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AQUATIC FIELD AND LAB METHODS II

NORTHWEST CENTER FOR SUSTAINABLE RESOURCES (NCSR)
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Aquatic Field and Lab Methods II

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Technology education programs in which this course is incorporated are described fully in the Center's report entitled, "Visions for Natural Resource Education and Ecosystem Science for the 21st Century." Copies are available free of charge.

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Course materials are also posted on our website:
www.ncsr.org

Please feel free to comment or provide input.

Wynn W. Cudmore, Ph.D., Principal Investigator
Northwest Center for Sustainable Resources
Chemeketa Community College
P. O. Box 14007
Salem, OR 97309
Phone: 503-399-6514
E-mail: wynn.cudmore@chemeketa.edu

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INTRODUCTION

This course has been adapted for use by the Northwest Center for Sustainable Resources. It is suitable for undergraduate and community college programs as well as secondary school programs.

Aquatic Field and Lab Methods II was designed to be a "capstone/learning community" course for final quarter sophomores majoring in natural resources and fisheries technology. As a follow-on to *Aquatic Field and Lab Methods I* (a 6 credit capstone course taught the previous quarter), this course continues to build on mastery of water quality data collection, analysis and presentation skills. This course also addresses the learning of biological sampling, analysis and presentation.

The first five (5) weeks of the quarter, are spent learning biological sampling techniques (i.e., plankton analysis, electroshocking, macroinvertebrate sampling, beach seining and otter trawling). Each student is assigned approximately 2-3 different methods so they can become "expert" in these methods. Once they master these, they are responsible for teaching them to the other students in the class. Mastery of the methods is then demonstrated to the instructor.

Also, during the first half of the quarter the class reads and seminars on a wide range of topics including ecosystem management, understanding ecosystems, stream channels, large woody debris in forested streams, stream temperature and aquatic habitats. The class begins learning and using some project planning and management tools outlined in *"The Memory Jogger II—A Pocket Guide of Tools for Continuous Improvement and Effective Planning."* Some initial experimental design work for the ecosystem study will be initiated in the last half of the course.

Between weeks 6-11 a comprehensive ecosystem study is carried out at a nearby field research site. An overnight ROPES/Challenge course is the first activity to develop the self confidence, self esteem, leadership and teamwork necessary to complete the ecosystem study. Students are required to keep field notebooks and formal journals during the entire quarter. Project planning is well underway by weeks 6-7. Project management continues from week 7-11. As the class takes on the roles of their chosen study, mission, goals and objectives, scope-of-work, and task and timelines are developed to complete the work plan.

The ecosystem study includes conducting a physical, chemical and biological baseline study at the field site. Students begin weekly monitoring of such characteristics as plankton in a lake, and macroinvertebrate populations in a creek. In addition, students use nets and electroshockers to sample fish populations in the lake and creek. Water quality sampling (using techniques learned during the previous quarter in *Aquatic Field and Lab Methods I*) could also be programmed into the weekly sampling schedule.

Products of this capstone course include: keeping a field notebook and journal; completing several problem sets (biometrics); participating in eight (8) seminars and facilitating at least one (1) of them; completing a team-prepared written research report of ecosystem study results; writing an article for a local newspaper; preparing a video of the learning community experience; attending one (1) natural resource meeting in the community; making two (2) oral presentations to the general public and a professional audience; and turning in a portfolio with work samples. In addition, students are required to write self evaluations at the end of the quarter, and students will evaluate their peers.

This is a 6-credit course which meets for 3 lecture hours and 6 lab hours per week. *Aquatic Field and Lab Methods II* allows students to learn the basic concepts, techniques and skills used in ecological surveys and research. The scientific method, experimental design, data collection, recording, analysis and presentations are emphasized. Basic statistical analyses using computers are routinely used in this course, and field training includes small boat handling and the skilled use and care of biological sampling equipment. Students work individually and in teams to safely carry out "real world" lake and stream surveys. Written research reports and students oral presentations are major components of the course.

TEXT AND REFERENCES

DePree, M. 1992. *Leadership Is An Art*. Del Publishing, a division of Bantam Doubleday Del Publishing Group, Inc. New York, NY. ISBN: 0-440-50324-8

Cox, G.W. 1996. *Laboratory Manual of General Ecology*. Seventh Edition. W.C. Brown Publishers. 278 pp.

Herman, S.G. 1994. *The Naturalist's Journal – The Grinnell System of Journel Writing*. The Evergreen State College. 275 pp.

COURSE DESCRIPTION

The primary goal of this field and laboratory course is for students to become skilled in the basic use of biological techniques and equipment that are used at the entry-level of employment in fisheries and natural resources. Along with learning the responsible, ethical, and safe use of these field and lab instruments and tools, students will be able to maintain them, troubleshoot and make minor repairs. Data collection, analysis and reporting of results (in both oral and written formats), are major objectives of this course. Students will work both individually, and in teams, to complete a wide array of learning outcomes.

COURSE OBJECTIVES

Upon successful completion of the course, students should be able to:

1. Demonstrate an understanding of the scientific method and experimental design in actual field research studies.

2. Work individually and in teams to plan and execute a biological field survey. Project planning will include mission, goals, objectives, scope of work, tasks and timelines.

3. Perform the following biological sampling methods with a high degree of knowledge, skill, precision and accuracy:

    a. plankton sampling.
    b. beach seining.
    c. otter trawling.
    d. macroinvertebrate sampling.
    e. mark-recapture population assessment.
    f. various aspects of timber, fish and wildlife sampling and monitoring.

4. Work safely, both individually and in teams, in a supervised, "real world" lab and field setting.

5. Demonstrate critical thinking skills through active participation in seminars, project planning, report writing and oral presentations of their research project.

6. Use previously learned problem solving, statistics and computer skills to evaluate and present results of field and lab exercises. Critically think about how economic, political, and social factors affect ecological policies and decision-making.

7. Maintain accurate, insightful, and timely field notebooks and journals throughout the term.

8. Creatively express themselves, orally and in writing, through field notebooks, journals, seminars, and written and oral presentations of formal research results; be able to use a variety of contemporary audio-visual techniques in their presentations.

STUDENT ASSESSMENT

Students may be assessed on a variety of activities which they have undertaken during the course.

Traditional assessment:

A. Quiz I – 25 pts
B. Quiz II – 50 pts
C. Mid-Term Exam – 100 pts
D. Oral Exam – 100 pts
E. Final Exam – 125 pts
F. Seminars – 16-20 pts each

Additional methods of assessment:

A. Field notebook and journals
C. Written research report
D. Presentation of field research to general and professional audiences
E. Demonstration of lab/field skills using the following equipment/methodologies:

1. Electroshocking
2. Surber Sampler
3. Ponar Dredge
4. Plankton Sampling/Counting
5. Beach Seining
6. Otter Trawling

F. Self-assessment of performance
G. Instructor evaluation of student performance (written)
H. Peer review of performance

TOPICS

I. Introduction to Biological Field and Laboratory Methods

A. Syllabus

1. Texts
2. Course Description
3. Course Objectives
4. Topics
5. Techniques of Instruction
6. Laboratory/Field Exercises
7. Methods of Evaluation
8. Other Instructional Materials, Equipment, Software and References

B. Course Outline

1. Field Notebooks and Journals
2. Seminaring
3. Project Planning/Field Research Project

II. Overview of an ecosystem study

A. Ecosystem management (seminar)
B. Literature research in ecology
C. Experimental design in ecological studies
D. Understanding ecosystems I (seminar)

III. Important considerations of ecological studies

A. Sampling design in ecological studies
B. Writing an ecological research paper
C. Understanding ecosystems II (seminar)

IV. Quantitative descriptions of ecological samples

A. Testing basic ecological hypothesis about samples
B. Stream channels (seminar)
C. Regression, correlation and analysis of variance

V. Management/leadership

A. Personal time management
B. Continuous Quality Improvement (CQI)
C. *Leadership Secrets of Atilla the Hun* (seminar)

VI. Using *Memory Jogger II*

A. "Changing Paradigms" (video)
B. Continuous Quality Improvement tools
C. Brainstorming
D. Developing an affinity diagram
E. Developing a tree diagram
F. Participating in the ROPES/Challenge course

VII. Project planning (ecosystem study)
- A. Defining our mission and scope-of-work
- B. Using project planning software for ecosystem study tasks and time lines
- C. How to conduct team meetings
- D. Creating a cause and effect diagram

VIII. Project management (ecosystem study)

- A. Collection and analysis of physical, chemical and biological data
- B. Process improvement (class activity)
- C. Fine sediments
- D. Large woody debris
- E. Stream temperature and aquatic habitat
- F. Creating a flow chart (class activity)
- G. Nominal group technique (class activity)
- H. Demonstration of ecological sampling skills (individual assessment)

IX. Learning to work together

- A. Conducting team meetings
- B. Preparing for oral presentations
- C. Editing and assembling final written report
- D Rehearsal for oral presentation

X. Assessment – Instructor's choice of method from variety of assessment choices.

LABORATORIES AND ACTIVITIES

Field Notebooks and Journals
Plankton Sampling and Analysis
Biometrics—Terminology and Problems
Stream Analysis—Electroshocking
Stream Analysis—Discharge
Macroinvertebrate Sampling—Surber Sampler, Ponar Dredge, and Multiple-Plate Sampler
Mark-Recapture Method—Lincoln Index
Gas Supersaturation
Beach Seining and Otter Trawling
Nekton Sampling

Additional activities:
Capstone I—Team Building (The ROPES Challenge)
Capstone II—Ecosystem Study (Learning Communities)

Seminars: Special Topics and Internet Research

DETAILED SCHEDULE

Week 1
Lecture: Introduction to Biological Field and Laboratory Methods
Reading: Preface; Notes to Students; Chapter 1; pp 1-8 (lab text)
Lecture: Designing An Ecological Study; Ecosystem Management

Week 2
Lecture: Literature Research In Ecology
Reading: Chapter 3; pp 21-25 (lab text)
Lecture: Experimental Design in Ecological Studies
Reading: Chapter 4; pp 26-32 (lab text); Understanding Ecosystems
Seminar Topic: Understanding Ecosystems—Part I
Assignment: Biometrics problem

Week 3
Lecture: Sampling Design in Ecological Studies
Reading: Chapter 5; pp 33-39 (lab text)
Lecture: Writing An Ecological Research Paper
Reading: Chapter 6; pp 39-42 (lab text)
Seminar Topic: Understanding Ecosystems—Part II

Week 4
Lecture: Quantitative Description of Ecological Samples
Reading: Chapter 7; pp 43-51 (lab text)
Lecture: Testing Basic Ecological Hypothesis About Samples
Reading: Chapter 8; pp 53-59 (lab text)
Seminar Topic: Stream Channels—The Link Between Forests and Fishes

Week 5
Lecture: Regression, Correlation, and Analysis of Variance
Handout Topic: Time Management
Lecture: Personal Time Management
Seminar Topic: Fine Sediments and Salmon Production
Reading: *Leadership Secrets of Atilla the Hun*

Week 6
Reading: Continuous Quality Improvement (CQI) Tools; pp1-22
Class Activity: Brainstorming (Affinity Diagram)
Reading: Continuous Quality Improvement (CQI) Tools; pp 23-30; 156-160
Seminar Topic: Fine Sediments and Salmon Production
Reading: *Memory Jogger II*, pp 23-30; 56-62; 91-114; 150-155

Week 7
Class Activity: Defining Our Mission and Scope-of-Work
Reading: *Memory Jogger II*, pp 23-30; 154-155
Class Activity: Team Meeting #1; Cause and Effect (Fishbone)
Reading: *Memory Jogger II*, pp 56-62
Seminar Topic: Large Woody Debris in Forested Streams in the PNW: Past, Present, and Future

Week 8
Class Activity: Team Meeting #2; Flow Charts
Reading: *Memory Jogger II*, pp 115-131
Class Activity: Process Improvement Exercise
Reading: *Memory Jogger II*, pp 115-131 (review)
Seminar Topic: Stream Temperature and Aquatic Habitat—Fisheries and Forestry Interactions

Week 9
Class Activity: Team Meeting #3; Process Improvement
Assignment: Self Evaluations and Portfolios
Class Activity: Nominal Group Technique (NGT)
Assignment: Results of NGT
Seminar: Learning To Work Together

Week 10
Class Activity: Team Meeting #4
Class Activity: Final Preparation for Oral Presentation to General Audience
Class Activity: Rehearse for Oral Presentation to a Professional Audience
Class Activity: Team Meeting #5

Week 11
Finals Week: Instructor's Choice of Assessment is conducted here

INSTRUCTION TECHNIQUES

Lectures are supplemented with whiteboard illustrations, overhead projections, and seminars about special reading assignments, discussions, and handout materials.

Laboratories include keeping an accurate field notebook and journal; use of active and passive fish sampling devices (i.e., electroshocker; plankton, gill, and trawl nets; beach seines); collecting, preserving, identifying and classifying biological specimens; and small boat handling; maintenance and repair. Students work individually and in teams to carry out ecological research studies. They are required to write and speak about their results.

OTHER MATERIALS, EQUIPMENT, SOFTWARE, AND REFERENCES

I. Sampling Equipment—Two (2) *Boston Whalers*, beach seines and otter trawls, electroshocker, and a wide array of biological sampling equipment and supplies.

II. AV Equipment (available to students)

A. Overhead projector and mylar projections
B. 35 mm cameras and film or digital cameras
C. LCD projectors
D. Slide projectors
E. Video cameras and blank video tapes

III. Hardware, Software, Video

IBM PC's (located in the classroom)

Word processing, presentation and spreadsheet software

Project planning software (PERT and Gantt)

REFERENCES

Robert, W. 1990. *Leadership Secrets of Atilla The Hun.* Warner Books. New York, NY.

Video, "Changing Paradigms," from the book by Joel A. Baker, *Discovering the Future: the Business of Paradigms.* 2nd Ed. Washington Society of Chartered Public Accountants. Registration # 72514. Videotape distributed by Chart House Learning Corporation (1-800-328-3789).

Standard Methods for the Examination of Water and Wastewater. 1985. 17th Ed. American Public Health Association (APHA), American Water Works Association (AWWA), and the Water Pollution Control Federation (WPCF).

Statistix for Windows—User's Manual. 1996. Analytical Software, P.O.B. 12185, Tallahassee, FL 32317-2185. 333pp.

Fisheries Techniques. 1995. Second Edition. American Fisheries Society. 732pp.

The Memory Jogger—A Pocket Guide of Tools for Continuous Improvement and Effective Planning. 1994. 1st Edition. GOAL/QPC. Methuen, MA.

The Team Handbook—How To Use Teams To Improve Quality. 1992. 18th Printing. Joiner Associates, Inc. Madison, WI.

Field Notebooks and Journals

INTRODUCTION

As natural resources, fisheries and/or GIS technicians, one of the most important tools you will need to possess will be to observe and record what goes on around you in the natural world. **Field notes** and **journals** are an important part of developing and improving your observation and data-collection skills over time, and you will be required to keep both a field notebook and journal this entire quarter. Observations, insights, and key information you learn in this class will not only help you in this class, but it will provide an excellent record for you to refer back to whenever needed. You will develop record-keeping skills by keeping field notes and journals on a daily basis. Periodically, both your field notebooks and journals will be turned in to be reviewed by the instructor or his/her assistant.

Use your text, "*The Naturalist's Field Journal,*" as your guide for both format and content for your field notebooks and journals (see course outline—text and references).

FIELD NOTEBOOKS

The field notebook is usually a *Rite-In-The-Rain* type (either the 3.5" x 5" size, or the spiral bound). Both are designed to fit in your shirt or back pocket. Both types can be purchased for about $3-4 at the college bookstore. You may fill up several notebooks during one quarter! Always write in your notebooks using a #2 pencil or black waterproof ink. Most inks will "run-in-the-rain." Remember too, that field notebooks **always** go with you into the field.

Use your "Rite-in-the-Rain" field notebooks to record your observations and measurements (when field data forms are not provided). Also record questions and insights and make diagrams and sketches whenever possible. At the end of the day, you should always transcribe your field notes into your journal.

Items in your field notebooks should describe: *who?, what?, when?, where?, why?* and *how?*

1. Front Covers:

Some important information to include on the front cover of your field notebooks includes:

- course number and course name
- a note that says, "If found, please return to: (your name, address and phone #)"

2. Inside the Front Cover:

Important information to write inside the front cover includes the following reminder checklist:

- date/time
- weather observations
- locations, landmarks
- routes to and from your destinations
- field notes from instructors or guest speakers (in the field)
- mileage to and from destinations
- plants, animals and habitats observed
- questions and/or insights
- *qualitative observations* based upon your senses of sight, smell, hearing, touch and taste
- *quantitative measurements* (when field data sheets are not provided)
- animal signs (tracks, scat, calls, etc.)
- maps, drawings and sketches
- who was with you on your data-collection trip?

NATURALIST'S JOURNAL

You will transcribe notes taken in the field into your journal. Unlike your field notebooks, your journal *does not* go into the field.

Your naturalist's journal consists of three major sections: The Journal, Species Account and Diary.

1. The Journal:

This first section is much like your field notebook. Much of the information you recorded that same day in the field can be transcribed to this section of your journal. *Things to avoid* in this section are personal feelings, emotions, and comments of a non-scientific nature (save these for your Diary). Journal entries are a bit more formal than field notebook entries (pay more attention to complete sentences, grammar and punctuation). Journal entries can serve as *legal evidence* and are often bound and placed in library collections.

Follow the examples in Herman's text for both format and content, and you can't go wrong.
It goes into great detail as to the specific format and style of the field journal. You will be required to learn and use this style. Consult Herman's text whenever you are in doubt. His book is based upon the *Grinnell System of Journal Writing*, which is a very disciplined type of technical writing.

2. *Species Account:*

The first time you see a particular species of a plant or animal, it should be noted in your field notebook. *Sketch it right on the spot!* Describe the habitat you found it living in. What other species of plant and animals were also living in this habitat? Describe what life stage or season of the year you observed the plant or animal.

As you transcribe this new species to your Species Account Section of your journal, describe in full detail what you observed the first time you noted it. This may take more than one page. You may have to look up some scientific information or even key out the species the first time you record it. As minimum, you should include:

- Common name
- Scientific name (genus and species)
- Other taxonomic classifications (phylum, class, order, and family)
- date/time of observation
- location
- habitat
- season of the year
- other plants and animals nearby
- ecological relationships (interactions with other members of the observed ecosystem)
- physical, chemical, and biological characteristics (size, color, smell, sex, life stage, texture, etc.)
- sketch of what you observed

With the second and subsequent sightings, you only have to make general observations such as date, time, location—place these entries directly below your previous entries for this species. Draw additional sketches if the appearance is different than previous observations (i.e., perhaps this eagle is an adult compared to the immature one you saw the first time; or you now observe a salmonberry [*Rubus spectabilis*] bush in winter—without leaves—as compared to the first observation in the spring of the year).

3. *Diary:*

The Diary section of your journal is where you can be the most creative. There are almost no restrictions on what you can write about in this section. Many students like to express their emotions and feelings about what they saw in the field that day, or how the class itself is going for them (good or bad). It provides a chance to "vent some steam" if necessary or "go nuts over something that really turned you on" that day. Remember that your diary entries will *be kept strictly confidential*. No one but your instructor and assistant will ever see what you write here. We often make comments in this section that either compliment or disagree with your comments and/or thoughts. Please keep our comments confidential too!

NOTE: *Try to write **equally** in all three (3) sections every day you write in your journal.*

PROCEDURE

For this first exercise, you will have a chance to use all of your senses to observe a "bit of nature" around you.

A. Field Scenario

You will choose a small portion (a few square meters) of the natural environment around you to observe for this exercise. This involves sitting quietly in your chosen spot and recording in your field notebooks all the required information (see the checklist you've put inside the front cover of your notebook), plus *everything* else you see, hear, smell, touch, or taste within your "private space." Pay special attention to the living things you observe: Where are they?, what are they doing?, what sounds do they make?, what do they look like? (provide a sketch), how big are they? (measure them), what color are they? (NOTE: *You should be equipped with a metric ruler and a hand lens*). Also note the physical and chemical environment: air temperature, weather, clouds, soil type, water conditions, elevation, etc.

Be sure to observe, both close in and far away from your chosen location, for at least 90 minutes. Record as much as you can during this time. (NOTE: *This field notebook scenario was the "brainchild" of Ms. Judy Moore, Biology Instructor, Yakima Valley Community College, Yakima, WA. It is included with her permission*).

B. Naturalist's Field Journal Exercise

The same day (or night) after you make your initial field observations in your field notebooks, take some time to transcribe your records *equally* in the three (3) sections of your journals. There should be plenty of information to fill a number of pages in each section. Be sure to consult Steve Herman's text for *format* and *content*. You may also have to consult taxonomic keys for animals and plants when you do your species accounts. (NOTE: *Sketches should be your own, not a photocopy of a picture from a book.)*

Remember that in the Diary Section of your journal, it's your chance to reflect on how this nature observation exercise related to you as a person. *How did you feel as you were sitting quietly observing and writing? Does this bring up any memories of your past? How do you, or humans in general, fit into or relate to nature today?* This section of your journal will feel very much like you are writing an English paper—you have "the freedom to explore and expound upon" your feelings, emotions, opinions and put them into writing, very unlike most scientific or technical writing you will do. And it's OK to take a few notes in your field notebook to jog your memory later when you write them in more detail in your diary.

LAB PRODUCTS

Turn in your field notebooks and journals.

Plankton Sampling and Analysis

INTRODUCTION

Plankton are defined as organisms suspended in a body of water which, because of their physical characteristics or size, are incapable of sustained mobility in directions counter to water currents. Most plankton are microscopic and of essentially neutral buoyancy. All of them drift with the currents (APHA, 1985).

Plankton consist of both plants (**phytoplankton**) and animals (**zooplankton**), and complex interrelationships exist among the various components of these groups. Chlorophyll-bearing plants such as algae usually constitute the greatest portion of the biomass of the plankton. Phytoplankton use the energy of sunlight to metabolize inorganic nutrients and convert them to complex organic materials (they are **autotrophic**). Zooplankton and other herbivores graze upon the phytoplankton (they are **heterotrophic**), and, in turn, are preyed upon by other organisms (i.e., fish and shellfish, and amphibians), thus passing the stored energy along to larger and usually more complex animals (among **trophic levels**). The heterotrophic animals in these food chains (or more commonly, food webs) release organic materials to the aquatic ecosystem (through waste products and when they die). These in turn are broken down into inorganic chemicals by bacteria (also heterotrophs). These inorganic nutrients are released into the water and become useable again by the **primary producers** (the phytoplankton). Thus we say, *"energy flows and nutrients cycle."*

Plankton may form the "base" of the food web in most aquatic ecosystems. Therefore data concerning them may be particularly significant to the water quality technician or pollution biologist. Phytoplankton "blooms" often cause extreme fluctuations of the dissolved oxygen in lakes. Phytoplankton both respire (use oxygen) and release oxygen (through photosynthesis) during any given 24-hour period. Large concentrations of plankton can cause offensive taste and odor problems in water or in the fish that inhabit those waters. They can also be aesthetically objectionable (causing the water to have a brownish, turbid appearance). The quantity of the phytoplankton occurring at a particular sampling location depends upon many factors including sampling depth, time of day, season of the year, nutrient content of the water, and presence of toxic materials.

During the next two lab/field exercises, you will learn to collect and analyze both phytoplankton and zooplankton from a lake. We will most likely be able to trace a plankton "bloom," which usually begins around early spring (April-May), and continues to increase until the nutrients in the lake become limiting. Zooplankton "blooms" often lag behind the blooms of phytoplankton by several days (or weeks). Since they are higher on the food pyramid than the phytoplankton, the biomass (or energy content) will be roughly one-tenth (1/10) that of the phytoplankton. We will sample and analyze both the phytoplankton and zooplankton on a weekly basis throughout this quarter to see if these trophic relationships hold true.

PROCEDURE

A. General Considerations

Before plankton samples are collected, a study design must be formulated. The objectives must be clearly defined, and the scope of the study must remain within the limitations of available personnel, time and money. Historical, biological, chemical and physical (especially hydrological) data should be examined when planning a study. Of particular importance are data concerning volume of the lake, flow, currents, prevailing wind direction, temperature, turbidity depth (light penetration), and a complete chemical analysis of the inorganic nutrients at all depths in the lake. (NOTE: *These observations were made in a previous class*). During the next two lab/field exercises, we will do some preliminary sampling and analysis to ensure we have the methodology mastered. We will then apply these techniques in later labs (e.g., lake studies).

We will locate our plankton sampling stations as near as possible to those selected for chemical, physical and bacteriological (Biological Oxygen Demand and fecal coliform) sampling stations to ensure maximum correlation of findings. We will establish a 100-meter surface transect by anchoring buoys at the beginning and end of the transect. NOTE: *Our sampling site was used by previous students, assuring the availability of historical data that will lead to a better understanding of our results and provide continuity to our current study.*

B. Sampling

For sampling phytoplankton and zooplankton in the lake, we will use the protocol described in American Public Health Association's (APHA) *Standard Methods for the Examination of Water and Wastewater* (1985), pp 1043-1083.

We will use either a fine mesh plankton net towed from our 12 ft. Alaskan boat or a water sampler to collect our samples. Plankton nets are preferred to bottles and traps when plankton are few or when only qualitative data are needed for analysis.

The **maximum volume (V)** of water is that which can be filtered through a net during an oblique tow (the tow line is maintained at a pre-determined angle [usually 60°] during the entire tow).

$V = p\, r^2 d$ where:
p = *pi* (3.14)
r = radius of the net orifice
d = distance (meters) the net is towed

After the sample has been collected, rinse the net with sufficient quantities of tap water (1-2 gallons) to ensure all plankton have been washed from the inside of the net into the attached glass vial. Fill out a sample label and place it inside the vial. Add preservative (if required) and screw on a lid. Run a replicate tow (if required). For today's lab, live samples will be examined. Store samples in an ice chest in the boat, and in a dark refrigerator in the lab. NOTE: *Keep the sample at ambient temperature and examine specimens as soon as possible after collection (if possible).*

C. Preservation

The most suitable phytoplankton preservative is *Lugol's Solution*, which can be used for all forms including flagellated phytoplankton.

D. Concentration or Dilution

The plankton contained in water samples often have to be concentrated (if too few are contained in the sample), or conversely, diluted if necessary. You will determine if concentration or dilution is necessary when you begin your analysis. *Standard Methods* gives exact instructions on pp 1054-55. It is important to record the amount of dilution or concentration used since these factors will be used in our final calculations of plankton density. A *Sedgewick-Rafter Counting Cell* will be discussed under Item F. below.

E. Microscopic Identification of Plankton

You will use both a compound and binocular microscope for identification of the phytoplankton and zooplankton in your mixed sample. A depression slide works well if there are some macroscopic zooplankton in the sample. Use a drop of *Protoslo* to immobilize "fast track" zooplankton. Use plankton keys supplied by the instructor to identify plankton. Try, at least, to key your specimens to common name, genus and species. Draw a sketch of each plankton on the sheets provided.

F. Plankton Counting

We will use the counting protocol specified in *Standard Methods* (pp 1057-1067). This method includes calibrating and using a *Whipple Micrometer Square* (Figures 3a. and 3b.) in the ocular lens system of our compound microscopes (*Standard Methods*, pp 1057-60), and a Sedgewick-Rafter (S-R) counting cell (pp 1061-63).

When determining the number of plankton in 1.0ml (within the Sedgwick-Rafter slide) you can practice two different techniques: 1) the "field" method and/or 2) the "strip" method.

G. Estimating Plankton Density In the Lake

Once we know how many phytoplankton are in 1.0 ml of our sample, we can multiply this times the number of ml in our sample (assuming no dilution or concentration). We can then calculate the volume of water sampled by our plankton net towed over the 100 meter transect ($V [m^3] = pr^2d$).

We know that the number of plankton in our sample came from the volume (V) sampled by our plankton net. Since we know the estimated volume of the lake (from supplied data) we can divide the total volume (V) of the lake by the volume of the plankton tow. We can then multiply our sample number of plankton by the number of *possible* plankton tow volumes in the entire lake and get a fair estimate of plankton number at this particular point in time. *Congratulations—you made it!*

LAB PRODUCTS

A. Be sure you wash plankton nets with fresh tap water thoroughly after each day's use. Hang nets in the specified drying area before stowing away. Wash and stow all other sampling equipment and supplies away where they belong.

B. Make sure you have recorded your observations and measurements in your field notebook.

C. Complete your drawings and data sheets for both your qualitative observations (keying out plankton) and your quantitative measurements (plankton counting, density calculations and graphic plot of plankton).

D. Turn in your field notebooks, your drawings, data sheets and a 2-3 page, individually prepared write-up of this lab/field exercise.

Plankton Data Sheet

Names of Samplers:

Type of Sample:_____
Location: _____
Subsurface tow: _____ft.
Length of Tow: _____m.

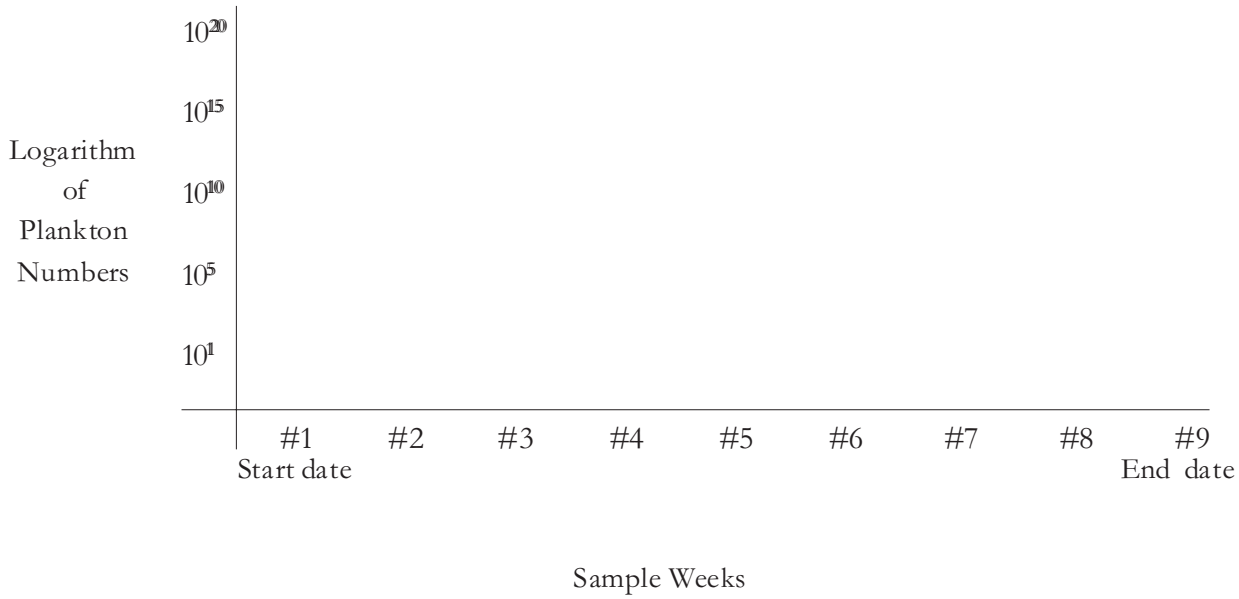
	Date / Time	Tow #1 (Surface)	Tow #2 (Surface)	Tow #1 (Subsurface)	Tow #2 (Subsurface)
#Phtoplankton					
#Zooplankton					

Calculations: *(Show work)*

Graphic Plot of Plankton Numbers

Lake _____ Months Sampled:_____

Names of Samplers:



Specimens Collected

Name _____ Section _____ Date _____

Specimen No.	Drawing
Division	
Common Name	
Genus (if known)	
Habitat	

NOTES FOR INSTRUCTORS

Be sure to read through this exercise and these notes fully before planning to implement this activity with students, as some information needs to be researched and reviewed beforehand.

A. General Considerations

We typically use a sampling site used by previous students, assuring the availability of historical data that will lead to a better understanding of our results and provide continuity to our current study.

When you design the comprehensive lake study, you may want to use a grid network established by Global Positioning Systems (GPS) and Geographic Information Systems (GIS). Also, you may want to establish two parallel transects (replicates) if your statistical design calls for them.

B. Sampling

For sampling phytoplankton and zooplankton in the lake, use the protocol described in American Public Health Association's (APHA) *Standard Methods for the Examination of Water and Wastewater* (1985), pp 1043-1083.

Once the sampling locations, depths and frequency have been determined, students should prepare for sampling by labelling sample containers (glass vials) with sufficient information to avoid confusion or error. Cut ¾" X 2" strips of *Rite-In-The-Rain* paper (20 strips per team) for labels. On the label (in pencil) indicate date, sampling station, study area (lake name), type of sample (plankton net), and depth (surface or subsurface). If subsurface sampling, describe how to estimate the depth of the tow. If the samples are to be preserved, list the type of preservative and the concentration on the label. Also; if replicate samples are taken, put the tow# on the label.

In their field notebook students should record the sample location, depth, type, date, time, meteorological conditions, turbidity (secchi disk reading), water temperature, and other significant observations. These field data may be invaluable when analytical results are interpreted, and often help to explain unusual changes caused by the variable character of the aquatic environment (APHA, 1985).

The **maximum volume (V)** of water is that which can be filtered through a net during an oblique tow (the tow line is maintained at a pre-determined angle [usually 60°] during the entire tow).

$V = p\, r^2 d$ where:
p = pi (3.14)
r = radius of the net orifice
d = distance (meters) the net is towed

Since the cosine of 60° is 0.5, the depth of the tow can be estimated based upon the principle of right triangles. For example, if the tow line length (the hypotenuse) is 50 ft., and a tow angle of

60° is maintained during the entire tow, then the depth is ½ of the length of the tow line (50 ft) minus the distance of the top of the tow line to the surface of the water. Thus, if the distance of the tow line above water is 4 ft., then the actual depth of the plankton net would be 21 feet.

C. Preservation

The most suitable phytoplankton preservative is *Lugol's Solution*, which can be used for all forms including flagellated phytoplankton. See *Standard Methods* (p. 1047) for preparing Lugol's Solution. Preserve zooplankton samples with 70% ethanol or 5% buffered formalin. Ethanol preservative is preferred if the zooplankton are to be stained (pg. 1052, Standard Methods, 1985). Formalin may be used for the first 48 hours of preservation with subsequent transfer to 70% ethanol.

D. Plankton Counting

Use the counting protocol specified in *Standard Methods* (pp 1057-1067). This method includes calibrating and using a *Whipple Micrometer Square* (Figures 3a. and 3b.) in the ocular lens system of a compound microscopes (*Standard Methods*, pp 1057-60), and a Sedgewick-Rafter (S-R) counting cell (pp 1061-63). The S-R counting cell (Figure 2b.) is a device commonly used for plankton counting because it is easily manipulated and produces reasonably reproducible results when used with a Whipple disc. The S-R cell is 50mm long by 20 mm wide by 1mm deep. Thus, the total area of the well is 1000 mm^2 and the volume when filled w/sample is 1000 mm^3 or 1.0 ml, or approximately 1.0 cm^3. This will be very useful when we start to extrapolate the number of plankton in 1.0 cc to the number in the column of water sampled by the net (# plankton/m^3) and finally, the number of plankton in the entire lake.

When determining the number of plankton in 1.0ml (within the Sedgwick-Rafter slide) you can practice two different techniques: 1) the "field" method and/or 2) the "strip" method. In the first, 10 randomly chosen fields of view (within the calibrated Whipple disc) are counted for phytoplankton and zooplankton. An average #/field is then calculated. Using the "strip" method, 3-4 full-length strips across the length of the S-R slide are counted. These are then averaged to get #/strip. You can then calculate the number of "field," or "strips," per S-R slide (based upon our original micrometer calculation for the Whipple disc). Then multiply or divide number of cells per milliliter by a correction factor to adjust for sample dilution or concentration. *Standard Methods* outlines the calculations to find # of plankton per ml. of sample for fields and strips on pp 1062-63.

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Biometrics—Terminology and Problems

Biometrics is an important tool used to study natural resource-related entities—whether they be trees, wildlife, invertebrates, or fish. The following provides an overview of concepts and related sample problems.

TERMINOLOGY

Statistics For our purposes, this may be defined as a collection of methods which have been developed for handling numerical data.

Biometrics The application of statistical methods to the solution of biological problems.

Experiment Any scientific endeavor where observations or measurements are made in order to draw inferences about the real world. Some experiments are rigidly controlled, whereas others can take the form of a study, survey or field collection.

Observation A record representing some property or characteristic of a real world object. Measurement is used in place of observation and implies a quantified observation.

Character of Interest That observation or measurement related to the purpose of the study. The character of interest is the characteristic being observed or measured; the measurement is recorded, analyzed and interpreted to draw an inference about the real world.

Experimental Unit *and* **Universe** The experimental unit is the object upon which the observation is made—it could be a single fish, an aquarium full of fish, or a lake full of fish. It must be clearly defined so as to restrict measurements to only those units of interest to the study. The universe is the set of all experimental units of interest in the study.

Population *and* **Sample** Population refers to the set of values for the characteristic of interest for the entire group of experimental units about which the inferences are to be made (the universe). Observations are not taken for all possible experimental units—only a sample is taken. A sample is a set of observations, usually only a small fraction of the total number that will be taken. Thus, samples represent a portion of the real world that has been selected for measurement.

Statistic *and* **Parameter** The sample statistic is an estimator of the population parameter, and there is a certain degree of variation among samples. When trying to characterize a population, we cannot obtain a perfect representation of that population by only

looking at a sample. For example, if we want to know about the population mean, we take an adequate sized sample and determine the sample mean. The population mean is a parameter. The sample mean is a statistic. The statistic is related to the parameter in the same way the sample is related to the population. Hence, we speak of population parameters and sample statistics.

Frequency Indicates how many times a particular score occurs in a collection of data.

Frequency Table A tabular arrangement of data, ranked in ascending or descending order of magnitude, together with the corresponding frequencies.

Frequency Histogram A set of rectangles having bases on a horizontal axis with centers at the given scores and heights equal to the corresponding frequencies (see figure 1 below).

Frequency Polygon A line graph of frequencies plotted against scores (can be obtained by connecting midpoints of tops of rectangles in the frequency histogram).

To illustrate the definitions above, consider a study in which chloride levels in a lake are to be determined. Twelve water samples are taken and chloride levels measured in μg/L. These measurements are given below:

100	101	99	101
100	100	99	102
100	98	101	102

These measurements may be arranged in a frequency table that represents how often each measurement occurred:

Frequency Table	
Chloride (μg/L)	**Frequency**
98	1
99	2
100	4
101	3
102	2

This table can then be used to generate a frequency histogram and frequency polygon as follows:

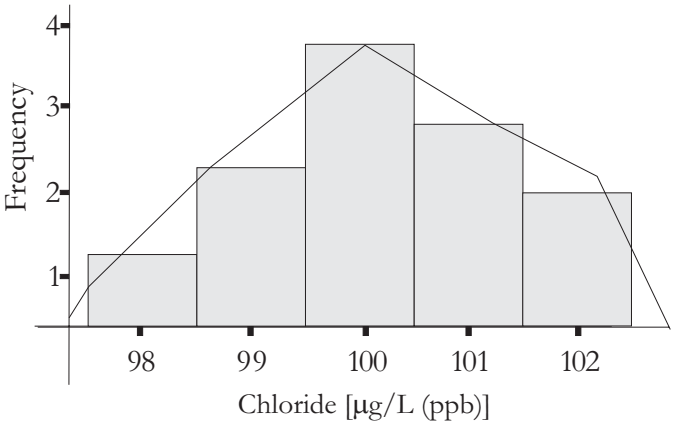

Figure 1. Frequency histogram for chloride concentrations (µg/L or parts per billion).

A. Measures of Central Tendency

Central Tendency The tendency of values to cluster about a particular value in a distribution of data.

Mode The value which occurs most frequently. In the example above, $\text{Mode} = 100.\ g/L$

Median Midpoint of a distribution of scores. It is most commonly used when the distribution is skewed or asymmetric.

If there are an odd number of observations, n, the median is $x_{(n+1)/2}$ where $x_{(n+1)/2}$ represents the $((n+1)/2)^{th}$ value in the frequency distribution.

If there are an even number of observations, the median is $(x_{n/2}+x_{(n/2+1)})/2$

$$\text{Median} = \frac{x_{n/2}+x_{(n/2+1)}}{2} = \frac{x_6+x_7}{2} = \frac{100+100}{2} = 100.\ g/L$$

Mean Arithmetic average for all the values in the sample distribution, denoted by $\bar{x}$.
The formula for calculating the *sample mean* is:

$$\bar{x} = \frac{x_1+x_2+x_3+\ldots+x_n}{n}$$

$$\bar{x} = \frac{\sum_{i=1}^{n} x_i}{n}$$

$$\bar{x} = \frac{\sum x}{n} \text{ where there are n observations in the sample}$$

$$\text{Mean} = \bar{x} = \frac{\sum x}{n} = \frac{98+2(99)+4(100)+3(101)+2(102)}{12} = 100.25.\ g/L$$

B. Measure of Dispersion

Dispersion Spread or variability of observations in a distribution.

Range The difference between the higher value and the lowest value.

$$Range = 102 - 98\ \mu g/L = 4\mu g/L$$

Average Deviation The sum of the deviation of the values from their mean, without regard to the sign, divided by the total number of values. The formula for calculating the average deviation (AD) is:

$$AD = \frac{\sum|x-\bar{x}|}{n}$$ where there are n observations.

n_x	x	$\lvert x-\bar{x}\rvert$	$n_x\lvert x-\bar{x}\rvert$
1	98	2.25	2.25
2	99	1.25	2.50
4	100	.25	1.00
3	101	.75	2.25
2	102	1.75	3.50
			11.50

$$AD = \frac{11.5}{12}\ \mu g/L$$

Note: Using the "absolute value" of the difference between each value of x and the mean $\bar{x}$ when solving for *average deviation*, is one way of disregarding the "+" or "-" signs.

Variance The sum of the squares of the deviation of the values from their mean divided by the total number of observations minus 1.

$$s^2 = \frac{\sum(x-\bar{x})^2}{n-1}$$ where there are n observations

n_x	x	$x-\bar{x}$	$(x-\bar{x})^2$	$n_x(x-\bar{x})^2$
1	98	-2.25	5.06	5.06
2	99	-1.25	1.56	3.12
4	100	-.25	.06	.24
3	101	+.75	.56	1.68
2	102	+1.75	3.06	6.12
				16.22

Note: By squaring the difference between each value of x and the mean x in determining the *variance*, we can disregard the "+" or "-" signs.

$s^2 =$

NOTE: *Using the divisor (n-1) instead of n in computing s^2 seems puzzling at first. It can be shown that dividing by n-1 provides an unbiased estimate of the population variance, whereas dividing by n produces estimates that are a bit too small (dividing by n tends to underestimate the population variance).*

Standard Deviation

The square root of the variance $s=\sqrt{\frac{\Sigma\left(x-\bar{x}\right)^2}{n-1}}$ where there are n observations

The formula adapted for the hand calculator is:

$s=\sqrt{\frac{\Sigma x^2-\frac{\left(\Sigma x\right)^2}{n}}{n-1}}$ where there are n observations

$\bar{n}_x$	x	$\bar{n}_x x$	x^2	$\bar{n}_x x^2$
1	98	98	9604	9604
2	99	198	9801	19602
4	100	400	10000	40000
3	101	303	10201	30603
2	102	202	10404	20808
Totals		1203		120617

$$s=\sqrt{\frac{120617-\frac{1203^2}{12}}{11}}$$

$$s=\sqrt{\frac{120617-120601}{11}}$$

$$s=\sqrt{\frac{16}{11}}=1.21\mu g/L$$

When using a hand-held calculator, the following calculation method may be helpful:
Subtracting a constant from each score in the distribution does not affect the variance or the standard deviation of the distribution. We can simplify the computations by first subtracting 100 from each score in the distribution, thus simplifying our calculations considerably.

$\bar{n}_x$	x	$\bar{n}_x x$	x^2	$\bar{n}_x x^2$
1	-2	-2	4	4
2	-1	-2	1	2
4	0	0	0	0
3	1	3	1	3
2	2	4	4	8
Totals		3		17

$$s^2 = \frac{\Sigma x^2 - \frac{(\Sigma x)^2}{n}}{n-1}$$

$$s^2 = \frac{17 - \frac{(3)^2}{12}}{11} = \frac{16.25}{11} = 1.48 \mu g / L$$

$$s = \sqrt{\frac{\Sigma x_i^2 - \frac{(\Sigma x_i)^2}{n}}{n-1}} = \sqrt{1.48} = 1.22 \mu g / L$$

Relative Standard Deviation

The standard deviation expressed as a fraction of the mean. The relative standard deviation is often expressed as a percent and referred to as the coefficient of variation:

$$c = \frac{(s)}{\bar{x}}(100)$$

The relative standard deviation is particularly helpful when comparing the precision of a number of determinations on a given substance at different levels of concentrations.

Normal Distribution Curve

Statisticians often represent data with theoretical curves which are smoother than frequency polygons, obtained from experiments in real life. However, frequency distribution or frequency polygons of experimental data often approximate a mathematical function called the "normal" distribution curve.

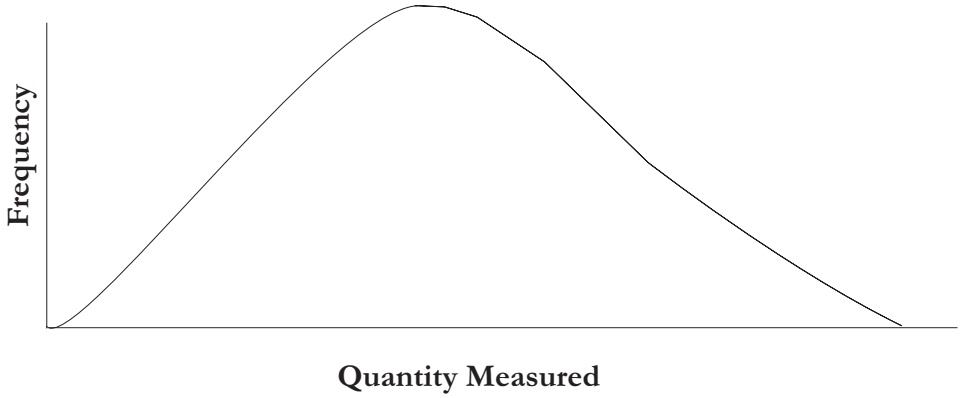

Figure 2. Normal Distribution Curve

In figure 3 the frequency polygon for chloride in water is a fairly good approximation of the "normal" curve.

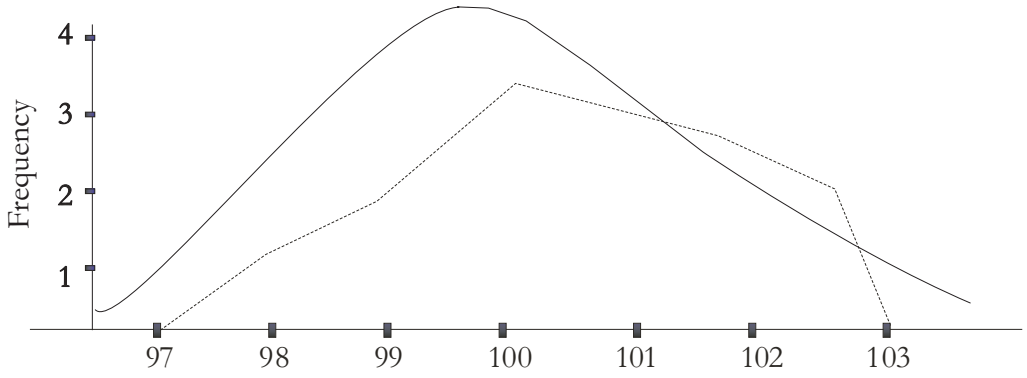

Figure 3. Normal Distribution Frequency Polygon

If a frequency distribution is a good approximation of the normal curve, we can use some facts about the normal curve to give us information about the frequency distribution. Figure 4 shows us the normal distribution in terms of the mean (μ), and the standard deviation (σ), and gives the percent of area under the curve between certain points.

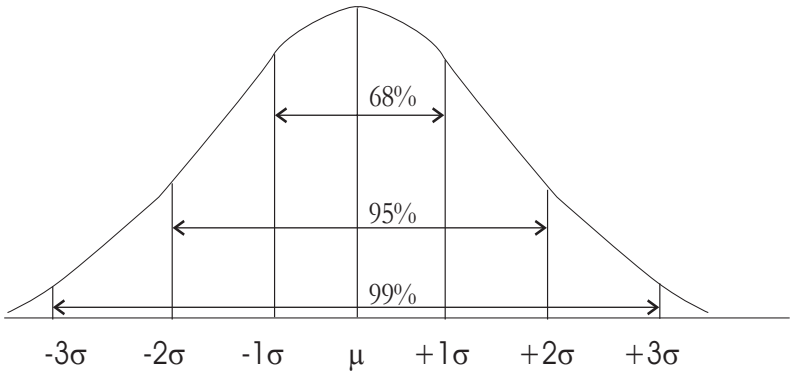

Figure 4. Normal Distribution Curve.

Figure 5 shows the frequency polygon obtained from the chloride determinations in terms of the mean ($\bar{x}$), and the standard deviation (s), and gives the percent of area under the curve between certain points.

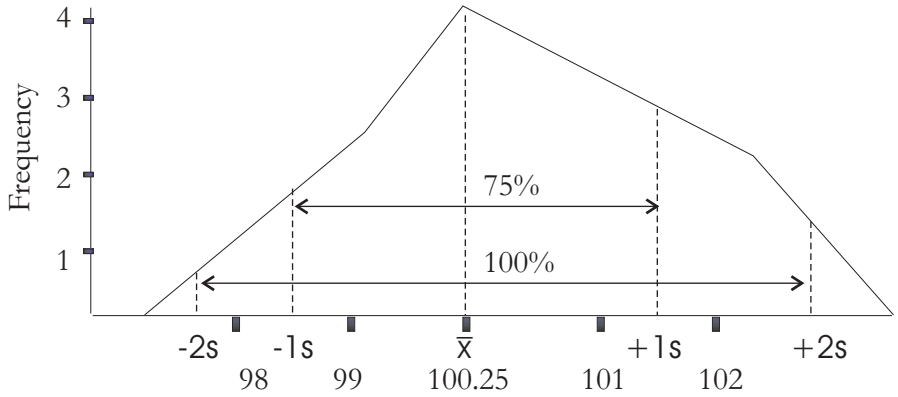

Figure 5. Comparison of Normal Curve and Frequency Polygon

Comparison of Figures 4 and 5 shows that we are justified in saying that the frequency distribution approximates the normal distribution. Specifically then, assuming a normal distribution we would expect 68% of the observations to lie within ± 1σ from the population mean when in fact, 75% of the observations were within ± 1s from the sample mean. Likewise assuming a normal distribution we would expect 95% of the observations to lie within ± 2δ from the population mean when in fact 100% of the observations were within ± 2s from the sample mean. In both cases the observed percentages are reasonably close to the expected percentages. Tests exist for determining whether or not a frequency distribution might reasonably be assumed to approximate the normal distribution.

Biometrics Problem

Codfish Length—Frequency Distribution

Given the following data collected on lengths of codfish, answer the questions.

Length (cm) (l)	Frequency ($\bar{n}$)	l	$\bar{n}$	l	$\bar{n}$	l	$\bar{n}$	l	$\bar{n}$	l	$\bar{n}$
25	2	39	18	53	15	67	--	81	--	95	--
26	7	40	15	54	8	68	--	82	2	96	--
27	8	41	13	55	6	69	--	83	1	97	--
28	9	42	13	56	11	70	1	84	--	98	--
29	13	43	19	57	7	71	1	85	--	99	--
30	12	44	19	58	4	72	--	86	1	100	--
31	9	45	21	59	5	73	1	87	1	101	--
32	15	46	13	60	1	74	--	88	--	102	1
33	7	47	19	61	2	75	--	89	--	103	--
34	7	48	21	62	1	76	--	90	--	104	--
35	5	49	8	63	2	77	1	91	--	105	--
36	12	50	22	64	--	78	1	92	1		
37	13	51	18	65	--	79	--	93	--		
38	16	52	18	66	2	80	1	94	--		
										Total	449=n

1. Plot the following data for length-frequency distribution of North Pacific black cod as frequency polygons and as histograms, first at 1 cm intervals, then at coarser groupings of 5 cm, 10 cm and 20 cm intervals.

    a. Compare the effect of using different base points for grouping (e.g., for the 5 cm group, use 25-29, 30-34, etc., or 27-31, 32-36, etc.). Use a 5 cm grouping only on this.

    b. The plots should be made on the same effective scale—(e.g., when using 5 cm intervals, the frequency in each interval will be about 5 times that when using 1 cm intervals so that the frequency should be reduced accordingly (i.e., for 1 cm intervals, 1 fish equals

1 unit on the graph paper; for 5 cm intervals, 1 fish equals 1/5 unit on graph paper; for 10 cm intervals, 1 fish equals1/10 unit on the graph paper, and 1/20 unit for 20 cm intervals).

c. The length (along the x-axis of your graph) should not be altered. Plotted this way, the various polygons and histograms should be nearly identical in appearance.

This example will illustrate the problem of the correct choice of class interval.

What appears to be the best grouping to use? Why?

2. You will find that many frequency distributions will, when plotted, appear as single peaks with more or less extensive tails above and below the peak.

a. Where does the peak (or peaks) appear in your distribution?

b. One or more quantities may be used to define the position of the peak of the distribution. The most commonly used is the **arithmetic** (or sample) **mean**, or more simply, just the **mean**. Calculate the mean by the longhand method described above (hand calculators are permitted). Show your work.

Formula for calculating sample mean:

$$\bar{x} = \frac{\sum_{i=1}^{n} x_i}{n}$$

(where there are "n" observations in the sample)

NOTE: *The numbers above and below the Σ (summation) symbol tell you what quantities to add up: Sum of x_i when i = 1, 2, 3... n^{th} (or last observation, in this case, the 449^{th} observation).*

c. Calculate the **mode** (the value at which the actual peak, or peaks, occur) and the **median**, or 50% point, which is the value such that half the individuals in the population have values less than the median, and half greater (use formulas given above). In most distributions, the median will lie between the mean and the mode, being rather close to the mean.

d. Where does your median lie?

3. a. Calculate the variance for these data.

b. The sample standard deviation is an average value of the deviation from the mean. It is calculated by taking the square root of the sample variance ($\sqrt{s^2}$). Calculate the sample standard deviation for this data.

4. Are your data normally distributed? How do you know?

5. Construct a distribution curve that shows the distribution of your sample data.

[This outline was prepared by L.A. Lederer, Statistician, Analytical Reference Service, Training Program, NCUIH, SEC. The original document and date where published is unknown.]

Stream Analysis—Electroshocking

INTRODUCTION

A. General Information

Electrofishing or **electroshocking** is the use of electricity to capture fish. **Electroshockers** are particularly useful in areas where uneven bottoms, fast flowing water, or obstructions (i.e., large amounts of wood debris) are present and make conventional collecting techniques difficult or impossible (APHA, *Standard Methods for the Examination of Water and Wastewater*, 1985). Electroshockers are non-selective in terms of size and type of fish caught; however, in the streams or ponds we will sample, we will be interested in all fish species living in these waters.

PROCEDURE

A. Operating Instructions

We will follow the operating instructions specified for the backpack electroshocker built by Smith-Root, Inc. (Vancouver, WA) commonly used for electrofishing in streams. This battery-powered unit delivers 150 minutes of continuous power before recharging is required. This unit delivers both D.C. and pulsed D.C. at 15-120 HZ and weighs 66 kg. The operating instructions are found both in the manual and on the instrument itself.

B. Safety Aspects

Electrofishing is hazardous work! The batteries used in electrofishing provide enough energy to electrocute a person. The current that passes through the body and vital organs is particularly dangerous (Reynolds, 1996). We will read safety precautions over as a class before we attempt any electroshocking.

Before any electroshocking begins, both while learning these procedures and once we use the technique for our field study, be aware of and practice the following safety procedures (Reynolds, 1996):

1. Never electroshock alone!

2. Avoid more than 2-3 samplers per team!

3. Have at least one *experienced* biologist or technician per team. That person must *actively* supervise.

4. Avoid operating near bystanders, pets or livestock in the creek or near the shore.

5. Electroshocking should proceed slowly and deliberately—avoid chasing fish.

6. Resist the urge to hand capture a stunned fish—the net person will use a net. A missed fish is better than a "shocking experience."

7. Always shut down the power source when equipment changes, repairs or any other non-routine situations arise.

8. Rest often enough to avoid fatigue.

9. Reread the procedures of this lab/field exercise before *each* and *every* sampling trip. (We have enclosed these safety procedures in plastic laminate and have attached it to the aluminum carrying case.)

NOTE: *Team supervisors must ensure all team members have read and understand these procedures*.

C. Field Sampling Scenario

Prior to beginning work, all students will be required to read and sign the attached *Acknowledgment of Electrofishing Orientation*.

1. Teams of 2-3 students will sample a 50-100 meter stretch of _____ Creek. Proceed as follows:

    a. Perform a conductivity measurement, and take a stream temperature. Record data in your field notebooks.

    b. The person with the backpack shocker and the net person will be dressed in hip boots or waders, elbow-length rubber gloves and raingear (if raining). Polarized sunglasses are extremely helpful. The netter should carry a fiberglass-handled shallow bag net. The person on shore should carry a partially-full, 5-gallon bucket of stream water (or water with MS-222, if fish are to be narcotized before examination). A second partially-full, 5-gallon bucket can be used as a recovery tank.

NOTE: *Fish should be fully recovered before release back to the stream.*

The stream bank person will key out each fish to common name, genus and species; determine life stage, length, and sex; and its general health and condition.

    c. Record all data on the field data sheet. This data sheet has been designed to allow a tally of each species to be kept.

    d. The next sampling team will continue its 50-100 meter survey upstream of the first beginning where the first team ended, and so on, with the third and fourth teams.

LAB PRODUCTS

A. Following this field exercise, all equipment, materials and supplies will be packed away and stored where directed. Be sure to put all batteries on the battery charger.

B. Ensure that all routine data (weather observations, etc.) and fish data have been recorded in your field notebooks and field data sheets.

C. Submit a 3-5 page, team-prepared, word processed or typewritten summary of this lab/field exercise. Your summary should have a title page, table of contents, 2-3 pages of text (including methods and materials, results—including tables and/or graphs), a discussion, and references cited. An appendix can be included if you choose to include your raw data, operating instructions, safety precautions, etc.

ACKNOWLEDGMENT OF ELECTROFISHING ORIENTATION

I have received instruction and orientation about electrofishing from my teacher. As a result, I understand and accept the following conditions:

1. Electrofishing (EF) is an inherently hazardous activity in which safety is the primary concern. The electrical energy used in EF is sufficient to cause death by electrocution.

2. During operation, it is critical to avoid contact with the electrodes and surrounding water. The EF field is most intense near the electrodes and can extend 5-10 m outward.

3. The electrodes are energized by the power source, a generator or battery, and controlled by safety switches; these switches must remain off until the signal is given to begin EF.

4. The power source has a main switch that must be turned off immediately if an emergency occurs.

5. The electrodes are usually metal probes suspended in the water. If direct current is used from a boat, the anodes (+) are in front of the boat to catch fish and the cathodes (–) may be suspended from the sides; both can produce electroshock. When a metal boat is the cathode, the boat is safe—as long as all metal surfaces inside it are connected to the hull.

6. Moveable anodes on a boat are dangerous, especially on metal boats. All electrodes on a conventional EF boat should be in the fixed position during operation.

7. Dry skin and clothing are good protection against electroshock. The body should be fully clothed during EF. Rubber knee boots are minimal foot protection, as are rubber gloves for the hands. A personal flotation device must be worn when the water is considered swift, cold, or deep. Ear protection is necessary for those working near the generator.

8. At least two members of the EF crew must have knowledge of CPR and first aid. A first aid kit and, in an EF boat, a fire extinguisher, must be within immediate reach during an operation. Electroshock can cause heart fibrillation or respiratory arrest; CPR can cure only the latter. The EF crew must know the location of the nearest defibrillation unit.

9. A communication system, particularly hand signals, must be available to all members of an EF crew. When multiple anodes are used in a portable EF operation, the buddy system must be used. Above all, NEVER OPERATE ALONE.

10. Stunned fish should be removed from the EF field as soon as possible and not subjected to continuous electroshock by being held in the dip net. Using the anode as a dip net is unhealthy for fish and people—*and should be avoided*.

11. An EF operation should proceed slowly and carefully; avoid chasing fish and other sudden maneuvers. Night activities require bright, bow-mounted headlights. Operations should cease during lightning or thunderstorms; use discretion during rain. Avoid EF too close to bystanders, pets or livestock.

12. All EF crew members must know who their leader is and recognize his or her authority as final in operational decisions. However, every crew member has the right to ask questions and to express concern about any safety aspect of an EF operation. A crew member has the right to decline participation in an EF operation, without fear of employer recrimination, if he or she feels unsafe in such participation.

_____ _____

Student Signature **Date**

I have discussed the above-named conditions with the employee and am satisfied that he or she understands them.

_____ _____

Teacher Signature **Date**

Adapted from a form used for employers and employees; Reynolds (1995), with permission. *In* Murphy B.R., and D.W. Willis. 1996. *Fisheries Techniques.* American Fisheries Society.

Names of Samplers: _____
_____
_____
_____

Electrofishing Data Sheet

Stream Location: _____ Stream Reach #_____

Date/ Time	Common Name	Genus and Species	Sex (M/F)	Fork Length	#Specimens (tally)

General Observations:

NOTES FOR INSTRUCTORS

Read the lab carefully, follow all safety protocols, and explain the following to students prior to doing this lab.

See *APHA, Standard Methods for the Examination of Water and Wastewater (1985); and Fisheries Techniques 2nd Edition (1996)* [devotes an entire chapter (pp 221-253)] for more about the art and science of electroshocking. Anyone interested in achieving mastery of this technique should spend considerable time reading and studying these references.

Electroshocking—Theory of Operation

An electric field in the water is produced by passing a current between two submersed electrodes or between an electrode and the ground. We use a newer style electroshocker that has only one electrode and a trailing cable that goes to ground (the water).

Depending upon their design, these electroshockers will produce either an **alternating current (AC)**, **direct current (DC)**, or in some cases both (a current inverter is required to produce both). AC stuns fish in its field, allowing them to be dipped from the water, whereas DC induces **galvanotaxis**—so that fish move toward one of the poles (the wand that contains the positive electrode). Once they swim into the electric field created by the anode (wand) and cathode (ground wire), they are stunned and can be netted. Newer DC electrofishers are usually battery powered. They are particularly effective in turbid water or in waters with numerous obstructions or heavy vegetation (since they tend to draw fish toward the electrode).

NOTE: *AC devices aren't used much anymore since they are more likely to kill or injure fish. Environmental factors such as water hardness and available electrolytes can impact the effectiveness of electroshocking, and this is why we always measure the conductivity of the waters we're sampling.*

Field Theory

As mentioned previously, the electrodes deliver voltage and current in the water to form a three-dimensional electrical field. The current flows between electrodes of opposite polarity (i.e., via flux lines) and voltage surrounds each electrode at right angles to the flux lines (i.e., equipotential lines). The field is non-homogeneous and weakens with distance from the electrode as energy dissipates in the water (Reynolds, 1996). The three parameters that apply to the electroshocker's circuit-voltage, current and resistance-differ at various points in the water. **Current density** is the current that flows through a 1-cm^2 plane (of water). **Voltage gradient** is the change in voltage over a 1-cm distance. **Resistivity** is a measure of the water's resistance (to carrying a current in the water). Water with an extremely high resistivity (i.e., deionized water) will be extremely poor at conducting a current. The inverse of resistivity is **conductivity** and is measured in μmhos/cm. Therefore, it is important to have some trace minerals in sampling waters in order for the electroshocker to be effective (the water must be able to conduct a current with a high enough

voltage to attract or stun the fish). Brackish water may contain too many minerals (electrolytes). Distilled water has very low conductivity (0.5 - 3.0 µmhos/cm). The conductivity of most freshwater bodies is between 50-1,500 µmhos/cm. On average, seawater is 500 times more conductive than fresh water (Reynolds, 1996).

Electroshocking Effects

Fish behavior in an electric field depends on the nature and intensity of the wave form applied. In any electrical field, a threshold of intensity must be reached to achieve a given behavioral response (Reynolds, 1996). Lamarque (1990) offers this review of effects on fish of an electric field:

"In responding to AC, a fish tends to assume a position perpendicular to the electric current (flux lines) thereby minimizing voltage gradient in its body. It may undulate in attempting rhythm with the AC cycle, exhibiting **oscillotaxis** (forced movement without orientation, a "thrashing" motion). At higher field intensity, **tetany**, or muscle contraction, occurs and the fish is immobilized. Fish responses are less predictable with AC than DC. Too much intensity can and will kill the fish."

In a DC field, a fish typically turns toward the **anode** and exhibits **electrotaxis** (forces swimming *towards* an electrode). As the fish nears the anode, a new threshold causes narcosis (muscle realization) and loss of equilibrium. While under **narcosis**, the fish may continue to swim, upside down, toward the anode. Tetany is achieved near the anode. Continuous DC elicits **taxis** and narcosis in fish if the appropriate threshold is reached. The effects of pulsed DC are less predictable. Depending upon fish size and species, taxis and narcosis may not occur. Continuous DC requires more power than does pulsed DC.

The importance of electrofishing-induced stress is often unrecognized by the technician or biologist because captured fish may appear normal. However, the **stress syndrome** results in an abnormal physiological state including a reduced respiratory level and **acidosis**. This stress may take hours and/or days for complete recovery. Recovering fish are also more susceptible to predation, less competitive and unable to feed. Wild fish require a longer period to recover than do hatchery fish (Mesa and Schreck, 1989).

Biological Factors Related to Electroshocking

Electrofishing tends to be more effective for some fish than others. Fish with fine scales (i.e., salmonids) tend to be more vulnerable than those with thick scales (i.e., ciprinids). Fish that inhabit shallower shoreline waters are also more susceptible to electroshocking. Electrofishing is selective for larger fish of a species. For example, adult salmon require a much lower voltage setting (about one tenth) than juvenile salmon. This may also be influenced by the territorial behavior of the adult salmon.

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Stream Analysis–Discharge

INTRODUCTION

Measuring **discharge**, or water velocity passing a cross section per unit of time, is a task performed by natural resource technicians as part of establishing permanent reference sites for gathering data about streams and rivers. The ability to accurately make and replicate stream channel measurements over a period of years, and through changes in personnel, is vital. We will use the procedure described on pages 44-48 in the U.S. Forest Service's *Stream Channel Reference Sites: An Illustrated Guide to Field Techniques* (April 1994) as our guide to performing this field test.

Stream discharge (Q) is the volume of water passing a cross-section of a stream per unit of time and is expressed as cubic feet per second (cfs). Discharge is simply velocity times cross sectional area (Q = VA). Cross-sectional area (A) is determined by stretching a tape across the channel to measure the distance (stream width) at the cross-section location. Depth is measured with a calibrated rod. Area = depth X width in small increments across the channel.

We will use a Marsh-McBirney current meter for our velocity measurements. We will also learn the float method for making velocity measurements when a flow meter is not available or when time is limited.

EQUIPMENT AND SUPPLIES
(Flow Meter and Float Method)

- Marsh-McBirney flow meter
- spare battery for flow meter
- field notebook and/or field data sheets
- clipboard
- pencils
- fiberglass cloth measuring tape (min. length 25 ft.)
- hand calculator
- chest waders or hip boots
- raingear
- polarized sunglasses (optional)
- stopwatch or wrist watch with second hand
- 5-10 floats (orange peels or water-soaked blocks of wood)

PROCEDURE

Current Velocity Measurements (Marsh-McBirney Flow Meter)

Procedures for this exercise are found in the Meter manual. Figure 58 shows the exact types of data you should enter in your field notebooks (or on your field data sheets).

Computing Discharge

We will calculate sample measurements and calculations in the laboratory before we go to the stream. Record your field data on the data sheets provided.

Float Method for Current Velocity

The float method is a simpler way to estimate discharge provided velocity has been previously measured and cross-sectional area calculated. Equipment for the float method measurement is simple: a measuring tape, timer (a stop watch or digital watch) and 5-10 floats. For floats, use an orange peel, a water-soaked block of wood, or other natural material that sinks at least halfway into the water, is visible from shore, won't be moved by wind and is expendable and non-polluting.

NOTES: *Previously measured cross sectional area (from midsection method used for flow meter calculations) can be used to find discharge (Q = VA), or if previous area data do not exist, an* ***average*** *cross sectional area can be calculated by averaging 3-4 width and depth measurements at the beginning, end, and along the channel segment being used.*

LAB PRODUCTS

Students should switch roles as note-taker and meter reader after the first measurements have been made (move to a new station 10 feet or so downstream for the second sampling). Each team of two students will turn in a one-page report that describes these two methods for measuring discharge. Include your two field data sheets as enclosures to your report. Be sure to discuss your results in your report. Turn in a one-page team report at the beginning of the next lab/field period. Be sure that the names of both the note taker and meter reader appear on these forms.

REFERENCE

U.S. Forest Service, 1994. *Stream channel reference sites: An illustrated guide to field techniques*. pp 44-48.

Discharge Field Data Sheet

Date & Time:
Location: Weather Observations:
Note Taker: Meter/Tape Reader:

Discharge Measurements (Marsh-McBirney Flow Meter)

Cross Section Measurement	Tape Distance (ft)	Width (ft)	Depth (ft)	Velocity (ft/sec)	Area (ft^2)	Q=Discharge (cfs)
1.						
2.						
3.						
4.						
5.						
6.						
7.						
8.						
9.						
10.						
11.						
12.						
13.						
14.						
15.						
16.						
17.						
18.						
19.						
20.						
21.						
22.						
23.						
24.						
25.						
(add *all* Q's from sub-sections to get total discharge)				Total Discharge		=cfs

Discharge Measurement Calculations
(Float Method)

Average Depth (Average of 3-4 measurements) = _____ ft.

Average Channel Width (Average of 3-4 measurements) = _____ ft.

(A) Area = Average depth X Average width = _____ $ft.^2$

(V) Velocity = $\frac{\text{Distance (ft.)}}{\text{Avg. Time (sec.)}}$

Distance = _____ft.

Time =_____sec.
_____sec.
_____sec.
_____sec.
_____sec.
_____sec.

Average Time = _____sec.

V = Distance (ft) = $\frac{\text{Distance (ft.)}}{\text{Avg. Time (sec.)}}$

Adjusted V = V X *Coefficient for stream bottom resistance = _____ft/sec.

Q = AV (adjusted) = _____cfs

*1. Coefficients for stream bottom resistance:

a. loose gravel, rocks = 0.8
b. smooth mud, sand, hardpan = 0.9
c. culvert = 1.0

2. Rate of flow (Q) X Correction factor for small streams (1.3) = _____cfs

Show your calculations below:

WATER DISCHARGE (VELOCITY)

Comments: Location:

Date/Time:

Method: Flow meter Measurements @ X-Sections Meter Used: Marsh McBirney

Note Taker: Meter Reader:

Tape Distance (ft.)	Width (ft.)	Depth (ft.)	Velocity (ft./sec.)	Area ($ft.^2$)	Q Discharge (cfs)	Discharge Using Float Method
						Average Depth (Estimated):_____
						Channel Width:_____
						(A) Area:_____
						(V) Velocity:
						(type of float used:_____)
						Distance:_____
						Time:
						sec._____
						sec._____
						sec._____
						sec._____
						sec._____
						Avg. Time: sec._____
						V=Distance (ft.) X CF (drag) = ft./sec.
						Avg. Time (sec.)
				Total cfs:		Q=AV=_____ft^2 x _____ ft./sec. =_____cfs

Macroinvertebrate Sampling
The Surber Sampler, Ponar Dredge/ Grab, and Multiple-Plate Sampler

NOTE: *This exercise will take 2 lab sessions to complete.*

INTRODUCTION

A. Surber Sampler

Quantitative samples of invertebrates from shallow streams have traditionally been taken by using a Surber sampler. A Surber sampler is a fine mesh net (with sides and a bag) attached to a hinged metal frame that is placed over a 1.0 ft. x 1.0 ft. x 4 in. deep stainless steel bottomless box frame that is buried to its rim in the stream substrate. The opening of the net is placed upstream. All substrate within the rim, and down to the bottom of the box frame, are agitated (or removed) to dislodge organisms, which drift into the collection bag (Rabeni, 1996).

The standard mesh size of the net is 9 threads/cm. While a smaller mesh size might increase the number of organism collected, it also will clog more easily and exert more resistance to the current than a larger mesh. This sampler is specific for the macrobenthos; many microcomponents of the benthos are not collected (APHA, 1985).

B. The Ponar Grab Sampler

This sampler is used extensively in medium to deep rivers, lakes and reservoirs. It has side plates and a screen on top of the sample compartment to prevent sample loss during closure. With one set of weights, the standard 23 X 23 cm sampler weighs 20 kg. The large surface disturbance associated with the Ponar grab can be reduced by installing hinged, rather than fixed, screen tops, thereby reducing the pressure wave associated with the samplers descent. This sampler is best used for sand, gravel, or small rocks with mud but can be used in all substrates except bedrock (APHA, 1985).

C. Multiple-Plate or Modified Hester-Dendy Sampler

The multiple-plate sample is constructed of 0.3 cm thick-tempered hardboard with 7.6 cm round plates and 2.5 cm round spacers that have center-drilled 1.6 cm holes. The plates and spacers are held together by a 0.63 cm diameter eye bolt and a nut at the bottom. There are 14 large plates and 24 spaces in each sampler. With the modified Hester-Dendy sampler, the top nine plates are separated by a single spacer, and plate 10 by two spacers. Plates 11 and 12 are separated by three spacers, and Plates 13 and 14 by four spacers. The sampler is approximately 14 cm long and 7.6 cm in diameter, and has an exposed surface area of approximately 1160 cm^2 and weighs about 0.45 kg.

Because it is cylindrical, the sampler fits a wide mouth container for shipping and storage (APHA, 1985). This sampler can be hung by a line into a lake or stream. It can be examined in total for colonization by macroinvertebrates, or the plates can be removed and examined (both qualitatively and quantitatively) one or more at a time over a period of days or weeks.

PROCEDURE

Equipment and supplies:

1. Surber Sampling

- Surber sampler
- 6 glass vials with caps
- 3 white porcelain cake pans (with grids)
- 9 strips of Rite-in-the-Rain labels
- 3 artist paint brushes
- 1 liter of 10% formalin solution
- field keys to macroinvertebrates
- field notebooks and field data sheets
- 3 clip boards w/rain cover
- hip boots or waders
- polarized sun glasses (optional)
- raingear (if needed)

2. Ponar Sampling

- Ponar grab sampler and 50 ft. line
- Stainless steel sieve tray
- 3 glass vials and caps
- 1 five gallon bucket
- 3 white porcelain cake pans (with grids)
- 3 strips of Rite-in-the-Rain labels
- 3 artist paint brushes
- 1 liter of 10% formalin solution
- field keys to macroinvertebrates
- field notebooks and data sheets

3. Multiple-Plate Sampling

- 3 Hester Dendy multiple-plate samplers
- 1 coil of nylon cord to attach sampler
- knife
- field notebook and data sheets

Teams of 2-3 students per team will practice using the Surber sampler in _____Creek.

LAB PRODUCTS

A. Data sheets for the Surber and Ponar portions of this lab will be turned in to the instructor.

B. Data sheets for the Hester Dendy sampler will be due at the end of the quarter when you turn in your field notebooks for final grading.

C. Prepare a team write-up (1-2 pp) for this lab exercise. Address the effectiveness of each method; when?, where?, why?, and how? each is used and the value of macroinvertebrate data to an overall watershed ecosystem study.

Page ___ of ___NAMES: _____

SURBER SAMPLER
DATA SHEET

Location #1 _____
Location #2 _____
Location #3 _____
Replicate # _____

Date/Time	Common Name	Genus/Species	#Found	Sketch	Comments

Page ___ of ___ NAMES: _____

PONAR GRAB SAMPLER
DATA SHEET

Location #1 _____
Location #2 _____
Location #3 _____
Replicate # _____

Date/Time	Common Name	Genus/Species	#Found	Sketch	Comments

Page ___ of ___ NAMES: _____

HESTER-DENDY
MULTIPLE-PLATE SAMPLER
DATA SHEET

Location #1 _____
Location #2 _____
Location #3 _____
Replicate # _____

Date/Time	Common Name	Genus/Species	#Found	Sketch	Comments

NOTES FOR INSTRUCTORS

Explain the following prior to or as students do the lab.

SURBER SAMPLER

Teams of 2-3 students per team should practice using the Surber sampler in _____Creek.

Duplicate Surber samples should be collected at three stations along your assigned stream reach. Sample at the beginning, middle and up-current end of your reach. Place the contents of your Surber bag into one white porcelain cake pan (with grid). Sort organisms into different sections of the pan using the artists paint brushes provided. Key out your specimens by common name, phylum, class, and order, if possible. Some specimens may be identified to the family, genus and species level. Specimens may be keyed at a later time too. Count the numbers of each species found. After sorting, keying out and enumerating your macroinvertebrates, place the contents in a glass vial, label with location, date/time, type of sampler used, replicate number, name of collector, and other pertinent information using the Rite-in-the-Rain strips. Use a pencil to record your information. Record observations in your field notebooks and data on your field data sheets.

PONAR GRAB SAMPLER

In your teams of 2-3, take three (3) samples with the Ponar grab from different locations on the lake. Pour your samples into the stainless steel sieve tray. Rinse with lake water from your 5-gallon bucket until all of the mud is washed away. Remove all foreign objects (rocks, wood chips, leaves etc.) by hand. Rinse your organisms into your white porcelain pan. Sort according to like organisms. Key them out to the greatest detail possible. Count each species. Make entries in your field notebooks and on your data sheets. After sorting, keying, counting and recording, place the contents of your tray into your glass vials. Label with the same information you used for your Surber samples.

HESTER-DENDY (HD) MULTIPLE-PLATE SAMPLER

Your team will place one HD multiple-plate sampler in each of three (3) locations. Your multiple-plate sampler work is done for today. You will be responsible for removing two (2) discs each week (beginning next week) until the quarter ends (or you run out of discs). Counting will include scraping your disc into a white porcelain pan and keying out and counting as in the Surber and Ponar protocols. You should also examine the phytoplankton and zooplankton. Calculate the surface area of the discs so that you can quantify the density of each species of macro-invertebrates and the phyto- and zooplankton. Fill out your data sheets and field notebooks each time you collect and analyze your samples. Your data will be used for your final research report.

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Mark-Recapture Method (Lincoln Index)

INTRODUCTION

The mark-recapture method of estimating animal density is used frequently in fisheries and wildlife studies (Pollack, et. al., 1990). This method was introduced in the late nineteenth century by C.G. Peterson and later re-introduced for studying bird populations by F.C. Lincoln in late 1930 (Begon, 1980). The method is based upon the fact that if we capture and **mark** some members of a total population, **recapture** them and then sample the population again to find out what proportion of the sample bears the mark, we can then, by proportionality, estimate the total population size.

The **proportionality** is:

Rearranging this proportion we get the more familiar formula:

Estimated population $\widehat{N} = \frac{Mn}{r}$

where:

$\widehat{N}$ = estimated total size of the population
M = number of animals marked in the population
n = total animals caught in the recapture sample
r = number marked in the recapture sample

NOTE: *This estimate is for the date of marking,* ***not*** *the date of recapture.*

This method is commonly used because it is relatively easy to accomplish, relatively inexpensive and can give fair estimates of population density in a rather short time frame. It is, however, somewhat inaccurate because:

1. $\widehat{N}$ is not a very precise estimator of N unless the sample size is large, or a high proportion of the population is marked.

2. Several **assumptions** may or may not be valid in an actual population (Cormack, 1993). These assumptions are:

   a) "marks" are not lost
   b) there is *no recruitment into* the population by reproduction, growth or immigration, or *emigration out of* the population
   c) marked and unmarked animals behave alike (mortality rates, activity, and response to traps are the same)

Jolly (1965) devised a capture-recapture statistical model that considers death and immigrating which gives more validity to this method.

PROCEDURE

Equipment and Supplies:

- 1 bag each of white and brown beans (of roughly the same size)
- 1 plastic bucket
- 5 hand calculators (with statistical functions)
- release-recapture data sheet

In this exercise we will use a small plastic bucket (represents a lake) filled with white beans (juvenile coho salmon). The object is to estimate the coho population density in the lake.

The instructor will begin by removing a handful of white beans from the bucket. These will be counted and put aside. An *equal* number of brown beans (representing those coho that were captured and marked) are put back into the bucket. All beans are thoroughly mixed.

Each team of students will then use a plastic cup (with a capacity of about 75-100 beans) to simulate a beach seine and scoop up a recaptured sample. Beans should be thoroughly mixed between scoops (seines). Students will enter their data in the space below. The instructor will supply the figure for the number of marked beans (coho) in the population. Compute $\widehat{N}$. Also enter other teams' estimates as they become available.

Number marked in sample (r) = _____

Sample size (n) = _____

Total marked in population (M) = _____

Your estimate of total population size ($\widehat{N}$) = _____

Other team estimates of total population size ($\widehat{N}$) = _____ , _____ , _____

DISCUSSION QUESTIONS

1. Get the *actual* value for $\widehat{N}$ from your instructor. How far off was the best estimate derived by sampling? The worst estimate? Calculate the mean ($\bar{x}$) of all the estimates. Calculate the median of all the estimates; and the mode, range, and standard deviation. Was this sampling method a close approximation of the *actual* population?

2. Why is the Lincoln index estimate for the time of marking rather than the time of recapture?

3. List several methods for capturing fish, amphibians, reptiles, birds and mammals.

4. List several methods for marking fish, amphibians, reptiles, birds and mammals.

5. Considering the actual population of coho in a lake, try to assess which of the assumptions necessary to validate the mark-recapture method—consider how it might be violated, and specifically, how.

6. What are "trap-shy" and "trap-prone" animals? How might each of them affect your estimate of population size?

7. Why isn't mortality considered an assumption for the mark-recapture method whereas reproduction is?

8. Indicate whether your estimate, $\widehat{N}$, would be too high or too low in each of the following circumstances:

a. Some coho lose their marks.

b. Immigration occurs.

c. Marked coho have higher mortality rates.

d. Marked coho are more likely to be seined.

e. Marked coho are more sluggish than unmarked ones.

Confidence Intervals:

It is desirable when using the Lincoln index to calculate a confidence interval, the interval within which you are confident (with a specific probability) that the true population lies. In general, the value we add to and subtract from (for the upper and lower confidence limits) any estimate is given by $K\sqrt{S^2}$, where S^2 is a variance and K is the normal curve variate. Unfortunately, the variance is not straight forward for the Lincoln index population estimate. Therefore, the formula we will use for our exercise today is:

$$S^2 = \frac{M^2 n(n-r)}{r^3}$$

Enter here your estimate of S^2: _____

Take the square root of S^2 and enter here your value of $\sqrt{S^2}$: _____

The useful normal curve variates (K) for the 95% probability is 1.96 (taken from a table of normal curve variates at different probability levels). Thus, now multiply 1.96 times S and enter it here:

_____

This is the value you will add to and subtract from your estimate of N ($\widehat{N}$) to give the upper and lower limits of the confidence interval.

Give those limits here: _____

Now write a comprehensive statement in the space below that gives the confidence intervals for the estimate of the coho population in the lake.

_____

_____

To get the 99% confidence limits for our population estimate, multiply S times 2.58 (the normal variate for 99% probability), and then add and subtract this value from your estimate of N.

Give those limits here: _____

[Some of the formuli listed above were taken from: Cox, G.W. 1996. *Laboratory Manual of General Ecology*. Seventh Edition]

LAB PRODUCTS

During the lab:

- Carry out the bean-counting and calculations.
- Participate in the discussion.
- Complete your calculations and fill in your data sheets and calculations.
- Exchange data with other teams. Use *your* estimate of N ($\widehat{N}$) for the calculations of confidence intervals (CI).

Following the Lab:

- Clean up your lab space. Separate brown and white beans and return them to their appropriate containers.
- Turn in one team data sheet with names of all team members in the upper right hand corner of the first page.

Turn in a 2-3 page team report that addresses the questions in the lab exercise. *Be as specific as possible!*

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Gas Supersaturation

INTRODUCTION

A. What Is Supersaturation?

Dissolved gasses are present in all fish-bearing waters, usually entering the water directly from the air. Normally, the dissolved gas pressure is equal to the atmospheric (barometric) pressure just above the air-water interface (14.7 PSI or 760 MM of mercury at sea level). NOTE: *100% saturation is considered normal and ideal.*

Occasionally, natural or man-made conditions occur that upset this balance. Supersaturation can be caused by dam spillways, waterfalls, power plant outflows, algae blooms and other sources. If the % saturation goes higher than 110%, aquatic life is threatened and fish kills can occur. If the % saturation goes below 80%, not enough dissolved oxygen is present to support aquatic life. Undersaturation (de-aerated waters, below 100% saturation) can occur in some well or spring waters and in still or stagnant ponds.

Supersaturation causes a disease in fish called the **"gas-bubble disease."** Air bubbles come out of solution in the blood vessels of fish causing blockage, rupture and death. This disease is similar to the bends experienced by human divers coming to the surface too quickly from a deep dive.

B. How was the Saturometer invented?

Large fish kills in the early 1970's on the Columbia and Snake river systems in the Pacific Northwest prompted the E.P.A. to take action to solve the problems. An inexpensive, field portable, accurate instrument to measure dissolved gas pressures at many various locations was needed. Dr. Ray F. Weiss, with the Scripps Institute of Oceanography, in conjunction with people from Region 10, E.P.A. in Seattle, designed the first operational saturometer. ECO Enterprises redesigned and started manufacturing these instruments in 1972.

Your instructor will provide you with pertinent background information, standards, and protocols.

PROCEDURE

We will visit our local fish hatchery. Students will work in teams of 2-4

1. Following the procedure demonstrated by the instructor or assistant, determine the barometric pressure using a mercurial barometer. This reading may be verified by checking with a local airport.

2. Using the ES-2 Weiss Saturometer (the long handled type) and following the correct protocol, determine the percent (%) saturation for the lake at the floating net pen. Use the same saturometer to find percent (%) saturation in the creek at the outfall below the lake.

   Record your measurements on the field data sheet provided. Exchange your instrument with other teams when completed.

3. Using the ES-3 Weiss Saturometer (the short-handled meter) determine the percent (%) saturation in the headbox inside the hatchery. Measure the percent (%) saturation in at least one (1) of the fish troughs containing coho salmon fry. Record your measurements on the field data sheet provided. Exchange your instrument with other teams when completed.

4. With your instructor's direction, perform your calculations, and record them on the data sheets provided.

EQUIPMENT AND SUPPLIES

- field notebook
- lab exercise and data sheets
- mercurial barometer
- ES-2 and ES-3 Weiss saturometer
- telephone
- hand-held calculator

LAB PRODUCTS

At the end of this lab you will be responsible for:

- Rinsing both the ES-2 and ES-3 saturometers and stowing them away according to the hatchery manager's directions.

- Complete your data sheets. Remember to obtain data from other teams to calculate mean and standard deviation for percent (%) saturation measurements taken by your team.

- Turn in one data sheet for your team at the beginning of the next lab/field period. Remember that each individual is responsible for the procedure, data collection and calculations for this exercise.

GAS SATURATION DATA SHEET

Date/Time	Location	Instrument Used	Percent (%) Saturation	Teams Average % Saturation	Teams S.D. of % Saturation	Team Member Names
	Location 1					
	Location 2					
	Location 3					
	Location 4					

Calculations: Percent (%) Saturation = $\frac{P\ atm + D\ P}{P\ atm} \times 100$

Location 1	Location 2	Location 3	Location 4

Barometric Pressure (from mercurial barometer) = _____ mm Hg
Barometric Pressure (from airport) = _____ mm Hg

Comments: _____

_____

_____

NOTE FOR INSTRUCTORS

NOTE Introductory information in this laboratory exercise has been used with permission from the "Field Manual for the ES-2 and ES-3 Weiss Saturometers," ECO Enterprises, 2821 N.E. 55th, Seattle, WA 98105. Be sure to check the sites where sampling is undertaken in this exercise to ensure you have access to similar facilities for your students.

The following is general information to share with your students. Important protocols for conducting this lab are included.

A. Principles of Operation

The saturometer operates basically as an artificial fish gill. Dimethyl silicone rubber tubing is used for the sensing membrane. This small, hollow tubing will allow the passage of dissolved gasses from the outside (water) to the interior (pressure gauge, referenced to local atmospheric pressure) for readout. Over 300 square inches (200 ft.) of this tubing is used in the ES-2 instrument.

The tubing (.025" O.D. x .012" I.D.) is wound on frames into a compact sensor. A perforated outer shell protects the membrane tubing from damage and allows rapid water transfer. A manually operated water-jet pump emits high velocity streams of water across the tubing, accelerating water transfer and removing air bubbles from the tubing that interfere with dissolved gas diffusion. The water-jet pump is not needed in undersaturated waters as no bubbles can form.

A specially calibrated low-volume pressure gauge is used to measure the dissolved gas pressure. This gauge measures from -100 to +400 mm of mercury with a full-scale accuracy of better than ± 1%. A pressure relief valve is located just behind the gauge to quickly vent internal pressure after a reading is taken, for testing, leak detection and calibration purposes.

B. Saturometer Use in Rivers, Lakes & Streams

Before industrial man started utilizing our waters for power generation, irrigation and flood control, the only source of supersaturation was waterfalls—which trapped atmospheric gasses and carried them into deep pools where the gasses went into solution. Nature's way of dissipating the resulting supersaturated condition was to take these waters over turbulent, shallow sections of the river that agitated the water, bringing the dissolved gasses in contact with the air-water interface where they could dissipate and return to the atmosphere.

The advent of large dams on many fish-bearing rivers and streams eliminated these turbulent sections by creating large, deep reservoirs that prevented effective dissipation of excess dissolved gas pressures. Dam spillways, when in operation and not properly designed, contributed more to the supersaturation levels. Hot water from power generating facilities (nuclear or other) entering these waters further contributed to this problem.

Unlike the air we breathe, the dissolved gas pressure and composition in water are highly variable and can be adversely affected by many different factors and conditions. The location, control,

regulation or elimination of these undesirable factors falls under the jurisdiction of many diverse disciplines: regulatory agencies such as the E.P.A., state, federal and private fisheries agencies, power companies and utility districts, the Departments of the Interior and Commerce, and private industry are all involved in controlling and maintaining a healthy aquatic environment for fish and fisheries-based industries.

The Weiss Saturometer, as a field portable accurate instrument to monitor dissolved gas parameters, has proven to be a valuable asset on a world-wide basis. Both rugged and inexpensive, fast, accurate readings can be obtained. At the monitoring site, the sensor membrane section is immersed in the water and the valve closed. A few minutes of agitation with the water pump (swishing back and forth) will indicate whether the water is supersaturated (pointer on gauge will move clockwise, +) or undersaturated (pointer on gauge will move counterclockwise, –). If undersaturated water is evident, no agitation or pumping is needed as air bubbles will not form on the tubing. If supersaturated conditions are present, vigorous agitation and pumping every 30 seconds of agitation should be used. If no change in the reading occurs, equilibration has been reached and that reading should be noted.

Other information should be recorded, preferably on a data sheet, and should include date, time, location, barometric pressure (either measured or calculated from weather service data and altitude charts), water and air temperature. If % nitrogen is needed, a dissolved oxygen determination should be made with either a D.O. probe or the Winkler method.

If tests at difficult sites or from a boat are required, it may be wise to use the wrist-straps on the ES-2 or tie a line around the handle to prevent loss of either type of instrument.

An alternative method of obtaining a saturometer reading is preferred by some—a pump or syringe is used to apply a positive pressure (or by sucking on the valve opening to provide a vacuum), then the valve is shut so a reading 50 to 100 mmHg above the expected reading is obtained. The instrument, immersed in the water to be tested, is then allowed to bleed down to the equilibration point. Agitation may still be needed in supersaturated waters. Both methods can be employed and the average value computed for very accurate readings. Either method takes generally 4-15 minutes to obtain a reading.

Factors that can alter the equilibration time and accuracy of the readings include:

1. Condition of the tubing—periodic cleansing of the membrane tubing in a detergent solution is recommended.

2. Temperature of the tubing—hot water immersion just prior to taking a reading speeds up reading time.

3. Water conditions—oil, industrial waste, other pollutants can coat the tubing adversely affecting equilibration time and instrument accuracy.

4. Restrictions to gas flow—swagelok fittings being tightened too far can pinch off the teflon tubing used to transfer pressure to the gauge. There are four "frame" windings in each

instrument and eight membrane tubing "ends" brought into the accumulator manifold. A pinch-off in one of the membrane frames will generally not impede accurate readings as the gas has essentially two directions it can flow to get to the manifold and gauge.

5. Leaks—a leak anywhere, no matter how small, will adversely affect both the equilibration time and final reading. NOTE: *The instrument must be maintained as a sealed system!*

Equilibration is defined as the point where the internal (dissolved) gas pressure equals the atmospheric pressure at the measurement location plus the gauge diaphragm and spring pressure that moves the gauge pointer to its stable position. After readings are taken, the lower section of the instrument should be rinsed off with fresh water, dried and the instrument should be stored in a plastic bag or suitable container away from excess humidity, direct sunlight or heat extremes.

If slow reading times are experienced, it may be due to the presence of water in the tubing. Long-term immersion is not recommended as some water vapor can traverse the membrane tubing wall and condense inside the tubing, causing a blockage condition. If this condition is expected, the instrument can be dried out in an electric oven at 180° F for one hour.

CAUTION!! *Do not dry the instrument or expose to open flame or fire as the tubing may be destroyed. Care should be taken to keep the gauge dry and free from corrosion. In taking field readings, do not allow foreign objects such as sticks, sand, etc., to enter the sensor area.*

C. Saturometer Use In Hatcheries, Research Facilities

An active saturometer-monitoring program in government or private hatcheries can prove of great value in preventing fish kills due to supersaturation or oxygen depletion and in regulating the aquatic atmosphere through dynamic control of aerators and source water selection, metering and composition control. Many hatcheries now employ almost totally artificial (man-made) control of the amount and type of dissolved gases present in their fish bearing waters. The advent of intensive fish farming programs and modern research facilities intensifies the necessity to accurately measure the composition and amount of dissolved gasses present. It has been proven that fish kills due to dissolved gas supersaturation are not necessarily due to nitrogen alone, but the sum total of all dissolved gas partial pressures—the exact condition that the Weiss Saturometers measure!

REMINDER: *It is still a necessity to maintain a proper nitrogen/oxygen balance to assure the fish have enough oxygen present to survive!*

Many hatcheries, fish farm facilities, and research institutes are now altering this nitrogen/oxygen balance to obtain optimum growth rates of fingerlings and fry, often with great success. The necessity of maintaining these de-aerated and then oxygen enriched waters at the optimal % saturation pressure dictates an active monitor and control program due to the complexity of these systems. One failure or mistake gone unnoticed for a given length of time may cause the loss of millions of young fish.

A sudden change in source water composition and % saturation occasionally occurs. The first warning signs of trouble without a saturometer-monitoring program are usually a lot of belly-up fish. With a monitoring program, such conditions can be detected and corrected before fatalities occur. The same is true if the aerator and circulations pump systems should a leak (usually at a fitting) in a high pressure or high velocity section of piping, or in a suction section on the inlet side of a pump occur, where atmospheric air can be injected into that point via venturi action causing supersaturation to be present past that point. If the condition persists, fatalities can occur. With a spot check saturometer-monitoring program, the leak can be located and fixed before fatal conditions occur.

Stagnant conditions can produce algae blooms, often in larger holding ponds. They produce prodigious amounts of dissolved oxygen that can reach harmful or fatal levels. By taking readings on a grid pattern using triangulation measurements, a thermal map can be constructed, pinpointing the algae bloom source. Localized herbicide then can be selectively applied to control this condition without poisoning the entire pond. And the presence of too many fish in a given body of water can lead to oxygen depletion. These conditions can be detected and corrected with an active saturometer monitoring program before fatalities develop.

These basic operating procedures outlined are used in hatcheries and other facilities. The ES-3 "mini" or lab saturometer may be better suited for use in smaller size fish tanks as it occupies less space and removes less dissolved gas from the water than the larger ES-2 instrument. The longer handle on the ES-2, however, allows hatchery personnel better access to outdoor holding ponds which may have water levels a few feet below the ground surface.

Saturometers can be sterilized with acetone, boiling water, dilute chlorine or other bacterial agents to prevent the possible spread of diseases from tank to tank or pond to pond.

D. Calculating Percent Saturation

Before immersion of the saturometer sensor section in water, the pressure gauge will indicate a zero reading. This is the point at which the pressure inside the instrument is equal to the local atmospheric (barometric) pressure. After a saturometer reading is taken, the gauge pointer will indicate a positive or negative dissolved gas pressure reading in millimeters of mercury. To convert this number into % saturation, the atmospheric pressure should be measured with an aneroid or mercurial barometer or calculated from altitude charts and local weather bureau or airport data. The following formula is then used:

$$\text{(Equation \#1) Percent (\%) Sat.} = \frac{P_{atm} + DP}{P_{atm}} \times 100$$

Where % Sat. is the total dissolved gas saturation (supersaturation or undersaturation), P atm = the local barometric pressure in mm Hg (760 mm Hg nominal at sea level), D P is the saturometer reading in mm Hg.
Example:

Barometric pressure is 765 mm Hg, P is + 100 mm Hg, therefore:

The actual dissolved gas pressure present in the water is 865 mm Hg.

NOTE: *For most applications this number (% Sat.) is the only number needed to assure safety to the aquatic environment and to assure legal requirements are within proper limits.*

E. Conversion Factors

Atomic Values: O_2 = 22.392 liters/mole
N_2 = 22.403 liters/mole
Ar = 22.390 liters/mole

Atomic Weights: O_2 = 31.9988 grams/mole
N_2 = 28.0134 grams/mole
Ar = 39.948 grams/mole

Composition of air: O_2 = 20.946%, N_2 = 78.084%, Ar = .934%

Concentration: 1 ppk = 1 mg/liter (approx./fresh water)
.001 gm/L = 1 oz./7400 gallons (approx./fresh water)
1 gm/liter = 1000 parts per million (approx./fresh water)

Conversion of ml/L to mg/L: 1 ml/L O_2 = 1.42903 mg/L
1 ml/L N_2 = 1.25043 mg/L
1 ml/L Ar = 1.78419 mg/L

Distance: 1 ft. = .3048 meters
1 meter = 3.281 feet

Hydrostatic Pressure: 1 meter of fresh water @ 20° C = 73.43 mm Hg
1 foot of fresh water @ 20° C = 22.89 mm Hg
1 meter of seawater @ 35 ppt, 20° C = 74.88 mm Hg
1 foot of seawater @ 35 ppt, 20° C = 23.34 mm Hg

Pressure: 1 Std. atmosphere = 760 mm Hg = 1.013 Bar = 10.35 meter (32.2 ft.) of fresh water at 20° C = 10.15 meter (32.56 ft.) of seawater at 35 ppt and 20° C = 14.22 lbs./sq. in. = 101.325 kilopascals = 760 torr @ 0° C = 1 kg/cm^2

Temperature: °C = 5/9 (F° - 32) °F = (9/5 °C) + 32 °K = °C + 273.15

Volume: 1 cc = 6.1 X 10^{-3} cu. in. 1 cu. ft. = 28.32 liters

REFERENCE

ECO Enterprises, 1985. *Weiss saturometer field manual for the ES-2 & ES-3 instruments*. ECO Enterprises, 2821 N.E. 55th, Seattle, WA 98105. (206) 525-4784 or (206) 523-9300; also, 1-800-426-6937 (Exc. WA).

Beach Seining and Otter Trawling

INTRODUCTION

A. Otter Trawls

Otter trawls are commonly used in surveys of demersal species, particularly in marine ecosystems and estuaries (Murphy and Willis, 1996). The general purpose of otter trawling is to provide indices of abundance of different species of finfish and shellfish and provide biological information for understanding their population dynamics. For research purposes, otter trawling is usually conducted from 5 meters to 150 meters in depth. Many bottom trawl surveys are combined with midwater trawling and hydroacoustic surveys (Hayes *et al.*, 1996).

One reason for the popularity of trawls is that they sample a discrete area of the bottom, or volume of the water column, over a specified time. While trawls are quite versatile in the variety of habitats they can sample, they do have some limitations. Trawls cannot be effectively used on bottom substrates that can snag the net. Coral reefs, aquatic macrophyte (kelp) beds, and rocky outcroppings are examples of habitats that can't be sampled with otter trawls.

B. Beach Seines

A beach seine is a type of encircling net. Encircling nets are used to trap fish actively by surrounding them in a fence-like wall of netting. Surrounding nets provide several advantages over otter trawls because the gear is easy to deploy, sampling is rapid, a large area can be sampled, the limits of the sampling area are well defined, and fish are captured live with minimal trauma (Murphy and Willis, 1996). These nets also allow for live release (Pierce *et al.*, 1990). Beach seines, unlike otter trawls, can be used without a boat, and can be fished by a single person.

Beach seines are often fished in a semicircle around the targeted fish and dragged to shore, herding the fish into the net (Murphy and Willis, 1996). Setting and hauling beach seines can be done in several ways. One method is for a single vessel or person wading in shallow water to fix a towline on shore and then to set a wing and the bunt offshore before turning in to shore and setting the second wing and towline. It's common to set the net up-current or up-wind in some cases, to give the net a chance to open to its greatest width. However, the lead-line must be well anchored to prevent the seine from lifting off the bottom if this technique is used. The net is hauled in by dragging in the towlines until the bunt reaches shore and the catch can be retrieved.

PROCEDURE

Many studies in which beach seines are used rely on a single haul to represent the fish community in the area being seined (Allen *et al.*, 1992). These researchers showed that species richness, species rank, and size distribution of dominant taxa were well represented by the first haul.

During the next two field exercises we will gather data that will either support or refute the above research findings.

Equipment and Supplies:

- 2 Boston Whaler boats
- 1 research otter trawl
- 1 straight beach seine
- field notebooks
- layered clothing
- raingear, hip boots, knee boots and/or waders
- this lab exercise and data sheets
- 6 five gallon plastic buckets
- 6 plastic specimen tubs
- camera and binoculars (optional)
- species identification keys (adults and juveniles)
- 2 YSI dissolved oxygen meters
- 2 YSI salinity, conductivity, temperature meters

The class will split into two teams (blue and gold crews). The blue team will do beach seining, with the instructor while the gold team will perform otter trawling with our instructional assistant. During the next lab/field period, teams will switch; the blue crew doing the otter trawling and the gold crew beach seining.

A. Beach Seining

Three side-by-side (replicate) beach seines will be made each sampling day. Teams should mark off three 150' sections along the beach by placing stakes in the sand/mud. The instructor will provide on-site details as to which beach seining method to use. Use the data sheets provided to record your observations and measurements.

You will be recording *species* of plants and animals found, *numbers* of each species, and you will be making some basic water quality measurements (dissolved oxygen, salinity, conductivity and water temperature) for each replicate.

NOTE: *Be sure to record routine observations in your field notebooks!*

B. Otter Trawling

Three (3) replicate otter trawls will be performed. Each trawl will be exactly 5 minutes long (once the trawl reaches the bottom).

The instructor will provide specific details as to the direction of tows, length of tows, and the setting and retrieval of the trawl. Like seining, you will be recording *species* of plants and animals found, *numbers* of each species found, and you will be making some basic water quality measurements (dissolved oxygen, salinity, conductivity, and water temperature) for each replicate. And again, be sure to record routine observations in your field notebooks.

C. Departure and Return

We will leave _____by vans at _____. We will return at _____. Be sure to develop a check off sheet for this exercise.

NOTE: *The instructor will ask you to review your list before we leave on this field exercise.*

LAB PRODUCTS

At the end of these lab/field exercises, you will be responsible for:

1. Rinsing the beach seine and trawl net with fresh water, and stretching them out to dry in the designated area.

2. Rinsing or wiping off all other equipment, boats and gear with fresh water, and temporarily placing all gear and equipment in the designated drying area. Once it's dry, stow it where you got it.

3. Complete your otter trawl and beach seine data summary sheets; make sure your field notebooks are complete; write a 4-5 page, word processed (or typed) *individual* research report about your results for these two lab/field exercises. Reports should contain a title page, table of contents, introduction, methods and materials, results and discussion, summary and/or conclusions, and bibliography. Use tables and graphs as needed. Your raw data tables and summaries can go in your appendix. Photos can be used as figures in your report.

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Allen, D.M., S.K. Service, and M.V. Ogburn-Mattews. 1992. *Factors influencing the collection efficiency of estuarine fish*. Transactions of the American Fisheries Society 121: 234-244.

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Marrow, J.E. 1980. *The freshwater fishes of Alaska*. Alaska Northwest Publishing Co., Anchorage, AK.

Scott, W.B. and E.J. Crossman. 1973. Bulletin 184. *Freshwater fishes of Canada*. Canadian Government Publishing Centre, Ottawa, Canada. ISBN 0-660-10239-0.

Names: _____ Beach Seine Data Sheet

Location: _____ Beach Seine #: _____ Stage of Tide: _____

Date/Time	Common Name	Genus & Species	# Sampled	Comments

Dissolved Oxygen: _____mg/L Salinity: _____ $^{o}/_{o}$ (ppt)

Conductivity: _____ µmhos Water Temperature: _____ ^{o}C

Page ___ of ___

Names: _____
_____
_____
_____

Beach Seine Data Summary

Date of Collection:_____
Location: _____

Common Name	Genus & Species	Total Number Per Seine	Comments
		Seine #_____ Seine #_____ Seine #_____ $\bar{x}$ = _____ S.D. = _____	

Names_____

Otter Trawling Data Sheet

Location: _____ Trawl #: _____

Stage of Tide: _____ Estimated Length of tow: _____ meters

Date/Time	Common Name	Genus & Species	# Sampled	Comments

Dissolved Oxygen: _____mg/L Salinity: _____ $^{o}/_{o}$ (ppt)

Conductivity: _____ µmhos Water Temperature: _____ °C

Page ___ of ___

Names: _____
_____
_____
_____

Otter Trawl Data Summary

Date of Collection:_____
Location: _____

Common Name	Genus & Species	Total Number Per Seine	Comments
		Trawl #_____ Trawl #_____ Trawl #_____ $\bar{x}$ = _____ S.D. = _____	

NOTE FOR INSTRUCTORS

NOTE: *Other types of sampling such as trap netting or gill netting may be substituted for the otter trawling if desired, as otter trawling is specific to marine sampling and specific equipment, such as boats with sufficient horse-power, is required for this exercise.*

BACKGROUND INFORMATION

A. Otter Trawls

Otter trawls are commonly used in surveys of demersal species, particularly in marine ecosystems and estuaries (Murphy and Willis, 1996). The general purpose of otter trawling is to provide indices of abundance of different species of finfish and shellfish and provide biological information for understanding their population dynamics. For research purposes, otter trawling is usually conducted from 5m down to 150 m in depth. Many bottom trawl surveys are combined with midwater trawling and hydroacoustic surveys (Hayes, Paola Ferrei, and Taylor, 1996).

A typical otter trawl features a head rope (with floats), a foot rope (with a chain to weigh the net down and scour the bottom), top and lower wings, a mouth and cod end, bridles that connect to a pair of trawl doors (that flare outward to keep the wings of the net open when towed), and two towing ropes that connect to a single or double set of booms on the towing vessel (Byrne and Nicolas, 1989).

Otter trawls are towed along the bottom. As the net is towed through the water, fish entering the net eventually tire and are "funneled" to the cod end where they are held until the net is retrieved. Most trawls are designed so that the cod end is tied shut while fishing and can be opened after retrieval, to make removing fish from the net easier (Hayes, Paola Ferrei, and Taylor, 1996). Most bottom trawls for scientific programs use otter doors. Otter doors for bottom trawls are relatively heavy in order to keep the net on the bottom, as well as to keep the wings of the net spread open (APHA, 1985). Most otter doors are rectangular and keep lateral pressure on the net by having the towing warps attached to front brackets, while bridles are attached to the back brackets. The bottoms of the doors typically have metal shoes to protect the doors from abrasion with the substrate (Byrne and Nicolas, 1989).

One reason for the popularity of trawls is that they sample a discrete area of the bottom, or volume of the water column, over a specified time. While trawls are quite versatile in the variety of habitats they can sample, they do have some limitations. Trawls cannot be effectively fished on bottom substrates that can snag the net. Coral reefs, aquatic macrophyte (kelp) beds, and rocky outcroppings are examples of habitats that can't be sampled with otter trawls.

Unless the trawl net is relatively small, it generally requires a powerful vessel to pull the net along the bottom. It also takes two or more people to handle an otter trawl safely, both in the water and when the net is on-board.

B. Beach Seines

A beach seine is a type of encircling net. Encircling nets are used to trap fish actively by surrounding them in a fence-like wall of netting. Surrounding nets provide several advantages over otter trawls because the gear is easy to deploy, sampling is rapid, a large area can be sampled, the limits of the sampling area are well defined, and fish are captured live with minimal trauma (Murphy and Willis, 1996). These nets also allow for live release (Pierce et al, 1990). Beach seines, unlike otter trawls, can be fished without a boat and can be operated by a single person.

The beach seine is made of mesh of uniform size and consists of two wings and a bunt section that holds the catch. In some seines, the bunt is enlarged to form a bag in the netting. The wings form a long vertical wall that funnels fish to the bunt. The beach seine also features a cork line at the top and lead line at the bottom of the net. A 5-6 foot tall pole is also attached to each wing. Towing warps or bridles can be attached to these poles.

Beach or haul seines are typically used in shallow waters where the net wall can extend from the surface to the bottom. Seines with a bagged bunt work better in lakes; bagless seines are used in rivers.

Beach seines are often fished in a semicircle around the targeted fish and dragged to shore, herding the fish into the net (Murphy and Willis, 1996). Setting and hauling beach seines can be done in several ways. One method is for a single vessel or person wading in shallow water to fix a towline on shore and then to set a wing and the bunt offshore before turning in to shore and setting the second wing and towline. It's common to set the net up-current or up-wind in some cases, to give the net a chance to open to its greatest width. However, the lead-line must be well anchored to prevent the seine from lifting off the bottom if this technique is used. The net is hauled in by dragging in the towlines until the bunt reaches shore and the catch can be retrieved.

A second method requires two vessels or two people wading in shallow water. In this case, the two vessels or people start together at the farthest point from the beach and set the bag. They open the seine parallel to the beach, each setting a wing and towline. Then the seine is pulled toward the shore, and the wings are brought together. The bag with the catch is held between the vessels or hauled ashore where the catch can be collected. Another common method of seining is for two fishers to set the net perpendicular to shore and then drag the net parallel to shore. At the end of the tow, the person farthest out turns in to shore and both wings are pulled ashore (Murphy and Willis, 1996).

Nekton Sampling

INTRODUCTION

Nekton are all the larger, aquatic, free-swimming animals in lakes, seas, and ponds, whose movements are largely independent of currents and waves. Some species considered to be nekton include fishes, squids and whales.

PROCEDURE

Today we will perform nekton sampling in Lake_____. We will be using hook and line to catch and tag resident cutthroat trout. We will use the methods from the *Mark and Recapture lab* to mark, release and recapture the cutthroat.

Equipment and Supplies:

- light spinning rod and reel (a hand line will do)
- bobbers
- worms (or other preferred baits)
- #6 or #8 worm hooks
- polarized sun glasses (optional)
- dip net
- spaghetti tags and tagging gun
- field notebook and nekton data sheets
- a *current & valid* Washington State fishing license
- keys to freshwater fishes (adults and juveniles)
- Washington State Fishing Regulations
- *...and don't forget to bring your luck!*

You are welcome to fish *anywhere* you like in Lake _____. You can fish individually or in teams of two.

Identify, count, and *release all* of the different species you catch today. Tag and release *only* cutthroat trout. Record all catch data on the data sheets provided.

LAB PRODUCTS

1. Read over this lab exercise and the current state fishing regulations for Western Washington lowland lakes. Also review the mark-recapture protocol.

2. *Fish like crazy for 3 hours and 50 minutes!!*

3. Record your catch data.

4. Turn in a 1-2 page individual report at the beginning of the next lab. Include advantages and disadvantages of hook and line as a nekton sampling method. Try to describe *the best technique* you can for catching cutthroat trout with hook and line (you are encouraged to consult with some of the more expert fishers in our class for this information!).

   Both individual and class results will be summarized. Try to explain the results you got and how they compared with data for the entire class.

REFERENCES

Eschmeyer, W.N. and E.S. Herald. 1983. *A field guide to Pacific coast fishes of North America.* Houghton Mifflin Co., Boston, Mass.

Samplers: _____

Nekton Sampling
Individual Data Sheet

Location (General): _____
Specific Location: _____

Date/Time	Common Name	Genus and Species	Number Caught	Number Marked	Number Released	Comments

General Information (techniques used; bait used; number of fish injured or killed; etc.):

Nekton Sampling
Class Data Sheet

Location: _____

Date/Time	Common Name	Genus and Species	Number Caught	Number Marked	Number Released	Comments

Capstone I: Team Building

INTRODUCTION

As we begin our "capstone" field project, we will focus on developing teamwork, project planning, self-esteem and self-confidence. We will also get a chance to practice our leadership, supervisory, and following and listening skills. One excellent way to begin enhancing all of the above behaviors, attitudes and skills is to participate in the **ROPES/Challenge course.**

NOTE: *ROPES is an incredibly effective process to develop teamwork*.

PROCEDURE

Wearing a sturdy pair of shoes with good arch and ankle support is essential. Bring layered clothing and raingear (NOTE: *we won't cancel if it rains or snows!*).

You don't have to be afraid of some of the more difficult challenges (i.e., the "pamper" pole and "hole-in-space"). You can ask your team's permission to excuse you from certain events. You'll be amazed just how much you will be able to accomplish, both individually and as a team!

LAB PRODUCTS

Following each event, teams will reflect on the significance of that event, either to us as individuals, or as a team. These periods of reflection are definitely as important as the physical challenges themselves.

There will be no write-ups due from this field exercise; however, you should reflect on this experience in the diary portion of your journal.

NOTES FOR INSTRUCTORS

This activity is one of the first "capstone" activities we do, starting about midway through the quarter. It provides opportunities for students to put into practive what they've learned thus far in this course. This type of activity is recommended to help students pull together their collective knowledge and apply what they've learned.

The ROPES Challenge Course is an example of an innovative team-building exercise. We've used it with adult (instructor) groups as well as students, and responses are almost always positive.

These courses are available mainly through outdoor adventure centers throughout the country. Or you may find this website useful, and a brief summary follows:

www.adventureropes.com/

Ropes Course

"We are a full service ropes/challenge course provider. We design, we build, we train, we teach. We conduct inspections on existing challenge courses and we perform upgrades on those courses to bring them into compliance with current safety standards. Our goal is to make the ropes course experience available to everyone who is willing to accept the challenge. We believe that experiential education is the most powerful tool ever developed to assist in learning."

Capstone II: Ecosystem Study

INTRODUCTION

This "capstone" will provide you the opportunity to put into practice all of the knowledge and skills learned in previous classes, and specifically, those skills learned in the first five (5) weeks of this course.

PROCEDURES

I. Project Planning for your Research Project

A. Brainstorming—Affinity and Tree Diagrams
B. Defining Scope-of-Work
C. Developing Tasks and Time Lines
D. Prioritizing Tasks
E. Assigning Personnel
    1. Individual assignments
    2. Team assignments

II. Project Management for your Research Project

A. Leadership and Supervision Skills
B. Teamwork Skills
C. Continuous Quality Improvement (CQI)

III. Project Completion and Delivery

A. Final Research Project
B. Oral Presentations

NOTES FOR INSTRUCTORS

THE CAPSTONE RESEARCH PROJECT

I use a *learning community approach* in this project. I am including some notes on learning communities in this section. Included are guidelines for student project planning and development, and presentation requirements.

A. SAMPLE RESEARCH REPORT OUTLINE

This is a sample of a research report structure that could be provided to students.

TITLE: *Management Implications of Rockfish Tagging Research off the Southwest Washington Coast*

I. Introduction

Begin with a general discussion of black rockfish; why tagging is necessary; past tagging efforts; and end with the purpose (or focus) of this research report (NOTE: *In a critical essay, this is your thesis statement*).

II. Life history and habitat of black rockfish

- A. Life history
- B. Habitat

III. Importance of black rockfish

- A. To humans (commercial and recreational fishing)
- B. To other animals (the food web)

IV. Description of the study area

- A. Use a section of a coastal navigation chart
- B. Photographs taken of coastline
- C. Describe the coastal features and bottom characteristics of the area from USCG's Coast Pilot

V. Description of research vessel and gear

- A. Interview a captain of a research vessel
- B. Photos taken during interview

VI. Methods and materials

A. Personnel

1. WDF&W captain, crew and biologists
2. Volunteer fishermen

B. Equipment used to locate rockfish

1. LORAN and GPS
2. Radar and side scanning sonar

C. Gear and bait used to catch rockfish

1. Poles and reels
2. Bait and lures

D. The tagging process

1. Tags (photos and/or drawings
2. Tagging techniques (describe protocol; include data sheet as a figure)

VII. Results and discussion

A. Compare past and present populations (use tables and graphs here)
B. Describe the current management plan for black rockfish in Washington State
C. Results of interviews regarding the effectiveness of this research with:

1. The captain and crew
2. State fisheries biologists
3. A volunteer fishermen
4. Personal opinion (as an observer)

VIII. Summary and/or conclusions

A. Discuss how the tagging results will influence future management of black rockfish.
B. Summarize how important it is to include the public in a state research project.

IX. Bibliography or references cited

A. Journal reprints
B. Personal communications (interviews)
C. Articles from sport fishing magazines

X. Appendices

A. Tagging protocol used by state agencies
B. Actual data sheets taken from tagging trip
C. State management plans for black rockfish
D. Recreational fishing regulations for black rockfish

B. ORAL PRESENTATIONS OF RESEARCH REPORTS

I. Preparation

Your presentation should last no longer than 20 minutes. You should allow 3-5 minutes for questions and answers. Your report should include:

a. *an introduction* ("tell them what you are going to tell them")
b. *the body* of your talk ("tell them")
c. *a summary* ("tell them what you just told them").

NOTE: *The format and organization of your presentation is just as important as its content!*

AV equipment and supplies will be available for your presentation. Ask the instructor or assistant to help you with these. They include:

a. white board (diagrams, drawings, etc.)
b. handouts
c. overheads (the instructor will provide)
d. slides
e. posters
f. Power-Point software

Useful tips:

- Remember to *practice* using AV equipment *before* your presentation.
- Practice your presentation in front of a mirror, your family, your roommate, your dog, or a fellow student.
- Time your presentation so that you finish in 16-17 minutes, allowing 3-4 minutes for questions from your audience.
- Note cards are acceptable, but *practice enough* so you only have to rely on them in a pinch. Do not read from them!
- Always face your audience, even when talking from a slide, an overhead, or the white-board. Relax and smile at your audience. Keep an eye on your audience to see if they are "in tune" with your presentation.
- Don't lean on the podium. Don't confine yourself to the podium.
- Walk around, it will help you to relax.

STUDENT-DEVELOPED PROGRAM OBJECTIVES

1. To learn about and practice effective leadership, management and interpersonal relationship skills—supervision, planning and organization, plan implementation, progress tracking, communications, coordination and cooperation.

2. To gain confidence and critical skills through practice of all five elements of language—reading, writing, speaking, thinking, and listening.

3. To gain perspectives on local and global social, technological and natural resource issues from a review of historical and contemporary writings, videos and discussions (seminars and guest speakers). To be comfortable with answers to those questions regarding these issues that are often incomplete and ambiguous.

4. To learn about and practice state-of-the-art techniques related to:

   - biological and chemical field and laboratory sampling and recording of data
   - experimental design and analysis of laboratory and field collected data utilizing existing data base statistics and graphics software
   - literature searching
   - technical writing for local and state agencies using prescribed formats; using advanced word processing of general and technically written letters, reports
   - grant writing
   - production of slide/tape, video and poster presentations
   - organizational work plans and progress evaluation

5. To learn the "art of seminaring"; especially, how to think and speak critically and ask good questions of texts, teachers, our fellow students and ourselves.

6. To gain perspectives on our own bias by considering the views of others.

7. To learn and practice the different stages of the writing process (general and technical)—gathering materials, shaping it into writing, revising what one has written, and editing it for publication and public presentation.

8. To practice working with others in exploring ideas, solving problems, and aiding others in development of basic skills.

9. To experience delight, excitement, fear, rapture, astonishment, and a host of other emotions that creative writing, thinking, and risk-taking can awaken.

10. To become better aware of and develop a personal framework of regarding ethics in the work place, journalism, our scientific endeavors, and conservation of natural resources.

11. To explore, through works of literature and textbooks, fundamental questions of our values, epistemologies, and lives in relation to technological change.

12. To learn to participate effectively and comfortably in an oral exchange of ideas about significant literary works; to be able to listen and to formulate ideas spontaneously and communicate them to others in large and small groups.

USING THE LEARNING COMMUNITY/LINKED COURSE APPROACH

The overall delivery of this research study will take on the nature of a "learning community," either as one which is done formally (team taught by two or more faculty), or informally, where two or more faculty "link" their individual courses together (i.e., Aquatic Field & Lab Methods *and* Technical Writing). Both will attempt to accomplish learning outcomes that *jointly* satisfy the requirements of each of the individual courses. Students may be enrolled in just one or perhaps *both/all* of the linked courses.

LEARNING COMMUNITIES—MARKETING AND ADVERTISING

Learning communities, if they involve team teaching of multiple courses, often require campus-wide marketing and advertising. A poster or advertisement included in the college's course catalog, or in the local newspaper, is an effective way of getting the message out about the course. It is important to inform and educate *all* divisions of the college (admissions, councilors, faculty, staff, administrators, etc.) about your learning community at least one quarter before it is taught. Planning by the faculty co-instructing a learning community should start at least two quarters in advance. *One-year ahead of time is not too far ahead!*

COURSE OBJECTIVES/TIME LINES

During the first week of the learning community, faculty and students will:

- Develop course objectives (requirements)
- Develop a typical weekly schedule
- Develop a corporate mission statement. The mission statement will continue to be developed throughout the quarter.

Brainstorming is used extensively during the first two weeks to develop an affinity diagram, a tree diagram and a set of time lines for the research study. These are all continuous quality improvement (CQI) tools that allow student to practice management, leadership, teamwork, organizational, facilitation, listening and following skills. Other quality management tools used are flow diagrams, fish bone diagrams and multivoting (nominal group technique) for gaining consensus. *The Memory Jogger Plus +* is an inexpensive pocket guide for students to learn these management and leadership tools.

SEMINARS

Seminaring is a key ingredient of a learning community. One of the major goals of our learning communities is to explore ecological and technological issues from various (i.e., organizational, historical, economic, social and political) perspectives.

ASSESSMENT

Assessment is considerably different (and usually more comprehensive) in a capstone learning community than a traditional class. Some of the assessment tools used include: a written self-evaluation by each student, an instructor-written evaluation of the student, comprehensive peer and instructor evaluations, and a portfolio which includes all of the "end products" (learning outcomes) including a final research report. This portfolio has proven very useful to the student/graduate for: 1) application to a 4-year college or university; and 2) applying for a job. It tends to provide a much more comprehensive assessment of the student than just a grade on a transcript.

ORAL PRESENTATIONS

Students will make presentations to both general and professional audiences at the end of the field exercise.

CHECKLIST FOR A SUCCESSFUL MEETING

Before the Event ~

_____ Firm up the entire program well in advance; one to two months, if possible.
_____ Be certain speakers know their topic and time requirements.
_____ Outline program contents for publicity channels, including places and dates.
_____ Complete planning for all physical facilities:

a. Meeting spaces; lunch and break areas.
b. Adequate exhibit spaces.
c. Registration supplies and personnel.
d. Audio-visual equipment and assistants.
e. Signs and bulletin boards for guidance of invited guests and speakers.
f. Invitations to guests, presenters, and the media.

_____ Line up community support and participation.
_____ Line up staff of hosts and hostesses.
_____ Print or copy brochures, programs.
_____ Arrange for photography and photographer.

During the Event ~

_____ Make sure someone is in charge; that everyone knows their responsibilities.
_____ Start sessions on time; announce at the beginning that this will be standard procedure.
_____ Keep sessions and speakers on schedule

_____ Public address, recording and audio-visual equipment:
  a. Have it ready and warmed up ahead of schedule; check that extra light bulbs are available for overhead and slide projector; if using hi-tech. computer equipment, learn how to use it, if passwords are required, etc.
  b. Have qualified operators available.
  c. Allow rehearsal time for all speakers to practice with A/V equipment.
  d. Assign someone to turn lights off and on.

_____ Make a good photographic record of the program.
_____ Keep the program on schedule.
_____ Give credits to the planners and host/hostesses.

After the Event ~

_____ Personalized thank-you letters to speakers and assistants.
_____ Return all borrowed equipment and/or supplies.
_____ Pay all bills promptly.

SUMMARY

Since each capstone learning community is "student-driven," there is no single outline to use as an example. Students are allowed many choices during the quarter. Faculty must relinquish the "control" or "authority" normally held in a traditional course. This *does not* mean, however, that the students will not be productive. The opposite is most often true. Once the synergy of the "community" kicks in (this often happens in the third to fifth week of the quarter, when students finally accept the fact that they are 100% responsible for *their own* learning), near miraculous learning occurs, and learning outcomes are accomplished within the prescribed time lines. All of this requires much "risk taking" on the parts of both the faculty and students making up the community.

TEMPLATE FOR END-PRODUCTS

The following is a template with examples for constructing a list of "end product" requirements to be completed by each student.

NOTES: *Instructors can insert categories here of their own design, based upon the end-products they decide upon. Students should be directed to pay close attention to specific due dates of each end product.*

End Products	Due date	Date Completed
Field notebook or journal	_____	_____
Written research report	_____	_____
Oral presentation of research	_____	_____
Demonstration of lab skills	_____	_____
Self evaluation	_____	_____
Instructor evaluation of student	_____	_____
Peer review	_____	_____

Guidelines for grading oral presentations of research	
Criteria	**Percentages**
Organization	20%
Content	20%
Use of AV materials	20%
Answers to Questions	20%
Public Speaking Skills	20%
Total	100%

SELF-ASSESSMENT PEER REVIEW OF PERFORMANCE

Name	Seminar	Lab/ Field Partici-pation	Project Plan-ning	Team-work	Leader-ship	Research Report (1st Draft)	Research Report (Final Draft)	Notes

To students:
Please rate yourself and your teammates performance with regard to the following area zero (0) being the lowest, five (5) the highest. Your ratings will be kept strictly confidential.

INSTRUCTOR EVALUATION OF STUDENT'S PERFORMANCE

Name	Seminar	Lab/ Field Partici-pation	Project Plan-ning	Team-work	Leader-ship	Research Report (1st Draft)	Research Report (Final Draft)	Notes

REFERENCES

Brassard, M. 1989. *Memory Jogger Plus +*. First Edition. GOAL/QPC. 13 Branch Street, Methuen, MA 01844.

Department of the Navy. 1992. *Handbook for Basic Process Improvement*. Chief of Naval Operations Executive Steering Committee and the Department of the Navy's TQL Office, Washington, D.C.

Weekly Seminar Assignment #1

Definitions of Ecosystem Management and Related Terms

Available on the Web at: classes.aces.uiuc.edu/NRES325/defin.html (Pages 1-8)

Answer the following questions regarding our seminar reading for this week. Write your answers on separate sheets of paper. You may also find it useful to jot down comments, questions, and page numbers on a separate sheet to bring to seminar.

1. What did Aldo Leopold and Eugene Odum say about ecosystem management? (pg 1) Roughly, what year(s) did each of these "visionaries" make these statements?

2. Write down, sentence by sentence, the entire definition of ecosystem management authored by the Federal Interageney Ecosystem Management Task Force (June 1995). After each sentence, write in your own words, what you think the Task Force meant by each statement. Continue until you have completed the definition. (pg 1)

3. What else does the Task Force's approach emphasize? (pg 1)

4. List and discuss the seven (7) elements that make up ecosystem management (pg 2) according to the Ecological Society of America's (ESA's) "The Scientific Basis for Ecosystem Management (1995)."

5. Summarize in one paragraph, what the ESA says about ecological science as a basis for ecosystem management. (pp 2-3)

6. What are some *basic differences* between the positions taken on ecosystem management between the U.S. Fish and Wildlife Service and the U.S. Forest Service? (pg 4)

7. Differentiate between ecosystem "health" and ecosystem "integrity." (pg 5)

8. List several ecosystem heath goal, objectives and indicators. (pg 6)

9. What are the five (5) main ecosystem management goals listed by Edward Grumbine?

10. What is biotic integrity according to James R. Karr?

Weekly Seminar Assignment #2

"Learning to Work Together"

Reference Material: *The Team Handbook—How To Use Teams To Improve Quality*. 1992. 18th Printing. Joiner Associates, Inc. Madison, WI.

You will be responsible for the contents of Chapter 6 of the "Team Handbook." No formal written questions will be required; however during seminar time you are required to reflect upon and write your responses to Question #1 (below) in the Diary Section of your journal.

Seminar (First Session)

1. After reading and studying Chapter 6 of the Team Manual thoroughly:

    a. Where do you think your team is at this time regarding: *forming, storming, norming or performing?*

    b. If your team has progressed past the forming stage, describe the *behaviors* that existed at earlier stages, and those *behaviors* that exist now (15 minutes for this exercise). Record behaviors at each stage on flip chart.

    c. What behaviors describe your team at your current stage of team development?

2. Discuss the ten (10) ingredients for a successful team (pp 6-11 to 6-21) regarding:

    a. *Indicators of potential trouble.* Which of these trouble indicators apply to your team? Brainstorm around your circle of members, having each team member offer one trouble indicator (that applies to your team) per round (no criticizing or discussion for this exercise; it is okay to pass). Facilitator should keep this moving fairly quickly. Recorder should write responses on a flip chart. Be sure however, that everyone gets a chance to get all of their "indicators" out in the open, and up on the chart. Go around the circle as many times as needed to complete this (about 20 minutes for this exercise).

    b. Brainstorm again, addressing the question "What can I do to help my team progress to the performing stage?" If you are already at the performing stage, address the question "What can I do to make our team perform even better?" Each member should begin his/her statement with: *"I can help our team be more successful by . . ."* Again, the facilitator should keep this moving, and the recorder should use a flip chart (allow about 20 minutes for this exercise).

Seminar Continues (Second session)

3. Teams get back together. Reporters use flip charts to report to the entire class. (Allow 10 minutes *maximum* for each team report. Time keepers—remind facilitator two minutes before time is up).

4. Class evaluation of this seminar exercise (10 minutes).

Quiz I

1. Define the following terms and list a living representative of each (be as specific as possible).

    a. plankton

    b. periphyton

    c. macrophyton

    d. macroinvertebrates

    e. autotrophic

    f. heterotrophic

2. Explain the significance of each of the following as indicators of polluted and/or clean water.

    a. plankton

    b. periphyton

    c. macrophyton

    d. macroinvertebrate

3. List the five (5) essential pieces of information to be included on sample labels that are inserted into sample containers immediately as plankton samples are collected.

Quiz II

1. Define the following biometrics terms:

    a. statistic

    b. biometrics

    c. parameter

    d. standard deviation

    e. observation

    f. mode

    g. character of interest

    h. universe

    i. experimental unit

    j. population

    k. sample

    l. frequency histogram

2. What is a normal distribution curve? Why are comparisons between our frequency polygon and the normal distributions curve useful?

3. List two (2) types of statistics that describe central tendency and two (2) that describe measure of dispersion. Briefly explain what they are.

4. Consider the following sample set of numbers: **2, 3, 3, 4, 4, 4, 8, 10, 12**

a. Construct of frequency table using the above data.

b. Find the range.

c. Calculate the mean.

d. What is the median?

e. What is the mode?

f. Average deviation?

g. Variance?

h. Standard deviation?

i. Draw a frequency polygon using the data (sample set of numbers) provided.

Sample
Mid-Term Exam
(100 points)

Answer *any* five (5) of the following. (use a spearate sheet of paper).

1. Explain how you would use the backpack electroshocker to sample fish populations in a creek. Be specific about start-up and shut-down procedures, voltage and wattage settings, type of current used and why, and what water quality measurements must be made prior to use of electroshocker? Why? What precautions should be taken to avoid undue stress to fish?

2. Describe in detail the use of seines, trawls, chemicals, and hook and line fishing as active fishing methods. Give advantages and disadvantages of each method and tell when each might be the preferable method over the others.

3. Describe two (2) types of entanglement devices and two (2) entrapment devices used in passive fish sampling. Be specific about how, when, and where used. Compare advantages and disadvantages.

4. Define plankton, periphyton, macrophyton, and macroinvertebrates. Be specific about the types of plants or animals that make up these major groups and where they might be found in the aquatic or marine environment. How do they serve as an "indicator" of pollution? Again, cite specific examples.

5. Define five (5) of the following biometric terms:

   a. experiment
   b. observation
   c. population
   d. sample
   e. character of interest
   f. universe
   g. experimental unit
   h. parameter
   i. statistic

6. Explain how you would determine the "standing plankton crop" or "biomass" (both qualitatively and quantitatively) in a lake. Be specific about equipment and techniques used in sample collection and analysis.

7. Your assignment is to research the change in the periphyton communities related to the installation of a secondary treatment plant on the _____ River. List all of the major and incidental observations that should be included in your field notes. You collect several periphyton samples to analyze in the laboratory. What information should be included in your field notes? What information should be included on the sample label you insert in the collection container?

8. List at least five (5) major types of observations to be included in field notes when collecting plankton or fish in a lake. Also list at least five (5) pieces of information to be included on labels to be inserted into sample container.

9. What is ecosystem management? What is adaptive management? In your discussion compare the way we have managed natural resources in the past with these new management philosophies, strategies and practices.

Oral Exam and Study Guide

1. You will be asked to verbally define and discuss five (5) of the terms from the attached list of watershed related terms. The instructor will randomly select terms, so you'll have to know them all.

2. Describe (in detail) each of the following physical, chemical and biological sampling procedures. Describe in terms of equipment needed, protocol for sampling and analysis, and the nature of the data collected (units of measurement, description of data sheets, use of data):

- Plankton sampling and analysis
- Surber sampling
- Otter trawling
- Beach seining
- Flow (water velocity or discharge)

3. Be prepared to discuss benefits (or disadvantages) of Continuous Quality Improvement (CQI) as a set of project planning and management tools to carry out our research study this quarter (i.e., did they help or hinder our goal to complete several major team "end products?"). You will be graded strictly on how well you defend your point of view (pro or con); not on what you think I want to hear.

4. Be prepared to discuss pros or cons of the "learning community" (teamwork, seminars, students teach other students, journals, oral presentations, portfolios) approach to learning versus more traditional approaches. Again, you will be graded on how well you defend your point of view; not on what you think the instructor wants to hear.

Watershed Related Terms

The following is a list of watershed-related terms that will be "fair game" for definitions and description for the final oral exam:

Accuracy
Adaptive management
Alkalinity
Baseline survey
Bed load
Benthic
Biodiversity
Bioregion
Buffer strips
Canopy
Channel forming discharge
Community
Conductivity
Coniferous
Control
CQI (Continuous Quality Improvement)
Cover
Cubic feet per second
Cumulative effect
Debris flow
Deciduous
Discharge
Dissolved load
Dissolved oxygen
DOM (Dissolved Organic Matter)
Ecosystem
Ecosystem management
Ecotone
Electroshocking
Ephemeral
Erosion
Experimental design
First order stream
Flood plain
Fluvial

Geomorphology
GIS (Geographic Information Systems)
Glacial flour
GPS (Global Positioning Systems)
Gradient
Gully erosion, gullying
Habitat units
Hardness
Hydraulic permit
Intermittent stream
LWD (Large woody debris)
Limiting factor
Macroinvertebrates
Mass wasting
Meander
Non-point source pollution
Parameter
pH (hydrogen ion concentration)
Plunge pool
Point source pollution
Pool
Population
Precision
Reach
Replicate
Riffle
Riparian zone
Scoured
Side Channel
Stream segment
Stream typing
Surface erosion
Suspended sediments
Synergistic
Tailout
Thalweg
Time lines
Total solids
TQM (Total Quality Management)
Watershed
Wetlands

NOTE: Portions of the study guide can be selected by the instructor and used as the oral exam.

Sample
Final Exam

Definitions

1. Define and give "real world" examples of the following biometrics terms.

a. observation

b. experimental unit

c. universe

d. character of interest

e. biometrics

Fill-Ins.

1. Parameters describe _____ while statistics describe _____. An experimental unit is to the _____, as a sample is to the _____.

2. A term that indicates how many times a particular score occurs in a collection of data is known as the _____.

3. The difference between the highest and lowest values in a distribution of values is called the _____, while the square root of the sum of squares of the deviation of the values from their mean, divided by the total number of observations minus one (1), is called the _____.

4. The arithmetic average of all values in the sample distribution is the _____; the value that occurs most frequently is the _____; and the midpoint of the distribution is the _____.

Short Answers.

1. List two (2) types of statistics that describe central tendency and two (2) that describe measure of dispersion. (Briefly tell what they are)

2. What is a normal distribution curve? Why are comparisons between our frequency polygon and the normal distribution curve useful?

3. How is standard deviation useful to the biometrician?

In your own words, list five (5) of the ten (10) principles for conducting biological sampling. Cite a "real world" example of each.

Definitions. Define the following terms and list a living representative of each *(be as specific as possible*).

a. plankton

b. periphyton

c. macrophyton

d. macroinvertebrates

e. autotrophic

f. heterotrophic

Short Answers. Explain how each of the following are of significance as indicators of polluted and/or clean water.

a. plankton

b. periphyton

c. macrophyton

d. macroinvertebrate

Essay. Answer any five (5) of the following.

1. List five (5) different physical, chemical and/or biological factors that should be controlled in a bioassay and explain why each is important to the final outcome of the experiment.

2. Describe in detail two (2) types of active fish sampling methods or devices, and two (2) types of passive methods or devices. Be specific about how, when, where, and why used. Give the advantages and disadvantages of each.

3. Describe how you would design a bioassay to determine the toxic effect of cupric ion (Cu++) on coho salmon in the Chehalis River. When might a static bioassay be appropriate? A flow-through system?

4. Explain the principal of electroshocking and how you would use the backpack electroshocker to sample fish populations in a creek. Be specific about start-up and shut-down procedures, voltage and wattage settings, type of current used and why, and what water quality measurements must be made prior to use of the electroshocker.

5. Explain how you would determine the "standing plankton crop" and "biomass" (both qualitative and quantitative) in a lake. Be specific about equipment and techniques used in sample collection and analysis.

6. List at least five (5) major types of observations to be included in field notes when collecting plankton or fish in a lake. Also list at least five (5) pieces of information to be included on label to be inserted into sample container.

7. Using the ten (10) principals of experimental design, describe how you would formulate your null hypothesis (describe each step in getting there) and how you would test whether bait type made any difference in catches of spot shrimp in Puget Sound.