Precision	Accuracy
	Percent	± *Percent*		
Horton Creek	2.3	83.7	Poor	Fair
Gance Creek	9.3	29.5	Poor	Fair
Frenchman Creek	2.8	49.0	Poor	Good
Johnson Creek	.0	.0	Excellent	Excellent
South Fork Salmon River	8.8	25.1	Poor	Good
Elk Creek	8.1	27.9	Poor	Poor

Table 43. — Accuracy, precision, and confidence intervals for gravel

Stream	Gravel	Confidence interval	Precision	Accuracy
	Percent	± *Percent*		
Horton Creek	74.1	6.4	Good	Fair
Gance Creek	73.8	5.4	Good	Fair
Frenchman Creek	58.9	7.8	Good	Poor
Johnson Creek	53.9	7.5	Good	Poor
South Fork Salmon River	47.1	7.1	Good	Good
Elk Creek	76.2	3.9	Excellent	Poor

APPENDIX 3 (con.)

Table 44. — Accuracy, precision, and confidence intervals for fine sediment

Stream		Percent fine sediment	Confidence interval	Precision	Accuracy
			± Percent		
Horton Creek	large	8.6	51.9	Poor	Fair
	fine	16.5	29.1	Poor	
Gance Creek	large	4.3	43.5	Poor	Fair
	fine	9.0	28.3	Poor	
Frenchman Creek	large	25.4	15.6	Fair	Poor
	fine	26.0	14.0	Fair	
Johnson Creek	large	22.9	13.2	Fair	Poor
	fine	23.7	15.0	Fair	
South Fork Salmon River	large	21.2	12.0	Fair	Good
	fine	21.6	12.5	Fair	
Elk Creek	large	4.5	30.0	Poor	Poor
	fine	10.3	4.7	Excellent	

Table 45. — Accuracy, precision, and confidence intervals for embeddedness

Stream	Embeddedness	Confidence interval	Precision	Accuracy
	Units	*± Percent*		
Horton Creek	3.5	4.3	Excellent	Fair
Gance Creek	3.3	4.6	Excellent	Fair
Frenchman Creek	2.6	6.4	Good	Excellent
Johnson Creek	2.7	5.4	Good	Good
South Fork Salmon River	2.6	7.7	Good	Poor
Elk Creek	3.5	3.7	Excellent	Good

Table 46. — Accuracy, precision, and confidence intervals for instream vegetative cover

Stream	Instream vegetative cover	Confidence interval	Precision	Accuracy
	Feet	*± Percent*		
Horton Creek	0.6	24.2	Poor	Fair
Gance Creek	.2	42.1	Poor	Poor
Frenchman Creek	.7	31.4	Poor	Good
Johnson Creek	.3	40.5	Poor	Good
South Fork Salmon River	6.5	8.1	Good	Poor
Elk Creek	5.5	11.0	Fair	Good

APPENDIX 4

Mathematical Proof of Needed Stream Depth Measurements

Given: A channel cross section underneath the transect, with water depths measured at one-fourth, one-half, and three-fourths the distance of the width of water. What is the average depth?

Cross section area = width × depth

so

$$\text{Average depth } (\bar{D}) = \frac{\text{Area (A)}}{\text{Width (W)}},$$

but the total area (A) and total width (W) are equal to the sum of the areas of the four parts of the cross section defined by the three depth measurements so

$$\bar{D} = \frac{A}{W} \sum_{i=1}^{4} \frac{\left(\frac{D_i + D_{i+1}}{2}\right)\left(\frac{W}{4}\right)}{W} = \sum_{i=1}^{4} \frac{D_i + D_i + 1}{8}$$

$$\bar{D} = \frac{D_1 + D_2 + D_2 + D_3 + D_3 + D_4 + D_4 + D_5}{8}$$

$$\bar{D} = \frac{D_1 + 2D_2 + 2D_3 + 2D_4 + D_5}{8},$$

but D_1 and $D_5 = 0$; therefore

$$\bar{D} = \frac{2D_2 + 2D_3 + 2D_4}{8} = \frac{D_2 + D_3 + D_4}{4}.$$

APPENDIX 5

Stream Habitat and Fishery Rating Variables that Failed to Show Promise

BANK-TO-BANK WIDTH

The bank-to-bank width is the distance from the top of the right streambank along the transect line to the top of the left streambank. The top of the bank is usually at that point where the verti cal slope of the bank sloping away from the water column changes to a horizontal slope. This measurement was recorded to the nearest foot (0.31 m), but for more accuracy it should be recorded in tenths of feet (0.03 m). After 3 years of measuring this attribute, we concluded we could not measure bank-to-bank width with precision with this approach because of the inability of the observers to accurately select the two points representing the top of the banks.

Confidence intervals around the means were extremely large and year-to-year precision and accuracy in the measurements rated very low. If this measurement is needed, it should be done in combination with the cross section profile, which allows the points to be permanently set or accurately determined.

HIGH WATER STREAM WIDTH

This measurement is taken as the measurement for existing water stream width, except that the high water measurement begins at the high water mark on one bank and ends at the high water mark on the opposite bank. This measurement was recorded to the nearest foot (0.31 m), but should be measured to a tenth of a foot (0.03 m). After 3 years of testing we concluded this measurement could not be taken accurately using this method. High water marks were constantly changing, were hard to define, and on some stream reaches within broad flat flood plains where the stream averaged 30 ft (9.1 m) in width during low flows, the high water stream width could average over 1,200 ft (366 m) or more. If this measurement is needed, it should be obtained by onsite checks during the high flow period to correctly mark the high water points on both banks.

SUN ANGLE

The angle made by the arc of the sun as it intercepts the midpoint of the transect is measured with a clinometer. The angle of the arc is easily determined by the day of the year. For uniformity, we used the sun's arc on August 1 for all measurements taken during the season.

The sum of the two clinometer readings that measure the angle on each side of the stream from the channel horizontal to the sun horizon are subtracted from 180 to obtain the sun arc degrees (fig. 27). Examples of conditions intercepting the rays and reducing the degrees of the arc are streamside vegetation, logs, debris, bridges, trees, high streambanks, and narrow canyons. We found this measurement correlated well with fish standing crop in our higher elevation streams with increasing sun arc resulting in increasing fish standing crop. The measurement had good year-to-year accuracy and precision rating and had narrow confidence intervals. Even with these merits, which are hard to find in most attributes, we are still not sure how to handle these data after they are collected.

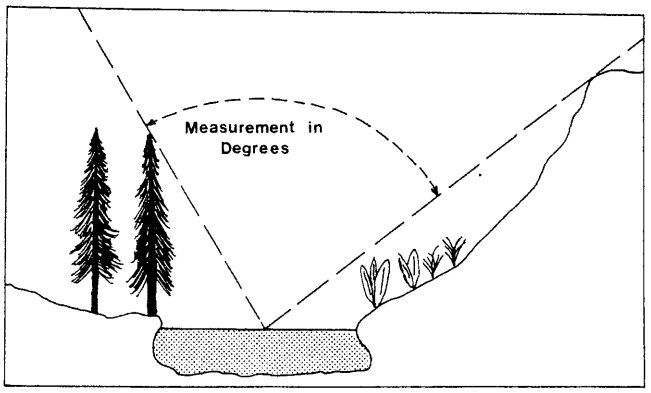

Figure 27. — Measurement of sun arc degrees.

STREAMBANK ROCK CONTENT

Streambanks intercepted by the transect line are evaluated for percent rock content. We found it difficult to rate rock content because streambank cover makes it impossible to determine the true composition of the bank. Only by digging soil pits could we get an accurate estimate because the streambank materials usually are lensed and change drastically in composition from one spot to another. We finally dropped this rating because of the difficulty in getting reliable data without digging pits. The rock content of the exposed channel is available from the substrate analysis and would be more meaningful. Streambank rock content is rated as shown:

Rating	Description
5	Over 95 percent of the bank material is more than 0.19 inch (4.7 mm) in particle size. The majority of the material is boulder or rubble.
4	From 75 to 94 percent of the bank material is more than 0.19 inch in particle size. The majority of the material is boulder or rubble, but if the majority is gravel, the rate is 3.
3	From 50 to 74 percent of the bank material is more than 0.19 inch in particle size. The majority of the material is boulder or rubble, but if the majority is gravel, the rate is 2.
2	From 25 to 49 percent of the bank material is over 0.19 inch in particle size diameter, but if the majority of the material is gravel, the rate is 1.
1	Less than 25 percent of the bank material is over 0.19 inch, but if the majority of the material is rubble and boulder, the rate is 2.

FISH STREAMSIDE ENVIRONMENT

This evaluation includes cover mainly as it relates to catchable size fish (table 47). Young-of-the-year and other age groups of small fish are not adequately considered in this evaluation.

APPENDIX 5 (con.)

Table 47. — Fish streamside environment rating

Description	Go to block	Rating
1a Contact zone is pool	2	
1b Contact zone is riffle	5	
2a Contact zone pool rates 5	3	
2b Contact zone pool rates 3 or 4	4	
2c Contact zone pool rates 1 or 2	5	
3a Cover is abundant		5
3b Cover is intermediate		4
3c Cover is lacking		3
4a Cover is abundant		4
4b Cover is intermediate		3
4c Cover is lacking		2
5a Cover is abundant		3
5b Cover is intermediate		2
5c Cover is lacking		1

The area to be evaluated is the border between the streambank or channel and the shoreline water columns. Only that area intercepted by the transect line is evaluated, although areas outside of this are considered to obtain the pool quality and cover ratings. High rating values would be considered to indicate better conditions for catchable salmonids. We have not evaluated this rating to see if it is of any value in predicting fish populations.

CHANNEL STABILITY

Stream channel stability rates the channel as to whether it is stable, aggrading, or eroding. It is an estimate of the rate the channel moves horizontally or vertically. Stream channel stability is rated as shown:

Rating	Description
3	Stable
2	Aggrading
1	Eroding

The rating is based on subjective judgment, so after 2 years of use, observer error, and inability to determine if the channel had scoured or filled, we discarded it. The cross-section profile measurements can indicate channel stability, but will not determine if the channel is aggrading or degrading. The chain method does determine this.

APPENDIX 6

FPSP-AI: A BASIC Computer Program Designed for the Hewlett-Packard HP9845 that Calculates Population Estimates Using a Removal-Depletion Maximum-Likelihood Formula.

```
160   !       CALCULATIONS MADE       CORRESPONDING VARIABLE NAMES
170   !
180   !  1.   Population estimate                1. Popest
190   !  2.   Population estimate variance       2. Popsizvar
200   !  3.   Pop. est. standard error           3. Sepopsiz
210   !  4.   Pop. est. upper conf. interval     4. Upconfintpop
220   !  5.   Pop. est. lower conf. interval     5. Loconfintpop
230   !
240   !  6.   Capture probability estimate       6. Captprob = Phat
250   !  7.   Capt. prob. estimate variance      7. Varcaptprob
260   !  8.   Capt. prob. est. std. error        8. Secaptprob
270   !  9.   Capt. prob. est. up. conf. int.    9. Upconfintcapt
280   ! 10.   Capt. prob. est. lo. conf. int.   10. Loconfintcapt
290   !
300   ! 11.   Chi square goodness-of-fit         11.  Chisquare
310   !
320   !.....................................................
330   !  SECTION 1:  INPUT INFORMATION.
340   !  Asks the user to input the number of removals
350   !  [Numofrmvls] made and the number of fish caught per
360   !  removal [Numfishprrmvl(Rmvl)].  Total catch [S] and
370   !  a function [C] are calculated for use in the
380   !  population estimate computation.
390   !
400   DIM Expnumfish(4),Numfishprrmvl(4)
410   INPUT "ENTER THE NAME OF THE STREAM",Stream$
420   PRINT Stream$
430   INPUT "ENTER THE NAME OF THF SPECIES",Species$
440   PRINT Species$
450   INPUT "HOW MANY REMOVALS?",Numofrmvls
460   PRINT Numofrmvls
470   T=Numofrmvls
480   FOR Rmvl=1 TO Numofrmvls
490     PRINT "HOW MANY FISH CAUGHT IN REMOVAL";Rmvl;"?"
500     INPUT Numfishprrmvl(Rmvl)
510     PRINT Numfishprrmvl(Rmvl)
```

APPENDIX 6 (con.)

```
520       S=S+Numfishprrmvl(Rmvl)
530       C=C+Numfishprrmvl(Rmvl)*Rmvl
540     NEXT Rmvl
550     !
560     !.....................................................
570     !  SECTION 2:  SEARCH FOR POP.EST. OF HIGHEST PROBABILITY.
580     !  Population estimates are calculated for S+0, S+1, S+2,
590     !  etc., until the population function [Theta] reaches a
600     !  maximum point.  This point corresponds to the
610     !  population estimate of maximum liklihood.  The variable
620     !  [I] is used to increment [S] by one each time through
630     !  the loop.  A summation term [Firstterm] is defined as
640     !  zero when [I]=0.
650     !
660     Firstterm=I=Theta=Oldtheta=0        ! Initialize variables.
670     Phat=S/C
680     GOTO 760    ! Calculation of summation term (Firstterm) is
690                 ! skipped when I=0 to prevent division by zero.
700                 ! Firstterm is set initially to zero.
710     I=I+1
720     Phat=S/(C+T*I)
730     Firstterm=Firstterm+LOG(1+S/I)     ! LOG(X) takes the
740                                        ! natural log of X.
750     Oldtheta=Theta
760     Theta=Firstterm+S*LOG(Phat)+(C-S+T*I)*LOG(1-Phat)
770     IF (Oldtheta<Theta) OR (I=0) THEN 710  !Looks for Theta
780                                            !to reach a max.
790     !
800     !.....................................................
810     !  SECTION 3:  POPULATION ESTIMATE STATISTICS.
820     !  This section is entered when [Theta] reaches a maximum
830     !  point (i.e. the loop in SECTION 2 has been exited).
840     !  The statistics corresponding to the maximum liklihood
850     !  estimate are calculated.
860     !
870     Popest=I-1+S
880     Captprob=Phat=S/(C+T*(I-1))
890
Popsizvar=Popest*(1-Phat)**T*(1-(1-Phat)**T)/((1-(1-Phat)**T)**2
-(T*Phat)**2*(1-Phat)**(T-1))
900     Sepopsiz=SQR(Popsizvar)
910     Tvalue=1.96     ! The T-value is assumed to be 1.96
920     Confintpop=Tvalue*Sepopsiz
930     Upconfintpop=Popest+Confintpop
940     Loconfintpop=Popest-Confintpop
950     IF Loconfintpop<S THEN Loconfintpop=S
960     !
970     !.....................................................
980     !  SECTION 4:  CAPTURE PROBABILITY STATISTICS.
990     !  The capture probability statistics corresponding to the
1000    !  maximum liklihood estimate are calculated.
1010    !
```

APPENDIX 6 (con.)

```
1020  Varcaptprob=(Captprob/Popest)**2*(Popsizvar/(1-Captprob)**
(T-1))
1030  Secaptprob=SQR(Varcaptprob)
1040  Confintcapt=Tvalue*Secaptprob
1050  Upconfintcapt=Captprob+Confintcapt
1060  Loconfintcapt=Captprob-Confintcapt
1070  IF Loconfintcapt<0 THEN Loconfintcapt=0
1080  !
1090  !.....................................................
1100  !  SECTION 5:  CHI SQUARE CALCULATION.
1110  !  The expected number of fish caught is calculated for
1120  !  each removal [Expnumfish(Rmvl)] and for the total catch
1130  !  [Exptotnumfish].  These numbers are compared with the
1140  !  actual number of fish caught to yield the chi square
1150  !  [Chisquare] statistic.
1160  !
1170  Exptotnumfish=Totnumfishcot=Chisqsumterm=0
1180  FOR Rmvl=1 TO Numofrmvls
1190    Expnumfish(Rmvl)=Popest*(1-Captprob)**(Rmvl-1)*Captprob
1200    Chisqsumterm=Chisqsumterm+(Numfishprrmvl(Rmvl)-Expnumfis
h(Rmvl))**2/Expnumfish(Rmvl)
1210    Exptotnumfish=Exptotnumfish+Expnumfish(Rmvl)
1220    Totnumfishcot=Totnumfishcot+Numfishprrmvl(Rmvl)
1230  NEXT Rmvl
1240  Chisquare=Chisqsumterm+(Totnumfishcot-Exptotnumfish)**2/Fx
ptotnumfish
1250  !
1260  !.....................................................
1270  !  SECTION 6:  OUTPUT.
1280  !  The information calculated above is printed.  PRINT
1290  !  USING and IMAGE statements allow formatting where
1300  !
1310  !  X is a blank space,
1320  !  K is a string constant,
1330  !  A is a string character (strings are left-justified
1340  !       with blanks filling out the rest of the field),
1350  !  D is a digit position,
1360  !  Z is also a digit position (leading zeros are replaced
1370  !       with 0 as a fill character).
1380  !
1390  !  Prefix numbers refer to the number of occurrences.  For
1400  !  example, 7X specifies seven blank spaces.
1410  !
1420  PRINTER IS 0   ! This statement activates the printer.
1430  PRINT USING "K,31A,K,25A";"STREAM:  ",Stream$,"SPECIES:  "
,Species$
1440  PRINT
1450  PRINT USING "K,7X,K,5D";"TOTAL CATCH","=",S
1460  IMAGE K,4X,K,5D,16X,K,6X,K,Z.5D
1470  PRINT USING 1460;"POPULATION EST","=",Popest,"CAPTURE PROB
","= ",Captprob
1480  IMAGE K,3X,K,DZ.3D,14X,K,Z.5D
```

APPENDIX 6 (con.)

```
1490  PRINT USING 1480;"POP EST STD ERR","= ",Sepopsiz,"CAPT PRO
B STD ERR = ",Secaptprob
1500  IMAGE K,4DZ.2D,12X,K,Z.5D
1510  PRINT USING 1500;"LOWER CONF INTRVL = ",Loconfintpop,"LOWE
R CONF INTRVL = ",Loconfintcapt
1520  PRINT USING 1500;"UPPER CONF INTRVL = ",Upconfintpop,"UPPE
R CONF INTRVL = ",Upconfintcapt
1530  PRINT
1540  IMAGE K,8X,K,DZ.4D,13X,K,4(4D,X)
1550  PRINT USING 1540;"CHI SQUARE","= ",Chisquare,"REMOVAL PATT
ERN:  ",Numfishprrmvl(1),Numfishprrmvl(2),Numfishprrmvl(3),Numfi
shprrmvl(4)
1560  PRINTER IS 16     ! Printer is turned off.
1570  END
```

EXAMPLE OF FPSP-AI OUTPUT.

STREAM: So. Fk. Salmon R. SPECIES: Rainbow Trout

TOTAL CATCH	= 111		
POPULATION EST	= 116	CAPTURE PROB	= 0.53110
POP EST STD ERR	= 3.481	CAPT PROB STD ERR	= 0.04964
LOWER CONF INTRVL	= 111.00	LOWER CONF INTRVL	= 0.43381
UPPER CONF INTRVL	= 122.82	UPPER CONF INTRVL	= 0.62839
CHI SQUARE	= 0.2638	REMOVAL PATTERN:	60 30 15 6

EXAMPLE OF FPSP-AI INTERNAL CALCULATIONS.

I	SUMM. TERM	CAPTURE PROB	THETA
0	0.0	0.587301587302	-128.109045019
1	4.71849887128	0.575129533679	-126.871876966
2	8.75273950942	0.563451776650	-126.206675228
3	12.3903256691	0.552238805970	-125.833238175
4	15.7489634363	0.541463414634	-125.640520400
5	18.8931157150	0.531100478469	-125.570077691
6	21.8635301806	0.521126760563	-125.586621814

Note that Theta is maximized when I=5. The population estimate (116) equals the total catch (111) plus I (5).

APPENDIX 7

Tolerance Quotients of Aquatic Macroinvertebrates (from Winget and Mangum 1979)

Table 48. — Tolerance quotients (TQ) of aquatic macroinvertebrates based upon tolerance to alkalinity, sulfate, and sedimentation including low stream gradients

Taxa	TQ
Phylum Coelenterata	108
Class Hydrozoa	108
Phylum Aschelminthes	108
Class Nematoda	108
Phylum Mollusca	108
Class Gastropoda	108
Family Lymnaidae	108
Lymnaea	108
Family Physidae	108
Physa	108
Family Planorbidae	108
Phylum Annelida	108
Class Hirudinea	108
Class Oligochaeta	108
Family Tubificidae	108
Tubifex	108
Family Lumbricidae	108
Lumbricus aquaticus	108
Phylum Platyhelminthes	
Class Turbellaria	108
Order Tricladida	108
Phylum Arthropoda	
Class Arachnida	
Suborder Hydracarina	108
Class Crustacea	108
Order Isopoda	108
Family Asellidae	108
Asellus	108
Order Amphipoda	108
Family Talitridae	108
Hyalella azteca	108
Family Gammaridae	108
Gammarus lacustris	108
Order Decapoda	108
Family Astacida	108
Pacifastacus gambeli	108
Cambarus laevis	108
Order Cladocera	108
Daphnia	108
Order Copepoda	108
Order Ostracoda	108
Class Insecta	108
Order Collembola	108
Family Poduridae	108
Podura aquatica	108
Family Entomobryidae	108
Order Megaloptera	72
Family Sialidae	72
Sialis	72
Family Corydalidae	72
Corydalus cognata	72
Order Lepidoptera	72
Family Pyralidae	72
Parargyractis kearfottalis	72
Order Ephemeroptera	72
Family Siphlonuridae	72
Ameletus	48
Siphlonurus occidentalis	72
Isonychia	48
Family Baetidae	72
Baetis spp.	72
Callibaetis	72
Pseudocloeon	72
Centroptilum	36
Dactylobaetis	36
Paracloeodes	72
Family Oligoneuriidae	36
Lachlania saskatchewanensis	36
Homeoneuria	36
Family Heptageniidae	48
Heptagenia	48
Stenonema	48
Cinygmula	21
Rhithrogena	21
Epeorus	21
Anepeorus	48
Family Leptophlebiidae	36
Paraleptophlebia	24
Leptophlebia	24
Choroterpes	36
Traverella	36
Family Tricorythidae	108
Tricorythodes	108
Leptohyphes	72
Family Ephemerellidae	48
Ephemerella	48
Ephemerella grandis	24
Ephemerella doddsi	4
Ephemerella coloradensis	18
Ephemerella tibialis	24
Ephemerella inermis	48
Ephemerella infrequens	48
Ephemerella spinifera	24
Family Ephemeridae	36
Ephemera simulans	36
Hexagenia limbata	36
Family Caenidae	72
Caenis	72
Brachycerus	72
Family Polymitarcidae	48
Ephoron	48
Order Odonata	
Family Cordulegastridae	72
Cordulegaster	72
Family Gomphidae	108
Gomphus	108
Erpetogomphus compositus	72
Ophiogomphus severus	108
Progomphus borealis	72
Family Aeshnidae	72
Aeshna	72
Anax	72
Oplonaeschna	72
Family Libellulidae	72
Cordulia shurtleffi	72
Erythemis	72
Leucorrhinia	72
Libellula	72
Sympetrum	72
Somatochlora	72
Family Agrionidae	108
Hetaerina americana	108
Calopteryx	108

APPENDIX 7 (con.)

Taxa	TQ
Family Lestidae	108
Archilestes	108
Lestes	108
Family Coenagrionidae	108
Argia	108
Amphiagrion	72
Enallagma	72
Ischnura	72
Coenagrion	72
Telebasis salva	72
Order Hemiptera	
Family Belastomatidae	72
Belastoma	72
Benacus	72
Lethocerus	72
Abedus	72
Family Corixidae	108
Callicorixa	108
Hesperocorixa	108
Corisella	108
Trichocorixa	108
Cenocorixa	108
Graptocorixa	108
Arctocorixa	108
Sigara	108
Family Gerridae	72
Gerris	72
Rheumatobates	72
Family Naucoridae	72
Ambrysus mormon	72
Pelocoris	72
Family Notonectidae	108
Notonecta	108
Buenoa	108
Family Veliidae	72
Microvelia americana	72
Rhagovelia distincta	72
Family Mesoveliidae	72
Mesovelia	72
Family Macroveliidae	72
Macrovelia	72
Order Plecoptera	
Family Nemouridae	36
Amphinemura	6
Malenka	36
Prostoia besametsa	24
Podmosta	12
Zapada	16
Nemoura	24
Family Capniidae	32
Capnia	32
Eucapnopsis	18
Isocapnia	24
Mesocapnia frisoni	32
Utacapnia	18
Family Taeniopterygidae	48
Taenionema	48
Doddsia	24
Oemopteryx	48
Family Leuctridae	18
Paraleuctra	18
Perlomymia	18
Family Pteronarcyidae	24
Pteronarcella badia	24
Pteronarcys californica	18
Pteronarcys princeps	24
Family Perlodidae	48
Megarcys signata	24
Skwala parallela	18

Taxa	TQ
Cultus aestivalis	12
Isogenoides	24
I. elongatus	24
I. zionensis	24
Kogotus modestus	18
Pictetiella expansa	18
Diura knowltoni	24
Isoperla	48
I. ebria	24
I. fulva	48
I. mormona	48
I. quinquepunctata	48
Family Chloroperlidae	24
Family Perlidae	24
Acroneuria abnormis	6
Claassenia sabulosa	6
Hesperoperla pacifica	18
Perlesta placida	24
Doronuria theodora	18
Order Trichoptera	
Family Rhyacophilidae	18
Rhyacophila	18
Atopsyche	18
Himalopsyche	18
Family Glossosomatidae	32
Glossosoma	24
Anagapetus	24
Protoptila	32
Culoptila	32
Family Philopotamidae	24
Chimarra	24
Doliphilodes (sortosa)	24
Wormaldia	24
Family Psychomyidae	108
Polycentropus	108
Nyctiophylax	108
Psychomyia	108
Tinodes	108
Family Hydropsychidae	108
Hydropsyche	108
Cheumatopsyche	108
Arctopsyche	18
Smicridea	72
Diplectrona	48
Macronema	48
Parapsyche	6
Family Hydroptilidae	108
Hydroptila	108
Agraylea	108
Ochrotrichia	108
Neotrichia	108
Ithytrichia	108
Oxyethira	108
Leucotrichia	108
Alisotrichia	108
Mayatrichia	108
Family Limnephilidae	108
Limnephilus	108
Dicosmoecus	24
Hesperophylax	108
Oligophlebodes	24
Apatania	18
Amphicosmoecus	18
Neothremma	8
Lenarchus	18
Chyranda	18
Psychoglypha	24
Ecclisomyia	24
Homophylax	18

APPENDIX 7 (con.)

Taxa	TQ
Allocosmoecus	18
Asynarchus	108
Clistorania	108
Grammotaulius	108
Imania	48
Neophylax	24
Onocosmoecus	18
Pycnopsyche	72
Family Leptoceridae	54
Oecetis	54
Leptocella	54
Triaenodes	54
Mystacides	54
Ceraclea	54
Family Lepidostomatidae	18
Lepidostoma	18
Family Brachycentridae	24
Brachycentrus	24
Micrasema	24
Oligoplectrum	24
Amiocentrus	24
Family Helicopsychidae	18
Helicopsyche borealis	18
Family Polycentropodidae	72
Polycentropus	72
Nictiophylax	72
Family Sericostomatidae	72
Gumaga	72
Order Coleoptera	
Family Haliplidae	54
Brychius	54
Haliplus	54
Peltodytes	54
Family Dytiscidae	72
Derovatellus	72
Laccophilus	72
Bidessus	72
Agabus	72
Hygrotus	72
Hydroporous	72
Oreodytes	72
Hybius	72
Rhanius	72
Dytiscus	72
Acilius	72
Cybister	72
Deronectes	72
Thermonectus	72
Coptotomus	72
Family Hydrophilidae	72
Helophorous	72
Hydrochara	72
Berosus	72
Enochrus	72
Hydrophilus	72
Tropisternus	72
Hydrobius	72
Paracymus	72
Crenitis	72
Ametor	72
Helochares	72
Laccobius	72
Enochrous	72
Cymbiodyta	72
Family Elmidae	108
Zaitzevia	
Narpus	
Stenelmis	
Dubiraphia	
Optioservus	108
Heterlimnius	
Elmis	
Simsonia	
Microcylloepus	
Lara	
Family Cyrinidae	108
Gyrinus	108
Family Amphizoidae	24
Amphizoa	24
Family Hydraenidae	72
Order Diptera	
Family Tipulidae	72
Antocha monticola	24
Dicranota	24
Hexatoma	36
Holorusia grandis	72
Helobia	36
Tipula	36
Family Psychodidae	36
Maruina	36
Psychoda	36
Pericoma	36
Family Blephariceridae	2
Bibiocephala grandis	2
Agathon	2
Family Deuterophlebiidae	4
Deuterophlebia coloradensis	4
Family Culicidae	108
Aedes	108
Culex	108
Anopheles	108
Mansonia	108
Psorophora	108
Culiseta	108
Family Dixidae	108
Dixa	108
Family Simuliidae	108
Family Chironomidae	108
Family Ceratopogonidae	108
Family Stratiomyidae	108
Euparyphus	108
Family Tabanidae	108
Tabanus	108
Family Rhagionidae	24
Atherix pachypus	24
Family Dolichopodidae	108
Family Empididae	108
Hermerodromia	108
Family Ephydridae	108
Ephydra	108
Family Muscidae	108
Limnophora	108
Family Syrphidae	108
Chrysogastera	108
Tubifera	108
Helophilus	108

APPENDIX 8

A Key to Community Tolerance Quotients (from Winget and Mangum 1979)

Table 49.—A key giving Predicted Community Tolerance Quotients (CTQp) for various combinations of gradient (percent), substrates, total alkalinity as milligrams per liter calcium carbonate, and sulfate as milligrams per liter sulfate for any given stream

	Go to key number	CTQp
1. Stream gradient 0.1-1.2	2	
1.3-3.0	15	
3.0	28	
2. Substrate mostly boulder and rubble	3	
Gravel and rubble	7	
Sand and boulder, Rubble or gravel	11	
3. Total alkalinity 0-199	4	
200-300	5	
>300	6	
4. Sulfate 0-149		51
150-300		71
>300		90
5. Sulfate 0-149		53
150-300		71
>300		90
6. Sulfate 0-149		90
150-300		96
>300		108
7. Total alkalinity 0-199	8	
200-300	9	
>300	10	
8. Sulfate 0-149		53
150-300		85
>300		103
9. Sulfate 0-149		55
150-300		86
>300		103
10. Sulfate 0-149		89
150-300		97
>300		108
11. Total alkalinity 0-199	12	
200-300	13	
>300	14	
12. Sulfate 0-149		60
150-300		90
>300		108
13. Sulfate 0-149		60
150-300		90
>300		108
14. Sulfate 0-149		90
150-300		99
>300		108
15. Substrate mostly boulder and rubble	16	
Gravel and rubble	20	
Sand and boulder, rubble or gravel	24	
16. Total alkalinity 0-199	17	
200-300	18	
>300	19	

APPENDIX 8 (con.)

	Go to key number	CTQp
17. Sulfate 0-149		50
150-300		65
>300		90
18. Sulfate 0-149		50
150-300		65
>300		90
19. Sulfate 0-149		90
150-300		96
>300		108
20. Total alkalinity 0-199	21	
200-300	22	
>300	108	
21. Sulfate 0-149		50
150-300		80
>300		103
22. Sulfate 0-149		55
150-300		80
>300		108
23. Sulfate 0-149		80
150-300		96
>300		108
24. Total alkalinity 0-199	25	
200-300	26	
>300	27	
25. Sulfate 0-149		66
150-300		88
>300		108
26. Sulfate 0-149		65
150-300		88
>300		108
27. Sulfate 0-149		85
150-300		93
>300		108
28. Substrate mostly boulder and rubble	29	
Gravel and rubble	33	
Sand and boulder, rubble or gravel	37	
29. Total alkalinity 0-199	30	
200-300	31	
>300	32	
30. Sulfate 0-149		50
150-300		62
>300		100
31. Sulfate 0-149		50
150-300		62
>300		108
32. Sulfate 0-149		85
150-300		90
>300		108
33. Total alkalinity 0-199	34	
200-300	35	
>300	36	
34. Sulfate 0-149		50
150-300		77
>300		108
35. Sulfate 0-149		50
150-300		77
>300		108

APPENDIX 8 (con.)

	Go to key number	CTQp
36. Sulfate 0-149		90
150-300		99
>300		108
37. Total alkalinity 0-199	38	
200-300	39	
>300	40	
38. Sulfate 0-149		80
150-300		100
>300		108
39. Sulfate 0-149		80
150-300		100
>300		108
40. Sulfate 0-149		100
150-300		108
>300		108

APPENDIX 9

Biotic Index for Chandler's Score

Table 50. — Biotic index for Chandler's score as adapted by Cook (1976) for an eastern North American stream

Groups present in sample	Increasing abundance				
	Present	Few	Common	Abundant	Very abundant
	Points scored				
Each species of Perlidae, Perlodidae, Chloroperlidae, Taeniopteryginae	90	94	98	99	100
Each species of Nemouridae (excluding Taeniopteryginae), Astacidae	84	89	94	97	98
Each species of Ephemeroptera (excluding *Baetis*)	79	84	90	94	97
Each species of cased caddis, Megaloptera, *Agrion* (Zygoptera)	75	80	86	91	94
Each species of *Ancylus*	70	75	82	87	91
Rhyacophila (Trichoptera)	65	70	77	83	88
Genera of *Dicranota, Limnophora,* Tipulidae	60	65	72	78	84
Genera of *Simulium, Pristina*	56	61	67	73	75
Genera of Coleoptera (excluding *Stenelmis*), Nematoda	51	55	61	66	72
— Ceratopongidae	47	50	54	58	63
— *Baetis* (Ephemeroptera), Anisoptera,	44	46	48	50	52
— *Stenelmis* (Coleoptera)					
— *Gammarus*	40	40	40	40	40
Each species of uncased caddis (excluding *Rhyacophila*),					
— Zygoptera (excluding *Agrion*)	38	36	35	33	31
Each species of Tricladida	35	33	31	29	25
Genera of Hydracarina	32	30	28	25	21
Each species of Mollusca (excluding *Ancylus*)	30	28	25	22	18
— Chironomids (excluding *C. riparius*)	28	25	21	18	15
Each species of *Glossiphonia*	26	23	20	16	13
Each species of *Asellus*	25	22	18	14	10
Each species of leech (excluding *Glossiphonia, Haemopsis*)	24	20	16	12	8
— *Haemopsis*	23	19	15	10	7
— *Tubifex* sp.	22	18	13	12	9
— *Nais*	20	16	10	6	2
Each of the air-breathing species	19	15	9	5	1
No animal life			0		

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Platts, William S.; Megahan, Walter F.; Minshall, G. Wayne. Methods for evaluating stream, riparian, and biotic conditions. Gen. Tech. Rep. INT-138. Ogden, UT: U.S. Department of Agriculture, Forest Service, Intermountain Forest and Range Experiment Station; 1983. 70 p.

This report develops a standard way of measuring stream, riparian, and biotic conditions and evaluates the validity of the measurements recommended. Accuracy and precision of most measurements are defined. This report will be of value to those persons documenting, monitoring, or predicting stream conditions and their biotic resources, especially those related to impacts from land uses.

KEYWORDS: methods, aquatic habitat, fish, streams, inventory, fish dynamics, riparian, stream channel, macroinvertebrates

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This publication reports research involving pesticides. It does not contain recommendations for their use, nor does it imply that the uses discussed here have been registered. All uses of pesticides must be registered by appropriate State and/or Federal agencies before they can be recommended.

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