Agricultural Science with Vernier

Agricultural Science Experiments using Vernier Sensors

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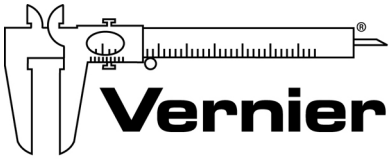

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Proper safety precautions must be taken to protect teachers and students during experiments described herein. Neither the authors nor the publisher assume responsibility or liability for the use of material described in this publication. It cannot be assumed that all safety warnings and precautions are included. Teachers must follow local regulations concerning safe handling, use, and disposal of the materials and chemicals associated with the experiments included in this manual.

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Cover Photo: Agricultural Science teachers Rosalea Riley and Rebecca Mahan of Virginia use a LabQuest and Light Sensor and Stainless Steel Temperature Probe to measure the light intensity and temperature in a greenhouse.

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Preface

This book contains a collection of experiments that can be helpful in teaching agricultural science. The experiments can be done using a variety of Vernier interfaces including the LabQuest, LabQuest Mini, LabPro, EasyLink, or the Texas Instruments CBL 2, for collecting, displaying, printing, graphing, and analyzing data. Using high-quality Vernier sensors will result in more convenient and more accurate results than many traditional methods. Your students will perform many new experiments, some with measurements not previously obtainable in the classroom or in the field.

Using Vernier data-collection technology opens up a large number of new agricultural science investigations—the experiments we have included here are only a small fraction of the many possibilities. Many are done in the classroom, but the equipment can easily be taken into the field, as well. The Vernier LabQuest is especially well-suited to a combination of field work and classroom work.

You will find a wide range of experiments in this book. Whether your agricultural science class is high school or college, plant or animal, you should find a large number of experiments in this book that match the scope and objectives of your course. Following each student experiment there is an extensive Teacher Information section with sample results, answers to questions, directions for preparing solutions, and other helpful hints regarding the planning and implementation of a particular experiment.

Experiments in this book can be used unchanged or they can be modified using the word-processing files provided on the accompanying CD. Students will respond differently to the design of the experiments, depending on teaching styles of their teachers, math background, previous experience using probeware, and the scope and level of the agricultural science course. Here are some ways to use the experiments in this book.

- Unchanged. You can photocopy the student sheets or print them from the accompanying CD, distribute them, and have the experiments done following the procedures as they are. Many students will be more comfortable if most of the steps used in data collection and analysis are included in each experiment.
- Slightly modified. The CD accompanying the book contains the Word files for all versions of the experiments in this book. Before producing student copies, you can change the directions to make them better fit your teaching circumstances. See *Appendix A* for instructions on using the CD.
- Extensively modified. This, too, can be accomplished using the accompanying CD. Some teachers will want to decrease the degree of detail in student instructions or add more rigor to the analysis sections for more advanced students.

We feel it is **very important** to have your students perform Experiment 1, "Introduction to Data Collection." This experiment has introductory details that are not included in other experiments in the book. The experiment can be completed quickly, leaving students plenty of time to explore other important capabilities of the data-collection software.

We also feel it is important for teachers to read the information presented in the appendices. They include valuable information that can help make you more comfortable with your initial use of this equipment. Here is a short summary of the information available in each appendix:

- *Appendix A* tells you how to use the word-processing files found on the CD.
- *Appendix B* tells you how to use Logger *Pro* 3 software to display or print graphs and data tables after importing data from LabQuest or a graphing calculator.

- *Appendix C* provides information on Vernier Software & Technology products for agricultural science.
- *Appendix D* provides a list of the equipment and supplies used in these experiments.
- *Appendix E* provides information on collecting GPS data and using GIS software to map data.
- *Appendix F* tells you how to use TI Connect to load the EasyData App onto your calculator and capture calculator screen images.
- *Appendix G* provides details about safety information.

We hope you find this book a helpful start in integrating data-collection technology into your agricultural science course.

Introduction to Data Collection

Data collection is a very important part of science. Meteorologists collect weather data over time to keep an historical record and to help make forecasts. Oceanographers collect data on the salinity (saltiness) of seawater to study changing trends in our Earth's oceans. While data have been collected by hand for thousands of years, the technology to collect data electronically has been around for fewer than 80 years. Only in the last 20 years has this technology been available to schools.

This experiment was designed to introduce you to two of the most common modes of data collection that will be used in this class. Part I will guide you through collecting and analyzing data over time. A Temperature Probe will be used to record the temperature of water for 60 seconds at a rate of one sample per second. In Part II, you will collect data using a mode called Events with Entry. This style of data collection allows you to collect one point of data, then will ask you to type in a corresponding value. In this experiment, the data collected will be the temperature of your hand and the value you type in will be your assigned group number.

OBJECTIVES

In this experiment, you will

- Become familiar with the Logger*Pro* computer program.
- Use a computer and a Temperature Probe to make measurements.
- Analyze a graph of the data.
- Use this graph to make conclusions about the experiment.
- Determine the response time of a Temperature Probe.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier Temperature Probe
two 250 mL beakers
cold tap water
hot tap water
ice

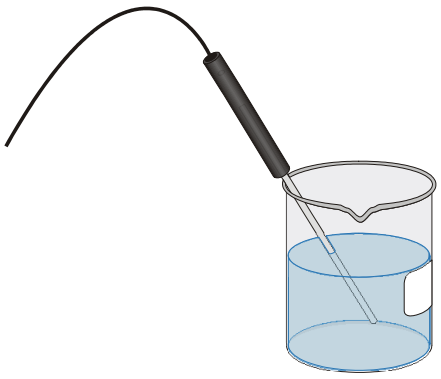


Figure 1

PROCEDURE

Part I Time Graph

1. Place about 100 mL of tap water into a 250 mL beaker. Add two or three ice cubes.
2. Connect the Temperature Probe to your Vernier computer interface.
3. Place the Temperature Probe into the cold water and stir briefly. Then position the probe in the cold-water beaker as shown in Figure 1. Note: Make sure the beaker will not tip over from the weight of the Temperature Probe.
4. Prepare the computer for data collection.
   a. Choose Open from the File menu.
   b. Open the *Agricultural Science with Vernier* folder.
   c. Open the file "01a Intro to Data".
5. Place about 150 mL of hot water into a second 250 mL beaker.
6. Click Collect to begin data collection. Do not stir or move the water.
7. When exactly 10 seconds have gone by, quickly move the Temperature Probe to the beaker containing hot water and allow the computer to continue data collection. Do not stir the water or move the Temperature Probe during the remainder of the data collection period.
8. Data collection will stop automatically after 60 seconds.
9. Remove the Temperature Probe from the beaker and dry it with a paper towel.
10. Determine the time at which the highest temperature was reached. There are several ways to accomplish this. Try them all.
    a. Move the cursor to the point on the graph where it appears the highest temperature was reached.
    b. As you move the cursor across the graph, notice that there is a live readout of the x- and y-coordinates at the bottom of the screen. This readout of the cursor location is a fast and easy way to interpret a graph.
    c. Click the Examine button, , on the toolbar. The cursor now includes a vertical line. As you move the cursor across the graph, it will jump from one data point to the next. The temperature and time values corresponding to its position will be displayed in the Examine Box.
    d. Use the left and right arrow keys to scroll across the highest portion of the curve. Which do you prefer, the mouse or the left and right arrows?
    e. Find the highest temperature. Record this temperature and the time when it was first reached in your data table.
    f. Close the Examine Box by clicking the upper-left corner of the box.
    g. To confirm the time when the highest temperature was first reached, use the scroll bars in the table to scroll through the table and examine the data.

11. Practice changing the graph scaling. In future experiments, you may want to change the scale of either axis of a graph. There are several ways to do this.
    a. To scale the temperature axis from 0 to 80°C instead of the present scaling, click the mouse on the "100" tickmark at the top of the axis. In the edit box that appears, type in "80" and press the ENTER key. Notice that the entire axis readjusts to the change you made. To scale the time axis from 0 to 150 seconds instead of the present scaling, click the mouse on the "60" tickmark at the right end of the axis. In the edit box that appears, type in "100" and press the ENTER key.
    b. Another way to change scaling is to click the Autoscale button, [A], on the toolbar. The computer will automatically rescale the axes for you. Try it.
12. Print copies of the graph as directed by your teacher.

Part II Events with Entry

13. Prepare the computer for data collection.
    a. Choose Open from the File menu.
    b. Open the *Earth Science with Vernier* folder.
    c. Open the file "01b Intro to Data" file.
14. Number the members of your group by age with the oldest being number one. Record the names in your data table. Add more lines if needed.
15. Click [Collect] to begin data collection.

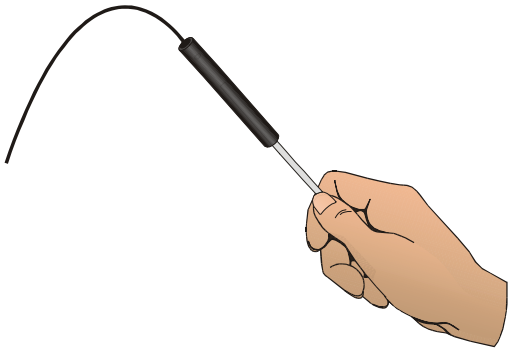


Figure 2

16. Measure the hand temperatures of your group members.
    a. Group member number one should pick up the Temperature Probe and hold its tip in the palm of his/her hand as shown in Figure 2.
    b. Watch the live temperature readout in the Meter window. When the temperature has stopped rising, click [Keep].
    c. You will be prompted to type in a number. Type in "1" for your group member number and press the ENTER key. The temperature and group member number have been saved in the data table.
17. Cool the Temperature Probe back down by placing it in the cold water from Part I. Monitor the temperature in the meter and remove it from the water when the temperature reaches 25°C.
18. Pass the Temperature Probe to the next group member.
19. Repeat Steps 16–18 until every group member has their hand temperature recorded.
20. Click [Stop] to end data collection.
21. Determine each person's hand temperature by using one of the methods described in Step 10. Record them in the data table.
22. Print copies of the graph as directed by your teacher.

DATA

Part I Time Graph

Maximum temperature (°C)	Elapsed time (s)

Part II Events with Entry

Group member number	Group member name	Maximum temperature (°C)
1		
2		
3		
4		
5		
6		
Group average		

PROCESSING THE DATA

Part I Time Graph

1. Describe the appearance of your graph in Part I.
2. Why is time plotted on the horizontal axis in this experiment?
3. Why is temperature plotted on the vertical axis?
4. Determine the Temperature Probe's response time. To do this, use your data to find how long it took for the Temperature Probe to reach the maximum temperature after moving it from the cold water to the hot water.
5. Explain how you determined your answer to Question 4.

Part II Events with Entry

6. Calculate your group's average for the maximum temperatures. Record the result in the data table.
7. Who had the hottest hand?
8. Who had the coldest hand?

TEACHER INFORMATION

Introduction to Data Collection

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

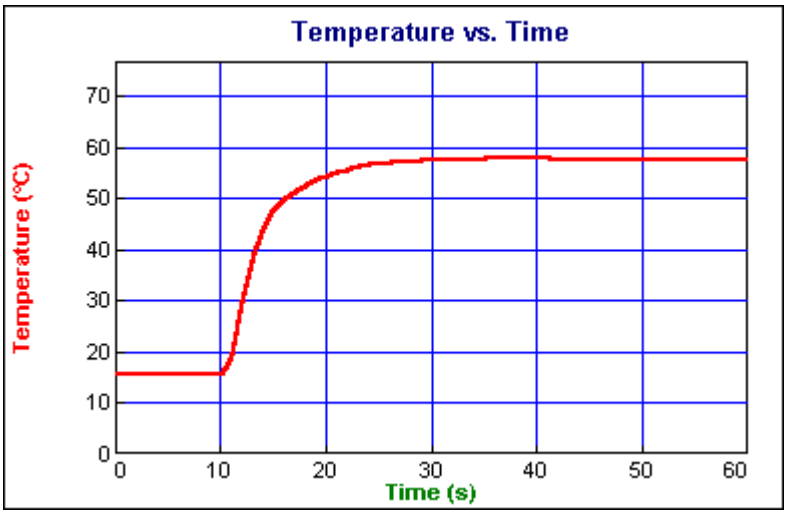
2. This experiment is intended to be used at the beginning of the school year to introduce you and/or your students to data collection with Vernier products. It also works well as a review if they have not used the products recently. The procedures in this experiment are more detailed than in the rest of the book. For this reason, it should be done first.

3. If you do not have hot tap water available in your classroom for Part I, water can be heated on a hot plate. A temperature of about 60°C works well.

4. As it is written, this experiment gives the students the option to print graphs of their data. If you prefer to have your students graph "by hand," instruct them to record data from the table at two-second intervals for this purpose.

SAMPLE DATA

Part I Time Graph



Maximum temperature (°C)	Elapsed time (s)
58.2	35

Part II Events with Entry

Group member number	Group member name	Maximum temperature (°C)
1	Starr C.	35.0
2	Kaden W.	32.5
3	Jeremy N.	33.4
4	Roberto G.	33.7
5	Patrice S.	32.1
6	Tonie L.	31.9
Group Average		33.1

ANSWERS TO QUESTIONS

Part I Time Graph

1. The curve is flat until the 20 seconds point, then it curves up rapidly. It slowly levels off at the maximum temperature.

2. Time is plotted on the horizontal axis because it is the independent variable. (The independent variable is plotted on the horizontal axis.)

3. Temperature is plotted on the vertical axis because it is the dependent variable in this experiment. (The dependent variable is plotted on the vertical axis.)

4. Answers will vary. In the example above, the response time is 31 seconds.

5. The response time was calculated by taking the time elapsed when the probe first reached the maximum temperature and subtracting 20 seconds.

Part II Events with Entry

6. See data table.

7. Answers will vary.

8. Answers will vary.

ACKNOWLEDGEMENT

We wish to thank Don Volz and Sandy Sapatka for their help in developing and testing this experiment.

Acids and Bases

Organisms are often very sensitive to the effect of acids and bases in their environment. They need to maintain a stable internal pH in order to survive—even in the event of environmental changes. Many naturally occurring biological, geological, and man-made chemicals are capable of stabilizing the environment's pH. This may allow organisms to better survive in diverse environments found throughout the earth. Teams will work in pairs, using one computer and two pH systems. One team will measure the effect of acid on biological materials, while the other team will measure the effect of base on biological materials. Each group will test the biological materials assigned to them, and all groups will share their data at the end of the class.

OBJECTIVES

In this experiment, you will

- Add an acid to a material and note the extent that it resists changes in pH.
- Add a base to a material and note the extent that it resists changes in pH.
- Work with classmates to compare the ability of different materials to resist pH changes.

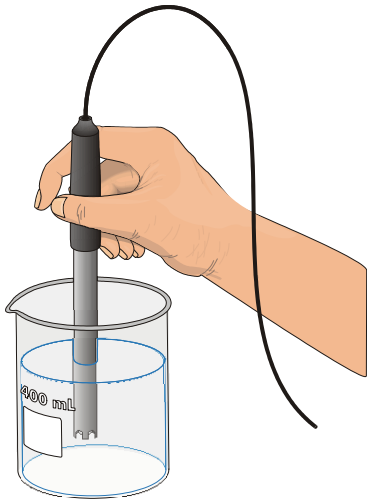


Figure 1

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier pH Sensor (one per team)
Various biological organisms (or parts of an organism), such as yeast, potato, orange juice, or a plant leaf solution.
Various non-biological materials, such as an antacid, buffer, carbonated water or soda, salt, or Alka-Seltzer solution.
two 250 mL beakers
one rinse bottle with distilled water
0.10 M HCl (acid) with dropper
0.10 M NaOH (base) with dropper
50 mL graduated cylinder
goggles
lab apron
two 50 mL beakers
Various simple biological materials, such as egg white, vitamin C, or gelatin solution.

PROCEDURE

1. Obtain and wear goggles.

2. Team A will use the pH probe in CH 1, while Team 2 will use the pH probe in CH 2. Before each use of the pH probe, you need to rinse the tip of the electrode thoroughly with distilled water. To do this, hold the pH electrode above a rinse beaker and use the rinse bottle to thoroughly rinse the electrode tip.

   Important: Do not let the pH electrode dry out. Keep it in a 250 mL beaker with about 100 mL of tap water when not in use. The tip of the probe is made of glass—it is fragile. Handle with care!

3. Connect the probes to the computer interface. Prepare the computer for data collection by opening the file "02 Acids and Bases" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

Testing the effect of acid and base on water

4. Label one of the 50 mL beakers *acidic* and label the other *basic*. Place 20 mL of distilled water in each beaker.

5. Rinse the pH probe thoroughly with distilled water, then place it into the beaker to be tested:
   - Team A: Place your probe in the beaker labeled *acidic*.
   - Team B: Place your probe in the beaker labeled *basic*.

6. Click Collect to begin making pH measurements.

7. The group will be entering the number of drops of acid or base added to the beaker. Before you begin, determine the initial pH of the solution. Click Keep, then type **0** in the text box and press ENTER.

8. Add acid or base to the solution. Stir each solution thoroughly after addition. **CAUTION:** Handle the hydrochloric acid with care. It can cause painful burns if it comes in contact with the skin. Sodium hydroxide solution is caustic. Avoid spilling it on your skin or clothing.
   - Team A: Add 5 drops of acid to the beaker labeled *acidic*.
   - Team B: Add 5 drops of base to the beaker labeled *basic*.

9. When the pH readings are stable click Keep. Enter the total number of drops of acid or base you have added to the water in the beaker. Type **5** in the text box and press ENTER.

10. Repeat Steps 8 through 9, adding 5 drops at a time until each team has added a total of 30 drops.

11. Click Stop when you have added a total of 30 drops.

12. Rinse the pH probe thoroughly and place the probe into the beaker of tap water. Clean the two 50 mL beakers.

13. Move your data to a stored data run. To do this, choose Store Latest Run from the Experiment menu. This will allow the data you obtained for water to be included in every future graph.

Testing the effect of acid and base on other materials

14. Test the effect of acid and base on a material assigned to you by your instructor:
    a. Obtain 20 mL of a solution to test from your instructor.
    b. Repeat Steps 5–12.
    c. Record the volume and pH values from the table in Table 1. Run 1 data will be the data collected using water. The data labeled Latest will be the data for your tested material.
    d. (optional) Print a copy of your graph. Enter your name(s) and the number of copies of the graph. The graph should have four lines on it—water with acid, water with base, your material with acid, and your material with base.
15. If time permits, repeat Step 14 for as many materials as you can. Before starting the next experiment, delete the latest run by choosing Delete Data Set ▸ Latest from the Data menu.
16. Obtain the pH values of any materials you did not test from your classmates. These values should be listed on the board. Record these values in Table 1.
17. Subtract the ΔpH of the acid from the ΔpH of the base to determine the Total Buffer Range. Record these values in Table 1.

DATA TABLE

Table 1										
Material rested	Add	pH, after adding this many drops								
		0	5	10	15	20	25	30	ΔpH	Total buffer range
	acid									
	base									
	acid									
	base									
	acid									
	base									
	acid									
	base									
	acid									
	base									
	acid									
	base									
	acid									
	base									
	acid									
	base									
	acid									
	base									

PROCESSING THE DATA

1. Make a series of graphs of the data obtained from other students. Alternatively, if instructed by your teacher, obtain a printout of each plot from other student teams. Construct the graphs so they appear similar to the plot your team made:
   - The horizontal axis has Volume scaled from 0 to 30 drops.
   - The vertical axis has pH scaled from 0 to 12.
   - The data you obtained for water should be included in every graph.
   - Construct one graph from the data in each row of Table 1.

2. Make a list of each material that was tested by the teams in your class. Place the most acidic material at the top of the list and the most basic material at the bottom of the list. Use the value corresponding to 0 drops of acid or base, as this value represents the natural acidity of the material.

Table 2		
Material	Initial pH	Rank
		most acidic
		2
		3
		4
		5
		6
		7
		least acidic

3. Put the materials tested into the following three categories:

Biological Organisms	Biological Chemicals	Non-Biological Chemicals

4. Calculate the pH change for each material. Record this in Table 1.

5. Make a second list of each material in Table 1. Place the material that had the largest Total Buffer Range at the top of the list in Table 3 and the smallest range at the bottom of the list.

Table 3		
Material	Total buffer range	Rank
		greatest change
		2
		3
		4
		5
		6
		7
		8
		least change

QUESTIONS

1. How should the pH of a material to test in the *Acidic* beaker compare to that in the *Basic* beaker before any acid or base is added? Why?

2. Referring to Question 1, does your data support your hypothesis? If not, what might cause the differences?

3. Generally, what was the effect of adding HCl to each solution? Was this true for every solution? Why do you think this happened the way it did?

4. Generally, what was the effect of adding NaOH to each solution? Was this true for every solution? Why do you think this happened the way it did?

5. Compare the various graphs of each substance. Why was it of value to include the plot of water in acid and water in base with every experiment?

6. Which class of materials, biological organisms, biological chemicals, or non-biological chemicals reacted most dramatically to the addition of acid or base? How does this relate to their complexity?

7. Which of the materials in Table 3 is the best buffer? The poorest buffer?

EXTENSION

1. Bring in common materials from home to test. How do you think they will respond? How did their response compare to your predictions?

TEACHER INFORMATION

Acids and Bases

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. To prepare the 0.1 M NaOH solution, use 4.0 g of solid NaOH pellets per 1 L of solution. **HAZARD ALERT:** Corrosive solid; skin burns are possible; much heat evolves when added to water; very dangerous to eyes; wear face and eye protection when using this substance. Wear gloves. Hazard Code: B—Hazardous.

3. To prepare the 0.1 M HCl solution, use 8.6 mL of concentrated acid per 1 L of solution. **HAZARD ALERT:** Highly toxic by ingestion or inhalation; severely corrosive to skin and eyes. Hazard Code: A—Extremely hazardous.

   The hazard information reference is: Flinn Scientific, Inc., *Chemical & Biological Catalog Reference Manual, 2000*, (800) 452-1261, www.flinnsci.com. See *Appendix G* for more information.

4. Try to make a 1% solution of the materials to test. It is not too critical to be exact. Add ~10 grams of material for each liter of solution.

5. Have the students help design the list of materials to use. Try to keep the three classes of materials balanced—biological organisms or tissues, biological chemicals, and non-biological chemicals.

   Good organisms or tissues to use might include blended liver, plant leaves, potato roots, yeast, fruit juices (from real fruit—not those <10% varieties!) or Euglena (if you culture them). Try to avoid oily materials—they will be difficult to clean off the probe.

   Good chemicals include starch, enzymes, gelatin, vitamin Bs or C, casein, egg white, or other simple, non-oily biochemicals.

   Good non-biological materials include a mix of buffers with non-buffers. Buffers might include soda water, Alka-Seltzer, phosphate buffer, Tums, etc. An interesting combination is aspirin and Bufferin. Good non-buffers include table salt and nitrogen fertilizer. It is fun to include rocks—try marble (calcium carbonate—a buffer in acid) and quartz.

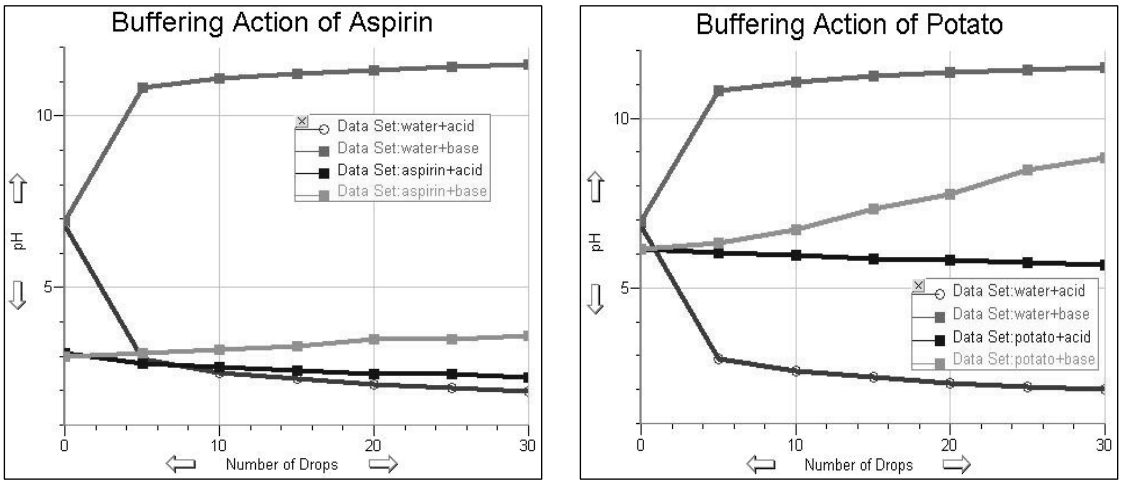
6. The stored calibration will work fine for this experiment. However, you may want to calibrate the pH probes before students use them. You can save this calibration as part of the experiment file. Refer to the teacher's section of experiment 18 for instructions on calibrating a pH Sensor.

7. Vernier Software sells a pH buffer package for preparing buffer solutions with pH values of 4, 7, and 10 (order code PHB). Simply add the capsule contents to 100 mL of distilled water. You can also prepare pH buffers using the following recipes:
   - pH 4.00: Add 2.0 mL of 0.1 M HCl to 1 L of 0.1 M potassium hydrogen phthalate.
   - pH 7.00: Add 582 mL of 0.1 M NaOH to 1 L of 0.1 M potassium dihydrogen phosphate.
   - pH 10.00: Add 214 mL of 0.1 M NaOH to 1 L of 0.05 M sodium bicarbonate.

8. Teams of students need to work together using the same computer and one interface. One team will be adding acid while a cooperating team adds base. They will need to keep synchronized! For example, when the acid group has added 20 drops, the base group must also have added 20 drops, or the plot will not be meaningful. You may also modify the experiment and use one pH Sensor at each computer.

SAMPLE DATA

Table 1										
Material Tested	Add	pH, after adding this many drops								
		0	5	10	15	20	25	30	ΔpH	Total Buffer Range
tap water	acid	6.81	2.91	2.53	2.35	2.19	2.08	1.99	–4.8	9.3
	base	6.95	10.83	11.08	11.24	11.34	11.42	11.48	4.5	
aspirin	acid	3.1	2.8	2.7	2.6	2.5	2.5	2.4	–0.7	1.3
	base	3.0	3.1	3.2	3.3	3.5	3.5	3.6	0.6	
Bufferin	acid	3.2	3.2	3.2	3.2	3.2	3.3	3.3	0.1	0.8
	base	3.2	3.4	3.6	3.7	3.8	3.9	4.1	0.9	
liver	acid	4.52	4.42	4.38	4.33	4.28	4.22	4.18	–0.3	1.6
	base	4.99	5.07	5.33	5.51	5.71	5.91	6.3	1.3	
egg white	acid	9.52	8.97	8.3	7.62	7.25	6.98	6.8	–2.7	3.6
	base	9.45	9.82	9.88	9.96	10.12	10.25	10.36	0.9	
gelatin	acid	5.75	5.11	4.84	4.68	4.45	4.2	4.06	–1.7	6.1
	base	5.75	7.62	9.18	9.56	9.78	9.99	10.13	4.4	
soda water	acid	4.65	3.36	2.6	2.5	2.4	2.3	2.3	–2.4	3.3
	base	4.45	4.7	4.9	5.0	5.1	5.2	5.3	0.9	
potato	acid	6.14	6.03	5.96	5.87	5.81	5.75	5.69	–0.4	3.1
	base	6.14	6.32	6.7	7.33	7.76	8.46	8.84	2.7	



Buffering action of aspirin and potato following the addition of acid and base

Classification of Materials		
Organisms or Tissues	Biological Chemicals	Non-Biological Chemicals
liver	aspirin	Bufferin
potato	gelatin	soda water
egg		water

Table 2		
Material	Initial pH	Rank
aspirin	3.0	most acidic
Bufferin	3.2	2
soda water	4.5	3
liver	4.7	4
gelatin	5.8	5
potato	6.1	6
tap water	6.9	7
egg	9.5	least acidic

Table 3		
Material	Total Buffer Range	Rank
tap water	9.3	worst
gelatin	6.1	2
egg	3.6	3
soda	3.3	4
potato	3.1	5
liver	1.6	6
aspirin	1.3	7
Bufferin	0.8	best

ANSWERS TO QUESTIONS

1. The values should be the same, since the same solution is in each beaker.

2. The actual results may vary. Possible reasons include:
   - The beakers were not cleaned equally well by the cooperative groups.
   - The probes were not calibrated equally, or differed slightly in their response.

3. The effect HCl had on each solution was to decrease its pH. Not all materials responded equally, and several did not respond much at all. These were better buffers.

4. The effect NaOH had on each solution was to increase its pH. Not all materials responded equally, and several did not respond much at all. These were better buffers.

5. Water acted as a control. The similarities and differences among the graphs can be noted more easily when each is compared to a single substance, such as water.

6. Non-biological chemicals, such as water and salt, reacted most dramatically to the addition of acid or base. Biologically complex materials and non-biological buffers resisted pH changes most. The non-biological materials were most divergent in their behavior. This is especially true if any of the graphs have a different scaling than the others.

   The order in which material reacted most dramatically to the addition of acid or base is: water, gelatin, egg white, soda water, potato, liver, aspirin, and Bufferin. The ranking by complexity is similar—water is the simplest material, followed by the two proteins. The soda water has a natural buffer, the bicarbonate ion. Soda water is very simple. Potato roots and livers are cellular material, thus more complex than any of the above. Finally, aspirin and Bufferin are simple chemicals with great buffering capacity.

   As a general rule, simple chemicals may or may not be good buffers, depending upon their make-up. Complex biological materials are almost always better buffers than simple ones, since there are usually a greater number of chemicals in cellular matter that serve as buffers.

7. Answers may vary. See Table 1 for sample values. Of these data, Bufferin is the best buffer and water is the poorest buffer.

Diffusion through Membranes

Diffusion is a process that allows ions or molecules to move from where they are more concentrated to where they are less concentrated. This process accounts for the movement of many small molecules across a cell membrane. Diffusion is the process by which cells acquire food and exchange waste products. Oxygen, for instance, might diffuse in pond water for use by fish and other aquatic animals. When animals use oxygen, more oxygen will diffuse to replace it from the neighboring environment. Waste products released by aquatic animals are diluted by diffusion and dispersed throughout the pond.

It is important to consider how the rate of diffusion of particles might be affected or altered.

- Diffusion may be affected by the steepness of the concentration gradient (the difference between the number of ions or molecules in one region of a substance and that in an adjoining region). The direction that a diffusing molecule or ion might travel in any particular direction is random. While the particles are diffusing, is there a net movement from where they are concentrated to where they are less concentrated?
- Diffusion might be affected by other different, neighboring particles. For instance, if oxygen diffuses towards a single-celled pond organism at a certain rate, will that rate be altered if some other molecule suddenly surrounded the organism? Would the presence of other molecules block or enhance the diffusion of a molecule, or would the molecule's rate be independent of particles that do not alter the concentration gradient?

One way to measure the rate of diffusion of ions is to monitor their concentration in solution over a period of time. Since ions are electrically charged, water solutions containing ions will conduct electricity. A Conductivity Probe is capable of monitoring ions in solution. This probe however, will not measure the amount of electrically neutral molecules dissolved in water. Salts, such as sodium chloride, produce ions when they dissolve in water. If you place a salt solution in a container such as dialysis tubing, the salt can travel through the very small holes in the tubing. When dialysis tubing containing a solution of salt ions is placed into a beaker of water, the ions can diffuse out of the tubing and into the surrounding water. In this way, you will be able to measure the diffusion of salts in a solution of water and determine how concentration gradients and the presence of other particles affect the diffusion of the salt across a membrane.

OBJECTIVES

In this experiment, you will

- Use a computer and Conductivity Probe to measure the ionic concentration of various solutions.
- Study the effect of concentration gradients on the rate of diffusion.
- Determine if the diffusion rate for a molecule is affected by the presence of a second molecule.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier Conductivity Probe
three 18 × 150 mm test tubes with rack
1%, 5%, and 10% salt water
400 mL beaker
dialysis tubing, 2.5 cm × 12 cm
dropper pipet or Beral pipet
scissors
stirring rod
5% sucrose (table sugar) solution
dental floss or clamp
ring stand and utility clamp

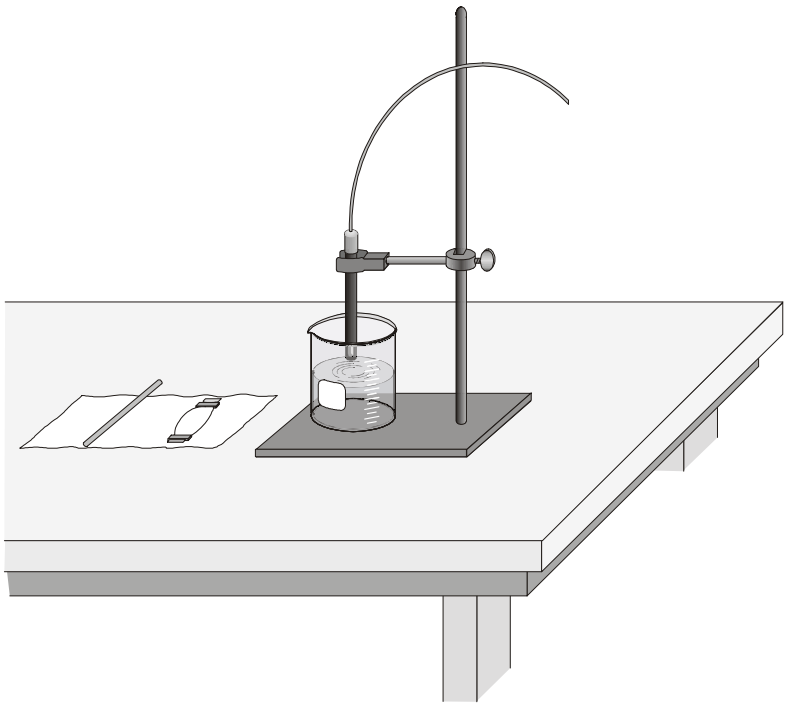


Figure 1

PROCEDURE

1. Connect the Conductivity Probe to the computer interface. Check to be sure the Conductivity Probe is set to the intermediate setting, 2000 µS/cm (equivalent to a concentration of 1000 mg/L).

2. Prepare the computer for data collection by opening the file "03 Membrane Diffusion" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

Part I Concentration gradients

3. Test whether different concentration gradients affect the rate of diffusion. You will place three solutions of differing salt concentrations (1%, 5%, and 10%) in distilled water. Each salt solution will be placed in a dialysis tube and allowed to diffuse into the surrounding water. When salt diffuses, the conductivity of water in the beaker will increase.

4. In Table 1, predict what you believe will happen in this set of experiments. How will the rate of diffusion change when a 10% salt solution is placed in contact with pure water compared to when a 1% salt solution is placed in contact with pure water?

5. Prepare the dialysis tubing. Obtain a piece of wet dialysis tube and a dialysis tubing clamp or a short (approximately 10 cm) length of dental floss. Using the clamp or floss, tie one end of the tube closed about 1 cm from the end, as shown in Figure 2.

6. Place a 1% salt solution into a section of dialysis tubing. To do this,
   a. Obtain about 15 mL of a 1% salt water solution in a test tube.
   b. Using a funnel or Beral pipet, transfer about 10 mL of the 1% salt water into the dialysis tube, as in Figure 2. **Note:** To open the tube, you may need to rub the tubing between your fingers a bit.
   c. Tie off the top of the dialysis tube with a clamp or a new length of dental floss. Try not to allow any air into the dialysis tube. The tube should be very firm after it is tied or clamped. Trim off any excess dental floss extending more than 1 cm from either knot.
   d. Wash the outside of the tubing with tap water thoroughly, so that there is no salt water adhering to the tubing.

Figure 2

7. Place 300 mL of water into a 400 mL beaker. If the conductivity of the tap water is low (50 mg/L or less), use tap water to fill the beaker. Otherwise, use distilled water.

8. Position the Conductivity Probe into the water as shown in Figure 1. Place the dialysis tube into the water. Be sure the tubing is submerged completely under the water. **Important:** Be sure to position the Conductivity Probe and dialysis tubing the same distance apart in each trial.

9. After stirring the solution for 30 seconds, begin data collection by clicking ▸ Collect. Stir the solution slowly and continuously throughout the two-minute data collection period. Data collection will automatically end after two minutes have passed.

10. Determine the rate of diffusion.
    a. Move the mouse pointer to the point where the data values begin to increase. Hold down the mouse button. Drag the pointer to the end of the data and release the mouse button.
    b. Click the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
    c. Record the slope of the line, *m*, as the rate of diffusion in Table 2.
    d. Close the linear regression floating box.

11. Remove one of the clamps. If the dialysis tubing is tied off with floss, use a pair of scissors and carefully cut one of the dental floss knots and discard the floss. If you accidentally make a cut in the tubing, replace it.

12. Empty all of the liquid out of the dialysis tube. Squeeze the excess liquid out with your fingers.

13. Obtain 15 mL of a 5% salt solution in a test tube. Repeat Steps 6–12, substituting this 5% salt solution for the 1% solution.

14. Obtain 15 mL of a 10% salt solution in a test tube. Repeat Steps 6–12, substituting this 10% salt solution for the 1% solution.

15. Examine your data closely and make a conclusion. Record your conclusion in Table 1.

Part II Effect of other molecules

16. Measure the rate of diffusion of salt while it is in the presence of another, non-conducting solution. Since sugar does not form ions in solution, it should not conduct electricity. Sugar will be added to the water to determine whether it interferes with the diffusion of salt.

17. In Table 1, predict what you believe will happen in this set of experiments. Will the non-conducting sugar in the water block or reduce the diffusion rate of salt? Why?

Test to determine if water or a sugar solution conducts electricity.

18. Place some water in a clean 400 mL beaker.

19. Test the total dissolved solids concentration of the water by placing a clean Conductivity Probe into it. Record the total dissolved solid concentration value in Table 3. The total dissolved solids value should be displayed in the meter at the right of the screen.

20. Obtain 300 mL of a 5% sugar solution in a clean 400 mL beaker.

21. Test the total dissolved solids concentration of the 5% sugar solution by placing a clean Conductivity Probe into it. Record the total dissolved solids value in Table 3. The total dissolved solids value should be displayed in the meter on the right of the screen.

Test if 5% sugar interferes with the diffusion of a 5% salt solution.

22. Repeat Steps 6–12, with the following changes:
    - Use 300 mL of sugar water in place of the water in Step 7.
    - Substitute a 5% salt solution for the 1% solution.
    - Record the rate of diffusion in Table 4.

23. Examine your data closely and make a conclusion. Record your conclusion in Table 1.

DATA

Table 1

	Prediction	Conclusion
Part I		
Part II		

Part I

Table 2	
Salt Concentration (%)	Rate of diffusion (mg/L/s)
1	
5	
10	

Part II

Table 3	
Solution	Concentration (mg/L)
Distilled water	
Sugar water	

Table 4: Summary of Data	
Solution	Rate of diffusion (mg/L/s)
5% salt	
5% salt / 5% sugar	

QUESTIONS

1. What conclusion can you draw from the data in Table 2?
2. How did your conclusion compare to your prediction for Part I? Can you account for any differences?
3. If the rates in any of the three experiments varied in Part I, calculate how much faster each rate was compared to that for the 1% salt solution. For instance, if the rate of the 1% solution was 1 μS/s and the rate of the 10% solution was 5 μS/s, then the rate of diffusion for the 10% solution would be (5/1) five times the rate of the 1% salt solution.
4. Compare the ionic concentration of pure water with a sugar water solution. How do you account for this?
5. What conclusion can you draw from the data in Tables 3 and 4?

EXTENSIONS

1. Make a plot of the rate of diffusion *vs*. the salt concentration in the dialysis bag. Using your plot, estimate the rate of diffusion of a 3% salt solution.
2. If the results of the experiments in Part I can be extrapolated to diffusion in living systems, how would a single-celled organism respond in an oxygen rich pond compared to an oxygen poor pond? Explain.
3. Design an experiment to determine the effect of temperature on the diffusion of salt. Perform the experiment you designed.

4. Ectotherms are organisms whose body temperature varies with the surrounding environment. On the basis of on your data from Extension Question 3, how do you expect the oxygen consumption of ectotherms to vary as the temperature varies? Explain.

5. If waste products of a single celled organism were released by the organism into the pond, how would that affect the organism's ability to obtain oxygen as readily?

TEACHER INFORMATION

Diffusion through Membranes

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. If the water in your area is very soft, you may want to use tap water instead of distilled water. Test to see if the conductivity of the tap water is less than about 50 mg/L salt.

3. Provide each group with pre-cut, hydrated dialysis tubing. The tubing must be soaked in water for at least ten minutes prior to use. The tubing should be soft and flexible.

4. Use dialysis tubing clamps if at all possible, as this will speed things up greatly. If desired, use dental floss or string to tie off the dialysis tubing. The floss works exceptionally well. You may want to show students how to tie off the dialysis tubes.

5. Have students check their dialysis tubes for leakage. This should be done before each experiment. Leaky tubes should be replaced.

6. Any sugar may be used in Part II. Table sugar is inexpensive and readily available.

7. To prepare 5% sugar solution, add 50 grams of sugar to make one liter of solution (300 mL per group is needed).

8. To prepare 1% salt solution, add 10 grams of NaCl to make one liter of solution (15 mL per group is needed).

9. To prepare 5% salt solution, add 50 grams of NaCl to make one liter of solution (30 mL per group is needed).

10. To prepare 10% salt solution, add 100 grams of NaCl to make one liter of solution (15 mL per group is needed).

11. The stored conductivity calibration (for 0–2000µS/cm) works well for this experiment. The ionic concentration is approximately proportional to the conductivity of the solution.

SAMPLE RESULTS

The following data may be different from students' results.

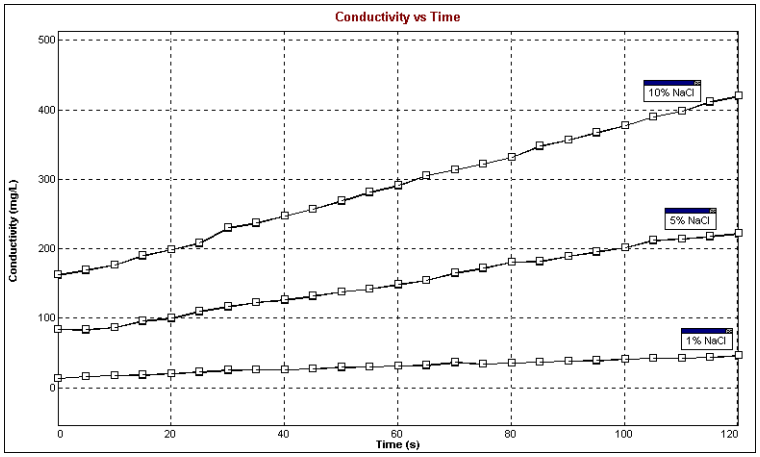
Part I

Table 2	
Salt concentration (%)	Rate of diffusion (mg/L/s)
1	0.25
5	1.23
10	2.19

Part II

Table 3	
Solution	Conductivity (mg/L)
Distilled water	1.5
Sugar water	1.9

Table 4: Summary of Data	
Solution	Rate of diffusion (mg/L/s)
5% salt	1.23
5% salt / 5% sugar	1.21



Diffusion through dialysis tubing of differing salt concentrations

ANSWERS TO QUESTIONS

1. Rate of diffusion should increase with increasing salt concentration.

2. The rate of diffusion should increase as the concentration gradient becomes steeper. The rate of the 10% salt solution should be the greatest and the rate of the 1% salt solution should be the lowest of the three.

3. The rate of the 10% salt solution should be approximately ten times that of the 1% solution, while the rate of the 5% salt solution should be five times that of the 1% solution.

4. The conductivity should be the same, as neither will conduct appreciably. Neither molecule is electrically charged.

5. Student answers will vary. The rates of diffusion should be the same.

Conducting Solutions

In this experiment, you will study the electrical conductivity of water and various water solutions. A solution can contain molecules, ions, or both. Some substances, such as sucrose ($C_{12}H_{22}O_{11}$) and glucose ($C_6H_{12}O_6$), dissolve to give a solution containing mostly molecules. An equation representing the dissolving of sucrose (table sugar) in water is:

$$C_{12}H_{22}O_{11}(s) \longleftrightarrow C_{12}H_{22}O_{11}(aq)$$

where (s) refers to a solid substance and (aq) refers to a substance dissolved in water. Other substances, such as calcium chloride ($CaCl_2$), dissolve in water to produce a solution containing mostly ions. An equation is:

$$CaCl_2(s) \longleftrightarrow Ca^{2+}(aq) + 2\ Cl^-(aq)$$

Calcium ions are necessary for muscle contraction, mitochondrial activity, bone formation, and many other metabolic processes. Organisms may obtain minerals such as calcium from their water supply, since ions dissolve in water.

You will determine conductivity of the solutions using a computer-interfaced Conductivity Probe. The unit of conductivity in this experiment is microsiemens per centimeter, or µS/cm.

OBJECTIVES

In this experiment, you will

- Write equations for the dissolving of substances in water.
- Use a Conductivity Probe to test the electrical conductivity of solutions.
- Determine whether molecules or ions are responsible for electrical conductivity of solutions.

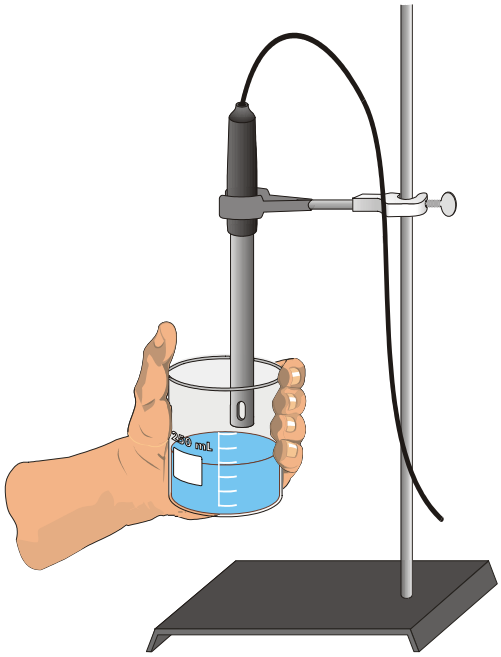


Figure 1

MATERIALS

computer
Vernier computer interface
Vernier Conductivity Probe
Logger*Pro*
sodium chloride, NaCl, solution
calcium chloride, $CaCl_2$, solution
aluminum chloride, $AlCl_3$, solution
ring stand
ethanol, C_2H_6O, solution
sucrose, $C_{12}H_{22}O_{11}$, solution
glucose, $C_6H_{12}O_6$, solution
stream or lake water
ocean water (optional)
various foods in solution
distilled water
utility clamp

PRE-LAB EXERCISES

Many of the materials you will be using today are found in common household items. A list of common names or uses can be found below:

Sodium chloride, NaCl	Common household salt
Calcium chloride, $CaCl_2$	Used to pickle cucumbers, or to help concrete cure in cold weather
Acetic acid, CH_3COOH	Vinegar
Ethanol, C_2H_6O	Found in gasoline or in alcoholic beverages. Usually obtained from yeast fermentation
Fructose, $C_6H_{12}O_6$	Fruit sugar
Sucrose, $C_{12}H_{22}O_{11}$	Table sugar, beet or cane sugar
Glucose, $C_6H_{12}O_6$	Corn or blood sugar

1. An equation representing the dissolving of sucrose in water is:

$$C_{12}H_{22}O_{11}(s) \longleftrightarrow C_{12}H_{22}O_{11}(aq)$$

Like solid sucrose, the substances glucose, $C_6H_{12}O_6(s)$, and ethanol, $C_2H_6O(l)$, dissolve in water to yield solutions containing mostly molecules. Write equations showing the dissolving of these two liquids in water in Table 1.

2. An equation showing the dissolving of $CaCl_2$ in water is:

$$CaCl_2(s) \longleftrightarrow Ca^{2+}(aq) + 2\ Cl^-(aq)$$

Like $CaCl_2$, the substances NaCl and $AlCl_3$ dissolve in water to give solutions containing mostly ions. Write equations showing the dissolving of these two solids in water in Table 2.

PROCEDURE

1. Obtain and wear goggles.
2. Secure the Conductivity Probe with the ring stand and utility clamp as shown in Figure 1.
3. Connect the Conductivity Probe to the computer interface. Check to be sure the Conductivity Probe is set to 0–20,000 μS/cm.
4. Prepare the computer for data collection by opening the file "04 Conducting Solutions" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

5. Test the conductivity of each solution listed in the data table. You can do the tests in any sequence.
   a. Carefully raise each vial and its contents up around the Conductivity Probe until the hole near the probe end is completely submerged in the solution being tested. **Important:** Since the two electrodes are positioned on either side of the hole, this part of the probe must be completely submerged.
   b. Briefly swirl the beaker contents. Once the conductivity reading in the meter has stabilized, record the value in Table 3.
   c. Before testing the next solution, clean the electrodes by surrounding them with a 250 mL beaker and rinse them with distilled water from a wash bottle. Blot the outside of the probe end dry using a tissue. It is *not* necessary to dry the *inside* of the hole near the probe end.

DATA

Table 1	
$C_6H_{12}O_6(s)$	$C_2H_6O(l)$

Table 2	
NaCl(s)	$AlCl_3(s)$

Table 3		
Solution	Material	Conductivity (µS/cm)
1	Distilled water	
2	Sodium chloride, NaCl	
3	Calcium chloride, $CaCl_2$	
4	Aluminum chloride, $AlCl_3$	
5	Ethanol, C_2H_6O	
6	Sucrose, $C_{12}H_{22}O_{11}$	
7	Glucose, $C_6H_{12}O_6$	
8	Tap water	
9	Stream water	
10	Ocean water	
11		
12		

QUESTIONS

1. Which solutions conduct electricity best, those containing mostly ions or those containing mostly molecules?

2. Does distilled water conduct electricity well? Explain.

3. Does tap water conduct electricity? Account for this observation.

4. Consider the conductivity readings for the NaCl, $CaCl_2$, and $AlCl_3$ solutions. What trend do you observe? Account for this trend.

5. How does the conductivity of ocean water compare to pond or stream water? How can you account for this?

6. Which foods in solution conducted electricity well? How can you account for this?

7. Suggest three other substances whose water solutions would conduct electricity well. Explain how you decided on your choices.

EXTENSIONS

1. Test your predictions for Question 7 above.

TEACHER INFORMATION

Conducting Solutions

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Two or more sets of the solutions can be made available in small beakers or jars.
3. The solutions can be prepared (using distilled water) as follows:

0.05 M NaCl (2.93 g/liter)	0.05 M sucrose (17.1 g/liter)
0.05 M $AlC1_3$ (6.7 g/liter)	0.05 M $CaCl_2$ (5.55 g/liter)
0.05 M ethanol (2.3 g or 2.9 mL/liter)	0.05 M glucose (9.0 g/liter)

4. A variety of food suspensions may be used. Both plant and animal foods might be considered.
5. To prepare food suspensions, cut the food into small pieces and blend for 5 to 10 seconds, or until finely chopped. Strain the food through cheesecloth and collect the resulting filtrate for testing. This way, students will be testing the resulting dilute solution that will contain varying amounts of ions and molecules. Avoid foods that are high in oil or fat content, as they may leave residues on the electrodes of the Conductivity Probe (see the probe user's guide that was shipped with the probe for further information).
6. Several sources of water can be tested, including stream, tap, ocean, and lake water. Students may want to bring samples in from home to test.
7. The calibration that is stored within the data-collection software will work fine for a comparison of different solutions. For more accurate conductivity readings, you (or your students) can do a 2-point calibration for each Conductivity Probe using air (0 conductivity value) and the calibration solution that came with the Conductivity Probe (1000 µS/cm value).
8. If you make measurements of ocean water, you will need to dilute samples to 1/4 of their original concentration by adding 100 mL of the salt water sample to 300 mL of distilled water. This diluted sample can then be measured using the Conductivity Probe at the high-range setting. Multiply the conductivity reading by 4 to obtain the actual conductivity.

SAMPLE RESULTS

Table 1		
Solution	Material	Conductivity (µS/cm)
1	Distilled water	0
2	Sodium chloride, NaCl	5214
3	Calcium chloride, $CaCl_2$	9362
4	Aluminum chloride, $AlCl_3$	11707
5	Ethanol, C_2H_6O	0
6	Sucrose, $C_{12}H_{22}O_{11}$	0
7	Glucose, $C_6H_{12}O_6$	0
8	Tap water	varies (20 – 1000)

ANSWERS TO QUESTIONS

1. The solutions containing mostly ions conduct best.

2. Distilled water does not conduct well because it contains few ions.

3. Tap water does conduct electricity. It contains Ca^{2+} Mg^{2+}, Fe^{3+} CO_3^{-2}, HCO_3^- and other ions that dissolve into water as it flows through and over soil and rocks.

4. The conductivity increases from NaCl through $AlC1_3$ because of the increasing number of ions. A formula unit of NaCl contributes two ions, $CaCl_2$ three ions, and $AlCl_3$ four total ions.

5. Ocean water conducts much more than pond water. It must have many more ions in it than pond water.

6. Answers may vary.

7. Any soluble ionic solid, and some soluble molecular substances, will give a conducting solution. Some common ionic solids that give conducting solutions include
   - The "no-salt" substitute, potassium chloride (KCl).
   - Salt peter, sodium nitrate ($NaNO_3$).
   - Ammonium chloride (NH_4Cl).
   - Epsom salts, magnesium sulfate ($MgSO_4$).
   - Drano®, sodium hydroxide (NaOH).
   - Muratic acid, hydrochloric acid (HCl), is an example of a conducting solution made by dissolving a molecular substance.

Osmosis

In order to survive, all organisms need to move molecules in and out of their cells. Molecules such as gases (e.g., O_2, CO_2), water, food, and wastes pass across the cell membrane. There are two ways that the molecules move through the membrane: *passive transport* and *active transport*. While active transport requires that the cell uses chemical energy to move substances through the cell membrane, passive transport does not require such energy expenditures. Passive transport occurs spontaneously, using heat energy from the cell's environment.

Diffusion is the movement of molecules by passive transport from a region in which they are highly concentrated to a region in which they are less concentrated. Diffusion continues until the molecules are randomly distributed throughout the system. Osmosis, the movement of water across a membrane, is a special case of diffusion. Water molecules are small and can easily pass through the membrane. Other molecules, such as proteins, DNA, RNA, and sugars are too large to diffuse through the cell membrane. The membrane is said to be semipermeable, since it allows some molecules to diffuse though but not others.

If the concentration of water on one side of the membrane is different than on the other side, water will move through the membrane seeking to equalize the concentration of water on both sides. When water concentration outside a cell is greater than inside, the water moves into the cell faster than it leaves, and the cell swells. The cell membrane acts somewhat like a balloon. If too much water enters the cell, the cell can burst, killing the cell. Cells usually have some mechanism for preventing too much water from entering, such as pumping excess water out of the cell or making a tough outer coat that will not rupture. When the concentration of water inside of a cell is greater than outside, water moves out of the cell faster than it enters, and the cell shrinks. If a cell becomes too dehydrated, it may not be able to survive. Under ideal conditions, the water concentration outside is nearly identical to that inside.

In this experiment, you will use a Gas Pressure Sensor to measure the rate of pressure change as water moves in to or out of the cell (dialysis tubes filled with various concentrations of syrup solution). The pressure generated is called osmotic pressure and is in response to the overall movement of molecules, both water and syrup, inside the dialysis cell.

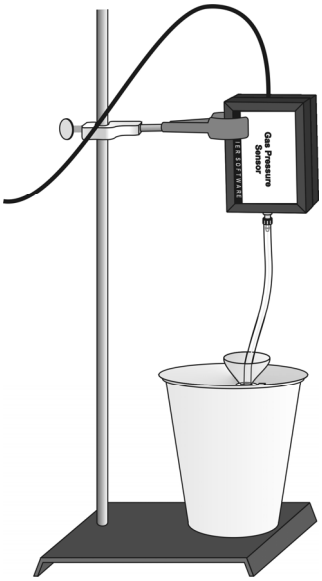


Figure 1

OBJECTIVES

In this experiment, you will

- Use a Gas Pressure Sensor to investigate the relationship between water movement and solute concentration.
- Determine the water potential of potato cells.

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier Gas Pressure Sensor
four 16 x 100 mm test tubes
20 cm piece of plastic tubing
test tube rack
25 mL graduated cylinder
various concentrations of syrup solutions
dialysis tubing, 2.5 cm x 15 cm
dialysis tubing clamp or floss
utility clamp
ring stand
plastic tubing clamp
plastic tubing with Luer connector
20 oz. Styrofoam cup or 600 mL beaker
warm water bath
plastic syringe

PROCEDURE

1. Place four test tubes in a rack and label them 90%, 80%, 70%, and 60%.
2. Fill each test tube with 15 mL of the corresponding syrup solution.
3. Connect the plastic tubing with the Luer connector to the valve on the Gas Pressure Sensor.
4. Connect the Gas Pressure Sensor to the computer interface. Prepare the computer for data collection by opening the file "05 Osmosis" from the *Agricultural Science with Vernier* folder of Logger *Pro*.

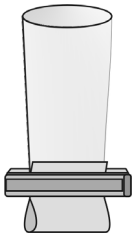


Figure 2

5. Using a ring stand and clamp, mount the pressure sensor above the insulated water container, as in Figure 1. **Note:** Be careful not to get any solution on the sensor.
6. Obtain a piece of wet dialysis tubing and dialysis tubing clamp and clamp off one end as shown in Figure 2.
7. Prepare the dialysis tubing.
   a. Connect the 20 cm segment of plastic tubing to the syringe and draw up the contents of the 90% syrup solution.
   b. Rub the open end of the dialysis tubing between your thumb and forefinger to create an opening.
   c. Insert the end of the plastic tube into the dialysis tubing and carefully inject the syrup solution as shown in Figure 3.
   d. Rinse the syringe and tubing with water and set it aside.

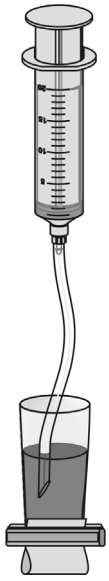


Figure 3

8. Complete the experimental setup.
   a. Slide a plastic tubing clamp on to the plastic tubing connected to the Gas Pressure Sensor.
   b. Insert the end of the tubing into the top of the dialysis tubing filled with the syrup solution so that it is about 1 cm above the liquid.
   c. Wrap the dialysis tubing tightly around the plastic tube.
   d. Slide the clamp over the end of the wrapped dialysis tubing and squeeze the tubing clamp shut as shown in Figure 4, leaving minimal airspace above the liquid.

Figure 4

9. Fill the insulated water container with 600 mL of 37°C water and place the clamped dialysis tube in the container. Check that the dialysis tubing holding the solution is completely submerged and without kinks. Wait five minutes.
10. After 5 minutes click Collect to begin data collection.
11. When data collection is finished, carefully release the tubing clamp by pushing up on one side of the clamp while pushing down on the other. **Important:** Do not disconnect the tubing from the sensor prior to releasing the tubing clamp. There will be enough pressure within the dialysis tubing to create a mess.
12. Rinse out the dialysis tubing and beaker.
13. Determine the rate of change in osmotic pressure.
    a. Move the mouse pointer to the point where the data values begin to increase. Hold down the left mouse button. Drag the mouse pointer to the end of the data and release the mouse button.
    b. Click the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
    c. Record the slope of the line, *m*, as the rate of change in osmotic pressure for a 90% syrup solution.
    d. Close the linear regression floating box.
14. Store the data by choosing Store Latest Run from the Experiment menu.
15. Repeat Steps 6–14 for the remaining syrup solutions.

DATA

Table 1	
Syrup concentration	Rate of pressure change (kPa/min)
90%	
80%	
70%	
60%	

Table 2	
Syrup solution concentration	Class average rate of pressure change (kPa/min)
90%	
80%	
70%	
60%	

PROCESSING THE DATA

1. On Page 2 of the experiment file, create a graph with the rate of pressure change (slope) on the y-axis and syrup concentration on the x-axis.

QUESTIONS

1. Which solutions, if any, produced a positive slope? Was water moving in or out of the cell (dialysis tubing) under these circumstances? Explain.
2. Which solutions, if any, produced a negative slope? Was water moving in or out of the cell under these circumstances? Explain.
3. Does syrup move in or out of the cell? Explain.
4. Examine the graph of the rate of pressure change *vs*. the syrup concentration. Describe any pattern in the data.
5. Use the graph to estimate the concentration of syrup that would yield no change in pressure. Why is this biologically significant?
6. When wilted plants are watered, they tend to become rigid. Explain how this might happen.
7. Explain the strengths and weakness of this dialysis model with respect to an animal and plant cell.
8. Discuss and explain potential reasons to account for variation in class average pressure change at specific syrup concentrations.
9. This exercise was carried out at initial temperatures of 37°C. Predict the effect of increased and decreased initial temperature on rates of pressure change for each of these different solution concentrations.
10. Predict the rates of pressure change if the dialysis tubing is placed in an insulated cup holding 1000 mL of 37°C water. Explain your reasoning.
11. Predict the rate of pressure change if the dialyisis tubing were filled with 10 mL of 80% syrup solution and 5 mL of 1.0 M sodium chloride solution. Explain your reasoning.

EXTENSION – WATER POTENTIAL

Water potential is a term used when predicting the movement of water into or out of plant cells. Water always moves from an area of higher water potential to an area of lower water potential. The symbol for water potential is the Greek letter Psi, Ψ. Water potential consists of a physical pressure component called pressure potential Ψ_p, and the effects of solutes called solute potential, Ψ_s.

Ψ	=	Ψ_p	+	Ψ_S
Water potential	=	Pressure potential	+	Solute potential

Distilled water in an open beaker has a water potential of zero. The addition of solute decreases water potential while the addition of pressure increases water potential. A water potential value can be positive, negative, or zero. Water potential is usually measured in bars, a metric measure of pressure. (1 kPa = .1 bar)

In this experiment, you will measure the percent change in mass of potato cores after they have soaked in various concentrations of sugar solutions for a 24 hour period. You will use this data to calculate the water potential of the potato cells.

MATERIALS

computer
Logger *Pro*
four potato cores
0 M, 0.33 M, 0.67 M, or 1.0 M sugar solution
250 mL beaker
plastic wrap
balance
paper towels

PROCEDURE

You will be assigned one or more of the sugar solutions in which to soak your potato cores.

1. Pour 100 mL of the assigned sugar solution into a 250 mL beaker.
2. Measure and record the mass of the four potato cores together.
3. Put the four cores into the beaker of sugar solution.
4. Cover the beaker with plastic wrap and allow it to stand for a 24-hour period.
5. Remove the cores from the beaker, blot with a paper towel, and determine the mass of the four cores together after soaking.
6. Calculate the percent change in mass and record your data for the sugar concentration tested in Table 2 as well as on the class data sheet.
7. Repeat Steps 1–6 for any additional assigned sugar solutions.

DATA

Table 2			
Sugar solution concentration	Initial mass (g)	Final mass (g)	Percent change in mass
0.00 M			
0.33 M			
0.67 M			
1.0 M			

Table 3 Class Data of Percent Change in Mass										
Sugar solution concentration	Group 1	Group 2	Group 3	Group 4	Group 5	Group 6	Group 7	Group 8	Total	Class average
0.00 M										
0.33 M										
0.67 M										
1.00 M										

Table 4	
Sugar molar concentration	=
Solute potential	=
Water potential	=

PROCESSING THE DATA

1. On Page 3 of the experiment file, create a graph with the percent change in mass on the y-axis and concentration of sugar on the x-axis.

2. Perform a linear regression to determine the molar concentration of sugar solution at which the mass of the potato cores does not change.

   a. Move the mouse pointer to the first data point. Press the left mouse button. Drag the pointer to the last data point.
   b. Click the Linear Fit button,  , to perform a linear regression. A floating box will appear with the formula for a best fit line.

c. Choose Interpolate from the Analyze menu. Move the mouse pointer along the regression line to the point where the line crosses the x axis. This point represents the molar concentration of sugar with a water potential equal to the potato core water potential. Record this concentration in Table 4.

3. Use the sugar molar concentration from the previous step to calculate the solute potential of the sugar solution.

   a. Calculate the solute potential of the sugar solution using the equation

$$\Psi_s = -iCRT$$

   where i = ionization constant (1.0 for sugar since it doesn't ionize in water)

   C = sugar molar concentration (determined from the graph)

   R = pressure constant (R = 0.0831 liter bars/mol-K)

   T = temperature (K)

   b. Record this value with units in Table 4.

4. Calculate the water potential of the solution using the equation

$$\Psi = \Psi_p + \Psi_s$$

   The pressure potential of the solution in this case is zero because the solution is at equilibrium. Record the water potential value with units in Table 4.

QUESTIONS

1. What may happen to an animal cell if water moves into it? How does this differ from what would happen in a plant cell?

2. What factors affect water potential?

3. If a plant cell has a higher water potential than its surrounding environment and the pressure is equal to zero, will water move in or out of the cell? Explain why.

TEACHER INFORMATION

Osmosis

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. The Extension on water potential has been added to this experiment to further address the concept of water movement in and out of cells.

3. Pre-soak 15 cm segments of dialysis tubing in water.

4. Prepare five sugar solutions according to specifications in Table 1. They should be 90%, 80%, 70%, and 60% with the tubes labeled accordingly. Contents of each tube should be capped and shaken until solution is homogeneous. Tubes are then placed in a test tube rack until ready for use.

Table 1

Tube composition	Amount of syrup (mL)	Amount of water (mL)
90%	13.5	1.5
80%	12.0	3.0
70%	10.5	4.5
60%	9.0	6.0

5. Make a warm water bath (37^{o}C) available to students.

6. If more than one sensor is available to each group, prepare and connect additional dialysis tubing/ syrup solution set-ups for each additional gas pressure sensor.

7. If more than one sample solution is being tested by the group, each run will need a separate container of warmed water.

SAMPLE RESULTS

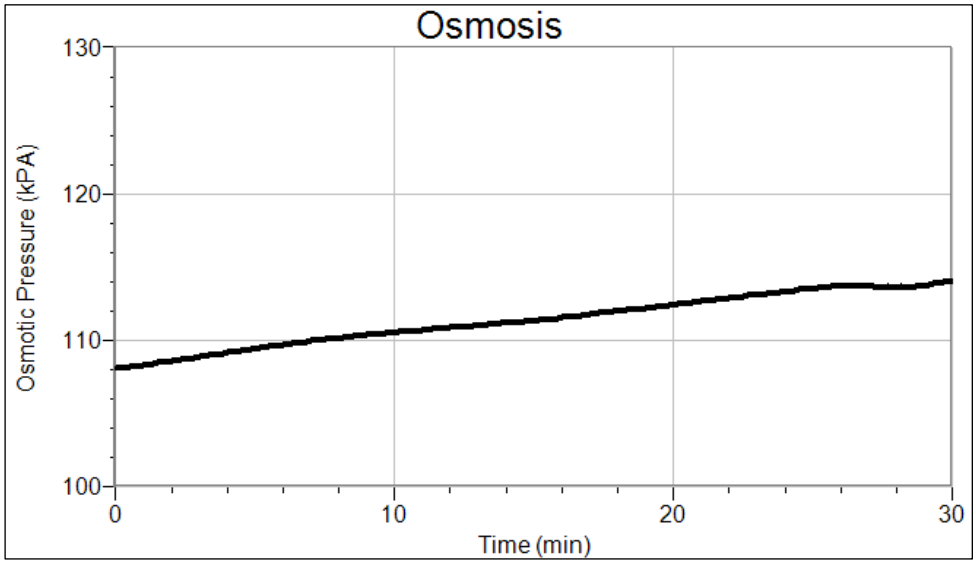


Table 1	
Concentration	Rate of pressure change (kPa/min)
90%	0.3
80%	0.27
70%	0.22
60%	0.16

EXTENSION SAMPLE DATA

Average Percent Change in Mass - Class Data	
0.00 M	18.5
0.33 M	1.6
0.67 M	–15.0
1.0 M	–18.0

Sugar molar concentration	0.41 M
Solute potential	–10.0 bars
Water potential	–10.0 bars

ANSWERS TO QUESTIONS

1. All solutions produce a positive slope as water will seek to move from high concentration into the syrup solution with a lesser water concentration. As the water moves into the "cell", the osmotic pressure increases since the larger sugar molecules are not able to diffuse the opposite direction across the membrane.

2. None of the solutions produced a negative slope as the solute inside the "cell" consisted of a solution of sugar molecules that were too large to diffuse through the membrane into the surrounding water. The only overall movement would be the water from outside the "cell" moving into the cell attempting to dilute the inner solution to a state similar to the outer region. Diffusion and the special case, osmosis, seek to establish equilibrium that would, if attained, result in a zero slope with concentrations being similar on both sides of the membrane.

3. Syrup, due to the large sugar molecules and the small pore size of the dialysis membrane, remains inside this cell.

4. All trials seem to proceed to a pressure that ultimately results in the rupture of the cell, time to reach this state appears to offer the only difference.

5. The syrup concentration that would yield no change in pressure would be one with possibly just a trace of syrup. This solution would be near isotonic. The significance of this isotonic situation is that the pressure maintained inside the cell is the same as that of the fluid surrounding the cell. This maintains cell size and function at normal osmotic pressure.

6. Cells in wilted plants are in some state of dehydration. For those plants that have not exceeded their critical wilting point, watering will rehydrate the cells and increase their turgor allowing the plant to resume life in a state of healthy water balance. The turgor is brought about by osmotic pressure within plant cells.

7. This dialysis model of a cell works in offering a descriptive and manipulative structure for studying the relative nature of a cell's plasma membrane. One can interact with this system by adjusting the solute conditions and recording changes in pressure. The drawback to this method is the matter of scale, membrane pore size, lack of control of ion movement, and inability to detect changes in solutions resembling those found in inter and intra cellular environments.

8. Some of the reason for variation would be technique differences between the groups, varying volumes of solution and air space in the dialysis tubing, varying initial temperatures and volumes of water surrounding the dialysis cell, and variations in the region of the graph selected to find rate of osmotic change.

9. Increasing the temperature would speed up the collisions of water molecules with the membrane tubing thereby increasing the rate of osmosis while lowering the temperature would slow the osmotic rate of change. In humans, temperature remains constant at 37°C so, as in this example, change will be due to variations in solution concentration.

10. By using a less insulated container more heat energy will be lost to the environment. This cooling of the water surrounding the dialysis cell will slow the rate of osmotic pressure change. However, there may be a trade-off between the doubling of the volume of water surrounding the cell and the cooling due to a material of lesser insulating ability.

ANSWERS TO EXTENSION QUESTIONS

1. An animal cell will swell and maybe burst when water moves into it. A plant cell has a cell wall that prevents the cell from bursting.
2. The two factors that affect water potential are solute potential and pressure potential.
3. Water will move out of the plant cell because water always moves from an area of higher water potential to an area of lower water potential.

FURTHER EXTENSIONS

Some other possible extensions include:

1. Design and perform an experiment to determine how table salt (NaCl) affects the rate of osmosis.
2. When Hannibal conquered Carthage, his soldiers salted the fields. Using your data, what effect do you think this had on plants? Explain.
3. When one makes beef jerky, strips of fresh beef are covered with salt for many hours. What effect would this have in the process?

Respiration of Sugars by Yeast

Yeast are able to metabolize some foods, but not others. In order for an organism to make use of a potential source of food, it must be capable of transporting the food into its cells. It must also have the proper enzymes capable of breaking the food's chemical bonds in a useful way. Sugars are vital to all living organisms. Yeast are capable of using some, but not all sugars as a food source. Yeast can metabolize sugar in two ways, *aerobically,* with the aid of oxygen, or *anaerobically*, without oxygen.

In this lab, you will try to determine whether yeast are capable of metabolizing a variety of sugars. When yeast respire aerobically, oxygen gas is consumed and carbon dioxide, CO_2, is produced. You will use a CO_2 Gas Sensor to monitor the production of carbon dioxide as yeast respire using different sugars. The four sugars that will be tested are glucose (blood sugar), sucrose (table sugar), fructose (fruit sugar), and lactose (milk sugar).

OBJECTIVES

In this experiment, you will

- Use a CO_2 Gas Sensor to measure concentrations of carbon dioxide.
- Determine the rate of respiration by yeast while using different sugars.
- Determine which sugars can be used as a food source by yeast.

Figure 1

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier CO_2 Gas Sensor
250 mL respiration chamber
5% glucose, sucrose, lactose, and fructose sugar solutions
600 mL beaker (for water bath)
Beral pipettes
hot and cold water
thermometer
four 10 × 100 mm test tube
yeast suspension

PROCEDURE

1. Prepare a water bath for the yeast. A water bath is simply a large beaker of water at a certain temperature. This ensures that the yeast will remain at a constant and controlled temperature. To prepare the water bath, obtain some warm and cool water from your teacher. Combine the warm and cool water in the 600 mL beaker until it reaches 38–40°C. The beaker should be filled with about 300–400 mL water. Leave the thermometer in the water bath during the course of the experiment to monitor the temperature of the water bath.

2. Obtain five test tubes and label them G, S, F, L, and W.

3. Obtain the four sugar solutions: glucose, sucrose, fructose, and lactose.
   a. Place 2 mL of the glucose solution in test tube G.
   b. Place 2 mL of the sucrose solution in test tube S.
   c. Place 2 mL of the fructose solution in test tube F.
   d. Place 2 mL of the lactose solution in test tube L.
   e. Place 2 mL of distilled water in test tube W.

4. Obtain the yeast suspension. Gently swirl the yeast suspension to mix the yeast that settles to the bottom. Put 2 mL of yeast into each of the five test tubes. Gently swirl each test tube to mix the yeast into the solution.

5. Set the five test tubes into the water bath.

6. Incubate the test tubes for 10 minutes in the water bath. Keep the temperature of the water bath constant. If you need to add more hot or cold water, first remove as much water as you will add, or the beaker may overflow. Use a beral pipet to remove excess water. While the test tubes are incubating, proceed to Step 7.

7. If your sensor has a switch, set it to the Low (0–10,000 ppm) setting. Connect the CO_2 Gas Sensor to the computer interface. Prepare the computer for data collection by opening the file "06 Yeast Respiration" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

8. When incubation is finished, use a beral pipet to place 1 mL of the solution in test tube G into the 250 mL respiration chamber. Note the temperature of the water bath and record as the actual temperature in Table 1.

9. Quickly place the shaft of the CO_2 Gas Sensor in the opening of the respiration chamber.

10. Begin measuring carbon dioxide concentration by clicking ▸ Collect. Data will be collected for 4 minutes.

11. When data collection has finished, remove the CO_2 Gas Sensor from the respiration chamber. Fill the respiration chamber with water and then empty it. Make sure that all yeast have been removed. Thoroughly dry the inside of the chamber with a paper towel.

12. Determine the rate of respiration:
    a. Move the mouse pointer to the point where the data values begin to increase. Hold down the left mouse button. Drag the pointer to the end of the data and release the mouse button.
    b. Click on the Linear Fit button,  , to perform a linear regression. A floating box will appear with the formula for a best fit line.
    c. Record the slope of the line, *m*, as the rate of respiration in Table 1.
    d. Close the linear regression floating box.
    e. Share your data with the class by recording the sugar type and respiration rate on the board.
13. Move your data to a stored run. To do this, choose Store Latest Run from the Experiment menu.
14. Use a notebook or notepad to fan air across the openings in the probe shaft of the CO_2 Gas Sensor for 1 minute.
15. Repeat Steps 8–14 for the other four test tubes.

DATA

Table 1		
Sugar tested	Actual temperature (°C)	Respiration tate (ppm/min)
Glucose		
Sucrose		
Fructose		
Lactose		
Water (control)		

Table 2: Class Averages	
Sugar tested	Respiration tate (ppm/min)
Glucose	
Sucrose	
Fructose	
Lactose	
Water	

PROCESSING THE DATA

1. When all other groups have posted their results on the board, calculate the average rate of respiration for each solution tested. Record the average rate values in Table 2.

2. On Page 2 of the experiment file, make a bar graph of rate of respiration *vs*. sugar type. The rate values should be plotted on the y-axis, and the sugar type on the x-axis. Use the rate values from Table 2.

QUESTIONS

1. Considering the results of this experiment, do yeast equally utilize all sugars? Explain.

2. Hypothesize why some sugars were not metabolized while other sugars were.

3. Why do you need to incubate the yeast before you start collecting data?

4. Yeast live in many different environments. Make a list of some locations where yeast might naturally grow. Estimate the possible food sources at each of these locations.

TEACHER INFORMATION

Respiration of Sugars by Yeast

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. To prepare the yeast solution, dissolve 7 g (1 package) of dried yeast for every 100 mL of water. Incubate the suspension in 37–40°C water for at least 10 minutes.

3. After the 10 minute incubation period, transfer the yeast to dispensing tubes. Each group will need about 12 mL of yeast.

4. To prepare the 5% sugar solutions, add 5 g of sugar per 100 mL of solution.

5. The stopper included with the older-style CO_2 Gas Sensor is slit to allow easy application and removal from the probe. When students are placing the probe in the respiration chamber, they should gently twist the stopper into the chamber opening. Warn the students not to twist the probe shaft or they may damage the sensing unit.

6. The CO_2 Gas Sensor relies on the diffusion of gases into the probe shaft. Students should allow a couple of minutes between trials so that gases from the previous trial will have exited the probe shaft. Alternatively, the students can use a firm object such as a book or notepad to fan air through the probe shaft. This method is used in Step 14 of the student procedure.

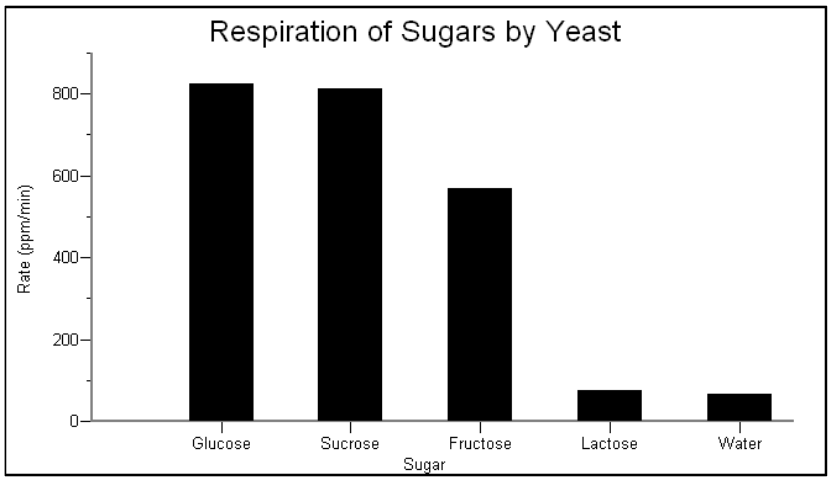
7. The stored calibration for the CO_2 Gas Sensor works very well for this experiment.

8. To conserve battery power, we suggest that AC Adapters be used to power the interfaces rather than batteries when working with the older-style CO_2 Gas Sensor.

SAMPLE RESULTS

Table 1

Sugar tested	Actual temperature (°C)	Respiration Rate (ppm/min)
Glucose	37°C	823.44
Sucrose	37°C	812.55
Fructose	37°C	568.15
Lactose	37°C	75.52
Water (control)	37°C	65.54



Rate of respiration of four sugars

ANSWERS TO QUESTIONS

1. Yeast cannot utilize all of the sugars equally well. While glucose, sucrose, and fructose can all be metabolized by yeast, lactose is not utilized at all.

2. Yeast may not have the proper enzymes to either transport lactose across its cell membrane, or it may not have the enzyme needed to convert it from a disaccharide to a monosaccharide.

3. It takes the yeast a few minutes to transport the sugar into the cell, to respire at a constant rate, and to reach the proper temperature.

4. Some yeast live on other organisms. If they are warm blooded, they may be near the optimal temperature for yeast respiration, 37°C. Many yeast live in soils. The temperature of soils may easily be measured at different times of the year.

Reflection and Absorption of Light

Would you feel cooler wearing a light or dark-colored shirt on a hot, sunny day? The color and texture of an object influences how much radiant energy from the sun it will absorb or reflect. Every color reflects a certain amount of light while absorbing the rest as heat energy. The amount of reflected light is called the color's *light reflectance value.* Dark colors with low light reflectance values tend to reflect little light while absorbing lots of heat energy, whereas light colors with high reflectance values reflect a lot of light and absorb little energy. People in warm, sunny climates are more likely to purchase light-colored cars since they don't heat up as quickly as dark-colored ones. Many house paints come with a predetermined light reflectance value to guide consumers when making color choices for their homes. Since the Earth's surface is made of many colors and textures, it is heated unevenly. Snow, ice, and clouds reflect a lot of energy back into space while green forests and vegetated lands absorb energy.

In this experiment, you will investigate the relationship between the percent reflectivity of various colors and the temperature change due to energy absorption. You will measure the amount of light reflected from paper of various colors using a Light Sensor and calculate percent reflectivity. You will also measure the temperature change of the air under the paper due to energy absorption by the paper using a Temperature Probe.

OBJECTIVES

In this experiment, you will

- Use a Light Sensor to measure the amount of reflected light.
- Calculate percent reflectivity of various colored paper.
- Use a Temperature Probe to measure the energy absorbed from light.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Light Sensor
Temperature Probe
4 cm piece of drinking straw
lamp and 150 W clear bulb
aluminum foil
white paper
black paper
2 other pieces of colored paper
ring stand
2 utility clamps
tape
ruler

Figure 1

PROCEDURE

1. Prepare the sensors for data collection.
   a. Tape the straw to the table surface as shown in Figure 1.
   b. Insert a Temperature Probe into the straw as far as it will go. Check to make sure the end of the Temperature Probe is not touching the tabletop.
   c. Place the piece of white paper over the Temperature Probe.
   d. Use a utility clamp and ring stand to fasten a Light Sensor 5 cm above a piece of colored paper as shown in Figure 2. The Light Sensor should be set on the 0–6000 lux position.
   e. Use the other utility clamp to fasten the lamp and bulb to the ring stand 10 cm above the paper.
   f. The classroom lights should be on.

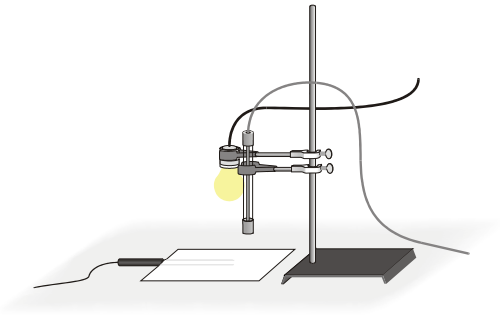


Figure 2

2. Connect the Light Sensor to Channel 1 and the Temperature Probe to Channel 2 of the Vernier computer interface.

3. Prepare the computer for data collection by opening the file "07 Reflect and Abs Light" in the *Agricultural Science with Vernier* folder.

4. Switch on the light bulb. Click Collect to begin data collection. Record the starting temperature.

5. When data collection is complete, record the final temperature. Click on the Illumination graph to select it. Click the Statistics button, , then click OK to display a Statistics box for the first run. Record the mean light reflection value (in lux). The lux is the SI unit for light illumination. Click on the Temperature graph to select it. Click the Statistics button, , then click OK. Verify the minimum and maximum readings for temperature.

6. Repeat Steps 4 and 5 for black paper and aluminum foil. If time allows, make and record readings for two additional colors of paper.

DATA

Color	White	Black	Aluminum	_____	_____
Starting Temperature (°C)					
Final Temperature (°C)					
Change in Temperature (°C)					
Reflection value (lux)					
Percent reflectivity	%	%	100 %	%	%

PROCESSING THE DATA

1. Subtract to find the change in temperature for each color paper.
2. Which color paper had the largest temperature increase?
3. Which color paper had the smallest temperature increase?
4. Solar collectors can be used to absorb the sun's radiation and change it to heat. What color would work best for solar collectors? Explain.
5. Calculate the percent reflectivity of each color paper using the relationship:

$$\% \text{ Reflectivity} = \frac{\text{reflection value for paper}}{\text{reflection value for aluminum}} \times 100$$

   Show your work in the data table above.

6. Which color paper has the highest reflectivity?
7. Which color paper has the lowest reflectivity?
8. What relationship do you see between percent reflectivity and temperature change?
9. What types of surfaces might give a planet a high reflectivity? Explain.
10. Does the planet Earth have high reflectivity? Why or why not?

EXTENSIONS

1. Design an experiment to test the reflectivity of sand, soil, water, and other materials. Perform the experiment you designed.
2. Design an experiment to test the effect of texture on reflectivity. Perform the experiment you designed.

TEACHER INFORMATION

Reflection and Absorption of Light

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. If you are using calculators for data collection, the student version of this activity contains two methods. Use Method 1 if you are collecting data with Light Sensor and an EasyTemp or a Temperature Probe connected to an EasyLink. Use Method 2 if you are collecting data with a Light Sensor and a Temperature Probe connected to a LabPro or CBL 2.

3. Heavy construction paper works well in this experiment. Try to obtain pieces with the same texture and thickness. Rectangular 10 cm x 20 cm pieces work well.

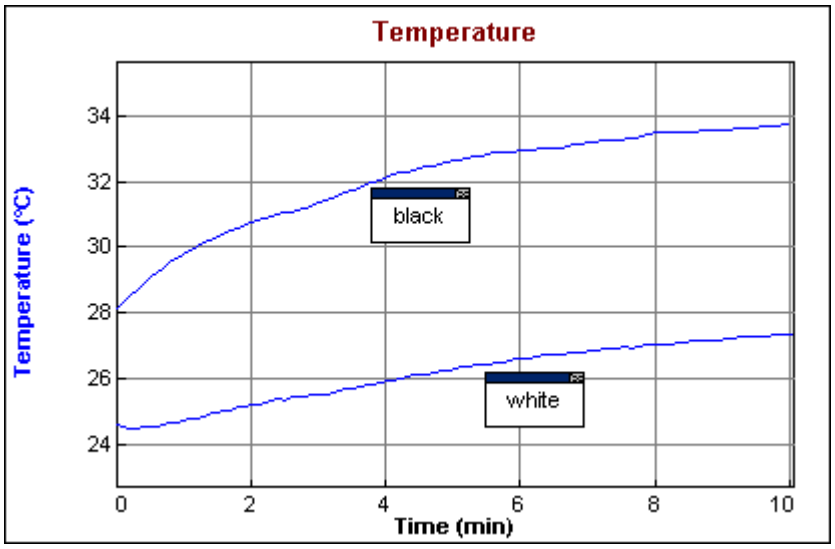
4. If you are using a TI Light Probe (TILT-BTA) for data collection, the sensor will measure Light level rather than Illumination.

5. Remind your students not to touch a hot bulb.

SAMPLE RESULTS

Color	White	Black	Aluminum	_____	_____
Starting temperature (°C)	24.6	28.2	27.4		
Final temperature (°C)	27.5	33.9	28.7		
Change in temperature (°C)	2.9	5.7	1.3		
Reflection value (lux)	2010	826	4496		
Percent reflectivity	44.7%	18.4%	100%	%	%

ANSWERS TO QUESTIONS



1. See the Sample Results.
2. Black paper had the largest temperature increase.
3. White paper had the smallest temperature increase.
4. Black would work best for a solar collector since it absorbs radiant energy best.
5. See the Sample Results.
6. White paper has the highest reflectivity.
7. Black paper has the lowest reflectivity.
8. The lower the reflectivity, the greater the temperature change.
9. Snow, ice, sand, clouds, and water would be expected to give a planet high reflectivity.
10. Planet Earth has high reflectivity because much of it is covered by snow, ice, sand, clouds, and water. The results of this experiment suggest that dark-colored parts of the Earth, such as forests and green cropland, would have lower reflectivity.

ACKNOWLEDGEMENT

We wish to thank Don Volz and Sandy Sapatka for their help in developing and testing this experiment.

Soil pH

When you think of pH, you probably think of liquid acids and bases. But soil can be acidic or basic, too. Soil pH, sometimes referred to as soil acidity, can be expressed using the *pH* scale. The pH scale ranges from 0 to 14. Soils with pH above 7 are basic or *sweet*. Soils with pH below 7 are acidic or *sour*. A soil with a pH of 7 is neither acidic nor basic, but is *neutral*.

Plant Nutrients	
Macronutrients	**Micronutrients**
Nitrogen	Iron
Phosphorus	Manganese
Potassium	Zinc
Sulfur	Copper
Calcium	Molybdenum
Magnesium	Cobalt
	Chlorine

The pH of soil is an important factor in determining which plants will grow because it controls which nutrients are available for the plants to use. Three primary plant nutrients – nitrogen, phosphorus, and potassium – are required for healthy plant growth. Because plants need them in large quantities, they are called *macronutrients*. They are the main ingredients of most fertilizers that farmers and gardeners add to their soil. Other nutrients such as iron and manganese are also needed by plants, but only in very small amounts. These nutrients are called *micronutrients*.

The availability of these nutrients depends not only on the amount but also on the form that is present, on the rate they are released from the soil, and on the pH of the soil. In general, macronutrients are more available in soil with high pH and micronutrients are more available in soil with low pH. Figure 1 shows the effect of pH on the availability of nutrients in the soil.

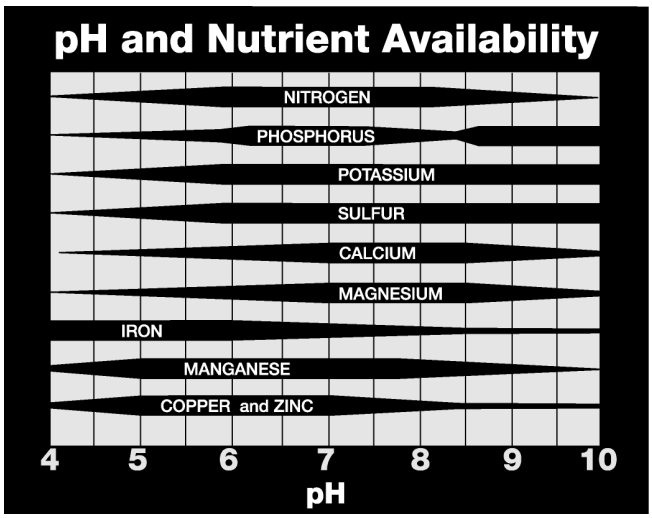


Figure 1

OBJECTIVES

In this experiment, you will

- Use a pH Sensor to measure the pH of soil samples.
- Identify any nutritional problems plants would have in that soil.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier pH Sensor
100 mL graduated cylinder
waste cup
distilled water
2 soil samples
two 250 mL beakers
wash bottle with distilled water
2 plastic spoons
paper towels

PROCEDURE

1. Prepare the water-soil mixture.
   a. Label two beakers "A" and "B".
   b. Place 50 g of Soil A into Beaker A. To avoid cross-contamination of the soils, leave this spoon in the beaker.
   c. Using a new spoon, place 50 g of Soil B into Beaker B. Leave the spoon in the beaker.
   d. Add 100 mL of distilled water to each beaker.
   e. Stir both mixtures thoroughly.
   f. Stir once every three minutes for 15 minutes.
   g. After the final stirring, let the mixtures settle for about five minutes. This allows the soil to settle out, leaving a layer of water on top for you to take your pH measurement. Continue with Steps 2–5 while you are waiting.

2. Connect the pH Sensor to the Vernier computer interface. **Important:** For this experiment your teacher already has the pH Sensor in pH soaking solution in a beaker; be careful not to tip over the beaker when connecting the sensor to the interface.

3. Prepare the computer for data collection by opening the file "08 Soil pH" from the *Agricultural Science with Vernier* folder.

4. Calibrate the pH Sensor.
   - If your teacher directs you to use the stored calibration, proceed to Step 5.
   - If your instructor directs you to perform a new calibration for the pH Sensor, follow this procedure.

   First Calibration Point

   a. Choose Calibrate ▸ CH1: pH from the Experiment menu and then click Calibrate Now.
   b. Place the sensor tip into the pH-7 buffer. Type **7** (the pH value of the buffer) in the edit box.
   c. When the displayed voltage reading for Reading 1 stabilizes, click Keep.

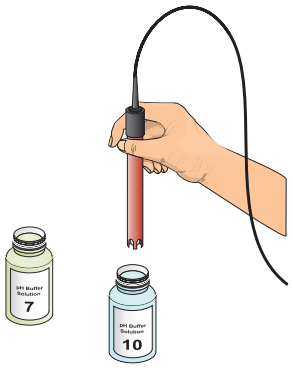


Figure 2

   Second Calibration Point

   d. Rinse the sensor with distilled water and place it in the pH-10 buffer solution.
   e. Type **10** (the pH value of the buffer) in the edit box.
   f. When the displayed voltage reading for Reading 2 stabilizes, click Keep, then click Done.

5. Measure the pH.
   a. Carefully place the tip of the pH Sensor into the liquid part of Beaker A. If your sensor has a glass bulb at the tip, make sure it is covered by the water.
   b. Note the pH reading in the meter.
   c. If the reading is stable, simply record the pH value in the data table.
6. If the reading is fluctuating, determine the *mean* (or average) value. To do this:
   a. Click Collect to begin a 10 second sampling run. **Important:** Leave the probe tip submerged for the 10 seconds that data is being collected.
   b. When the sampling run is complete, click on the Statistics button, , to display the statistics box on the graph.
   c. Record the mean pH value in your data table.
7. Rinse the pH Sensor with distilled water and repeat Steps 5 and 6 for the sample in Beaker B.
8. Rinse the pH Sensor with distilled water and return it to its storage container.

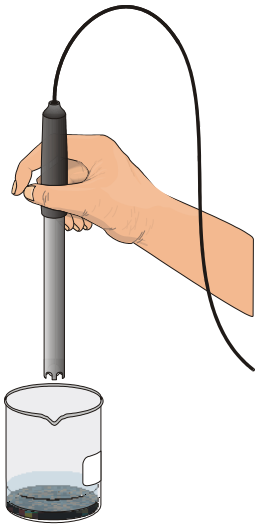


Figure 3

9. Your instructor will tell you whether you should keep the soil for further testing or clean up at this time.

DATA

	Sample A	Sample B
Soil pH		

PROCESSING THE DATA

1. Are the soils acidic, basic, or neutral?
2. Plants growing in these soils might have trouble obtaining enough of some essential nutrients. According to Figure 1, which nutrients might be in short supply for each of the soils?

EXTENSIONS

1. Research the function of each nutrient and what symptoms a plant would have if they were not getting enough.
2. Test soil samples from your backyard or another environment and compare to your first results. Are the results the same or different? Try to explain why.
3. Research how farmers adjust the pH of soils. Design and conduct an experiment to test the effectiveness of their methods.

TEACHER INFORMATION

Soil pH

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. The "pH soaking solution" used in this experiment is pH 7 buffer solution. It can be purchased from chemical supply companies. Vernier Software & Technology sells a package of capsules for preparing buffer solutions of pH 4, 7, and 10 (Order Code PHB). We recommend that you remove the pH Sensor from its storage bottle before class. If the pH Sensor is soaking in a beaker with pH soaking solution, students will have an easier time taking measurements.

3. The stored calibration for the pH Sensor works well for this experiment. If you want the most accurate results possible, you can have the students perform the 2-point calibration described in the student procedures.

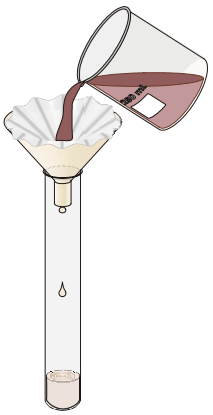
4. You can provide the soil samples, students can bring samples from home, or they can get samples from the schoolyard. Try to get samples of different types of soil to make the results more interesting. One sample could be limed as described in Item 7 to vary the pH between samples.

5. All soil samples should be dry. If you have a drying oven, allow 12 hours at 100°C or 24 hours at 80°C. If air-drying, allow at least 2–3 days depending on moisture content.

6. Water to soil ratios of 1:1, 2:1, 5:1, and even up to 10:1 are used in soil pH field testing. In general, the lower the ratio the better. However, there must be enough water to take a proper reading without harming the glass bulb on the pH electrode. A water to soil ratio of 2:1 is used in the procedure but may easily be changed using the Word files on the CD.

7. Show your students how to properly rinse a pH Sensor using a wash bottle filled with distilled water.

8. Some soil particles may float at the top of the water. This is not a problem as long as the glass bulb at the tip of the sensor is in full contact with the water. If desired, the mixture could be filtered using a funnel and filter paper as shown at right.



9. Soils that are too acidic can be adjusted to a higher pH by using a liming agent. Calcium carbonate, $CaCO_3$, magnesium carbonate, $MgCO_3$, and various oxides of calcium are common liming materials. Ground dolomitic limestone is a popular choice because it contains calcium carbonate and magnesium carbonate. You could lime some soil to vary the results or have students perform an extension to investigate this procedure.

10. The stored calibration for the pH Sensor works well for this experiment. If you want the most accurate results possible, you can have the students perform the 2-point calibration described in the student procedure.

SAMPLE RESULTS

	Sample A	Sample B
Soil pH	5.7	7.2

ANSWERS TO QUESTIONS

1. Answers will vary. For the sample data, Soil Sample A is acidic and Soil Sample B is basic.

2. Answers will vary. For the sample data:

   Soil Sample A probably isn't getting enough phosphorus and may be low in nitrogen and potassium as well. Soil Sample B is probably low in iron and manganese.

ACKNOWLEDGEMENT

We wish to thank Don Volz and Sandy Sapatka for their help in developing and testing this experiment.

Soil Salinity

Soil salinity is a measure of the saltiness of the soil. Many plants have trouble growing in soil that contains too much salt. High soil salinity makes it more difficult for plants to get water from the soil and can interfere with their obtaining the proper nutrients. The table below provides a general idea of the effect salinity has on plants.

Soil can become saline by the natural weathering of minerals, irrigation, or run-off from salted roads. Poor drainage and hot, dry weather also contribute to the build-up of salt in the soil. Sodium chloride, NaCl, is the most common salt, but others such as calcium chloride, $CaCl_2$, and magnesium sulfate, $MgSO_4$, are often present as well.

Soil salinity is determined by measuring the electrical conductivity of a soil-water mixture. The higher the salinity of the soil, the higher the conductivity of this mixture will be.

Table 1 Plant Response to Salinity

Salinity (dS/m)	Plant response
0 – 2	few problems
2 – 4	some sensitive plants have trouble
4 – 8	most plants have trouble
8 – 16	only some plants will survive
above 16	very few plants will survive

In this experiment, you will use a Conductivity Probe to measure the salinity of several soils. The unique units of soil salinity require a special note. Soil salinity is commonly reported in units of dS/m, deciSiemens per meter.

OBJECTIVES

In this experiment, you will

- Use a Conductivity Probe to measure the salinity of soil samples.
- Predict plant response to the salinity of the soil.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier Conductivity Probe
100 mL graduated cylinder
waste container
tissue
distilled water
2 soil samples
2 cups
wash bottle with distilled water
2 plastic spoons
paper towels
10 dS/m salinity standard (optional)

PROCEDURE

1. Prepare the water-soil mixture in a 2:1 ratio. Note: If your samples have already been prepared, proceed directly to Step 2.
   a. Label two beakers "A" and "B".
   b. Place 50 g of Soil A into Beaker A. To avoid cross-contamination of the soils, leave this spoon in the beaker.
   c. Using a new spoon, place 50 g of Soil B into Beaker B. Leave the spoon in the beaker.
   d. Add 100 mL of distilled water to each beaker.
   e. Stir both mixtures thoroughly.
   f. Stir once every three minutes for 15 minutes. Continue with Steps 2–4 while waiting.

2. Connect the Conductivity Probe to the Vernier computer interface. The switch on the Conductivity Probe should be on the 0–20000 µS/cm setting (equivalent to 0–20 dS/m).

3. Prepare the computer to collect data by opening the file "09 Soil Salinity" from the *Agricultural Science with Vernier* folder.

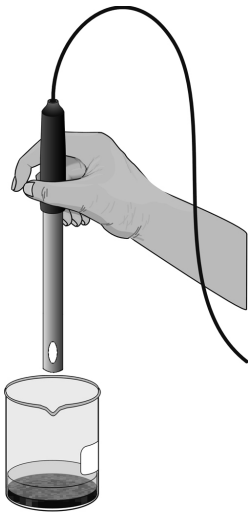
4. You are now ready to calibrate the Conductivity Probe.
   - If your teacher directs you to use the stored calibration, proceed directly to Step 5.
   - If your instructor directs you to perform a new calibration for the Conductivity Probe, follow this procedure:

   First Calibration Point

   a. Choose Calibrate ▸ CH1: Conductivity (dS/m) from the Experiment menu and then click Calibrate Now.
   b. Perform the first calibration point by placing the Conductivity Probe into distilled water. The hole near the tip of the probe should be covered completely.
   c. Type **0** (the salinity value) in the edit box.
   d. When the displayed voltage reading for Reading 1 stabilizes, click Keep.

   Second Calibration Point

   e. Place the Conductivity Probe into the 10 dS/m salinity standard solution. The hole near the tip of the probe should be covered completely.
   f. Type **10** (the salinity value) in the edit box.
   g. When the displayed voltage reading for Reading 2 stabilizes, click Keep, then click Done.
   h. Rinse the electrode with distilled water and gently blot it dry with a tissue.



5. You are now ready to collect salinity data.
   a. Place the tip of the electrode into Sample A. The hole near the tip of the probe should be completely covered by the water-soil mixture.
   b. Monitor the salinity value in the meter.
   c. When stable, record the salinity in your data table.
   d. Rinse the Conductivity Probe with distilled water.

6. Repeat Step 5 for Sample B.

DATA

	Sample A	Sample B
Soil Salinity (dS/m)		

PROCESSING THE DATA

1. Describe two ways in which soil can become saline.
2. According to Table 1, how would plants respond to each of your soil samples?

EXTENSIONS

1. Test soil samples from your backyard or another environment and compare to your first results. Are the results the same or different? Try to explain why.
2. Design and conduct an experiment to study the effect of soil salinity on plants.

TEACHER INFORMATION

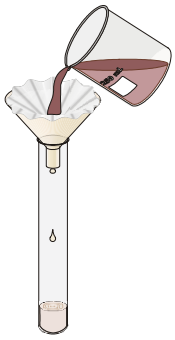
Soil Salinity

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. If your students will be testing for soil pH as well as salinity, you may want them to use the same samples for both. If your class periods are long enough, they may be able to perform both tests on the same day. If not, the beakers of soil and water should be covered to prevent evaporation.

3. You can provide the soil samples, students can bring samples from home, or they can get samples from the schoolyard. Try to get samples of different types of soil to make the results more interesting. One sample could be spiked with fertilizer or salt for variety.

4. The standard method of determining soil salinity is to make a saturated paste of soil and water, then extract the water using vacuum filtration. The EC_e, electrical conductivity of the extract, is then determined using a conductivity meter. However, water to soil ratios of 2:1 and even 5:1 are sometimes used in the field and are used in this lab for simplicity. The soil-water mixture can be filtered if desired (as shown), but the results of the filtered compared to the unfiltered do not vary significantly.



5. All soil samples should be dry. If you have a drying oven, allow 12 hours at 100°C or 24 hours at 80°C. If air-drying, allow at least 2–3 days depending on moisture content.

6. The salinity of soil is commonly reported in units of decisiemens per meter, dS/m.

7. The experiment file students are instructed to open already has a stored calibration for salinity in decisiemens per meter, dS/m. The stored calibration works well for this experiment. If the best accuracy is desired, however, a 2-point calibration can be performed using distilled water and a 10 dS/m standard. To prepare the 10 dS/m standard, add 4.60 g of NaCl to enough distilled water to prepare 1 liter of solution.

SAMPLE RESULTS

	Sample A	Sample B
Soil salinity (dS/m)	0.9	4.1

ANSWERS TO QUESTIONS

1. Soil can become saline by the natural weathering of minerals, irrigation, or run-off from salted roads.

2. Answers will vary. For the sample data, Soil Sample A would have no problems. Most plants would have some trouble with Sample B.

Soil Temperature

How do flowers and other plants know when to start growing in the spring? How do farmers know when it is safe to plant their crops? Soil temperature plays an important role in both of these decisions. Each spring, soil is heated from above by warmer air and by solar radiation. Once the soil reaches a certain temperature, it is time to plant and grow.

Soil temperature changes more slowly than the air temperature, so there is always a lag time between the extremes of air temperatures and soil temperatures. Because of daily temperature fluctuations, the soil could be cooler than the air in the daytime and warmer than the air in the nighttime.

Soil temperatures also change with depth. The deeper the soil, the more constant the temperature will be. Because of this, when referring to soil temperatures, the depth at which the measurements were taken is also important. Figure 1 shows the average soil temperatures across the United States at a depth of 4 inches. This is the depth used by the U.S. Department of Agriculture (USDA) and the National Oceanographic and Atmospheric Administration (NOAA) in their *Weekly Weather and Crop Bulletin*. This particular figure shows data from April 2002. If you look carefully, you can see the isotherms indicating the regions where various crops such as wheat and corn can develop.

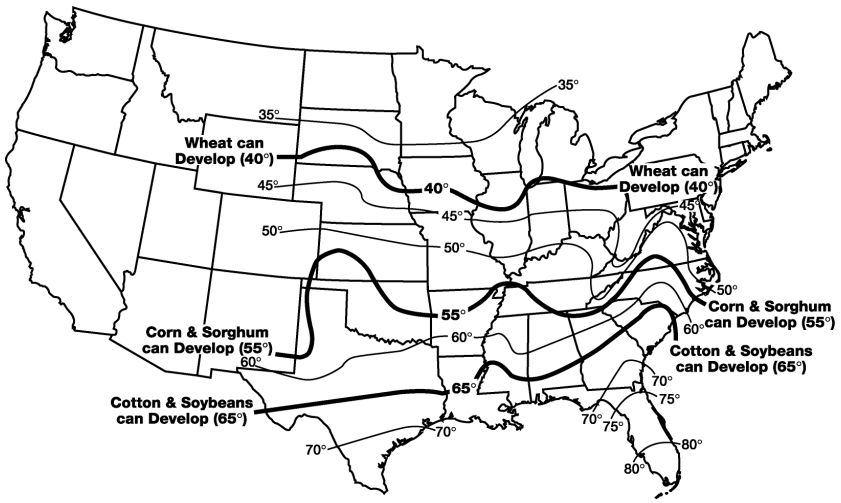


Figure 1: Soil temperatures at a depth of four inches.

In this experiment, you will use Temperature Probes to monitor the soil temperature at three different depths. A lamp and a bowl of ice will be used to simulate day and night over a two-day period. You will observe how soil temperatures vary at different depths and the timing of these variations.

OBJECTIVES

In this experiment, you will

- Simulate temperature changes over a two-day period.
- Use Temperature Probes to measure the temperature of soils at different depths.
- Explain your results.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
3 Temperature Probes
tape
plastic milk jug containing soil
bowl
lamp
ruler
ice

PROCEDURE

1. Connect the three Temperature Probes to Channels 1–3 of the Vernier computer interface.

2. Prepare the computer for data collection by opening the file "10 Soil Temperature" from the *Agricultural Science with Vernier* folder.

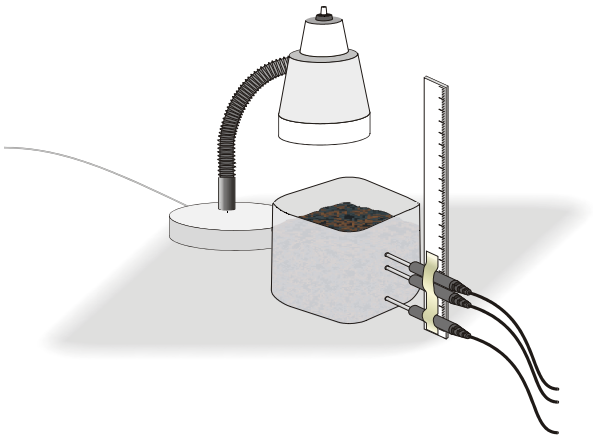


Figure 2

3. A plastic milk jug has already been prepared with soil. On one side, you should find three small holes, at 1 cm, 3 cm and 7 cm below the soil surface.
   a. Insert Probe 1 (the Probe in Channel 1) into the hole that is 1 cm below the soil surface. Push the probe in far enough so that the tip of the probe is in the center of the jug.
   b. Insert Probe 2 the same distance into the hole that is 3 cm below the soil surface.
   c. Insert Probe 3 the same distance into the hole that is 7 cm below the soil surface.

4. The Temperature Probes must be parallel to the table during data collection. Secure them in this position by taping them to a ruler as shown in Figure 2.

5. Position the lamp so that the bulb is between 5 and 10 cm from the soil surface. Do NOT turn it on yet! Once it is in position, move it slightly off to the side to make room for the bowl of ice to be placed on the soil. Later, when you are instructed to turn on the lamp, move it back over the soil.

6. Fill the bowl with ice.

7. When everything is ready, place the bowl of ice on the surface of the soil as shown in Figure 3 and click Collect to begin data collection.

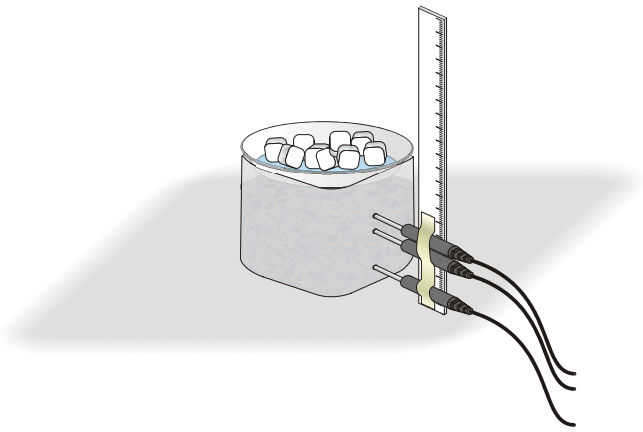


Figure 3

8. Once every five minutes, you will need to change the setup. These changes will simulate the temperature changes over a two-day period. Watch the time in the meter and use the chart below to make your changes.

Time (minutes)	Change to Setup	Time of Day (simulated)
0	Place bowl of ice on soil	Nighttime
5	Remove ice and position lamp above soil (do not turn lamp on)	Morning
10	Turn on lamp	Daytime
15	Turn off lamp and move it aside	Evening
20	Place bowl of ice on soil	Nighttime
25	Remove ice and position lamp above soil (do not turn lamp on)	Morning
30	Turn on lamp	Daytime
35	Turn off lamp and move it aside	Evening
40	Data collection will stop	

9. Data collection will stop after 40 minutes.

10. Autoscale your graph by clicking the Autoscale button, , on the toolbar.

11. Analyze your data to determine the temperature changes.

    a. Click the Statistics button, , and select all three Temperature Probes.
    b. Click OK.

c. Find the minimum and maximum temperatures for each sensor and record them in the data table. **Note:** You may need to move the Statistics boxes around so that they are all visible and it is clear which one is associated with which line.
d. Subtract to find the change in temperature for each sensor and record them in your data table.

12. Print or sketch your graph according to your teacher's instructions.

DATA

	1 cm depth	3 cm depth	7 cm depth
Maximum temperature (°C)			
Minimum temperature (°C)			
Change in temperature (°C)			

PROCESSING THE DATA

1. Study your graph. Describe the shapes of the three lines. Refer to the lines as the 1 cm line, the 3 cm line, and the 7 cm line, indicating their depth beneath the soil surface.

2. Propose an explanation for why the three lines have different shapes.

3. Study the timing of the temperature changes.
   a. Did the rising and falling temperatures reach their peaks and valleys at the same time?
   b. How long after the light was turned off did the 1 cm line reach its first temperature peak?
   c. How long after the 1 cm line reached its first peak did the 3 cm line reach its peak?

4. Propose an explanation for your answers to Question 3.

EXTENSIONS

1. Move the experiment outside and measure temperatures over longer periods of time. Describe how the results compare to the simulated exercise in class.

2. Explain how a blanket of snow could actually protect plants in the soil from freezing.

TEACHER INFORMATION

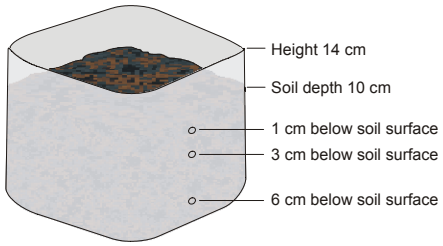
Soil Temperature

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. This experiment calls for three Temperature Probes. This may mean that students need to combine into larger groups or that some of the groups perform the experiment one day and the rest of the class perform it the next.

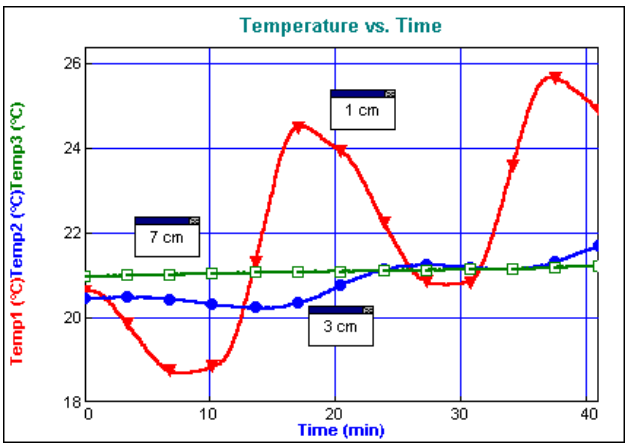
3. If you are using calculators for data collection, this experiment can be performed with calculators from the TI-83 Plus or TI-84 Plus families and a LabPro or CBL 2. Ideally it is not performed with Easy products because all runs of data should be collected at the same time under similar conditions.

4. The soil container should be made up ahead of time. Make one per group. To make the soil container,
   a. Cut the top off of a plastic milk jug so that the remainder is 14 cm high.
   b. Using an awl (or a tough ball-point pen), poke three holes in the side of the jug. The holes should be large enough for the Temperature Probe to easily fit and lined up vertically at 1, 3, and, 7 cm below the surface of the soil.
   c. Fill the jug with soil 10 cm deep.



SAMPLE RESULTS

	1 cm depth	3 cm depth	7 cm depth
Maximum temperature (°C)	25.72	21.73	21.25
Minimum temperature (°C)	18.72	20.26	21.00
Change in temperature (°C)	7.00	1.47	0.25



ANSWERS TO QUESTIONS

1. The 1 cm line went up in temperature when the light was turned on and went down when the ice was applied. Of the three, it had the largest temperature swings. The 3 cm line also went up and down in temperature, but there was a time lag and it had smaller temperature swings. The 7 cm line stayed fairly flat.

2. The closer to the surface the measurement was taken, the larger the temperature swing. This is because the change in temperature was always applied from the top. Therefore, the soil closest to the top is most affected.

3. a. No, the temperature peaks and valleys occurred at different times.
   b. Answers will vary. For the sample data, the first peak in the 1 cm line came 2.6 minutes after the light was turned off.
   c. Answers will vary. For the sample data, the lag time between 1 cm and 3 cm was approximately 9 minutes.

4. As the soil is warmed by the lamp or cooled by the ice, that change in temperature takes time to move through the soil. The heat from the lamp may only take a short time to reach 1 cm into the soil, but it will take several minutes to reach the soil to the 3 cm line.

Soil Moisture

Dry soil is made up of minerals, organic material, and air pockets, called *pore spaces*. In well-aerated soils, a typical volumetric ratio would be 55% solids and 45% pore space. As water is added to the soil, the pore spaces begin to fill with water. Soil that seems damp to the touch might have 55% solids, 35% pore space and 10% water. This would be an example of 10% volumetric water content. The maximum water content in this scenario is 45% because at that value, all the available pore space has been filled with water. This soil is referred to as being saturated, because at 45% volumetric water content, the soil can hold no more water.

Over time, soil moisture changes as soils collect, store, and release water. Collection occurs as water enters the soil through surface pores in a process called permiation. When forces of retention within soil are greater than removal forces, water storage is possible. Water release takes place when plants uptake water, evaporation occurs, or gravitational forces overcome retention.

Many factors influence the rate at which water will permeate a soil and how quickly soil moisture is lost, including surface cover, environmental factors such as relative humidity and air movement, and the characteristics of the soil itself. In this activity, you will compare two types of soil to determine how soil particle size influences soil moisture over time.

OBJECTIVES

In this experiment, you will

- Learn the technique for measuring soil moisture using a Soil Moisture Sensor.
- Determine the volumetric soil water content of a soil sample.
- Examine how soil particle size influences soil moisture.

MATERIALS

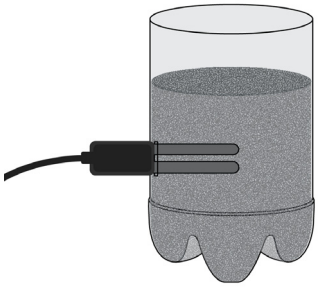
computer
Vernier computer interface
Logger*Pro*
Vernier Soil Moisture Sensor
soil sample
plastic container
water

PROCEDURE

1. Connect a Soil Moisture Sensor and the data-collection interface. Prepare the computer for data collection by opening the file "11 Soil Moisture" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

2. Obtain the soil sample assigned by your instructor. Record your soil type in the data table.

3. Position the Soil Moisture Sensor.
   a. Insert the Soil Moisture Sensor into the slot in the side of the container. Ensure that the surface of the soil is 5.0 cm above the top edge of the sensor. **Note:** The long axis of the sensor should be placed horizontally, with the short axis or "blades" oriented vertically as shown in the figure.
   b. Press down on the soil along either side of the sensor with your fingers. Continue to compact the soil around the sensor by pressing down on the soil with your fingers until you have made at least five passes along the sensor. This step is important, as the soil adjacent to the sensor surface has the strongest influence on the sensor reading.



4. Click ▸ Collect to start data collection.
5. Calculate the amount of water required to be equivalent to 1 cm of rain falling on the soil sample.
6. After 3 hours, add the amount of water that you calculated. In your data table, record the amount of time that has elapsed since data collection began.
7. Leave the setup undisturbed for the remainder of data collection (7 days).

DATA TABLE

Table 1: Your Data	
Soil type (e.g. clay loam)	
Initial soil moisture (%)	
Elapsed time when water was added (hr)	
Elapsed time when water reached sensor (hr)	
Time for water to reach sensor (hr)	
Rate of permeation (cm/hr)	
Maximum soil moisture (%)	
Elapsed time when maximum saturation was reached (hr)	
Time required to reach maximum saturation (hr)	
Final soil moisture (%)	

Table 2: Class Results		
Soil type		
Average permeation rate (cm/hr)		

DATA ANALYSIS

1. Use the graph to determine the initial, maximum, and final soil moisture. Record the values in your data table.
2. Use the graph to determine the amount of time that had elapsed when the water reached the Soil Moisture Sensor and record the value.
3. Calculate and record the time it took for the water to permeate to the level of the Soil Moisture Sensor.

   time water reached sensor – time water was added = time for water to reach sensor

4. Calculate and record the rate at which water permeated the soil.
5. Calculate and record the time it took for maximum saturation to be reached.

   time maximum saturation was recorded – time water was added
   = time for maximum saturation to be reached

QUESTIONS

1. Which type of soil had the fastest permeability rate?
2. Describe how soil particle size influences the rate at which water permeates soil.
3. Which soil had the highest maximum soil moisture?
4. Which soil reached maximum soil moisture more quickly? Explain why this is the case.
5. How does soil particle size affect the rate at which soil will lose soil moisture?
6. How is soil moisture important to plant growth and production?

EXTENSIONS

1. How is soil moisture influenced by soil temperature?
2. How is soil moisture influenced by environmental factors such as the relative humidity of the air?
3. How does compost, mulch, or the presence of foliage influence soil moisture?
4. Do transpiration and soil moisture affect each other?

Experiment
11

Soil Moisture

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. If you are using calculators for data collection, this activity can be performed with calculators from the TI-83 Plus or TI-84 Plus families and a LabPro or CBL 2. It cannot be performed with Easy products because data are collected over a 7-day period.

3. The procedure directs students to position the Soil Moisture Sensor so that the top edge is 5.0 cm below the surface of the soil. To accommodate the sensor, each group will need enough soil to fill their container to a depth of approximately 12 cm.

4. Any container that can accommodate a 12 cm deep soil sample will work for this experiment. One option is to use 2 L soda bottles. Cut off the top part o the bottle. Using a box knife, cut a small vertical slot in the side of the container into which the Soil Moisture Sensor can be inserted. The slot should be located so that there is room for at least 5 cm of soil above and below the sensor.

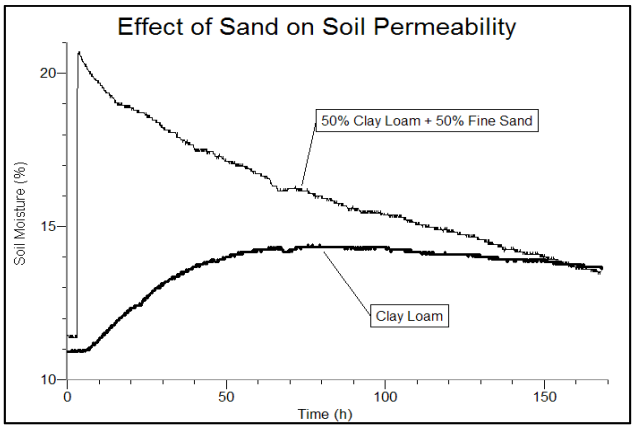
5. Make available at least two types of soil and assign different soils to different groups. Sandy loam and a soil that is a mixture of 50% sandy loam and 50% fine sand make a good pair for comparison.

6. Start with dry soil. It does not need to be dried in an oven but should be sufficiently dry to show a change in soil moisture after water is added.

7. To reduce the variability of environmental factors such as temperature, wind, and relative humidity when students are comparing their results, soil samples should be stored under similar conditions throughout the entire data-collection period.

8. If you are collecting data with calculators, EasyData will automatically enter long-term data-collection mode because of the length of data collection. Follow the directions in the student version of the lab and on the calculator screen to collect and retrieve the data. We recommend keeping the calculator connected to the data-collection interface while data are being collected. If you choose to disconnect the calculator, ensure that you keep track of which calculator is connected to each data-collection interface as the calculators must be re-united with the same data-collection interface to retrieve data.

SAMPLE RESULTS



Seven day study of soil moisture

Soil type	clay loam	50% clay loam + 50% fine sand
Initial soil moisture (%)	10.9	11.4
Elapsed time when water was added (hr)	3	3
Elapsed time when water reached sensor (hr)	6.7	3.1
Time for water to reach sensor (hr)	3.7	0.1
Rate of permeation (cm/hr)	1.2	45
Maximum soil moisture (%)	14.4	20.7
Elapsed time when maximum saturation was reached (hr)	75.7	3.6
Time required to reach maximum saturation (hr)	72.7	0.6
Final soil moisture (%)	13.7	13.5

ANSWERS TO QUESTIONS

1. The permeation rate was much higher in the soil that was a mixture of 50% clay loam and 50% fine sand.
2. Water permeates sand-rich soils (large particles) much faster than clay-rich soils (small particles).
3. The soil that was a mixture of 50% clay loam and 50% fine sand had a higher soil moisture.
4. The soil that was a mixture of 50% clay loam and 50% fine sand reached maximum soil moisture more quickly because the water was not absorbed as much by the soil because it has greater mineral content.
5. The results show that sand-rich soil loses water faster than clay-rich soil.
6. Plants become stressed without enough available water and will not produce as well.

Computer
12A

Photosynthesis and Respiration

Plants make sugar, storing the energy of the sun into chemical energy, by the process of photosynthesis. When they require energy, they can tap the stored energy in sugar by a process called cellular respiration.

The process of photosynthesis involves the use of light energy to convert carbon dioxide and water into sugar, oxygen, and other organic compounds. This process is often summarized by the following reaction:

$$6\ H_2O + 6\ CO_2 + \text{light energy} \rightarrow C_6H_{12}O_6 + 6\ O_2$$

Cellular respiration refers to the process of converting the chemical energy of organic molecules into a form immediately usable by organisms. Glucose may be oxidized completely if sufficient oxygen is available by the following equation:

$$C_6H_{12}O_6 + 6\ O_2 \rightarrow 6\ H_2O + 6\ CO_2 + \text{energy}$$

All organisms, including plants and animals, oxidize glucose for energy. Often, this energy is used to convert ADP and phosphate into ATP.

OBJECTIVES

In this experiment, you will

- Use a CO_2 Gas Sensor to measure the amount of carbon dioxide consumed or produced by a plant during respiration and photosynthesis.
- Determine the rate of respiration and photosynthesis of a plant.

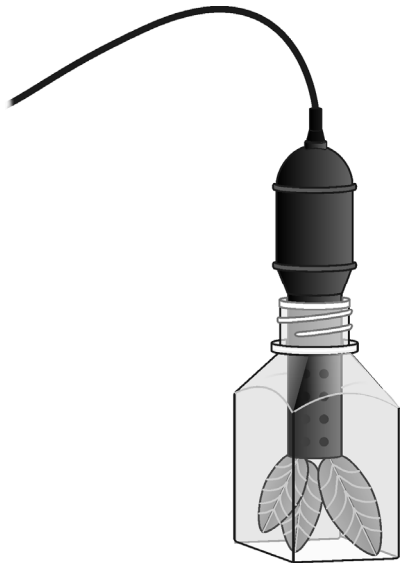


Figure 1

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier CO_2 Gas Sensor
aluminum foil
250 mL respiration chamber
plant leaves
500 mL tissue culture flask
lamp
forceps

PROCEDURE

1. If your sensor has a switch, set it to the Low (0-10,000 ppm) setting. Connect the CO_2 Gas Sensor to the Vernier interface.
2. Prepare the computer for data collection by opening the file "12A Photosyn-Resp (CO2)" from the *Agricultural Science with Vernier* folder of Logger*Pro*.
3. Obtain several leaves from the resource table and blot them dry, if damp, between two pieces of paper towel.
4. Place the leaves into the respiration chamber, using forceps if necessary. Wrap the respiration chamber in aluminum foil so that no light reaches the leaves.
5. Place the CO_2 Gas Sensor into the bottle as shown in Figure 1. Wait 3 minutes before proceeding to Step 6.
6. Click Collect to begin data collection. Data will be collected for 10 minutes.
7. When data collection has finished, determine the rate of respiration:
   a. Move the mouse pointer to the point where the data values begin to increase. Hold down the left mouse button. Drag the pointer to the point where the data ceases to rise and release the mouse button.
   b. Click on the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
   c. Record the slope of the line, *m*, as the rate of respiration in Table 1.
   d. Close the linear regression floating box.
8. Move your data to a stored run. To do this, choose Store Latest Run from the Experiment menu.
9. Remove the aluminum foil from around the respiration chamber.
10. Fill the tissue culture flask with water (not the respiration chamber) and place it between the lamp and the respiration chamber. The flask will act as a heat shield to protect the plant leaves.
11. Turn the lamp on. Place the lamp as close to the leaves as reasonable. Do not let the lamp touch the tissue culture flask. Note the time. The lamp should be on for 3 minutes prior to beginning data collection.
12. After the three-minute time period is up, click Collect to begin data collection. Data will be collected for 10 minutes.

13. When data collection has finished, determine the rate of photosynthesis:
    a. Move the mouse pointer to the point where the data values begin to decrease. Hold down the left mouse button. Drag the pointer to the point where the data ceases to decline and release the mouse button.
    b. Click the Linear Fit button, , to perform a linear regression. Choose "Latest: CO2" and a floating box will appear with the formula for a best fit line.
    c. Record the slope of the line, *m*, as the rate of photosynthesis in Table 1.
    d. Close the linear regression floating box.
14. Print a graph showing your photosynthesis and respiration data.
    a. Label each curve by choosing Text Annotation from the Insert menu. Enter "Photosynthesis" in the edit box. Repeat to create an annotation for the "Respiration" data. Drag each box to a position near its respective curve.
    b. Print a copy of the graph, with both data sets displayed. Enter your name(s) and the number of copies of the graph you want.
15. Remove the plant leaves from the respiration chamber, using forceps if necessary. Clean and dry the respiration chamber.

DATA

Table 1	
	Rate of respiration/photosynthesis (ppt/min)
In the dark	
In light	

QUESTIONS

1. Was either of the rate values positive? If so, what is the biological significance of this?
2. Was either of the rate values negative? If so, what is the biological significance of this?
3. Do you have evidence that cellular respiration occurred in leaves? Explain.
4. Do you have evidence that photosynthesis occurred in leaves? Explain.
5. List five factors that might influence the rate of Carbon Dioxide production or consumption in leaves. Explain how you think each will affect the rate?

EXTENSIONS

1. Design and perform an experiment to test one of the factors that might influence the rate of Carbon Dioxide production or consumption in Question 5.
2. Compare the rates of photosynthesis and respiration among various types of plants.

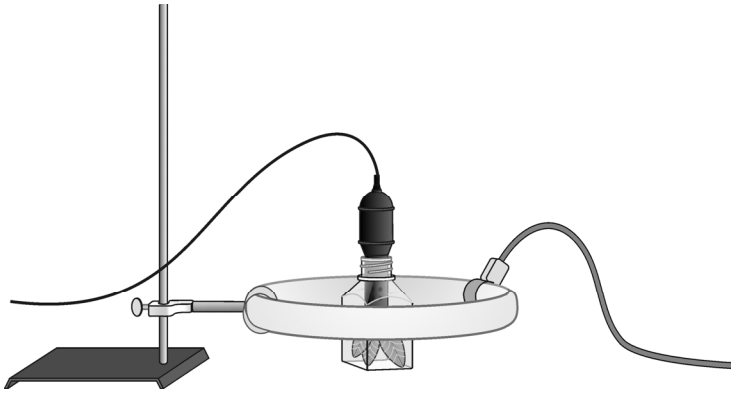
TEACHER INFORMATION

Photosynthesis and Respiration

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. Spinach leaves purchased from a grocery store work very well and are readily available any time of the year. Do not purchase the pre-packaged spinach in a bag. For best results, keep the leaves cool until they are to be used. Just before use, expose the leaves to bright light for 5 minutes.

3. A fluorescent ring lamp works very well since it bathes the plant in light from all sides and it gives off very little heat. When using a ring lamp as shown below, it is not necessary to use a heat shield.



4. If tissue culture flasks are not available, a beaker or flask of water will also work. The tissue culture flask is very thin, however, and will allow leaves to receive much more light from the same lamp.

5. On a nice, sunny day, this experiment may be performed using sun light. If so, no heat shield is needed.

6. When students are placing the probe in the respiration chamber, they should gently twist the stopper into the chamber opening. Warn the students not to twist the probe shaft or they may damage the sensor.

7. To conserve battery power, we suggest that AC Adapters be used to power the interfaces rather than batteries when working with the CO_2 Gas Sensor.

SAMPLE RESULTS

Table 1	
Leaves	Rate of respiration/photosynthesis (ppt/min)
In the dark	0.039
In light	–0.076

ANSWERS TO QUESTIONS

1. The CO_2 rate value for leaves in the dark was a positive number. The biological significance of this is that CO_2 is produced during respiration. This causes the concentration of CO_2 to increase, as sugar is oxidized and broken into CO_2, water, and energy.

2. The rate value for leaves in the light was a negative number. The biological significance of this is that CO_2 is consumed during photosynthesis. This causes the concentration of CO_2 to decrease, as the CO_2 is converted into glucose.

3. Yes, cellular respiration occurred in leaves, since CO_2 increased when leaves were in the dark and photosynthesis was not possible.

4. Yes, photosynthesis occurred in leaves, since CO_2 decreased when leaves were exposed to light.

5. Answers may vary. They might include:

   a. A greater number of leaves should increase the rate, since there are more chloroplasts to undergo photosynthesis and more cells to require energy through cellular respiration.
   b. A greater light intensity will increase the rate of photosynthesis. It may not affect the rate of cellular respiration, however.
   c. A cooler room may decrease both rates, as cellular metabolism decreases in cooler weather.
   d. Facing the top of the leaves toward the light should increase the rate of photosynthesis, since the chloroplasts are closer to the light source.
   e. If the plants overheat due to the heat from the lamp, they may wilt and stop functioning. This will decrease all rates.
   f. If there are too many leaves, diffusion may be restricted and prevent accurate readings. This may apparently decrease both rates.
   g. If water vapor increases in the chamber, the both rates will appear to be more positive than they should be, as water vapor absorbs infrared light. Since the CO_2 Gas Sensor functions by measuring the amount of infrared light reaching a photodetector, water vapor will effect the readings.

Computer
12B

Photosynthesis and Respiration

Plants make sugar, storing the energy of the sun into chemical energy, by the process of photosynthesis. When they require energy, they can tap the stored energy in sugar by a process called cellular respiration.

The process of photosynthesis involves the use of light energy to convert carbon dioxide and water into sugar, oxygen, and other organic compounds. This process is often summarized by the following reaction:

$$6\ H_2O + 6\ CO_2 + \text{light energy} \rightarrow C_6H_{12}O_6 + 6\ O_2$$

Cellular respiration refers to the process of converting the chemical energy of organic molecules into a form immediately usable by organisms. Glucose may be oxidized completely if sufficient oxygen is available by the following equation:

$$C_6H_{12}O_6 + 6\ O_2 \rightarrow 6\ H_2O + 6\ CO_2 + \text{energy}$$

All organisms, including plants and animals, oxidize glucose for energy. Often, this energy is used to convert ADP and phosphate into ATP.

OBJECTIVES

In this experiment, you will

- Use an O_2 Gas Sensor to measure the amount of oxygen gas consumed or produced by a plant during respiration and photosynthesis.
- Determine the rate of respiration and photosynthesis of a plant.

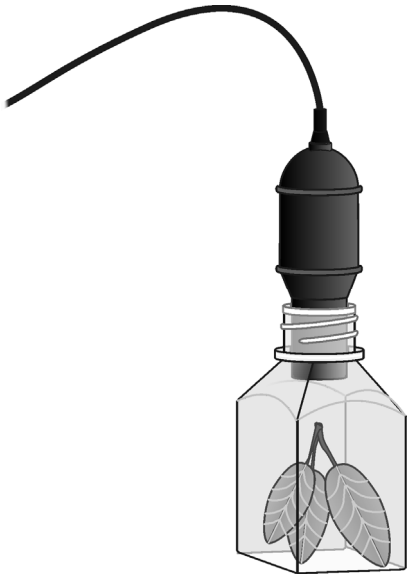


Figure 1

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier O_2 Gas Sensor
aluminum foil
250 mL respiration chamber
plant leaves
500 mL tissue culture flask
lamp
forceps

PROCEDURE

1. Connect the O_2 Gas Sensor to the Vernier interface.
2. Prepare the computer for data collection by opening the file "12B Photosyn-Resp (O2)" from the *Agricultural Science with Vernier* folder of Logger*Pro*.
3. Obtain several leaves from the resource table and blot them dry, if damp, between two pieces of paper towel.
4. Place the leaves into the respiration chamber, using forceps if necessary. Wrap the respiration chamber in aluminum foil so that no light reaches the leaves.
5. Place the O_2 Gas Sensor into the bottle as shown in Figure 1. Gently push the sensor down into the bottle until it stops. The sensor is designed to seal the bottle without the need for unnecessary force. Wait 3 minutes before proceeding to Step 6.
6. Click Collect to begin data collection. Data will be collected for 10 minutes.
7. When data collection has finished, determine the rate of respiration:
   a. Move the mouse pointer to the point where the data values begin to decrease. Hold down the left mouse button. Drag the pointer to the point where the data ceases to decline and release the mouse button.
   b. Click on the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
   c. Record the slope of the line, *m*, as the rate of respiration in Table 1.
   d. Close the linear regression floating box.
8. Move your data to a stored run. To do this, choose Store Latest Run from the Experiment menu.
9. Remove the aluminum foil from around the respiration chamber.
10. Fill the tissue culture flask with water (not the respiration chamber) and place it between the lamp and the respiration chamber. The flask will act as a heat shield to protect the plant.
11. Turn the lamp on. Place the lamp as close to the leaves as reasonable. Do not let the lamp touch the tissue culture flask. Note the time. The lamp should be on for 3 minutes prior to beginning data collection.
12. After the three-minute time period is up, click Collect to begin data collection. Data will be collected for 10 minutes.

13. When data collection has finished, determine the rate of photosynthesis:
    a. Move the mouse pointer to the point where the data values begin to increase. Hold down the left mouse button. Drag the pointer to the point where the data ceases to rise and release the mouse button.
    b. Click on the Linear Fit button, , to perform a linear regression. Choose "Latest: Oxygen Gas" and a floating box will appear with the formula for a best fit line.
    c. Record the slope of the line, *m*, as the rate of photosynthesis in Table 1.
    d. Close the linear regression floating box.
14. Print a graph showing your photosynthesis and respiration data.
    a. Label each curve by choosing Text Annotation from the Insert menu. Enter "Photosynthesis" in the edit box. Repeat to create an annotation for the "Respiration" data. Drag each box to a position near its respective curve. Adjust the position of the arrow heads.
    b. Print a copy of the graph, with both data sets displayed. Enter your name(s) and the number of copies of the graph you want.
15. Remove the plant leaves from the respiration chamber, using forceps if necessary. Clean and dry the respiration chamber.

DATA

Table 1	
	Rate of photosynthesis/respiration (ppt/min)
In the dark	
In light	

QUESTIONS

1. Was either of the rate values a positive number? If so, what is the biological significance of this?
2. Was either of the rate values negative? If so, what is the biological significance of this?
3. Do you have evidence that cellular respiration occurred in leaves? Explain.
4. Do you have evidence that photosynthesis occurred in leaves? Explain.
5. List five factors that might influence the rate of oxygen production or consumption in leaves. Explain how you think each will affect the rate?

EXTENSIONS

1. Design and perform an experiment to test one of the factors that might influence the rate of oxygen production or consumption in Question 5.
2. Compare the rates of photosynthesis and respiration among various types of plants.

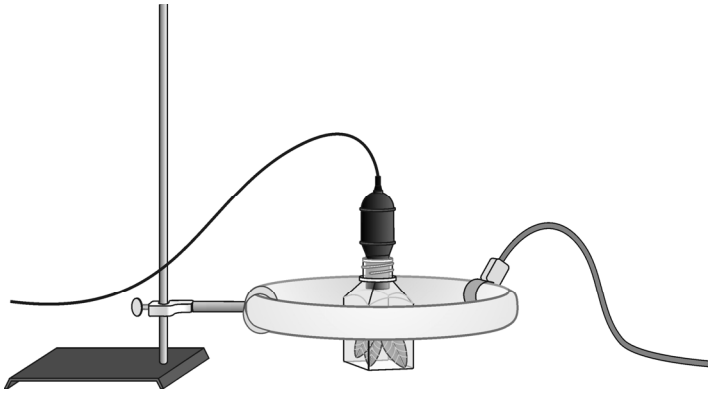
TEACHER INFORMATION

Photosynthesis and Respiration

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. Spinach leaves purchased from a grocery store work very well and are readily available any time of the year. For best results, keep the leaves cool until they are to be used. Just before use, expose the leaves to bright light for 5 minutes.

3. A fluorescent ring lamp works very well since it bathes the plant in light from all sides and it gives off very little heat. When using a ring lamp as shown below, it is not necessary to use a heat shield.



4. If tissue culture flasks are not available, a beaker or flask of water will also work as a heat shield. The tissue culture flask is very thin, however, and will allow leaves to receive much more light from the same lamp.

5. On a nice, sunny day, this experiment may be performed using sun light. If so, no heat shield is needed.

6. To extend the life of the O_2 Gas Sensor, always store the sensor upright in the box in which it was shipped.

SAMPLE RESULTS

Table 1	
Leaves	Rate of respiration/photosynthesis (ppt/min)
In the dark	–0.138
In light	0.271

ANSWERS TO QUESTIONS

1. The rate value for leaves in the light was a positive number. The biological significance of this is that O_2 is produced during photosynthesis. This causes the concentration of O_2 to increase, as the O_2 is converted into glucose.

2. The rate value for leaves in the dark was a negative number. The biological significance of this is that O_2 is consumed during cellular respiration. This causes the concentration of O_2 to decrease as glucose is oxidized for energy.

3. Yes, cellular respiration occurred in leaves, since O_2 decreased when leaves were in the dark and photosynthesis was not possible.

4. Yes, photosynthesis occurred in leaves, since O_2 increased when leaves were exposed to light.

5. Answers may vary. They might include:
   a. A greater number of leaves should increase the rate, since there are more chloroplasts to undergo photosynthesis and more cells to require energy through cellular respiration.
   b. A greater light intensity will increase the rate of photosynthesis. It may not affect the rate of cellular respiration, however.
   c. A cooler room may decrease both rates, as cellular metabolism decreases in cooler weather.
   d. Facing the top of the leaves toward the light should increase the rate of photosynthesis, since the chloroplasts are closer to the light source.
   e. If the plants overheat due to the heat from the lamp, they may wilt and stop functioning. This will decrease all rates.
   f. If there are too many leaves, diffusion may be restricted and prevent accurate readings. This may apparently decrease both rates.

Photosynthesis and Respiration

Plants make sugar, storing the energy of the sun into chemical energy, by the process of photosynthesis. When they require energy, they can tap the stored energy in sugar by a process called cellular respiration.

The process of photosynthesis involves the use of light energy to convert carbon dioxide and water into sugar, oxygen, and other organic compounds. This process is often summarized by the following reaction:

$$6\ H_2O + 6\ CO_2 + \text{light energy} \rightarrow C_6H_{12}O_6 + 6\ O_2$$

Cellular respiration refers to the process of converting the chemical energy of organic molecules into a form immediately usable by organisms. Glucose may be oxidized completely if sufficient oxygen is available by the following equation:

$$C_6H_{12}O_6 + 6\ O_2 \rightarrow 6\ H_2O + 6\ CO_2 + \text{energy}$$

All organisms, including plants and animals, oxidize glucose for energy. Often, this energy is used to convert ADP and phosphate into ATP.

OBJECTIVES

In this experiment, you will

- Use an O_2 Gas Sensor to measure the amount of oxygen gas consumed or produced by a plant during respiration and photosynthesis.
- Use a CO_2 Gas Sensor to measure the amount of carbon dioxide consumed or produced by a plant during respiration and photosynthesis.
- Determine the rate of respiration and photosynthesis of a plant.

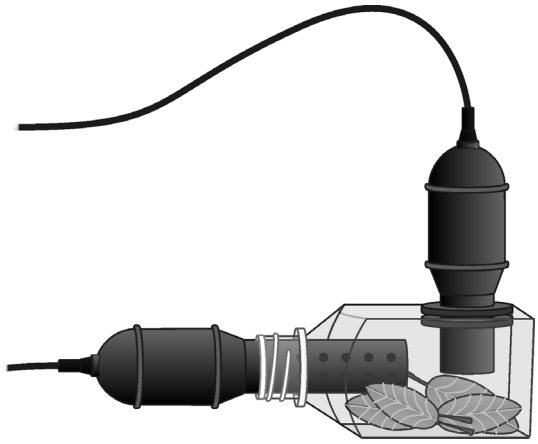


Figure 1

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier O_2 Gas Sensor
Vernier CO_2 Gas Sensor
BioChamber 250
plant leaves
500 mL tissue culture flask
lamp
aluminum foil
forceps

PROCEDURE

1. If your CO_2 Gas Sensor has a switch, set it to the Low (0–10,000 ppm) setting. Connect the CO_2 Gas Sensor to Channel 1 and the O_2 Gas to Channel 2 of the Vernier computer interface.

2. Prepare the computer for data collection by opening the file "12C Photosyn-Resp (CO2 and O2)" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

3. Obtain several leaves from the resource table and blot them dry, if damp, between two pieces of paper towel.

4. Place the leaves into the BioChamber 250, using forceps if necessary. Wrap the respiration chamber in aluminum foil so that no light reaches the leaves.

5. Place the O_2 Gas Sensor into the BioChamber 250 as shown in Figure 1. Insert the sensor snugly into the grommet. The O_2 Gas Sensor should remain vertical throughout the experiment. Place the CO_2 Gas Sensor into the neck of the respiration chamber as shown in Figure 1.Wait 10 minutes before proceeding to Step 6.

6. Click ▸ Collect to begin data collection. Collect data for fifteen minutes and click ■ Stop.

7. When data collection has finished, determine the rate of respiration:
   a. Click anywhere on the CO_2 graph. Move the mouse pointer to the point where the data values begin to increase. Hold down the left mouse button. Drag the pointer to the point where the data ceases to increase and release the mouse button.
   b. Click on the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
   c. Record the slope of the line, *m*, as the rate of respiration in Table 1.
   d. Close the linear regression floating box.
   e. Repeat Steps 7a–d for the O_2 graph. However, you will need to move the mouse pointer to the point where the data values begin to decrease. Hold down the mouse button and drag to the point where the data ceases to decrease.

8. Move your data to a stored run. To do this, choose Store Latest Run from the Experiment menu.

9. Remove the aluminum foil from around the respiration chamber.

10. Fill the tissue culture flask with water (not the respiration chamber) and place it between the lamp and the respiration chamber. The flask will act as a heat shield to protect the plant leaves.

11. Turn the lamp on. Place the lamp as close to the leaves as reasonable. Do not let the lamp touch the tissue culture flask. Note the time. The lamp should be on for 5 minutes prior to beginning data collection.

12. After the five-minute time period is up, click ▸ Collect to begin data collection. Collect data for 15 minutes and click ■ Stop.

13. When data collection has finished, determine the rate of photosynthesis:
    a. Click anywhere on the CO_2 graph. Move the mouse pointer to the point where the data values begin to decrease. Hold down the left mouse button. Drag the pointer to the point where the data ceases to decrease and release the mouse button.
    b. Click on the Linear Fit button, , to perform a linear regression. Choose "Latest CO2" and a floating box will appear with the formula for a best-fit line.
    c. Record the slope of the line, *m*, as the rate of photosynthesis in Table 1.
    d. Close the linear regression floating box.
    e. Repeat steps 13a–d for the O_2 graph. However, you will need to move the mouse pointer to the point where the data values begin to increase, hold down the mouse button and drag to the point where the data ceases to increase.

14. Print a graph showing your photosynthesis and respiration data.
    a. Label each curve by choosing Text Annotation from the Analyze menu. Enter "Photosynthesis" in the edit box. Repeat to create an annotation for the "Respiration" data. Drag each box to a position near its respective curve. Adjust the position of the arrow heads.
    b. Print a copy of the graph, with both data sets displayed. Enter your name(s) and the number of copies of the graph you want.

15. Remove the plant leaves from the respiration chamber, using forceps if necessary. Clean and dry the respiration chamber.

DATA

Table 1

	CO_2 rate of production/consumption (ppt/min)	O_2 rate of production/consumption (ppt/min)
In the dark		
In the light		

QUESTIONS

1. Was either of the rate values for CO_2 a positive number? If so, what is the biological significance of this?

2. Was either of the rate values for O_2 a negative number? If so, what is the biological significance of this?

3. Do you have evidence that cellular respiration occurred in leaves? Explain.

4. Do you have evidence that photosynthesis occurred in leaves? Explain.

5. List five factors that might influence the rate of oxygen production or consumption in leaves. Explain how you think each will affect the rate?

EXTENSIONS

1. Design and perform an experiment to test one of the factors that might influence the rate of oxygen production or consumption in Question 5.

2. Compare the rates of photosynthesis and respiration among various types of plants.

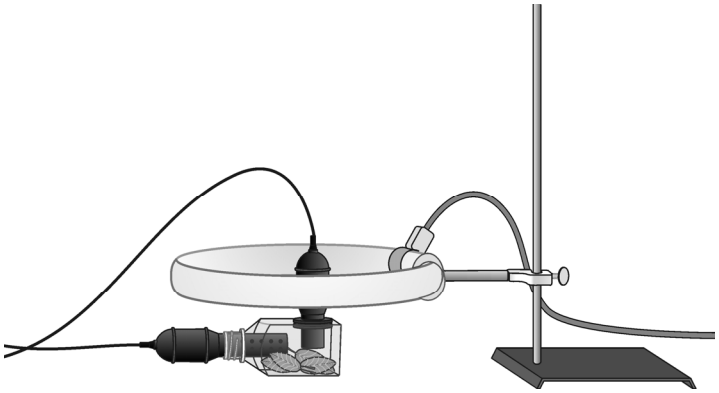
TEACHER INFORMATION

Photosynthesis and Respiration

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. Spinach leaves purchased from a grocery store work very well and are readily available any time of the year. Do not purchase the pre-packaged spinach in a bag. For best results, keep the leaves cool until they are to be used. Just before use, expose the leaves to bright light for 5 minutes.

3. A fluorescent ring lamp works very well since it bathes the plant in light from all sides and it gives off very little heat. When using a ring lamp as shown below, it is not necessary to use a heat shield.



4. If tissue culture flasks are not available, a beaker or flask of water will also work. The tissue culture flask is very thin, however, and will allow leaves to receive much more light from the same lamp.

5. On a nice, sunny day, this experiment may be performed using sun light. If so, no heat shield is needed.

6. To extend the life of the O_2 Gas Sensor, always store the sensor upright in the box in which it was shipped.

7. The waiting time before taking data may need to be adjusted depending on the rate of diffusion of the oxygen gas and the carbon dioxide gas. Monitor the gas concentrations and start collecting data when the levels of gas begin to move in the correct direction. It may take up to 15 minutes for the Oxygen Gas level to begin increasing once the light is turned on.

8 When students are placing the probe in the respiration chamber, they should gently twist the stopper into the chamber opening. Warn the students not to twist the probe shaft or they may damage the sensor.

9. To conserve battery power, we suggest that AC Adapters be used to power the interfaces rather than batteries when working with the CO_2 Gas Sensor.

SAMPLE RESULTS

Table 1		
Leaves	CO_2 rate of production/consumption (ppm/min)	O_2 rate of production/consumption (%/min)
In the dark	39.0	–0.0138
In the light	–75.6	0.0271

ANSWERS TO QUESTIONS

1. The CO_2 rate value for leaves in the dark was a positive number. The biological significance of this is that CO_2 is produced during respiration. This causes the concentration of CO_2 to increase, as sugar is oxidized and broken into CO_2, water, and energy.

2. The O_2 rate value for leaves in the dark was a negative number. The biological significance of this is that O_2 is consumed during cellular respiration. This causes the concentration of O_2 to decrease as glucose is oxidized for energy.

3. Yes, cellular respiration occurred in leaves, since O_2 decreased when leaves were in the dark and photosynthesis was not possible.

4. Yes, photosynthesis occurred in leaves, since O_2 increased when leaves were exposed to light.

5. Answers may vary. They might include:
   a. A greater number of leaves should increase the rate, since there are more chloroplasts to undergo photosynthesis and more cells to require energy through cellular respiration.
   b. A greater light intensity will increase the rate of photosynthesis. It may not affect the rate of cellular respiration, however.
   c. A cooler room may decrease both rates, as cellular metabolism decreases in cooler weather.
   d. Facing the top of the leaves toward the light should increase the rate of photosynthesis, since the chloroplasts are closer to the light source.
   e. If the plants overheat due to the heat from the lamp, they may wilt and stop functioning. This will decrease all rates.
   f. If there are too many leaves, diffusion may be restricted and prevent accurate readings. This may apparently decrease both rates.

Transpiration

Water is transported in plants, from the roots to the leaves, following a decreasing water potential gradient. *Transpiration*, or loss of water from the leaves, helps to create a lower osmotic potential in the leaf. The resulting transpirational pull is responsible for the movement of water from the xylem to the mesophyll cells into the air spaces in the leaves. The rate of evaporation of water from the air spaces of the leaf to the outside air depends on the water potential gradient between the leaf and the outside air.

Various environmental factors, including those conditions which directly influence the opening and closing of the stomata, will affect a plant's transpiration rate. This experiment will measure transpiration rates under different conditions of light, humidity, temperature, and air movement. The data will be collected by measuring pressure changes as the plant takes up water into the stem.

OBJECTIVES

In this experiment, you will

- Observe how transpiration relates to the overall process of water transport in plants.
- Use a computer interface and a Gas Pressure Sensor to measure the rate of transpiration.
- Determine the effect of light intensity, humidity, wind, and temperature on the rate of transpiration of a plant cutting.

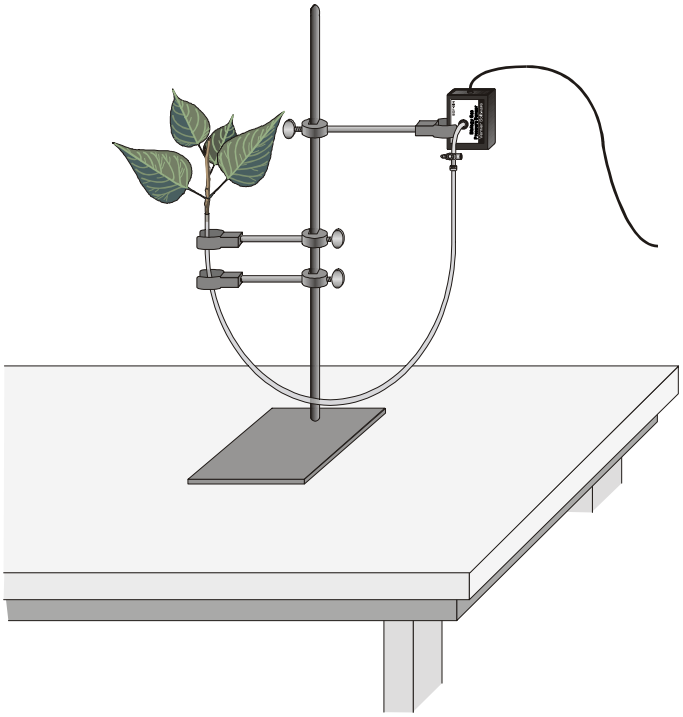


Figure 1

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier Gas Pressure Sensor
utility clamps
ring stand
plant cuttings
plastic tubing clamps
dropper or Beral pipette
razor blade or scalpel
metric ruler
masking tape
100 watt light source
plastic gallon size bag with twist tie
heater, small electric
fan with slow speed
aerosol spray container or plant mister
plastic syringe

PROCEDURE

1. Position the ring stand, utility clamps, and Gas Pressure Sensor as shown in Figure 1.

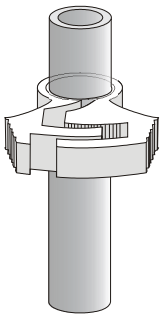


Figure 2

2. Prepare the plastic tubing.
   a. Connect the plastic syringe to one end of a 36–42 cm piece of plastic tubing.
   b. Place the other end of the tubing into water and use the syringe to draw water up into the tubing until it is full. Tap the tubing to expel any air bubbles that form inside the tube.
   c. Slip a plastic tubing clamp onto the tubing as shown in Figure 2.
   d. Bend the tubing into a U shape with both ends up. Remove the syringe, leaving the tubing full of water.

3. Select a plant that has a stem roughly the same diameter as the opening of the plastic tubing. Using a scalpel or razor blade, carefully cut the plant one inch above the soil. Place the plant under water against a hard surface and make a new cut at a 45° angle near the base of the stem.

4. Connect the plant to the tubing.
   a. The plastic tubing has a white plastic connector at one end that allows you to connect it to the valve on the Gas Pressure Sensor. Raise the end of the tubing with the connector until you see water beginning to drip out of the other end.
   b. Carefully push the cut stem of the plant down into the end of the tubing where the water is dripping out. Be careful not to allow any air bubbles to form between the cut portion of the stem and the water in the tube.
   c. Push the plant down as far as it will go without damaging the plant. At least one centimeter of the plant stem should fit into the tubing. If the stem is too large for the tubing, cut the stem at a higher point where it is smaller.
   d. Squeeze the tubing clamp shut as tight as possible as shown in Figure 3.

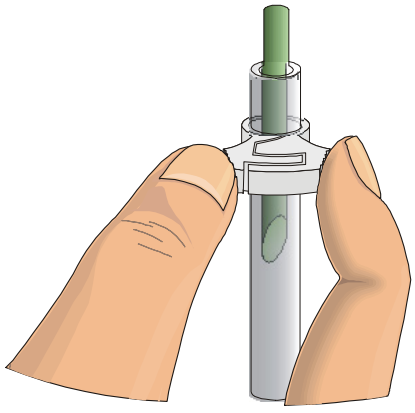


Figure 3

5. When the tubing clamp is shut tight, invert your plant cutting to check for any leaks. If water does leak out, turn the plant right side up and try tightening the clamp further.

   Important: Be sure the tubing is filled completely with water. The water column must be flush with the stem. There should be no air visible at the base of the stem. If water moves down the tube away from the stem after it has been inserted, check for a leak in the system.

6. Connect the plastic tubing to the sensor valve. **Caution**: Do not allow water to enter the valve of the Gas Pressure Sensor.

7. Secure the plant in an upright position with the utility clamps as shown in Figure 1. It should be positioned so that the cut stem is about 8 cm below the water level at the other end of the tubing, as shown in Figure 1.

8. Place a mark on the tube at the starting water level to allow you to refill the tube to the proper level in Step 17.

9. Place your plant setup in an area where the wind, humidity, and temperature are reasonably constant. This will be your control setup.

10. Allow the system 5 minutes to adjust to the environment. While the system is adjusting, set up the computer.

11. Connect the Gas Pressure Sensor to the computer interface. Prepare the computer for data collection by opening the file "13 Transpiration" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

12. Check the base of the plant stem in the water tube to make sure that no air bubbles or air pockets have formed that will prevent the plant from taking up water. If an air pocket has formed, refit the plant in the tubing before initiating data collection in Step 13.

13. After the plant has equilibrated for 5 minutes, click Collect to begin data collection. Data will be collected for 15 minutes.

14. When data collection has finished, find the rate of transpiration for your plant. To do this,

    a. Move the mouse pointer to the point where the pressure values begin to decrease. Click the mouse button and drag the pointer to the end of the data, then release the mouse button.
    b. Click the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
    c. Record the slope of the line, *m*, in Table 1 as the rate of transpiration for the control. Close the floating box.

15. (Optional) Double click anywhere on the graph and enter "Transpiration: Control" as the graph title. Print a copy of your graph. Enter your name(s) and the number of copies of the graph.

16. Design an experiment to simulate one of the following environmental factors, as assigned by your teacher:
    - the effect of light intensity
    - the effect of the wind blowing on the plant
    - the effect of humidity
    - the effect of temperature
    - the effect of another self-identified environmental variable

    Be sure to address the following questions in your design:
    - What is the essential question being addressed?
    - What assumptions are made about the system being measured?
    - Can those assumptions be easily verified?
    - Will the measurements provide the necessary data to answer the question under study?
17. After checking your procedure with your teacher, obtain the materials needed for the experiment and perform the tests. Refill the water level in the tube to the same level marked in Step 8. Record your values in Table 1.

PROCESSING THE DATA

1. Determine the surface area of all the leaves on your plant cutting by the following method:
    a. Cut all the leaves (not stems) off your plant and determine their mass using a balance.
    b. Estimate the total leaf surface area in cm^2 for your plant by cutting out a section of leaf 5 cm × 5 cm.
    c. Determine the mass for this leaf section and divide by 25 cm^2 to find the mass of 1 cm^2 of leaf.
    d. Divide the total mass of the leaves by the mass of 1 cm^2 to find the total leaf surface area.
    e. Record the calculated surface area in Table 1.
2. Calculate the rate of transpiration/surface area. To do this, divide the rate of transpiration by the surface area for each plant. These rate values can be expressed as kPa/min/cm^2. Record the rate/area in Table 1.
3. Subtract the control (rate/area) value from the experimental value. Record this adjusted rate in the last column of Table 1.
4. Record the adjusted rate for your experimental test on the board to share with the class. Record the class results in Table 2 for each of the environmental conditions tested. If a condition was tested by more than one group, take the average of the values and record in Table 2.
5. Make a bar graph that shows the effect of different environmental conditions on the transpiration of water in plant cuttings. Using the data in Table 2 plot the adjusted rate for each test on the y-axis and the test label on the x-axis.

DATA

Table 1				
Test	Slope (kPa/min)	Surface area (cm^2)	Rate/area ($kPa/min/cm^2$)	Adjusted rate ($kPa/min/cm^2$)
Experimental _____				
Control				

Table 2	
Class Data	
Test	Adjusted rate ($kPa/min/cm^2$)
Light	
Humidity	
Wind	
Temperature	

QUESTIONS

1. How was the rate of transpiration affected in each of the experimental situations as compared to the control?

2. Which variable resulted in the greatest rate of water loss? Explain why this factor might increase water loss when compared to the others.

3. What adaptations enable plants to increase or decrease water loss? How might each affect transpiration?

EXTENSIONS

1. Using a compound microscope, identify the vascular tissues of a plant stem. Describe the function of each tissue type identified.

   a. Obtain a section of stem from the plant you used during the transpiration experiment.
   b. Using a nut-and-bolt microtome, carefully cut 6 cross sections of the plant stem. The cross sections should be cut as thin as possible.
   c. Place each of the cross sections in a dish or cup of 50% ethanol solution for 5 minutes.
   d. Remove the cross sections from the alcohol and place them in a dish containing toludine blue O stain for 5 minutes.

e. Rinse the cross sections with distilled water and mount them on a microscope slide with a drop of 50% glycerin. Place a cover slip on the slide and examine the cross sections using a compound microscope.
f. On a separate sheet of paper, make a drawing of the cross sections. Identify and label the cell and tissue types described by your teacher.

2. Test cuttings from a variety of different plant species. How does each compare?

3. Count the number of stoma/cm^2 for each of the plants in Extension 1. How does this relate to the plant's ability to transpire water?

4. Design an experiment to test for the variables in Question 3.

TEACHER INFORMATION

Transpiration

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. You should leave water out overnight in a beaker or cup to allow any excess dissolved air to escape. This will ensure that no air bubbles form in the tube at the cut end of the stem. If air bubbles form, it may be necessary to restart your experiment. If bubbles do form, remove the plant and tubing from the two utility clamps and allow the plant to hang towards the ground with the other end of the tubing pointing up. Carefully tap on the sides of the tubing to loosen any bubbles—they will float to the water's surface at the other end. Once all bubbles are removed, check the plant's seal at the tube. Secure your plant in the tubing and restart the data collection.

3. There is not always an immediate change in the transpiration rate. Allow the plant to spend a few extra minutes under a particular condition before initiating data collection. This will give the plant the necessary time to adjust. When the transpiration rate changes drastically the stomata will close, decreasing the transpiration rate. If the length of data collection is extended, you will be able to see on the graph when the stomata have closed and the rate slows down.

4. Many plants work well for this experiment. Plants that have been used include tomato, strawberry, bean, geranium, cyclamen, and even honeysuckle. For best results, we recommend using plants with numerous leaves. Tomato plants work very well and have been used to collect the sample data for this activity. One possible extension of this experiment would be to have the students use different plant species under similar conditions and evaluate how different plants have adapted to prevent water loss.

5. The thick-wall plastic tubing that comes with the Gas Pressure Sensor is well suited for this lab. The inner diameter of the tubing is 3 mm and may be too small for some plant specimens. Science supply companies carry thick-wall plastic tubing, with a larger inner diameter, that will work well on larger plant stems. They also sell tubing connectors that will allow you to connect the larger tubing to the tubing provided with the Gas Pressure Sensor.

6. Emphasize to your students the importance of providing an airtight fit with all plastic-tubing connections.

7. The Vernier Barometer sensor can also be used to perform this experiment. If you already have a Barometer and wish to do this activity, you will need to order the following parts from Vernier Software & Technology:

   Pressure Sensor Accessories Kit (order code PS-ACC)

   Plastic 3-Way Pressure Sensor Valve (order code PSV)

8. The plastic tubing clamps used in the student procedure may be purchased from Vernier:

   Plastic Tubing Clamps (order code PTC: package of 100)

SAMPLE DATA

Table 1	
Test	Adjusted rate ($kPa/min/cm^2$)
Control	-2.39×10^{-3}
Light	-5.63×10^{-3}
Humidity	-0.52×10^{-3}
Wind	-0.16×10^{-3}

ANSWERS TO QUESTIONS

1. It is typically predicted that the light and wind will increase the rate of transpiration. This may not be apparent until after correction for surface area differences. Sometimes the wind, if too strong, may cause the leaves to droop or fold up, and in this case they may transpire less. Stomates may close to counter the dehydration. If this happens, discuss the nature of science experimentation, e.g., the expected may not always be the result. Usually, after correction for surface area, the high humidity plant will transpire less than a control. A student may question whether the light increased the temperature of the leaf. If the light was too close to the plant, temperature may indeed be a variable without a control.

2. Answers will vary—usually the light will produce the greatest rate of water loss. High light intensity increases water loss due to increased photosynthesis. Wind removes water vapor from the surface of the leaf more rapidly. It may increase the evaporation rate by increasing the gradient between water in the leaf air spaces and water vapor in the air.

3. Plants can increase or decrease water loss by:
   - closing the stomata during water stress.
   - reducing the number of stomata.
   - waxy cuticles.
   - fleshy, thick leaves.
   - hairy surfaces.
   - reducing the overall leaf surface area.

Computer
14A

Cell Respiration

Cell respiration refers to the process of converting the chemical energy of organic molecules into a form immediately usable by organisms. Glucose may be oxidized completely if sufficient oxygen is available by the following equation:

$$C_6H_{12}O_6 + 6O_2(g) \rightarrow 6\ H_2O + 6\ CO_2(g) + \text{energy}$$

All organisms, including plants and animals, oxidize glucose for energy. Often, this energy is used to convert ADP and phosphate into ATP. It is known that peas undergo cell respiration during germination. Do peas undergo cell respiration before germination? The results of this experiment will verify that germinating peas do respire. Using your collected data, you will be able to answer the question concerning respiration and non-germinating peas.

Using the CO_2 Gas Sensor, you will monitor the carbon dioxide produced by peas during cell respiration. Both germinating and non-germinating peas will be tested. Additionally, cell respiration of germinating peas at two different temperatures will be tested.

OBJECTIVES

In this experiment, you will

- Use a CO_2 Gas Sensor to measure concentrations of carbon dioxide.
- Study the effect of temperature on cell respiration.
- Determine whether germinated and non-germinated peas respire.
- Compare the rates of cell respiration in germinated and non- germinated peas.

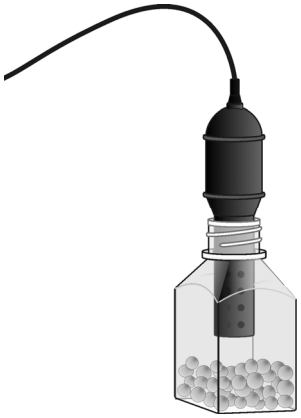


Figure 1

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier CO_2 Gas Sensor
250 mL respiration chamber
25 germinating peas
25 non-germinating peas
ice cubes
thermometer
two 100 mL beakers

PROCEDURE

1. If your sensor has a switch, set it to the Low (0–10,000 ppm) setting. Connect the CO_2 Gas Sensor to Channel 1 of the Vernier computer interface.
2. Prepare the computer for data collection by opening the "14A Cell Resp" file in the *Agricultural Science with Vernier* folder.
3. Obtain 25 germinating peas and blot them dry between two pieces of paper towel. Use the thermometer to measure the room temperature. Record the temperature in Table 1.
4. Place the germinating peas into the respiration chamber.
5. Place the shaft of the CO_2 Gas Sensor in the opening of the respiration chamber.
6. Wait one minute, then begin measuring carbon dioxide concentration by clicking Collect. Data will be collected for 5 minutes.
7. Remove the CO_2 Gas Sensor from the respiration chamber. Place the peas in a 100 mL beaker filled with cold water and an ice cube. The cold water will prepare the peas for part II of the experiment.
8. Use a notebook or notepad to fan air across the openings in the probe shaft of the CO_2 Gas Sensor for 1 minute.
9. Fill the respiration chamber with water and then empty it. Thoroughly dry the inside of the respiration chamber with a paper towel.
10. Determine the rate of respiration:
    a. Move the mouse pointer to the point where the data values begin to increase. Hold down the left mouse button. Drag the mouse pointer to the end of the data and release the mouse button.
    b. Click the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
    c. Record the slope of the line, *m*, as the rate of respiration for germinating peas at room temperature in Table 2.
    d. Close the linear regression floating box.
11. Move your data to a stored run. To do this, choose Store Latest Run from the Experiment menu.
12. Obtain 25 non-germinating peas and place them in the respiration chamber
13. Repeat Steps 5–11 for the non-germinating peas.

Part II Germinating peas, cool temperatures

14. Remove the peas from the cold water and blot them dry between two paper towels.
15. Repeat Steps 5–11 to collect data with the cold germinating peas.

16. Print a graph showing all three data runs.
    a. Label all three curves by choosing Text Annotation from the Insert menu, and typing "Room Temp Germinated" (or "Room Temp Non-germinated", or "Cold Germinated") in the edit box. Then drag each box to a position near its respective curve. Adjust the position of the arrowhead.
    b. Print a copy of the graph, with all three data sets and the regression lines displayed. Enter your name(s) and the number of copies of the graph you want.

DATA

Table 1	
Condition	Temperature (°C)
room	

Table 2	
Peas	Rate of respiration (ppm/min)
Germinating, room temperature	
Non-germinating, room temperature	
Germinating, cool temperature	

QUESTIONS

1. Do you have evidence that cell respiration occurred in peas? Explain.
2. What is the effect of germination on the rate of cell respiration in peas?
3. What is the effect of temperature on the rate of cell respiration in peas?
4. Why do germinating peas undergo cell respiration?

EXTENSIONS

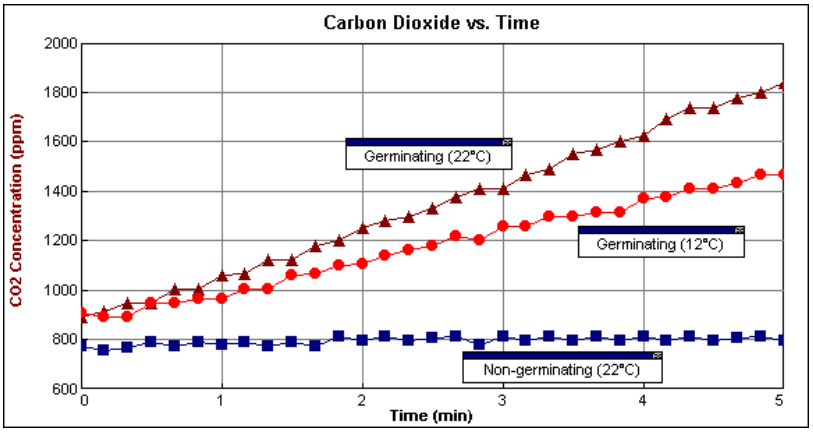
1. Compare the respiration rate among various types of seeds.
2. Compare the respiration rate among seeds that have germinated for different time periods, such as 1, 3, and 5 days.
3. Compare the respiration rates of various small animal types, such as insects or earthworms.

TEACHER INFORMATION

Cell Respiration

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Allow the seeds to germinate for three days prior to the experiment. Prior to the first day, soak them in water overnight. On subsequent days, roll them in a moist paper towel and place the towel in a paper bag. Place the bag in a warm, dark place. Check each day to be sure the towels remain very moist. If time is short, the peas can be used after they have soaked overnight. For best results, allow them to germinate for the full three days.
3. Heavy condensation buildup in the respiration chamber can interfere with readings from the CO_2 Gas Sensor. This can be a source of error if the peas are very wet when placed in the respiration chamber. Before placing the peas in the respiration chamber, blot them dry with a paper towel.
4. The stopper included with the older-style CO_2 Gas Sensor is slit to allow easy application and removal from the probe. When students are placing the probe in the respiration chamber, they should gently twist the stopper into the chamber opening. Warn the students not to twist the probe shaft or they may damage the sensing unit.
5. The CO_2 Gas Sensor relies on the diffusion of gases into the probe shaft. Students should allow a couple of minutes between trials so that gases from the previous trial will have exited the probe shaft. Alternatively, the students can use a firm object such as a book or notepad to fan air through the probe shaft. This method is used in the student procedure.
6. The morning of the experiment fill a 1 L beaker with ice and water so that students will have cold water. Students will also need access to ice.
7. The calibration stored in this experiment file works well for this experiment. Initial readings that seem slightly high or low will still reflect an accurate change in gas levels.
8. To conserve battery power, we suggest that AC Adapters be used to power the interfaces rather than batteries when working with the CO_2 Gas Sensor.

SAMPLE RESULTS



CO_2 respired by germinating and non-germinating peas

Table 1	
Condition	Temperature (°C)
room	22
cold water	10

Table 2	
Peas	Respiration rate (ppm/min)
Germinating, room temperature	194.5
Non-germinating, room temperature	6.6
Germinating, cool temperature	122.1

ANSWERS TO QUESTIONS

1. Yes, the carbon dioxide concentration *vs*. time graph clearly indicates that carbon dioxide is being produced at a steady rate when germinating peas are in the respiration chamber.

2. Germination greatly accelerates the rate of cellular respiration. This reflects a higher rate of metabolic activity in germinating seeds. In most experiments, non-germinating seeds do not seem to be respiring. Occasionally, however, some respiration is detectable.

3. Warm temperatures increase the rate of respiration. This reflects a higher rate of metabolic activity in warm germinating seeds than in cool seeds.

4. It is necessary for germinating seeds to undergo cellular respiration in order to acquire the energy they need for growth and development. Unlike their mature relatives, seeds do not yet have the necessary photosynthetic abilities needed to product their own energy sources.

Cell Respiration

Cell respiration refers to the process of converting the chemical energy of organic molecules into a form immediately usable by organisms. Glucose may be oxidized completely if sufficient oxygen is available by the following equation:

$$C_6H_{12}O_6 + 6O_2(g) \rightarrow 6\ H_2O + 6\ CO_2(g) + \text{energy}$$

All organisms, including plants and animals, oxidize glucose for energy. Often, this energy is used to convert ADP and phosphate into ATP. It is known that peas undergo cell respiration during germination. Do peas undergo cell respiration before germination? The results of this experiment will verify that germinating peas do respire. Using your collected data, you will be able to answer the question concerning respiration and non-germinating peas.

Using the O_2 Gas Sensor, you will monitor the oxygen consumed by peas during cell respiration. Both germinating and non-germinating peas will be tested. Additionally, cell respiration of germinating peas at two different temperatures will be tested.

OBJECTIVES

In this experiment, you will

- Use an O_2 Gas Sensor to measure concentrations of oxygen.
- Study the effect of temperature on cell respiration.
- Determine whether germinated and non-germinated peas respire.
- Compare the rates of cell respiration in germinated and non- germinated peas.

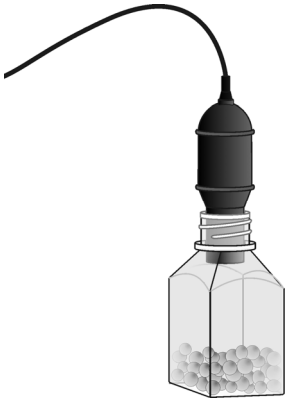


Figure 1

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier O_2 Gas Sensor
two 100 mL beakers
25 germinating peas
25 non-germinating peas
250 mL respiration chamber
ice cubes
thermometer

PROCEDURE

1. Connect the O_2 Gas Sensor to the computer interface.
2. Prepare the computer for data collection by opening the file "14B Cell Resp O2" from the *Agricultural Science with Vernier* folder of Logger*Pro*.
3. Obtain 25 germinating peas and blot them dry between two pieces of paper towel. Use the thermometer to measure the room temperature. Record the temperature in Table 1.
4. Place the germinating peas into the respiration chamber.
5. Place the O_2 Gas Sensor into the bottle as shown in Figure 1. Gently push the sensor down into the bottle until it stops. The sensor is designed to seal the bottle without the need for unnecessary force.
6. Wait two minutes, then begin collecting data by clicking Collect. Data will be collected for 10 minutes.
7. When data collection has finished, remove the O_2 Gas Sensor from the respiration chamber. Place the peas in a 100 mL beaker filled with cold water and an ice cube.
8. Fill the respiration chamber with water and then empty it. Thoroughly dry the inside of the respiration chamber with a paper towel.
9. Determine the rate of respiration:
   a. Click the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
   b. Record the slope of the line, *m*, as the rate of respiration for germinating peas at room temperature in Table 2.
   c. Close the linear regression floating box.
10. Move your data to a stored run. To do this, choose Store Latest Run from the Experiment menu.
11. Obtain 25 non-germinating peas and place them in the respiration chamber
12. Repeat Steps 5–10 for the non-germinating peas.

Part II Germinating peas, cool temperatures

13. Remove the peas from the cold water and blot them dry between two paper towels.
14. Repeat Steps 5–9 to collect data with the germinating peas at a cold temperature.
15. Print a graph showing all three data runs.
    a. Label all three curves by choosing Text Annotation from the Insert menu, and typing "Room Temp Germinated" (or "Room Temp Non-germinated", or "Cold Germinated") in the edit box. Then drag each box to a position near its respective curve. Adjust the position of the arrow head.
    b. Print a copy of the graph, with all three data sets and the regression lines displayed. Enter your name(s) and the number of copies of the graph you want.

DATA

Table 1	
Condition	Temperature (°C)
room	

Table 2	
Peas	Rate of respiration (%/min)
Germinating, room temperature	
Non-germinating, room temperature	
Germinating, cool temperature	

QUESTIONS

1. Do you have evidence that cell respiration occurred in peas? Explain.
2. What is the effect of germination on the rate of cell respiration in peas?
3. What is the effect of temperature on the rate of cell respiration in peas?
4. Why do germinating peas undergo cell respiration?

EXTENSIONS

1. Compare the respiration rate among various types of seeds.
2. Compare the respiration rate among seeds that have germinated for different time periods, such as 1, 3, and 5 days.
3. Compare the respiration rate among various types of small animals, such as insects or earthworms.

TEACHER INFORMATION

Cell Respiration

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), EasyData or DataMate (calculators), and DataPro (Palm handhelds) can be found on the CD that accompanies this book. See *Appendix A* for more information.

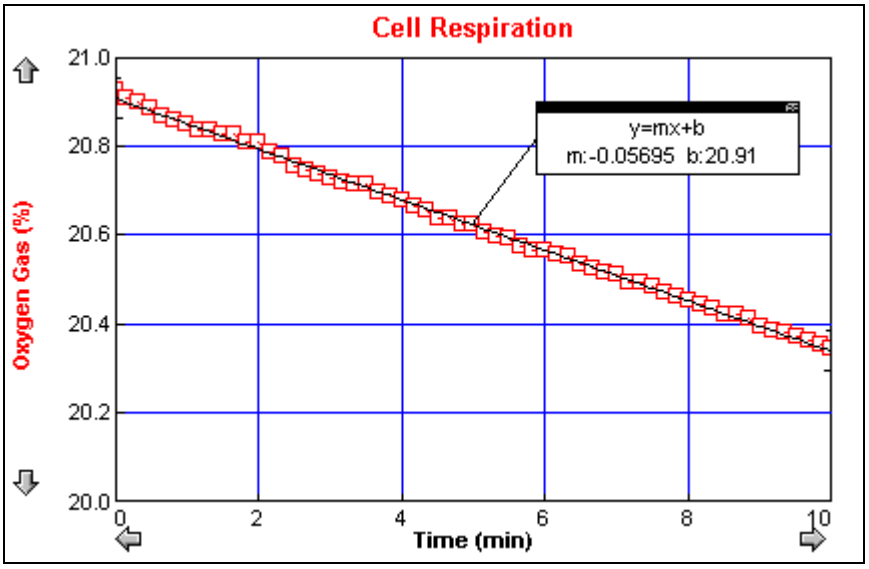
2. Allow the seeds to germinate for three days prior to the experiment. Prior to the first day, soak them in water overnight. On subsequent days, roll them in a moist paper towel and place the towel in a paper bag. Place the bag in a warm, dark place. Check each day to be sure the towels remain very moist. If time is short, the peas can be used after they have soaked overnight. For best results, allow them to germinate for the full three days.

3. To extend the life of the O_2 Gas Sensor, always store the sensor upright in the box it was shipped in.

4. The morning of the experiment fill a 1 L beaker with ice and water so that students will have cold water for Step 7. Students will also need access to ice.

5. The calibration stored in the data-collection software works well for this experiment. The calibration is for the O_2 Gas Sensor (%).

SAMPLE RESULTS



O_2 respired by germinated peas

Table 1	
Condition	Temperature (°C)
Room	24
Cold water	9

Table 2	
Peas	Respiration rate (%/min)
Germinating, room temperature	–0.057
Non-germinating, room temperature	–0.002
Germinating, cool temperature	–0.039

ANSWERS TO QUESTIONS

1. Yes, the oxygen concentration *vs*. time graph clearly indicates that oxygen is being consumed at a steady rate when germinating peas are in the respiration chamber.

2. Germination greatly accelerates the rate of cellular respiration. This reflects a higher rate of metabolic activity in germinating seeds. In most experiments, non-germinating seeds do not seem to be respiring. Occasionally, however, some respiration is detectable.

3. Warm temperatures increase the rate of respiration. This reflects a higher rate of metabolic activity in warm germinating seeds than in cool seeds.

4. It is necessary for germinating seeds to undergo cellular respiration in order to acquire the energy they need for growth and development. Unlike their mature relatives, seeds do not yet have the necessary photosynthetic abilities needed to produce their own energy sources.

Cell Respiration

Cell respiration refers to the process of converting the chemical energy of organic molecules into a form immediately usable by organisms. Glucose may be oxidized completely if sufficient oxygen is available according to the following equation:

$$C_6H_{12}O_6 + 6O_2(g) \rightarrow 6\ H_2O + 6\ CO_2(g) + \text{energy}$$

All organisms, including plants and animals, oxidize glucose for energy. Often, this energy is used to convert ADP and phosphate into ATP. Peas undergo cell respiration during germination. Do peas undergo cell respiration before germination? Using your collected data, you will be able to answer the question regarding respiration and non-germinating peas.

Using the CO_2 Gas Sensor and O_2 Gas Sensor, you will monitor the carbon dioxide produced and the oxygen consumed by peas during cell respiration. Both germinating and non-germinating peas will be tested. Additionally, cell respiration of germinating peas at two different temperatures will be investigated.

OBJECTIVES

In this experiment, you will

- Use an O_2 Gas Sensor to measure concentrations of oxygen gas.
- Use a CO_2 Gas Sensor to measure concentrations of carbon dioxide gas.
- Study the effect of temperature on cell respiration.
- Determine whether germinating peas and non-germinating peas respire.
- Compare the rates of cell respiration in germinating and non-germinating peas.

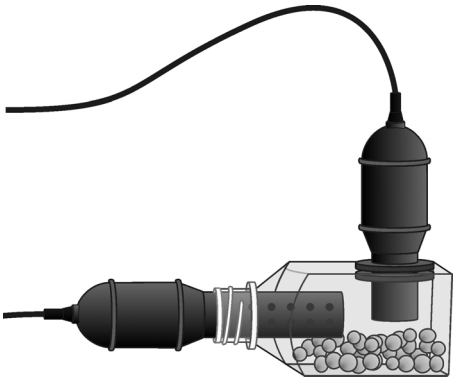


Figure 1

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier CO_2 Gas Sensor
Vernier O_2 Gas Sensor
BioChamber 250
25 germinating peas
25 non-germinating peas
250 mL respiration chamber
ice cubes
thermometer
two 100 mL beakers

PROCEDURE

1. If your CO_2 Gas Sensor has a switch, set it to the Low (0–10,000 ppm) setting. Connect the CO_2 Gas Sensor to Channel 1 and the O_2 Gas Sensor to Channel 2 of the Vernier computer interface.

2. Prepare the computer for data collection by opening the file "14C Cell Respiration (CO2 and O2)" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

3. Obtain 25 germinating peas and blot them dry between two pieces of paper towel. Use the thermometer to measure the room temperature. Record the temperature in Table 1.

4. Place the germinating peas into the respiration chamber.

5. Place the O_2 Gas Sensor into the BioChamber 250 as shown in Figure 1. Insert the sensor snugly into the grommet. The O_2 Gas Sensor should remain vertical throughout the experiment. Place the CO_2 Gas Sensor into the neck of the respiration chamber as shown in Figure 1.

6. Wait four minutes for readings to stabilize, then begin collecting data by clicking Collect. Collect data for ten minutes and click Stop.

7. When data collection has finished, remove the sensors from the respiration chamber. Place the peas in a 100 mL beaker filled with cold water and ice.

8. Fill the respiration chamber with water and then empty it. Thoroughly dry the inside of the respiration chamber with a paper towel.

9. Determine the rate of respiration:
   a. Click anywhere on the CO_2 graph to select it. Click the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
   b. Record the slope of the line, *m*, as the rate of respiration for germinating peas at room temperature in Table 2.
   c. Close the linear regression floating box.
   d. Repeat Steps 9a–c for the O_2 graph.

10. Move your data to a stored run. To do this, choose Store Latest Run from the Experiment menu.

11. Obtain 25 non-germinating peas and place them in the respiration chamber

12. Repeat Steps 5–10 for the non-germinating peas.

Part II Germinating peas, cool temperatures

13. Remove the peas from the cold water and blot them dry between two paper towels.

14. Repeat Steps 5–9 to collect data with the cold germinating peas.

15. Print a graph of concentration *vs.* volume showing all three data runs.
    a. Click anywhere on the CO_2 graph. Label all three curves by choosing Text Annotation from the Insert menu, and typing "Room Temp Germinated" (or "Room Temp Non-germinated", or "Cold Germinated") in the edit box. Then drag each box to a position near its respective curve. Adjust the position of the arrow head.
    b. Print a copy of the graph, with all three data sets and the regression lines displayed. Enter your name(s) and the number of copies of the graph you want.
    c. Click on the O_2 graph and repeat the process to print a copy of the O_2 graph.

DATA

Table 1	
Condition	Temperature (°C)
Room	

Table 2		
Peas	CO_2 Rate of respiration (ppt/min)	O_2 Rate of consumption (ppt/min)
Germinating, room temperature		
Non-germinating, room temperature		
Germinating, cool temperature		

QUESTIONS

1. Do you have evidence that cell respiration occurred in peas? Explain.
2. What is the effect of germination on the rate of cell respiration in peas?
3. What is the effect of temperature on the rate of cell respiration in peas?
4. Why do germinating peas undergo cell respiration?

EXTENSIONS

1. Compare the respiration rate among various types of seeds.
2. Compare the respiration rate among seeds that have germinated for different time periods, such as 1, 3, and 5 days.
3. Compare the respiration rate among various types of small animals, such as insects or earthworms.

TEACHER INFORMATION

Cell Respiration

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. Allow the seeds to germinate for three days prior to the experiment. Prior to the first day, soak them in water overnight. On subsequent days, roll them in a moist paper towel and place the towel in a paper bag. Place the bag in a warm, dark place. Check each day to be sure the towels remain very moist. If time is short, the peas can be used after they have soaked overnight. For best results, allow them to germinate for the full three days.

3. The O_2 Gas Sensor should always be stored upright in the box in which it was shipped.

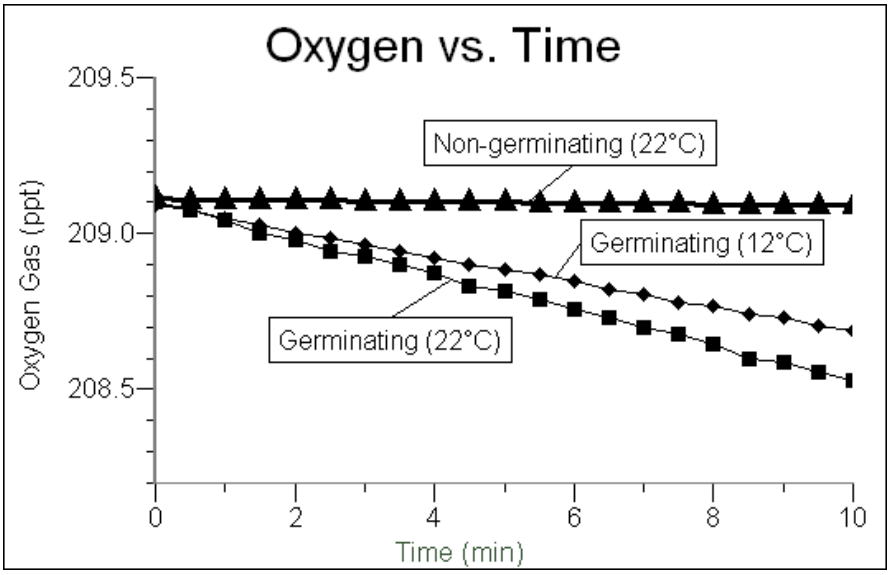
4. The morning of the experiment, fill a 1 L beaker with ice and water so that students will have cold water. Students will also need access to ice.

5. The calibrations stored in this experiment file for both sensors work well for this experiment. Initial readings that seem slightly high or low will still reflect an accurate change in gas levels.

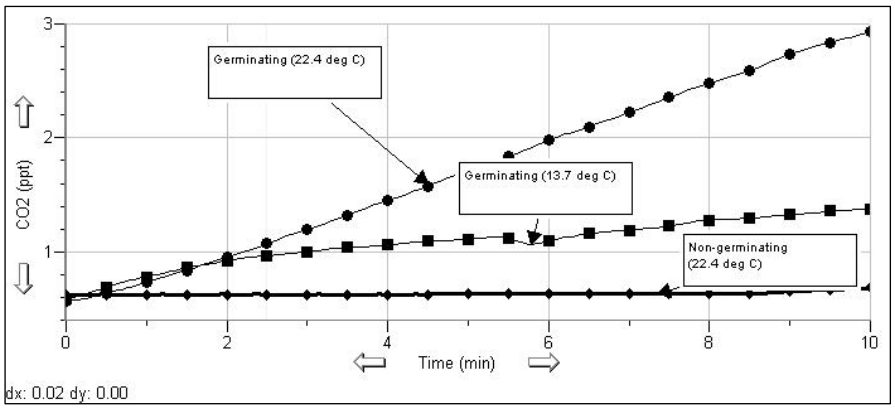
6. The stopper included with the older-style CO_2 Gas Sensor is slit to allow easy application and removal from the probe. When students are placing the probe in the respiration chamber, they should gently twist the stopper into the chamber opening. Warn the students not to twist the probe shaft or they may damage the sensing unit.

7. To conserve battery power, we suggest that AC Adapters be used to power the interfaces rather than batteries when working with the CO_2 Gas Sensor.

SAMPLE RESULTS



O_2 consumed by germinated peas



CO_2 levels of germinating and non-germinating peas

Table 1	
Condition	Temperature (°C)
room	22.4
cold water	13.7

Table 2		
Peas	CO_2 rate of respiration (ppt/min)	O_2 rate of consumption (ppt/min)
Germinating, room temperature	0.249	–0.152
Non-germinating, room temperature	0.003	–0.002
Germinating, cool temperature	0.066	–0.087

ANSWERS TO QUESTIONS

1. Yes, the oxygen concentration *vs*. time graph clearly indicates that oxygen is being consumed at a steady rate when germinating peas are in the respiration chamber. The carbon dioxide concentration *vs.* time graph indicates that carbon dioxide is being produced at a steady rate.

2. Germination greatly accelerates the rate of cellular respiration. This reflects a higher rate of metabolic activity in germinating seeds. In most experiments, non-germinating seeds do not seem to be respiring.

3. Warm temperatures increase the rate of respiration. This reflects a higher rate of metabolic activity in warm germinating seeds than in cooler seeds.

4. It is necessary for germinating seeds to undergo cellular respiration in order to acquire the energy they need for growth and development. Unlike their mature relatives, seeds do not yet have the necessary photosynthetic abilities needed to produce their own energy sources.

The Greenhouse Effect

Greenhouses allow gardeners to grow plants in cold weather. This is because the air inside the greenhouse stays warmer than the outside air. Short wavelength radiation from the sun passes through the glass, warming the interior of the greenhouse. The longer-wavelength radiation emitted does not pass through glass and is trapped in the greenhouse. This, along with the lack of mixing between the inside and outside air, keeps the greenhouse consistently warm.

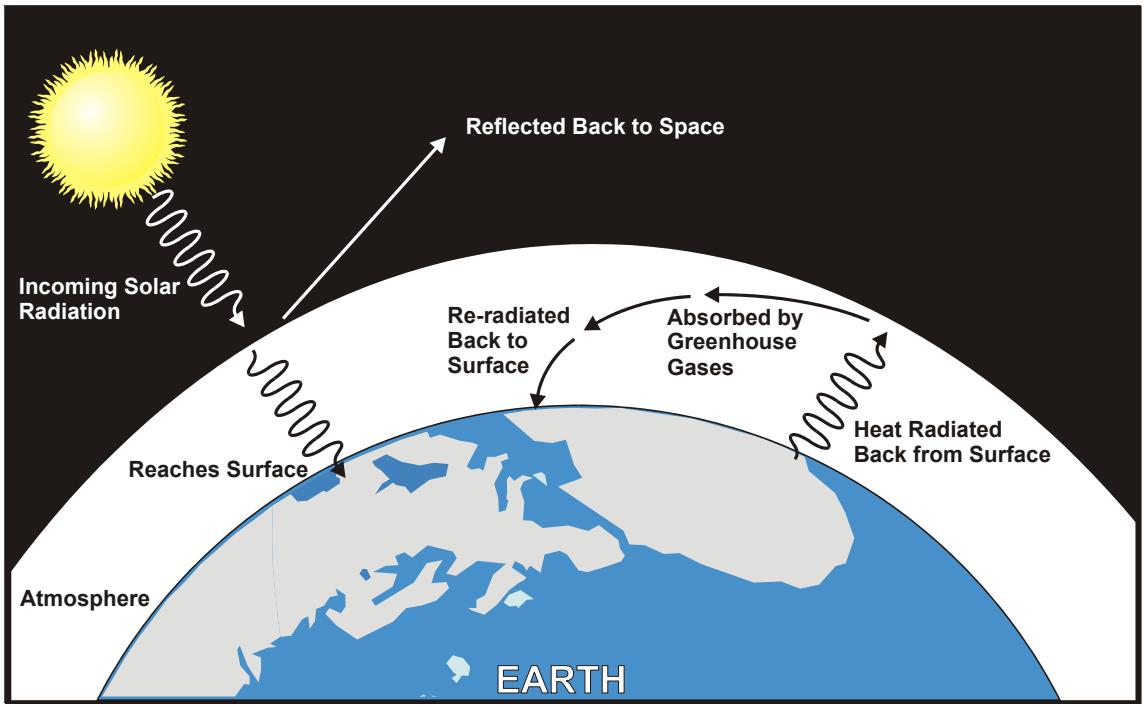


Figure 1

On a larger scale, the greenhouse effect helps keep our planet warm. Figure 1 shows how short wavelength radiation from the sun passes through the atmosphere, warming the Earth. The longer wavelength energy is then trapped by certain gases called greenhouse gases. The greenhouse gases most responsible are water vapor, carbon dioxide, methane, and nitrous oxide.

In this experiment, you will use two Temperature Probes to measure and compare the temperatures in model greenhouses under various conditions. In Part I, you will investigate the role of a plastic covering over the top of the model greenhouse. In Part II, you will investigate the effect of increased levels of two greenhouse gases – water vapor, H_2O, and carbon dioxide, CO_2.

OBJECTIVES

In this experiment, you will

- Use Temperature Probes to measure temperatures in a model greenhouse and a control.
- Use the results to make conclusions about the greenhouse effect.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
2 Temperature Probes
lamp with 100 watt bulb
2 rulers
tape
two 600 mL beakers
soil
plastic wrap

PROCEDURE

Part I The Effect of a Plastic Cover

1. Connect the Temperature Probes to the Vernier computer interface.
2. Tape Temperature Probe 1 and Temperature Probe 2 each to a ruler as shown in Figure 2. The probe tips should each be 3 cm from the ruler ends and the tape should not cover the probe tips.
3. Prepare the computer for data collection by opening the file "15 Greenhouse Effect" from the *Agricultural Science with Vernier* folder.

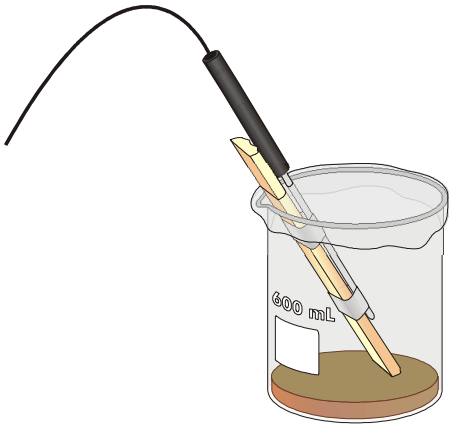


Figure 2

4. Obtain two beakers and prepare them for data collection.
   a. Place a layer of soil 1 cm deep in each beaker.
   b. Place the Temperature Probes into the beakers as shown in Figure 2.
   c. Cover the top of Beaker 1 (the beaker containing Probe 1) tightly with plastic wrap. There should not be too much excess plastic wrap covering the sides of the beaker. Beaker 1 is your covered greenhouse and Beaker 2 is the control.
   d. Position a lamp bulb the same distance from both beakers. The bulb should be about 5 cm above the tabletop and the same distance from the two probe tips.
5. Click Collect to begin data collection. Turn on the lamp.
6. Monitor the time in the meter. When 5 minutes have passed, turn off the lamp. Data will continue to be collected.
7. At the 10 minute mark, turn the lamp back on. Data collection will stop after 15 minutes.
8. When data collection stops, turn the lamp off.
9. Turn on the Examine feature by clicking on the Examine button, , on the toolbar.
10. Move the cursor to the 0 minute mark on the graph. Use the Examine box to determine the temperatures in Beaker 1 and Beaker 2 and record them in your data table.
11. Use the same method to determine the temperatures at the 5, 10, and 15 minute marks and record them in your data table.
12. Print copies of the graph as directed by your teacher.

13. Choose Store Latest Run from the Experiment menu.

Part II The Effect of Greenhouse Gases

14. Cover Beaker 2 tightly with plastic wrap. The ruler and probe should still be in place.
15. Lift the edge of the plastic wrap on Beaker 1 to make an opening.
16. Take a deep breath and hold it for as long as is comfortable. Without touching your lips to the beaker, exhale into the opening filling Beaker 1 with your breath.
17. Reseal the plastic over the top of the beaker and reposition near the lamp. Important: Make sure the beakers are the same distance from the lamp and are being illuminated evenly as in Part I.
18. Click Collect to begin data collection. Turn on the lamp.
19. Monitor the time in the meter. When 5 minutes have passed, turn off the lamp. Data will continue to be collected.
20. At the 10 minute mark, turn the lamp back on. Data collection will stop after 15 minutes.
21. When data collection stops, turn the lamp off.
22. Turn on the Examine feature by clicking on the Examine button, , on the toolbar.
23. Record the temperature values at the 0, 5, 10, and 15 minute marks in your data table. **Important:** Be sure that you are reading the "Latest" temperature values and not the Run 1 values from the Examine box.
24. (Optional). Print copies of the graph.

DATA

Part I The Effect of a Plastic Cover

	Probe 1 Greenhouse	Probe 2 Control	Temperature difference
0 minute temperature (°C)			
5 minute temperature (°C)			
10 minute temperature (°C)			
15 minute temperature (°C)			

Part II The Effect of Greenhouse Gases

	Probe 1 Greenhouse gases	Probe 2 Control	Temperature difference
0 minute temperature (°C)			
5 minute temperature (°C)			
10 minute temperature (°C)			
15 minute temperature (°C)			

PROCESSING THE DATA

Part I The Effect of a Plastic Cover

1. In the spaces provided in your data table, subtract to find the temperature differences.
2. During periods when the lamp was on, did the covered beaker warm faster or slower than the control?
3. Give a possible explanation for your answer to Question 2.
4. During periods when the lamp was off, did the covered beaker cool faster or slower than the control?
5. Give a possible explanation for your answer to Question 4.
6. Explain why a closed automobile heats up in the sun.

Part II The Effect of Greenhouse Gases

7. In the spaces provided in your data table, subtract to find the temperature differences.
8. What two important greenhouse gases did the exhaled air contain?
9. During periods when the lamp was on, did the beaker with greenhouse gases warm faster or slower than the control?
10. Give a possible explanation for your answer to Question 9.
11. During periods when the lamp was off, did the covered beaker warm faster or slower than the control?
12. Give a possible explanation for your answer to Question 11.
13. In what way is the greenhouse effect good for the Earth?
14. In what ways might the greenhouse effect become a problem for the Earth?

EXTENSIONS

1. Repeat the experiment using the sun as the light source.

2. Run the experiment for two hours. How are the results different than your results for the 15 minute data-collection period? Explain the differences.

3. Repeat the experiment using plastic containers instead of glass ones. Discuss any differences that result.

TEACHER INFORMATION

The Greenhouse Effect

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. If you are using calculators for data collection, his activity can be performed with calculators from the TI-83 Plus or TI-84 Plus families and a LabPro or CBL 2. It can not be performed with Easy products because all runs of data must be collected at the same time under similar conditions.

3. Each group needs two temperature probes for this activity. You may need to combine groups or instruct students to collect data in rounds

4. The 600 mL beakers could be replaced with 400 mL or 250 mL beakers if needed.

5. Make sure the students do not have too much excess plastic wrap on the sides of their beakers. This will affect their results.

6. Use a plastic wrap that clings well to glass. You do not want to have to use tape to keep the plastic tight on the beaker.

7. Black paper could be substituted for soil.

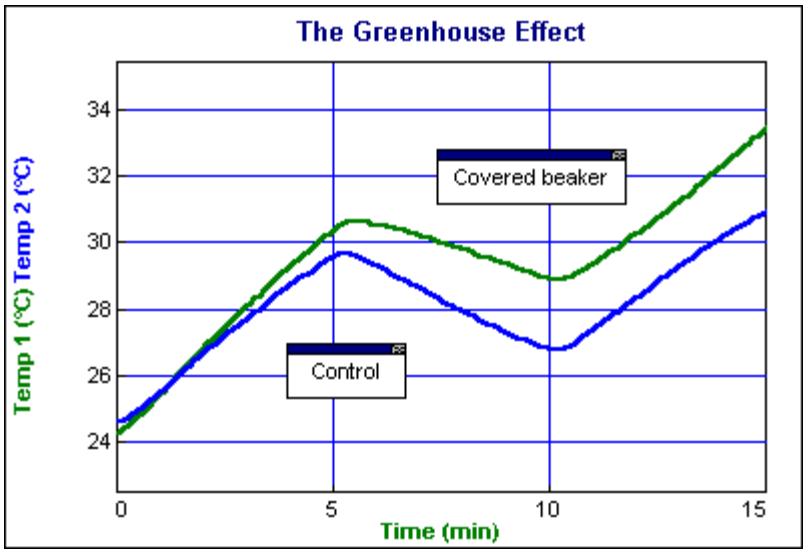
8. If the experiment is performed on a very humid day, the effects of the water vapor in Part II will be minimized. The increased CO_2 levels should still cause a measurable difference in temperature, however.

9. Try to use rulers of the same color for each group. A dark-colored ruler in one beaker and a light-colored ruler in the other could affect the students' results.

SAMPLE RESULTS

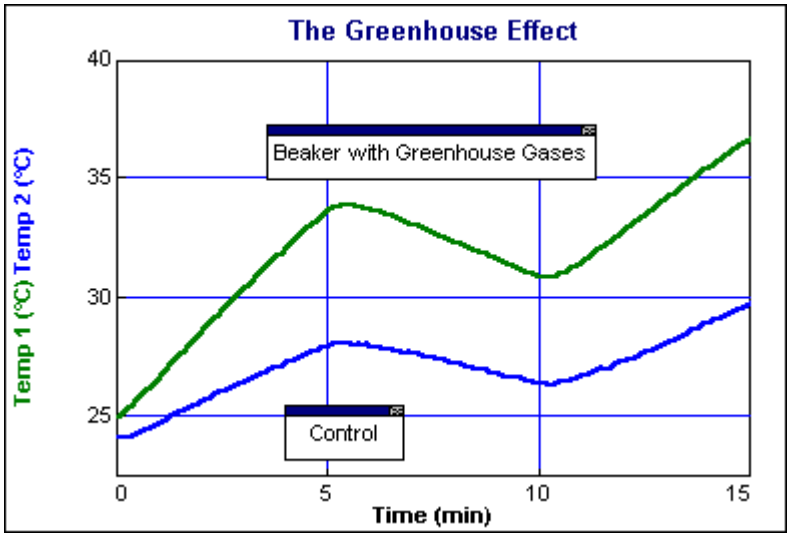
Part I The Effect of a Plastic Cover

	Probe 1 Greenhouse	Probe 2 Control	Temperature difference
5 minute temperature (°C)	30.5	29.7	0.8
10 minute temperature (°C)	29.0	26.9	2.1
15 minute temperature (°C)	33.4	30.9	2.5



Part II The Effect of Greenhouse Gases

	Probe 1 Greenhouse Gases	Probe 2 Control	Temperature Difference
5 minute temperature (°C)	33.8	28.1	5.7
10 minute temperature (°C)	31.0	26.5	4.5
15 minute temperature (°C)	36.6	29.7	6.9



ANSWERS TO QUESTIONS

Part I The Effect of a Plastic Cover

1. See the Sample Results.
2. When the lamp was on, the covered beaker warmed up faster.
3. The light was able to enter both beakers, but the resulting heat was trapped in the beaker with the covering.
4. When the lamp was off, the covered beaker cooled off more slowly.
5. The covering kept the warm air on the inside from mixing with the cold air on the outside.
6. Just like the covered beaker, light will pass through the glass in the windows of an automobile. The resulting heat is then trapped by the closed windows.

Part II The Effect of Greenhouse Gases

7. See the Sample Results.
8. Increased levels of carbon dioxide, CO_2, and water vapor, H_2O, are found in human breath.
9. When the lamp was on, the beaker with greenhouse gases warmed faster than the control.
10. Greenhouse gases absorb heat better than other gases in the air, so the beaker with more greenhouse gases will become warmer.
11. When the lamp was off, the beaker with greenhouse gases cooled faster than the control.
12. In this case, both beakers are covered. The warm air inside the beakers cannot mix with the cooler air outside. Because there is a greater temperature difference between the beaker with greenhouse gases and the air, it will cool faster.
13. The greenhouse effect keeps Earth at a relatively moderate temperature. Without it, temperature extremes would be unbearable for life to exist.
14. Human-caused emissions of greenhouse gases could cause the atmosphere to warm more than it should. This could cause major shifts in climates and weather patterns.

ACKNOWLEDGEMENT

We wish to thank Don Volz and Sandy Sapatka for their help in developing and testing this experiment.

Energy in Food

Food supplies energy for all animals—without it we could not live. The quantity of energy stored in food is of great interest to humans. The energy your body needs for running, talking, and thinking comes from the foods you eat. Not all foods contain the same amount of energy, nor are all foods equally nutritious for you. An average person should consume a minimum of 2,000 kilocalories per day. That is equivalent to 8,360 kilojoules. Calories and joules are both units of energy. We will use joules in this lab since it is the accepted SI metric standard.

You can determine energy content of food by burning a portion of it and capturing the heat released to a known amount of water. This technique is called *calorimetry*. The energy content of the food is the amount of heat produced by the combustion of 1 gram of a substance. It is measured in kilojoules per gram (kJ/g).

OBJECTIVES

In this experiment, you will

- Use a computer to measure temperature changes.
- Monitor the energy given off by food as it burns.
- Determine and compare the energy content of different foods.

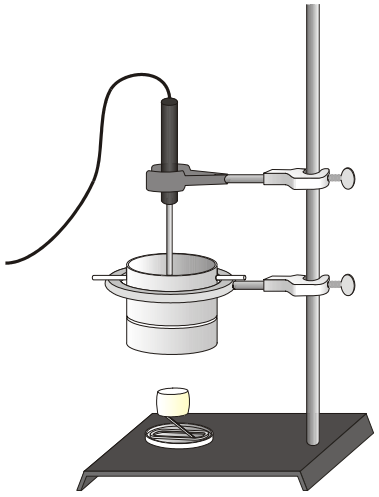


Figure 1

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Temperature Probe
100 mL graduated cylinder
balance
food holder
two food samples (nut, popcorn, or marshmallow)
matches
ring stand and 10 cm ring
small can
split 1-hole stopper
two stirring rods
utility clamp
warm and cool water
wooden splint
two 1-hole rubber stoppers

PROCEDURE

1. Obtain and wear goggles.

2. Obtain a sample of food and a food holder similar to the one shown in Figure 1. Mount the food onto the food holder so that it can burn without damaging the holder. Find and record the initial mass of the food sample and food holder. **CAUTION:** *Do not eat or drink in the laboratory.*

3. Connect the Temperature Probe to the computer interface. Prepare the computer for data collection by opening the file "16 Energy in Food" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

4. Set up the apparatus shown in Figure 1.
   a. Determine the mass of an empty can. Record the value in Table 1.
   b. Place about 50 mL of cold water into the can.
   c. Determine and record the mass of the can plus the water.
   d. Insert a stirring rod through the holes in the top of the can and hold it in place with two one-hole stoppers. Position the can 2.5 cm (~1 inch) above the food sample.
   e. Use a utility clamp and split stopper to suspend the temperature probe in the water. The probe should not touch the bottom or side of the can.

5. Click ▶ Collect to begin data collection. Record the initial (minimum) temperature of the water in Table 1.

6. Remove the food sample from under the can and use a wooden splint to light it. Quickly place the burning food sample directly under the center of the can. Stir the water constantly. **CAUTION***: Keep hair and clothing away from an open flame.*

7. If the temperature of the water exceeds 60°C, blow the flame out. Do not stop the computer yet.

8. After 4 minutes, if the food is still burning, blow the flame out. Record the maximum temperature of the water in Table 1.

9. Once the water temperature begins to decrease, end data collection by clicking ■ Stop.

10. Determine the final mass of the food sample and food holder.

11. Place burned food, matches, and wooden splints in the container supplied by your instructor.

12. You can confirm your data by clicking the Statistics button, . The minimum temperature (t_2) and maximum temperature (t_1) are listed in the floating box.

13. Repeat Steps 4–12 for a second food sample. Be sure to use a new 50 mL portion of cold water.

DATA

Table 1		
Measurements	Sample 1	Sample 2
Food used		
Mass of empty can (g)		
Mass of can plus water (g)		
Minimum temperature of water (°C)		
Maximum temperature of water (°C)		
Initial mass of food (g)		
Final mass of food (g)		

Table 2		
Calculations	Sample 1	Sample 2
Mass of water (g)		
Δt of water (°C)		
Δmass of food (g)		
Energy gained by water (J)		
Energy content of food (J/g)		

PROCESSING THE DATA

Record the following calculations in Table 2. Show your work in Table 3.

1. Calculate the change in mass of water. Show your calculations.
2. Calculate the change in mass of each food sample. Show your calculations.
3. Calculate the changes in the temperature of the water, Δt. Record this in Table 2. Show your calculations.
4. Calculate the energy gained by the heated water. Show your calculations. To do this, use the following equation:

   Energy gained by water = (mass of water) x (Δt of water) x (4.18 J/g°C)

5. Convert the energy you calculated in Step 3 to kilojoules (1 kJ = 1000 J).
6. Use your answer in Step 4 to calculate the energy content of each food sample (in kJ/g):

   Energy content of food = Energy gained by water / Δmass of food

Table 3		
Calculation	Sample 1	Sample 2
Δm		
Δt		
Energy gained		
Energy content		

7. Record your results and the results of other groups below.

Table 4			
Class Results			
Food Type	Food Type	Food Type	Food Type
Energy content (kJ/g)			
Average			

QUESTIONS

1. Which of the foods has the greatest energy content?
2. Which of the tested foods is the best energy source? Why?
3. What was the original energy source of the foods tested?
4. Why might some foods with a lower energy content be better energy sources than other foods with a higher energy content?
5. Would you expect the energy content values that you measured to be close to the value listed in dietary books? Why?

EXTENSION

1. Determine the energy content of other combustible foods.

TEACHER INFORMATION

Energy in Food

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), EasyData or DataMate (calculators), and DataPro (Palm handhelds) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. Any four nuts could be used for this experiment. Walnuts, pecans, peanuts, and almonds are easy to obtain and give excellent results.

3. The water can should be approximately 1.5 to 2 inches in diameter and about 3 inches long. A small juice can will do. Drill two holes in the can just under the metal rim, large enough so that a solid glass rod can easy fit in. The can will be suspended by the glass rod with a one-hole rubber stopper at each end. The rubber stoppers will rest on a metal 4 inch ring.

4. The food stand is made using a cork stopper, size 7 or larger, and a paper clip. Straighten one end of the paper clip and push it into the bottom of the cork stopper. Bend the other end of the paper clip into a ring so it will cup the food sample.

5. The two rubber stoppers on the very end of the stirring rod holding the can will prevent the can from slipping off the ring stand.

6. Heat is lost to the environment during this experiment as the fuel is burning. Therefore, the energy content students measure will not be similar to the published values. This lab is still valid, however, since the heat lost to the environment is nearly proportional in every experiment. If the physical conditions in every experiment are the same, the energy contents will be proportional. Several key factors include:
   - The distance from the bottom of the can to the flame (or table top) should be equal.
   - The cans should be of equal dimensions.
   - The flame should not be in a breeze.

7. Provide each lab group with a container to discard their burnt foods. The charred pieces will make a mess otherwise. Soot will accumulate on the outside of the calorimeter can. Provide a paper towel for students to set the can onto between experiments. Soap may be needed after this experiment!

8. The Vernier temperature calibrations that are stored in the data-collection software will work fine for this experiment.

9. Use of nuts, especially peanuts, is being restricted and phased out of schools due to increasing numbers of allergic reactions and the heightened sensitivity some students exhibit.

10. If you are collecting data on a calculator, we suggest that you clear all other programs and miscellaneous data off of the calculators to make room in the memory for collected data before loading EasyData.

SAMPLE RESULTS

Table 1			
Measurements	Sample 1	Sample 2	Sample 3
Food used	walnut	almond	pine nut
Mass of empty can (g)	28.51	28.51	28.51
Mass of can plus water (g)	77.35	78.27	76.99
Initial temperature of water (°C)	21.6	22.5	22.3
Final temperature of water (°C)	47.7	49.8	31.4
Initial mass of food (g)	12.85	12.92	12.30
Final mass of food (g)	12.25	12.18	12.10

Table 2			
Calculations	Sample 1	Sample 2	Sample 3
Mass of water (g)	49.18	49.76	48.48
Δt of water (°C)	26.1	27.3	9.1
Δmass of food (g)	0.60	0.74	0.20
Energy gained by water (J)	5330	5680	1840
Energy content of food (J/g)	8880	7670	9220

Since this experiment is designed for 9th and 10th grade students, the mathematics has been simplified. Here is a more complete description of some of the mathematical reasoning.

The law of conservation of energy states that the energy lost by the food should equal the energy gained by the water.

$$\Delta E_{\text{food}} = \Delta E_{\text{water}}$$

The energy lost to the environment is nearly proportional in every experiment so it can be ignored. The energy gained by the water can be calculated using the equation below where m_{water} is the mass of the water in grams, C_p is the heat capacity of water which is equal to 4.186 J/g°C, and Δt is the change in temperature in °C.

$$\Delta E_{\text{water}} = m_{\text{water}} \bullet C_p \bullet \Delta t$$

Since the energy gained by the water is equal to the energy lost by the food, then the energy lost by the food can be found by using this equation.

$$\Delta E_{\text{food}} = \Delta E_{\text{water}} = m_{\text{water}} \bullet C_p \bullet \Delta t$$

To calculate how much energy would be lost by one gram of the same food, divide the energy lost by the food by the mass of food that did burn.

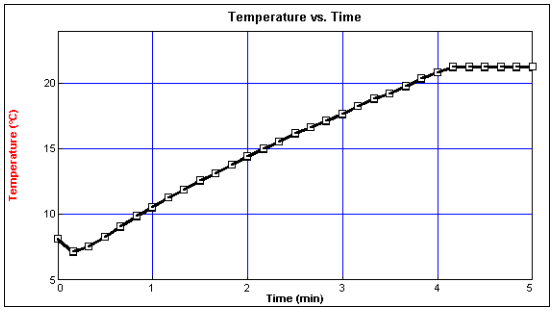
$$\text{Energy content} = \frac{\Delta E_{\text{food}}}{1\text{ gram}} = \frac{m_{\text{water}} \bullet C_p \bullet \Delta t}{\Delta m_{\text{food}}}$$

After converting joules to kilojoules, this gives us an answer similar to that in the student handout. We define the term *energy content* to be that amount of energy that can be obtained by the combustion of one gram of food.

The energy contents of a few sample foods are listed below. Note that these should be proportional to the measured energy contents, not equal to them, since heat was lost to the environment, and combustion was not complete.

Food	Energy content (kJ/g)
almond	26.8
brazil nuts	29.0
cashews	25.5
coconut (dry)	23.8
kidney beans	25.9

Food	Energy content (kJ/g)
lard	37.6
lima bean	14.3
peanuts	25.9
walnut	29.3



Walnut burning and warming water

ANSWERS TO QUESTIONS

1. Answers may vary, depending upon the type of food used. In the above experiment, pine nuts had the highest heat content, followed by walnuts, then almonds.

2. Answers may vary. The food with the highest energy content is the best energy source.

3. The sun is the source of energy for land plants. Plants can transform this radiant energy into chemical energy. This energy is used to manufacture many different substances, possibly in the form of fats, carbohydrates, or other high energy chemicals. When these chemicals are broken down, they release energy.

4. Some high energy foods might be indigestible. Food with a high energy content might be high in cholesterol or saturated fats. These may be harmful to some people in large amounts.

5. No, the measured values should be lower than those listed in a reference book.
   - There would be a certain amount of heat lost to the air and is not used to heat the water.
   - Soot that collects indicates incomplete combustion of the food. The published values assume complete combustion.

Computer
17A

Enzyme Action: Testing Catalase Activity

Many organisms can decompose hydrogen peroxide (H_2O_2) enzymatically. Enzymes are globular proteins, responsible for most of the chemical activities of living organisms. They act as *catalysts,* substances that speed up chemical reactions without being destroyed or altered during the process. Enzymes are extremely efficient and may be used over and over again. One enzyme may catalyze thousands of reactions every second. Both the temperature and the pH at which enzymes function are extremely important. Most organisms have a preferred temperature range in which they survive, and their enzymes most likely function best within that temperature range. If the environment of the enzyme is too acidic or too basic, the enzyme may irreversibly *denature*, or unravel, until it no longer has the shape necessary for proper functioning.

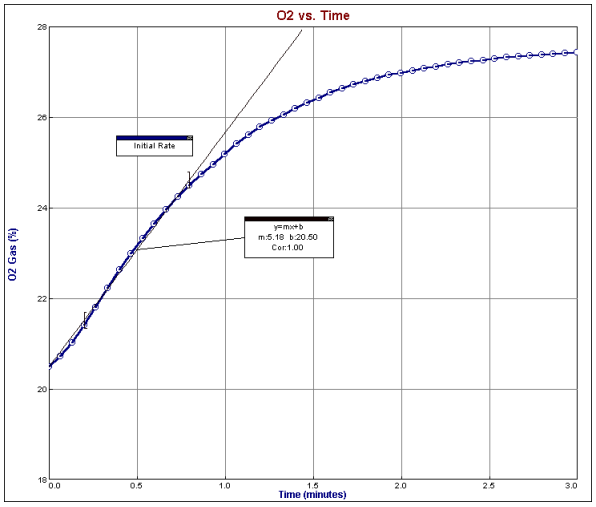
H_2O_2 is toxic to most living organisms. Many organisms are capable of enzymatically destroying the H_2O_2 before it can do much damage. H_2O_2 can be converted to oxygen and water, as follows:

$$2\ H_2O_2 \rightarrow 2\ H_2O + O_2$$

Although this reaction occurs spontaneously, the enzyme catalase increases the rate considerably. Catalase is found in most living organisms. A great deal can be learned about enzymes by studying the rates of enzyme-catalyzed reactions. The rate of a chemical reaction may be studied in a number of ways including:

- measuring the rate of appearance of a product (in this case, O_2, which is given off as a gas)
- measuring the rate of disappearance of substrate (in this case, H_2O_2)
- measuring the pressure of the product as it appears (in this case, O_2).

In this experiment, you will measure the rate of enzyme activity under various conditions, such as different enzyme concentrations, pH values, and temperatures. It is possible to measure the concentration of oxygen gas formed as H_2O_2 is destroyed using an O_2 Gas Sensor. If a plot is made, it may appear similar to the graph shown.



At the start of the reaction, there is no product, and the concentration is the same as the atmosphere. After a short time, oxygen accumulates at a rather constant rate. The slope of the curve at this initial time is constant and is called the *initial rate*. As the peroxide is destroyed, less of it is available to react and the O_2 is produced at lower rates. When no more peroxide is left, O_2 is no longer produced.

OBJECTIVES

In this experiment, you will

- Use a computer and an O_2 Gas Sensor to measure the production of oxygen gas as hydrogen peroxide is destroyed by the enzyme catalase or peroxidase at various enzyme concentrations.
- Measure and compare the initial rates of reaction for this enzyme when different concentrations of enzyme react with H_2O_2.
- Measure the production of oxygen gas as hydrogen peroxide is destroyed by the enzyme catalase or peroxidase at various temperatures.
- Measure and compare the initial rates of reaction for the enzyme at each temperature.
- Measure the production of oxygen gas as hydrogen peroxide is destroyed by the enzyme catalase or peroxidase at various pH values.
- Measure and compare the initial rates of reaction for the enzyme at each pH value.

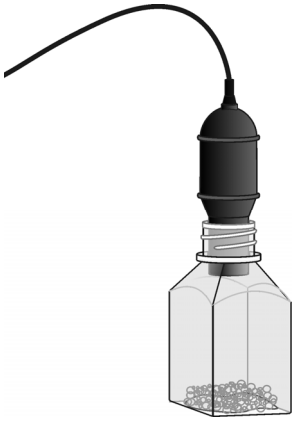


Figure 1

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier O_2 Gas Sensor
400 mL beaker
10 mL graduated cylinder
250 mL Nalgene bottle
three dropper pipettes
3.0% H_2O_2
enzyme suspension
three 18 × 150 mm test tubes
ice
pH buffers
test tube rack
thermometer

PROCEDURE

1. Obtain and wear goggles.

2. Connect the O_2 Gas Sensor to the computer interface. Prepare the computer for data collection by opening the file "17A Enzyme (O2)" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

Part I Testing the Effect of Enzyme Concentration

3. Place three test tubes in a rack and label them 1, 2, and 3. Fill each test tube with 5 mL of 3.0% H_2O_2 and 5 mL of water.

4. Initiate the enzyme catalyzed reaction.
   a. Using a clean dropper pipette, add 5 drops of enzyme suspension to test tube 1.
   b. Begin timing with a stopwatch or clock.
   c. Cover the opening of the test tube with a finger and gently invert the test tube two times.
   d. Pour the contents of the test tube into a clean 250 mL Nalgene bottle.
   e. Place the O_2 Gas Sensor into the bottle as shown in Figure 1. Gently push the sensor down into the bottle until it stops. The sensor is designed to seal the bottle without the need for unnecessary force.
   f. When 30 seconds has passed, Click ▸ Collect to begin data collection.

5. When data collection has finished, remove the O_2 gas sensor from the Nalgene bottle. Rinse the bottle with water and dry with a paper towel.

6. Move your data to a stored run. To do this, choose Store Latest Run from the Experiment menu.

7. Collect data for test tubes 2 and 3.
   - Add 10 drops of the enzyme solution to test tube 2. Repeat Steps 4–6.
   - Add 20 drops of the enzyme solution to test tube 3. Repeat Steps 4–5.

8. Using the mouse, select the initial linear region of your data on the graph. Click on the Linear Fit button, . Click OK and a best-fit linear regression line will be shown for each run selected. In your data table, record the value of the slope, *m*, for each of the three solutions. (The linear regression statistics are displayed in a floating box for each of the data sets.)

9. To print a graph of concentration *vs.* volume showing all three data runs.
   a. Label all three curves by choosing Text Annotation from the Insert menu, and typing "5 Drops" (or "10 Drops", or "20 Drops") in the edit box. Then drag each box to a position near its respective curve. Adjust the position of the arrow head.
   b. Print a copy of the graph, with all three data sets and the regression lines displayed. Enter your name(s) and the number of copies of the graph you want.

10. Determine the rate of reaction for each of the time intervals listed in Table 3 using the procedure outlined in Step 8. Record the rates for all three data runs in the Table 3.

Part II Testing the Effect of Temperature

Your teacher will assign a temperature range for your lab group to test. Depending on your assigned temperature range, set up your water bath as described below. Place a thermometer in your water bath to assist in maintaining the proper temperature.

- 0–5°C: 400 mL beaker filled with ice and water.
- 20–25°C: No water bath needed to maintain room temperature.
- 30–35°C: 400 mL beaker filled with very warm water.
- 50–55°C: 400 mL beaker filled with hot water.

11. Rinse the three numbered test tubes used for Part I. Fill each test tube with 5 mL of 3.0% H_2O_2 and 5 mL of water. Place the test tubes in the water bath. The test tubes should be in the water bath for 5 minutes before proceeding to Step 12. Record the temperature of the water bath, as indicated on the thermometer, in the space provided in Table 4.

12. Find the rate of enzyme activity for test tubes 1, 2, and 3.
    - Add 10 drops of the enzyme solution to test tube 1. Repeat Steps 4–6.
    - Add 10 drops of the enzyme solution to test tube 2. Repeat Steps 4–6.
    - Add 10 drops of the enzyme solution to test tube 3. Repeat Steps 4–5.

13. Repeat Step 8 and record the reaction rate for each data set in Table 4. Calculate and record the average rate in Table 4.

14. Record the average rate and the temperature of your water bath from Table 4 on the class data table. When the entire class has reported their data, record the class data in Table 5.

Part III Testing the Effect of pH

15. Place three clean test tubes in a rack and label them pH 4, pH 7, and pH 10.

16. Add 3 mL of 3% H_2O_2 and 3 mL of a pH buffer to each test tube, as in Table 1.

Table 1

pH of buffer	Volume of 3% H_2O_2 (mL)	Volume of buffer (mL)
pH 4	5	5
pH 7	5	5
pH 10	5	5

17. Using the test tube labeled pH 4, add 10 drops of enzyme solution and repeat Steps 4–6.

18. Using the test tube labeled pH 7, add 10 drops of enzyme solution and repeat Steps 4–6.

19. Using the test tube labeled pH 10, add 10 drops of enzyme solution and repeat Steps 4–5.

20. Repeat Steps 8 and 9 to calculate the rate of reaction and print your graph. Record the reaction rate for each pH value in Table 6.

DATA

Part I Effect of Enzyme Concentration

Table 2	
Test tube label	Slope, or rate (%/min)
5 drops	
10 drops	
20 drops	

Table 3 Time intervals (minutes)					
Rates	0–0.5 min	0.5–1.0 min	1.0–1.5 min	1.5–2.0 min	2.0–3.0 min
5 Drops					
10 Drops					
20 Drops					

Part II Effect of Temperature

Table 4	
Test tube label	Slope, or rate (%/min)
Trial 1	
Trial 2	
Trial 3	
Average	
Temperature range:____°C	

Table 5 (Class Data)	
Temperature tested	Average rate

Part III Effect of pH

Table 6	
Test tube label	Slope, or rate (%/min)
pH 4	
pH 7	
pH 10	

PROCESSING THE DATA

1. On Page 2 of this experiment file, create a graph of the rate of enzyme activity *vs*. temperature. Plot the rate values for the class data in Table 5 on the y-axis, and the temperature on the x-axis. Use this graph to answer the questions for Part II.

QUESTIONS

Part I Effect of Enzyme Concentration

1. How does changing the concentration of enzyme affect the rate of decomposition of H_2O_2?

2. What do you think will happen to the rate of reaction if one increases the concentration of enzyme to 25 drops? Predict what the rate would be for 30 drops.

Part II Effect of Temperature

3. At what temperature is the rate of enzyme activity the highest? Lowest? Explain.

4. How does changing the temperature affect the rate of enzyme activity? Does this follow a pattern you anticipated?

5. Why might the enzyme activity decrease at very high temperatures?

Part III Effect of pH

6. At what pH is the rate of enzyme activity the highest? Lowest?

7. How does changing the pH affect the rate of enzyme activity?

EXTENSIONS

1. Different organisms often live in very different habitats. Design a series of experiments to investigate how different types of organisms might affect the rate of enzyme activity. Consider testing a plant, an animal, and a protist.

2. Presumably, at higher concentrations of H_2O_2, there is a greater chance that an enzyme molecule might collide with H_2O_2. If so, the concentration of H_2O_2 might alter the rate of oxygen production. Design a series of experiments to investigate how differing concentrations of the substrate hydrogen peroxide might affect the rate of enzyme activity.

3. Design an experiment to determine the effect of boiling the catalase on the rate of reaction.

4. Explain how environmental factors affect the rate of enzyme-catalyzed reactions.

TEACHER INFORMATION

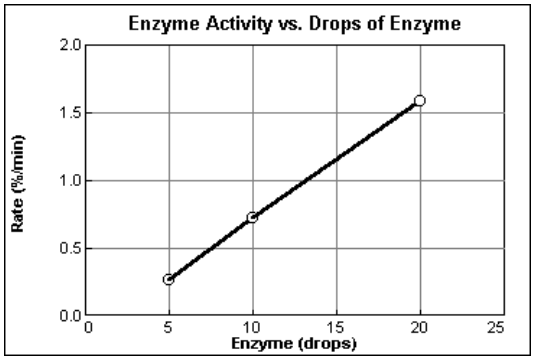
Enzyme Action: Testing Catalase Activity

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. This experiment may take a single group several lab periods to complete. A good breaking point is after the completion of Step 10, when students have tested the effect of different enzyme concentrations. Alternatively, if time is limited, different groups can be assigned one of the three tests and the data can be shared.
3. Your hot tap water may be in the range of 50–55°C for the hot-water bath. If not, you may want to supply pre-warmed temperature baths for Part II, where students need to maintain very warm water. Warn students not to touch the hot water.
4. Many different organisms may be used as a source of catalase in this experiment. If enzymes from an animal, a protist, and a plant are used by different teams in the same class, it will be possible to compare the similarities and differences among those organisms. Often, either beef liver, beef blood, or living yeast are used.
5. To prepare the yeast solution, dissolve 7 g (1 package) of dried yeast per 100 mL of 2% glucose solution. A 2% glucose is made by adding 20 g of glucose to enough distilled water to make 1 L of solution. Incubate the suspension in 37–40°C water for at least 10 minutes to activate the yeast. Test the experiment before the students begin. The yeast may need to be diluted if the reaction occurs too rapidly. The reaction in Step 4, with 6 mL of 1.5% hydrogen peroxide, and 5 drops of suspension produces enough oxygen to exceed a measured concentration of 22% in 40 to 60 seconds.
6. To prepare a liver suspension, homogenize 0.5 to 1.5 g of beef liver in 100 mL of cold water. You will need to test the suspension before use, as its activity varies greatly depending on its freshness. Dilute the suspension until the reaction in Step 4, with 6 mL of 1.5% hydrogen peroxide, and 5 drops of suspension produces enough oxygen to exceed a measured concentration of 22% in 40 to 60 seconds. The color of the suspension will be a faint pink. Keep the suspension on ice until used in an experiment.
7. You can purchase 3% H_2O_2 at any supermarket. If refrigerated, bring it to room temperature before starting the experiment.
8. To extend the life of the O_2 Gas Sensor, always store the sensor upright in the box in which it was shipped.
9. Vernier Software sells a pH buffer package for preparing buffer solutions with pH values of 4, 7, and 10 (order code PHB). Simply add the capsule contents to 100 mL of distilled water.

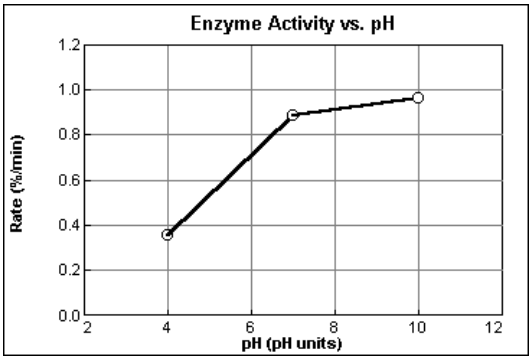
10. You can also prepare pH buffers using the following recipes:
    - pH 4: Add 2.0 mL of 0.1 M HCl to 1000 mL of 0.1 M potassium hydrogen phthalate.
    - pH 7: Add 582 mL of 0.1 M NaOH to 1000 mL of 0.1 M potassium dihydrogen phosphate.
    - pH 10: Add 214 mL of 0.1 M NaOH to 1000 mL of 0.05 M sodium bicarbonate.
11. You may need to let students know that at pH values above 10, enzymes will become denatured and the rate of activity will drop. If you have pH buffers higher than 10, have students perform an experimental run using them.

SAMPLE RESULTS

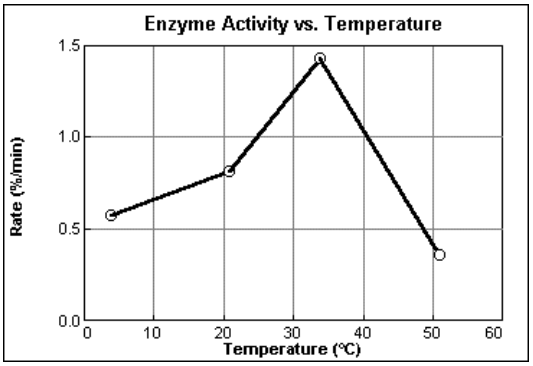
Sample class data	
Test tube label	Slope, or rate (%/min)
5 drops	0.27
10 drops	0.73
20 drops	1.59
0–5°C range: 4°C	0.58
20–25°C range: 21°C	0.82
30–35°C range: 34°C	1.43
50–55°C range: 51°C	0.36
pH 4	0.36
pH 7	0.89
pH 10	0.97



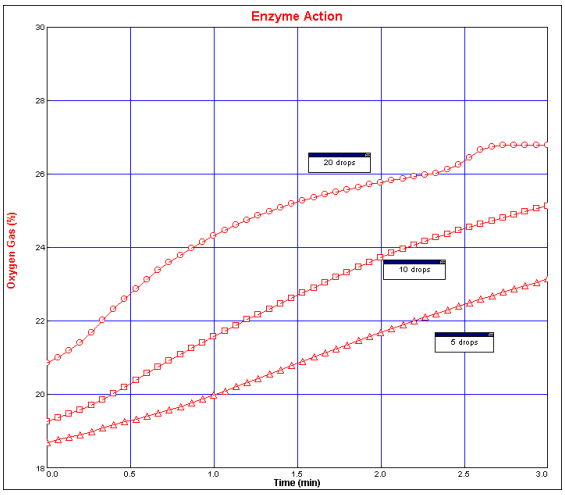
The effect of H_2O_2 concentration on the rate of enzyme activity



The effect of pH on the rate of enzyme activity



The effect of temperature on the rate of enzyme activity



Sample Data: Effect of H_2O_2 concentration on the rate of enzyme activity.

ANSWERS TO QUESTIONS

1. The rate should be highest when the concentration of enzyme is highest. With higher concentration of enzyme, there is a greater chance of an effective collision between the enzyme and H_2O_2 molecule.
2. Roughly, the rate doubles when the concentration of enzyme doubles. Since the data are somewhat linear, the rate is proportional to the concentration. At a concentration of 30 drops, the rate would be about 2.90 %/min.
3. The temperature at which the rate of enzyme activity is the highest should be close to 30°C. The lowest rate of enzyme activity should be at 60°C.
4. The rate increases as the temperature increases, until the temperature reaches about 50°C. Above this temperature, the rate decreases.
5. At high temperatures, enzymes lose activity as they are denatured.
6. Student answers may vary. Activity is usually highest at pH 10 and lowest at pH 4.
7. Student answers may vary. Usually, the enzyme activity increases from pH 4 to 10. At low pH values, the protein may denature or change its structure. This may affect the enzyme's ability to recognize a substrate or it may alter its polarity within a cell.

Enzyme Action: Testing Catalase Activity

Many organisms can decompose hydrogen peroxide (H_2O_2) enzymatically. Enzymes are globular proteins, responsible for most of the chemical activities of living organisms. They act as *catalysts,* as substances that speed up chemical reactions without being destroyed or altered during the process. Enzymes are extremely efficient and may be used over and over again. One enzyme may catalyze thousands of reactions every second. Both the temperature and the pH at which enzymes function are extremely important. Most organisms have a preferred temperature range in which they survive, and their enzymes most likely function best within that temperature range. If the environment of the enzyme is too acidic or too basic, the enzyme may irreversibly *denature*, or unravel, until it no longer has the shape necessary for proper functioning.

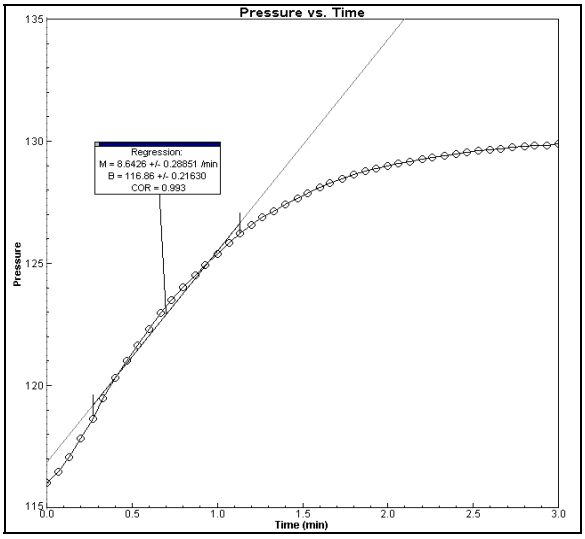
H_2O_2 is toxic to most living organisms. Many organisms are capable of enzymatically destroying the H_2O_2 before it can do much damage. H_2O_2 can be converted to oxygen and water, as follows:

$$2\ H_2O_2 \rightarrow 2\ H_2O + O_2$$

Although this reaction occurs spontaneously, the enzyme catalase increases the rate considerably. Catalase is found in most living organisms. A great deal can be learned about enzymes by studying the rates of enzyme-catalyzed reactions. The rate of a chemical reaction may be studied in a number of ways including:

- measuring the pressure of the product as it appears (in this case, O_2)
- measuring the rate of disappearance of substrate (in this case, H_2O_2)
- measuring the rate of appearance of a product (in this case, O_2 which is given off as a gas)

In this experiment, you will measure the rate of enzyme activity under various conditions, such as different enzyme concentrations, pH values, and temperatures. It is possible to measure the pressure of oxygen gas formed as H_2O_2 is destroyed. If a plot is made, it may appear similar to the graph shown.



At the start of the reaction, there is no product, and the pressure is the same as the atmospheric pressure. After a short time, oxygen accumulates at a rather constant rate. The slope of the curve at this initial time is constant and is called the *initial rate*. As the peroxide is destroyed, less of it is available to react and the O_2 is produced at lower rates. When no more peroxide is left, O_2 is no longer produced.

OBJECTIVES

In this experiment, you will

- Use a computer and Gas Pressure Sensor to measure the production of oxygen gas as hydrogen peroxide is destroyed by the enzyme catalase or peroxidase at various enzyme concentrations.
- Measure and compare the initial rates of reaction for this enzyme when different concentrations of enzyme react with H_2O_2.
- Measure the production of oxygen gas as hydrogen peroxide is destroyed by the enzyme catalase or peroxidase at various temperatures.
- Measure and compare the initial rates of reaction for the enzyme at each temperature.
- Measure the production of oxygen gas as hydrogen peroxide is destroyed by the enzyme catalase or peroxidase at various pH values.
- Measure and compare the initial rates of reaction for the enzyme at each pH value.

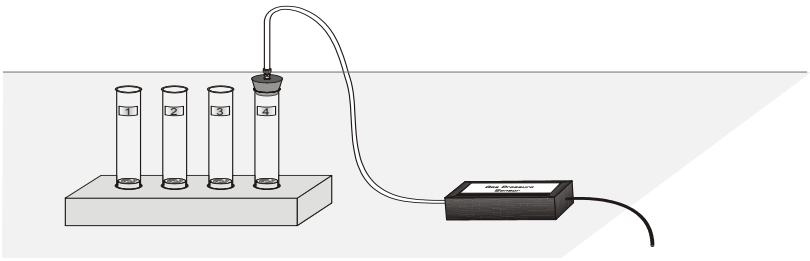


Figure 1

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier Gas Pressure Sensor
1-hole rubber stopper assembly
10 mL graduated cylinder
250 mL beaker of water
3% H_2O_2
600 mL beaker
enzyme suspension
four 18 x 150 mm test tubes
ice
pH buffers
test tube rack
thermometer
three dropper pipettes

PROCEDURE

1. Obtain and wear goggles.
2. Connect the Gas Pressure Sensor to the computer interface. Prepare the computer for data collection by opening the file "17B Enzyme (Pressure)" from the *Agricultural Science with Vernier* folder of Logger *Pro*.
3. Connect the plastic tubing to the valve on the Gas Pressure Sensor.

Part I Testing the Effect of Enzyme Concentration

4. Place four test tubes in a rack and label them 1, 2, 3, and 4. Partially fill a beaker with tap water for use in Step 5.
5. Add 3 mL of water and 3 mL of 3% H_2O_2 to each test tube.

6. Using a clean dropper pipette, add 1 drop of enzyme suspension to Test Tube 1. **Note**: Be sure not to let the enzyme fall against the side of the test tube.

Table 1		
Test tube label	Volume of 3% H_2O_2 (mL)	Volume of water (mL)
1	3	3
2	3	3
3	3	3
4	3	3

7. Stopper the test tube and gently swirl to thoroughly mix the contents. The reaction should begin. The next step should be completed as rapidly as possible.
8. Connect the free-end of the plastic tubing to the connector in the rubber stopper as shown in Figure 3. Click Collect to begin data collection. Data collection will end after 3 minutes.
9. If the pressure exceeds 130 kPa, the pressure inside the tube will be too great and the rubber stopper is likely to pop off. Disconnect the plastic tubing from the Gas Pressure Sensor if the pressure exceeds 130 kPa.
10. When data collection has finished, disconnect the plastic tubing connector from the rubber stopper. Remove the rubber stopper from the test tube and discard the contents in a waste beaker.

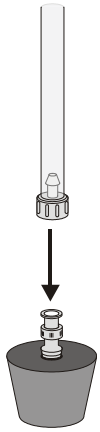


Figure 2

11. Find the rate of enzyme activity:
    a. Move the mouse pointer to the point where the data values begin to increase. Hold down the mouse button. Drag the mouse pointer to the point where the pressure values no longer increase and release the mouse button.
    b. Click the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best-fit line.
    c. Record the slope of the line, *m*, as the rate of enzyme activity in Table 4.
    d. Close the linear regression floating box.
12. Find the rate of enzyme activity for test tubes 2–4.
    a. Add 2 drops of the enzyme solution to test tube 2. Repeat Steps 7–11.
    b. Add 3 drops of the enzyme solution to test tube 3. Repeat Steps 7–11.
    c. Add 4 drops of the enzyme solution to test tube 4. Repeat Steps 7–11.

Part II Testing the Effect of Temperature

13. Place four clean test tubes in a rack and label them T 0–5, T 20–25, T 30–35, and T 50–55.

14. Add 3 mL of 3% H_2O_2 and 3 mL of water to each test tube, as shown in Table 2.

Table 2

Test tube label	Volume of 3% H_2O_2 (mL)	Volume of water
T 0–5	3	3
T 20–25 (room temp)	3	3
T 30–35	3	3
T 50–55	3	3

15. Measure the enzyme activity at 0–5°C:
    a. Prepare a water bath at a temperature in the range of 0–5°C by placing ice and water in a 600 mL beaker. Check that the temperature remains in this range throughout this test.
    b. Place Test Tube T 0–5 in the cold water bath until the temperature of the mixture reaches a temperature in the 0–5°C range. Record the actual temperature of the test-tube contents in the blank in Table 4.
    c. Add 2 drops of the enzyme solution to Test Tube T 0–5. Repeat Steps 7–11.

16. Measure the enzyme activity at 30–35°C:
    a. Prepare a water bath at a temperature in the range of 30–35°C by placing warm water in a 600 mL beaker. Check that the temperature remains in this range throughout this test.
    b. Place Test Tube T 30–35 in the warm water bath until the temperature of the mixture reaches a temperature in the 30–35°C range. Record the actual temperature of the test-tube contents in the blank in Table 4.
    c. Add 2 drops of the enzyme solution to Test Tube T 30–35. Repeat Steps 7–11.

17. Measure the enzyme activity at 50–55°C:
    a. Prepare a water bath at a temperature in the range of 50–55°C by placing hot water in a 600 mL beaker (hot tap water will probably work fine). Check that the temperature remains in this range throughout this test.
    b. Place Test Tube T 50–55 in the warm water bath until the temperature of the mixture reaches a temperature in the 50–55°C range. Record the actual temperature of the test-tube contents in the blank in Table 4.
    c. Add 2 drops of the enzyme solution to Test Tube T 50–55. Repeat Steps 7–11.

18. Measure the enzyme activity at 20–25°C (room temperature):
    a. Record the temperature of Test Tube T 20–25 in Table 4.
    b. In the tube labeled T 20–25, add 2 drops of the enzyme solution. Repeat Steps 7–11.

Part III Testing the Effect of pH

19. Place three clean test tubes in a rack and label them pH 4, pH 7, and pH 10.

20. Add 3 mL of 3% H_2O_2 and 3 mL of each pH buffer to each test tube, as in Table 3.

Table 3		
pH of buffer	Volume of 3% H_2O_2 (mL)	Volume of buffer (mL)
pH 4	3	3
pH 7	3	3
pH 10	3	3

21. In the tube labeled pH 4, add 2 drops of the enzyme solution. Repeat Steps 7–11.

22. In the tube labeled pH 7, add 2 drops of the enzyme solution. Repeat Steps 7–11.

23. In the tube labeled pH 10, add 2 drops of the enzyme solution. Repeat Steps 7–11.

DATA

Table 4	
Test tube label	Slope, or rate (kPa/min)
1 drop	
2 drops	
3 drops	
4 drops	
0–5°C range: _____ °C	
20–25°C range: _____ °C	
30–35°C range: _____ °C	
50–55°C range: _____ °C	
pH 4	
pH 7	
pH 10	

PROCESSING THE DATA

Enzyme concentration plot

1. On Page 2 of this experiment file, create a graph of the rate of enzyme activity *vs.* enzyme concentration. The rate values should be plotted on the y-axis, and the number of drops of enzyme on the x-axis. The rate values are the same as the slope values in Table 4.

Temperature plot

2. On Page 3 of this experiment file, create a graph of the rate of enzyme activity *vs*. temperature. The rate values should be plotted on the y-axis, and the temperature on the x-axis. The rate values are the same as the slope values in Table 4.

pH plot

3. On Page 4 of this experiment file, create a graph of rate of enzyme activity *vs*. pH. The rate values should be plotted on the y-axis, and the pH on the x-axis. The rate values are the same as the slope values in Table 4.

QUESTIONS

Part I Effect of Enzyme Concentration

1. How does changing the concentration of enzyme affect the rate of decomposition of H_2O_2?

2. What do you think will happen to the rate of reaction if the concentration of enzyme is increased to five drops? Predict what the rate would be for 5 drops.

Part II Effect of Temperature

3. At what temperature is the rate of enzyme activity the highest? Lowest? Explain.

4. How does changing the temperature affect the rate of enzyme activity? Does this follow a pattern you anticipated?

5. Why might the enzyme activity decrease at very high temperatures?

Part III Effect of pH

6. At what pH is the rate of enzyme activity the highest? Lowest?

7. How does changing the pH affect the rate of enzyme activity? Does this follow a pattern you anticipated?

EXTENSIONS

1. Different organisms often live in very different habitats. Design a series of experiments to investigate how different types of organisms might affect the rate of enzyme activity. Consider testing a plant, an animal, and a protist.

2. Presumably, at higher concentrations of H_2O_2, there is a greater chance that an enzyme molecule might collide with H_2O_2. If so, the concentration of H_2O_2 might alter the rate of oxygen production. Design a series of experiments to investigate how differing concentrations of the substrate hydrogen peroxide might affect the rate of enzyme activity.

3. Design an experiment to determine the effect of boiling the catalase on the reaction rate.

4. Explain how environmental factors affect the rate of enzyme-catalyzed reactions.

TEACHER INFORMATION

Enzyme Action: Testing Catalase Activity

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. This experiment may take a single group several lab periods to complete. A good breaking point is after the completion of Step 12, when students have tested the effect of different enzyme concentrations. Alternatively, if time is limited, different groups can be assigned one of the three tests and the data can be shared.

3. Your hot tap water may be in the range of 50–55°C for the hot-water bath. If not, you may want to supply pre-warmed temperature baths for Step 17, where students need to maintain very warm water. Warn students not to touch the hot water.

4. Many different organisms may be used as a source of catalase in this experiment. If enzymes from an animal, a protist, and a plant are used by different teams in the same class, it will be possible to compare the similarities and differences among those organisms. Often, either beef liver, beef blood, or living yeast are used.

5. To prepare the yeast solution, dissolve 7 g (1 package) of dried yeast per 100 mL of 2% glucose solution. Incubate the suspension in 37–40°C water for at least 10 minutes to activate the yeast. Test the experiment before the students begin. The yeast may need to be diluted if the reaction occurs too rapidly. The reaction in Step 12, with 3 mL of 3% hydrogen peroxide, 3 mL of water, and 2 drops of suspension should produce a pressure of 1.3 atmospheres in 40 to 60 seconds.

   To prepare a 2% sugar solution, add 20 grams of sugar to make one liter of solution (100 mL per group is needed).

6. To prepare a liver suspension, homogenize 0.5 to 1.5 g of beef liver in 100 mL of cold water. You will need to test the suspension before use, as its activity varies greatly depending on its freshness. Dilute the suspension until the reaction in Step 12, with 3 mL of 3% hydrogen peroxide, 3 mL of water, and 2 drops of suspension produces a pressure of 130 kPa in 40 to 60 seconds. The color of the suspension will be a faint pink. Keep the suspension on ice until used in an experiment.

7. You can purchase 3% H_2O_2 from any supermarket. If refrigerated, bring it to room temperature before starting the experiment.

8. Emphasize to your students the importance of providing an airtight fit with all plastic-tubing connections and when closing valves or twisting the stopper into a test tube.

9. The accessory items used in this experiment are the #1 single hole stopper fitted with a tapered valve connector and the section of plastic tubing fitted with Luer-lock connectors.

10. The length of plastic tubing connecting the rubber stopper assemblies to each gas pressure sensor must be the same for all groups. It is best to keep the length of tubing reasonably small to keep the volume of gas in the test tube low. **Note:** If pressure changes during data collection are too small, you may need to decrease the total gas volume in the system. Shortening the length of tubing used will help to decrease the volume.

11. If the Vernier Gas Pressure Sensor or Biology Gas Pressure Sensor is unavailable, the Vernier Pressure Sensor may be used as an alternative.

12. Vernier Software sells a pH buffer package for preparing buffer solutions with pH values of 4, 7, and 10 (order code PHB). Simply add the capsule contents to 100 mL of distilled water.

13. You can also prepare pH buffers using the following recipes:
    - pH 4: Add 2.0 mL of 0.1 M HCl to 1000 mL of 0.1 M potassium hydrogen phthalate.
    - pH 7: Add 582 mL of 0.1 M NaOH to 1000 mL of 0.1 M potassium dihydrogen phosphate.
    - pH 10: Add 214 mL of 0.1 M NaOH to 1000 mL of 0.05 M sodium bicarbonate.

14. You may need to let students know that at pH values above 10 enzymes will become denatured and the rate of activity will drop. If you have pH buffers higher than 10, have students perform an experimental run using them.

SAMPLE RESULTS

Table 5

Test tube label	Slope or rate (kPa/min)
1 drop	10.23
2 drops	44.98
3 drops	59.36
4 drops	98.26
0–5 °C range: 4°C	41.43
20–25 °C range: 21°C	48.02
30–35 °C range: 34°C	73.85
50–55 °C range: 51°C	27.55
pH 4	36.57
pH 7	66.86
pH 10	75.27

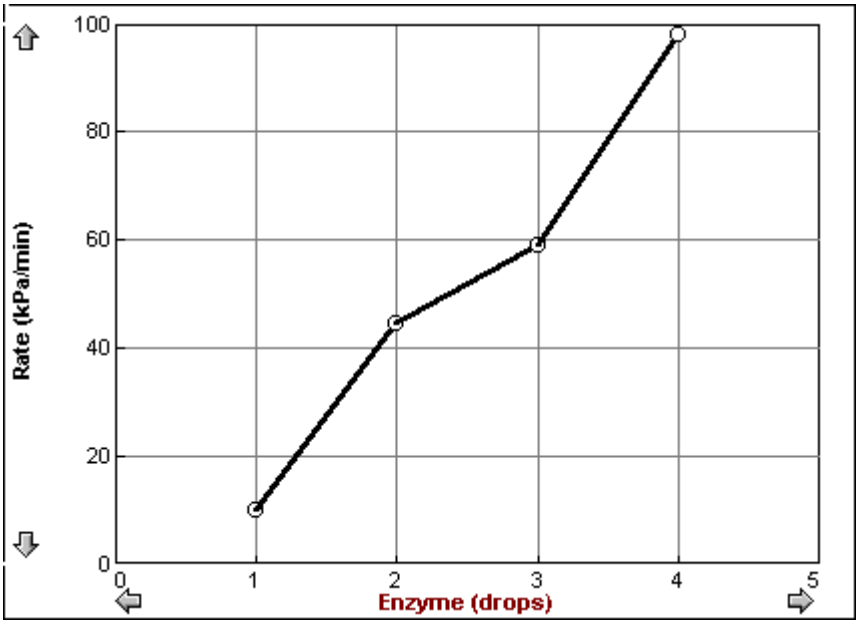


Figure 1: The effect of enzyme concentration on the rate of activity.

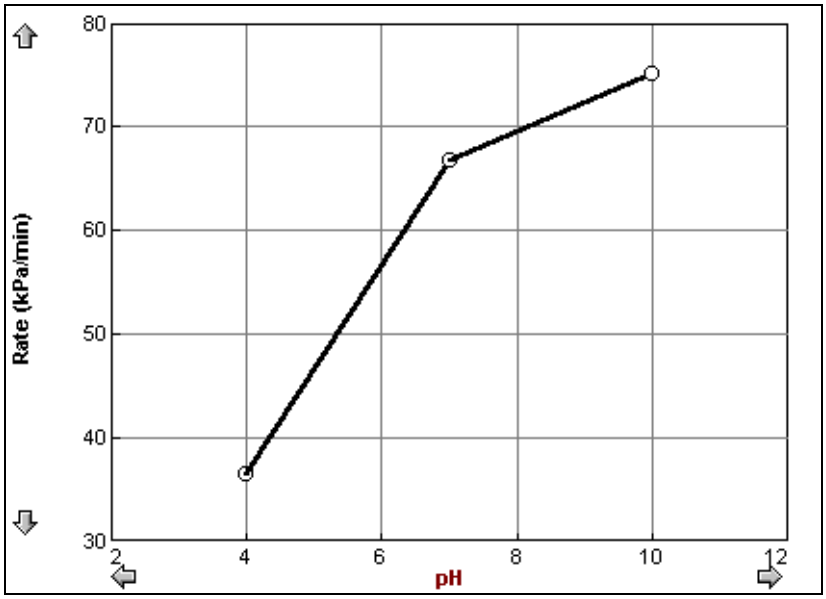


Figure 2: The effect of pH on the rate of enzyme activity

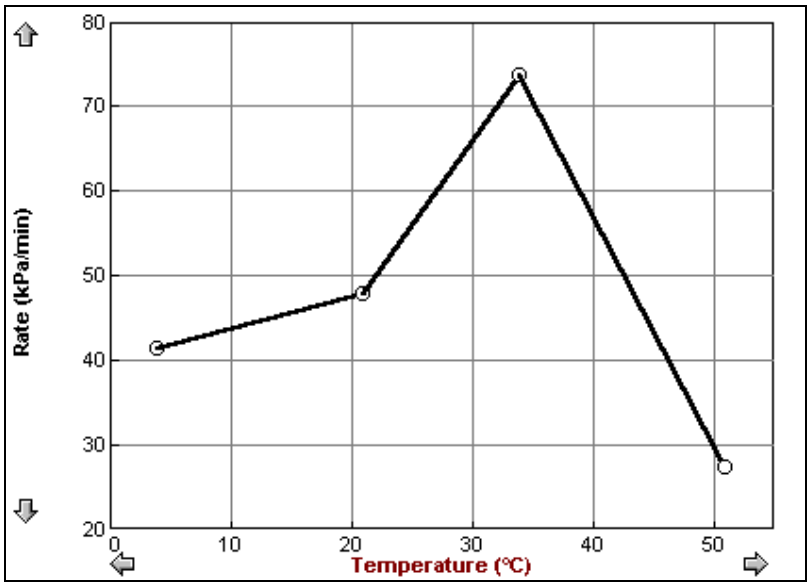


Figure 3: The effect of temperature on the rate of enzyme activity

ANSWERS TO QUESTIONS

1. The rate should be highest when the concentration of enzyme is highest. With higher concentration of enzyme, there is a greater chance of an effective collision between the enzyme and H_2O_2 molecule.

2. Roughly, the rate doubles when the concentration of enzyme doubles. Since the data are somewhat linear, the rate is proportional to the concentration. At a concentration of 5 drops, the rate in the above experiment should be about 111 kPa/min.

3. The temperature at which the rate of enzyme activity is the highest should be close to 30°C. The lowest rate of enzyme activity should be at 60°C.

4. The rate increases as the temperature increases, until the temperature reaches about 50°C. Above this temperature, the rate decreases.

5. At high temperatures, enzymes lose activity as they are denatured.

6. Student answers may vary. Activity is usually highest at pH 10 and lowest at pH 4.

7. Student answers may vary. Usually, the enzyme activity increases from pH 4 to 10. At low pH values, the protein may denature or change its structure. This may affect the enzyme's ability to recognize a substrate or it may alter its polarity within a cell.

Lactase Action

Lactose, a disaccharide sugar found naturally in mammalian milk, is utilized by infants as one of their initial sources of energy. During infancy, mother's milk is often the child's sole source of nutrition. This milk sugar, lactose, must undergo an enzymatic reaction that separates the disaccharide molecule into two monosaccharides; glucose and galactose. This action is carried out in the cells lining the small intestine. The enzyme facilitating the 'breakage' reaction is called lactase. After the split, the resulting simple sugar molecules are released and the lactase enzyme is available to react again. Glucose molecules are absorbed and transported to the liver while galactose molecules undergo another enzymatic reaction converting them to glucose.

Human utilization of milk as a food source varies across the globe. The adaptive production of sufficient lactase is a trait expressed in cultures that relied on dairy products over the generations. People from cultures lacking reliance on dairy products are prone to lactose intolerance, missing the level of lactase production necessary to metabolize the lactose molecule from milk. When dietary lactose escapes lactase action, the molecule proceeds to the large intestine where it is subjected to bacterial fermentation. As increased amounts of lactose pass through the small intestine without conversion, anaerobic bacteria in the colon increase fermentative gas production and discomfort, typical symptoms of lactose intolerance.

In this lab, you will assess the functioning of lactase. One way is to determine if the enzyme is converting the disaccharide into glucose and galactose by measuring the amount of glucose produced. You can use glucose test strips, originally made for diabetics to detect glucose levels. The test strip turns a range of colors to indicate the sugar's concentration in solution.

An alternative test for lactase activity measures the production of CO_2 gas by yeast. Presumably, yeast are unable digest lactose. Yeast metabolize glucose aerobically during respiration, according to the equation:

$$C_6H_{l2}O_6 + 6\ O_2 \rightarrow 6\ CO_2 + 6\ H_2O + \text{energy}$$

water is produced and CO_2 is released as the sugar is broken down in glycolysis. By monitoring the production of CO_2, we can use yeast to indicate lactase activity.

OBJECTIVES

In this experiment, you will

- Test the action of lactase.
- Use glucose test paper to monitor the presence of glucose.
- Determine if yeast can metabolize glucose, lactose, or galactose.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier CO_2 Gas Sensor
lactase solution
5% galactose solution
5% glucose solution
5% lactose solution
10 mL graduated cylinder
600 mL beaker (or water bath)
pipet
three 18 × 150 mm test tubes
hot and cold water
stopwatch
Tes-Tape or other glucose test paper
test-tube rack
thermometer
yeast suspension

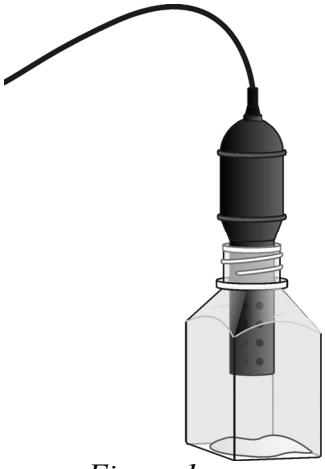


Figure 1

PROCEDURE

Testing for the Production of Glucose

1. You will determine if lactase can produce glucose from the conversion of lactose.
2. Obtain two test tubes and label them 1 and 2.

Table 1

Test tube	Lactose sugar solution (mL)	Lactase
1	2.5	2 drops
2	2.5	none

3. Obtain the lactose sugar solution. Add 2.5 mL of the sugar solution to both test tubes, as listed in Table 1. **Note:** Do not add the lactase to the test tubes until Step 7.

4. Prepare a water bath for the sugar solutions. The water level in the tank should cover 3/4 or more of the test tube while maintaining a temperature between 35°C and 37°C. If a water bath is not available, combine some warm and cool water in the 600 mL beaker to establish and maintain a 35°C–37°C bath. If you need to add more hot or cold water to maintain a constant temperature, first remove about as much water as you will be adding, or the beaker may overflow. Use a basting bulb or a Beral pipet to remove excess water.
5. Measure the glucose concentration.
   a. If the test paper is supplied in a continuous strip, tear off a small piece (0.5 cm) of glucose test paper. Otherwise, obtain one test strip.
   b. Using a dropper pipet, withdraw a drop or two of sugar solution from test tube 1.
   c. Place one drop of sugar solution onto the glucose test paper.
   d. Follow the instructions on the glucose test paper package to develop the test paper. This usually requires a 30 or 60 second wait before you compare the color of the tape to the supplied color chart.
   e. Record the approximate concentration of glucose in Table 3.
   f. Discard any sugar solution remaining in the dropper. Rinse the dropper by taking up clean water and expelling it into a waste beaker.
6. Repeat Step 5 for the second test tube.
7. Add lactase to test tube 1.
   a. Place 2 drops of lactase into test tube 1.
   b. Gently mix the contents of the tube.
8. Set both tubes in the water bath. Start the stopwatch. Be sure to keep the temperature of the water bath in the 35°C–37°C range.
9. Incubate the test tubes for 10 minutes, taking a glucose test once a minute for 10 minutes. Repeat Step 5 and record the concentrations of glucose in Table 3 once every minute.

Testing for the Ability of Yeast to Metabolize Sugars

10. If your CO_2 Gas Sensor has a switch, set it to the Low (0–10,000 ppm) setting. Connect the Vernier CO_2 Gas Sensor to the Vernier computer interface.
11. Prepare the computer for data collection by opening the file "18A Lactase Act (CO2)" from the *Agricultural Science with Vernier* folder of Logger*Pro*.
12. Check the water bath so it remains at a constant temperature range between 35°C and 37°C.
13. You will perform one of the five tests outlined in Table 2 and obtain the results of the other tests from your classmates. Your instructor will assign the test you will be performing. Record the test number in Table 4.

Table 2			
Test	Sugar	Yeast	Enzyme
1	2.5 mL lactose	2.5 mL	8 drops lactase
2	2.5 mL lactose	2.5 mL	none
3	2.5 mL glucose	2.5 mL	none
4	2.5 mL galactose	2.5 mL	none
5	None (water only)	2.5 mL	none

14. Prepare the sugar/yeast solution.
    a. Place 2.5 mL of the assigned solution into a clean test tube.
    b. Obtain the yeast suspension. Gently swirl the yeast suspension to mix the yeast that settles to the bottom. Add 2.5 mL of yeast to the test tube and mix the solution.
15. Transfer all of the sugar/yeast solution in the test tube to the 250 mL respiration chamber.
16. Quickly place the shaft of the CO_2 Gas Sensor in the opening of the respiration chamber.
17. Click Collect to begin data collection.
18. Data collection will end after 4 minutes. Remove the CO_2 Gas Sensor from the respiration chamber.

PROCESSING THE DATA

Determine the *rate* of respiration. The rate of respiration can be measured by examining the *slope* of the pressure *vs*. time plot for each test.

1. Determine the rate of respiration:
    a. Move the mouse pointer to the point where the CO_2 concentration begins to increase linearly. Hold down the mouse button. Drag the pointer to the point where the concentration begins to level off and release the mouse button.
    b. Click the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
    c. Record the slope of the line, *m*, as the respiration rate in Table 4.
    d. Close the linear regression floating box.
    e. Share your data with other classmates by recording the test tube number and the respiration rate on the board.
2. Print the graph of CO_2 concentration *vs*. time if directed to do so by your instructor.
3. Average the rate values for each of the four tests performed by the class and record them in Table 5.
4. On Page 2 of the experiment file, make a graph of respiration rate *vs.* sugar/enzyme combination.

5. Fill the respiration chamber with water and then empty it. Make sure that all yeast have been removed from the respiration chamber. Thoroughly dry the inside of the respiration chamber with a paper towel.

6. If time permits, repeat Steps 13–18 for another one of the solutions.

7. On Page 2 of the experiment file, make a graph of respiration rate *vs.* sugar/enzyme combination.

DATA

Table 3: Glucose Concentrations		
Time (minutes)	Lactose + Lactase	Lactose only
0		
1		
2		
3		
4		
5		
6		
7		
8		
9		
10		

Table 4: Your Results	
Test	Respiration rate (kPa/min)

Table 5		
Test	Type of sugar / enzyme	Respiration rate (ppm/min) average
1	Lactose + Lactase	
2	Lactose only	
3	Glucose	
4	Galactose	
5	None (water only)	

QUESTIONS

1. From the results of this experiment, how does lactase function? What is your evidence?
2. Considering the results of this experiment, can yeast utilize all of the sugars equally well? Explain.
3. Hypothesize why some sugars are not utilized by yeast while other sugars are metabolized.
4. How did the results of testing lactase's activity using glucose test paper compare with the results of using yeast as an indicator of activity? What is your evidence?
5. Which test tube served as a control in this experiment? What did you conclude from the control? How did this affect the interpretation of data in this experiment?

EXTENSIONS

1. Design an experiment to test the activity of Beano on the sugar melibiose. Does Beano have any effect on the sugar lactose? From the results of this experiment, how does Beano function? What is your evidence?
2. Design an experiment to test whether Beano has any effect on the sugar lactose.
3. Design and carry out an experiment to determine the optimal pH range of activity for lactase.
4. Design and carry out an experiment to determine the functional relationship between rate and lactase concentration.

TEACHER INFORMATION

Lactase Action

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Lactaid Fast Act caplets work well as a source for lactase and are available in most drugstores or the pharmacy section of supermarkets. Each caplet contains 9000 FCC Lactase Units. These tablets are time sensitive and may exhibit diminished action if used after their expiration date. **Note:** An FCC Lactase Unit is not the same as an enzyme unit (U) or a katal.
3. To prepare the lactase solution from Lactaid Fast Act caplets:
   a. Grind and pulverize one Lactaid Fast Act caplet with a mortar and pestle.
   b. Scrape the powder into a 50 mL sealable container.
   c. Add 20 mL of distilled water to the container with the powdered caplet.
   d. Agitate thoroughly until the powder is fully incorporated into the solution.
   e. Refrigerate the solution until it is needed and then store it in an ice bath when being used. The refrigerated solution retains activity for several days.
   f. The lactase solution can be filtered, if desired

   Under this formulation, 1 mL will contain approximately 450 FCC Lactase Units and 1 drop will contain approximately 18 FCC Lactase Units.
4. Another form of lactase is β -Galactosidase derived from Kluyveromyces lactis. This liquid formulation is ready to use and requires refrigeration. It can be ordered from Sigma-Aldrich, www.sigmaaldrich.com, order code G3665.
5. In this experiment, students will test the activity of lactase in two ways:
   - The presence of glucose using glucose test strips.
   - The ability of yeast to respire aerobically using the glucose that is enzymatically released. Yeast will not utilize lactose or galactose. They will, however, utilize glucose. Lactose releases glucose into solution when the disaccharide is converted by the lactase enzyme.
6. The stored calibration works well for this experiment.
7. Glucose test strips used by diabetics may be obtained at most drug stores or through most biological suppliers. Check that the strips are ones designed for urinalysis, not blood. These tablets are time sensitive and may exhibit diminished action if used after their expiration date. The resolution of the glucose test paper is not adequate for students to use for enzyme kinetic studies. Since the wet dye on the glucose test paper may stain desks, supply students with a container to discard used strips.
8. Use a glass Pasteur or a plastic pipette as the dropper. It should be long enough to easily withdraw a few drops of sugar solution from the test tube.
9. To prepare the necessary sugar solutions, add 5 g of sugar to enough distilled water to bring the solution to 100 mL.

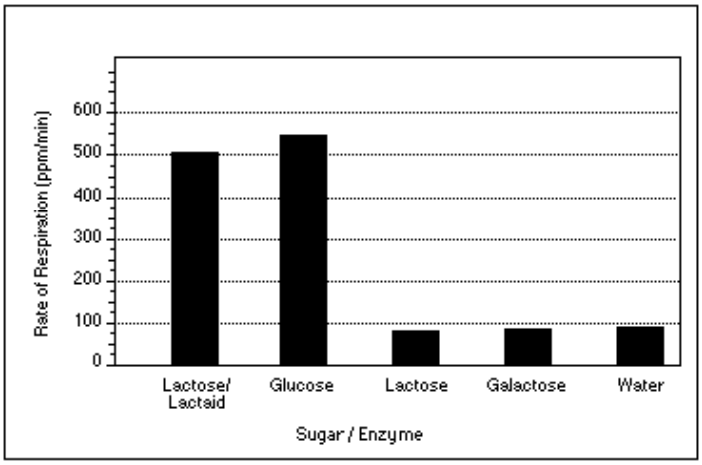
10. To prepare the yeast stock solution, dissolve 7 g (1 package) of dried yeast per 100 mL of water. Maintain the yeast suspension in a water bath at a temperature between 35–40° C. After an initiation period of ten minutes, students can transfer 3 mL of the yeast suspension to a tube that will serve as their groups yeast source. **Note:** If pressure changes during data collection are too small, you may need to increase the amount of yeast suspension being used in each trial.
11. The CO_2 Gas Sensor is dependent on the diffusion of gases into the probe shaft. Students should allow a couple of minutes between trials so that gases from the previous trial will have diffused out of the probe shaft. Alternatively, the students can use a firm object such as a book or notepad to fan air through the probe shaft.
12. The stopper included with the older-style CO_2 Gas Sensor is slit to allow it to be easily added or removed from the probe. When students are placing the probe in the respiration chamber, they should gently twist the stopper into the chamber opening. Warn the students not to twist the probe shaft or they may damage the sensing unit.
13. To conserve battery power, we suggest that AC Adapters be used to power the interfaces rather than batteries when working with the older-style CO_2 Gas Sensor.

SAMPLE RESULTS

The following data may be different from students' results. The actual values depend upon the viability and concentration of the yeast, among other factors.

Table 4

Test	Type of Sugar / Enzyme	Rate of Respiration (ppm/min)
1	Lactose + Lactase	500.99
2	Lactose	88.32
3	Glucose	538.56
4	Galactose	92.76
5	None (water only)	95.68



ANSWERS TO QUESTIONS

1. Lactase acts by converting lactose into glucose and, presumably, galactose. Yeast are not able to metabolize lactose, but can metabolize glucose. Yeast are able to use the lactose solution only *after* it has been acted upon by lactase. Since lactose is a disaccharide that can hydrolyze into glucose and galactose, lactase must have caused that hydrolysis.
2. Yeast cannot utilize all of the sugars equally well. Glucose was metabolized much more efficiently than lactose or galactose, as shown by their rates of respiration..
3. Yeast may not have the proper enzymes to transport certain sugars across its cell membrane, or it may not have the enzyme needed to convert sugar from disaccharides to monosaccharides.
4. The results of the two experiments should agree. The glucose test tape experiment indicated the presence of glucose with lactase + lactose. The yeast were able to respire with the products of lactase + lactose. Since glucose was the only sugar utilized by yeast in this experiment, one of the products of enzymatic activity presumably was glucose.
5. Test 5 served as the control. It contained only water and yeast in order to verify that the yeast need sugar to respire. Without the control, a person could conclude that the yeast can respire without any sugar.

Lactase Action

Lactose, a disaccharide sugar found naturally in mammalian milk, is utilized by infants as one of their initial sources of energy. During infancy, mother's milk is often the child's sole source of nutrition. This milk sugar, lactose, must undergo an enzymatic reaction that separates the disaccharide molecule into two monosaccharides; glucose and galactose. This action is carried out in the cells lining the small intestine. The enzyme facilitating the 'breakage' reaction is called lactase. After the split, the resulting simple sugar molecules are released and the lactase enzyme is available to react again. Glucose molecules are absorbed and transported to the liver while galactose molecules undergo another enzymatic reaction converting them to glucose.

Human utilization of milk as a food source varies across the globe. The adaptive production of sufficient lactase is a trait expressed in cultures that relied on dairy products over the generations. People from cultures lacking reliance on dairy products are prone to lactose intolerance, missing the level of lactase production necessary to metabolize the lactose molecule from milk. When dietary lactose escapes lactase action, the molecule proceeds to the large intestine where it is subjected to bacterial fermentation. As increased amounts of lactose pass through the small intestine without conversion, anaerobic bacteria in the colon increase fermentative gas production and discomfort, typical symptoms of lactose intolerance.

In this lab, you will assess the functioning of lactase. One way is to determine if the enzyme is converting the disaccharide into glucose and galactose by measuring the amount of glucose produced. You can use glucose test strips, originally made for diabetics to detect glucose levels. The test strip turns a range of colors to indicate the sugar's concentration in solution.

An alternative test for lactase activity measures the production of CO_2 gas by yeast. Presumably, yeast are unable digest lactose direclty. Yeast metabolize glucose anaerobically during fermentation, according to the equation:

$$C_6H_{l2}O_6 \rightarrow 2\ CH_3CH_2OH + 2\ CO_2 + \text{energy}$$

ethanol is produced and CO_2 is released as the sugar breaks down in glycolysis. By monitoring pressure change caused by the production of CO_2, we can use yeast to indicate lactase activity.

OBJECTIVES

In this experiment, you will

- Test the action of lactase.
- Use glucose test paper to monitor the presence of glucose.
- Determine if yeast can metabolize glucose, lactose, or galactose.

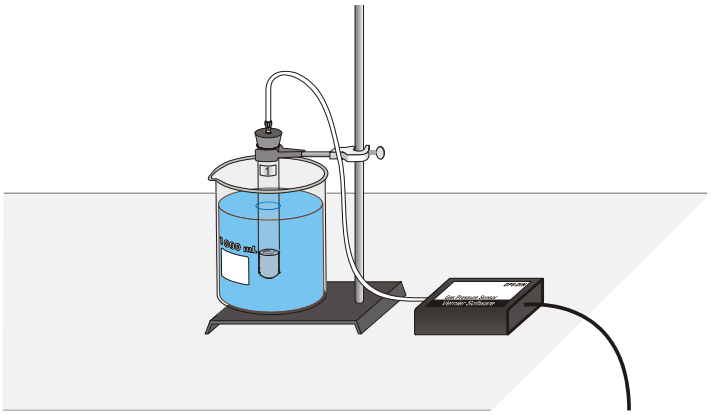


Figure 1

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier Gas Pressure Sensor
1-hole rubber stopper and tubing assembly
lactase solution
5% galactose solution
5% glucose solution
5% lactose solution
10 mL graduated cylinder
600 mL beaker (for water bath)
dropper or Beral pipet
cooking oil (canola, olive, or corn)
three 18 × 150 mm test tubes
hot and cold water or a water bath
stopwatch
Tes-Tape or other glucose test paper
test-tube rack
thermometer
yeast suspension

PROCEDURE

Testing for the Production of Glucose

1. You will determine if lactase produces glucose during the conversion of lactose.
2. Obtain two test tubes and label them test tube 1 and test tube 2.

Table 1

Test tube	Lactose sugar solution (mL)	Lactase
1	2.5	2 drops
2	2.5	none

3. Obtain the lactose sugar solution. Add 2.5 mL of the sugar solution to both test tubes, as listed in Table 1. **Note:** Do not add the lactase to the test tube until Step 7.
4. Prepare a water bath for the sugar solutions. The water level in the tank should cover 3/4 or more of the test tube while maintaining a temperature between 35°C and 37°C. If a water bath is not available, combine some warm and cool water in the 600 mL beaker to establish and maintain a 35°C–37°C bath. If you need to add more hot or cold water to maintain a constant temperature, first remove about as much water as you will be adding, or the beaker may overflow. Use a basting bulb or a Beral pipet to remove excess water.

5. Measure the glucose concentration.
   a. If the test paper is supplied in a continuous strip, tear off a small piece (0.5 cm) of glucose test paper. Otherwise, obtain one test strip.
   b. Using a clean pipette, withdraw a drop or two of sugar solution from test tube 1.
   c. Place one drop of sugar solution onto the glucose test paper.
   d. Follow the instructions on the glucose test paper package to develop the test paper. **Note:** This usually requires a 30 or 60 second wait before you compare the color of the tape to the supplied color chart.
   e. Record the approximate concentration of glucose in Table 3.
   f. Discard any sugar solution remaining in the dropper. Rinse the pipette by taking up clean water and expelling it into a waste beaker.
6. Repeat Step 5 for the second test tube.
7. Add lactase to test tube 1.
   a. Place 2 drops of lactase solution into test tube 1 only.
   b. *Gently* mix the contents of the tube.
8. Set both tubes in the water bath. Start the stopwatch. Be sure to keep the temperature of the water bath in the 35°C–37°C range.
9. Incubate the test tubes for 10 minutes, taking a glucose test once every minute for 10 minutes. Repeat Step 5 and record the concentrations of glucose in Table 3 once every minute.

Testing for the Ability of Yeast to Ferment Sugars

10. Connect the Gas Pressure Sensor to the computer interface. Prepare the computer for data collection by opening the file "18B Lactase Act (Press)" from the *Agricultural Science with Vernier* folder of Logger *Pro*.
11. Connect the plastic tubing to the valve on the Gas Pressure Sensor.
12. Check the water bath so it remains at a constant temperature range between 35°C and 37°C.
13. You will perform one of the five tests outlined in Table 2 and obtain the results of the other tests from your classmates. Your instructor will assign the test you will be performing. Record the test number in Table 4.

Table 2

Test	Sugar	Yeast	Enzyme
1	2.5 mL lactose	2.5 mL	8 drops lactase
2	2.5 mL lactose	2.5 mL	none
3	2.5 mL glucose	2.5 mL	none
4	2.5 mL galactose	2.5 mL	none
5	none (water only)	2.5 mL	none

14. Prepare the sugar/yeast solution:
    a. Place 2.5 mL of the assigned solution into a clean test tube.
    b. Obtain the yeast suspension. Gently swirl the yeast suspension to mix the yeast that settles to the bottom. Add 2.5 mL of yeast to the test tube and mix the solution.
15. Carefully add 1 mL of oil on top of the yeast/sugar mixture as shown in Figure 2. This will serve to seal the mixture from free oxygen. Be careful to not get oil on the inside wall of the test tube.
16. Set the test tube in the water bath.
17. Insert the single-holed rubber-stopper into the test tube. **Note**: *Firmly* twist the stopper for an *airtight* fit. Secure the test tube with a utility clamp and ring-stand as shown in Figure 1.

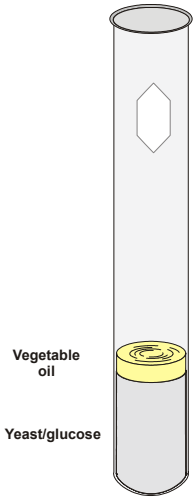


Figure 2

18. Connect the free end of the plastic tubing to the connector on the rubber stopper as shown in Figure 3. **Note:** Make sure that the test tube is at submerged at least 3/4 of the way in the water bath. The temperature of the air in the tube must be constant.
19. Click Collect to begin collecting pressure data. Maintain the temperature of the water bath during the course of the experiment.
20. Data collection will end after 15 minutes. Monitor the pressure readings displayed on the screen. If the pressure exceeds 135 kilopascals, the pressure inside the tube will be too great and the rubber stopper is likely to pop off. Disconnect the plastic tubing from the Gas Pressure Sensor if the pressure exceeds 135 kilopascals.
21. When data collection has finished, disconnect the plastic tubing connector from the rubber stopper. Remove the rubber stopper from the test tube, discard the contents in a waste beaker, and clean the test tube.

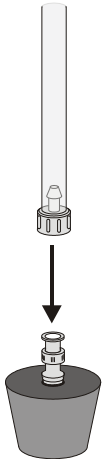


Figure 3

PROCESSING THE DATA

Determine the *rate* of fermentation. The rate of fermentation can be measured by examining the *slope* of the pressure *vs*. time plot for each test.

1. Find the rate of fermentation:
    a. Move the mouse pointer to the point where the pressure values begin to increase linearly. Hold down the mouse button. Drag the pointer to the point where the pressure begins to level off and release the mouse button.
    b. Click the Linear Fit button, , to perform a linear regression. A floating box will appear with the formula for a best fit line.
    c. Record the slope of the line, *m*, as the fermentation rate in Table 4.
    d. Share your data with other classmates by recording the test tube number and the fermentation rate.
2. Print the graph of pressure *vs*. time if directed to do so by your instructor.
3. Average the rate values for each of the four tests performed by the class and record them in Table 5.

4. On Page 2 of the experiment file, make a graph of fermentation rate *vs.* sugar/enzyme combination.

5. If time permits, repeat Steps 14–21 for another one of the solutions.

DATA

Table 3: Glucose Concentrations		
Time (minutes)	Lactose + Lactase	Lactose only
0		
1		
2		
3		
4		
5		
6		
7		
8		
9		
10		

Table 4: Your Results	
Test	Fermentation rate (kPa/min)

Table 5		
Test	Type of sugar / enzyme	Fermentation rate (kPa/min) Average
1	Lactose + Lactase	
2	Lactose only	
3	Glucose	
4	Galactose	
5	None (water only)	

QUESTIONS

1. From the results of this experiment, how does lactase function? What is your evidence?
2. Considering the results of this experiment, can yeast utilize all of the sugars equally well? Explain.
3. Hypothesize why some sugars are not metabolized by yeast while other sugars are.
4. How did the results of testing lactase's activity using glucose test paper compare with those using yeast as an indicator of activity? What is your evidence?
5. Which test tube served as a control in this experiment? What did you conclude from the control? How did the control affect the interpretation of data in this experiment?

EXTENSIONS

1. Design an experiment to test the activity of Beano® on the sugar melibiose. Does Beano have any effect on the sugar lactose? From the results of this experiment, how does Beano function? What is your evidence?
2. Design an experiment to test whether Beano has any effect on the sugar lactose.
3. Design and carry out an experiment to determine the optimal pH range of activity for lactase.
4. Design and carry out an experiment to determine the functional relationship between rate and lactase concentration.

TEACHER INFORMATION

Lactase Action

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. Lactaid Fast Act caplets work well as a source for lactase and are available in most drugstores or the pharmacy section of supermarkets. Each caplet contains 9000 FCC Lactase Units. These tablets are time sensitive and may exhibit diminished action if used after their expiration date. **Note:** An FCC Lactase Unit is not the same as an enzyme unit (U) or a katal.

3. To prepare the lactase solution from Lactaid Fast Act caplets:
   a. Grind and pulverize one Lactaid Fast Act caplet with a mortar and pestle.
   b. Scrape the powder into a 50 mL sealable container.
   c. Add 20 mL of distilled water to the container with the powdered caplet.
   d. Agitate thoroughly until the powder is fully incorporated into the solution.
   e. Refrigerate the solution until it is needed and then store it in an ice bath when being used. The refrigerated solution retains activity for several days.
   f. The lactase solution can be filtered, if desired

   Under this formulation, 1 mL will contain approximately 450 FCC Lactase Units and 1 drop will contain approximately 18 FCC Lactase Units.

4. Another form of lactase is β -Galactosidase derived from Kluyveromyces lactis. This liquid formulation is ready to use and requires refrigeration. It can be ordered from Sigma-Aldrich, www.sigmaaldrich.com, order code G3665.
5. In this experiment, students will test the activity of lactase in two ways:
   - The appearance of glucose using Tes-Tape or other glucose test paper.
   - The ability of yeast to respire anaerobically using the glucose that is enzymatically released. Yeast will not utilize lactose, or galactose. They will, however, utilize glucose. Lactose releases glucose into solution when the disaccharides are converted into monosaccharides enzymatically.

6. If time allows, you may consider having students do a second trial. This would increase the chances that they have at least one trial in which there is a significant pressure change.

7. Glucose test paper, used by diabetics, may be obtained at most drug stores. The resolution of the glucose test paper is not adequate for students to use for enzyme kinetic studies. Since the wet dye on the glucose test paper may stain desks, supply students with a container to discard used strips.

8. Use a glass Pasteur or a plastic Beral pipette for the dropper pipette. It should be long enough to easily withdraw a few drops of sugar solution from the test tube.

9. You will need to assign each group one of the five tests detailed in Table 2. If two or more groups perform the same test and record their values on the board, the class should use the average of these values. If there are sufficient measurements, take the average of the data after the highest and lowest values are discarded.

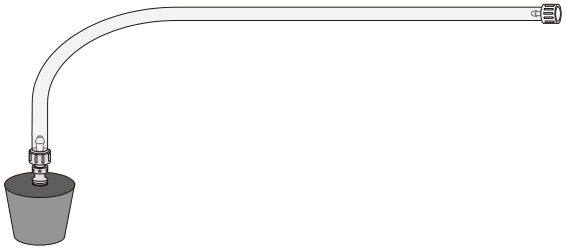
10. Students should record their rate value on the board for other groups to use.

11. To prepare the yeast solution, dissolve 7 g (1 package) of dried yeast per 100 mL of water. Incubate the suspension in 37–40°C water for at least 10 minutes.

12. To prepare the 5% sugar solutions, add 5 g of sugar per 100 mL of solution.

13. After the 10 minute incubation period, transfer the yeast to dispensing tubes. Each group will need about 3 mL of yeast.

14. The accessory items used in this experiment are the #1 single hole stopper fitted with a tapered valve connector and the section of plastic tubing fitted with Luer-lock connectors.



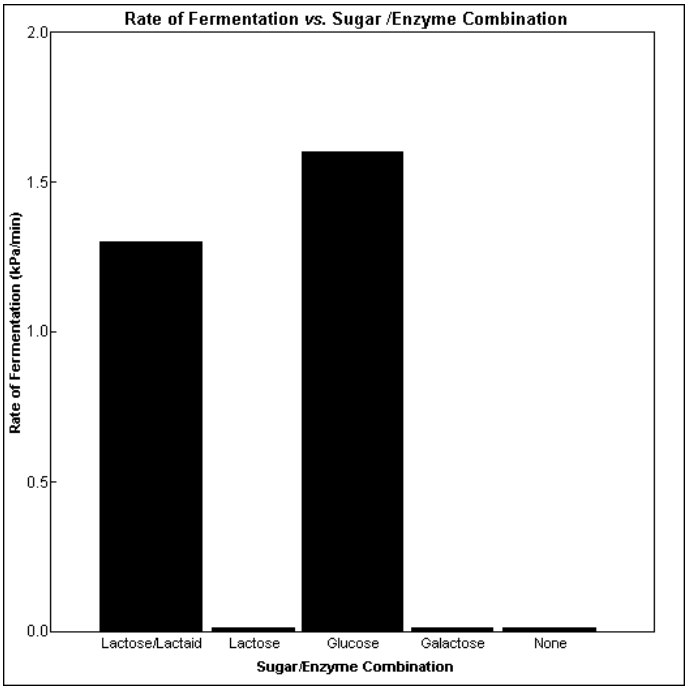
15. The length of plastic tubing connecting the rubber stopper assemblies to each gas pressure sensor must be the same for all groups. It is best to keep the length of tubing reasonably small to keep the volume of gas in the test tube low. **Note:** If pressure changes during data collection are too small, you may need to decrease the total gas volume in the system. Shortening the length of tubing used will help to decrease the volume.

SAMPLE RESULTS

The following data may be different from students' results. The actual values depend upon the viability and concentration of the yeast, among other factors.

Table 5

Test	Type of sugar/enzyme	Rate of fermentation (kPa/min)
1	Lactose + Lactase	1.3
2	Lactose	0
3	Glucose	1.6
4	Galactose	0
5	None (water only)	0



Rate of fermentation values for each test

ANSWERS TO QUESTIONS

1. Lactase acts by converting lactose into glucose and, presumably, galactose. Yeast are not able to metabolize lactose, but can metabolize glucose. Yeast are able to use the lactose solution only *after* it has been acted upon by lactase. Since lactose is a disaccharide that can hydrolyze into glucose and galactose, lactase must have caused that hydrolysis.

2. Yeast cannot utilize all of the sugars equally well. Of all the sugars tested, yeast can only metabolize glucose—not lactose or galactose. The rates of the latter three tests were zero.

3. Yeast may not have the proper enzymes to transport lactose across its cell membrane, or it may not have the enzyme needed to convert it from a disaccharide to a monosaccharide.

4. The results of the two experiments should agree. The glucose test tape experiment indicated the presence of glucose with lactase + lactose. The yeast were able to respire with the products of lactase + lactose. Since glucose was the only sugar utilized by yeast in this experiment, one of the products of enzymatic activity presumably was glucose.

5. Test 5 served as the control. It contained only water and yeast in order to verify that the yeast need sugar to respire. Without the control, a person could conclude that the yeast can respire without any sugar.

Oxygen Gas and Respiration

The process of breathing accomplishes two important tasks for the body. During inhalation, oxygen-rich air is brought into your lungs. During exhalation, air depleted in oxygen and rich in carbon dioxide is forced out. Oxygen is then transported to the cells where it is used in the process of respiration, yielding carbon dioxide as a product.

$$C_6H_{12}O_6 + 6\ O_2 \rightarrow 6\ CO_2 + 6\ H_2O + \text{energy}$$

glucose oxygen carbon dioxide water

Gas exchange takes place in the lungs at the membrane between the alveoli and the pulmonary capillaries. It is here that oxygen diffuses into the bloodstream and carbon dioxide diffuses out. Under normal circumstances, there is an equilibrium between the oxygen and carbon dioxide levels in the blood. Several mechanisms are involved in maintaining this balance. One such mechanism involves chemoreceptors. These specialized cells respond to changes in carbon dioxide, oxygen and H^+ concentrations and influence the body's ventilation patterns to maintain the proper balance of blood gases.

In this experiment, you will determine what factors affect how long you can hold your breath. You will be tested under two different conditions. The first condition is normal breathing. The second condition is immediately following hyperventilation. *Hyperventilation* is when your breathing rate is greater than what is necessary for proper exchange of oxygen and carbon dioxide. This will be achieved by a period of rapid breathing prior to holding your breath.

OBJECTIVES

In this experiment, you will

- Use an O_2 Gas Sensor to determine residual oxygen levels in exhaled air.
- Evaluate how internal O_2 and CO_2 concentrations influence breathing patterns.

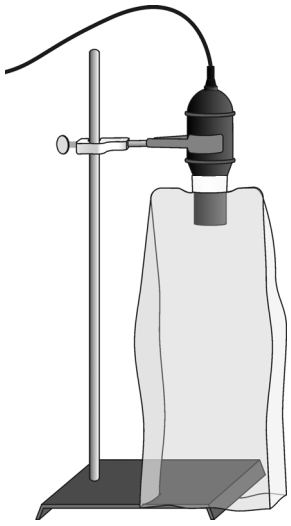


Figure 1

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier O_2 Gas Sensor
ring stand
test tube clamp
bread bag

PRELIMINARY QUESTIONS

1. How long do you think you can hold your breath?
2. When you hold your breath, what do you think happens to the oxygen concentration in your lungs? Explain.
3. When you hold your breath, what do you think happens to the carbon dioxide concentration in your lungs? Explain.
4. On average, people can hold their breath for a minute. What do you think prevents people from holding their breath for 2 or 3 minutes?

PROCEDURE

Each person in a lab group will take turns being the subject and the tester. When it is your turn to be the subject, your partner will be responsible for operating the equipment.

1. Secure the O_2 Gas Sensor using a test tube clamp and ring stand as shown in Figure 1. The plastic bread bag should already be taped to the sensor.
2. Connect the O_2 Gas Sensor to the Vernier computer interface.
3. Prepare the computer for data collection by opening the "19 Oxygen and Respiration" file in the *Agricultural Science with Vernier* folder.
4. When you begin collecting data, it is important that data collection begins at the same point the subject begins to hold his breath.
   a. Have the subject take a deep breath and hold it. Immediately click Collect to begin data collection. The subject should hold his breath as long as possible.
   b. When the subject can no longer hold his breath, he should blow his breath into the bread bag and twist the open end shut. This should result in the bread bag filled with the air the subject was holding in his lungs. Allow data collection to proceed for the full 120 seconds.
   c. When data collection has finished, open the bread bag and pull it back over the sensor exposing the sensor to room air. Leave the bag in that position until you are prepared to collect data again.
   d. Click the Examine button, . The cursor will become a vertical line. As you move the mouse pointer across the screen, the oxygen and time values corresponding to its position will be displayed in the box at the upper-left corner of the graph. Scroll across the data to determine how long the subject held his breath. Record the time in Table 1. Determine the maximum and minimum oxygen concentrations and record them in Table 1. To remove the examine box, click the upper-left corner of the box.
5. Move your data to a stored run. To do this, choose Store Latest Run from the Experiment menu.

6. Collect data following mild hyperventilation.
   a. Pull the bread bag back down off of the sensor in preparation for data collection.
   b. Have the subject take 10 quick deep breaths, forcefully blowing out all air after each breath. The subject should then take an 11th breath and hold it. Immediately click ▶ Collect to begin data collection. The subject should hold his breath as long as possible.
   c. When the subject can no longer hold his breath, he should blow his breath into the bread bag and twist the open end shut. This should result in the bread bag filled with the air the subject was holding in his lungs. Allow data collection to proceed for the full 120 seconds.
   d. When data collection has finished, open the bread bag and pull it back over the sensor exposing the sensor to room air.
   e. Click the Examine button, . The cursor will become a vertical line. As you move the mouse pointer across the screen, the oxygen and time values corresponding to its position will be displayed in the box at the upper-left corner of the graph. Scroll across the data to determine how long the subject held his breath. Record the time in Table 1. Determine the maximum and minimum oxygen concentrations and record them in Table 1. To remove the examine box, click the upper-left corner of the box.
7. Both runs should now be displayed on the same graph. Use the displayed graph and the data in Table 1 to answer the questions below.

DATA

Table 1

	Breath held (s)	Maximum O_2 concentration (%)	Minimum O_2 concentration (%)	Change in oxygen (%)
Normal				
Hyperventilation				

QUESTIONS

1. Did the oxygen concentration change as you expected? If not, explain how it was different.
2. Did the amount of time you held your breath change after hyperventilation (taking the 10 quick breaths)? If so, did the time increase or decrease? Explain.
3. After hyperventilation, was the resulting concentration of oxygen in your exhaled breath higher or lower than in the first attempt? How much did it change? What do you contribute this to?
4. On the first trial, what do you believe forced you to start breathing again?
5. On the second trial, what do you believe forced you to start breathing again?
6. Based on your answers to questions 4 and 5, does the concentration of oxygen or carbon dioxide have a greater influence on how long one can hold his breath?

TEACHER INFORMATION

Oxygen Gas and Respiration

This exercise is meant as an investigative study. Results may vary from those printed here. Variations in results may be due to different body mass, age, fitness, and sex of the students.

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. The best bag to use for this experiment is a bread bag. This is the same bag that comes with a loaf of bread that you purchase from the super market. The advantages to this type of bag are that it is pliable, large, and easy to obtain.

3. To secure the bag to the O_2 Gas Sensor, cut a small hole the size of a half dollar and feed the sensor through the hole. Use tape to seal the bag to the sensor and prevent any air from escaping at that junction. Most any type of tape will work.

4. Once the sensor is mounted on the ring stand with the test tube clamp, students can rotate the sensor enough so that it is pointing more in their direction. This may prove easier to use rather than having the sensor pointing straight down.

5. Students may find it easier to hold their breath if they are facing away from the computer screen.

6. Students with asthma or other respiratory ailments should not participate as the subject in this experiment.

ANSWERS TO PRELIMINARY QUESTIONS

1. Student answers will vary as to how long they think they can hold their breath.

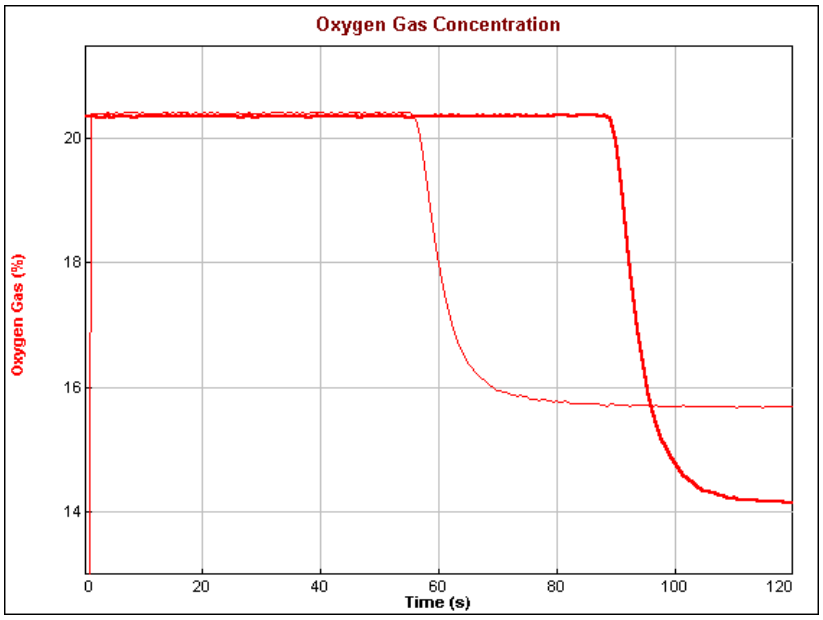
2. Oxygen levels in the lungs are going to drop as more oxygen moves into the bloodstream.

3. Carbon dioxide levels in the lungs are going to increase as more moves out of the bloodstream.

4. The main factor that prevents people from holding their breath longer is the increase in carbon dioxide concentration in the blood. Carbon dioxide, oxygen, and H^+ concentrations all contribute to voluntary control of ventilation. The body is far more sensitive to changes in carbon dioxide concentration than it is to changes in oxygen. Because of this, voluntary control of ventilation is primarily controlled by the concentration of carbon dioxide found in the blood.

SAMPLE RESULTS

Table 1				
	Breath held (s)	Maximum O_2 concentration (%)	Minimum O_2 concentration (%)	Change in oxygen (%)
Normal	55	20.5	15.7	4.8
Hyperventilation	89	20.4	14.2	6.2



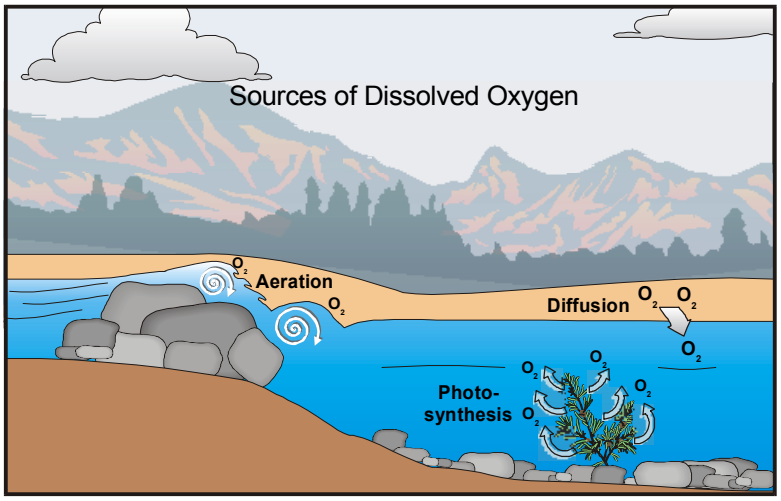
Oxygen concentration data after normal breathing and hyperventilation

ANSWERS TO QUESTIONS

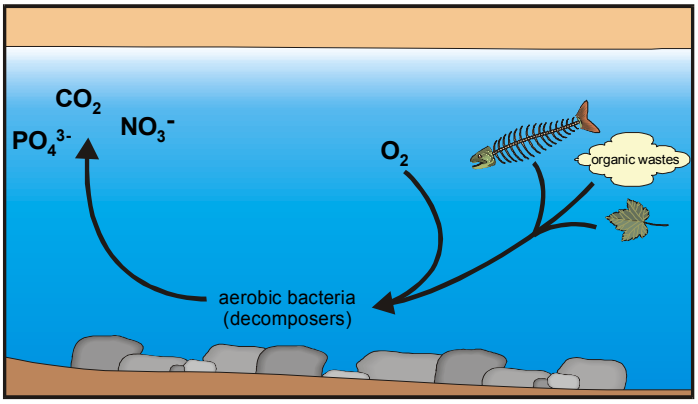
1. Answers vary.
2. During hyperventilation, the carbon dioxide concentration in the body drops. This should result in a person being able to hold their breath significantly longer than normal.
3. After hyperventilation, students will likely find that the oxygen concentration of their expired air was lower than under normal breathing conditions. Since they are able to hold their breath longer, more oxygen will be taken up and less will be expired.
4. Answers vary.
5. Answers vary.
6. Initially, students will believe that they are forced to breath again because they have run out of air. After seeing that they used more air in the second trial when they hyperventilated, they should reason that oxygen was not the determining factor. This should lead them to the conclusion that carbon dioxide is the reason they were forced to breath again.

Biochemical Oxygen Demand

Oxygen available to aquatic organisms is found in the form of *dissolved oxygen*. Oxygen gas is dissolved in a stream through aeration, diffusion from the atmosphere, and photosynthesis of aquatic plants and algae. Plants and animals in the stream consume oxygen in order to produce energy through respiration. In a healthy stream, oxygen is replenished faster than it is used by aquatic organisms. In some streams, aerobic bacteria decompose such a large volume of organic material that oxygen is depleted from the stream faster than it can be replaced. The resulting decrease in dissolved oxygen is known as the *Biochemical Oxygen Demand* (BOD).



When it rains, organic material found in the soil is transported in the rainwater to streams and rivers. Additional organic material accumulates in the stream when aquatic organisms die. Bacteria and other microorganisms decompose this organic material. In a healthy body of water, this process has only a slight impact on dissolved oxygen levels. It serves to release vital nutrients, such as nitrates and phosphates, which stimulate algae and aquatic plant growth. If the amount of decomposing organic material is too high, dissolved oxygen levels can be severely reduced. In a body of water with large amounts of decaying organic material the dissolved oxygen levels may drop by 90%—this would represent a high BOD. In a mountain stream with low levels of decaying organic material, the dissolved oxygen levels may drop by only 10% or 20%—a low BOD.



Organic materials, such as leaves, fallen trees, fish carcasses, and animal waste, end up in the water naturally and are important in the recycling of nutrients throughout the ecosystem. Organic materials that enter the water as a result of human impact can be considered sources of pollution.

Expected Levels

BOD levels are dependent on the body of water being tested. Shallow, slow-moving waters, such as ponds and wetlands, will often have large amounts of organic material in the water and high BOD levels. A water sample from a pond could have an initial dissolved oxygen reading of 9.5 mg/L. After the five-day incubation period, the dissolved oxygen could be down to 1 mg/L

resulting in a high BOD level of 8.5 mg/L. In contrast, a water sample collected from a cold mountain stream with an initial dissolved oxygen reading of 11 mg/L may have decreased to 9 mg/L after incubation, resulting in a BOD of only 2 mg/L. Use Table 1 as a rough guide for the data you gather[1].

Table 1: Interpretation of BOD Levels

BOD Level (mg/L)	Status
1–2 mg/L	Clean water with little organic waster.
3–5 mg/L	Moderately clean water with some organic waste.
6–9 mg/L	Lots of organic material and many bacteria.
>10 mg/L	Very poor water quality. Large amounts of organic material in the water.

Summary of Methods

Included in this test are the procedures for High and Low BOD levels. Decide beforehand, based on expected BOD levels (see Table 1), which procedure is appropriate for the water you are testing. Only one of the two tests should be performed.

Method 1: Low BOD Levels (0–6 mg/L)

BOD is calculated from two separate dissolved oxygen measurements made using the Dissolved Oxygen Probe. The initial dissolved oxygen reading is taken at the sampling site using the procedures outlined in Test 5. Using a light-free sample bottle, a water sample is collected at the same site. The sample is transported back to the lab and incubated at 20°C for a total of five days. After five days, the incubated sample is tested for dissolved oxygen. The oxygen reading at the end of the five days is subtracted from the initial reading. The resulting value is the BOD level.

Method 2: High BOD Levels (> 6 mg/L)

This method is recommended when testing stagnant or polluted waters, in which all of the dissolved oxygen may be consumed before the end of the 5-day period. The initial dissolved oxygen test, sampling, storage and incubation, are performed in the same manner as found in Method 1. Differences for Method 2 are:

- Five water samples are collected.
- A sample is tested for dissolved oxygen every 24 hours for five days.
- If, before the fifth day, the dissolved oxygen concentration falls below 4.0 mg/L, oxygen is added to the remaining samples by aeration.
- Add each bottle's change in dissolved oxygen concentration to obtain the BOD value.

[1] Table 1 is from the Student Watershed Research Project manual, 3rd Edition 1996.

Method 1: LOW BOD (0–6 mg/L)

Materials

computer
Vernier computer interface
Logger *Pro*
Vernier Dissolved Oxygen Probe
D.O. Electrode Filling Solution
Sodium Sulfite Calibration Solution
250 mL beaker
5 BOD bottles
aluminum foil
100% calibration bottle
wash bottle with distilled water
tissues or paper towels
pipet
small plastic or paper cup (optional)

Testing Procedure

Day 0

1. Obtain an initial dissolved oxygen reading at the site you are testing for BOD. If another student group is performing the dissolved oxygen test outlined in Test 5, then copy their dissolved oxygen readings onto the BOD Data & Calculations sheet under the heading of Initial Dissolved Oxygen. If no one is taking an initial dissolved oxygen reading at this site, you must perform the dissolved oxygen test prior to beginning this test.

2. Collect three water samples for the BOD test at the same location the initial dissolved oxygen reading was measured. Using the glass BOD sample bottles, submerge each sample bottle 10 cm below the water's surface and keep it there for 1 minute. When a minute has elapsed, the bottle should be void of any air bubbles and completely full. Place the bottle lid back on the bottle and screw the lid down tight while still submerged. Each bottle should be completely covered with aluminum foil or black tape to block out any light.

3. If the time between collection of samples and incubation of samples is greater than 30 minutes, place the filled bottles in an ice chest until they can be placed in an incubator. If the time is less than 30 minutes, simply keep the filled bottles out of direct sunlight.

4. Upon returning to the lab, place the BOD bottles in an incubator or dark closet at about 20°C. The bottles should remain in the incubator or closet for five days until you are ready to perform the final dissolved oxygen measurement in the Day-5 procedure below.

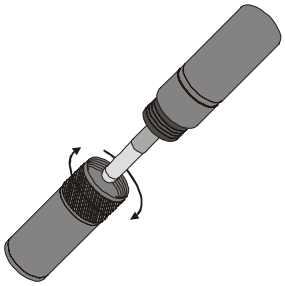
Day 5

When five days have passed, perform Steps 5–12 to test the incubated samples for dissolved oxygen. If possible, try to test the samples at roughly the same time of day they were collected.

5. Position the computer safely away from the water. Keep water away from the computer at all times.

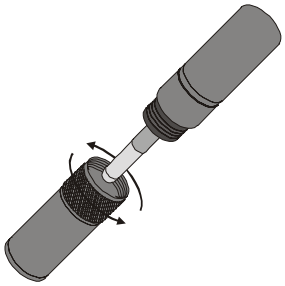
6. Connect the Dissolved Oxygen Probe to Channel 1 of the Vernier interface.

7. You are now ready to prepare the Dissolved Oxygen Probe for use.
   a. Remove the blue protective cap if it is still on the tip of the probe.
   b. Unscrew the membrane cap from the tip of the probe.
   c. Using a pipet, fill the membrane cap with 1 mL of D.O. Electrode Filling Solution.
   d. Carefully thread the membrane cap back onto the electrode.
   e. Place the probe into a container of water.



Remove membrane cap

Add electrode filling solution



Replace membrane cap

8. Prepare the computer for data collection by opening the file "20 BOD" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

9. It is necessary to warm up the Dissolved Oxygen Probe for 5–10 minutes before taking readings. To warm up the probe, leave it connected to the interface, with Logger *Pro* running, for 5–10 minutes. The probe must stay connected at all times to keep it warmed up. If disconnected for a few minutes, it will be necessary to warm up the probe again.

10. You are now ready to calibrate the Dissolved Oxygen Probe.
    - If your instructor directs you to use the calibration stored in the experiment file, then proceed to Step 11.
    - If your instructor directs you to perform a new calibration for the Dissolved Oxygen Probe, follow this procedure.

    Zero-Oxygen Calibration Point

    a. Choose Calibrate ▸ CH1: Dissolved Oxygen (mg/L) from the Experiment menu and then click Calibrate Now.
    b. Remove the probe from the water bath and place the tip of the probe into the Sodium Sulfite Calibration Solution. **Important:** No air bubbles can be trapped below the tip of the probe or the probe will sense an inaccurate dissolved oxygen level. If the voltage does not rapidly decrease, tap the side of the bottle with the probe to dislodge the bubble. The readings should be in the 0.2 to 0.5 V range.

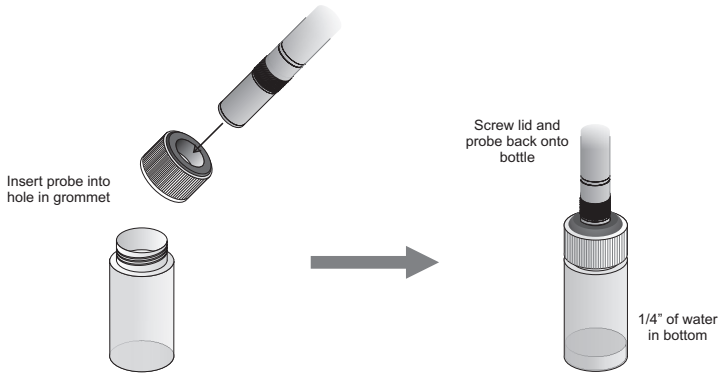
Insert probe at an angle

Submerge probe tip 1-2 cm

    c. Type **0** (the value in mg/L) in the edit box.
    d. When the displayed voltage reading for Reading 1 stabilizes, click Keep.

    Saturated D.O. Calibration Point

    e. Rinse the probe with distilled water.
    f. Unscrew the lid of the calibration bottle provided with the probe. Slide the lid and the grommet about 1/2 inch onto the probe body.



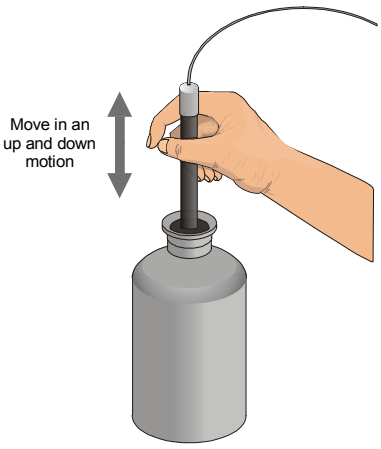
g. Add water to the bottle to a depth of about 1/4 inch and screw the bottle into the cap, as shown. Keep the probe in this position for about a minute. **Important:** Do not touch the membrane or get it wet during this step.

h. Type the correct saturated dissolved oxygen value (in mg/L) from Table 2 (for example, **8.66**) using the current barometric pressure and air temperature values. If you do not have the current air pressure, use Table 3 to determine the approximate air pressure at your altitude.

i. When the displayed voltage reading for Reading 2 stabilizes (readings should be above 2.0 V), click Keep and then click Done.

11. Remove the water samples from the incubator.

12. You are now ready to collect dissolved oxygen concentration data.



a. Submerge the probe tip in the BOD bottle. Gently move the probe in and up-and-down motion, while keeping the tip in the water at all times.

b. Click Collect to begin data collection.

c. Click Keep to begin a 10 s sampling run. **Important:** Leave the probe tip submerged for the 10 seconds that data is being collected.

d. When the sampling run is complete, stop data collection and record the dissolved oxygen value on the Data & Calculations sheet as the final 5 day DO reading.

13. Return to Step 12 to obtain a reading for the other two samples. When all readings have been taken, rinse the tip of the probe and secure it in the calibration bottle filled with water.

DATA & CALCULATIONS

Low BOD Levels (0–6 mg/L)

Stream or lake: _____ Time of day: _____

Site name: _____ Student name: _____

Site number: _____ Student name: _____

Date: _____ Student name: _____

	A	B	C
Sample	Initial dissolved oxygen (mg/L)	Final dissolved oxygen (mg/L)	BOD (mg/L)
Example	10.8 mg/L	6.7 mg/L	4.1 mg/L
1			
2			
3			
		Average	

Column Procedure:

A. Record dissolved oxygen reading from DO test performed at the sample site.
B. Record dissolved oxygen reading from DO test after incubation of sample for five days.
C. Calculate BOD = A – B = C

Field Observations (e.g., weather, geography, vegetation along stream) _____

_____

_____

_____

Test Completed: _____ Date: _____

Method 2: HIGH BOD (>6 mg/L)

Materials

computer
Vernier computer interface
Logger *Pro*
Vernier Dissolved Oxygen Probe
D.O. Electrode Filling Solution
Sodium Sulfite Calibration Solution
250 mL beaker
5 BOD bottles
aluminum foil
100% calibration bottle
wash bottle with distilled water
tissues or paper towels
pipet
small plastic or paper cup (optional)

Testing Procedure

Day 0

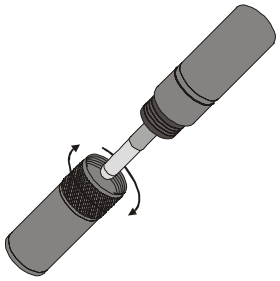
1. Obtain an initial dissolved oxygen reading at the site you are testing for BOD. If another student group is performing the dissolved oxygen test outlined in Test 5, then copy their dissolved oxygen readings onto the BOD Data & Calculations sheet as the Initial and Final Dissolved Oxygen reading for Day 0. If no one is taking an initial dissolved oxygen reading at this site, you must perform the dissolved oxygen test prior to beginning this test.
2. Collect five water samples for the BOD test at the same location the initial dissolved oxygen reading was measured. Using the glass BOD sample bottles, submerge each sample bottle 10 cm below the water's surface and keep it there for 1 minute. When one minute has elapsed, the bottle should be void of any air bubbles and completely full. Place the bottle lid back on the bottle and screw the lid down tight while still submerged. Each bottle should be completely covered with aluminum foil or black tape to block out any light. Using tape, label the bottles 1 through 5.
3. If the time between collection of samples and incubation of samples is greater than 30 minutes, place the filled bottles in an ice chest until they can be placed in an incubator. If the time is less than 30 minutes, simply keep the filled bottles out of direct sunlight.
4. Upon returning to the lab, place the BOD bottles in an incubator or dark closet set at 20°C. The bottles should remain in the incubator, or closet until you are ready to perform the first dissolved oxygen measurement in the Day 1–5 procedure below.

Day 1–5

When 24 hours have passed, perform Steps 5–13 to test an incubated sample for dissolved oxygen. The sample should be tested at approximately the same time of day the initial dissolved oxygen measurement was made.

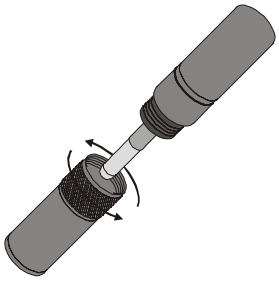
5. Position the computer safely away from the water. Keep water away from the computer at all times.
6. Connect the Dissolved Oxygen Probe to Channel 1 of the Vernier interface.
7. You are now ready to prepare the Dissolved Oxygen Probe for use.
   a. Remove the blue protective cap if it is still on the tip of the probe.
   b. Unscrew the membrane cap from the tip of the probe.
   c. Using a pipet, fill the membrane cap with 1 mL of D.O. Electrode Filling Solution.

d. Carefully thread the membrane cap back onto the electrode.
e. Place the probe into a container of water.



Remove membrane cap

Add electrode filling solution



Replace membrane cap

8. Prepare the computer for data collection by opening the file "06 BOD" from the *Agricultural Science with Vernier* folder of Logger*Pro*.
9. It is necessary to warm up the Dissolved Oxygen Probe for 5–10 minutes before taking readings. To warm up the probe, leave it connected to the interface, with Logger *Pro* running, for 5–10 minutes. The probe must stay connected at all times to keep it warmed up. If disconnected for a few minutes, it will be necessary to warm up the probe again.
10. You are now ready to calibrate the Dissolved Oxygen Probe.
    - If your instructor directs you to use the calibration stored in the experiment file, then proceed to Step 11.
    - If your instructor directs you to perform a new calibration for the Dissolved Oxygen Probe, follow this procedure.

Zero-Oxygen Calibration Point

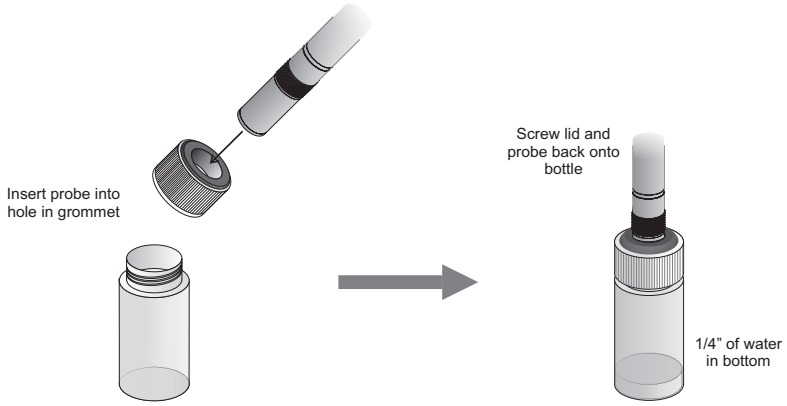
a. Choose Calibrate ▸ CH1: Dissolved Oxygen (mg/L) from the Experiment menu and then click Calibrate Now.
b. Remove the probe from the water bath and place the tip of the probe into the Sodium Sulfite Calibration Solution. **Important:** No air bubbles can be trapped below the tip of the probe or the probe will sense an inaccurate dissolved oxygen level. If the voltage does not rapidly decrease, tap the side of the bottle with the probe to dislodge the bubble. The readings should be in the 0.2 to 0.5 V range.
c. Type **0** (the value in mg/L) in the edit box.
d. When the displayed voltage reading for Reading 1 stabilizes, click Keep.

Insert probe at an angle

Submerge probe tip 1-2 cm

Saturated D.O. Calibration Point

e. Rinse the probe with distilled water.
f. Unscrew the lid of the calibration bottle provided with the probe. Slide the lid and the grommet about 1/2 inch onto the probe body.



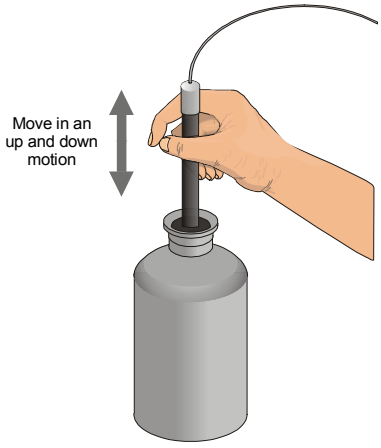
g. Add water to the bottle to a depth of about 1/4 inch and screw the bottle into the cap, as shown. Keep the probe in this position for about a minute. **Important:** Do not touch the membrane or get it wet during this step.

h. Type the correct saturated dissolved oxygen value (in mg/L) from Table 2 (for example, **8.66**) using the current barometric pressure and air temperature values. If you do not have the current air pressure, use Table 3 to determine the approximate air pressure at your altitude.

i. When the displayed voltage reading for Input 1 stabilizes (readings should be above 2.0 V), click Keep and then click Done.

11. Remove a water sample from the incubator.

12. You are now ready to collect dissolved oxygen concentration data.



a. Submerge the probe tip in the BOD bottle. Gently move the probe in and up-and-down motion, while keeping the tip in the water at all times.

a. Click Collect to begin data collection.

b. Click Keep to begin a 10 s sampling run. **Important:** Leave the probe tip submerged for the 10 seconds that data is being collected.

c. When the sampling run is complete, stop data collection and record the mean dissolved oxygen concentration value on the Data & Calculations sheet in the row for the day you are testing.

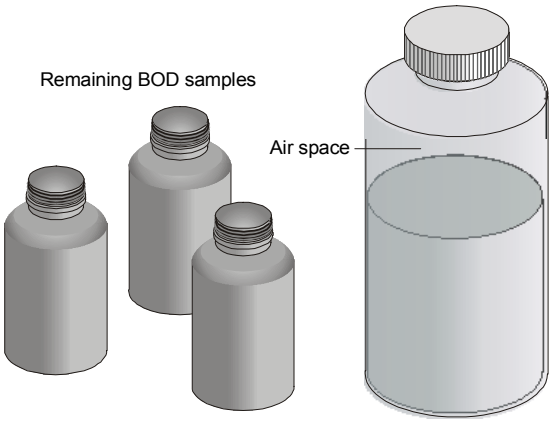
13. If the dissolved oxygen level falls below 4.0 mg/L before Day 5 it will be necessary to aerate the remaining water samples.

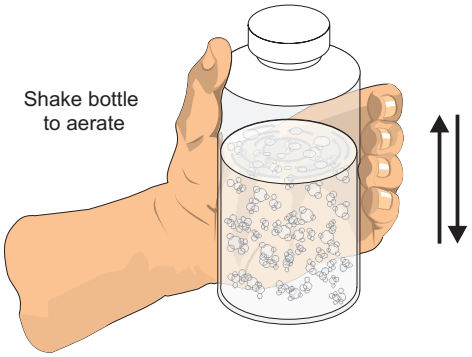
    a. Pour all remaining BOD water samples into a clean container or bottle that can be closed with a lid. Be sure to leave several inches of airspace in the top of the container. If this is not possible, you will need to find a larger container.

    b. Vigorously shake the closed container for one minute. Uncap for 30 seconds then close again. Shake for another minute and then pour the water back into the BOD bottles. This should sufficiently aerate the water, bringing the dissolved oxygen levels well above the 4.0 mg/L mark.

    c. Measure the dissolved oxygen of one of the BOD bottles as described in Step 12. Close all of the BOD bottles, making sure that they are all brim full with no air space visible within.

    d. Record the new dissolved-oxygen reading as the Initial Dissolved Oxygen for the next day on the Data & Calculations sheet. For example, if you are testing on Day 3, then record the new dissolved oxygen reading under the Initial Dissolved Oxygen column heading of Day 4.





14. In 24 hours, repeat Steps 5–13 to determine the dissolved-oxygen concentration of the next sample bottle. Continue testing a sample each day until all five samples have been tested. Calculate BOD as described on the Data & Calculations sheet.

DATA & CALCULATIONS

High BOD Levels (>6 mg/L)

Stream or lake: _____ Time of day: _____

Site name: _____ Student name: _____

Site number: _____ Student name: _____

Date: _____ Student name: _____

A		B	C	D
Aeration √	Day	Initial dissolved oxygen (mg/L)	Final dissolved oxygen (mg/L)	ΔDO (mg/L)
	Example	10.2 mg/L	7.7 mg/L	2.5 mg/L
	Example	7.7 mg/L	5.1 mg/L	2.6 mg/L
	Day 0			
	Day 1			
	Day 2			
	Day 3			
	Day 4			
	Day 5			

E	BOD	

Column Procedure:

A. If it was necessary to aerate the sample, place a check in column A.
B. Record final dissolved-oxygen reading from previous 24 hour test or first initial reading.
C. Record dissolved-oxygen reading from DO test performed on current day.
D. Calculate Δ DO = B – C = D
E. BOD = Sum of column D (day 0 through 5).

Field Observations (e.g., weather, geography, vegetation along stream)_____

_____

Test Completed: _____ Date: _____

CALIBRATION TABLES

Table 2: 100% Dissolved Oxygen Capacity (mg/L)												
	770 mm	**760 mm**	**750 mm**	**740 mm**	**730 mm**	**720 mm**	**710 mm**	**700 mm**	**690 mm**	**680 mm**	**670 mm**	**660 mm**
0°C	14.76	14.57	14.38	14.19	13.99	13.80	13.61	13.42	13.23	13.04	12.84	12.65
1°C	14.38	14.19	14.00	13.82	13.63	13.44	13.26	13.07	12.88	12.70	12.51	12.32
2°C	14.01	13.82	13.64	13.46	13.28	13.10	12.92	12.73	12.55	12.37	12.19	12.01
3°C	13.65	13.47	13.29	13.12	12.94	12.76	12.59	12.41	12.23	12.05	11.88	11.70
4°C	13.31	13.13	12.96	12.79	12.61	12.44	12.27	12.10	11.92	11.75	11.58	11.40
5°C	12.97	12.81	12.64	12.47	12.30	12.13	11.96	11.80	11.63	11.46	11.29	11.12
6°C	12.66	12.49	12.33	12.16	12.00	11.83	11.67	11.51	11.34	11.18	11.01	10.85
7°C	12.35	12.19	12.03	11.87	11.71	11.55	11.39	11.23	11.07	10.91	10.75	10.59
8°C	12.05	11.90	11.74	11.58	11.43	11.27	11.11	10.96	10.80	10.65	10.49	10.33
9°C	11.77	11.62	11.46	11.31	11.16	11.01	10.85	10.70	10.55	10.39	10.24	10.09
10°C	11.50	11.35	11.20	11.05	10.90	10.75	10.60	10.45	10.30	10.15	10.00	9.86
11°C	11.24	11.09	10.94	10.80	10.65	10.51	10.36	10.21	10.07	9.92	9.78	9.63
12°C	10.98	10.84	10.70	10.56	10.41	10.27	10.13	9.99	9.84	9.70	9.56	9.41
13°C	10.74	10.60	10.46	10.32	10.18	10.04	9.90	9.77	9.63	9.49	9.35	9.21
14°C	10.51	10.37	10.24	10.10	9.96	9.83	9.69	9.55	9.42	9.28	9.14	9.01
15°C	10.29	10.15	10.02	9.88	9.75	9.62	9.48	9.35	9.22	9.08	8.95	8.82
16°C	10.07	9.94	9.81	9.68	9.55	9.42	9.29	9.15	9.02	8.89	8.76	8.63
17°C	9.86	9.74	9.61	9.48	9.35	9.22	9.10	8.97	8.84	8.71	8.58	8.45
18°C	9.67	9.54	9.41	9.29	9.16	9.04	8.91	8.79	8.66	8.54	8.41	8.28
19°C	9.47	9.35	9.23	9.11	8.98	8.86	8.74	8.61	8.49	8.37	8.24	8.12
20°C	9.29	9.17	9.05	8.93	8.81	8.69	8.57	8.45	8.33	8.20	8.08	7.96
21°C	9.11	9.00	8.88	8.76	8.64	8.52	8.40	8.28	8.17	8.05	7.93	7.81
22°C	8.94	8.83	8.71	8.59	8.48	8.36	8.25	8.13	8.01	7.90	7.78	7.67
23°C	8.78	8.66	8.55	8.44	8.32	8.21	8.09	7.98	7.87	7.75	7.64	7.52
24°C	8.62	8.51	8.40	8.28	8.17	8.06	7.95	7.84	7.72	7.61	7.50	7.39
25°C	8.47	8.36	8.25	8.14	8.03	7.92	7.81	7.70	7.59	7.48	7.37	7.26
26°C	8.32	8.21	8.10	7.99	7.89	7.78	7.67	7.56	7.45	7.35	7.24	7.13
27°C	8.17	8.07	7.96	7.86	7.75	7.64	7.54	7.43	7.33	7.22	7.11	7.01
28°C	8.04	7.93	7.83	7.72	7.62	7.51	7.41	7.30	7.20	7.10	6.99	6.89
29°C	7.90	7.80	7.69	7.59	7.49	7.39	7.28	7.18	7.08	6.98	6.87	6.77
30°C	7.77	7.67	7.57	7.47	7.36	7.26	7.16	7.06	6.96	6.86	6.76	6.66
31°C	7.64	7.54	7.44	7.34	7.24	7.14	7.04	6.94	6.85	6.75	6.65	6.55

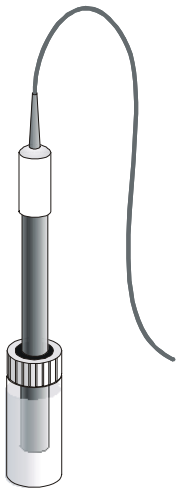
Table 3: Approximate Barometric Pressure at Different Elevations					
Elevation (feet)	**Pressure (mm Hg)**	**Elevation (feet)**	**Pressure (mm Hg)**	**Elevation (feet)**	**Pressure (mm Hg)**
0	760	2000	708	4000	659
250	753	2250	702	4250	653
500	746	2500	695	4500	647
750	739	2750	689	4750	641
1000	733	3000	683	5000	635
1250	727	3250	677	5250	629
1500	720	3500	671	5500	624
1750	714	3750	665	5750	618

TEACHER INFORMATION

Biochemical Oxygen Demand

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. In order for the Dissolved Oxygen Probe to warm up and stay polarized, power to the sensor must be continuous. LabPro, LabQuest, LabQuest Mini, and CBL 2 deliver continuous power once the data-collection software is started even if the screen goes to sleep. However, EasyLink used with a TI-84 graphing calculator and the EasyData App stops powering the sensor when the calculator goes to sleep. The calculator goes to sleep to conserve battery power if no user interaction is detected for 3 minutes. If power to the sensor is disrupted, the sensor must be warmed up for another 5–10 minutes before calibrating or taking readings. To avoid having to warm up the sensor again, students must press a button on the calculator every few minutes to keep the calculator awake.



3. The probe tip should be in water during the warm-up period. You could place the probe into a cup or beaker with water or you could use the DO calibration bottle. Simply fill the DO calibration bottle with water, fit the probe down into the lid, and tighten the lid onto the bottle. The probe tip should be submerged in the water until you calibrate or take samples.

4. When calibrating the Dissolved Oxygen Probe, it is important to be patient and permit the readings to stabilize.
   - At the zero oxygen point, the voltage should be somewhere between 0.2 V and 0.5 V. If it is not, make sure there is not an air bubble at the tip of your electrode. If you suspect your sodium sulfite solution may have gone bad, mix up some fresh or obtain a new bottle from Vernier (order code DO-CAL).
   - At the saturated oxygen point, the voltage should be above 2.0 V. If it is not, make sure the electrode is not actually touching the water in the bottle. Thoroughly rinse the electrode with distilled water again and gently blotted it dry with a paper towel being careful not to touch the membrane.

5. As the Dissolved Oxygen Probe measures dissolved oxygen, it removes O_2 from the water sample at the junction of the probe membrane. If you leave the probe in one spot in the water sample, you will see your dissolved-oxygen readings drop. To prevent this, it is important that students stir the probe gently through the sample as they take readings.

6. The gas-permeable plastic membrane on the Dissolved Oxygen Probe can become clogged by dirt and oil over time. It is best to advise students to avoid touching the membrane at any time. If the water being sampled is murky or dirty, rinse the probe tip with distilled water.

7. When students are collecting their samples, they should avoid sampling close to the surface of the water. This will cut down on the amount of silt and particulate collected in the samples.

8. Students will use the dissolved-oxygen concentration measured in Test 5 as their initial dissolved-oxygen reading. Students should collect their BOD samples from the same spot used for Test 5. It is best to take the samples about 10 cm below the water's surface.

9. Water samples should be incubated at 20°C ±1°C in an incubator. If an incubator is not available, it is possible to use a dark closet or drawer. This may not be as controlled as the incubator, but adequate for this test.

10. Sample bottles should be made of glass and completely covered with black electrical tape, black spray paint, or aluminum foil. If you prefer, amber or darkened glass bottles can be purchased from science supply companies rather than covering bottles with tape. It is necessary to block out all light, in order to prevent photosynthetic activity from within the water sample.

11. The instructions for the Low BOD Level test advise the students to collect three water samples. This can be cut back to a single sample, if desired. The two alternate samples are collected to improve the accuracy of the BOD test by taking the average of the three samples. The High BOD Level test requires five samples collected as a *minimum*. This enables students to measure a sample a day for the five-day period. To improve accuracy, you may choose to have students collect 10 or 15 samples.

12. If BOD levels are suspected to be high, you should have your students perform the High BOD Level test. During the course of the test it is likely that dissolved oxygen levels will fall below 4.0 mg/L before the test is concluded. If this occurs, the students must follow the instructions in the procedure and aerate the remaining BOD samples. When the samples have been aerated, it is necessary to take an initial dissolved-oxygen reading from the freshly aerated samples. Enter this new reading on the Data & Calculations sheet as the Initial Dissolved Oxygen reading for the following day.

13. Once samples are collected in the field, they should be placed away from any light in an ice chest. The samples should remain in the cooler until placed in the incubator back in the lab.

14. When collecting waters samples, students are instructed to submerge the sample bottles for 2 minutes. This allows the water to mix enough so that no oxygen is introduced into the bottle during filling.

15. When performing the High BOD test, you can simplify set up procedure by devoting a single interface to measuring dissolved oxygen. The DO Probe can be stored in a beaker of water overnight, rather than emptying the filling solution and storing it dry. The next day, have your students allow the DO Probe to warm up for 10 minutes. As long as the interface was not used for any other purpose since the last DO measurement, the initial calibration equation will still be stored. Therefore, students can skip the calibration steps and continue with the procedure.

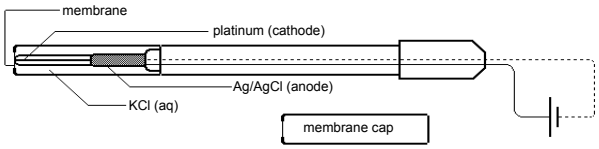
16. The student procedure provides several alternatives for loading or performing a Dissolved Oxygen Probe calibration:
    - The easiest option is to use the stored calibration.
    - Another option is to perform a two-point calibration in the lab, as described in the student procedure. After this calibration is completed, record the slope and intercept values. When students are ready to use the Dissolved Oxygen Probe, they can simply manually enter the calibration values. Since there is very little change in Dissolved Oxygen Probe performance over short periods of time, we think this is a good way for students to handle dissolved-oxygen calibrations.
    - The third option is to have students perform the two-point calibration described in the student procedure.

17. The Single Point data-collection mode (EasyData), "Average over 10 seconds" feature (LabQuest App), and "Use 10 second average" option (Logger *Pro*) were designed to make measurements easier and more accurate. When they are used, the interface takes readings for 10 seconds. These readings are averaged and this average value is recorded. This has several advantages:

    - It eliminates the need for students to choose one value over another if that value is fluctuating.
    - If the readings are fluctuating a little, an average of the values is desirable.
    - It requires the students to hold the sensor in the water longer than they might tend to otherwise.

How the Dissolved Oxygen Probe Works

The Vernier Dissolved Oxygen Probe is a Clark-type polarographic electrode that senses the oxygen concentration in water and aqueous solutions. A platinum cathode and a silver/silver chloride reference anode in KCl electrolyte are separated from the sample by a gas-permeable plastic membrane.



A fixed voltage is applied to the platinum electrode. As oxygen diffuses through the membrane to the cathode, it is reduced:

$$\tfrac{1}{2}\ O_2 + H_2O + 2e^- \longrightarrow 2\ OH^-$$

The oxidation taking place at the reference electrode (anode) is:

$$Ag + Cl^- \longrightarrow AgCl + e^-$$

Accordingly, a current will flow that is proportional to the rate of diffusion of oxygen, and in turn to the concentration of dissolved oxygen in the sample. This current is converted to a proportional voltage, which is amplified and read by any of the Vernier interfaces.

Storage of the Dissolved Oxygen Probe

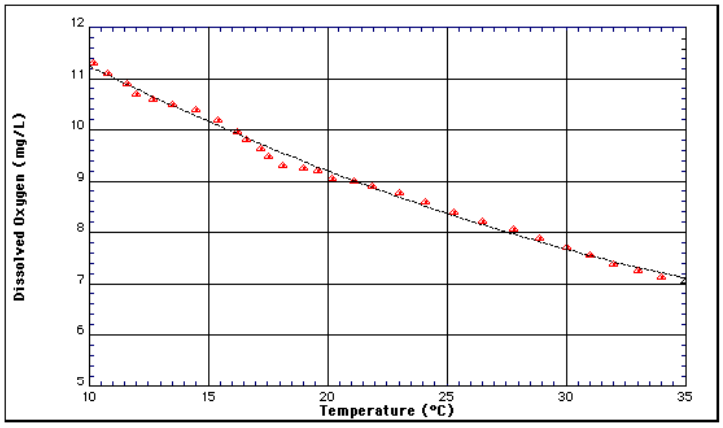
Follow these steps when storing the electrode:

- **Long-term storage (more than 24 hours):** Remove the membrane cap and rinse the inside and outside of the cap with distilled water. Shake the membrane cap dry. Also rinse and dry the exposed anode and cathode inner elements (blot dry with a lab wipe). Reinstall the membrane cap loosely onto the electrode body for storage. Do not screw it on tightly.
- **Short-term storage (less than 24 hours):** Store the Dissolved Oxygen Probe with the membrane end submerged in about 1 inch of distilled water.

Automatic Temperature Compensation

Your Vernier Dissolved Oxygen Probe is automatically temperature compensated, using a thermistor built into the probe. The temperature output of this probe is used to automatically compensate for changes in permeability of the membrane with changing temperature. If the probe was not temperature compensated, you would notice a change in the dissolved oxygen reading as temperature changed, even if the actual concentration of dissolved oxygen in the solution did not change. Here are two examples of how automatic temperature compensation works:

- If you calibrate the Dissolved Oxygen Probe in the lab at 25°C and 760 mm Hg barometric pressure (assume salinity is negligible), the value you entered for the saturated oxygen calibration point would be 8.36 mg/L (see Table 3). If you were to take a reading in distilled water that is saturated with oxygen by rapid stirring, you would get a reading of 8.36 mg/L. *If* the water sample is then cooled to 10°C with no additional stirring, the water would no longer be saturated (cold water can hold more dissolved oxygen than warm water). Therefore, the reading of the temperature-compensated Dissolved Oxygen Probe would still be 8.36 mg/L.
- If, however, the solution was cooled to 10°C *and* continually stirred so it remained saturated by dissolving additional oxygen, the temperature-compensated probe would give a reading of 11.35 mg/L—the value shown in Table 3. **Note:** Temperature compensation *does not mean* that the reading for a saturated solution will be the same at two different temperatures—the two solutions have different concentrations of dissolved oxygen, and the probe reading should reflect this difference.



Saturated Dissolved Oxygen vs. Temperature Data

Effects of Insulation on on Animal Temperature

Sheep, cattle, and other livestock have techniques to protect themselves against changes in temperature. Most animals have a hide or fleece to provide warmth and protection against cold and wet conditions. In summer animals sweat to increase heat loss through evaporation and may seek out natural shade sources such as trees or stay close to water.

Animal producers and owners also have many ways of managing animals to provide proper care and shelter. Barns and shelters can protect animals from cold, rain, and snow in the winter. Barns can also provide shade in hot summer months. Sometimes, animal producers will use misting systems to help cool animals.

Sheep producers are able to shear their animals to help regulate body temperature. Does it make sense to shear a sheep right before winter? In this activity you will explore the effect of insulation on maintaining body temperature.

OBJECTIVES

In this experiment, you will

- Analyze temperature changes over time.
- Simulate winter conditions on sheared and non-sheared "sheep."
- Consider how environmental factors influence animal well-being.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
2 Temperature Probes
insulated cup
non-insulated cup
large plastic container
warm water
ice water

PROCEDURE

1. Connect the Temperature Probes to the computer interface. Prepare the computer for data collection by opening the file "21 Animal Temperature" from the *Agricultural Science with Vernier* folder of Logger*Pro*.

2. Put some ice water in the large container. The depth of the water should be about equal to the depth of the water that will be in your cups.

3. Put equal amounts of warm water in both the insulated and the non-insulated cups. These are your sheep. Place one temperature probe into the insulated cup and the other in the non-insulated cup. Make sure the tips of the probes are NOT touching the sides or the bottoms of the cups!

4. Before you start data collection, allow the probes to adjust to the temperature of the water.

5. While one team member is stirring the water with the probes (without hitting the sides!), another person can start data collection by clicking ▶ Collect. After data have been collected for 30 seconds, the person holding the cups should lower them into the ice water. **Important**: Keep stirring until data collection is complete.

6. Print or sketch your graph as directed. Label the data for the different cups.

DATA TABLE

	Non-insulated cup	Insulated cup
Initial temperature (°C)		
Final temperature (°C)		
Change in temperature (°C)		

QUESTIONS

1. What does the insulation represent in this experiment?

2. What physiological system does the stirring represent?

3. Describe the results you see in the graph. Explain the difference, if any, between data from the two cups.

4. Explain how this experiment relates to the timing of sheep shearing.

5. This experiment models wintertime conditions. What are challenges for animals in the summer? How do they overcome these challenges? How does a farmer or rancher help the animals overcome these challenges?

EXTENSIONS

1. Perform an experiment that would model summertime conditions.

2. Conduct a research project to find out more about the effects of the environment on animals and how animals maintain constant body temperatures. Design an experiment to test the effect of other environmental factors on animal health.

TEACHER INFORMATION

Effects of Insulation on on Animal Temperature

1. The student pages with complete instructions for data-collection using LabQuest App, Logger *Pro* (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

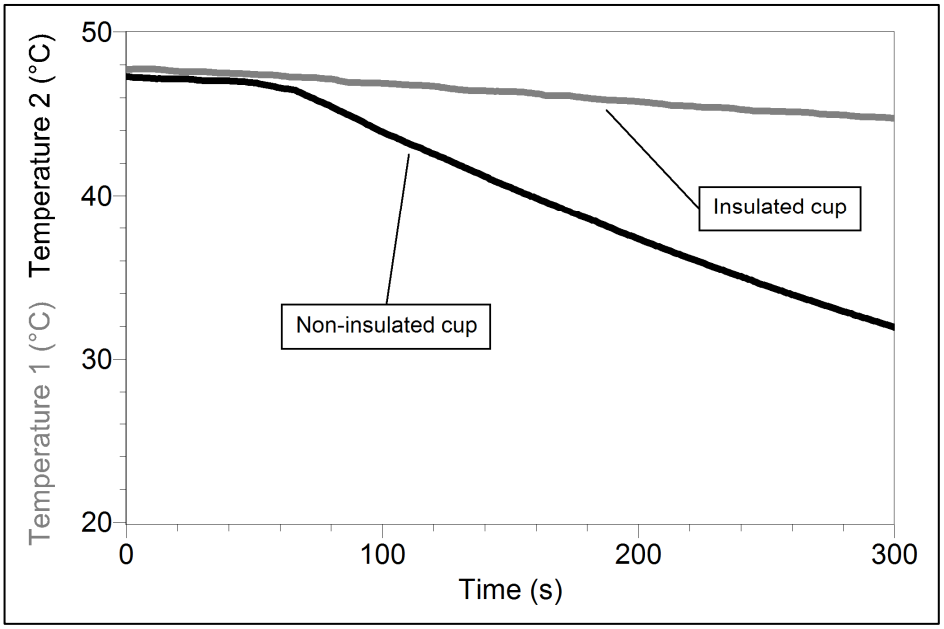
2. The cups should have approximately the same shape and volume. Filling them half to 2/3 with hot tap water works well.

3. Provide enough ice water that students can fill up the container to a level approximately equal to the level of the water inside the cup.

4. Each group needs two temperature probes for this activity. You may need to combine groups or instruct students to collect data in rounds.

SAMPLE DATA

	Non-insulated cup	Insulated cup
Initial temperature (°C)	47.1	47.1
Final temperature (°C)	31.9	44.8
Change in temperature (°C)	15.2	2.3



ANSWERS TO QUESTIONS

1. The insulation represents fleece or wool on the sheep.

2. The circulatory system

3. Answers will vary. In general students should identify that there is a steeper slope (more cooling) for the data for the non-insulated cup.

4. Sheep should not be sheared just before the cold season so that the fleece can help insulate through the cold months.

5. In summer, animals can overheat. They could be sheared, provided shade, or misted with water.

Lemon "Juice"

"Juice" is a slang term sometimes used for electricity. Batteries are made up of one or more electrochemical cells. Electrochemical cells often consist of two different materials in an electrolytic solution and connected to each other by a wire. In this experiment, you will study some basic principles of electrochemical cells using the juice of a lemon as the electrolyte. You will place small pieces of two different materials into the lemon, and a computer will be used to measure and display the voltages produced.

OBJECTIVES

In this experiment, you will

- Build several electrochemical cells.
- Use a computer to measure and display cell voltages.
- Discover which combinations produce a voltage.
- Decide which combination makes the "best" battery.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier Voltage Probe
2 alligator clips
a lemon
scalpel
graphite pencil (C)
iron nail (Fe)
magnesium strip (Mg)
zinc strip (Zn)
paper towel

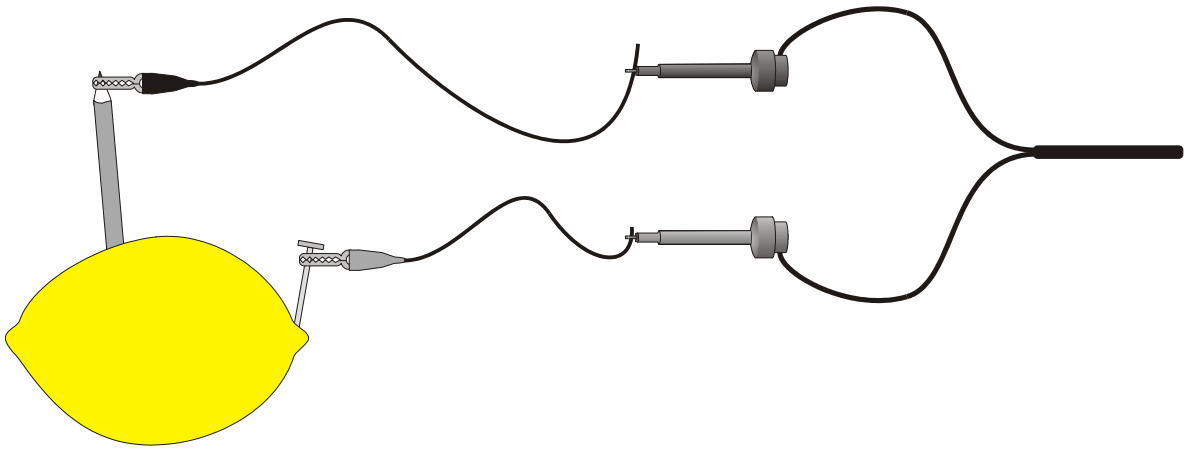


Figure 1

PROCEDURE

1. Use a pen to make two parallel marks 1 cm long and 2 cm apart on a lemon. Use a scalpel to cut 2 slits in the lemon peel at the marks.
2. Attach the red Voltage Probe lead to one alligator clip and the black probe lead to a second alligator clip as shown in Figure 1. You will be attaching the alligator clips to the test materials during this experiment in order to prevent corrosion of the Voltage Probe leads.
3. Connect the probe to the computer interface. Prepare the computer for data collection by opening the file "22 Lemon Juice" from the *Agricultural Science with Vernier* folder.
4. Insert a short graphite pencil, sharpened at both ends, into one of the slots and an iron nail into the other. Hook the alligator clip attached to the red probe lead to the pencil. Hook the alligator clip attached to the black probe lead to the iron nail.
5. Record the voltage reading. Observe whether the voltage reading stays constant, rises, or drops. Record your observations.
6. Switch the positions of the alligator clips. Record the voltage reading and your observations.
7. Repeat Steps 4–6 for the other combinations listed in the data table. Dry the materials after each use.

DATA

Probe Lead Red	Probe Lead Black	Voltage (V)	Observations	Probe Lead Red	Probe Lead Black	Voltage (V)	Observations
C	Fe			Fe	C		
C	Mg			Mg	C		
C	Zn			Zn	C		
Fe	Mg			Mg	Fe		
Fe	Zn			Zn	Fe		
Mg	Zn			Zn	Mg		

PROCESSING THE DATA

1. What happens to the voltage reading if a cell is hooked up backwards?
2. Which combination gives the highest voltage?
3. Which combination(s) gives the steadiest voltage?
4. Which combination would make the best battery? Explain.
5. The chemical activity of metal is shown by the size of the voltage reading when the metal is paired with carbon in a cell. A high voltage indicates high chemical activity. Rank the three metals (Fe, Mg, and Zn) according to chemical activity, from highest to lowest.

EXTENSIONS

1. Measure the voltage of "lemon cells" connected in series.
2. Try the experiment using other fruits and vegetables.
3. Do the experiment using other metals, such as aluminum, copper, and lead.

TEACHER INFORMATION

Lemon "Juice"

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. Number 2 pencils cut to a length of about 15 cm and sharpened at both ends serve well as carbon electrodes.

3. Any iron nail should work. Magnesium ribbon cut in 4 cm lengths and 4 cm × 1 cm pieces of zinc work well.

4. Consider cutting the slits in the lemons prior to class. The lemons can be reused.

5. The computer procedure has students record voltage data from the meter (without clicking the Collect button). Another possibility is to have students use the Selected Events mode for each of the 12 trials. The Logger*Pro* file for this experiment is already set up for this option. Simply have your students click the Collect button and click the Keep button when the reading is stable. This saves the voltage reading along with its trial number in the table.

SAMPLE RESULTS

Test Lead Red	Test Lead Black	Voltage (V)	Observations	Test Lead Red	Test Lead Black	Voltage (V)	Observations
C	Fe	0.67	steady	Fe	C	–0.52	drops
C	Mg	1.80	drops	Mg	C	–1.70	drops
C	Zn	1.22	steady	Zn	C	–0.91	drops
Fe	Mg	1.08	drops	Mg	Fe	–0.99	steady
Fe	Zn	0.51	steady	Zn	Fe	–0.39	steady
Mg	Zn	–0.79	steady	Zn	Mg	0.86	steady

ANSWERS TO QUESTIONS

1. If the cell is hooked up backwards, the reading is negative when using a LabPro interface.

2. The carbon-magnesium cell gives the highest voltage reading.

3. The carbon-zinc cell generally gives the steadiest reading. Other cell readings are sometimes very constant, too.

4. If the criterion is highest voltage reading, then the carbon-magnesium cell is the "best." If a steady reading is desired, then the carbon-zinc cell would be chosen as "best."

5. The activity ranking of the three metals (from highest to lowest) is Mg, Zn, Fe. The standard reduction potentials are –2.37, –0.76, and –0.44 V, respectively.

Ohm's Law

The fundamental relationship among the three important electrical quantities *current*, *voltage*, and *resistance* was discovered by Georg Simon Ohm. The relationship and the unit of electrical resistance were both named for him to commemorate this contribution to physics. One statement of Ohm's law is that the current through a resistor is proportional to the voltage across the resistor. In this experiment you will see if Ohm's law is applicable to several different circuits using a Current Probe and a Voltage Probe.

Current and voltage can be difficult to understand, because they cannot be observed directly. To clarify these terms, some people make the comparison between electrical circuits and water flowing in pipes. Here is a chart of the three electrical units we will study in this experiment.

Electrical Quantity	Description	Unit	Water Analogy
Voltage or Potential Difference	A measure of the Energy difference per unit charge between two points in a circuit.	Volt (V)	Water Pressure
Current	A measure of the flow of charge in a circuit.	Ampere (A)	Amount of water flowing
Resistance	A measure of how difficult it is for current to flow in a circuit.	Ohm (Ω)	A measure of how difficult it is for water to flow through a pipe.

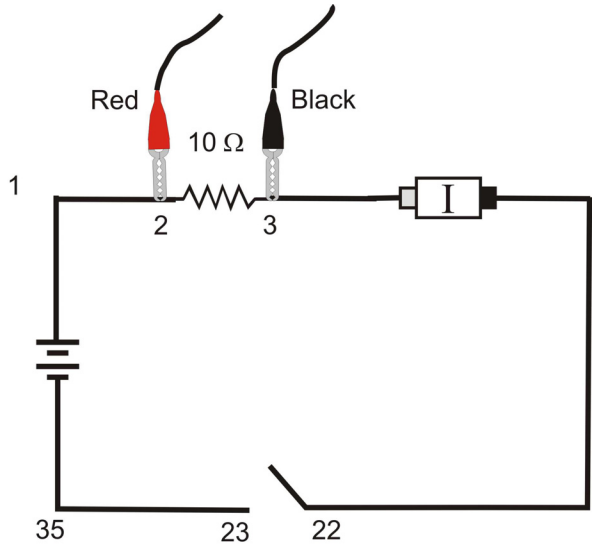


Figure 1

OBJECTIVES

- Determine the mathematical relationship between current, potential difference, and resistance in a simple circuit.
- Compare the potential *vs*. current behavior of a resistor to that of a light bulb.

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier Current Probe
Vernier Differential Voltage Probe
adjustable 5 volt DC power supply
Vernier Circuit Board, **or**
wires
clips to hold wires
switch
two resistors (about 10 and 50 Ω)
light bulb (6.3 V)

PRELIMINARY SETUP AND QUESTIONS

1. Connect the Current Probe to Channel 1 and the Differential Voltage Probe to Channel 2 of the computer interface.

2. Open the file "23 Ohms Law" in the *Agricultural Science with Vernier* folder.

3. With the power supply turned off, connect the power supply, 10 Ω resistor, wires, and clips as shown in Figure 1. Take care that the positive lead from the power supply and the red terminal from the Current & Voltage Probe are connected as shown in Figure 1. **Note:** Attach the red connectors electrically closer to the positive side of the power supply.

4. Click Zero. A dialog box will appear. Click OK to zero both sensors. This sets the zero for both probes with no current flowing and with no voltage applied.

5. Have your teacher check the arrangement of the wires before proceeding. Turn the control on the DC power supply to 0 V and then turn on the power supply. Slowly increase the voltage to 5 V. Monitor the meter in Logger *Pro* and describe what happens to the current through the resistor as the potential difference across the resistor changes. If the voltage doubles, what happens to the current? What type of relationship do you believe exists between voltage and current?

PROCEDURE

1. Record the value of the resistor in the data table.

2. Make sure the power supply is set to 0 V. Click Collect to begin data collection. Monitor the voltage and current. Click Keep.

3. Increase the voltage on the power supply to approximately 0.5 V. Click Keep.

4. Increase the voltage by about 0.5 V. Click Keep. Repeat this process until you reach a voltage of 5.0 V.

5. Click Stop and set the power supply back to 0 V.

6. Print a copy of the graph. Are the voltage and current proportional? Click the Linear Fit button, . Record the slope and *y*-intercept of the regression line in the data table, along with their units.

7. Repeat Steps 1–6 using a different resistor.

8. Replace the resistor in the circuit with a 6.3 V light bulb. Repeat Steps 2–5, but this time increase the voltage in 0.1 V steps up to 5.0 V.

9. To compare slopes of data at different parts of the curve, first click and drag the mouse over the first 3 data points. Click the Linear Fit button, , and record the slope of the regression line in the data table. Be sure to enter the units of the slope.

10. Click and drag the mouse over the last 10 points on the graph. Click the Linear Fit button, , and record the slope of the regression line in the data table.

DATA TABLE

	Slope of regression line (V/A)	Y-intercept of regression line (V)
Resistor ____ Ω		
Resistor ____ Ω		
Light bulb (first 3 pts)		
Light bulb (last 10 pts)		

ANALYSIS

1. As the potential across the resistor increased, the current through the resistor increased. If the change in current is *proportional* to the voltage, the data should be in a straight line and it should go through zero. In these two examples how close is the *y*-intercept to zero? Is there a proportional relationship between voltage and current? If so, write the equation for each run in the form potential = constant × current. (Use a numerical value for the constant.)

2. Compare the constant in each of the above equations to the resistance of each resistor.

3. Resistance, *R*, is defined using $R = V/I$ where *V* is the potential across a resistor, and *I* is the current. *R* is measured in ohms (Ω), where 1 Ω = 1 V/A. The constant you determined in each equation should be similar to the resistance of each resistor. However, resistors are manufactured such that their actual value is within a tolerance. For most resistors used in this lab, the tolerance is 5% or 10%. Check with your instructor to determine the tolerance of the resistors you are using. Calculate the range of values for each resistor. Does the constant in each equation fit within the appropriate range of values for each resistor?

4. Do your resistors follow Ohm's law? Base your answer on your experimental data.

5. Describe what happened to the current through the light bulb as the potential increased. Was the change linear? Since the slope of the linear regression line is a measure of resistance, describe what happened to the resistance as the voltage increased. Since the bulb gets brighter as it gets hotter, how does the resistance vary with temperature?

6. Does your light bulb follow Ohm's law? Base your answer on your experimental data.

EXTENSIONS

1. Investigate Ohm's law for reverse currents in resistors. Turn off the power supply and reverse the connections on the power supply. Turn the power supply back on and take data from 5.0 V to 0 V. Do not stop data collection. Turn off the power supply, restore the connections to the circuit to their original configuration, and turn the power supply back on. Take data from 0 to 5 V as before. Is the current still proportional to the potential across the resistor?

2. Investigate the behavior of other electrical devices such as diodes, LEDs, and Zener diodes. Make one run, then reverse the direction of the device and repeat.

3. Use a low voltage AC power supply and measure current and voltage as a function of time in a simple circuit. Compare the two graphs. Create a graph of voltage *vs.* current. Perform a linear regression over this data and compare to the resistance in the circuit.

TEACHER INFORMATION

Ohm's Law

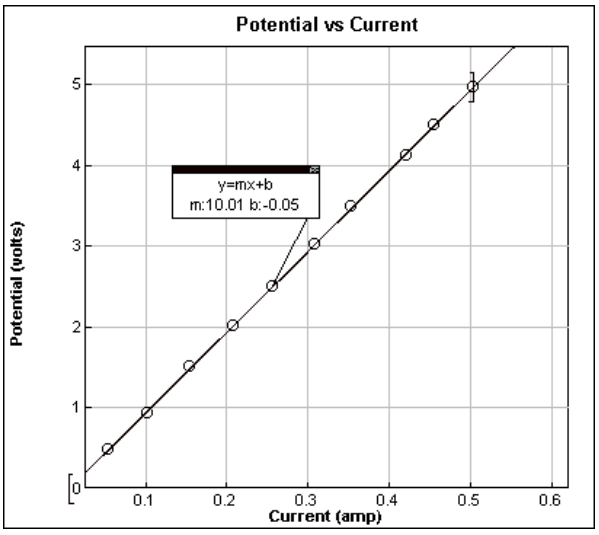
1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. If you use small resistances as we recommend, be sure to use power resistors capable of carrying the current. The sample data shown below was collected with Radio Shack 10 Ω Power Resistors (2 resistors cost about $1) and Radio Shack 50 Ω Power Resistors (2 resistors cost about $1). These resistors are rated to 10 watts.

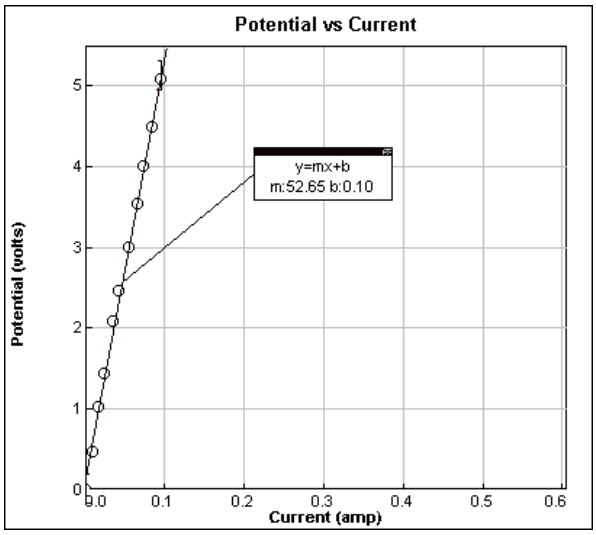
3. Radio Shack #40 Light Bulbs (two bulbs cost about $1) operate at 6.3 V and work well for the second part of the lab. If you use a smaller voltage light bulb, be sure to warn your students not to exceed the voltage rating of the bulb.

4. We recommend a power supply such as the Extech Digital DC Power Supply, available from Vernier Software & Technology.

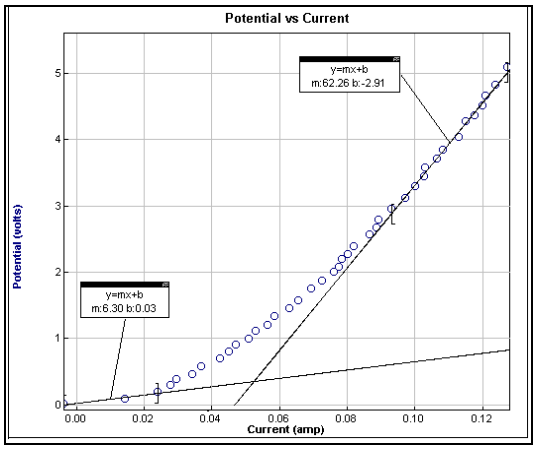
SAMPLE DATA



Potential vs. Current for 10 Ω Resistor



Potential vs. Current for 50 Ω Resistor



Potential vs. Current for 6.3 V Light Bulb

Device	Slope of regression line (V/A)	Y-intercept of regression line (V)
Resistor 10 Ω	10.0	-0.05
Resistor 50 Ω	52.7	0.10
Light bulb (first 3 pts)	6.3	
Light bulb (last 10 pts)	63.0	

ANSWERS TO PRELIMINARY QUESTIONS

5. Voltage increases as current increases. It appears the current and voltage are proportional.

ANSWERS TO ANALYSIS QUESTIONS

1. In both of these runs the potential or voltage and current are directly proportional. The *y*-intercepts are very close to zero and the data follow a straight line.

2. In each case the slope of the regression line is very close to the resistance value. The slope of the regression line for the with 10 Ω resistor was 10.0 V/A, or 10.0 Ω. The slope of the regression line for the 50 Ω resistor was 52.7 V/A.

3. Each of the resistors tested had a tolerance of 10%. The acceptable range for the 50-Ω resistor would be between 45 and 55 Ω. The 10 Ω resistor would be between 9 and 11 Ω. The resistance determined from the slopes of the regression line fit within these ranges.

4. Since the potential *vs.* current data follow a proportional relationship, the resistors tested do follow Ohm's law.

5. The graph of voltage *vs.* current for the light bulb is not linear. The slope of the first few data points is smaller than the last data. This implies that the resistance increases as the bulb brightness increases. For a light bulb filament, resistance increases with temperature.

6. The potential *vs.* current data of the light bulb are not proportional; therefore, we conclude that light bulb filaments do not follow Ohm's law.

Energy Content of Fuels

Energy content is an important property of fuels. This property helps scientists and engineers determine the usefulness of a fuel. Energy content is the amount of heat produced by the burning of 1 gram of a substance, and is measured in joules per gram (J/g).

You can determine energy content of a fuel by burning an amount of the fuel and capturing the heat released in a known mass of water in a calorimeter. If you measure the initial and final temperatures, the energy released can be calculated using the equation

$$H = \Delta t \cdot m \cdot C_p$$

where H = heat energy absorbed (in J), Δt = change in temperature (in °C), m = mass (in g), and C_p = specific heat capacity (4.18 J/g°C for water). Dividing the resulting energy value by grams of food burned gives the energy content (in J/g).

OBJECTIVES

In this experiment, you will

- Use a computer to measure temperature.
- Use a computer to analyze data.
- Use a balance.
- Determine energy content.
- Compare the energy content of different fuels.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Vernier Temperature Probe
fuel sample (candle, oil, or alcohol)
ring stand and 10 cm (4") ring
utility clamp
slit stopper
100 mL graduated cylinder
2 stirring rods
balance
small can
cold water
matches

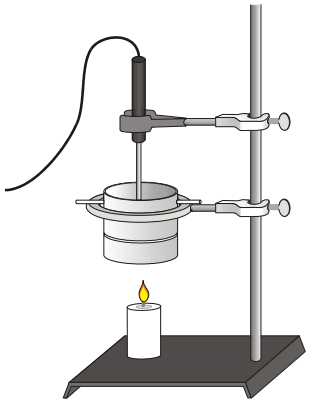


Figure 1

PROCEDURE

1. Obtain and wear goggles.

2. Connect the probe to the computer interface. Prepare the computer for data collection by opening the file "24 Energy of Fuels" from the *Agricultural Science with Vernier* folder.

3. Find and record the initial mass of the lamp with fuel sample or candle assigned to you. Make sure no more than 0.5 cm of wick sticks out of a lamp.

4. Set up the apparatus shown in Figure 1.
   a. Determine and record the mass of the empty can.
   b. Place cold water from the teacher into the can. Use 100 mL for candles and 200 mL for alcohol and oil.
   c. Determine and record the mass of the can plus water.
   d. Use a 10 cm ring and stirring rod to suspend the can about 5 cm above the candle or lamp.
   e. Use a utility clamp and slit stopper to suspend the Temperature Probe in the water. The probe should not touch the bottom of the can.

5. Click ▸ Collect to begin measuring temperature. Remember: The Temperature Probe must be in the water for at least 45 seconds before you begin! After 3 or 4 temperature readings have been made, light the lamp or candle. Record the initial temperature. Heat the water until its temperature reaches 40°C and then extinguish the flame. **CAUTION**: *Keep hair and clothing away from open flames.*

6. Stir the water until the temperature stops rising. Record this final temperature. Click ■ Stop to end data collection.

7. Determine the final mass of the lamp and fuel or candle.

8. To confirm the initial temperature, examine the initial data points in the table. To confirm the final temperature, click the on the graph to select it and then click the Statistics button, . The maximum temperature is listed in the statistics box on the graph.

9. Repeat the procedure using a different fuel. Start with cold water again.

DATA

	Trial 1	Trial 2
Fuel used		
Mass of lamp and fuel or candle (initial) (g)		
Mass of lamp and fuel or candle (final) (g)		
Mass of empty can (g)		
Mass of can plus water (g)		
Initial water temperature (°C)		
Final water temperature (°C)		

PROCESSING THE DATA

1. Calculate the change in water temperature, Δt, for each sample by subtracting the initial temperature from the final temperature ($\Delta t = t_{final} - t_{initial}$).

2. Calculate the mass (in g) of the water heated for each sample. Subtract the mass of the empty can from the mass of the can plus water.

3. Use the results of Steps 1 and 2 to determine the heat energy gained by the water (in J). Use the equation

$$H = \Delta t \cdot m \cdot C_p$$

   where H = heat absorbed (in J), Δt = change in temperature (in °C), m = mass of the water heated (in g), and C_p = specific heat capacity (4.18 J/g°C for water).

4. Calculate the mass (in g) of fuel burned. Subtract the final mass from the initial mass.

5. Use the results of Steps 3 and 4 to calculate the energy content (in J/g) of the fuel samples.

6. Record your results and the results of other groups in the Class Results table below.

Class Results			
Fuel Type	Fuel Type	Fuel Type	Fuel Type
Energy content (J/g)	Energy content (J/g)	Energy content (J/g)	Energy content (J/g)
Average:	Average:	Average:	Average:

7. Which of the fuels has the greatest energy content?
8. List two other factors, in addition to energy content, that might be important in choosing a fuel.

EXTENSIONS

1. Make a bar graph comparing the fuels tested in your class.
2. Design an experiment to compare heat content of different alcohols or oils.

TEACHER INFORMATION

Energy Content of Fuels

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Alcohol burners, available from science supply companies, work well with either alcohol or lamp oil. Because alcohol evaporates rapidly, the mass of the alcohol and burner should be determined immediately before and after use.
3. Small soup cans work well. After removing the paper and top, place two holes, large enough to fit a stirring rod, near the top. Be sure to remove all sharp edges. Many teachers prefer to use aluminum beverage cans.
4. Have ice water available and make sure all ice is removed from the water to be used. Water initially at 4–5°C gives best results, because starting 17–19°C below and finishing 17–19°C above room temperature tends to equalize heat exchange with the room.
5. Have the candles mounted on small pieces of cardboard. The cardboard bases will catch candle drippings.

SAMPLE RESULTS

Fuel used	candle
Mass of lamp and fuel or candle (initial) (g)	24.82
Mass of lamp and fuel or candle (final) (g)	24.18
Mass of empty can (g)	40.44
Mass of can plus water (g)	139.96
Initial water temperature (°C)	5.6
Final water temperature (°C)	41.2

PROCESSING THE DATA RESULTS

1. $\Delta t = 41.2 - 5.6 = 35.6°C$
2. 139.96 – 40.44 = 99.52 g water heated
3. $H = 35.6°C \times 99.52\ g \times 4.18\ J/g°C = 14{,}810\ J$ gained
4. 24.82 – 24.18 = 0.64 g candle burned
5. 14,810 J / 0.64 g = 23,140 J/g

6. Typical class averages are:

candle (paraffin wax)	23,000–25,000 J/g
lamp oil	16,000–18,000 J/g
ethyl alcohol	12,000–14,000 J/g

   Actual values are about 42.0 kJ/g and 26.8 kJ/g for paraffin wax and ethyl alcohol, respectively. You may want to discuss factors causing the values obtained in this experiment to be lower than the actual values.

7. Measured in J/g, candle wax has the greatest energy content.

8. Other possible factors to consider in the selection of a fuel are convenience, cost, availability, burning efficiency, and renewability.

Photovoltaic Cells

Energy produced by the sun is called *solar energy*. It is produced during nuclear reactions that take place throughout the volume of the sun. The energy travels to Earth in the form of light. *Photovoltaic (PV) cells*, or solar cells, change the light energy to electrical energy that can be used to power calculators, cars or even satellites. A photovoltaic cell is usually made of a semiconducting material such as silicon. When light strikes the cell, it provides enough energy to move electrons through the cell producing an electric current. A single photovoltaic cell is approximately the size of a fingernail and puts out a very small current when struck by the light. Objects requiring higher currents to operate can be powered by wiring large numbers of photovoltaic cells together.

Items powered by solar energy are said to be using *solar power*. Streetlights that must operate in the dark store the energy in a battery while the sun is shining and then use the energy at night. Scientists working in remote places rely on solar power to operate their computers and equipment. What things can you think of that are powered by solar energy?

In Part I of this experiment, you will measure the current and voltage produced by a photovoltaic cell when exposed to sunlight. You will calculate the power output of the cell using the relationship

$$P = V I$$

Power = voltage × current

You will also calculate the efficiency of the photovoltaic cell when converting the energy from the sun into electrical energy.

In Part II the relationship between the wavelength of the light striking the photovoltaic cell and power output will be investigated.

OBJECTIVES

In this experiment, you will

- Use a Current Probe to measure current output.
- Use a Voltage Probe to measure voltage output.
- Use a Light Sensor to measure light intensity.
- Calculate power output.
- Calculate efficiency.
- Investigate the relationship between wavelength of light and power output.

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Current Probe
Voltage Probe
Light Sensor
photovoltaic cell
wire leads with alligator clips
100 Ω resistor
blue, green, and red filters
sunshine
ruler

PRE-LAB QUESTIONS

1. In Part I of this experiment, you will determine the efficiency of the photovoltaic cell. A cell that converts all of the light energy into electrical energy is said to be 100% efficient. What do you predict to be the efficiency of the cell? Express your answer in percent form with 100% representing that all of the sunlight is converted to electrical energy and 0% representing that none of the sunlight is converted to electrical energy.

2. In Part II of this experiment, you will investigate the relationship between the wavelength of light striking the photovoltaic cell and the power output of the cell. Do you think that the wavelength of light will affect the power output? If so, which wavelength – red, blue, or green – will produce the highest power output?

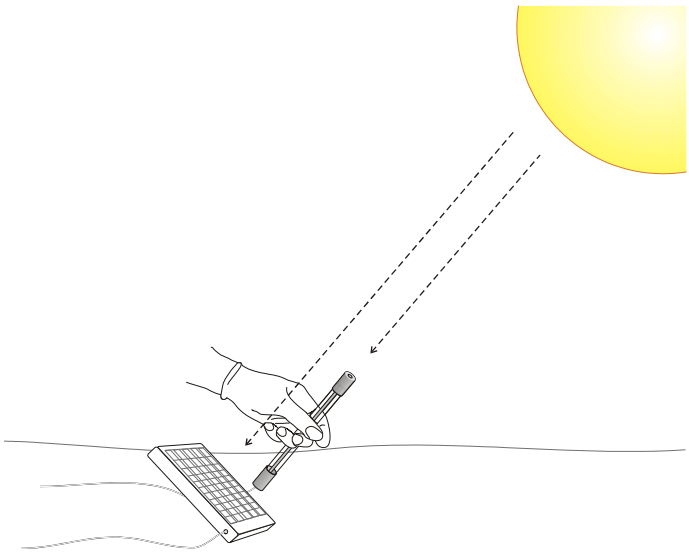


Figure 1

PROCEDURE

Part I Determining Power Output

1. Connect the Current Probe to Channel 1, the Voltage Probe into Channel 2, and the Light Sensor to Channel 3 of the Vernier computer interface. Set the Light Sensor to the 0–150,000 lux range.

2. Open the file "25 Photovoltaic Cells" in the *Agricultural Science with Vernier* folder.

3. Connect together the two voltage leads (red and black) of the Voltage Probe. Click Zero then zero both Current and Potential. This sets the zero for both probes with no current flowing and with no voltage applied.

4. Connect the series circuit shown in Figure 2. The red terminal of the Current Probe should be toward the + terminal of the photovoltaic cell. Look at the bottom of the PV cell to determine polarity. Connect the red lead of the Voltage probe to the wire coming from the + terminal of the PV cell and the black lead to the wire leading to the – terminal.

5. Tilt the PV cell toward the sun. Hold the Light Sensor at the same angle. The light intensity reading is displayed in the Illumination meter. Record this value in the data table.

6. Click Collect to begin data collection.

7. When data collection is complete, click on the Current graph to make it active. Click the Statistics button, , to display a Statistics box for the Current Probe. Record the mean current reading in the data table.

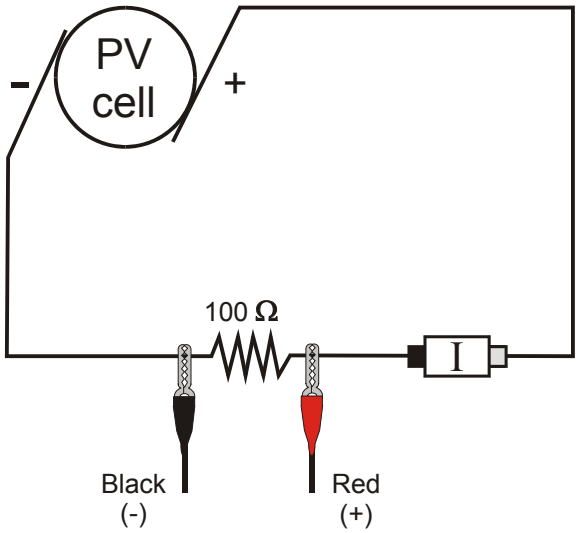


Figure 2

8. Click on the Voltage graph. Click the Statistics button, , to display a Statistics box for the Voltage Probe. Record the mean voltage reading in the data table.

9. Repeat steps 5–8 for two more trials. Keep the tilt of the PV cell and Light Sensor the same in all three trials.

Part II Effect of Wavelength

10. Repeat Steps 5–8 once more and record the mean values for current and voltage and the illumination value in the data table for Part II.

11. Place a blue filter over the tip of the Light Sensor. Tilt the Light Sensor toward the sun. Record the light intensity in the data table. Place the blue filter over the PV cell and tilt the PV cell toward the sun. Repeat Steps 6–8. Record the mean values for current and voltage in the data table.

12. Repeat Step 11, first with a red filter, and then with a green filter.

DATA

Part I Determining Power Output

	Current (A)	Voltage (V)	Illumination (lux)
Trial 1			
Trial 2			
Trial 3			
Average			

Instructions for completing the following table can be found in the Processing the Data section.

Power Output (W)	
Number of cells on panel	
Area of each cell (cm^2)	
Total area of solar cells (m^2)	
Power per square meter (W/m^2)	
Power from the sun (W/m^2)	
Panel efficiency (%)	

Part II Effect of Wavelength

	Current (A)	Voltage (V)	Illumination (lux)	Power (W)
No filter				
Blue filter				
Red filter				
Green filter				

PROCESSING THE DATA

1. Calculate the average current, voltage and illumination values for Part I.
2. Calculate the power output using the equation

$$P = V I$$

Power = voltage × current

3. Examine the open PV cell and record the number of cells on the panel.
4. Determine the area of one cell in cm^2. Remember, the area of a rectangle is length × width and the area of a triangle is ½ base × height. Draw a diagram of one cell and label any measurements that will help when calculating the area.
5. Calculate the total area of the cells in m^2 using the equation

$$\frac{\text{Number of cells on panel} \times \text{Area of one cell}}{10{,}000\ cm^2/m^2}$$

6. Determine the power per square meter output of the PV cell by dividing the power output by the total area of the cell.
7. Determine the power per square meter output of the sun by dividing the average illumination value by 75 since 1 W/m^2 = 75 lux.
8. Calculate the efficiency of the PV cell using the equation

$$\frac{\text{Power per square meter of PV cell}}{\text{Power per square meter of sun}} \times 100\%$$

9. How does the efficiency of your PV cell compare to your predicted efficiency?
10. What factors may contribute to the lack of efficiency of the PV cell?
11. Calculate the power output for each of the trials in Part II using the equation

$$P = V I$$

Power = voltage × current

12. Which of the colors had the highest power output? Which had the lowest output?
13. What can you conclude about the effect of wavelength on power output?
14. What types of conditions would contribute to an ideal location for PV cells used for electricity to heat a home?

EXTENSIONS

1. Determine the effect of the amount of light shining on a PV cell on the power output.
2. Determine the relationship between the angle of incidence of the light and the power output of a PV cell.
3. Determine whether efficiency depends on the load resistor.

TEACHER INFORMATION

Photovoltaic Cells

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. If you are using calculators for data collection, this activity can be performed with calculators from the TI-83 Plus or TI-84 Plus families and a LabPro or CBL 2. It cannot be performed with Easy products because all runs of data must be collected at the same time under similar conditions.

3. Photovoltaic cells may be purchased from sources such as Radio Shack or Edmund Scientific. The cell used for the design of this experiment is a 1.5 V 200 mA solar cell from Scientifics, Stock No. #B30373-35, www.scientificsonline.com, (1-800-728-6999).

4. Filters may be purchased from science supply companies or your local theatrical lighting store.

5. This experiment works best in bright sunlight. Some solar cells may be sensitive enough to do this experiment with a full spectrum bulb.

6. The illumination value will fluctuate due to obstructions of the sunlight. Have the students record an average illumination value.

7. This experiment was designed using a Current Probe, a Differential Voltage Probe, and a Vernier Light Sensor. If you have older Vernier equipment, be sure to choose the calibration that matches the sensors you are using.

SAMPLE RESULTS

Part I Determining Power Output

	Current (A)	Voltage (V)	Illumination (lux)
Trial 1	0.225	0.200	99800
Trial 2	0.227	0.190	99756
Trial 3	0.228	0.220	99820
Average	0.227	0.203	99792

Power Output (W)	0.046
Number of cells on panel	3
Area of each cell (cm^2)	7.47
Total area of solar cells (m^2)	2.24×10^{-3}
Power per square meter (W/m^2)	20.5
Power from the sun (W/m^2)	3992
Panel efficiency	0.51%

Part II Effect of Wavelength

	Current (A)	Voltage (V)	Illumination (lux)	Power (W)
No filter	0.200	0.225	99800	0.045
Blue filter	0.070	0.093	14800	0.0065
Red filter	0.104	0.127	8008	0.0132
Green filter	0.062	0.084	6650	0.0052

ANSWERS TO QUESTIONS

1–8. See the Sample Results.

9. The efficiency of this PV cell is very low. This experiment was performed in Oregon in April when the sun's light intensity is not at its greatest.

10. The efficiency of a PV cell depends on the type of cell technology, the quality of the cell's materials, the quality of the cell's manufacture, and the intensity of the sun.

11. See the Sample Results.

12. The red filter had the highest power output followed by green and then blue.

13. The PV cell is most sensitive to the longer wavelengths of red light. It is least sensitive to the shorter wavelengths of blue light.

14. An ideal location would have long hours of sunlight that shine directly on the PV cell. A tracking device to turn the PV cells with the sun would result in more energy from cell.

Wind Power

Power from the wind has become an increasingly popular option for electricity generation. Unlike traditional energy sources such as coal, oil, and gas that contribute large quantities of carbon dioxide to the atmosphere, wind power relies on a non-polluting, renewable, ever-present resource—the wind. In recent years, the cost of harnessing energy from the wind has become more affordable making it a viable alternative for many communities.

A wind turbine generally consists of a two- or three-bladed propeller made of aluminum or fiberglass mounted on the top of a tall tower. It converts energy from the mechanical energy of moving air to electrical energy by means of a generator. The wind causes the shaft of the turbine to spin which in turn causes a generator to produce electricity.

In this experiment, you will measure the power output of a wind turbine, investigate the relationship between power output and wind speed, and determine the relationship between power output and rotor shape.

You will use a small motor as a generator and a pinwheel as the turbine. The power output of the pinwheel can be determined by measuring the current and voltage produced by the motor. Power is determined using the relationship

$$P = V I$$

Power = voltage x current

OBJECTIVES

In this experiment, you will

- Use a Current Probe to measure current output.
- Use a Voltage Probe to measure voltage output.
- Calculate power output.
- Determine the relationship between power output and wind speed.
- Determine the relationship between power output and rotor shape.

MATERIALS

computer
Vernier computer interface
Logger*Pro*
Voltage Probe
Current Probe
ring stand
utility clamp
3-speed fan
pinwheel templates of each shape
modeling clay
5 cm piece of drinking straw
scissors
1-hole punch
propeller shaft adapter
3 wire leads with alligator clips
1 Ω resistor
metric ruler
1.5 V DC motor
plastic tubing clamps (optional)

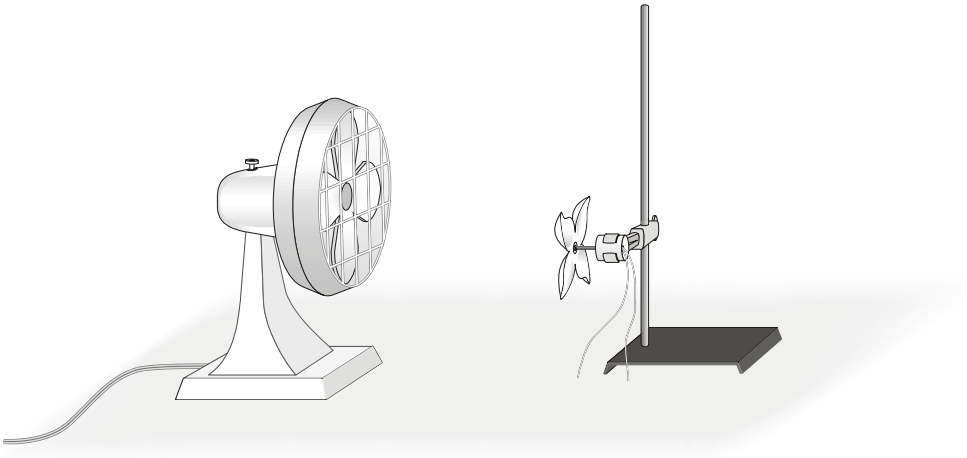


Figure 1

PRE-LAB QUESTIONS

1. In Part I of this experiment, you will increase the wind speed while keeping the rotor diameter constant. What effect do you think this will have on the power output of your wind turbine?

2. In Part II of this experiment, you will change the shape of your rotor while keeping the wind speed constant. What effect do you think this will have on the power output of your wind turbine?

PROCEDURE

Part I Effect of Wind Speed

1. Cut out the square pinwheel design by cutting along all of the lines on the template. Cut out the hole in the center of the pinwheel and on the end of each of the blades using the 1-hole punch. Push the straw through the center of the pinwheel. Carefully bring each of the blades in toward the center of the pinwheel and thread the straw through each of the blades. Be careful not to tear the paper. Place a bit of clay on the end of the straw to keep the blades from spinning off the straw. Put a piece of tape or a plastic tubing clamp on the straw behind the pinwheel to keep the paper from sliding.

2. Measure the diameter of the pinwheel at its widest point and record it in the data table.

3. Put the propeller shaft adapter on the shaft of the motor. Insert the propeller shaft adapter into the end of the straw. You may wish to use a plastic tubing clamp to secure the straw to the shaft. Secure the motor to the ring stand using a utility clamp as shown in Figure 1.

4. Connect the Current Probe to Channel 1 and the Voltage Probe to Channel 2 of the Vernier computer interface.

5. Connect the motor, 1 Ω resistor, wires, and clips as shown in Figure 2. Take care that the red lead from the motor and the red terminal of the Current Probe are connected

6. Prepare the computer for data collection by opening the file "26 Wind Power" in the *Agricultural Science with Vernier* folder of Logger *Pro*.

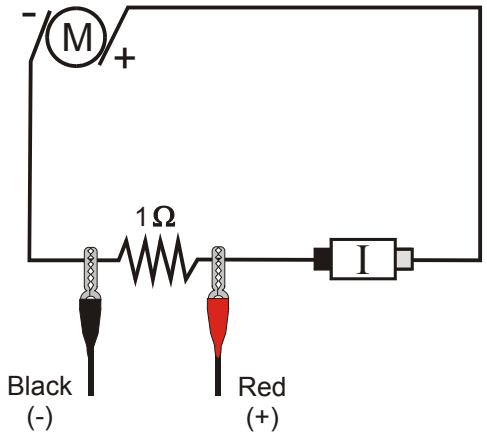


Figure 2

7. Since the direction of spin of the pinwheel depends on its design, you will need to check to see that both the current and voltage readings are positive.
   a. Blow on the pinwheel.
   b. Look at the live readouts and note whether either reading is negative or zero.
   c. If the current reading is negative, disconnect the alligator clips from the wires on the motor and switch them.
   d. If the voltage reading is negative or zero, unclip the voltage probe clips and switch them.

8. Click [Zero]. A dialog box will appear. Click [OK] to zero both sensors. This sets the zero for both probes with no current flowing and with no voltage applied.

9. Place the pinwheel about 15 cm in front of the fan. Turn on the fan to the high setting. Wait for 60 seconds until the fan reaches a constant velocity.

10. Click [Collect] to begin data collection.

11. When data collection is complete turn the fan to the medium setting. Click anywhere on the Current graph. Select the Statistics button, , then click [OK] to display a Statistics box for the Current Probe data. The mean current value is displayed in the statistics box on the graph. Record the mean current value in the data table.

12. Click anywhere on the Voltage graph to select it and click the Statistics button, . Click OK to display a Statistics box for the Voltage Probe data. Record the mean voltage value in the data table.

13. Repeat Steps 9–12 with the fan on the medium setting and then again on the low setting. Be careful not to change the location of the pinwheel or fan between trials.

Part II Effect of Rotor Shape

14. Cut out two of the rectangular pinwheel designs. Fold along the dotted lines and punch out the center holes. Put both pinwheel cutouts on the straw shaft and position them perpendicular to each other. Measure the diameter of the pinwheel and record it in the data table.

15. Place the pinwheel in front of the fan. Turn on the fan to the high setting. Wait for 60 seconds until the fan reaches a constant velocity.

16. Click ▸ Collect to begin data collection.

17. When data collection is complete click anywhere on the Current graph to select it. Click the Statistics button, , then click OK to display a Statistics box for the Current Probe data. Record the mean current value in the data table.

18. Click anywhere on the Voltage graph to select it and click on the Statistics button, . Click OK to display a Statistics box for the Voltage Probe data. Record the mean voltage value in the data table.

19. Repeat Steps 15–18 with the fan on the medium setting and then again on the low setting. Be careful not to change the location of the pinwheel or fan between trials.

DATA

	Square Design			Rectangular Design		
Rotor Diameter (cm)						
	Current (A)	Voltage (V)	Power (W)	Current (A)	Voltage (V)	Power (W)
Low Speed						
Medium Speed						
High Speed						

PROCESSING THE DATA

1. In the space provided in the data table, multiply current and voltage to determine the power output of the turbine.

2. What is the relationship between power output and wind speed in Part 1?

3. What is the relationship between power output and rotor shape?

4. What are some characteristics of an ideal location to build a *wind farm*, a grouping of many wind turbines? What make these characteristics ideal?

5. What are some advantages of using wind power over power from traditional means such as fossil fuels? What are some disadvantages?

EXTENSIONS

1. Compare the power output of rotors made from materials of different stiffnesses.

2. Investigate the effect of rotor shape on power output of other rotor shapes of the same diameter as the ones in this experiment.

3. Investigate the relationship between rotor diameter and power output.

4. Use an anemometer to measure the wind speed in each of the trials in Part 1. Determine the mathematical relationship between wind speed and power output.

TEACHER INFORMATION

Wind Power

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. If you are using calculators for data collection, this activity can be performed with calculators from the TI-83 Plus or TI-84 Plus families and a LabPro or CBL 2. It can not be performed with Easy products because all runs of data must be collected at the same time under similar conditions.

3. Copies made on card stock work well for the pinwheel.

4. This experiment was designed using a 1.5–3.0 Volt DC Motor, # 273-223, available from Radio Shack.

5. The resistor should be a 1 Ω 10 W power resistor.

6. The propeller adapter, Graupner Propeller adapter No. 6053.20, is available from Hobby Lobby International, Inc., 615-373-1444, www.hobby-lobby.com. They are the United States distributor for Graupner products. It may also be available from your local hobby store.

7. Clay inserted in the straw before mounting on the propeller adapter seems to hold the pinwheel nicely in place.

8. Plastic tubing clamps (pkg of 100) are available from Vernier Software & Technology, Order Code PTC.

9. The rectangular design propeller may not turn when the fan is on the low setting.

10. This experiment was designed using a Differential Current Probe and a Voltage Probe. It may be done using the Current Probe from the Current & Voltage System. It may also be done using a ULI Voltage Probe or the Voltage Probe from the Current & Voltage System. Make sure to choose the calibrations that match the sensors you are using.

SAMPLE RESULTS

	Square Design			Rectangular Design		
Rotor Diameter (cm)	12.7			10.2		
	Current (A)	Voltage (V)	Power (W)	Current (A)	Voltage (V)	Power (W)
Low Speed	0.017	0.004	6.8×10^{-5}	-	-	-
Medium Speed	0.034	0.015	5.1×10^{-4}	0.017	0.004	6.8×10^{-5}
High Speed	0.057	0.039	2.2×10^{-3}	0.028	0.010	2.8×10^{-4}

ANSWERS TO QUESTIONS

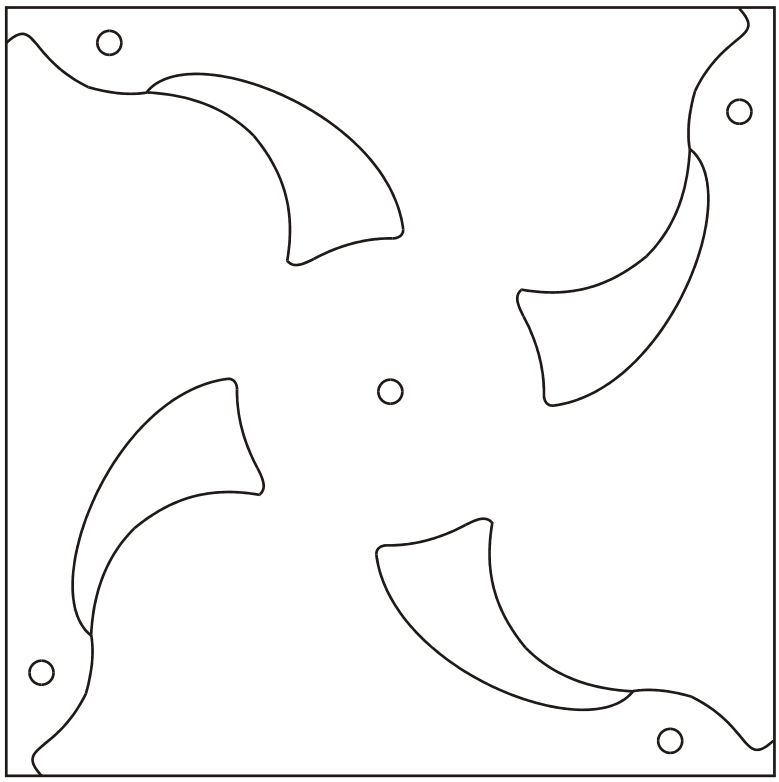
1. See the Sample Results.

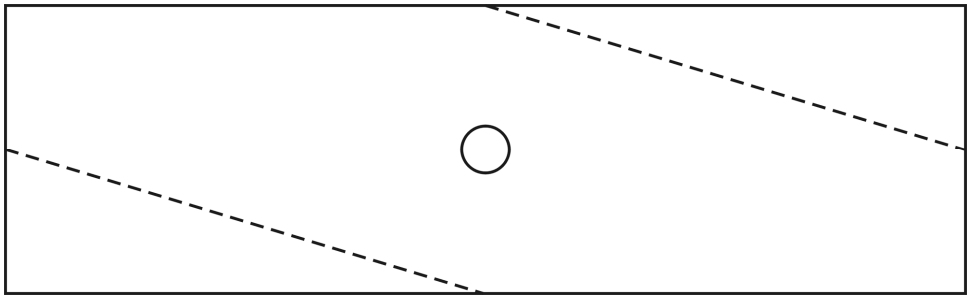
2. In Part I, the power output increases as the wind speed increases.

3. In Part II, the power output of the square design rotor is much higher than that of the rectangular design rotor.

4. A wind farm should be located where the wind blows regularly and at optimum speeds for the rotor design. These conditions are ideal since the turbine only produces power when it is turning. The energy output of a wind turbine is determined by multiplying the power output by the amount of time the power is produced. The longer the turbine spins, the more power is produced.

5. Wind power is beneficial because it uses fewer non-renewable resources and causes less air pollution than traditional coal-fired power plants. The turbines also require little land space leaving more land for farming.





Watershed Testing

There are many reasons for determining water quality. You may want to compare the water quality upstream and downstream to locate a possible source of pollutants along a river or stream. Another reason may be to track the water quality of a watershed over time by making measurements periodically. When comparing the quality of a watershed at different times, it is important that measurements be taken from the same location and at the same time of day.

In 1970, the National Sanitation Foundation, in cooperation with 142 state and local environmental specialists and educators, devised a standard index for measuring water quality. This index, known as the Water Quality Index, or *WQI*, consists of nine tests to determine water quality. These nine tests are; temperature, pH, turbidity, total solids, dissolved oxygen, biochemical oxygen demand, phosphates, nitrate, and fecal coliform. A graph for each of the nine tests indicates the water quality value (or Q-value) corresponding to the data obtained. Once the Q-value for a test has been determined, it is multiplied by a weighting factor. Each of the tests is weighted based on its relative importance to a stream's overall quality. The resulting values for all nine tests are totaled and used to gauge the stream's health (excellent, good, medium, poor, or very poor).

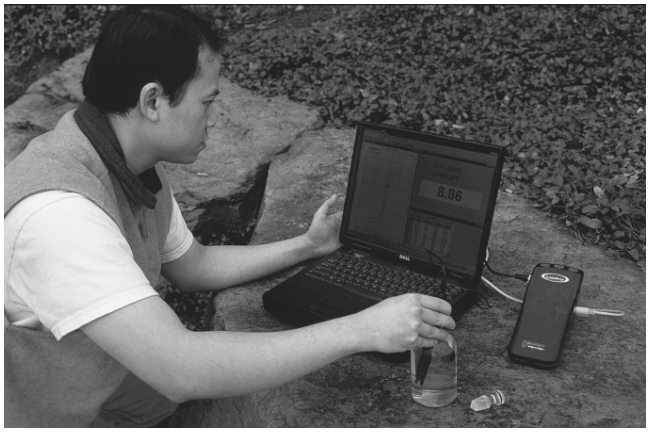
While the WQI can be a useful tool, it is best used in light of historical data. Not all streams are the same, and without historical data it is difficult to determine if a stream is truly at risk. For example, a stream may earn a very low WQI value and appear to be in poor health. By looking at historical data, however, you may find that samples were collected just after a heavy rain with an overflow from the local city sewer system and do not accurately reflect the stream's health.

For the purpose of this exercise, you will perform only four of the WQI tests: water temperature, dissolved oxygen, pH, and total dissolved solids. A modified version of the WQI for these four tests, will allow you to determine the general quality of the stream or lake you are sampling.

OBJECTIVES

In this experiment, you will

- Use a Dissolved Oxygen, Temperature, Conductivity, and pH Probe to make on-site measurements.
- Calculate the water quality based on your findings.



MATERIALS

computer (laptop)
Vernier computer interface
Vernier Temperature Probe
Vernier Conductivity Probe
Vernier pH Sensor
Vernier Dissolved Oxygen Probe
Logger*Pro*
water sampling bottle, stoppered
small plastic cup or beaker

PRE-LAB PROCEDURE

Important: Prior to each use, the Dissolved Oxygen Probe must warm up for a period of 10 minutes as described below. If the probe is not warmed up properly, inaccurate readings will result. Perform the following steps to prepare the Dissolved Oxygen Probe.

1. Prepare the Dissolved Oxygen Probe for use.
   a. Remove the blue protective cap.
   b. Unscrew the membrane cap from the tip of the probe.
   c. Using a pipet, fill the membrane cap with 1 mL of DO Electrode Filling Solution.
   d. Carefully thread the membrane cap back onto the electrode.
   e. Place the probe into a container of water.

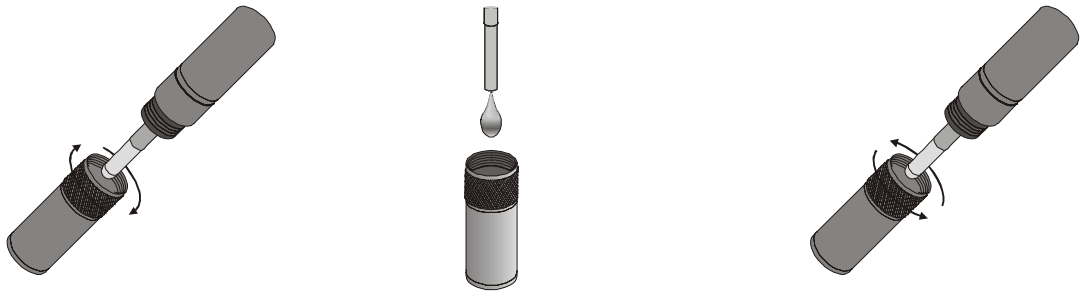


Figure 1

2. Connect the Dissolved Oxygen Probe to Channel 1 of the Vernier computer interface. Connect the Conductivity Probe to Channel 2. Set the selector switch on the side of the Conductivity Probe to the 0–2000 µS/cm range.
3. Prepare the computer for data collection by opening the file "27a Watershed Testing" from the *Agricultural Science with Vernier* folder of Logger*Pro*. The meters display the live dissolved oxygen and TDS (Total Dissolved Solids) readings.
4. It is necessary to warm up the Dissolved Oxygen Probe for 5–10 minutes before taking readings. To warm up the probe, leave it connected to the interface, with Logger *Pro* running. The probe must stay connected at all times to keep it warmed up. If disconnected for a few minutes, it will be necessary to warm up the probe again.
5. You are now ready to calibrate the Dissolved Oxygen Probe.
   - If your instructor directs you to use the calibration stored in the experiment file, then continue to the Procedure.
   - If your instructor directs you to perform a new calibration for the Dissolved Oxygen Probe, use the procedure at the end of this lab.

PROCEDURE

The data collection is performed in two parts. First, you will measure dissolved oxygen and total dissolved solids concentration of the water sample from the lake or stream being studied. Then you will measure pH and temperature of the lake or stream water.

Measuring Dissolved Oxygen and Total Dissolved Solids

Because the Dissolved Oxygen Probe requires 5–10 minutes to polarize before it can be used, it will already be connected to the interface, and will be in a sample of distilled water.
Important: The warm-up must be done in order to get accurate dissolved oxygen readings.

6. Choose a desirable location to perform your measurements. It is best to obtain samples as far from the shore edge as is safe. Your site should be representative of the watershed as a whole.
7. Rinse the sampling bottle out a few times with stream water. Fill the sampling bottle so that it is completely full and stopper the bottle under water. This should minimize the amount of atmospheric oxygen that gets into the water until the measurements have been made.
8. Position the computer and the probes safely away from the water. The computer will be damaged if it gets wet.
9. Remove the Dissolved Oxygen Probe from the storage bottle. Place the probe into the water sampling bottle. Gently and continuously swirl it to allow water to move past the probe's tip. After 30 seconds, or when the dissolved oxygen reading stabilizes, record the dissolved oxygen reading, in mg/L, in Table 1. Return the Dissolved Oxygen Probe to the storage bottle.
10. Place the Conductivity Probe into the water sampling bottle and gently swirl to allow water to move past the probe's tip. The Conductivity Probe is measuring Total Dissolved Solids (TDS). When the TDS reading stabilizes, record it in Table 1. Disconnect the Dissolved Oxygen Probe and Conductivity Probes from the interface. Important: Handle both probes with care. The Dissolved Oxygen Probe should be reconnected to the interface as soon as Step 12 is completed (so that it remains polarized).

Measuring Temperature and pH

11. Connect the Temperature Probe to Channel 1 and the pH Sensor to Channel 2. Prepare the computer for data collection by opening the file "27b Watershed Testing" from the *Agricultural Science with Vernier* folder of Logger*Pro*.
12. Place the Temperature Probe into the center of the water sampling bottle. When the reading stabilizes, record the temperature reading in Table 1. Disconnect the Temperature Probe from the interface and reconnect the Dissolved Oxygen Probe (so it stays polarized).
13. Using the small plastic cup, obtain some stream water to rinse the pH Sensor.
14. Remove the pH Sensor from the storage bottle. Rinse the pH electrode thoroughly with the stream water. Then place the electrode into the water sampling bottle and gently swirl. When the reading stabilizes, record the pH value in Table 1.
15. Repeat Steps 2–3 and 6–14 at one more location 6 meters from the first location.
16. Repeat Steps 2–3 and 6–14 at two locations about 1.6 km from the first location.

DATA

Table 1				
Location	Dissolved oxygen (mg/L)	pH	Total dissolved solids (mg/L)	Temperature (°C)
Site 1a				
Site 1b				
Average				
Site 2a				
Site 2b				
Average				

Temperature Difference: _____

Table 2 DO (% Saturated)			
	Dissolved oxygen (mg/L)	DO in saturated water	% Saturated
Site 1			
Site 2			

PROCESSING THE DATA

1. Calculate the averages for measurements at each location and record the results in Table 1.
2. Determine the % saturation of dissolved oxygen:
   a. Copy the value of dissolved oxygen measured at each site from Table 1 to Table 2.
   b. Obtain the barometric pressure, in mm Hg, using either a barometer or a table of barometric pressure values according to elevation (your instructor will provide either the barometer reading or the table of values).
   c. Note the water temperature at each site.
   d. Using the pressure and temperature values, look up the level of dissolved oxygen for air-saturated water (in mg/L) from a second table provided by your instructor. Record the results for each site in Table 2.
   e. To determine the % saturation, use this formula:

$$\% \text{ saturation} = \frac{\text{measured D.O. level}}{\text{saturated D.O. level}} \times 100$$

   f. Record the % saturation of dissolved oxygen in Table 3.

3. Using Tables 3–5, determine the water quality value (Q value) for each of the following measurements: dissolved oxygen, pH, and TDS. You may need to interpolate to obtain the correct Q values. Record your result in Table 7 for Site 1 and in Table 8 for Site 2.

Table 3	
Dissolved Oxygen (DO) Test Results	
DO (% saturation)	Q Value
0	0
10	5
20	12
30	20
40	30
50	45
60	57
70	75
80	85
90	95
100	100
110	95
120	90
130	85
140	80

Table 4	
pH Test Results	
pH	Q Value
2.0	0
2.5	1
3.0	3
3.5	5
4.0	8
4.5	15
5.0	25
5.5	40
6.0	54
6.5	75
7.0	88
7.5	95
8.0	85
8.5	65
9.0	48
9.5	30
10.0	20
10.5	12
11.0	8
11.5	4
12.0	2

4. Subtract the two average temperatures from the sites that are about 1.6 km apart. Record the result as the temperature difference in the blank below Table 1.

5. Using Table 6 and the value you calculated above, determine the water quality value (Q value) for the temperature difference measurement. You may need to interpolate to obtain the correct Q values. Record your result in Table 7 and Table 8. The temperature Q-value will be the same in both tables.

6. Multiply each Q-value by the weighting factor in Table 7 for Site 1 and in Table 8 for Site 2. Record the total Q-value in Tables 7–8.

7. Determine the overall water quality of your stream by adding the four total Q-values in Table 7 for Site 1 and in Table 8 for Site 2. Record the result in the line next to the label "Overall Quality." The closer this value is to 100, the better the water quality of the stream at this site.

   Note that this quality index is not a complete one—this value uses only four measurements. For a more complete water quality determination, you should measure fecal coliform counts, biological oxygen demand, phosphate and nitrate levels, and turbidity. It is also very valuable to do a "critter count"—that is, examine the macroinvertebrates in the stream.

Table 5	
Total Dissolved Solids (TDS) Test Results	
TDS (mg/L)	Q Value
0	80
50	90
100	85
150	78
200	72
250	65
300	60
350	52
400	46
450	40
500	30

Table 6	
Temperature Test Results	
ΔTemp (°C)	Q Value
0	95
5	75
10	45
15	30
20	20
25	15
30	10

Table 7 Site 1			
Test	Q-Value	Weight	Total Q-Value
DO		0.38	
pH		0.24	
TDS		0.16	
Temperature		0.22	

Overall Quality: _____

Table 8 Site 2			
Test	Q-Value	Weight	Total Q-Value
DO		0.38	
pH		0.24	
TDS		0.16	
Temperature		0.22	
		Overall Quality:	

QUESTIONS

1. Using your measurements, what is the quality of the watershed? Explain.
2. How do you account for each of the measurements? For example, if the pH of the downstream site is very low, and you took measurements above and below an auto repair station, perhaps battery acid leaked into the stream.

   DO:

   pH:

   TDS:

   Temperature:
3. How did measurements between the two sites compare? How might you account for any differences, if any?
4. Compare the measurements you obtained with those from previous months or years. Has the water quality improved, remained about the same, or declined? Explain.
5. Why would you expect the DO in a pond to be less than in a rapidly moving stream? If applicable, did your measurements confirm this assumption? Explain.
6. What could be done to improve the quality of the watershed?

CALIBRATION PROCEDURE

1. Perform the calibration.

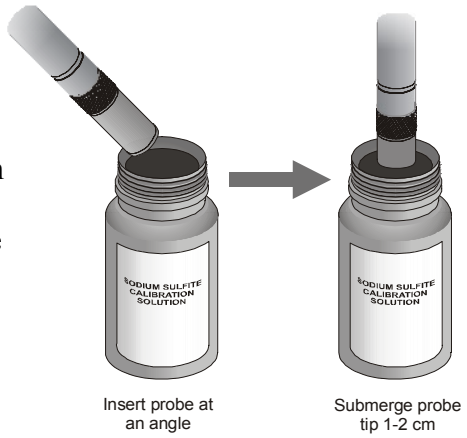


Figure 2

Zero-Oxygen Calibration Point

a. Choose Calibrate ▸ CH1: Dissolved Oxygen (mg/L) from the Experiment menu and then click Calibrate Now.
b. Remove the probe from the water and place the tip of the probe into the Sodium Sulfite Calibration Solution. **Important:** No air bubbles can be trapped below the tip of the probe or the probe will sense an inaccurate dissolved oxygen level. If the voltage does not rapidly decrease, tap the side of the bottle with the probe to dislodge any bubbles. The readings should be in the 0.2 to 0.5 V range.
c. Type **0** (the known value in mg/L) in the edit box.
d. When the displayed voltage reading for Reading 1 stabilizes, click Keep.

Saturated DO Calibration Point

e. Rinse the probe with distilled water and gently blot dry.
f. Unscrew the lid of the calibration bottle provided with the probe. Slide the lid and the grommet about 2 cm onto the probe body.

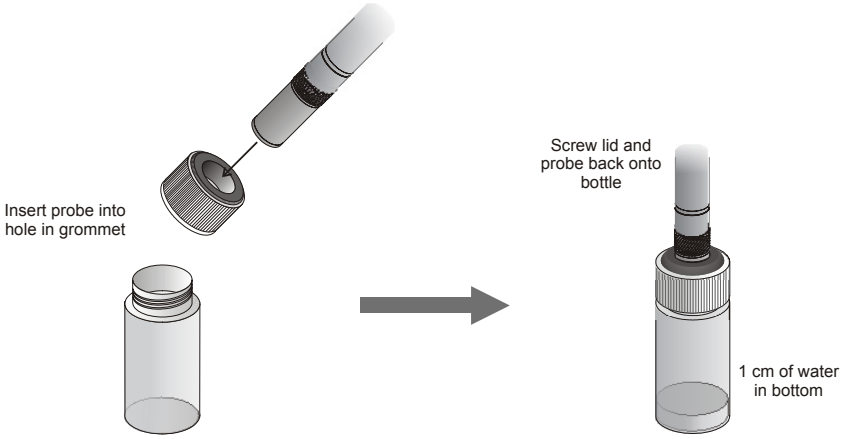


Figure 3

g. Add water to the bottle to a depth of about 1 cm and screw the bottle into the cap, as shown. **Important:** Do not touch the membrane or get it wet during this step. Keep the probe in this position for about a minute.
h. Type the correct saturated dissolved-oxygen value (in mg/L) from Table 9 (for example, **8.66**) using the current barometric pressure and air temperature values. If you do not have the current air pressure, use Table 10 to estimate the air pressure at your altitude.
i. When the reading for Reading 2 stabilizes (readings should be above 2.0 V), click Keep.
j. Choose Calibration Storage from the calibration pull-down menu. Choose Experiment File (calibration stored with the current document). Click Done.
k. From the File menu, select Save As. Save the current experiment file with a new name.
l. Prepare the probe for transport by filling the calibration bottle half full with water. Secure the Dissolved Oxygen Probe far enough down in the bottle that the membrane is completely covered by water. Screw the calibration bottle lid completely onto the bottle so that no water will leak out.

CALIBRATION TABLES

Table 9: 100% Dissolved Oxygen Capacity (mg/L)												
	770 mm	**760 mm**	**750 mm**	**740 mm**	**730 mm**	**720 mm**	**710 mm**	**700 mm**	**690 mm**	**680 mm**	**670 mm**	**660 mm**
0°C	14.76	14.57	14.38	14.19	13.99	13.80	13.61	13.42	13.23	13.04	12.84	12.65
1°C	14.38	14.19	14.00	13.82	13.63	13.44	13.26	13.07	12.88	12.70	12.51	12.32
2°C	14.01	13.82	13.64	13.46	13.28	13.10	12.92	12.73	12.55	12.37	12.19	12.01
3°C	13.65	13.47	13.29	13.12	12.94	12.76	12.59	12.41	12.23	12.05	11.88	11.70
4°C	13.31	13.13	12.96	12.79	12.61	12.44	12.27	12.10	11.92	11.75	11.58	11.40
5°C	12.97	12.81	12.64	12.47	12.30	12.13	11.96	11.80	11.63	11.46	11.29	11.12
6°C	12.66	12.49	12.33	12.16	12.00	11.83	11.67	11.51	11.34	11.18	11.01	10.85
7°C	12.35	12.19	12.03	11.87	11.71	11.55	11.39	11.23	11.07	10.91	10.75	10.59
8°C	12.05	11.90	11.74	11.58	11.43	11.27	11.11	10.96	10.80	10.65	10.49	10.33
9°C	11.77	11.62	11.46	11.31	11.16	11.01	10.85	10.70	10.55	10.39	10.24	10.09
10°C	11.50	11.35	11.20	11.05	10.90	10.75	10.60	10.45	10.30	10.15	10.00	9.86
11°C	11.24	11.09	10.94	10.80	10.65	10.51	10.36	10.21	10.07	9.92	9.78	9.63
12°C	10.98	10.84	10.70	10.56	10.41	10.27	10.13	9.99	9.84	9.70	9.56	9.41
13°C	10.74	10.60	10.46	10.32	10.18	10.04	9.90	9.77	9.63	9.49	9.35	9.21
14°C	10.51	10.37	10.24	10.10	9.96	9.83	9.69	9.55	9.42	9.28	9.14	9.01
15°C	10.29	10.15	10.02	9.88	9.75	9.62	9.48	9.35	9.22	9.08	8.95	8.82
16°C	10.07	9.94	9.81	9.68	9.55	9.42	9.29	9.15	9.02	8.89	8.76	8.63
17°C	9.86	9.74	9.61	9.48	9.35	9.22	9.10	8.97	8.84	8.71	8.58	8.45
18°C	9.67	9.54	9.41	9.29	9.16	9.04	8.91	8.79	8.66	8.54	8.41	8.28
19°C	9.47	9.35	9.23	9.11	8.98	8.86	8.74	8.61	8.49	8.37	8.24	8.12
20°C	9.29	9.17	9.05	8.93	8.81	8.69	8.57	8.45	8.33	8.20	8.08	7.96
21°C	9.11	9.00	8.88	8.76	8.64	8.52	8.40	8.28	8.17	8.05	7.93	7.81
22°C	8.94	8.83	8.71	8.59	8.48	8.36	8.25	8.13	8.01	7.90	7.78	7.67
23°C	8.78	8.66	8.55	8.44	8.32	8.21	8.09	7.98	7.87	7.75	7.64	7.52
24°C	8.62	8.51	8.40	8.28	8.17	8.06	7.95	7.84	7.72	7.61	7.50	7.39
25°C	8.47	8.36	8.25	8.14	8.03	7.92	7.81	7.70	7.59	7.48	7.37	7.26
26°C	8.32	8.21	8.10	7.99	7.89	7.78	7.67	7.56	7.45	7.35	7.24	7.13
27°C	8.17	8.07	7.96	7.86	7.75	7.64	7.54	7.43	7.33	7.22	7.11	7.01
28°C	8.04	7.93	7.83	7.72	7.62	7.51	7.41	7.30	7.20	7.10	6.99	6.89
29°C	7.90	7.80	7.69	7.59	7.49	7.39	7.28	7.18	7.08	6.98	6.87	6.77
30°C	7.77	7.67	7.57	7.47	7.36	7.26	7.16	7.06	6.96	6.86	6.76	6.66

Table 10: Approximate Barometric Pressure at Different Elevations					
Elevation (m)	**Pressure (mm Hg)**	**Elevation (m)**	**Pressure (mm Hg)**	**Elevation (m)**	**Pressure (mm Hg)**
0	760	800	693	1600	628
100	748	900	685	1700	620
200	741	1000	676	1800	612
300	733	1100	669	1900	604
400	725	1200	661	2000	596
500	717	1300	652	2100	588
600	709	1400	643	2200	580
700	701	1500	636	2300	571

TEACHER INFORMATION

Watershed Testing

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. When setting up the Dissolved Oxygen Probe, be sure to remove the blue plastic cap from the end of the probe. The cap is made of a soft plastic material and easily slides off the probe end.

3. In order for the Dissolved Oxygen Probe to warm up and stay polarized, power to the sensor must be continuous. LabPro, LabQuest, LabQuest Mini, and CBL 2 deliver continuous power once the data-collection software is started even if the screen goes to sleep. However, EasyLink used with a TI-84 graphing calculator and the EasyData App stops powering the sensor when the calculator goes to sleep. The calculator goes to sleep to conserve battery power if no user interaction is detected for 3 minutes. If power to the sensor is disrupted, the sensor must be warmed up for another 5–10 minutes before calibrating or taking readings. To avoid having to warm up the sensor again, students must press a button on the calculator every few minutes to keep the calculator awake.

4. The Dissolved Oxygen Probe must be calibrated the day of use. Follow the pre-lab procedure to prepare and calibrate the Dissolved Oxygen Probe. To save time, you may wish to calibrate the probe and record the calibration values on paper. The students can then skip the pre-lab procedure and they will have the calibration values available for manual entry in case the values stored in the program are lost.

5. To ensure the most accurate measurements of pH, the pH System should be calibrated prior to use. Refer to the teacher's section of experiment 18 for additional calibration information.

6. When transporting the Dissolved Oxygen Probe to the field site, you should store it in the plastic calibration bottle filled with distilled water. This plastic bottle is shipped with the Dissolved Oxygen Probe. It is important that the students understand the fragile nature of the electrode membrane and proper handling procedures.

7. A glass-stoppered water sampling bottle is recommended for collecting samples. Filling this bottle to the brim, followed by stoppering, will prevent additional oxygen from dissolving after water is collected.

8. Two sites 1.6 km apart should be selected for comparison. Have students take samples at two points for each site. Each of the sample points should be approximately 6 m (20 feet) apart.

9. To determine the D.O. concentration for a solution saturated with dissolved oxygen, refer to Table 9 and Table 10. **Important:** Be sure to bring a copy of these tables on the day you collect and test water samples! Use Table 10 to estimate barometric pressure using your approximate elevation above sea level. Temperature and barometric pressure values can then be used in Table 9 to determine the saturated level of dissolved oxygen, in mg/L. Use this formula to calculate % saturation of dissolved oxygen:

$$\% \text{ saturation} = \frac{\text{measured D.O. level}}{\text{saturated D.O. level}} \times 100$$

10. When measuring total dissolved solids, you may wish to have students use the 0–200 µS/cm (equal to 100 mg/L TDS) range to improve accuracy. This should only be done if TDS levels are below 100 mg/L.

11. A more complete water quality index can be obtained by measuring fecal coliform counts; biological oxygen demand, phosphate and nitrate levels, and turbidity. It is also very valuable to do a "critter count"—that is, examine the macroinvertebrates in the stream.

    For more information on the Water Quality Index, you may be interested in the Vernier book, *Water Quality with Vernier*.

SAMPLE DATA

Table 1				
Location	Dissolved Oxygen (mg/L)	pH	Total Dissolved Solids (mg/L)	Temperature (°C)
Site 1 Average	10.2	7.4	88.4	11.0
Site 2 Average	8.1	7.4	94.0	8.0

Table 2 DO (% Saturated)			
	Dissolved Oxygen (mg/L)	DO in Saturated Water	% Saturated
Site 1	10.2	11.1	91.9
Site 2	8.1	11.9	68.0

Table 7 Site 1			
Test	Q-Value	Weight	Total Q-Value
DO	97	0.38	36.9
pH	95	0.24	22.8
TDS	84	0.16	13.4
Temperature	85	0.22	18.7
		Overall Quality:	91.8

Table 8 Site 2			
Test	Q-Value	Weight	Total Q-Value
DO	70	0.38	26.6
pH	95	0.24	22.8
TDS	83	0.16	13.3
Temperature	85	0.22	18.7
		Overall Quality:	81.4

ANSWERS TO QUESTIONS

1. The water quality indices for the above sites are 91.8 and 81.4. These are very high indices, considering that they were obtained in an urban Seattle watershed. The first site was from a small, rapidly moving stream (~3.4 m^3/s), and the second from a pond 1.6 km upstream. Other measurements corroborated these measurements—the water quality was very high.

2. Answers will vary.

3. Answers will vary. The two sites compared equally except for the DO value. Since water at the second site was hardly moving, it had less dissolved oxygen than in rapidly moving, highly aerated water.

4. Answers will vary.

5. Water in rapidly moving stream is aerated as it flows through riffles, and may have more dissolved oxygen than in slowly moving water.

6. Answers will vary.

Interdependence of Plants and Animals

Plants and animals share many of the same chemicals throughout their lives. In most ecosystems, O_2, CO_2, water, food and nutrients are exchanged between plants and animals. In this lab, you will be designing your own experiments to determine the relationships between two organisms—a plant (Elodea) and an animal (a snail).

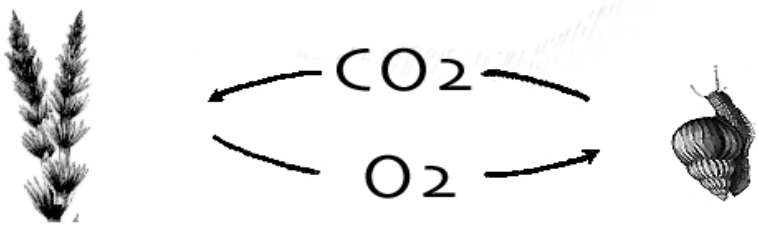


Figure 1

Several hypotheses have been discussed in the past about possible relationships between the Elodea and the snail. You will test to determine how oxygen and CO_2 are exchanged among Elodea plants, snails, and the water both exist in.

To perform the necessary tests, you will need to determine the presence of CO_2. An easy way to do this is to monitor the pH of the pond water. If CO_2 dissolves in water, it forms carbonic acid, H_2CO_3, and the pH decreases. If CO_2 is removed from pond water, the amount of carbonic acid goes down and the pH increases. One can use a computer to monitor the pH and determine whether CO_2 is released into the pond water or is taken from the water. Dissolved oxygen (DO) can be monitored with the aid of a Dissolved Oxygen Probe. Increases or decreases in the amount of dissolved oxygen can be rapidly assessed with this probe.

OBJECTIVES

In this experiment, you will

- Use a computer and a Dissolved Oxygen Probe to measure the dissolved oxygen in water.
- Use a computer and a pH Sensor to measure the pH of water.
- Use pH measurements to make inferences about the amount of CO_2 dissolved in water.
- Determine whether snails consume or produce oxygen and CO_2 in water.
- Determine whether plants consume or produce oxygen and CO_2 in the light.
- Determine whether plants consume or produce oxygen and CO_2 in the dark.

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier Dissolved Oxygen Probe
Vernier pH Sensor
eight 25 × 150 mm screw top test tubes
distilled wash water
250 mL beaker
Aluminum foil
Parafilm
pond snails
pond water
sprigs of elodea
two test tube racks

PRE-LAB PROCEDURE

Important: Prior to each use, the Dissolved Oxygen Probe must warm up for a period of 10 minutes as described below. If the probe is not warmed up properly, inaccurate readings will result. Perform the following steps to prepare the Dissolved Oxygen Probe.

1. Prepare the Dissolved Oxygen Probe for use.
   a. Remove the blue protective cap.
   b. Unscrew the membrane cap from the tip of the probe.
   c. Using a pipet, fill the membrane cap with 1 mL of DO Electrode Filling Solution.
   d. Carefully thread the membrane cap back onto the electrode.
   e. Place the probe into a container of water.

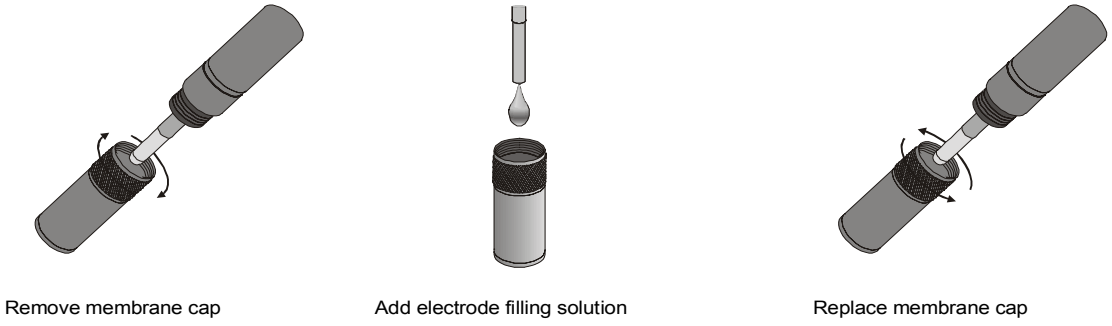


Figure 2

2. Connect the Dissolved Oxygen Probe to Channel 2 of the Vernier interface. Connect the pH Sensor to Channel 1.
3. Prepare the computer for data collection by opening the file "28 Plants and Animals" from the *Agricultural Science with Vernier* folder of Logger*Pro*. Two meters will be displayed. The left one is set to read pH while the right one is set for dissolved oxygen in mg/L.
4. It is necessary to warm up the Dissolved Oxygen Probe for 5–10 minutes before taking readings. To warm up the probe, leave it connected to the interface, with Logger *Pro* running. The probe must stay connected at all times to keep it warmed up. If disconnected for a few minutes, it will be necessary to warm up the probe again.
5. You are now ready to calibrate the Dissolved Oxygen Probe.
   - If your instructor directs you to use the calibration stored in the experiment file, then proceed to Step 6.
   - If your instructor directs you to perform a new calibration for the Dissolved Oxygen Probe, use the procedure at the end of this lab.

PROCEDURE

Day 1

6. Obtain and wear an apron.
7. Obtain and label eight test tubes 1–8.
8. Set test tubes 1–4 in one test tube rack and test tubes 5–8 in a second test tube rack.

9. Fill each tube with pond water.

10. Place one snail each in test tubes 2, 4, 6, and 8.

11. Place one sprig of elodea in test tubes 3, 4, 7, and 8. Each set of four tubes should appear similar to those in Figure 3.

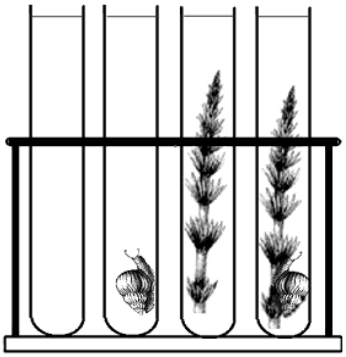


Figure 3

12. Wrap test tubes 5–8 in aluminum foil to make each light tight.

13. Remove the pH Sensor from the storage bottle. Rinse the probe thoroughly with distilled water. Place the probe into test tube 1 and gently swirl to allow water to move past the probe's tip. When the reading displayed in the meter stabilizes, record the pH value in Table 1.

14. Repeat Step 13 for each of the other seven test tubes.

15. When all of the pH readings have been taken, rinse the pH Sensor and return it to the pH storage bottle.

16. Place the Dissolved Oxygen Probe into test tube 1 so that it is submerged half the depth of the water. Gently and continuously move the probe up and down a distance of about 1 cm in the tube. This allows water to move past the probe's tip. Note: Do not agitate the water, or oxygen from the atmosphere will mix into the water and cause erroneous readings.

17. When the dissolved oxygen reading stabilizes (~30 seconds), record its value in Table 1.

18. Repeat Steps 16–17 for each of the other seven test tubes.

19. When all of the dissolved oxygen readings have been taken, rinse the Dissolved Oxygen Probe and return it to the distilled water beaker.

20. Completely fill each test tube with pond water and tighten the cap onto the tube. Do not allow any air bubbles to remain in any of the test tubes. Unscrew each cap slightly, so that they just barely open. Wrap each tube with Parafilm so that they do not leak water. The Parafilm will expand, if necessary, to accommodate any pressure build-up in a tube. No oxygen or carbon dioxide should enter or leave a tube.

21. Place test tubes 1–8 near the light source, as directed by your instructor.

22. Predict how the pH and dissolved oxygen will change in each tube. Write a short statement that explains your reasoning. Be specific about the roles of both the snail and elodea. Be prepared to discuss your reasoning in class on Day 2.

Day 2

23. Open the file saved on Day 1. Repeat Steps 1–6 to set up the pH Sensor and Dissolved Oxygen Probe.

24. Repeat Steps 13–21 to take pH and DO readings for each of the test tubes.

25. Now, the elodea will use the environment established by the snail and the snail will use the environment established by the elodea. Remove the snail from test tube 2 and the elodea from test tube 3. Place the snail in test tube 3 and the elodea in test tube 2. **Note**: Try not to aerate the water during the transfer.

26. Remove the snail from test tube 6 and the elodea from test tube 7. Place the snail in test tube 7 and the elodea in test tube 6.

27. Measure the pH and DO of test tubes 1–3 and test tubes 5–7. Record the results in Table 2. These values should be similar to those measured before the transfer. If not, the water may have been mixed too vigorously with the atmospheric air.

28. Completely fill test tubes 1–3 and test tubes 5–7 with water and tighten the cap onto each tube, as in Step 20. Wrap each slightly opened test tube with Parafilm. Place test tubes 1–3 and test tubes 5–7 near the light source, as in Step 21.

29. Return the snails and elodea from test tubes 4 and 8, as directed by your instructor. Clean and return the test tubes.

Day 3

30. Repeat Steps 13–19 for test tubes 1–3 and 5–7. Record the results in Table 2.

31. Return the snails and elodea, as directed by your instructor. Clean and return the test tubes.

DATA

Table 1						
Test Tube	pH Day 1	pH Day 2	ΔpH	DO Day 1	DO Day 2	ΔDO
1						
2						
3						
4						
5						
6						
7						
8						

Table 2						
Test Tube	pH Day 2	pH Day 3	ΔpH	DO Day 2	DO Day 3	ΔDO
1						
2						
3						
5						
6						
7						

PROCESSING THE DATA

1. Calculate the change in pH for Tables 1–2. Record your results in Tables 1–2.
2. Calculate the change in dissolved oxygen for Tables 1–2. Record your results in Tables 1–2.

QUESTIONS

1. Consider the snails. Comparing the readings from day 1 to day 2, answer the following questions:
   a. Do snails produce or consume CO_2 when in the light?
   b. Do snails produce or consume oxygen when in the light?

c. Which test tubes allow you to answer this? Which test tube is the experimental test tube? Which test tube is the control test tube?
d. Do snails produce or consume CO_2 when in the dark?
e. Do snails produce or consume oxygen when in the dark?
f. Which test tubes allow you to answer this? Which test tube is the experimental test tube? Which test tube is the control test tube?

2. Consider the elodea. Comparing the readings from day 1 to day 2, answer the following questions:

a. Do elodea produce or consume CO_2 when in the light?
b. Do elodea produce or consume oxygen when in the light?
c. Which test tubes allow you to answer this? Which test tube is the experimental test tube? Which test tube is the control test tube?
d. Do elodea produce or consume CO_2 when in the dark?
e. Do elodea produce or consume oxygen when in the dark?
f. Which test tubes allow you to answer this? Which test tube is the experimental test tube? Which test tube is the control test tube?

3. Consider the elodea placed in the snail's water on days 2–3. Comparing the readings from day 2 to day 3, answer the following questions:

a. Do elodea produce or consume CO_2 when in the light?
b. Do elodea produce or consume oxygen when in the light?
c. Which test tubes allow you to answer this? Which test tube is the experimental test tube? Which test tube is the control test tube?
d. Do elodea produce or consume CO_2 when in the dark?
e. Do elodea produce or consume oxygen when in the dark?
f. Which test tubes allow you to answer this? Which test tube is the experimental test tube? Which test tube is the control test tube?

4. Consider the snail placed in the elodea's water on days 2–3. Comparing the readings from day 2 to day 3, answer the following questions:

a. Do snails produce or consume CO_2 when in the light?
b. Do snails produce or consume oxygen when in the light?
c. Which test tubes allow you to answer this? Which test tube is the experimental test tube? Which test tube is the control test tube?
d. Do snails produce or consume CO_2 when in the dark?
e. Do snails produce or consume oxygen when in the dark?
f. Which test tubes allow you to answer this? Which test tube is the experimental test tube? Which test tube is the control test tube?

5. Summarize the relationship between snails and plants in a pond. Explain your reasoning.

6. Interpret the results of Test Tube 4 and Test Tube 8. Compare your findings to the results obtained from Table 2.

7. How do your conclusions compare to your predictions in Step 22?

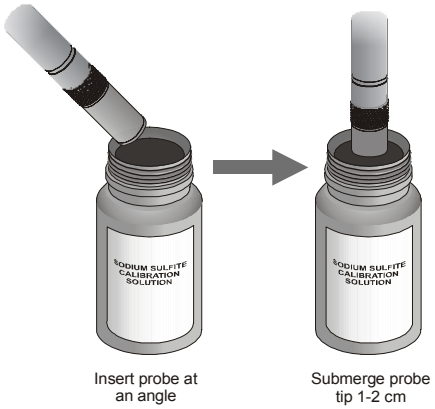
CALIBRATION PROCEDURE

1. Perform the calibration.

   a. Choose Calibrate ▸ CH2: Dissolved Oxygen (mg/L) from the Experiment menu and then click Calibrate Now.

   Zero-Oxygen Calibration Point

   b. Remove the probe from the water and place the tip of the probe into the Sodium Sulfite Calibration Solution. **Important:** No air bubbles can be trapped below the tip of the probe or the probe will sense an inaccurate dissolved oxygen level. If the voltage does not rapidly decrease, tap the side of the bottle with the probe to dislodge any bubbles. The readings should be in the 0.2 to 0.5 V range.

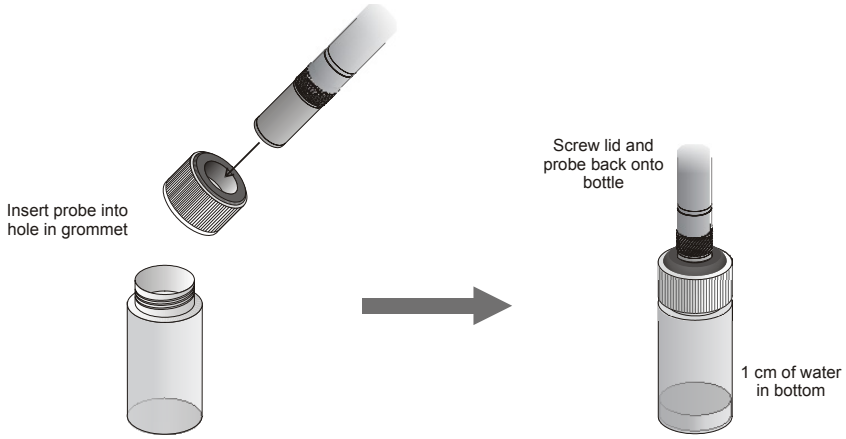
   c. Type **0** (the known value in mg/L) in the edit box.

   d. When the displayed voltage reading for Reading 1 stabilizes, click Keep.

   Saturated DO Calibration Point

   e. Rinse the probe with distilled water and gently blot dry.

   f. Unscrew the lid of the calibration bottle provided with the probe. Slide the lid and the grommet about 2 cm onto the probe body.

   g. Add water to the bottle to a depth of about 1 cm and screw the bottle into the cap, as shown. **Important:** Do not touch the membrane or get it wet during this step. Keep the probe in this position for about a minute.

   h. Type the correct saturated dissolved-oxygen value (in mg/L) from Table 3 (for example, **8.66**) using the current barometric pressure and air temperature values. If you do not have the current air pressure, use Table 4 to estimate the air pressure at your altitude.

   i. When the displayed voltage reading for Reading 2 stabilizes (readings should be above 2.0 V), click Keep.

   j. Choose Calibration Storage from the calibration pull-down menu. Choose Experiment File (calibration stored with the current document). Click Done.

   k. From the File menu, select Save As and save the current experiment file with a new name.

CALIBRATION TABLES

Table 3: 100% Dissolved Oxygen Capacity (mg/L)												
	770 mm	**760 mm**	**750 mm**	**740 mm**	**730 mm**	**720 mm**	**710 mm**	**700 mm**	**690 mm**	**680 mm**	**670 mm**	**660 mm**
0°C	14.76	14.57	14.38	14.19	13.99	13.80	13.61	13.42	13.23	13.04	12.84	12.65
1°C	14.38	14.19	14.00	13.82	13.63	13.44	13.26	13.07	12.88	12.70	12.51	12.32
2°C	14.01	13.82	13.64	13.46	13.28	13.10	12.92	12.73	12.55	12.37	12.19	12.01
3°C	13.65	13.47	13.29	13.12	12.94	12.76	12.59	12.41	12.23	12.05	11.88	11.70
4°C	13.31	13.13	12.96	12.79	12.61	12.44	12.27	12.10	11.92	11.75	11.58	11.40
5°C	12.97	12.81	12.64	12.47	12.30	12.13	11.96	11.80	11.63	11.46	11.29	11.12
6°C	12.66	12.49	12.33	12.16	12.00	11.83	11.67	11.51	11.34	11.18	11.01	10.85
7°C	12.35	12.19	12.03	11.87	11.71	11.55	11.39	11.23	11.07	10.91	10.75	10.59
8°C	12.05	11.90	11.74	11.58	11.43	11.27	11.11	10.96	10.80	10.65	10.49	10.33
9°C	11.77	11.62	11.46	11.31	11.16	11.01	10.85	10.70	10.55	10.39	10.24	10.09
10°C	11.50	11.35	11.20	11.05	10.90	10.75	10.60	10.45	10.30	10.15	10.00	9.86
11°C	11.24	11.09	10.94	10.80	10.65	10.51	10.36	10.21	10.07	9.92	9.78	9.63
12°C	10.98	10.84	10.70	10.56	10.41	10.27	10.13	9.99	9.84	9.70	9.56	9.41
13°C	10.74	10.60	10.46	10.32	10.18	10.04	9.90	9.77	9.63	9.49	9.35	9.21
14°C	10.51	10.37	10.24	10.10	9.96	9.83	9.69	9.55	9.42	9.28	9.14	9.01
15°C	10.29	10.15	10.02	9.88	9.75	9.62	9.48	9.35	9.22	9.08	8.95	8.82
16°C	10.07	9.94	9.81	9.68	9.55	9.42	9.29	9.15	9.02	8.89	8.76	8.63
17°C	9.86	9.74	9.61	9.48	9.35	9.22	9.10	8.97	8.84	8.71	8.58	8.45
18°C	9.67	9.54	9.41	9.29	9.16	9.04	8.91	8.79	8.66	8.54	8.41	8.28
19°C	9.47	9.35	9.23	9.11	8.98	8.86	8.74	8.61	8.49	8.37	8.24	8.12
20°C	9.29	9.17	9.05	8.93	8.81	8.69	8.57	8.45	8.33	8.20	8.08	7.96
21°C	9.11	9.00	8.88	8.76	8.64	8.52	8.40	8.28	8.17	8.05	7.93	7.81
22°C	8.94	8.83	8.71	8.59	8.48	8.36	8.25	8.13	8.01	7.90	7.78	7.67
23°C	8.78	8.66	8.55	8.44	8.32	8.21	8.09	7.98	7.87	7.75	7.64	7.52
24°C	8.62	8.51	8.40	8.28	8.17	8.06	7.95	7.84	7.72	7.61	7.50	7.39
25°C	8.47	8.36	8.25	8.14	8.03	7.92	7.81	7.70	7.59	7.48	7.37	7.26
26°C	8.32	8.21	8.10	7.99	7.89	7.78	7.67	7.56	7.45	7.35	7.24	7.13
27°C	8.17	8.07	7.96	7.86	7.75	7.64	7.54	7.43	7.33	7.22	7.11	7.01
28°C	8.04	7.93	7.83	7.72	7.62	7.51	7.41	7.30	7.20	7.10	6.99	6.89
29°C	7.90	7.80	7.69	7.59	7.49	7.39	7.28	7.18	7.08	6.98	6.87	6.77
30°C	7.77	7.67	7.57	7.47	7.36	7.26	7.16	7.06	6.96	6.86	6.76	6.66

Table 4: Approximate Barometric Pressure at Different Elevations					
Elevation (m)	**Pressure (mm Hg)**	**Elevation (m)**	**Pressure (mm Hg)**	**Elevation (m)**	**Pressure (mm Hg)**
0	760	800	693	1600	628
100	748	900	685	1700	620
200	741	1000	676	1800	612
300	733	1100	669	1900	604
400	725	1200	661	2000	596
500	717	1300	652	2100	588
600	709	1400	643	2200	580
700	701	1500	636	2300	571

TEACHER INFORMATION

Interdependence of Plants and Animals

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. The Dissolved Oxygen Probe must be calibrated the first day of use. Follow the pre-lab procedure to prepare and calibrate the Dissolved Oxygen Probe. To save time, you may wish to calibrate the probe and record the calibration values on paper. The students can then skip the pre-lab procedure. They will have the calibration values available for manual entry in case the values stored in the program are lost.

3. In order for the Dissolved Oxygen Probe to warm up and stay polarized, power to the sensor must be continuous. LabPro, LabQuest, LabQuest Mini, and CBL 2 deliver continuous power once the data-collection software is started even if the screen goes to sleep. However, EasyLink used with a TI-84 graphing calculator and the EasyData App stops powering the sensor when the calculator goes to sleep. The calculator goes to sleep to conserve battery power if no user interaction is detected for 3 minutes. If power to the sensor is disrupted, the sensor must be warmed up for another 5–10 minutes before calibrating or taking readings. To avoid having to warm up the sensor again, students must press a button on the calculator every few minutes to keep the calculator awake.

4. As a time-saving measure, instruct the students at the end of class to leave the data-collection program running. This will keep power going to the probes. When the next group of students comes in, they can begin at Step 6 of the procedure. They can skip the pre-lab procedure because the initial group of students has completed all of the setup. Have the last group of students for the day shut everything off and put things away.

5. The pond water should be adjusted to pH 7 before class begins. Use 0.1 M NaOH or 0.1 M HCl to adjust the pH. Be sure the elodea are fresh and healthy.

6. Florescent lamps should be used as a source of light. They should be on for the entire 24-hour period, set a few inches from the tubes. If the tubes are water tight, as they should be, test tube racks are not necessary. Students can place them horizontally on a table and the light can be lowered until it is just above the tubes.

7. Wrap the test tubes thoroughly in aluminum foil if they require darkness, or place them in a darkened part of the room. If there are not a sufficient number of test tube racks for these, place the set of four wrapped tubes in a small beaker.

8. Between classes, store the Dissolved Oxygen Probes in a beaker of distilled water. At the end of the day, be sure to empty out the electrode filling solution in the Dissolved Oxygen Probe and rinse the inside of the membrane cap with distilled water.

9. If you have a pH System, but do not have a Dissolved Oxygen Probe, the experiment may be modified to indirectly investigate carbon dioxide levels using only the pH System.

10. When taking dissolved oxygen readings, the students should allow ample time for the readings to stabilize. In some instances this can take 60 seconds.

11. Each student team should use the same set of equipment to make measurements each day.

12. When setting up the Dissolved Oxygen Probe, be sure to remove the blue plastic cap from the end of the probe. The cap is made of a soft plastic material and easily slides off the probe end.

13. Elodea canadensis is a good alternative for those who live in any area to which it is illegal to ship Elodea. Other aquatic plants may work equally well.

ANSWERS TO QUESTIONS

1. Consider the snails.
   a. Snails produce CO_2 when in the light. The pH decreased, meaning the acidity increased. Higher acidity means more CO_2 is dissolved. A snail was the only organism present to produce the CO_2.
   b. Snails consume O_2 when in the light. The DO decreased, so less O_2 is dissolved. A snail was the only organism present to consume the O_2.
   c. Experimental Test Tube: 2. Control Test Tube: 1.
   d. Snails produce CO_2 when in the dark. The pH decreased, so the acidity increased. Higher acidity means more CO_2 is dissolved. A snail was the only organism present to produce the CO_2.
   e. Snails consume O_2 in the dark. The DO decreased, so less O_2 is dissolved. A snail was the only organism present to consume the O_2.
   f. Experimental Test Tube: 6. Control Test Tube: 5.

2. Consider the Elodea.
   a. Elodea consume CO_2 when in the light. The pH increased, so the acidity decreased. Lower acidity means less CO_2 is dissolved. Elodea was the only organism present to consume the CO_2.
   b. Elodea produce O_2 when in the light. The DO increased, so more O_2 is dissolved. Elodea were the only organism present to make the O_2.
   c. Experimental Test Tube: 3. Control Test Tube: 1.
   d. Elodea produce CO_2 in the dark. The pH decreased, so the acidity increased. Higher acidity means more CO_2 is dissolved. Elodea were the only organism present to produce the CO_2.
   e. Elodea consume O_2 in the dark. The DO decreased, so less O_2 is dissolved. Elodea was the only organism present to consume the O_2.
   f. Experimental Test Tube: 7. Control Test Tube: 5.

3. Consider the elodea placed in the snail's water on days 2–3.
   a. Elodea consumes the CO_2 that snails release when in the light. CO_2 increased when a snail was alone in the pond water, yet it decreased when elodea replaced the snail. Some of the CO_2 used by the plant must have come from the snail. The increase in pH was greater for elodea in Tube 2 of Table 2 than in Tube 3, Table 1, so more CO_2 was consumed from water the snail was in.
   b. Elodea produces O_2 when in the light, as above. The DO increased, so more O_2 is dissolved.
   c. Experimental Test Tube: 2. Control Test Tube: 1.

d. The pH change in Test Tube 6 from Day 2 to Day 3 was negative. This indicates that the plant did not remove the CO_2; rather, it was added as the plant respired. Elodea did not consume CO_2 in the dark, and did not use the CO_2 from the snail.
e. Elodea consumed O_2 when in the dark. The DO decreased, so the plant respired when in the dark.
f. Experimental Test Tube: 6. Control Test Tube: 5.

4. Consider the snail placed in the elodea's water on days 2–3.
   a. The snail did release CO_2 in the light, as during Day 1 – Day 2. The pH decreased, so the acidity increased. Higher acidity means more CO_2 is dissolved.
   b. Snails consume O_2 that elodea produce in the light. 2b (above) shows that plants make O_2 in the light. The DO change in Test Tube 3 from Day 2 to Day 3 was negative. This indicated that the snail removed the O_2 made by elodea.
   c. Experimental Test Tube: 3. Control Test Tube: 1.
   d. The snail did release CO_2 in the dark, as the pH decreased.
   e. The snail did consume O_2 while in the dark, as the DO decreased.
   f. Experimental Test Tube: 7. Control Test Tube: 5.

5. Here is a summary of the relationship between snails and plants in a pond:
   - Snails can produce CO_2 in both light and dark conditions.
   - Elodea produce CO_2 in the dark, but consume CO_2 when illuminated with light.
   - In the light, elodea can use the CO_2 that snails produce.
   - In the dark, elodea cannot use the CO_2 that snails produce.
   - Snails can consume O_2 in both light and dark conditions.
   - Elodea produce O_2 in the light, but consume O_2 when in the dark.
   - In the light, snails can use the O_2 elodea produce.
   - In the dark, elodea do not produce O_2, so snails cannot use it.

6. Answers may vary. The answer depends upon the rate of photosynthesis vs. the rate of respiration in Test Tube 4.
   - If the rate of respiration by the snail and plant was greater than the rate of photosynthesis, 3a and 3b might be answered differently. The pH would remain low and the amount of DO would also be low.
   - If the rate of respiration by the snail and plant was less than the rate of photosynthesis, 4a and 4b might be answered differently. The pH difference would be higher, indicating a removal of CO_2. The amount of dissolved oxygen would also be high. This might mask the respiration by both plant and animal.

7. Answers may vary.

Biodiversity and Ecosystems

Biodiversity is critical in any self-sustaining environment. Complex and diverse ecological systems are made up of many organisms and a huge variety of interactions. Simple ecosystems have few organisms, few interactions, and are often fragile. All ecosystems, whether diverse or sparse, involve an intimate interaction of living things with their abiotic environment. Biodiversity implies variety, and variety in an ecosystem often ensures a greater chance of survival in a changing world.

The Earth is losing its biodiversity at a worrisome rate. Humans simplify ecosystems for many reasons: to increase the agricultural base, to make way for cities and industrial zones, or for aesthetic reasons, such as making lawns and gardens. This practice has direct effects upon many abiotic factors within an environment. The air temperatures found in cities, for instance, are usually significantly higher than that in surrounding, non-urbanized areas. Such cities are said to produce *heat islands*. An area's biodiversity has profound effects upon the physical and biological makeup of an ecosystem.

OBJECTIVES

In this experiment, you will

- Examine how biodiversity affects an environment's temperature.
- Determine how animal diversity changes in different environments.
- Work with your classmates to compare biodiversity in areas with different plant patch sizes.

MATERIALS

computer
Vernier computer interface
Temperature Probe
Logger *Pro*
notebook
meter stick
string or twine

PROCEDURE

1. Choose two sites, one that is diverse with a fair variety of different types of plants. Call this Site A. Find a simple site, such as a grassy lawn. Call this Site B. Two such sites might be like those shown here:

Site A

Site B

2. Connect the Temperature Probe to the computer interface. Prepare the computer for data collection by opening the file "29 Biodiversity" from the *Agricultural Science with Vernier* folder of Logger *Pro*.

Site A:

3. Using a meter stick, measure out a one-square meter area at Site A. Mark the area with string or twine.

Identifying organisms

4. Examine this area closely. Your group will need to make several decisions:
   - How will we identify an organism? What is a grass organism? One blade? A group from one set of roots? A patch of grass?
   - How will we count similar organisms? If there are thousands of one type of organism in your area, do you count each one, or find ways of estimating? What ways of estimating are reasonably accurate? Since many animals and birds move in and out of an area, over what time period will you count organisms?
   - How will we identify a plant patch? A patch of one type of plant is an area of similar plants that are physically separate from another area of similar plants. Patches need not be the same size. You will need to distinguish one patch from another.

   Discuss how your group will determine each of these.

   Record your decisions and the rationale for your choices.

5. Now, record information about the different living organisms in your area. Include estimates of the following:
   - The type of organism. The actual name is not important. You might write something like:
     - a maple tree
     - tall (20–30 cm) grass with wide blades
     - cut grass, 3 cm long
     - beetle #1 (0.5 cm long, black)
     - bee #1 (yellow and black stripes about 1 cm long)
     - a fly flew in and out of the area
   - the approximate number of each type of organism.
   - the number of plant patches in the 1 m^2 area.

6. Using a soil borer or a trowel, examine a small sample of soil. Record the depth of the humus in your soil in Table 1. Humus is made of decaying organic matter and is usually darker in color than the non-organic soil. Note any animals in your sample.

7. Examine your data from Steps 5 and 6 and record each of the following in Table 1:
   a. The number of different animals in your area
   b. The number of different plants in your area
   c. The number of plant patches in your area

Identifying physical factors

8. Place a meter stick vertically in your area. One end of the stick (reading 0 cm) should be on the soil. If leaf litter exists, move it aside and place the stick on the dirt. The other end (100 cm) should be in the air, so that the stick is as vertical as possible.

9. Place the temperature probe at the top of the meter stick (at a height of 100 cm).

10. You are ready to begin the measurements. Start measuring by clicking Collect. Note that a new button, Keep, is available.

11. Allow the temperature reading to stabilize then click Keep. This button tells the computer that you want to record a measurement.

12. A text box will appear. Enter the height of the sensor. At a height of 100 cm, type **100** in the text box and press ENTER.

13. Move the sensor down to 90 cm. Repeat Steps 11 and 12.

14. Continue measuring the temperature at heights of 80, 70, 60, 50, 40, 30, 20, 10, and 0 cm. To do so, repeat the procedure in Step13. When all measurements have been made click Stop.

15. Obtain the temperature values from the table and record them in Table 2.

16. Move your data to a stored data run. To do this, choose Store Latest Run from the Experiment menu. This will allow you to keep the first plot on the screen while you are measuring Site B.

Site B

17. Repeat Steps 5–15 at Site B. Save your data when finished.

Class Data

18. Obtain the data found by each team in your class and record their values in Table 3. Calculate the averages for each column and record your results in the bottom row of Table 3.

DATA

Table 1		
Count in your area	Site A (diverse)	Site B (sparse)
Number of different animals		
Number of different plants		
Number of plant patches		
Humus depth (cm)		

Table 2											
Height (cm)	100	90	80	70	60	50	40	30	20	10	0
Site A Temperature (°C)											
Site B Temperature (°C)											

Table 3								
Team	# of Animals		# of Plants		# of Patches		Humus depth (cm)	
	Site A	Site B	Site A	Site B	Site A	Site B	Site A	Site B
1								
2								
3								
4								
5								
6								
7								
8								
9								
10								
Average								

QUESTIONS

1. How would you compare Sites A and B? Include a comparison of the biodiversity at each site.

2. Which site supported a *larger* animal population? Which site supported a *more diverse* animal population? Hypothesize why this might be so.

3. Examine your data in Table 3. How does the average number of plants compare to the average number of plant patches?

4. What seems to be a more meaningful indicator of biodiversity—a count of the number of plants, or a count of the number of plant patches? Explain your answer.

5. How would each group's definition of what a plant was affect the results in Table 3 and the answer to Question 4?

6. Compare your plots of temperature *vs*. height for the two sites. If you performed Extension 1, compare plots of the measurements for the two sites.

7. Using your answer to Question 6, summarize how living organisms affect both biotic and abiotic factors in an ecosystem.

8. Which ecosystem, the complex one found in Site A or the simple one found in Site B, requires a greater expenditure of human resources to maintain? Explain your answer.

9. If each ecosystem experienced a fundamental environmental change, which would be more likely to survive? Explain your reasoning.

10. Summarize your conclusions of this field experiment.

EXTENSIONS

1. What other environmental measurements (in addition to temperature) could be taken that would help one understand how abiotic variables are affected by living organisms? Design an experiment to measure these variables.

2. Write an essay that discusses how an animal might perceive a patch site.

3. Make two possible food pyramids from your data, one from Site A and the other from Site B. Discuss how biodiversity affects the two food pyramids.

TEACHER INFORMATION

Biodiversity and Ecosystems

1. The student pages with complete instructions for data-collection using LabQuest App, Logger Pro (computers), and EasyData (calculators) can be found on the CD that accompanies this book. See *Appendix A* for more information.

2. If possible, this lab should be performed on a warm, dry day.

3. Students will need class time to discuss how they will define their plants and plant patches. This should be done while (or after) the students visualize their study areas. Since groups will be sharing data, it is best if the class comes to a common agreement prior to data collection.

   A patch is an area where plants are separate from other plants. This can be a single organism or a large number of similar organisms. Students will need to determine how to distinguish one patch from another.

4. Have student groups select a wide variety of locations for Site A. This will provide ample information when the groups share data. The area should have some plants at least 0.5 m tall.

5. A lawn or playing field is a good choice for Site B. They usually are part of a very simple ecosystems.

6. Allow sufficient time at each site so that students observe animals that move into and out of their site. Their site includes a vertical column as high as they can observe.

7. It is strongly recommended that students perform Extension 1. In the data below, light and relative humidity were measured in addition to temperature. Students may need class time to decide what additional measurements to take. You will need to advise students what sensors and probes you have available.

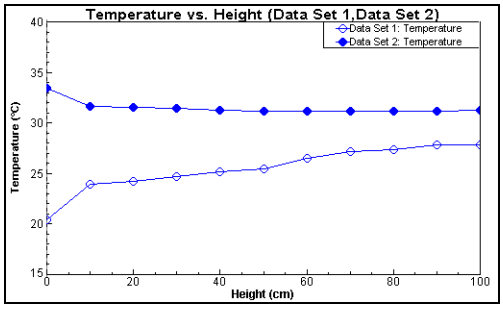
SAMPLE RESULTS

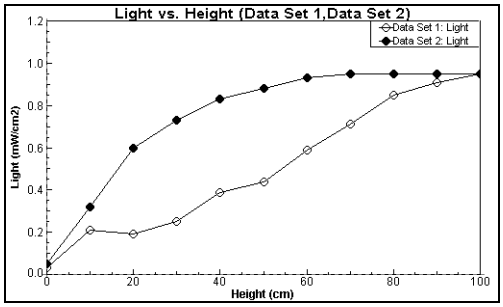
The following data may be different from students' results. The data below came from the two sites in Figure 1. In this discussion, Extension 1 was performed using a light and relative humidity sensor.

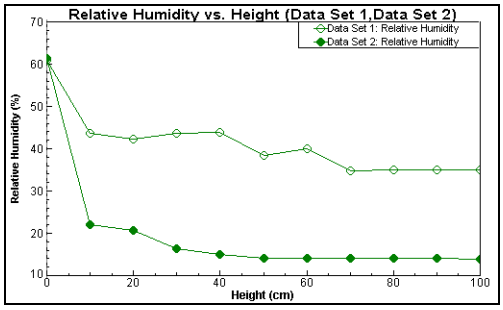
Table 1

Count in your area	Site A	Site B
Number of different animals	8	0
Number of different plants	9	2
Number of plant patches	14	4
Humus depth (cm)	5	1

Table 2						
	Site A (diverse)			Site B (sparse)		
Height (cm)	Temp (°C)	Relative humidity (%)	Light	Temp (°C)	Relative humidity (%)	Light
100	27.8	35.0	0.95	31.3	13.9	0.95
90	27.8	35.0	0.91	31.2	14.0	0.95
80	27.4	34.9	0.85	31.2	14.1	0.95
70	27.2	34.8	0.71	31.2	14.0	0.95
60	26.5	39.9	0.59	31.2	14.1	0.93
50	25.5	38.3	0.44	31.2	14.1	0.88
40	25.2	43.9	0.39	31.3	15.0	0.83
30	24.7	43.7	0.25	31.4	16.4	0.73
20	24.2	42.2	0.19	31.5	20.6	0.60
10	23.9	43.6	0.21	31.6	22.1	0.32
0	20.4	61.0	0.030	33.4	61.3	0.05







These plots were obtained from the sites in Figure 1.

At Site A, the relative humidity remains high, the temperatures are quite cool, and the light reflected is lower due to the plant life.

At Site B, a grassy lawn, the relative humidity is high only at ground level, the temperatures are consistently high, and the light reflected is consistently higher. This provides a much narrower habitat range than in Site A.

ANSWERS TO QUESTIONS

1. Answers will vary. In this sample, 17 types of organisms were found at Site A, while only two were at Site B. The biodiversity at Site A was greater than at Site B.

2. Site A supported more kinds and a higher population of animals. The range of habitats available to animals was greater, as was the potential food supply.

3. Answers may vary, although generally, the number of different patches of plants is greater in the more diverse area. The number of individual plants in either area may be quite high, however.

4. In the study area measured above, there are thousands of blades of grass, but grass was one of only two types of plants present at site B. A count of the number of plant patches seems to be a more meaningful indicator of biodiversity than a count of the number of plants.

5. Using different definitions will introduce extra variables into the experiment. For instance, if two groups examined the same area and obtained vastly different numbers, the two group's combined data would appear to be totally unrelated. If one group's method of defining plants and/or patches was used to measure all of the sites, the numbers could be compared in a meaningful way.

6. In this discussion, Extension 1 was performed using a light and relative humidity sensor.
   a. At Site A, the temperatures are quite cool due to the plant life. At Site B, a grassy lawn, the temperatures are consistently high. They may be highest at the ground level, since there is no shade.
   b. The light increases dramatically when there is little vegetation, and is not as intense when plants absorb light energy.
   c. At Site A, the relative humidity remains high due to the plant life. At Site B, a grassy lawn, the relative humidity is high only at ground level. It plummets drastically just above the height of the grass.

7. A diverse habitat provides a wide range of environments for plants and animals to live. The hot, dry conditions found in Site B are inhospitable to life compared to the moist, cool environment found in Site A. The plants at Site A tend to
   - provide shade, keeping the animals, the plant roots, the soil, and some plants cool.
   - trap the moisture, so that it does not evaporate into the atmosphere as rapidly.
   - provide a rich and more varied food supply.
   - keep the temperatures moderate and consistent under the shade.

8. Many resources were used to maintain a grassy lawn. Fossil fuels were combusted, metals processed for the mower, and human time and energy were constantly applied to maintain a simple ecosystem. Few human energy or resources were expended to maintain Site A—it was left alone for over 30 years.

9. Site A is more resilient to change. For instance, if a disease came though that destroyed one specie of grass, Site B might be devastated, while Site A would only be slightly affected. Because of the many different types of plants and animals present, the disappearance of any one organism would not be devastating at Site A.

Appendix

A

Using the Electronic Resources

The electronic resources, accessed through your account at www.vernier.com, contain the following:

- **Student**
  - **EasyData**–Contains the calculator instructions for each of the 29 student experiments in this book.
  - **LabQuest**–Contains the LabQuest App instructions for each of the 29 student experiments in this book.
  - **Logger Pro**–Contains the computer instructions for each of the 29 student experiments in this book.
- **Teacher–** Contains the teacher information pages for each experiment in this book.

Using the *Agricultural Science with Vernier* Word-Processing Files

All file names begin with the experiment number, followed by an abbreviation of the title; e.g., 01 Intro Data Collection is the file name used for *Experiment 1, Introduction to Data Collection.* This provides a way for you to edit the experiments to match your lab situation, your equipment, or your style of teaching. The files contain all figures, text, and tables in the same format as printed in *Agricultural Science with Vernier*.

Appendix

B

Using Logger *Pro* to Transfer Data to a Computer

You may elect to use the Vernier Logger *Pro* program to transfer data from LabQuest or TI graphing calculator to a computer. Logger *Pro* has many graphing features, such as labels and units for axes, autoscaling, and modification of axes. Printed graphs will have a better resolution and appearance than printed screens of the LabQuest or TI graphing calculator display. Data tables can be displayed and printed with side-by-side columns and headings. Logger *Pro* also provides advanced data-analysis features, such as curve fitting, statistical analysis, and calculated spreadsheet columns.

The directions below are for Logger *Pro* version 3.6 or newer.

Transferring data from LabQuest

If you are not prompted to retrieve the data from LabQuest, Logger *Pro* can open files saved in the LabQuest application.

1. Connect LabQuest to your computer with a USB cable.
2. Start Logger *Pro* on your computer.
3. Choose LabQuest Browser ► Open… from the File menu.
4. You'll see a standard file selection dialog showing the files available on your LabQuest. Select the file name you want, and click Open. Logger *Pro* will open the LabQuest file, displaying any data, graphs, and notes.

Transferring data from a TI graphing calculator

Logger *Pro* software has an option for importing data lists from all of the TI graphing calculators.

1. Connect the TI Connectivity cable or the USB direct cable to the serial or USB port of your computer and to the port on your calculator.
2. Turn on the calculator.
3. Start Logger *Pro* on your computer.
4. Choose Import from►TI Device from the File menu. A dialog box appears with directions for importing data.
5. From the pull-down menu, choose the USB port or serial port (COM 1-4 on a PC, modem or printer port on a Macintosh) to which the cable is connected.[1]
6. Click on the Scan for Calculator button. The calculator model you are using should now be identified, and you should see a message, "Ready to Import."

[1] If you are using a PC serial cable, identify whether it is a gray or black cable.

7. Select the lists that you wish to import by clicking on each of them. (To select more than one list on a Macintosh, hold down the Command and Shift keys while you click.)
8. Click OK to send the lists to the computer. The lists will appear in columns in the data table in Logger *Pro*. They will be labeled with the simple list names from the calculator. If you want to rename them or add units, double-click on the heading in the data table and enter new labels and units.
9. Click the Refresh Catalog button if you have connected a new interface or calculator to the computer.

Appendix C

Vernier Products for Agricultural Science

All software and laboratory interfacing hardware required for the experiments contained in this book are available from Vernier Software & Technology and can be found in this appendix.

LabQuest

Vernier LabQuest provides a portable and versatile data-collection device for any class studying biology. It can be used as a computer interface, as a stand-alone device, or in the field. It has built-in graphing and analysis software and a vivid color touch screen. It is compatible with existing Vernier sensors. It has a rechargeable, high-capacity internal battery. It also has a built-in temperature sensor and microphone.

LabQuest Mini

The Vernier LabQuest Mini is a low-cost data-collection interface that connects to the USB port of a computer and has five sensor ports.

LabPro

Vernier LabPro offers another option for data collection in biology. A wide variety of Vernier probes and sensors can be connected to each of the four analog ports and two sonic/digital ports. LabPro is connected to a computer using a serial or USB port or to a TI graphing calculator.

EasyLink

EasyLink is a single channel interface that plugs into the USB port of the TI-84 Plus or TI-84 Plus Silver Edition calculator. It is used with the EasyData App for data collection. EasyLink's flexibility and ease of use make it perfect for a variety of activities in science and math. It supports over 30 analog sensors, including Gas Pressure, pH, and Conductivity, among others.

Data-Collection Software

Computer

Logger *Pro* is the data-collection software for collecting data on a computer. Logger *Pro* software now comes with a free site license for both Windows and Macintosh, so you only need to order one copy of Logger *Pro* for your school or college department.

Software	Macintosh and Windows CD
Logger *Pro*	(order code: LP)

LabQuest

LabQuest App is the data-collection application used to collect data when using LabQuest as a standalone device.

Calculator

The EasyData App controls the data gathering process, and makes data analysis easier after experiments are completed. See *Appendix B* for information on transferring the program to your calculator.

Vernier Products for Agricultural Science

Item	Order Code
Vernier LabQuest	LABQ
Vernier LabQuest Mini	LQ-MINI
Vernier LabPro interface	LABPRO
Vernier EasyLink	EZ-LINK
CO_2 Gas Sensor	CO2-BTA
Conductivity Probe	CON-BTA
Current Probe	DCP-BTA
Differential Voltage Probe	DVP-BTA
Dissolved Oxygen Probe	DO-BTA
Gas Pressure Sensor	GPS-BTA
Light Sensor	LS-BTA
O_2 Gas Sensor	O2-BTA
Tris-Compatible Flat pH Sensor	FPH-BTA
Soil Moisture Sensor	SMS-BTA
Stainless Steel Temperature Probe	TMP-BTA

Vernier Sensors for Agricultural Science

CO_2 Gas Sensor

The CO_2 Gas Sensor measures gaseous carbon dioxide levels It has two settings: low range (0–10,000 ppm) and high range (0–100,000 ppm). This probe is great for measuring changes in CO_2 levels during plant photosynthesis and respiration. With this sensor, you can easily monitor changes in CO_2 levels occurring in respiration of organisms as small as crickets or beans! A chamber with probe attachment is included for running controlled experiments with small plants and animals.

Conductivity Probe

This probe is great for environmental testing for salinity, total dissolved solids (TDS), or conductivity in water samples. Biology students can use it to investigate the difference between ionic and molecular compounds, strong and weak acids, salinity, or ionic compounds that yield different ratios of ions. The Conductivity Probe can monitor concentration or conductivity at three different sensitivity settings: 0–200 µS/cm, 0–2000 µS/cm, and 0–20,000 µS/cm.

Current Probe and Differential Voltage Probe

Use our Current Probe and/or Differential Voltage Probe to monitor currents and voltages in low-voltage DC and AC circuits. The differential voltage range is ±6 V. The current range is ±0.6 A. These sensors are ideal for most "battery and bulb" circuits, or to explore series and parallel circuits.

Dissolved Oxygen Probe

Use the Dissolved Oxygen Probe to determine the concentration of oxygen in aqueous solutions in the range of 0–14 mg/L (ppm). It has built-in temperature compensation and a fast response time. This probe is great for water quality, biology, or ecology. Included with the probe is a zero-oxygen solution, two membrane caps, a 100% calibration bottle, and electrode filling solution. Replacement membranes are available (order code MEM).

Gas Pressure Sensor

The Gas Pressure Sensor can be used for a variety of experiments in biology where gases, such as oxygen and carbon dioxide, are either produced or consumed in a reaction. The pressure range is 0 to 2.1 atm (0 to 210 kPa). It comes with a variety of pressure-sensor accessories, including a syringe, plastic tubing with two Luer-lock connectors, two rubber stoppers with Luer-lock adapters, and one two-way valve.

Light Sensor

Our Light Sensor approximates the human eye in spectral response and can be used over three different illumination ranges, selected with a switch. It can be used for inverse square law experiments or for studying solar energy.

O_2 Gas Sensor

The O_2 Gas Sensor measures oxygen concentration in air. Many of the experiments currently performed using the CO_2 Gas Sensor can be performed or complemented using the O_2 Gas Sensor. Due to its wide measurement range (0–27% oxygen by volume), it can also be used to monitor oxygen concentration during human respiration.

Tris-Compatible Flat pH Sensor

The Flat pH Sensor use a double-junction electrode, making it compatible with Tris buffers and solutions containing proteins. The flat shape also makes it ideal for measuring the pH of semisolids such as food or soil. Range: 0 to 14 pH units

Soil Moisture Sensor

The Soil Moisture Sensor uses capacitance to measure the percent volumetric water content of soil. Use it to measure the loss of soil moisture over time due to evaporation and plant uptake or evaluate optimum soil moisture contents for various species of plants.

Stainless Steel Temperature Probe

The Stainless Steel Temperature Probe is an accurate, durable, and inexpensive sensor for measuring temperature. Range: –40°C to +135°C

Appendix
D

Equipment and Supplies

A list of equipment and supplies for all the experiments is given below. The amounts listed are for a class of up to 30 students working in groups of two, three, or four students in a classroom equipped with eight computers. The materials have been divided into **nonconsumables**, **consumables**, and **chemicals**. Most consumables and chemicals will need to be replaced each year. Most nonconsumable materials may be used many years without replacement. Some substitutions can be made.

Nonconsumables

Item	Amount	Experiment
alligator clips	16	22
balance	2	5, 16, 24
basting bulb	8	5
beaker, 100 mL	16	14a-c
beaker, 250 mL	16	1, 2, 5, 8, 9, 17b, 20, 28
beaker, 400 mL	8	3, 17a
beaker, 50 mL	16	2
beaker, 600 mL	24	5, 6, 7, 15, 17b, 18a-b
BioChamber 250	8	12c, 14c
bottle, BOD, 300 mL (available from Hach Company)	8	20
bottle, Nalgene, 250 mL	8	6, 12a-c, 14a-c, 17a,
bottle, sampling, 500 mL	8	27
bottle, spray	2	13
bowl	8	10
bulb, incandescent (100 W)	8	15
bulb, incandescent, clear (150 W)	8	7
can, small, metal	8	16, 24
clamp, utility	16	3, 4, 5, 7, 13, 16, 19, 26
clamp, dialysis tubing	16	3, 5
clamp, plastic tubing	16	5, 13
clay	1 stick	26
CO2–O2 Tee	8	12c, 14c

container, plastic	8	11
cup, plastic	8	8
cup, small, plastic	8	27
cup, large, Styrofoam	8	5
fan	2	13
filter, blue	8	25
filter, green	8	25
filter, red	8	25
floodlight, 100 watt	8	13
food holder	8	16
forceps	8	12a-c, 18a-b
goggles	class set	most
graduated cylinder, 10 mL	8	17a-b, 18a-b
graduated cylinder, 50 mL	8	2
graduated cylinder, 100 mL	8	8, 9, 16, 24
heater, small electric	2	13
hole punch (single)	1	26
lab apron	class set	2
lamp, desk	8	7, 10, 12a-c, 15
meter stick	8	29
motor, DC, 1.5 V	8	26
milk jug (1 gallon)	8	10
paper, black	8 pieces	7
paper, various colors	16 pieces	7
paper, white	8 pieces	7
pencil	8	5, 22
photovoltaic cell	8	25
plastic tubing clamps	8	26
plastic tubing w/Luer-lock fitting	16	5, 17b, 18b
power supply, low-voltage, DC, 6 V range, 3 amp rating (current controlled)	8	23

propeller shaft adaptor	8	26
razor blade, knife or scalpel	8	13, 22
resistor, 1 Ω	8	26
resistor, 100 Ω	8	25
ring stand	8	3, 4, 5, 7, 13, 16, 19, 24
ring, 10 cm	8	16, 24
rubber stopper assembly	16	5, 17b, 18b
ruler, metric	8	7, 10, 13, 15, 25, 26
scissors	2	3, 26
spoon, plastic	8	8, 9
stirring rod, glass	16	3, 16, 24
stopper, rubber, single-hole	16	16, 24
stopper with plastic connector	8	5
stopper, rubber, split, single-hole	8	16
stopwatch	8	18a-b
string	32 meters	5, 29
switch, single-pole, double-throw	8	23
syringe, plastic	8	5, 13
test tube rack	16	3, 17a-b, 18a-b, 28
test tube, 10 × 100 mm	32	6
test tube, 18 × 150 mm	40	3, 17a-b, 18a-b
test tube, 25 × 150 mm screw top	64	14
thermometer	8	6, 17a-b, 18a-b, 28a-c
tissue culture flask, 500 mL	8	12a-c
trowel, flat-bladed	8	11
wash bottle	8	2, 8
water bath	1	18a-b
wire leads	24	25, 26

Consumables

Item	Amount	Experiment
Alka Seltzer®	5 tablets	2
aluminum foil	1 roll	7, 12a-c, 20, 28
antacid tablets	20 g	2
aspirin	20 g	2
bag, plastic (bread or produce)	16	19
bag, plastic (gallon)	4	13
beral pipets	100	3, 13
beral pipets, 1 mL graduated	200	6, 17a-b, 18a-b
Bufferin®	20 g	2
candle	8	24
centrifuge tube, small	30	5
dental floss	1 roll	3
dialysis tubing, 2.5 cm × 12 cm	1/2 roll	3, 5
distilled water	30 L	2, 3, 4, 8, 9, 28
egg white	20 g	2
fruit juice	200 mL	2
gelatin	200 mL	2
graph paper	32 pcs	13
ice	5 bags	1, 10, 14a-c, 17a-b
Lactaid FastAct Caplet	1 box	18a-b
lamp oil	100 mL	24
leaves, fresh picked	25	12a-c
lemon	8	22
liver	20 g	2, 17a-b
Maple syrup	500 mL	5
marble (rock)	20 g	2
marshmallows	8	16
matches	8 boxes	16, 24
nail, small, iron	8	22

nuts, various	8	16
paper towels	roll	8, 9, 22
Parafilm, 5 x 5 cm	64	28
peas (garden)	400	14a-c
pipet, disposable	8	20
plant cuttings	8	13
plant, aquatic (elodea or anacharis)	16	28
plastic wrap	roll	15
pond water	7 L	4, 28
popcorn, popped	8	16
potato, whole	9	2
quartz (rock)	20 g	2
snails, aquatic	16	28
soda water	200 mL	2
soil samples (see Teacher section for specifics)	varies	8, 9, 10, 11, 15
spinach, fresh	2 bunches	12a-c
starch	20 g	2
straw, plastic	16	7, 26
tape, masking	1 roll	7, 10, 13, 15
Tes-Tape or other glucose test paper	1 pkg	18a-b
towels, paper	30	5
vitamin B	20 g	2
vitamin C	20 g	2
wooden splint	16	16
yeast	6 pkgs	2, 6, 17a-b, 18a-b

Chemicals

Item	Amount	Experiment
aluminum chloride	10 g	4
buffer solution, pH 4	500 mL	2, 17a-b
buffer solution, pH 7	1 liter	2, 8, 17a-b
buffer solution, pH 10	500 mL	2, 17a-b
calcium chloride	10 g	4
ethanol	500 mL	4, 24
fructose	5 g	6
galactose	5 g	18a-b
glucose	25 g	4, 6, 18a-b
hydrochloric acid (0.1 M)	100 mL	2
hydrogen peroxide (3%)	750 mL	17a-b
lactose	10 g	6, 18a-b
magnesium ribbon	1/4 roll	22
sodium chloride (table salt)	350 g	2, 3, 4
sodium hydroxide (0.1 M)	100 mL	2
sucrose	600 g	3, 4, 5, 6
zinc, sheet	1/4 sheet	22

Suppliers

Carolina Biological Supply Co.
1-800-334-5551
www.carolina.com

A. Daigger & Company
1-800-621-7193
www.daigger.com

Flinn Scientific Inc.
1-800-452-1261
www.flinnsci.com

Ward's Natural Science
1-800-962-2660
www.wardsci.com

Hach Company
1-800-227-4224
www.hach.com

Appendix

E

Mapping Your Data Using GPS and GIS

In agricultural science field work, knowing the exact locations of your sampling sites can be very important. Simply seeing the locations displayed on a map can add a new dimension to data analysis. Their relationships to each other and their surroundings can sometimes help explain results, or can generate even more questions. Either way, adding this geospatial component to the inquiry process can be beneficial.

There are two components involved in mapping your data: GPS and GIS. A GPS receiver is used to identify and save your location and GIS software is used to map it.

Obtaining GPS (Global Positioning System) Data

The Vernier GPS Sensor is a simple and inexpensive way to collect location information. This USB dongle connects directly to the Vernier LabQuest or your laptop and reports latitude, longitude, and altitude just as if it was another sensor. Many models of Garmin GPS units are also compatible with LabQuest and Logger *Pro*. Check the Vernier web site, www.vernier.com, for the latest list of compatible units.

Vernier GPS Sensor

Bringing GPS Data into Logger *Pro*

To map your data, you will want it in Logger *Pro* computer software. If you collected GPS data on LabQuest in the field, simply connect LabQuest to a computer running Logger *Pro* and you will be prompted to import the data. If the data have been saved on LabQuest, connect the LabQuest to a computer running Logger *Pro*. In Logger *Pro*, choose LabQuest Browser from the File menu and locate the file.

If you want to use a supported Garmin GPS unit in the field, save the locations as waypoints. When back inside, connect the GPS to the computer running Logger *Pro* and you will be prompted to retrieve the waypoints. Figure 1 shows three sample waypoints.

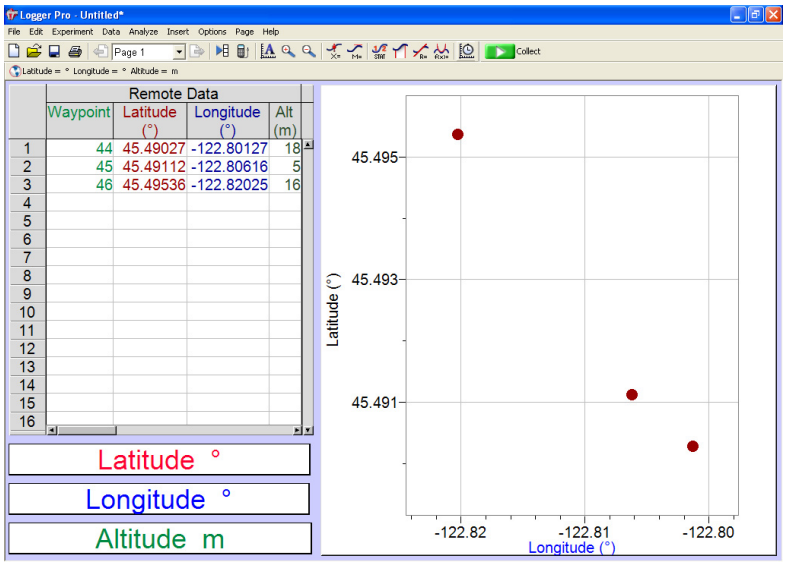


Figure 1: GPS waypoints in Logger Pro

If you are taking a laptop computer into the field, either the Vernier GPS Sensor or a supported Garmin unit can be connected directly to the computer and Logger *Pro* will treat it like a sensor, collecting live latitude, longitude, and altitude data. The Events with Entry data-collection mode is ideal for this purpose as it allows you to enter the site name as you go.

Mapping your Data Using Google™ Maps

Once your location data is in Logger *Pro*, you will probably want to view it on a map. If your computer is connected to the internet, Google Maps is one easy way to display your data. To do this:

1. Open the Logger *Pro* file that contains the geographic data.

2. Choose Export As → Google Map… from the File menu. The dialog shown in Figure 2 will be displayed, allowing you to change markers and annotations that will appear on the map.

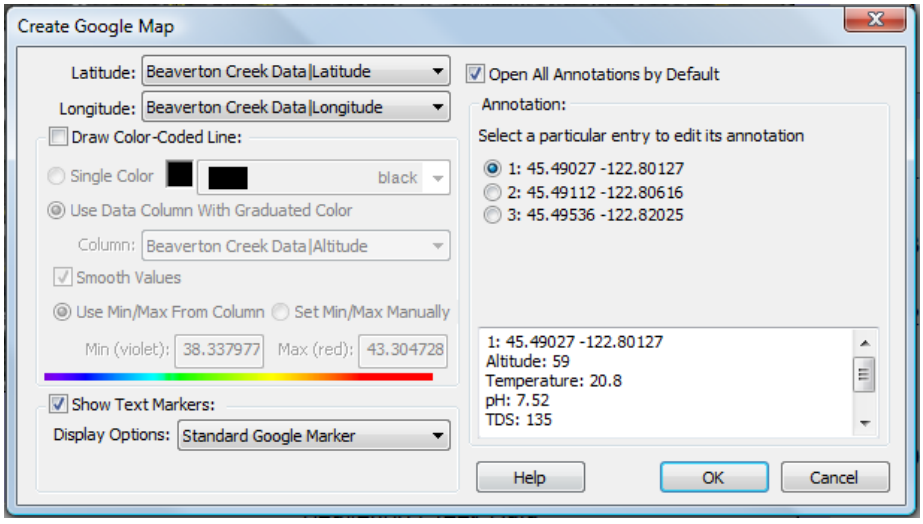


Figure 2: Logger Pro *Create Google Map dialog*

3. Click OK. Your default internet browser will be launched and your data will automatically be plotted on the Google map, as shown in Figure 3.

Figure 3: Data points marked in Google map.

4. The resulting map can then be inserted into Logger *Pro* as a picture as shown in Figure 4.
   a. Capture and save an image of the map with a tool such as Grab© for Macintosh or SnagIt© for Windows.
   b. In Logger *Pro*, choose Picture→ Picture Only… from the Insert menu.
   c. Navigate to and select your map file.
   d. Arrange your page objects manually or choose Auto Arrange from the Page menu.

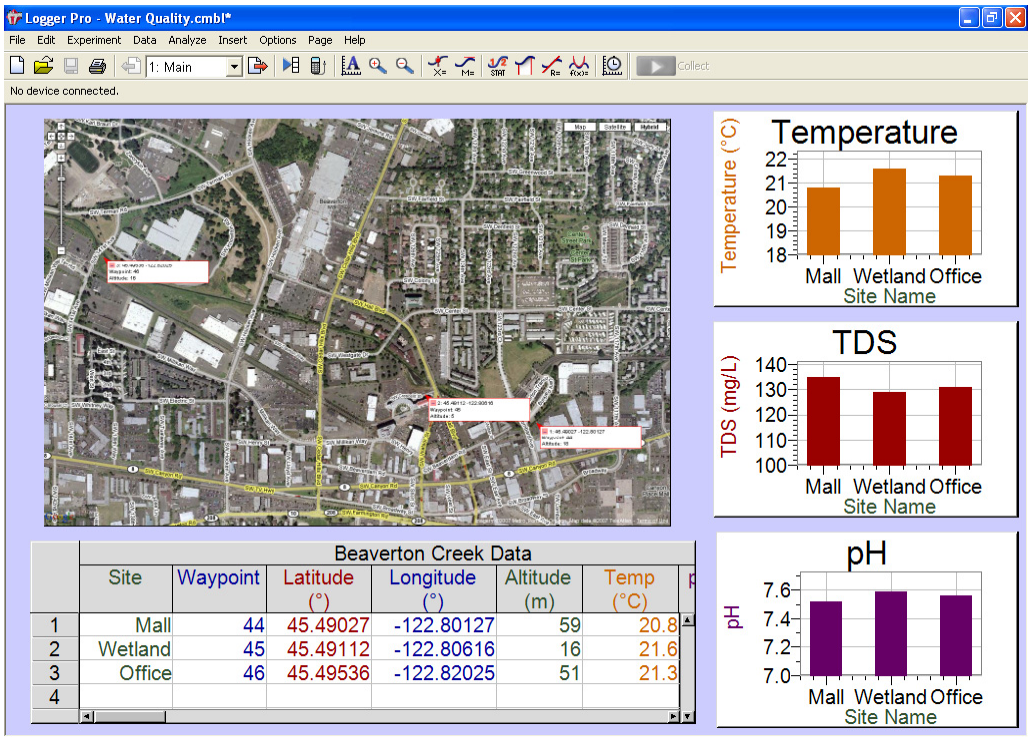


Figure 4: Logger Pro *with inserted Google map image.*

GIS (Geographic Information System)

Geographic Information System, GIS, is the term used to describe a system which displays, manages, and analyzes data which is spatially referenced to the Earth. GIS software is used for everything from scientific investigation, to urban planning, to marketing. It is particularly important for agricultural studies because it can show the relationships between the collected data and its environment. The data in Figure 1, for example, doesn't show us much on the Logger *Pro* graph. Once mapped as in Figure 3, however, we can see that this is a fairly urban location with few trees and lots of roads and parking lots. How might these factors influence the temperature, TDS, and pH values measured at each site? GIS can help answer these questions.

There are many different GIS software programs available, ranging from free tools such as ArcGIS Explorer and Google Earth, to ArcView, the gold standard of GIS software. The most common choices for educators include:

ArcGIS Explorer

ArcGIS Explorer is a free virtual globe from ESRI, the world leader in GIS software. You can use it to add your own data, plus you can add photos, reports, videos, and other information.

ArcGIS Explorer can be downloaded at www.esri.com/arcgisexplorer

ArcView

ArcView is full-featured GIS software for visualizing, analyzing, creating, and managing data with a geographic component. Once you know where your sampling sites are located, ArcView allows you to visualize, explore, and analyze this data, revealing patterns, relationships, and trends that are not readily apparent in graphs and tables.

ArcView is part of the ArcGIS Desktop collection of GIS software products from ESRI. Educational pricing is available. For information on purchasing ArcView, visit an ESRI reseller such as GISetc: Educational Technology Consultants. GISetc specializes in GIS for education and can be found on the web at www.gisetc.com.

Appendix

F

Using TI Connect: Loading EasyData and Capturing Calculator Screen Images

This appendix gives an overview of loading and updating the EasyData application on your TI-83 Plus or TI-84 Plus graphing calculators. It also describes how to download calculator screen images, such as graphs, to a computer.

EasyData is part of the bundle of APPS that come preloaded on all new TI-83 Plus, TI-84 Plus and TI-84 Plus Silver Edition graphing calculators. To do the activities in this book, you will need to use version 2.0 or newer. To check to see if you have the correct version of EasyData on your calculator, press APPS and scroll through the list of applications. With EasyData highlighted, press ENTER to launch the App and note the version number on the introductory screen. If your graphing calculator does not contain an appropriate version of EasyData, you can download the latest version for free from the Vernier web site www.vernier.com/easy and use TI Connect to transfer it to your graphing calculator.

Before loading EasyData onto your calculator, make sure you have a recent version of the calculator operating system. If you are using a TI-84 Plus calculator, you will need operating system version 2.30 or newer. If you are using a TI-83 Plus calculator, you will need operating system version 1.18 or newer. Operating system updates can be downloaded from the Texas Instruments web site at education.ti.com.

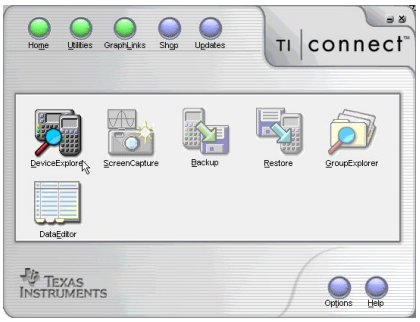
Loading EasyData

TI Connect for Windows and Macintosh is simple and easy to use. First, be sure you have the TI Connect software installed on your computer. If you do not, you can download this software for free from the Texas Instruments web site at www.education.ti.com/ticonnect. When you have downloaded the EasyData App onto your computer, follow the instructions below to transfer the EasyData app to your graphing calculator.

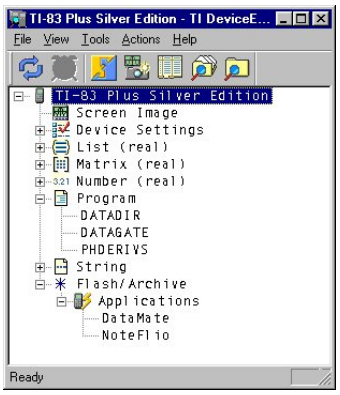
Windows Computers running Windows 7, Vista, 98, 2000, or XP

1. Connect the TI Connectivity cable or the USB direct cable to the serial or USB port of your computer and to the port on your graphing calculator.

2. Start the TI Connect software on your computer. Within TI Connect, click on Device Explorer. You may be asked to identify which computer port your cable is plugged into.



3. The software will identify the attached calculator and call up a window showing the calculator's contents.



4. To load EasyData onto a TI graphing calculator, simply drag the EasyData file from wherever you have it saved on your computer to the Device Explorer window, and it will copy onto your graphing calculator.

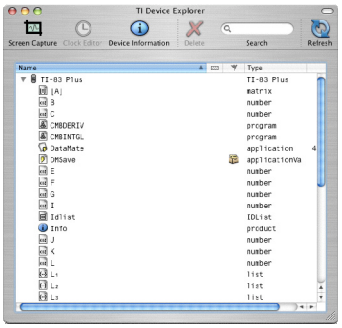
5. The EasyData App should now be loaded into your calculator. To confirm this, press APPS on the calculator to display the loaded applications.

Macintosh Computers running Mac OS X 10.4, 10.5, and 10.6.

1. Connect the TI Connectivity cable or the USB direct cable to the serial or USB port of your computer and to the port on your graphing calculator.

2. Turn the calculator on. On the computer, start TI Connect and double-click on TI Device Explorer.

3. The program will identify the attached calculator and call up a window displaying the calculator's contents.



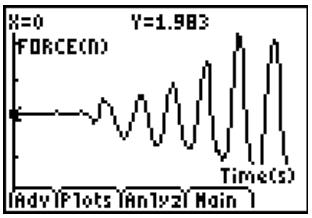
4. To load EasyData onto a TI graphing calculator, simply drag the EasyData file from wherever you have it saved on your computer to the Device Explorer window, and it will copy onto your graphing calculator.

5. The EasyData App should now be loaded into your calculator. To confirm this, press APPS on the calculator to display the loaded applications.

Capturing a Calculator Screen Image

Many experiments in this book contain an optional step that involves printing the graph displayed on the calculator screen. To capture and print a calculator screen image, you need a TI Connectivity cable or a USB direct cable and TI Connect software.

Before doing an experiment that requires printing, you may want to show your students how to print graphs. The process described below produces screen images directly from the calculator, such as seen here.



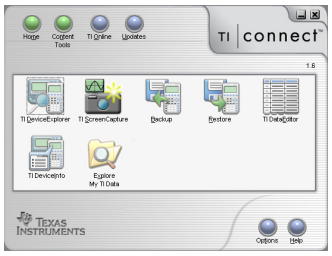
A graph of force vs. time as displayed in the EasyData application

Windows Computers running Windows 7, Vista, 98, 2000, or XP

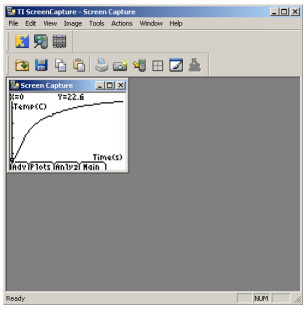
1. On your calculator, display the screen you want to capture.

2. Connect the TI Connectivity cable or the USB direct cable to the serial or USB port of your computer and to the port on your graphing calculator.

3. Start the TI Connect software on your computer. Within TI Connect, click ScreenCapture.



4. The captured screen will appear in TI Screen Capture window, and it can now be printed or saved to a file. To capture an additional screen, select Get Screen from the Tools menu.



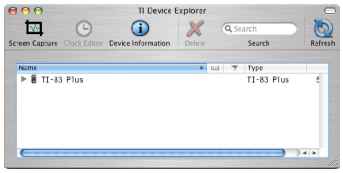
Macintosh Computers running Mac OS X 10.4, 10.5, and 10.6.

1. On your calculator, display the screen you want to capture.

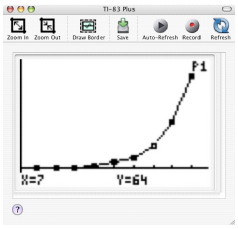
2. Connect the TI Connectivity cable or the USB direct cable to the serial or USB port of your computer and to the port on your graphing calculator.

3. Start the TI Connect software on your computer. Within TI Connect, double-click TI Device Explorer.

4. The TI Device Explorer window will appear displaying the contents of the calculator's memory.



5. Click the "Screen Capture" toolbar control. The calculator's screen image will appear in a separate window. The captured screen can now be printed or saved to a file.



Appendix
G

Safety Information

Chemical Hazard Information

The reference source for the chemical hazard information in this book is the 2002 edition of Flinn Scientific's *Chemical & Biological Catalog Reference Manual*. Flinn Scientific, Inc. is an acknowledged leader in the areas of chemical supply, apparatus and laboratory equipment supply, and chemical safety. Flinn's *Chemical & Biological Catalog Reference Manual* is an outstanding reference to be used as you order chemicals, store chemicals, mix solutions, use chemicals in you classroom, and dispose of chemicals. Most of the chemicals and the equipment used in *Agricultural Science with Vernier* are available from this catalog. We strongly urge you to obtain and use a current copy of the above mentioned publication by contacting Flinn Scientific at the address below:

Flinn Scientific, Inc.
P.O. Box 219
Batavia, Illinois 60510
Telephone (800) 452-1261
www.flinnsci.com

The Flinn hazard code is used in the teacher information section of many tests in *Biology with Computers* to describe any possible hazards associated with the chemical reagents used. The Flinn hazard code (A–D) is defined as follows:

A. Extremely Hazardous. This category includes, but is not limited to, concentrated acids, severely toxic, severely corrosive, unstable and /or explosive chemicals.

B. Hazardous. This category includes, but is not limited to, chemicals that are toxic/poisons, corrosive, contain heavy metals, and/or are alleged/proven carcinogens.

C. Somewhat Hazardous. This category includes, but is not limited to, chemicals that are highly flammable/combustible, moderately toxic and/or oxidants.

D. Relatively Non-Hazardous. This category includes, but is not limited to, chemicals that are irritants and/or allergens.

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(by Experiment Number)