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Group Projects in Large and Small Classes

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Individual accountability is often viewed as an obstacle to group learning techniques. We have developed procedures for group projects that include individualized components for large (ca. 500) and small (ca. 50) non-majors introductory biology classes. Projects for the large class focus on library research while an experimental approach is required in the smaller class. In each case the students are provided with a step-by-step outline which necessitates individual contributions as well as group effort.

The projects have been received with a great deal of enthusiasm and have replaced the traditional laboratory report in our large class. Students in groups of three choose their own topics and each student gives a combination oral/poster presentation on one aspect of the topic at the end of the semester. The combined presentations must provide a cohesive overview of the topic.

The projects for the smaller class provide an occasion to “do” science and have resulted in a better understanding of, and appreciation for, science and its methodology. The student teams present a group-designed poster and each student gives part of an oral presentation at the end of the semester. In addition, each student completes a group evaluation form which includes estimates of each member’s contribution.

Teaching General Biology Using the Open Lab Format

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The Department of Biology at Kennesaw State College has been using an open-lab format in general education offerings for more than 15 years. The primary advantage is the efficient use of space and personnel in lab courses with large numbers of students. At Kennesaw State, for example, one lab with a capacity of 24 students can be used to accommodate 650 students per quarter. Another primary advantage is the flexibility afforded both students and faculty. Both faculty and commuter students appreciate the ability to vary lab hours around other work and family obligations, and the use of part-time faculty in the labs increases the options given faculty for time-release.

The primary disadvantages of an open-lab format are lack of communication and of responsibility. Faculty must use some class time to clarify lab expectations, and efforts must be made to ensure general faculty agreement on lab issues and assignments. Some students, away from the supervision of their professor, spend less time on lab exercises, and faculty must be encouraged, at times, to increase their lab emphasis.

The practical details of operating open labs vary from one school to another. At Kennesaw, the general biology laboratories are open approximately 60 hours per week, including Saturday morning. Faculty members staff the lab for 3 hours per week per section taught, and the remaining hours are the responsibility of part-time faculty.

Students entering the lab sign in on attendance records. Each receives a seat assignment and a plastic bin of appropriate non-disposable materials (glassware, forceps, marking pencils, etc.)

The lab manual contains detailed instructions and a series of questions to be answered as the student progresses through the lab. The instructions are divided up into sections, or stations, and the appropriate expendable materials (solutions, dialysis tubing, etc.) are arranged on benches around the room. Materials for each station are clearly marked. Laboratory assistants replace these materials as needed and are also available to help with check-in procedures when the lab instructor requests help.

Once the students have finished the lab, or have reached a logical stopping point, they check out of the lab, returning their bins of materials. They complete the lab report out of class and turn it in to their professor.

The primary function of the lab instructor is supervision of the lab, particularly helping students understand the process and the material. Faculty are encouraged to be pro-active, circulating among the students and discussing lab procedures and results.

The open-lab format has been successful at Kennesaw State College. You are welcome to direct questions to either of the authors.

DNA Sequence Analysis

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During the past 20 years rapid progress has been made in DNA cloning and sequencing technologies. The goal of much of this technology is to provide tools to help us understand gene structure and regulation, and also to characterize the protein sequence encoded by genes. It is certain that more knowledge in these areas will provide insight into basic cellular processes and ultimately assist in the diagnosis and treatment of disease. Consequently, it is important to introduce these ideas early to undergraduates with interests in medicine as well as basic and biomedical research. At Emory we have developed an advanced laboratory course in Genetics to address this issue. One section of this course was designed to introduce students to computer-assisted analyses of DNA.

The careful analysis of large amounts of DNA sequence is now routinely performed with the assistance of computers. Many analyses cannot be performed in a reasonable time frame without such programs. Even a slow computer will perform analyses in a fraction of a second.

GENEPRO (Riverside Scientific, 206/842-9498) is a simple-to-use, yet sophisticated program that runs on a microcomputer. In our course students work with a *Drosophila* sequence. The sequence derives from a gene named “mastermind”; this gene is required for normal *Drosophila* nervous system development. In the lab each student is given an “unknown” sequence. Initially students learn to work with the DNA sequence and find restriction enzyme sites as well as areas of the sequence likely to encode proteins. The deduced protein sequence is then examined for particular structural features that may provide initial clues to function (i.e., hydrophobic regions that may span a membrane). Finally, students use the deduced amino acid sequence to search sequence databases for related proteins. All students score hits in the database and then use associated literature references to write a short paper. The following is an outline of the types of analyses that can be performed on this sequence:

DNA analysis: (1) restriction sites, (2) composition, (3) inverse, (4) translate, (5) open reading frame search, (6) codon usage, (7) highlight a sequence.

Protein analysis: (1) structural tests, (2) composition, (3) reverse translate, (4) highlight a sequence.

Homology analysis: (1) dot matrix, (2) homology search.

Teaching DNA Structure and Function on a Shoestring Budget

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The experimental evidence, that enabled scientists to identify deoxyribonucleic acid (DNA) as the genetic material (Avery et al., 1944; Hershey and Chase, 1952), and enabled Watson and Crick (1953) to create their model for the structure of DNA, can be demonstrated in the classroom in a variety of ways. Much of the research into the nature and function of the genetic material is possible naturally through mutations, defined as heritable changes in the sequence of nucleotides in the DNA. Now, however, changes in the DNA structure can be generated in the classroom laboratory using a variety of molecular techniques (Cohen, 1975). A laboratory-generated technique called splicing makes possible the ability to cut and to ligate together DNAs from a variety of different types of organisms. Through the process of splicing, a change has been created in the DNA that will result in a new material that has the ability to create new life forms capable of destroying human-produced materials and to alter life so that not only is the present affected, but the future as well (Miller, 1992; Murray, 1991).

Many high school and undergraduate biology laboratory teachers find themselves operating on a shoestring budget and are always searching for ways to provide some form of a hands-on experience to students when teaching DNA structure and function. Examples such as gene mutation, mutation frequency, DNA replication (DNA synthesis) and transcription (RNA synthesis), messenger RNA translation (protein synthesis), and genetic engineering are classical and modern-day concepts in which DNA structure and function can be taught. The two exercises described below demonstrate how inexpensive, cost-saving items, and sometimes basic household products, may be used to teach certain aspects and concepts in DNA structure and function.

Exercise 1: DNA Structure, Function, and Regulation

Purpose: After completion of Exercise 1, students should be able to: (1) describe or diagram the basic chemical structure of a single-stranded and double-stranded nucleic acid strand and distinguish chemically between DNA and varieties of RNA; (2) give the basic sequences of one strand of DNA and predict that of a complementary strand of DNA; (3) point out degeneracy, redundancy, and the flexibility of the genetic code using a standard genetic code table that is provided; (4) calculate mutation frequencies following one, two, or three rounds of cell division (DNA replication); and (5) draw a diagram illustrating an operon, a group of contiguous, coordinately controlled genes, and describe how it functions.

Introduction: The experimental evidence, that enabled scientists to identify DNA as the genetic material, and enabled Watson and Crick to create their model for the structure of DNA, are discussed in this exercise. The processes by which information encoded in the DNA is decoded and expressed in the cell are also discussed. Much of the research into the nature and function of the genetic material is possible through mutations, defined as heritable changes in the sequence of

nucleotides in the DNA. Students will be introduced to the mechanisms of gene regulation in both eukaryotic and prokaryotic cells.

Diagrams: models of DNA; models of nucleotides showing double and triple bonds; illustrations representing repression, induction, and constitutive synthesis of the lactose operon; and genetic code table. *Equipment:* small portable typewriters or personal computers with keyboards for typing letters, words, and sentences that are used to represent codes, codons, nucleotides, amino acid sequences, and mutation frequencies. *Supplies:* plastic snap beads of varying lengths and colors to illustrate nucleotide, amino acid sequences, and mutation sites.

Directions: Students will complete the DNA replication and transcription, and RNA translation of a given nucleotide sequence using the genetic code table, nucleotide sequence, and DNA models provided. Students are given 1-minute, 2-minute, and 3-minute time periods to type from the nucleotide sequence of a single-stranded DNA: (1) the daughter DNA molecule (double-stranded) after one replication cycle; (2) the RNA transcript; and (3) the amino acid sequence of the polypeptide (protein) chain formed after translation of messenger RNA transcript. In the 1-, 2-, or 3-minute specific time periods, point mutations (base substitution, addition, or deletion of a letter or space on the message) can be illustrated using a simple sentence to be typed in specific 1-, 2-, or 3-minute time periods, such as “the guest are now here”. That represents the normal or single-stranded parent DNA molecule. The following examples show the single-stranded daughter DNA molecule after a base substitution or a base addition mutation.

Base substitution: “the guest are not here”

Base addition: “the guest are nowhere”

A period (.) or an “x” can be used to create nonsense or missense mutations.

Given the single-stranded DNA strand below and the following gene expression hierarchy: (1) show the replication (double-stranded daughter DNA molecules); (2) show the mRNA strand; (3) show the transfer RNAs, tRNAs (anticodons), required for amino acid peptide bond elongation; and (4) show amino acid sequence of the polypeptide formed in the messenger RNA translation product. Upon completing the exercise from each level of gene expression: (1) determine the nature of the mutations; (2) the mutation rates in base pairs per minute or amino acid substitutions per minute; and (3) the mutation frequencies in base or base pair changes and amino acid substitutions in units per molecule (e.g., one amino acid substitution per polypeptide).

Single-stranded parent DNA strand: TACCGTTTGAGCGGGCCCAAAGTGAATGGCATTAAA

Exercise 2: Gene Manipulation Through Recombinant DNA Techniques

Purpose: After completing Exercise 2, students will be able to: (1) describe the primary techniques utilized in recombinant DNA experiments; (2) summarize the problems involved in cloning a single gene; (3) describe the action of restriction endonucleases and their specific function in recombinant DNA experiments; and (4) identify the role of plasmids, circular DNAs that replicate independently of the cell’s chromosomes, in recombinant DNA experiments.

Introduction: A laboratory-generated technique called splicing makes possible the ability to cut and to ligate (join) together DNAs from a variety of different types of organisms. Through the process of splicing, a change has been created in the DNA that will result in a new material that has the ability to create new life forms capable of destroying human-produced materials and to alter life so that not only is the present affected, but the future as well. In addition to splicing, DNAs can be chemically synthesized and transferred into different types of cells in which the genes may even

function under appropriate conditions. DNA is currently being used to diagnose the potential for the development of certain diseases and for the medical treatment of certain diseases (Miller, 1992). Much of the above is possible due to recombinant DNA technology.

A vital key in the development of recombinant DNA technology was the discovery of restriction endonucleases and their importance as chemical scalpels (Cohen, 1975). The importance of these enzymes as tools in recombinant DNA technology lies in their specificity for a particular substrate. Each enzyme attacks a specific sequence of nucleotide bases in the DNA double helix. Some DNA restriction endonucleases attack symmetrical (palindromic) sequences composed of four to seven nucleotides while other enzymes attack asymmetrical sequences of four to five nucleotides long. The enzyme cuts the DNA into pieces called “resection fragments”. In this exercise students are introduced to the rationale for the procedures and molecules used in DNA technology.

Diagrams: restriction endonuclease maps of various plasmid DNA cloning vectors (pBR322) and restriction endonuclease charts showing restriction recognition sites. *Supplies:* plastic snap beads of varying length and colors to illustrate vector (plasmid) DNA, insert (cloned) DNA, antibiotic-resistant DNA, restriction enzyme sites, and recombinant DNA molecules.

Directions: Below are two nucleotide sequences of double-stranded DNA and a list of restriction endonucleases and their recognition sequence and site. Determine the restriction endonuclease that will cut *both* pieces of DNA and indicate the sites of attack and the number of fragments produced.

DNA sequence:

1. CCAGTCGTTAACGAATTCGTCGACGTCGAC
GGTCAGCAATTGCTTAAGCAGCTGCAGCTG
2. ACGGGTTAACCCAATGGATCCCAAGTTAACGGTACC
TGCCAATTGGGTTACCTAGGGTTCAATTGCCATGG

Restriction endonucleases*	Recognition site**
Eco RI	G/AATTC
Bam HI	G/GATCC
Hpa I	GTT/AAC
Sal I	G/TCGAC

* Each enzyme recognizes a symmetrical or palindromic sequence

** The “/” indicates the cut site

Make a chain of: 10 black snap beads, 6 red snap beads, 4 green snap beads, 3 blue snap beads, and 2 yellow snap beads. Form a circle (plasmid) by connecting these five snap bead chains together. The green snap bead chain = insert DNA; the red snap bead chain = antibiotic-resistant DNA; the blue snap bead chain = origin of DNA replication (ori); the yellow snap bead chain = restriction sites; and the black snap bead chain = vector DNA. Now, describe the structure and function of your recombinant DNA molecule.

Finally, construct hypothetical, but logical, models of recombinant DNA molecules using different numbers and combinations of colored snap bead chains. After Exercise 2 has been completed, answer the following questions: (1) Are your DNA molecules recombinant? (2) Are your recombinant DNA molecules logically constructed? (3) Will your recombinant DNA molecules replicate? (4) Do your recombinant DNA molecules demonstrate unique cloning

strategies? (5) Can your recombinant DNA molecule be easily screened and selected if present in a recombinant DNA library (gene bank)?

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Using Plant Tissue Culture to Investigate Plant Cell Differentiation and Dedifferentiation

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One of the challenges in teaching developmental biology is designing laboratory exercises that introduce important concepts and techniques, provide some latitude for independent study, and which are not prohibitively expensive. Described here is an experimental project that uses plant tissue culture techniques to examine cell differentiation in carrot. The specific questions asked in this experiment are: (1) Does the ability of carrot tissue to form callus depend upon the type of tissue examined?, and 2) Does the ability of carrot tissue to form callus depend upon the age of the tissue?

The techniques used in this project are relatively straightforward: students prepare sterile seedlings, prepare callus cultures, monitor, measure, and record callus growth, and analyze data. Seeds are sterilized by soaking them for 15 seconds in 15% silver nitrate. The seed suspension is then quickly poured through a funnel lined with a cone of filter paper. After seeds had dried for 2–4 hours, they are sprinkled onto a petri dish containing suitable medium (see below). Seedling cultures are incubated in the light. After germination, the sterile seedlings are used to start callus cultures. It is essential that sterile technique be maintained throughout these procedures. For our tissue type experiment, students separate root, shoot, and leaf tissue. The tissues are cut into pieces approximately 5 mm in length. Such tissues are placed on media suitable for callus formation (see below). Incubation was in the dark at 28°C. Cultures are examined once or twice per week. The percentage of tissues forming callus is recorded as are the lengths, widths, and general appearance of each tissue piece.

This project is very successful both in terms of experimental results and student enthusiasm. In carrot, both the age and the types of tissue determined how well callus was able to form. Students were pleased about learning tissue culture techniques and their group work and record keeping skills were improved. Class discussion of the experimental results generated several insights about similarities and differences between plant and animal development and as well as ideas for further extensions to this experiment. The project can be adapted for use in introductory courses or in upper-division offerings including cell biology, developmental biology, botany, or plant physiology.

Standard culture media for growing plants from seed: MS salts (Murashige-Skoog salt base, can be purchased from Carolina Biological Co. and other suppliers), 100 mg of myo-inositol, 30 g of sucrose, and 1 liter of distilled or deionized water. Adjust pH to 5.8, add 9 g of agar, and autoclave for 20 minutes with a slow exhaust. (Pre-made media can also be purchased from Carolina Biological Co. and other suppliers.)

Carrot callus induction medium: Same recipe and instructions as above except add 0.5 mg/liter of 2,4-dichlorophenoxyacetic acid (2,4-D). (Pre-made carrot callus induction medium can be purchased from Carolina Biological Co. or other suppliers.)

Bioluminescence in Transformed Bacterial Cells

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E. coli cells can be transformed with a plasmid incorporating a gene for bioluminescence. This causes the cells to glow in the dark. Students who perform this transformation exercise are invariably fascinated with the phenomenon, and even more intrigued when they learn that the *lux* genes were isolated originally from a bacterium found in a fish. The *lux* gene plasmid was produced by my colleague, Dr. Joseph J. Shaw, while working at the University of California at Davis.

Students work in teams of six to transform competent *E. coli* cells with two different plasmids to obtain cells of two phenotypes which are then compared to control (untransformed) cells. Each team is given three tubes of competent *E. coli* cells. One tube is labeled “control”, and receives no plasmids. The second tube is labeled “plasmid #1”; #1 plasmids, which confer resistance to the antibiotic ampicillin, are added to this tube. The third tube is labeled “plasmid #2”; #2 plasmids, which carry genes for both ampicillin resistance and bioluminescence, are added to this tube. All tubes are then taken through the transformation procedure.

Students then transfer half of the cells from each tube onto plates of plain agar and the other half onto plates of agar containing ampicillin. The cells are allowed to grow overnight at 37°C. These plates are observed first in a dark room to determine which of them exhibit bioluminescence and then visually to determine which plates have colonies that are ampicillin resistant.

This simple yet stimulating experiment provides beginning students with an easily understood introduction to molecular genetics. Further information, a full description of the exercise, and/or stabs of the needed cells are available upon request from the author.

Making and Staining Fresh Plant Hand-Sections

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Introductory biology students often come to the course with very little knowledge of or interest in plants, and examination of prepared slides often generates little enthusiasm. To increase the students' interest in and enjoyment of plants, a standard staining procedure was modified to produce usable stained sections more rapidly. Of course, sections which have been cut with a microtome, fixed, and stained will be better than those the majority of students can produce with this method. However, the excitement the students exhibit when they produce their own slides in a short period of time makes up for the lack of clear detail in some of the slides, and quite a few of the slides are spectacular. This sectioning method does not work well with thin, soft material such as young roots. The tips of young carrots produce reasonably good sections, although they look significantly different from the traditional *Ranunculus* root sections. Cross-sections of stems of marigold, *Vinca rosea*, carnation, and brassica varieties are easy to make and look good, as do sections of many woody stems. It is best to look at leaf sections unstained at first and then stain some to see the arrangement of vascular tissue. The leaves of *Vinca rosea*, brassicas, carnations, and snapdragons have given good results.

Student Instructions: You will stain the material with 0.2% toluidine blue which stains tissues differently depending upon their chemical composition. Lignified cell walls, such as those of fibers, tracheids, and vessels, stain blue or bluish-green. Cell walls that are not lignified stain shades of purple.

1. Place the material on a clean slide and hold it down with your index finger so that only a very small portion is exposed. Press the flat edge of a new razor blade against your fingertip, angled slightly away from your finger. Make a series of 10 or more sections with a rapid, short, slicing motion, pushing the blade back very slightly each time without moving your finger. Wash the slices off the razor blade onto a clean glass slide with a little water and a dissecting needle.
2. Spread out the sections and discard any that are obviously too thick. Keep partial sections since they often have at least one edge which is quite thin.
3. Carefully draw off the water by placing a kimwipe at the edge and letting it soak up the liquid.
4. Add 1 or 2 drops of 0.2% toluidine blue stain to the sections on the slide and let it stain for 5 to 15 seconds.
5. Quickly draw off the stain just as you did the water.
6. At once, add a few drops of water to wash off any remaining stain and draw off this water.
7. Add more water. Blot it off again if it looks blue.
8. Add water and carefully spread out the sections.
9. Put on a cover slip and examine the slide. If the sections are stained too darkly, wipe off the slide and try again. If the sections are not stained darkly enough, either make a new slide and leave the stain on for longer, or draw off the water and stain the section a little more.
10. Be sure to adjust the light and the fine focus so that you see all there is to see.

An Inexpensive Pressure Transducer for Measuring Aerobic and Anaerobic Respiration

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Metabolic rate is commonly measured by quantifying the amount of gas (either carbon dioxide or oxygen) that is released or consumed over a period of time. These experiments are traditionally performed with a Warburg-type manometer which tends to be fragile, awkward to set up for the inexperienced student, and provides relatively imprecise measurements. A sensitive but sturdy electronic device which can precisely measure the change in pressure produced by a reaction system has been developed at Mt. Holyoke

College. This pressure transducer outputs a voltage that is proportional to the change in pressure between two inlets. Therefore, students can monitor pressure change over time using either a strip chart recorder or a simple voltmeter (the PT-60 pressure transducer has two output ranges: 5 mV/mm Hg and 50 mV/mm Hg). The cost of the transducer is comparable to the price of a standard glass manometer with a side-arm reaction vessel.

We have developed experiments in aerobic respiration (using plants and animals) and anaerobic respiration (yeast fermentation) that use the PT-60. Currently, we are devising experiments that use the pressure transducer to measure oxygen evolution in photosynthesis.

Recording Action Potentials from Earthworm Giant Axons

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There are two major ways of increasing action potential conduction velocity: increasing the diameter of the axon and adding a myelin sheath to the axon. Since invertebrates do not have myelin they use large diameter fibers to convey information rapidly.

In this exercise action potentials are recorded extracellularly from the giant nerve fibers of the earthworm, *Lumbricus* sp. The earthworm nerve cord has three giant fibers: one medial giant and two lateral giants. These fibers monitor sensory input and rapidly send their output to motor neurons which cause longitudinal muscles to contract.

This exercise requires no surgery and can be used to examine a variety of action potential related concepts. It is an excellent introduction to instrumentation as well.

Earthworms are anesthetized by immersion in 15% ethanol for approximately 15 minutes and placed ventral side down in a dissecting pan. Five insect pin electrodes are simply placed into the earthworm along the midline as shown in Figure 1. Two of the electrodes are connected to a stimulator. The other three are connected to the input of an AC amplifier. The outputs of the amplifier and stimulator are connected to a recording device (e.g., an oscilloscope or a data acquisition system and computer). Figure 2 shows records taken from a computer screen.

Conduction velocity (V) can be determined by comparing the latency of the responses (T) with the distance (D) from the stimulating electrode (S -) to the first recording electrode. Two different distances are used so that conduction velocity ($V = (D_2 - D_1)/(T_1 - T_2)$). Refractory periods can be determined by putting the stimulator in the twin pulse mode and changing the delay to provide different times between the two pulses.

This exercise was modified from one described by B. Oakley and R. Schafer (*Experimental Neurobiology: A Laboratory Manual*, University of Michigan Press, Ann Arbor, Michigan, 366 pages, 1984).

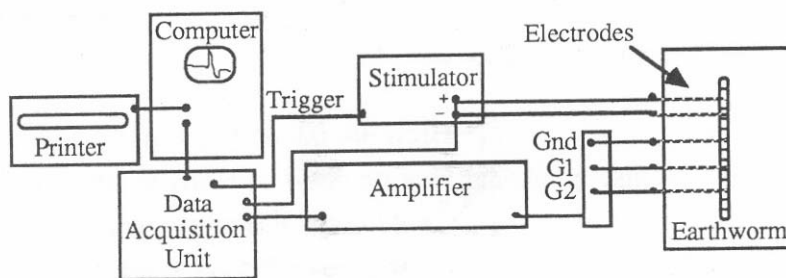


Figure 1. Schematic diagram of the set-up used to record action potentials extracellularly from earthworm giant fibers.

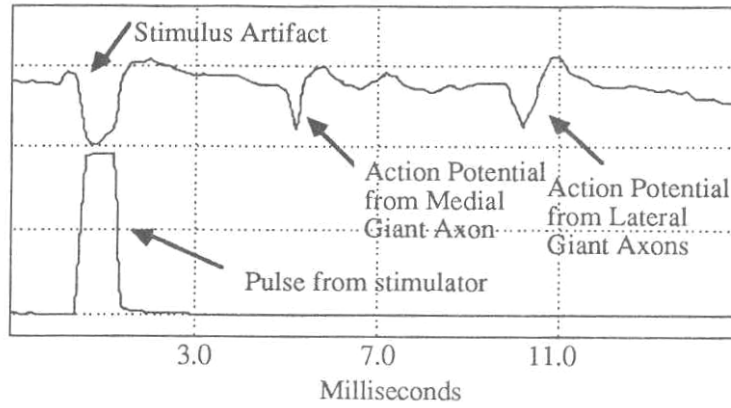


Figure 2. Computer printout showing oscilloscope traces of action potentials recorded extracellularly from earthworm medial and lateral giant axons. A stimulus pulse from the stimulator is also shown.

Exploring a Fresh Mammalian Heart in a Learning-Cycle Laboratory

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When seeking materials for laboratories, use the local butcher shop as one supply source. In my classes I use fresh hearts, bones, and muscles tissue from a farming-community or urban kosher shops. On ordering a fresh beef heart, I ask butchers to leave long vessels. Before using it, I remove some of the surrounding fat and close small vessels and extraneous knife cuts with a sewing needle and thread.

Students use the fresh heart as the first exploratory activity in a laboratory on the structure and function of the heart (Wilke, 1993a, 1993b). On examining the heart with sight and touch and by running water into its vessels, students develop structure-function concepts such as the heart has two separate flow pathways and each pathway flows unidirectionally. In additional activities, students explore detailed heart structure in preserved whole and preserved frontally-sectioned hearts. They determine the structural details that explain the observed flow patterns in the fresh heart.

To check and reinforce their understanding, students apply their concepts in two ways: (1) Students compare the structure of the postnatal heart to the fetal heart and infer prenatal blood-flow patterns (or they can compare it to the three-chambered frog heart). (2) They predict the effects of described human structural abnormalities on blood flow and the individual.

The learning-cycle teaching strategy consists of three components: exploration, concept formation, and application. In these laboratories, students use hands-on activities to develop and apply concepts. Student-designed experiments may be in either the exploration or application component. In this teaching strategy, the teacher does *not* deliver facts but is a facilitator of students' concept development through questions that encourage their thinking. If you are interested in additional information on the learning cycle, I recommend a monograph of the National Association for Research in Science Teaching (NARST) by Lawson et al. (1989). Mosby-Year Book Inc. produced a 27-minute video tape about the learning-cycle teaching strategy, which shows students engaged in this heart laboratory.

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———. 1993b. Teacher's guide to accompany exploring biology today. Mosby-Year Book, St. Louis, 217 pages.

Nematodes of the Cockroach

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Leidynema appendiculata and *Hammerschmidtella diesingi* are common parasites of the American cockroach, *Periplaneta americana*. They are easily teased from the hindgut, are large enough (2 mm), to see with the unaided eye, and are excellent specimens for examining internal anatomy. Demonstrating their presence to students adds an interesting dimension to the study of cockroaches in a zoology laboratory, provides a context in which to discuss symbiosis, and introduces the subject of human parasitism since these nematodes are oxyuroid relatives of the human pinworm, *Enterobius vermicularis*.

After anesthetizing the cockroach by cooling, remove the head, legs, and wings. Open the abdomen and expose the digestive tract in a dissecting dish containing insect saline. Remove the section of the tract between the gizzard (which is easily recognized) and the anus, and place it in a watch glass of saline. Tease open the hindgut and observe with a stereoscopic microscope. Nematodes will be readily visible as they migrate out of the gut. By placing the nematodes on a microscope slide, adding a coverslip, and examining with a compound microscope, internal anatomical features are readily seen.

A list of several older references which provide diagrams of these nematodes along with three recent articles are:

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Plant Competition from the Field and Laboratory Perspective

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This series of introductory labs provide our first-year students the opportunity to explore the relative strengths and limitations of field versus laboratory work. Experimental design, hypothesis testing, data collection, and interpretation are emphasized. The first part of this project takes place in the field. Students conduct a survey of trees in nearby woods. Each team of four students identifies and measures all trees (greater than 5 cm in diameter) in a different 400 m² site. Groups from all lab sections pool their results and then plot the average tree diameter versus the number of trees per 400 m².

Students observe that trees in less densely populated study sites tend to be larger than trees in denser study sites. They are asked what they can infer from these data and to consider whether they have proven that competition is occurring among the trees. We also discuss whether other factors might influence how trees respond to crowding.

Based upon their field results, groups of three to four students each design and implement an experiment to examine how plants respond to crowding in laboratory conditions. They also test whether certain factors might influence how plants respond to crowding. Nutrient or water availability, soil pH, and varying temperature, are a few of the parameters students have examined. Each group examines a different factor.

Four to five weeks after planting the seeds, students harvest the plants and weigh them. Students assess how their plants responded to crowding. They evaluate whether the other factor they studied had an effect on the plants response to crowding. They also compare their laboratory results to the field survey and each group presents an oral report of their findings.

Influence of Nutrients and Zooplankton Grazing on Phytoplankton: A Collaborative Lab

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The purpose of the collaborative lab is two-fold. First it allows the gathering of more extensive data without using an unmanageable amount of time from a laboratory experience. Secondly, it allows introductory students to understand some of the concepts and experiences more advanced courses offer.

This lab experience looks at the producers (phytoplankton) and the primary consumers (zooplankton) of the open water of a lake. Phytoplankton are defined as floating or weakly swimming photosynthetic autotrophs such as diatoms, green algae, and cyanobacteria. Zooplankton are defined as floating or weakly swimming heterotrophs, mostly microscopic, such as protozoans, rotifers, and microcrustaceans (e.g., copepods and cladocerans).

Phytoplankton require light, water, and nutrients (nitrogen, phosphorus, potassium, etc.), among other things. If any of these requirements is in short supply (limited), the growth rate of the phytoplankton will be held at a level lower than possible given unlimited resources. Obviously in a lake, water is not a limiting resource. Light, near the surface where the phytoplankton and zooplankton are found, is also sufficient not to be limiting. Nutrients, however, can often be limiting. Growth rates of phytoplankton may also be limited via predation by zooplankton. Conversely zooplankton growth may be limited by the amount of phytoplankton present.

Four replicate stations were established in a local lake. Each station contained four submerged 5-gallon (19 liter) plastic carboys:

Carboy 1: "CONTROL" (lake water)

Carboy 2: "+ PO₄" (lake water plus 400 µg/liter KH₂PO₄)

Carboy 3: "- Zooplankton" (lake water filtered to remove zooplankton)

Carboy 4: "+ PO₄ / - Zooplankton" (filtered lake water plus 400 µg/liter KH₂PO₄)

One week and 4 weeks after the stations were established samples were taken from each carboy for phosphorus analysis, zooplankton identification and enumeration, and phytoplankton identification and enumeration.

Phytoplankton, particularly diatoms, definitely increased with the addition of phosphorus and the elimination of zooplankton. There is some indication that zooplankton increased as the phytoplankton increased.

The students in the *courses* contributed as follows: (1) Introductory Biology: collected samples, identified and enumerated zooplankton, and ran phosphate analysis. (2) Ecology: set up the experiment, identified and enumerated zooplankton, and ran phosphate analysis. (3) Phycology: collected samples, and identified and enumerated phytoplankton.

Using Displays to Augment Biology Laboratories

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Displays are a part of many biology departments and can provide much useful information if not some color to a room or hallway. Not only are they important in providing learning material outside the classroom, but they can be interesting and amusing. Displays can elaborate on topics discussed in lectures and labs. In our department we have glass tables located in the hallway where displays are available for viewing. Every semester a teaching assistant is assigned to be the curator of exhibits in order to fulfill a semester job. Each week the display is changed and corresponds to the laboratory exercise of that week. We have found that these displays are useful to students studying for tests, and faculty and the public find them interesting and amusing.

The discussion in the ABLE mini workshop included how to create eye-catching and artistic displays and consisted of some pointers and helpful hints that may turn a display case into an interesting exhibit. These suggestions included the headline or main image; amount of information in the display; setting up the display to guide a viewer through it; emphasizing the most important points with images, color, different typefaces; having someone else look at the display and critique it; and bringing the display all together. This last point is important since the display is a designed work, not just a collection of information. The creative displays and their relevance to laboratory exercises in our department have been successful in gaining much attention and are worth having in any biology department.

Update on AP Biology Labs

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The AP Biology course is designed to be the equivalent of a college introductory biology course taken by biology majors during their first year. It is intended that the AP Biology course be taken by high school students after the successful completion of a first course in high school biology and one in chemistry as well. Since one-fourth to one-third of the credit in most college introductory courses is derived from laboratory work, the same emphasis is required in an AP course, therefore laboratory experience must be included in all AP Biology courses. The AP Biology Development Committee has developed a set of 12 laboratories to provide a standard as an aid to teachers for integration of laboratories into the AP Biology curriculum (the titles of these laboratories are given below). These laboratories have been introduced over the years from 1988 through 1994.

The AP Biology Development Committee is now in the process of revising the laboratories based on two surveys recently undertaken: (1) a survey of colleges and universities that provides input on the number and type of laboratory investigations done in introductory college courses, and (2) a survey of AP Biology teachers that provides input on the workability of the 12 current AP Biology labs in the high school setting.

1. Diffusion and Osmosis
2. Enzyme Catalysis
3. Mitosis and Meiosis
4. Plant Pigments and Photosynthesis
5. Cell Respiration
6. Molecular Biology
7. Genetics of *Drosophila*
8. Population Genetics and Evolution
9. Transpiration
10. Physiology of the Circulatory System
11. Behavior: Habitat Selection
12. Dissolved Oxygen and Aquatic Primary Productivity