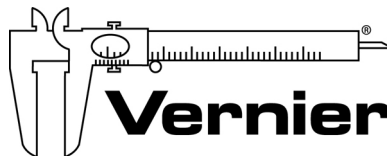


Human Physiology with Vernier

Diana Gordon
Steven L. Gordon, M.D.



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Human Physiology with Vernier

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About the Authors

Diana Gordon graduated from Linfield College with a B.A. in biology, and received her MAT from Lewis and Clark College. While teaching biology at St. Mary's Academy in Portland, Diana received NABT's Outstanding Biology Teacher of the Year award for the state of Oregon. In 2001, she joined the Tech Support team at Vernier as a biology specialist. During her years at Vernier she conducted many workshops throughout the country, training teachers in the use of Vernier technology. She is currently a Public Information Officer for the Oregon Health and Science University. She and her husband, Dr. Steve Gordon, share an interest in and commitment to the improvement of science education and are popular lecturers on the topics of science, pseudoscience, and medical quackery.

Steve Gordon, M.D. received his B.A. in zoology from the University of California at Berkeley, and his M.D. from the University of California, Los Angeles Medical School. After graduating from medical school, he and Diana moved to Oregon, where he completed an internal medicine residency at Oregon Health and Science University. Dr. Gordon is board certified in internal medicine and has maintained a full-time general internal medicine practice with Northwest Permanente for over 20 years. He continues to train second-year medical students in a clinical setting, helping them to translate basic knowledge of physiology into clinical relevance. Along with his wife, Diana, he maintains an active interest in science education.

Proper safety precautions must be taken to protect teachers and students during experiments described herein. Neither the authors nor the publisher assumes responsibility or liability for the use of material described in this publication. It cannot be assumed that all safety warnings and precautions are included.

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Sensors Used in Experiments

		Surface Temperature	Heart Rate	Blood Pressure	EKG Sensor	Hand Dynamometer	Spirometer	Oxygen Gas Sensor	25-g Accelerometer
1	Warming Function of Nasal Passageways	X							
2	Effect of Vascularity on Skin Temperature Recovery	X							
3	Heart Rate as a Vital Sign		X						
4	Heart Rate and Exercise		X						
5	Heart Rate Response to Baroreceptor Feedback		X						
6	Effect of Coughing on Heart Rate		X						
7	Blood Pressure as a Vital Sign			X					
8	Blood Pressure and Exercise			X					
9	Diurnal Blood Pressure Variation			X					
10	Heart Rate and Blood Pressure as Vital Signs		X	X					
11	Heart Rate, Blood Pressure, and Exercise		X	X					
12	Analyzing the Heart with EKG				X				
13	Introduction to EMG				X				
14A	Neuromuscular Reflexes (with Accelerometer)				X				X
14B	Neuromuscular Reflexes (without Accelerometer)				X				
15	Muscle Function Analysis				X				
16	Grip Strength Comparison					X			
17	Grip Strength and Muscle Fatigue					X			
18	EMG and Muscle Fatigue				X	X			
19	Lung Volumes and Capacities						X		
20	Respiratory Response to Physiologic Challenges						X		
21	Analysis of Lung Function						X		
22	Oxygen and Aerobic Metabolism						X	X	
23	Oxygen Extraction by the Lungs							X	
24	Effect of "Dead Space" on Oxygen Exchange							X	

Preface

This book contains 24 human physiology experiments using the Vernier LabQuest and Vernier LabPro. The experiments employ eight sensors, including the Vernier EKG Sensor, Hand-Grip Heart Rate Monitor, Hand Dynamometer, Blood Pressure Sensor, Spirometer, Surface Temperature Sensor, Oxygen Gas Sensor, and 25-g Accelerometer.

You will find that the experiments in this book will appeal to students at many levels, including high school, advanced placement, college, and even first-year medical school. The experiments are designed to encourage students to think about the physiology of the various human organ systems by measuring the activity of the upper respiratory tract, the lungs, the heart, the nervous system, and the musculoskeletal system. Homeostasis is an underlying theme, and several experiments illustrate the response of the human body to stress or external influences. Others include activities that simulate disease states to enable students to compare normal and abnormal functioning of a particular system.

Where possible, we have pointed out the medical relevance of each experiment with examples and problems drawn from clinical experience and from the perspective of medical personnel. We believe that this will make the experiments more interesting for students. In many experiments, students are asked to compare results or collect results from the class. This approach reveals the variability that is inherent in the study of different individuals; the variability that makes the study of human physiology and the practice of medicine so challenging. The experiments in this book are written with directions that correspond to computer-based data collection, using Vernier Logger *Pro*[®] software on a Windows or Macintosh computer and to data collection using Vernier LabQuest and LabQuest App.

The printed pages of this book contain the computer instructions. LabQuest instructions can be found on the accompanying CD. The files for the student pages for each experiment included on the CD in the back-inside cover of this book are Microsoft Word[®] files. See *Appendix A* for details about using this CD. The Word files on the CD are identical to the printed versions of the student experiments in this book.

In all cases, purchase of this book entitles you to use these word-processing files in any way you wish in your physiology course. This includes making edits to our files to match your own teaching style or lab-investigation pedagogy. We also give you permission to include these files or edited versions in your course lab manual, as long as this manual is not distributed beyond your school boundaries.

Instructors will find extensive support information in this book:

- Following each student experiment, there is an extensive Teacher Information section with sample results, sample graphs, answers to questions, directions for preparing solutions, and other helpful hints regarding the planning and implementation of a particular experiment.
- *Appendix B* provides information on Vernier products for human physiology.
- *Appendix C* documents all equipment and supplies for doing all experiments in this book, divided into nonconsumables and consumables.
- *Appendix D* provides safety information associated with the experiments in this book.

Which experiments should you use? This depends, of course, on the scope and sequence of your human physiology course. We do not expect that every experiment in your physiology course will require a Vernier sensor to be used. As you can see from the Table of Contents of this book, however, Vernier sensors can become an important laboratory tool for your students.

We would like to thank Gretchen Stahmer DeMoss for directing this project. Her insights and contributions have been invaluable and greatly appreciated. We are grateful to Kelly Redding for her outstanding illustrations, which help to clarify many of the physiologic principles discussed in the experiments. Thanks also to Alex Plank, who provided excellent editorial support, and to John Wheeler, Henry Le, and Peter Paul Lalic, who developed several new sensors to support this venture. Finally, we would like to thank Dan Holmquist, who gave us the opportunity to help launch Vernier's entrance into the area of human physiology.

Warming Function of Nasal Passageways

Nasal passageways transport air to and from the lungs and also serve to warm and humidify this air. The nasal turbinates provide a large surface area and rich blood supply for a process of heat exchange which contributes to the maintenance of a constant body temperature (see Figure 1). Warming of inhaled air improves lung function and removal of heat from the exhaled air helps to prevent excessive heat loss from the body. Conditions such as viral infections, allergies, and deviated septae can all lead to plugging of nasal passageways and mouth breathing. Inhaling colder air through the mouth can irritate the lungs and worsen conditions such as bronchitis and asthma.

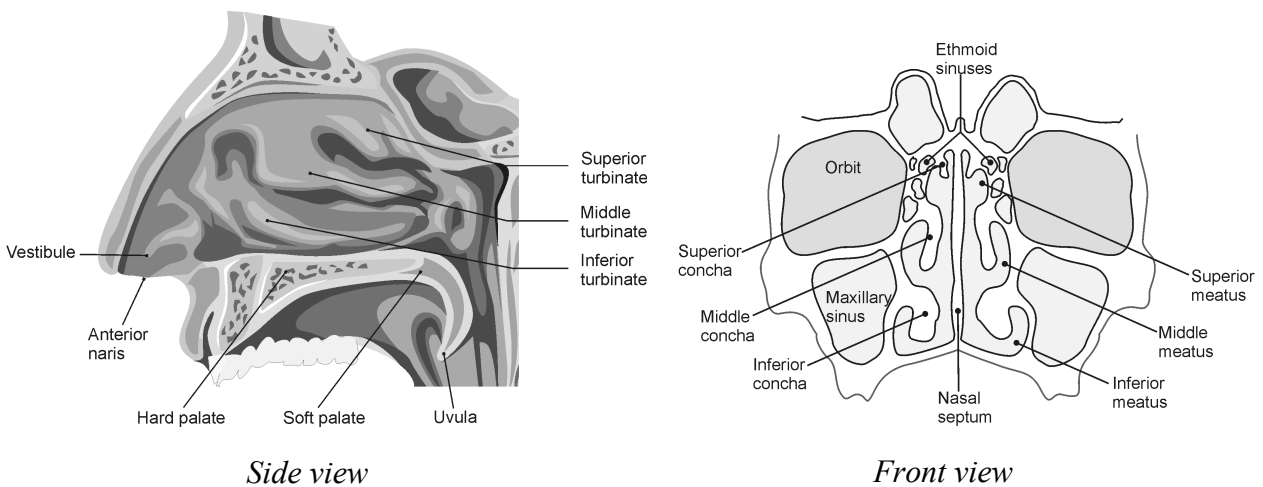


Figure 1

In this experiment, you will use the Vernier Surface Temperature Sensor to measure the temperature of exhaled air. Air that has passed through the nasal passageways will be compared with air that was inhaled through the mouth. In both cases, the air spends time in the lungs where heat is acquired from exposure to the extensive lung tissue alveolar surface area (see Figure 2). However, air that has passed through the nasal passageways arrives at the lungs already partially warmed.

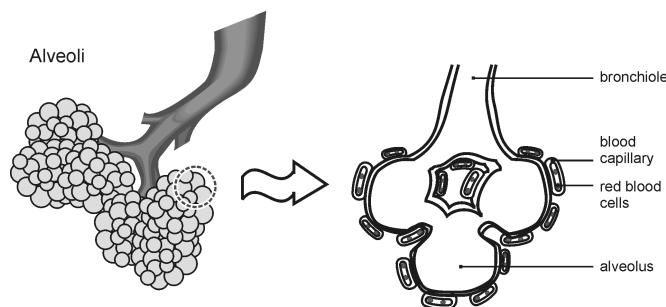


Figure 2

OBJECTIVES

In this experiment, you will

- Compare the temperature of air that has passed through the nasal passageways with air that has not.
- Evaluate the contribution of nasal passages vs. lungs to the warming of the air we breathe.

MATERIALS

computer
Vernier computer interface
Logger *Pro*

Vernier Surface Temperature Sensor
plastic straws
tape

PROCEDURE

Comparison of Temperature of Air Inhaled Through Nose and Air Inhaled Through Mouth.

1. Connect the Surface Temperature Sensor to the Vernier computer interface. Open the file “01 Warming Function” from the *Human Physiology with Vernier* folder.
2. Insert the end of the Surface Temperature Sensor in one end of a plastic straw such that the tip of the sensor is approximately 2 cm into the straw. Fold the wire back over the straw and affix it to the outside of the straw with tape (see Figure 3).
3. Click to begin data collection. Record a baseline temperature for approximately 5 s or until a stable baseline temperature is reached.
4. After the baseline temperature has stabilized, begin taking normal breaths through the nose and exhaling by blowing your breath out the mouth and through the straw (approximately 2–3 s per inhalation and 2–3 s per exhalation). Remove the straw from the mouth after each exhalation. Continue this process until data collection is complete. Data will be collected for 40 s.
5. Store this run by choosing Store Latest Run from the Experiment menu.
6. Click to establish a temperature baseline. If the temperature is higher than the baseline temperature of the first run, allow the baseline temperature to fall to a level close to what it was at the start of the first run. You may speed this process by inhaling and/or blowing through the straw. Once your baseline temperature has been reached, click .
7. Click to begin data collection. Record a baseline temperature for about 5 s.
8. After the baseline temperature has stabilized, begin taking normal breaths through the mouth and exhaling through the straw (approximately 2–3 s per inhalation and 2–3 s per exhalation). Use a nose clip or pinch the nose closed to prevent accidental nose breathing. Remove the straw from the mouth during each inhalation. Continue this process until data collection is complete. Data will be collected for 40 s.
9. Click the Statistics button, . Check the boxes in front of Latest and Run 1, then click . Record the maximum and minimum temperatures to the nearest 0.1°C for each run in Table 1.

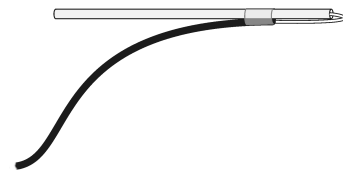


Figure 3

10. Subtract minimum from maximum values to obtain the temperature change and record these values as Δt . Then, subtract the Δt values for mouth inhalation (Latest) from nose inhalation (Run 1) and record this value in the column labeled $\Delta t_{\text{Run 1}} - \Delta t_{\text{Latest}}$.

DATA

Table 1			
	Run 1 (nose inhalation)	Latest (mouth inhalation)	$\Delta t_{\text{Run 1}} - \Delta t_{\text{Latest}}$
Maximum temperature (°C)			
Minimum temperature (°C)			
Δt (°C)			

DATA ANALYSIS

1. Reviewing your data, what conclusions can you reach regarding where inhaled air receives most of its heat?

2. What proportion of the difference in temperature between inhaled and exhaled air can be attributed to the nasal passageways when nose breathing? Assume that the warming contribution of the mouth is negligible. (For the denominator, use the Δt value for Run 1 in Table 1).

3. The average surface area of the nasal passageways is 160 cm² and the average surface area of the alveoli in the lungs is approximately 100 m² (1 × 10⁶ cm²). Use the data you collected to calculate the amount of heat contribution per unit area of
 - (a) nasal epithelium surface area to which the air is exposed.

 - (b) alveolar surface area to which the air is exposed.

In both cases, ignore confounding variables such as the volume of air that stays in the bronchi (also called *dead space*) and the volume of air that does not leave the lungs, also called *functional residual capacity*.

Computer 1

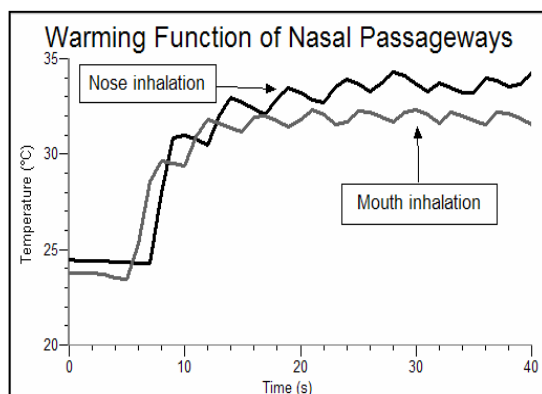
4. Describe a situation in which it would be beneficial to breathe through the mouth.

TEACHER INFORMATION

Warming Function of Nasal Passageways

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Lab bench surfaces should be well cleaned or covered with clean disposable covers to prevent straws from contacting harmful chemicals or residues.
3. It is not necessary to use more than a single loop of tape to connect the sensor to the straw; this method will facilitate moving the sensor from straw to straw as different subjects perform the experiment.

SAMPLE DATA



Sample data

	Run 1 (nose inhalation)	Latest (mouth inhalation)	$\Delta t_{\text{Run 1}} - \Delta t_{\text{Latest}}$
Maximum temperature (°C)	34.4	32.3	
Minimum temperature (°C)	24.5	23.5	
Δt (°C)	9.9	8.8	1.1

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Most of the heat is acquired from the lungs.
2. The difference between exhaled temperatures from nose and mouth breathing is divided by the temperature of exhaled air from nose breathing to obtain a value in the range of 10–15%.

Experiment 1

3. (a) Air warmed in the nasal passageways can be found using the following equation.

$$\frac{\Delta t_{Run1} - \Delta t}{160 \text{ cm}^2}$$

Answers should be approximately $7 \times 10^{-3} \text{ }^\circ\text{C}/\text{cm}^2$.

- (b) Air warmed in the lungs can be found using the following equation.

$$\frac{\Delta t_{Latest} - \Delta t}{1 \times 10^6 \text{ cm}^2}$$

Answers should be approximately $1 \times 10^{-5} \text{ }^\circ\text{C}/\text{cm}^2$.

4. Students may describe any situation in which the body attempts to rid itself of excess heat, such as exercising or working in a hot environment.

Effect of Vascularity on Skin Temperature Recovery

Homeostasis refers to the body's ability to maintain internal conditions (e.g., temperature, pH, hydration) within the narrow limits that are optimal for the continuation of metabolic processes. When these optimal conditions are disturbed by a change in the environment, body systems work to return them to normal.

Many of the chemical reactions and cellular processes necessary to sustain human life occur most readily at a body temperature of approximately 37.0°C (98.6°F). Homeostatic mechanisms work to maintain this temperature, regardless of changes in the external environment. Changes in temperature are sensed by the skin, which is well-designed to counteract these changes. Beneath the protective epidermal layer of the skin lies the dermis, which contains sweat and oil glands and a rich blood supply (see Figure 1).

The dermal tissues influence body temperature by either allowing or diverting the blood's access to the surface of the skin. When an area is exposed to the cold, small arterioles in the dermis constrict, allowing less than normal blood flow to that area and protecting core body temperature. When cold is removed, the blood supply to the skin increases again as the arterioles dilate. Exposure to heat causes blood vessels to dilate, allowing heat to dissipate from the skin's surface, and resulting in a flushed or red appearance. Other examples of the regulation of body temperature through the action of the skin's blood supply include the dilatation seen with embarrassment and the constriction and resulting paleness that occurs with fear. Dilatation also occurs with ingestion of alcohol.

Human skin temperature is maintained at approximately 33°C (91°F). This temperature can be referred to as the skin temperature "set point." Regions of skin with higher *vascularity* (containing more blood vessels) will return more quickly to the set point after a disturbance than regions with less vascularity. In this experiment, you will compare the rate of recovery from cold in two different skin regions and draw conclusions about the vascularity of these areas.

Important: Inform your instructor of any possible health problems that might be exacerbated if you participate in this exercise.

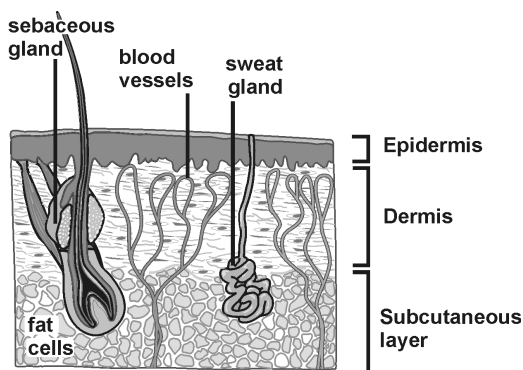


Figure 1

OBJECTIVES

In this experiment, you will

- Compare the rate of recovery from cold in two different skin regions.
- Correlate rate of recovery with vascularity.


MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier Surface Temperature Sensor

tape
ice
soap and water or rubbing alcohol
towel (paper or cloth)

PROCEDURE

Temperature Recovery by Skin of Upper Arm

1. Connect the Surface Temperature Sensor to the Vernier computer interface. Open the file “02 Skin Temperature” from the *Human Physiology with Vernier* folder.
2. Remove excess oil from the skin over the biceps with soap and water or alcohol. Tape the Surface Temperature Sensor to the upper arm, over the area of the biceps. Be sure to tape the thermistor end (the tip) of the sensor directly to the arm (see Figure 2).
3. Click to begin data collection. Collect data for 50 s to obtain a baseline recording of the temperature. Click to end data collection.
4. Choose Store Latest Run from the Experiment menu to store the data.
5. Remove the Surface Temperature Sensor from the arm. Obtain a piece of ice and hold the ice over the area of the upper arm to which the Surface Temperature Sensor was affixed. Hold the ice cube in place for 30 s.
6. Remove the ice and quickly blot the area dry with a towel. DO NOT RUB as friction can cause an increase in skin temperature.
7. Tape the Surface Temperature Sensor to the upper arm again, in the same area of the biceps where the ice was held.
8. Click to begin data collection. Data will be collected for 120 s.
9. Store this run by choosing Store Latest Run from the Experiment menu.
10. Determine the rate of recovery.
 - a. Select the data collected from 0–50 s by clicking and dragging to highlight this region.
 - b. Click on the Linear Fit button, , and check the box next to Run 2.
 - c. Click and a best fit linear regression line will be shown for the selected run. The linear regression statistics are displayed in a floating box.
 - d. In Table 1, record the value of the slope, m , for the run.
 - e. Close the linear fit box by clicking the \times in the corner of the box.

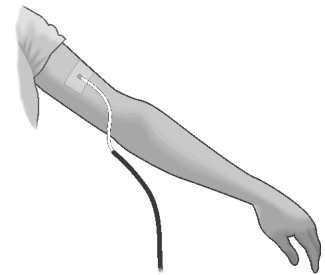


Figure 2

Temperature Recovery by Facial Skin

11. Tape the Surface Temperature Sensor to the face, below the cheek bone, approximately 3 cm from the corner of the mouth, looping the sensor wire over the ear for stability (see Figure 3). Remove excess oil from the skin with soap and water or alcohol to improve the adhesion of the tape to the skin. Be sure to tape the thermistor end (the tip) of the sensor directly to the cheek.
12. Click to begin data collection. Collect data for 50 s to obtain a baseline recording of the temperature. Click to end data collection.
13. Store this run by choosing Store Latest Run from the Experiment menu.
14. Remove the Surface Temperature Sensor from the face. Obtain a piece of ice and hold the ice over the area of the cheek to which the Surface Temperature Sensor was affixed. Avoid placing the ice cube on the cheek bone, as this may cause headache. Hold the ice cube in place for 30 s.
15. Remove the ice and quickly blot the area dry with a towel. **Important:** Do not rub as friction can cause an increase in skin temperature.
16. Tape the Surface Temperature Sensor to the face again, in the same area where the ice was touching.
17. Click to begin data collection. Data collection will continue for 120 s.
18. Determine the rate of recovery.
 - a. Select the data collected from 0–50 s by clicking and dragging to highlight this region.
 - b. Click on the Linear Fit button, , and check the box next to Run 2.
 - c. Click and a best fit linear regression line will be shown for the selected run. The linear regression statistics are displayed in a floating box.
 - d. In Table 1, record the value of the slope, m , for the run.

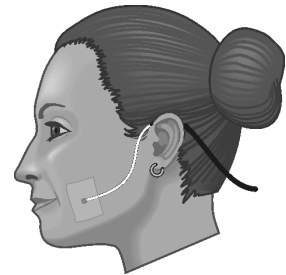


Figure 3

DATA

Table 1	
Run	Slope, or rate (°C/s)
Run 2 (upper arm)	
Latest (cheek)	

DATA ANALYSIS

1. Is the baseline temperature recovered within the 2 minutes during which data is collected? Estimate how long it would take for full recovery to be achieved for each of the two runs. Relate this to everyday experiences where you have been exposed to cold.

Computer 2

2. Which area of skin tested (upper arm or face) had the most rapid recovery of temperature after cooling? Explain this result.
3. Alcohol causes dilatation of arterioles and a sensation of warmth. Would you recommend that someone who is stranded in the snow drink alcohol to keep warm? Why or why not?
4. A condition called hyperthermia (heat prostration) can result when the body's homeostatic mechanisms are no longer adequate to counter the effect of high external temperatures. On the basis of what you know about vasoconstriction and vasodilation as methods of temperature regulation in the body, describe the skin color of someone who is in the first stages of hyperthermia.

EXTENSION

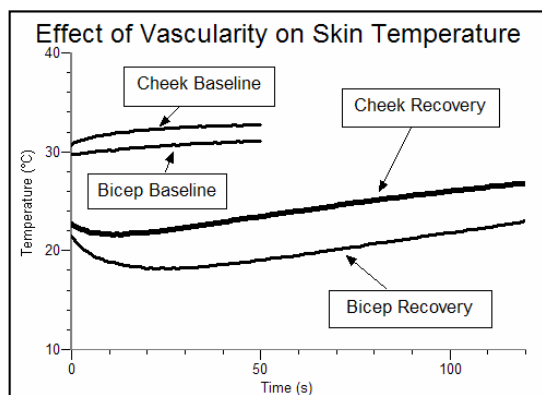
1. Perform the procedure on different areas of the body (i.e., over large muscles, such as the thigh and calf muscles; or over areas with little subcutaneous fat, such as the palm, kneecap, and shin). Compare your results with the result obtained in the original experiment.
2. Perform the procedure placing a blanket over the area on which the sensor is attached. Does this alter the recovery time?

TEACHER INFORMATION

Effect of Vascularity on Skin Temperature Recovery

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. To ensure that the tape holding the Surface Temperature Sensor stays in place, provide rubbing alcohol (or alcohol wipes) so that students may remove make-up or oils from the skin.
3. Any kind of clear tape (e.g., cellophane) will work well for this experiment and allow proper placement of the thermistor end of the Surface Temperature Sensor.

SAMPLE DATA



Sample data for skin temperature recovery

Run	Slope, or rate ($^{\circ}\text{C}/\text{s}$)
Run 2 (upper arm)	-0.02
Latest (cheek)	0.03

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Skin temperature recovers only partially during the 2 minutes of data collection. It would likely take another minute or two for full recovery to occur. Students may relate this to experiences of coming out of a cold swimming pool, lake, or other body of water.
2. The face recovered more rapidly, as demonstrated by the greater slope value in Table 1. This is due to the fact that the face possesses a richer blood supply than the skin of the upper arm. This vascularity leads to the well-known “blush response,” which occurs with embarrassment, anger, or other emotions.

Experiment 2

3. It is not a good idea to drink alcohol when you are cold. Dilatation of arterioles in the skin creates a sensation of warmth to the skin sensors, but allows greater heat loss and more rapid cooling of the core body temperature. This may hasten hypothermia.
4. In the first stages of heat stroke, the body is dilating surface blood vessels in an attempt to release heat. This results in a flushed, “lobster-like” appearance.

Heart Rate as a Vital Sign

Since the earliest days of medicine, heart rate has been recognized as a *vital sign* (an indicator of health, disease, excitement, and stress). Medical personnel use heart rate to provide clues as to the presence of many medical conditions. Reflex changes in heart rate are one of the body's most basic mechanisms for maintaining proper perfusion to the brain and other tissues. Low blood volume caused by bleeding or dehydration results in the heart beating faster as it attempts to maintain adequate blood pressure. Excitement, stress, and anxiety activate the autonomic nervous system, which may also speed the heart rate and raise blood pressure.

The autonomic nervous system consists of sympathetic and parasympathetic branches, which have opposing effects on the circulatory and other organ systems. Sympathetic activation raises blood pressure in addition to pulse. After an initial activation of the sympathetic nervous system, the increase in blood pressure stretches nerve fibers in the baroreceptors (see Figure 1). This results in a reflex activation of the parasympathetic nervous system, which, through actions opposite to those of the sympathetic nervous system, helps to restore homeostasis.

In this experiment, you will observe how the heart responds to cold stimulus applied peripherally. In this case, cold will act as a noxious stimulus, activating the “fight or flight” response through the sympathetic nervous system.

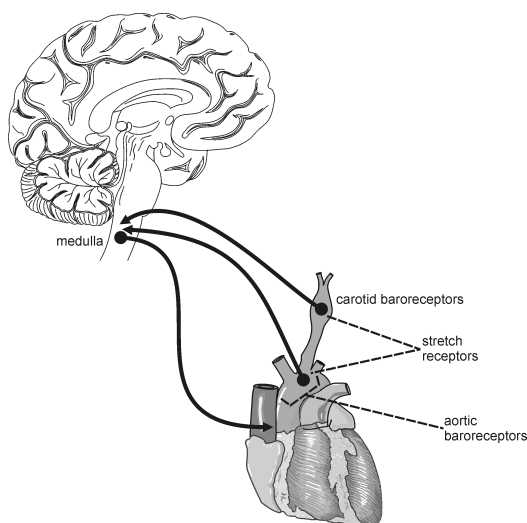


Figure 1

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of heart rate.
- Compare heart rate before and after exposure to cold stimulus.
- Observe an example of sympathetic nervous system activation (“fight or flight response”).

MATERIALS

- | | |
|--|--|
| computer | ice water bath |
| Vernier computer interface | towel |
| Logger Pro | saline solution in dropper bottle
(only for use with Exercise HR Monitor) |
| Vernier Hand-Grip Heart Rate Monitor or | |
| Vernier Exercise Heart Rate Monitor | |

PROCEDURE

Select one person from your lab group to be the subject.

1. Connect the receiver module of the Heart Rate Monitor to the Vernier computer interface.
2. Open the file “03 Heart Rate Vital Sign” from the *Human Physiology with Vernier* folder.
3. Prepare to collect data.
 - a. Sit in a chair, facing away from the computer.
 - b. Prepare to submerge your foot in the ice water bath by removing your shoe and sock.
 - c. Position your foot adjacent to the ice water bath, but do not put it in the bath yet.
4. Set up the Heart Rate Monitor. Follow the directions for your type of Heart Rate Monitor.

Using a Hand-Grip Heart Rate Monitor

- a. The receiver and one of the handles are marked with a white alignment arrow as shown in Figure 2. Locate these two arrows.
- b. Have the subject grasp the handles of the Hand-Grip Heart Rate Monitor so that their fingers are in the reference areas indicated in Figure 3. Hold the handles vertically.
- c. Have someone else hold the receiver near the handles so that the two alignment arrows are pointing in the same direction and are at approximately the same height as shown in Figure 2. **Note:** The receiver must stay within 60 cm of the handles during data collection.

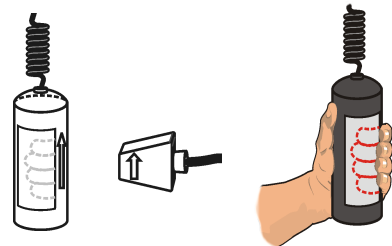


Figure 2

Figure 3

Using an Exercise Heart Rate Monitor

- a. Depending upon your size, select a small- or large-size elastic strap. Secure one of the plastic ends of the elastic strap to the transmitter belt. It is important that the strap provide a snug fit of the transmitter belt.
- b. Wet each of the electrodes (the two textured oval areas on the underside of the transmitter belt) with 3 drops of saline solution.
- c. Secure the transmitter belt against the skin directly over the base of the rib cage (see Figure 4). The POLAR logo on the front of the belt should be centered. Adjust the elastic strap to ensure a tight fit.
- d. Take the receiver module of the Heart Rate Monitor in your right hand. **Note:** The receiver must stay within 60 cm of the handles during data collection.

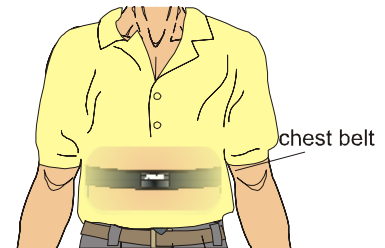


Figure 4

5. To determine that everything is set up correctly, click to begin monitoring heart rate. Note that there may be up to a 30 second delay before data are seen. The readings should be within the normal range of the individual, usually between 55 and 80 beats per minute. Click when you have determined that the equipment is operating properly, and proceed to the next step.
6. Collect data to observe the effect of submerging your foot in an ice water bath. **Note:** Read over this step prior to beginning data collection to familiarize yourself with the process.
 - a. Click .
 - b. If the baseline is not stable, repeat Steps 5. If the baseline is stable, plunge your foot into the ice water bath at 40 s.
 - c. Remove your foot from the ice water bath 30 s after immersion (when data have been collected for 70 s) and rest it on the towel.
 - d. Remain seated and allow data collection to continue for the full 240 s data-collection period.
7. Click and drag over the area of the graph where the resting (“baseline”) heart rate is displayed. Click the Statistics button, . The Statistics box will appear with the statistics calculated for the selected region. Record the mean resting heart rate, to the nearest whole number, in Table 1.
8. Move the Statistics brackets to highlight the total time of data collection. The values in the statistics box will be recalculated to reflect this change. Record the maximum and minimum heart rates, to the nearest whole numbers, and the corresponding times at which these rates are graphed, in Table 1.
9. Move the statistics brackets to highlight the region of the graph beginning at 40 s (when the foot was immersed in the ice water bath) and ending at the first peak (see Figure 5). Record the maximum heart rate value in Table 1. In the corresponding Time column record (to the nearest whole number) the Δx value displayed at the lower left corner of the graph.

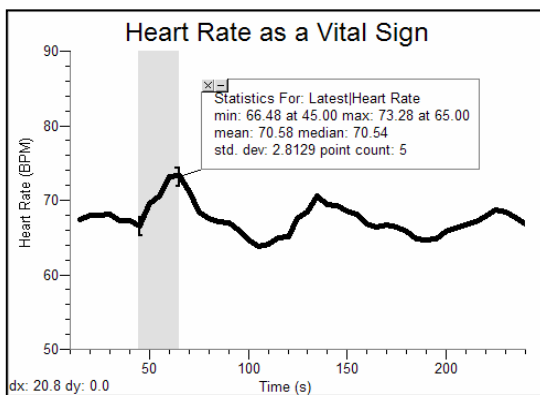


Figure 5

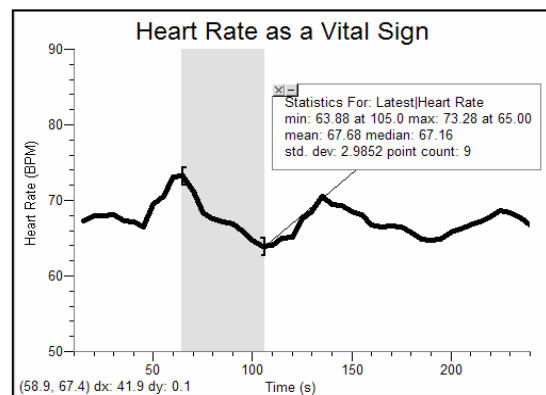


Figure 6

10. Move the Statistics brackets to enclose the region of the graph beginning at the first peak and ending at the lowest point in the valley that follows (see Figure 6). Record the minimum heart rate value as the Rebound heart rate in Table 1. Record the Δx value in the corresponding Time column.

DATA

Table 1		
Condition	Heart rate (beats/minute)	Time (s)
Resting heart rate		
Maximum heart rate		
Rebound heart rate		

DATA ANALYSIS

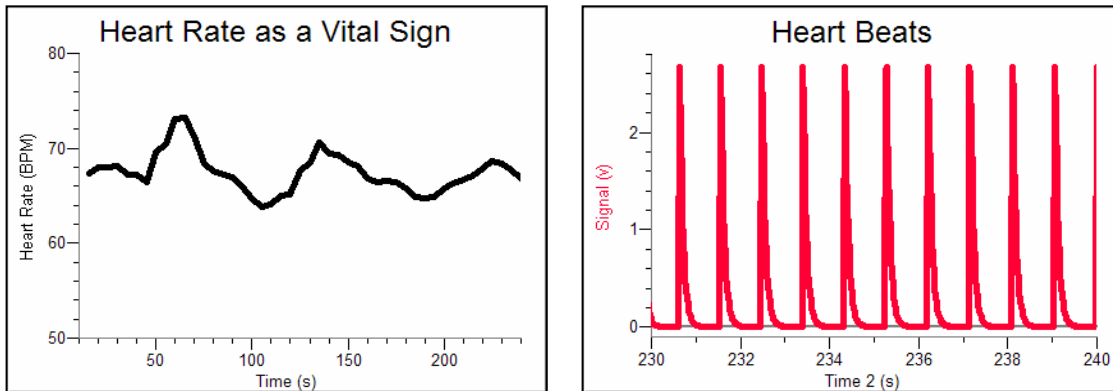
1. How long after immersion did your heart rate reach its maximum value? Explain the physiologic mechanism that led to this change in heart rate.
2. Describe the changes in heart rate that occurred after the maximum value. How can you explain the minimum heart rate value? How would you explain the heart rate variations seen in the remainder of the experiment?
3. How long after the maximum heart rate did it take to arrive at your rebound heart rate? What can you say about the relative speed of physiologic response to a stimulus *vs.* the speed of mechanisms that are designed to maintain homeostasis?
4. If the heart rate is too slow there is inadequate blood pressure to maintain perfusion to the brain. This can lead to loss of consciousness (fainting). Keeping in mind the autonomic nervous system responses that you observed in this experiment, explain the sequence of events that results in a severely frightened person fainting.

TEACHER INFORMATION

Heart Rate as a Vital Sign

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. This lab exercise can be done with either of two types of heart rate monitors: the Hand-Grip Heart Rate Monitor or the Exercise Heart Rate Monitor. The Hand-Grip Heart Rate Monitor, which consists of two hand grips and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through the hand grips and relaying the information to the plug-in receiver. The Exercise Heart Rate Monitor, which consists of a chest belt and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through a belt worn around the chest and relaying the information to the plug-in receiver.
3. When using either the Hand-Grip Heart Rate Monitor or the Exercise Heart Rate Monitor there will be a 15 s delay while data are collected before the first point is plotted on the upper graph. Thereafter, a point will be plotted every 5 s.
4. Computer monitors can be a source of electrical interference for both designs of the Heart Rate Monitors. To ensure that this is not a factor in your students' data-collection activities, move the receiver module as far away from the computer monitor as possible.
5. The receiver module of both designs of the Heart Rate Monitors will receive signals from the closest transmitter source. This may result in an apparent doubling of the subjects' heart rates. To avoid confusion or erroneous readings, have test subjects from different lab teams maintain a distance of at least 2 m from each other.
6. When using the Hand-Grip Heart Rate Monitor, it is important that students closely follow the instructions for holding the handles. The hand grips are marked for the right or left hand, each showing where the individuals' fingers and palms should be placed. In addition, the left hand grip and the receiver are both marked with alignment arrows. When collecting data, it is important that the arrow labels on each of these devices be in alignment (see figures in the student version of the experiment). Failure to follow these instructions will result in unreliable data.
7. It is important to have good contact between the transmitter belt and the test subject when using the Exercise Heart Rate Monitor. It is very important that the belt fit snugly. Both electrodes should be wetted with either saline solution or contact lens solution. A 5% salt solution works well and can be prepared by adding 5 g per 100 mL of solution. Typical symptoms of inadequate contact with the electrodes are a noisy signal with erroneous peaks, missing heart beat readings, or a flat-line display. If the students receive a flat reading with no heart rate detected, have them move the transmitter and receiver closer together. The range of the transmitter in the chest belt is 60–80 cm.

SAMPLE DATA



Sample data

Condition	Heart rate (beats/minute)	Time (s)
Resting heart rate	68	
Maximum heart rate	73	24
Rebound heart rate	64	42

ANSWERS TO THE DATA ANALYSIS QUESTIONS

- Answers will vary, but should be in the range of 30–40 s. Factors such as weight, regular exercise, health, medications, and tolerance to cold, may affect individual response. Exposure to cold causes severe enough discomfort to activate the sympathetic nervous system, which acts to increase the heart rate.
- Following the maximum value, most students will report a slowing of the heart to a rate below the resting heart rate. The initial increase in heart rate (and blood pressure) activates baroreceptors which in turn cause parasympathetic activation and a slowing of the heart rate. In this case, the slowing briefly “overshoots” the resting heart rate. Some students may even report a “see-sawing” effect, where the heart rate increases and decreases in an almost sinusoidal pattern while homeostasis is being restored.
- Answers will vary, but should be in the range of 30–50 s. The initial physiologic response to stimuli is more rapid than compensatory responses. There are obvious benefits to a rapid response to danger. A more measured return to homeostasis lessens the see-sawing effect as the sympathetic and parasympathetic systems are achieving new balance.
- When a person is severely frightened, there is an overwhelming sympathetic response. This increases heart rate and blood pressure dramatically. The compensatory parasympathetic response is likewise much greater than normal, causing marked slowing of the heart rate and a drop in blood pressure and circulation to the brain.

Heart Rate and Exercise

The adaptability of the heart can be observed during exercise, when the metabolic activity of muscle tissue increases. The cardiovascular system, consisting of the heart and blood vessels, responds to exercise with an increase in heart rate and strength of contraction with each beat, resulting in a higher *cardiac output* (quantity of blood pumped through the heart per unit of time). Physically fit people can deliver a greater volume of blood in a single heartbeat than unfit individuals and can sustain a greater work level before reaching a maximum heart rate. Being more physically fit also leads to a more rapid recovery of resting heart rate.

In this experiment, you will observe how the heart responds to the increased metabolic demand of muscles during exercise.

Important: Do not attempt this experiment if physical exertion will aggravate a health problem. Inform your instructor of any possible health problems that might be exacerbated if you participate in this exercise.

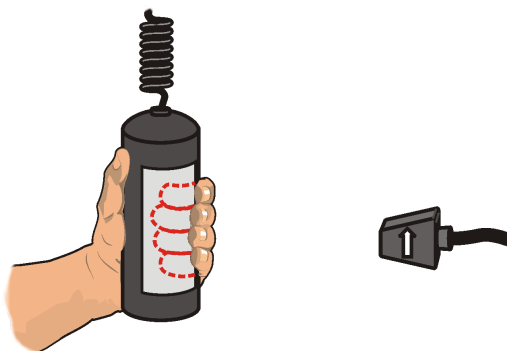


Figure 1

OBJECTIVES

In this experiment, you will

- Determine the effect of exercise on heart rate.
- Correlate the fitness level of individual with amount of daily exercise.

MATERIALS

computer
Vernier computer interface
Logger Pro

Vernier Hand-Grip Heart Rate Monitor **or**
Vernier Exercise Heart Rate Monitor
saline solution in dropper bottle
(only for use with the Exercise HR Monitor)

PROCEDURE

Select one or more persons from your lab group to be the subject(s).

1. Connect the receiver module of the Heart Rate Monitor to the Vernier computer interface.
2. Open the file “04 Heart Rate and Exercise” from the *Human Physiology with Vernier* folder.
3. Set up the Heart Rate Monitor. Follow the directions for your type of Heart Rate Monitor.

Using a Hand-Grip Heart Rate Monitor

- a. The receiver and one of the handles are marked with a white alignment arrow as shown in Figure 2. Locate these two arrows.
- b. Have the subject grasp the handles of the Hand-Grip Heart Rate Monitor so that their fingers are in the reference areas indicated in Figure 3. Hold the handles vertically.
- c. Have someone else hold the receiver near the handles so that the two alignment arrows are pointing in the same direction and are at approximately the same height as shown in Figure 2. **Note:** The receiver must stay within 60 cm of the handles during data collection.

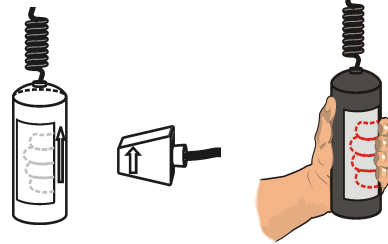


Figure 2

Figure 3

Using an Exercise Heart Rate Monitor

- a. Depending upon your size, select a small or large size elastic strap. Secure one of the plastic ends of the elastic strap to the transmitter belt. It is important that the strap provide a snug fit of the transmitter belt.
 - b. Wet each of the electrodes (the two textured oval areas on the underside of the transmitter belt) with 3 drops of saline solution.
 - c. Secure the transmitter belt against the skin directly over the base of the rib cage (see Figure 4). The POLAR logo on the front of the belt should be centered. Adjust the elastic strap to ensure a tight fit.
 - d. Take the receiver module of the Heart Rate Monitor in your right hand. Remember that the receiver must be within 80 cm of the transmitter in the Heart Rate Monitor belt.
4. Stand quietly facing your table or lab bench.
 5. To determine that everything is set up correctly, click to begin monitoring heart rate. Note that there may be up to a 30 second delay before data are seen. The readings should be within the normal range of the individual, usually between 55 and 80 beats per minute. Click when you have determined that the equipment is operating properly, and proceed to Step 6.
 6. Click to begin data collection. If the baseline appears stable, begin to run in place at 40 s. Continue data collection while running in place for the next 60 s.
 7. At approximately 100 s, stop running and stand in place while your heart rate slows toward its resting pre-exercise value. Data will be collected for 200 s.

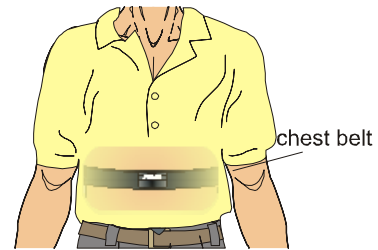



Figure 4

- Click and drag over the area of the graph where the resting heart rate is displayed (from 0 to approximately 40 s). This will highlight the region of interest.
- Click the Statistics button, . Record the mean resting heart rate in Table 1.
- Drag the right hand bracket to the right edge of the graph, until all the data points are highlighted. The values in the Statistics box will be adjusted based on the data within the brackets. Record the maximum heart rate in Table 1.
- Move the statistics brackets to highlight the area of the graph beginning with the maximum heart rate and ending with the first data point that matches the initial baseline value (or the last point graphed, if baseline is not achieved). Record the Δx value displayed at the lower left corner of the graph as the recovery time in Table 1.

DATA

Table 1	
Condition	
Resting heart rate (bpm)	
Maximum heart rate (bpm)	
Recovery time (s)	

DATA ANALYSIS

- Normal resting heart rates range from 55–100 beats per minute. What was the subject's resting heart rate? How much did the subject's heart rate increase above resting rate with exercise? What percent increase was this?
- How does the subject's maximum heart rate compare with other students in the group or class? Is this what you expected?
- Recovery time has been shown to correlate with degree of physical fitness. How does the subject's recovery rate compare to that of your classmates? Is this what you expected?

4. Congestive heart failure is a condition in which the strength of contraction with each beat may be significantly reduced. For example, the ventricle may pump only half the usual volume of blood with each beat. Would you expect a person with congestive heart failure to have a faster or slower heart rate at rest? With exercise?
5. Medications are available which can slow the heart or speed it up. If a patient complains of feeling poorly and has a heart rate of 120 beats/min, should you administer a medicine to slow the rate?

EXTENSION

Design an anonymous survey to be taken by each member of your class. In the survey, ask questions that you think might influence the test results (examples might include: gender, age, exercise regimen, coffee drinking within 2 hours of the experiment, and smoking status). Compare and contrast resting heart rates and recovery rates after exercise within and among these groups.

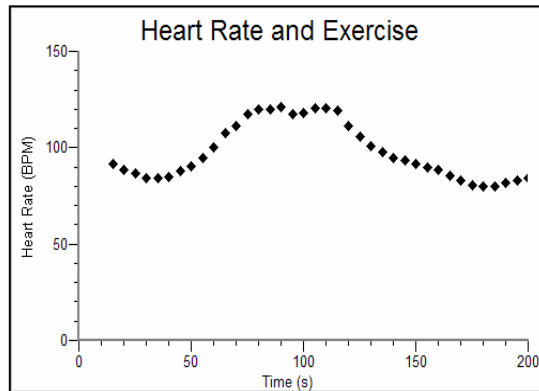
TEACHER INFORMATION

Heart Rate and Exercise

The privacy of your students needs to be respected in this exercise. We suggest that the test subjects be completely voluntary. No student should be required to perform the test if he/she is uncomfortable sharing data with others. In cases where questions refer to personal data and comparison to class data, this should be collected anonymously and compiled by the teacher.

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. This lab exercise can be done with either of two types of heart rate monitors: the Hand-Grip Heart Rate Monitor or the Exercise Heart Rate Monitor. The Hand-Grip Heart Rate Monitor, which consists of two hand grips and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through the hand grips and relaying the information to the plug-in receiver. The Exercise Heart Rate Monitor, which consists of a chest belt and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through a belt worn around the chest and relaying the information to the plug-in receiver.
3. When using either the Hand-Grip Heart Rate Monitor or the Exercise Heart Rate Monitor there will be a 15 s delay while data are collected before the first point is plotted on the upper graph. Thereafter, a point will be plotted every 5 s.
4. Computer monitors can be a source of electrical interference for both designs of the Heart Rate Monitors. To ensure that this is not a factor in your students' data-collection activities, move the receiver module as far away from the computer monitor as possible.
5. The receiver module of both designs of the Heart Rate Monitors will receive signals from the closest transmitter source. This may result in an apparent doubling of the subjects' heart rates. To avoid confusion or erroneous readings, have test subjects from different lab teams maintain a distance of at least 2 m from each other.
6. When using the Hand-Grip Heart Rate Monitor, it is important that students closely follow the instructions for holding the handles. The hand grips are marked for the right or left hand, each showing where the individuals' fingers and palms should be placed. In addition, the left hand grip and the receiver are both marked with alignment arrows. When collecting data, it is important that the arrow labels on each of these devices be in alignment (see figures in the Student version of the experiment). Failure to follow these instructions will result in unreliable data.
7. It is important to have good contact between the transmitter belt and the test subject when using the Exercise Heart Rate Monitor. It is very important that the belt fit snugly. Both electrodes should be wetted with either saline solution or contact lens solution. A 5% salt solution works well and can be prepared by adding 5 g per 100 mL of solution. Typical symptoms of inadequate contact with the electrodes are a noisy signal with erroneous peaks, missing heart beat readings, or a flat-line display. If the students receive a flat reading with no heart rate detected, have them move the transmitter and receiver closer together. The range of the transmitter in the chest belt is 60–80 cm.

SAMPLE DATA



Sample graph showing a student's heart rate data for this experiment

Table 1	
Condition	
Resting heart rate (bpm)	87
Maximum heart rate (bpm)	121
Recovery time (s)	57

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Answers will vary. Factors such as weight, regular exercise, health, medications, etc., may play a part in determining the resting and exercise heart rate.
2. Answers will vary. Factors such as effort, weight, regular exercise, health, medications, etc., may play a part in determining the maximum heart rate achieved by students in this experiment.
3. Answers will vary. Factors such as weight, regular exercise, health, medications, etc., may play a part in determining the resting and exercise heart rate.
4. A person with congestive heart failure will tend to have a more rapid heart rate at rest and with exercise. If less blood is pumped during each heart beat, the heart will attempt to compensate by beating more rapidly.
5. Students should be able to answer no to this question based on their understanding of the heart rate as a homeostatic mechanism. The heart may beat rapidly to compensate for other problems, such as blood loss and low blood pressure. In this case, slowing the heart with medication may lead to a precipitous and fatal fall in blood pressure. One should not attempt to manipulate the heart rate before ascertaining the underlying cause of the abnormality.

Heart Rate Response to Baroreceptor Feedback

One of the homeostatic mechanisms of the human body serves to maintain a fairly constant blood pressure. Major determinants of blood pressure are heart rate, amount of blood pumped with each beat (*stroke volume*), and the resistance of the arterial system which is receiving the blood. The heart rate is influenced by *baroreceptors*, special sensors in tissues in the aortic arch and carotid arteries which contain nerve endings that respond to stretching (see Figure 1). An increase or decrease in stretch sends signals to the medulla in the brain which in turn acts on the heart through the vagus nerve, completing what is called a *feedback loop*. Sudden increase in pressure in the heart or carotid arteries causes an increase in stretch of the baroreceptor sensors and results in a decrease in heart rate. Sudden lowering of pressure causes the opposite effect. This feedback loop enables us to function in a gravity environment.

Most people have experienced the sensation of dizziness after standing abruptly from a seated or squatting position. This effect can be seen in healthy individuals, but it is accentuated in the elderly and in certain conditions including dehydration and Parkinson's disease. In these cases, the increase in heart rate may be significant but is still not able to make up for an insufficiency of the other two contributors to blood pressure (i.e., low blood volume or poor regulation of the resistance of the arterial system by the sympathetic nervous system). One of the first tests performed by doctors on patients who complain of dizziness is to check the blood pressure and pulse with the patient lying down and then standing. A drop in blood pressure of 20 points or an increase in heart rate of 20 points with standing is considered significant. This condition is called *orthostatic hypotension*.

In this experiment, you will observe heart rate response to squatting and to standing from a squatting position. In the former, there is a rapid increase in venous return to the heart as veins in the leg muscles are compressed. This causes a sudden increase in stroke volume and pressure sensed by the baroreceptors. In standing from a squatting position, there is a sudden reduction in venous return to the heart because of "pooling" of blood in the legs. This results in a decrease in stroke volume and pressure.

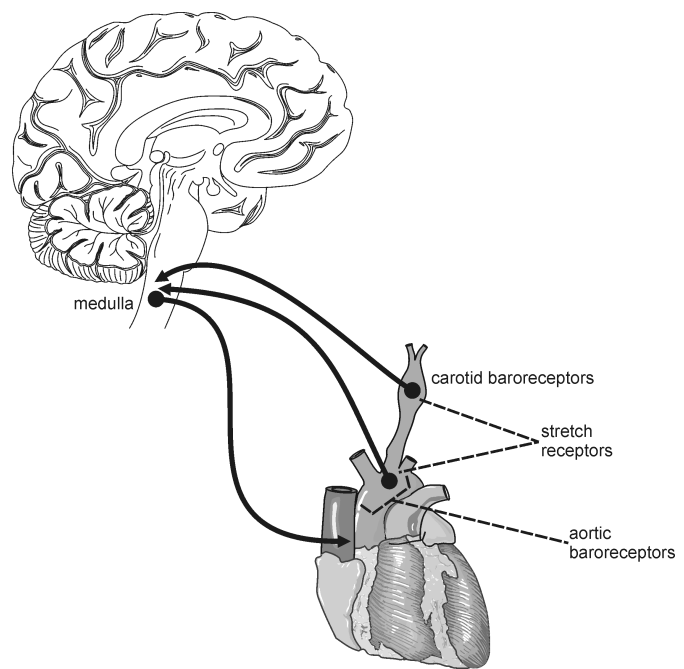


Figure 1

Important: Do not attempt this experiment if you suffer from knee pain or dizzy spells. Inform your instructor of any possible health problems that might be exacerbated if you participate in this exercise.

OBJECTIVES

In this experiment, you will

- Observe pulse response to sudden squatting.
- Observe pulse response to sudden standing from a squatting position.
- Correlate pulse response to sympathetic nervous system function.

MATERIALS

computer
Vernier computer interface
Logger Pro

Vernier Hand-Grip Heart Rate Monitor **or**
Vernier Exercise Heart Rate Monitor
saline solution in dropper bottle
(only for use with Exercise HR Monitor)

PROCEDURE

1. Connect the receiver module of the Heart Rate Monitor to the Vernier computer interface. Open the file “05 Heart Rate Response” from the *Human Physiology with Vernier* folder.
2. Set up the Heart Rate Monitor. Follow the directions for your type of Heart Rate Monitor.

Using a Hand-Grip Heart Rate Monitor

- a. The receiver and one of the handles are marked with a white alignment arrow as shown in Figure 2. Locate these two arrows.
- b. Have the subject grasp the handles of the Hand-Grip Heart Rate Monitor so that their fingers are in the reference areas indicated in Figure 3. Hold the handles vertically.
- c. Have someone else hold the receiver near the handles so that the two alignment arrows are pointing in the same direction and are at approximately the same height as shown in Figure 2. **Note:** The receiver must stay within 60 cm of the handles during data collection.



Figure 2



Figure 3

Using an Exercise Heart Rate Monitor

- a. Depending upon your size, select a small or large size elastic strap. Secure one of the plastic ends of the elastic strap to the transmitter belt. It is important that the strap provide a snug fit of the transmitter belt.
- b. Wet each of the electrodes (the two textured oval areas on the underside of the transmitter belt) with 3 drops of saline solution.
- c. Secure the transmitter belt against the skin directly over the base of the rib cage (see Figure 4). The POLAR logo on the front of the belt should be centered. Adjust the elastic strap to ensure a tight fit.
- d. Take the receiver module of the Heart Rate Monitor in your right hand. Remember that the receiver must be within 80 cm of the transmitter in the Heart Rate Monitor belt.

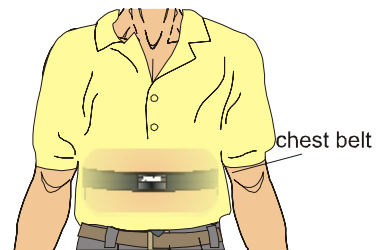


Figure 4

Heart Rate Response to Baroreceptor Feedback

3. To determine that everything is set up correctly, click to begin monitoring heart rate. Note that there may be up to a 30 second delay before data are seen. The readings should be within the normal range of the individual, usually between 55 and 80 beats per minute. Click when you have determined that the equipment is operating properly, and proceed to Step 4.
4. After at least 60 s of stable baseline data has been collected, rapidly lower yourself into a squatting position. Maintain this position until your heart rate returns to the initial baseline rate.
5. After obtaining 10–20 s of stable heart rate values, rise rapidly to a standing position. Continue to record data until the baseline heart rate has been achieved, or until the end of the run. Data will be collected for 400 s.
6. Click and drag to highlight the baseline data prior to squatting. Click the Statistics button, , and record the mean value (to the nearest whole number) as the Baseline heart rate in Table 1.
7. Drag the right bracket to include the last data point of the run. Record the minimum and maximum heart rates for the total run (to the nearest whole number) in Table 1. These values will be displayed in the Statistics box.
8. On the graph, drag the Statistics brackets to select the region between the heart rate immediately prior to squatting and the maximum or minimum heart rate (first peak or valley) that follows squatting. Record the Δx value (time) displayed in the lower left corner of the graph in Table 2 (to the nearest whole number) as “Response time 1.”
9. Repeat Step 8 for the following regions:
 - a. From the maximum or minimum heart rate following squatting to the beginning of a new stable heart rate. Record the Δx value (time) displayed in the lower left corner of the graph in Table 2 as “Recovery time 1.”
 - b. The region just prior to standing and the maximum heart rate after standing. Record the Δx value (time) displayed in the lower left corner of the graph in Table 2 as “Response time 2.”
 - c. The region between the maximum heart rate after standing and the point at which the heart rate has re-stabilized (i.e., stable for at least 40 s). Record the Δx value (time) displayed in the lower left corner of the graph in Table 2 as “Recovery time 2.”

DATA

Table 1		
Baseline heart rate (bpm)	Minimum heart rate (bpm)	Maximum heart rate (bpm)

Table 2			
Baroreceptor response time 1: Squatting (s)	Recovery time 1 (s)	Baroreceptor response time 2: Standing (s)	Recovery time 2 (s)

DATA ANALYSIS

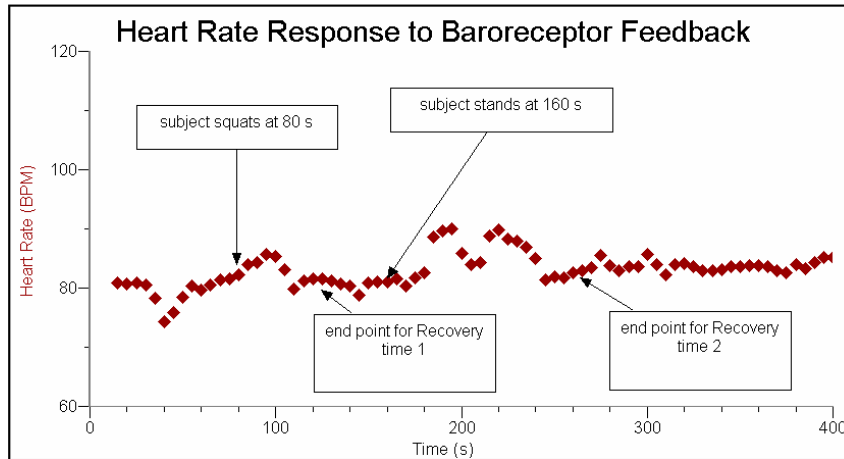
1. How much and in which direction (increase or decrease) did the heart rate change as a result of
 - a. standing?
 - b. squatting?
2. Changing the heart rate is only one of a variety of homeostatic mechanisms that maintain a fairly constant blood pressure during changes in body position. The sympathetic nervous system helps by adjusting peripheral resistance in the arterial system. As this occurs the heart rate is able to normalize again. Compare the duration of the initial direction of heart rate change after standing to the recovery time. What does your data tell you about the relative speed of the change in peripheral vascular resistance as compared to that of the heart rate response?
3. Dizziness may result from low blood pressure and can occur in patients who take medicines which impair the ability of the heart to increase its rate. Given what you have learned from your data, which daily activities would be most likely to cause dizziness in people who take these medications?
4. Using your knowledge of heart rate response to a decrease in blood volume returning to the heart, suggest a way to evaluate (without the use of medical equipment) whether significant blood loss has occurred in an accident victim.
5. The majority of astronauts who are in a microgravity environment for several weeks will experience orthostatic hypotension and dizziness on return to Earth. What are possible mechanisms for this?

TEACHER INFORMATION

Heart Rate Response to Baroreceptor Feedback

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. You may find it helpful to affix the receiver to a lab bench with tape or to a ring stand.
3. This lab exercise can be done with either of two types of heart rate monitors: the Hand-Grip Heart Rate Monitor or the Exercise Heart Rate Monitor. The Hand-Grip Heart Rate Monitor, which consists of two hand grips and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through the hand grips and relaying the information to the plug-in receiver. The Exercise Heart Rate Monitor, which consists of a chest belt and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through a belt worn around the chest and relaying the information to the plug-in receiver.
4. When using either the Hand-Grip Heart Rate Monitor or the Exercise Heart Rate Monitor there will be a 15 s delay while data are collected before the first point is plotted on the upper graph. Thereafter, a point will be plotted every 5 s.
5. Computer monitors can be a source of electrical interference for both designs of the Heart Rate Monitors. To ensure that this is not a factor in your students' data-collection activities, move the receiver module as far away from the computer monitor as possible.
6. The receiver module of both designs of the Heart Rate Monitors will receive signals from the closest transmitter source. This may result in an apparent doubling of the subjects' heart rates. To avoid confusion or erroneous readings, have test subjects from different lab teams maintain a distance of at least 2 m from each other.
7. When using the Hand-Grip Heart Rate Monitor, it is important that students closely follow the instructions for holding the handles. The hand grips are marked for the right or left hand, each showing where the individuals' fingers and palms should be placed. In addition, the left hand grip and the receiver are both marked with alignment arrows. When collecting data, it is important that the arrow labels on each of these devices be in alignment (see figures in the Student Version of the experiment). Failure to follow these instructions will result in unreliable data.
8. It is important to have good contact between the transmitter belt and the test subject when using the Exercise Heart Rate Monitor. It is very important that the belt fit snugly. Both electrodes should be wetted with either saline solution or contact lens solution. A 5% salt solution works well and can be prepared by adding 5 g per 100 mL of solution. Typical symptoms of inadequate contact with the electrodes are a noisy signal with erroneous peaks, missing heart beat readings, or a flat-line display. If the students receive a flat reading with no heart rate detected, have them move the transmitter and receiver closer together. The range of the transmitter in the chest belt is 60–80 cm.

SAMPLE DATA



Sample data

Table 1		
Baseline heart rate (bpm)	Minimum heart rate (bpm)	Maximum heart rate (bpm)
79	76	92

Table 2			
Baroreceptor response time 1: Squatting (s)	Recovery time 1 (s)	Baroreceptor response time 2: Standing (s)	Recovery time 2 (s)
21	29	31	72

ANSWERS TO THE DATA ANALYSIS QUESTIONS

- (a) Answers will vary. Three cardiovascular responses occur after squatting. First, there is a sudden increase in venous return to the heart caused by compression of veins in the legs. Second, there is the response of the heart to an increased volume of blood returning to the atria through the inferior vena cava. Finally, the resulting increase in stroke volume stretches baroreceptors, leading to a reflex decrease in heart rate.

Because of the variability and interaction of these three responses, the heart rate response to squatting will vary from individual to individual. A portion of the class will experience a decrease in heart rate. These individuals show a greater Baroreceptor sensitivity. Others will have a less sensitive Baroreceptor response and will show an increase in heart rate.

In the sample data, there was a decrease in heart rate with squatting.

- (b) Students should report an increase in heart rate after standing.

2. The response times were faster than the recovery times in both cases. This indicates that additional homeostatic mechanisms were slower to come into action than the heart response.
3. People on medicines that cause heart rate slowing are most likely to become dizzy when first getting out of bed or out of a chair. It is especially common for them to complain of dizziness during gardening, due to the squatting involved in this activity.
4. Blood loss results in a decrease in blood pressure. This will be accompanied by an increase in heart rate. A rapid pulse would indicate that possibly some significant blood loss has occurred. A normal pulse is reassuring.
5. Any answer that suggests “use it or lose it” is correct. The muscles involved in maintaining venous return from the lower extremities have weakened from disuse. Students may compose other answers such as fluid shifts that occur in space. Astronauts tend to have fluid shifts toward the upper body while in space. On reentry the sudden effect of gravity is similar to sudden standing from a squatting position.

Effect of Coughing on Heart Rate

Involuntary coughing is the result of irritation of special sensory nerves in the respiratory tract. This helps to clear potentially damaging substances from the lungs (water, foreign bodies, dust, infection, mucous, etc.). Coughing can be more deleterious than helpful, causing discomfort, preventing sleep, or leading, in some cases, to dizziness or loss of consciousness (known as *cough syncope*).

The physiologic effects resulting from a cough are numerous. There is marked increase in intrathoracic pressure just prior to expulsion of air. When blood pressure is normal, this leads to a decrease in venous return to the right side of the heart and a decrease in cardiac output. On the other hand, a cough-induced increase in intrathoracic pressure may provide a form of “internal cardiopulmonary resuscitation” in a heart attack victim whose blood pressure is falling dangerously low. In this case, coughing can be as effective as the external chest compressions of CPR in raising blood pressure and providing better blood circulation to vital tissues.

Coughing, and the resulting wide fluctuations in intrathoracic pressure it produces, causes reflex stimulation of the autonomic nervous system. The sympathetic nervous system is an “activating system,” preparing the body for a “flight or fight response” by increasing heart rate and blood pressure. The parasympathetic nervous system acts through the vagus nerve to slow the heart and to lower blood pressure. Both the sympathetic and parasympathetic systems may be stimulated or inhibited by physiologic stimuli or medications.

The following table shows potential heart rate response to stimulation or inhibition of the sympathetic or parasympathetic nervous systems:

Branch of Autonomic Nervous System	Heart Rate Response to	
	Stimulation	Inhibition
Sympathetic	↑	↓
Parasympathetic	↓	↑

In this experiment, you will observe the response of heart rate to coughing, and correlate this response to activity in the autonomic nervous systems.

Important: Do not attempt this experiment if you suffer from asthma or any condition that may be aggravated by repeated coughing.

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of the heart rate at rest.
- Observe the change in heart rate that occurs with repetitive coughing.
- Correlate heart rate response to autonomic nervous system activity.

MATERIALS

computer
Vernier computer interface
Logger Pro

Vernier Hand-Grip Heart Rate **or**
Vernier Exercise Heart Rate Monitor
saline solution in dropper bottle
(only for use with the Exercise HR Monitor)

PROCEDURE

Select one person from your lab group to be the subject.

1. Connect the receiver module of the Heart Rate Monitor to the Vernier computer interface. Open the file “06 Effect of Coughing” from the *Human Physiology with Vernier* folder.
2. Set up the Heart Rate Monitor. Follow the directions for your type of Heart Rate Monitor.

Using a Hand-Grip Heart Rate Monitor

- a. The receiver and one of the handles are marked with a white alignment arrow as shown in Figure 1. Locate these two arrows.
- b. Have the subject grasp the handles of the Hand-Grip Heart Rate Monitor so that their fingers are in the reference areas indicated in Figure 2. Hold the handles vertically.
- c. Have someone else hold the receiver near the handles so that the two alignment arrows are pointing in the same direction and are at approximately the same height as shown in Figure 1. **Note:** The receiver must stay within 60 cm of the handles during data collection.

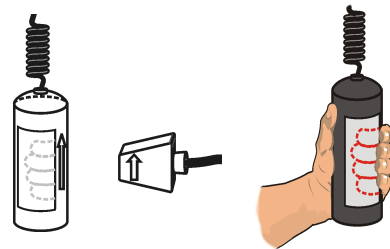


Figure 1

Figure 2

Using an Exercise Heart Rate Monitor

- a. Depending upon your size, select a small or large size elastic strap. Secure one of the plastic ends of the elastic strap to the transmitter belt. It is important that the strap provide a snug fit of the transmitter belt.
 - b. Wet each of the electrodes (the two textured oval areas on the underside of the transmitter belt) with 3 drops of saline solution.
 - c. Secure the transmitter belt against the skin directly over the base of the rib cage (see Figure 3). The POLAR logo on the front of the belt should be centered. Adjust the elastic strap to ensure a tight fit.
 - d. Take the receiver module of the Heart Rate Monitor in your right hand. Remember that the receiver must be within 80 cm of the transmitter in the Heart Rate Monitor belt.
3. Click to begin data collection. There will be a 15 s delay while data are collected before the first point is plotted on the upper graph. Thereafter, a point will be plotted every 5 s. Obtain approximately 45 s of graphed data as a baseline heart rate.
 4. At approximately 60 s, begin to cough continuously (every 1–3 s). Heart rate will change in response to the coughing. When the heart rate levels off for at least 15 s (or reverses direction), stop coughing. Data will be collected for 400 s.

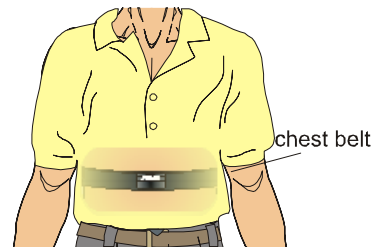



Figure 3

5. Click and drag to highlight the baseline heart rate data points on the graph. Click the Statistics button, , and record the mean value to the nearest whole number as the Baseline heart rate in Table 1.
6. Drag both of the Statistics brackets to enclose the region where the heart rate levels off. Record the mean heart rate to the nearest whole number as the Maximum heart rate in Table 1. Close the Statistics box by clicking the × in the corner of the box.
7. Determine the difference between the Maximum heart rate and the Baseline heart rate and record this value in Table 1.
8. Determine the time between the onset of coughing and the beginning of the plateau. To do this, click and drag between approximately 60 s (time at onset of coughing) and the beginning of the plateau. Record the Δx value that is displayed at the lower left corner of the graph to the nearest whole number in Table 1.
9. Determine the recovery time. To do this, click and drag between the last point on the plateau and the point at which the heart rate returns to baseline. If the baseline heart rate is not achieved before the end of the experiment, drag to include the last data point recorded. Record the Δx value that is displayed at the lower left corner of the graph to the nearest whole number in Table 1.

DATA

Table 1				
Baseline heart rate (bpm)	Maximum heart rate (bpm)	Δ Heart rate (Max–baseline) (bpm)	Response time (s)	Recovery time (s)

DATA ANALYSIS

1. In what direction did your heart rate change in this experiment? According to the table in the Introduction, what are the possible explanations for this change?
2. There are medications that can selectively block the action of either sympathetic or parasympathetic influences on the heart. How could such medications be used to determine which of these systems is responsible for a change in heart rate such as was seen in this experiment?

Computer 6

3. Compare the response and recovery times recorded in Table 1. List possible survival advantages of the differences you see.
4. The parasympathetic and sympathetic nerve supplies are severed during heart transplantation and are not surgically repaired. Would a heart transplant recipient's heart rate change with coughing (or with a severe fright)?
5. You are in a remote location and a member of your party complains of chest pain and dizziness. You find that his pulse is 35 bpm. You immediately call 911 and are told that it will take 15 minutes for the helicopter to arrive. You know that CPR should not be performed on conscious individuals. Drawing from the knowledge you have gained from this experiment, what might be done to improve your patient's pulse and blood pressure?

EXTENSION

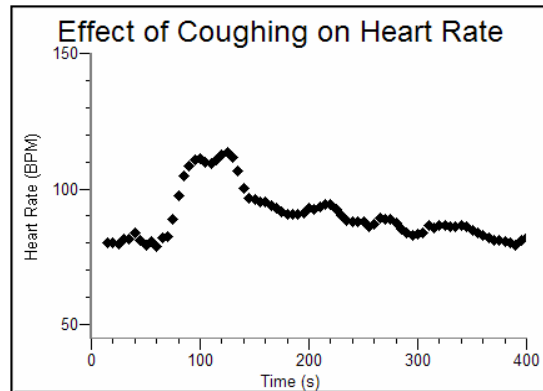
Perform the experiment with different coughing frequencies and intensities.

TEACHER INFORMATION

Effect of Coughing on Heart Rate

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Position students in such a way that they are coughing away from other students. Students with active colds should not participate in this experiment.
3. Repetitive coughing will be more comfortable for students if small inhalations are used.
4. This lab exercise can be done with either of two types of heart rate monitors: the Hand-Grip Heart Rate Monitor or the Exercise Heart Rate Monitor. The Hand-Grip Heart Rate Monitor, which consists of two hand grips and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through the hand grips and relaying the information to the plug-in receiver. The Exercise Heart Rate Monitor, which consists of a chest belt and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through a belt worn around the chest and relaying the information to the plug-in receiver.
5. When using either the type of Heart Rate Monitor, there will be a 15 s delay while data are collected before the first point is plotted on the upper graph. Thereafter, a point will be plotted every 5 s.
6. Computer monitors can be a source of electrical interference for both the Hand-Grip and the Exercise Heart Rate Monitor. To ensure that this is not a factor in your students' data-collection, move the receiver module as far away from the computer monitor as possible.
7. The receiver module of both designs of the Heart Rate Monitors will receive signals from the closest transmitter source. This may result in an apparent doubling of the subjects' heart rates. To avoid confusion or erroneous readings, have test subjects from different lab teams maintain a distance of at least 2 m from each other.
8. When using the Hand-Grip Heart Rate Monitor, students should avoid moving their arms when they cough as this may affect data collection. For this reason, it may be helpful to rest the sensor handles on the thighs or lab bench during the experiment. Also, it is important that students closely follow the instructions for holding the handles. The hand grips are marked for the right or left hand, each showing where the individuals' fingers and palms should be placed. In addition, the left hand grip and the receiver are both marked with alignment arrows. When collecting data, it is important that the arrow labels on each of these devices be in alignment (see figures in the student version of the experiment). Failure to follow these instructions will result in unreliable data.
9. It is important to have good contact between the transmitter belt and the test subject when using the Exercise Heart Rate Monitor. It is very important that the belt fit snugly. Both electrodes should be wetted with either saline solution or contact lens solution. A 5% salt solution works well and can be prepared by adding 5 g per 100 mL of solution. Typical symptoms of inadequate contact with the electrodes are a noisy signal with erroneous peaks, missing heart beat readings, or a flat-line display. If the students receive a flat reading with no heart rate detected, have them move the transmitter and receiver closer together. The range of the transmitter in the chest belt is 60–80 cm.

SAMPLE DATA



Student's heart rate response to repeated coughing

Table 1				
Baseline heart rate (bpm)	Maximum heart rate (bpm)	Δ Heart rate (Max-Baseline) (bpm)	Response time (s)	Recovery time (s)
82	119	37	51	274

ANSWERS TO THE DATA ANALYSIS QUESTIONS

- Heart rate increases during coughing for two possible reasons: Either the sympathetic system has greater stimulation or the parasympathetic system has greater inhibition. Without further testing, it is not possible to determine which of these factors is at work in this experiment.
- Answers will vary. Students should reason that the increase in heart rate seen in this experiment was due either to the sympathetic stimulation or parasympathetic inhibition. If the action of one of the systems is blocked (i.e., use a beta blocker to inhibit the sympathetic nervous system) and the heart rate increases, there must have been inhibition of the parasympathetic system. The converse is also true. However it has been found experimentally that coughing acts through the parasympathetic system.
- Recovery time is much longer than response time. Students may postulate that response to danger must be rapid to prepare for a burst of musculoskeletal activity (the “fight or flight” response), whereas there is no compelling survival advantage to a quick return to baseline. Students may notice some undulations in the recovery data as homeostasis is restored. During recovery, there is interplay between the sympathetic and parasympathetic nervous systems (which are opposing systems), that leads to overcorrection. A more rapid recovery would likely lead to greater overcorrection, and larger fluctuations from homeostatic levels.

4. Because both sympathetic and parasympathetic nervous pathways have been severed, a heart transplant recipient will not exhibit a rapid increase in heart rate when frightened or coughing. Students may be interested to learn that there may be a more gradual response seen with fright or stress because of the influences of circulating hormones that are released (e.g., epinephrine, cortisol). It may also be interesting to discuss with students the heart's intrinsic ability to change rate during exercise in response to an increased venous return, which causes stretching of the myocardium and stronger contraction of the ventricles (called *Starling's Mechanism*).
5. On the basis of what has been learned from this experiment (coughing increases heart rate), students should recommend that the patient begin coughing. Students who have taken first aid or CPR courses may also recommend that the patient lie down and elevate the legs above the heart to increase blood pressure. Other stimuli might also be applied, such as shaking or shouting.

Blood Pressure as a Vital Sign

Blood pressure is a measure of the changing fluid pressure within the circulatory system. It varies from a peak pressure produced by contraction of the left ventricle, to a low pressure, which is maintained by closure of the aortic valve and elastic recoil of the arterial system. The peak pressure is called *systole*, and the pressure that is maintained even while the left ventricle is relaxing is called *diastole* (see Figure 1).

Mean arterial pressure (MAP) is not a simple average of the two pressures, because the duration of diastole is twice that of systole. MAP is used by emergency room and intensive care unit personnel as a measure of the adequacy of blood supplied to vital tissues (such as the brain, heart, and kidneys) when the blood pressure is dangerously low.

Blood pressure is traditionally reported with the *systolic* pressure stated first and the *diastolic* pressure stated second. In adults, 120/80 and below is considered normal blood pressure. High blood pressure is 140/90 or above. The seriousness of low blood pressure, as well as the health risks of high blood pressure (also called *hypertension*), have been elucidated over the past several decades. High blood pressure is a major risk factor for a number of health problems including strokes and congestive heart failure. Diet and exercise are beneficial, but many people require medication for optimal blood pressure control.

In this experiment, you will examine your blood pressure using the Vernier Blood Pressure Sensor. You will compare blood pressures taken before and after exposure to cold. The cold stimulus activates the sympathetic nervous system, resulting in hemodynamic changes that prepare the body for a “fight or flight” response (i.e., when fighting or running from danger).

The sensitivity of blood pressure to harmful external or internal injuries makes it useful as a *vital sign*, an indicator of health, disease, excitement, and stress.

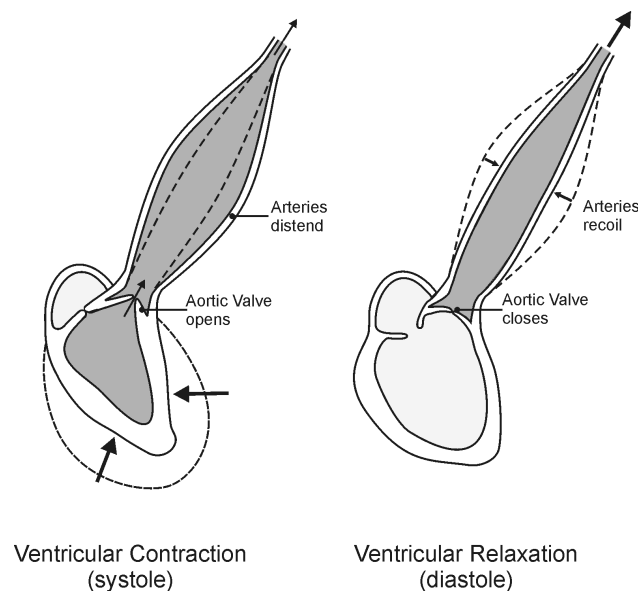


Figure 1

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of blood pressure.
- Compare blood pressure before and after exposure to cold stimulus.
- Observe an example of sympathetic nervous system activation (“fight or flight” response).

MATERIALS

computer
Vernier computer interface
Logger Pro

Vernier Blood Pressure Sensor
ice water bath
towel (paper or cloth)

PROCEDURE

Select one or more persons from your lab group to be the subject(s).

Part I Baseline Blood Pressure

1. Connect the Blood Pressure Sensor to the Vernier computer interface. There are two rubber tubes connected to the pressure cuff. One tube has a black Luer-lock connector at the end and the other tube has a bulb pump attached. Connect the Luer-lock connector to the stem on the Blood Pressure Sensor with a gentle half turn.
2. Open the file “07 Blood Press Vital Sign” from the *Human Physiology with Vernier* folder.
3. Attach the Blood Pressure cuff firmly around the upper arm, approximately 2 cm above the elbow. The two rubber hoses from the cuff should be positioned over the biceps muscle (brachial artery) and not under the arm (see Figure 2).
4. Have the subject sit quietly in a chair with his or her forearm resting on a table surface. *The person having his or her blood pressure measured must remain still during data collection; there should be no movement of the arm or hand during measurements.*
5. Click to begin data collection. Immediately pump the bulb pump until the cuff pressure reaches at least 160 mm Hg. Stop pumping. The cuff will slowly deflate and the pressure will fall. During this time, the systolic, diastolic, mean arterial pressures, and pulse will be calculated by the software. These values will be displayed on the computer screen. When the cuff pressure drops below 50 mm Hg, the program will stop calculating blood pressure. At this point, you can terminate data collection by clicking . Release the pressure from the cuff, but do not remove it.
6. Enter the pulse and the systolic, diastolic, and mean arterial pressures in Table 1.
7. Store the data by choosing Store Latest Run from the Experiment menu.

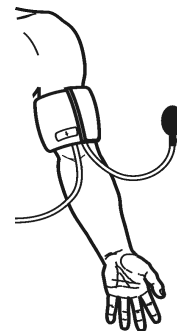


Figure 2

Part II Blood Pressure Response to Cold

8. Prepare an ice water bath for use in the next step. The subject will be instructed to place his or her opposite hand (the one to which the Blood Pressure cuff is not attached) in the ice water bath for 15 s.
9. Collect data to examine the body's response to cold.
 - a. With the cuff still attached, have the subject from Part I put the hand of his or her non-cuffed arm in the ice water bath.
 - b. As soon as the subject's hand enters the ice water bath, click .
 - c. Pump the bulb until the cuff pressure reaches at least 160 mm Hg, then stop pumping.
 - d. When data have been collected for 15 s, have the subject remove his or her hand from the ice water bath.
 - e. The systolic, diastolic, and mean arterial pressures will be calculated by the software. These values will be displayed on the computer screen. When the blood pressure readings have stabilized (after the pressure drops to 50 mm Hg), the program will stop calculating blood pressure. At this point, you can terminate data collection by clicking . Release the pressure from the cuff, and remove the cuff from the subject's arm.
10. Enter the systolic, diastolic, and mean arterial pressures, and the pulse in Table 2.

DATA

Table 1–Baseline Blood Pressure			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (beats/minute)

Table 2–Blood Pressure Response to Cold			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (beats/minute)

DATA ANALYSIS

1. Describe the trends that occurred in the systolic pressure, diastolic pressure, mean arterial pressure, and pulse with cold stimulus. How might these be useful in a “fight or flight” response?

Computer 7

- Vasovagal syncope* is a condition in which severe pain or fright activates the parasympathetic nervous system instead of the sympathetic nervous system, resulting in fainting. Keeping in mind that the parasympathetic system causes a response opposite to that of the sympathetic system, describe the hemodynamic changes that would explain this.
- As a vital sign, blood pressure is an indicator of general health. A high blood pressure (140/90 or higher) increases the risk of cardiovascular disease and strokes. Collect the systolic and diastolic pressures for the class and calculate the average for each. Rate the class average blood pressure using the following scale:

Blood Pressure	Category
140/90 or higher	High
120–139/80–89	Pre-hypertension
119/79 or below	Normal

EXTENSION

Blood pressure is traditionally obtained by using a stethoscope to listen to the brachial artery. The pumping of air into the blood pressure cuff acts to stop the blood flow through this artery. As the pressure is released, the blood again is allowed to flow. When the blood begins to flow, pulsations can be heard through the stethoscope. The pressure in the cuff at that time can be noted, and corresponds closely to the systolic blood pressure. As pressure continues to be released from the cuff, the pulsations of the artery become less audible. The pressure at which they disappear has been found to approximate the diastolic pressure. These sounds are known as *Korotkoff Sounds*.

With a stethoscope, obtain the blood pressure of a classmate by listening for the appearance and disappearance of pulsations as the pressure in the cuff is released. Compare this to the blood pressure you obtained with the Vernier Blood Pressure Sensor.

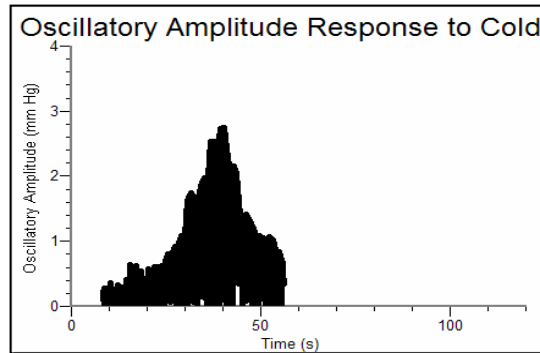
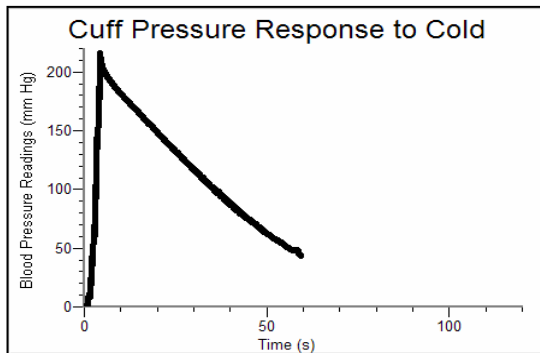
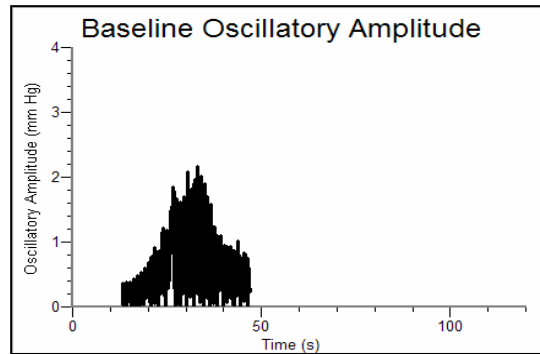
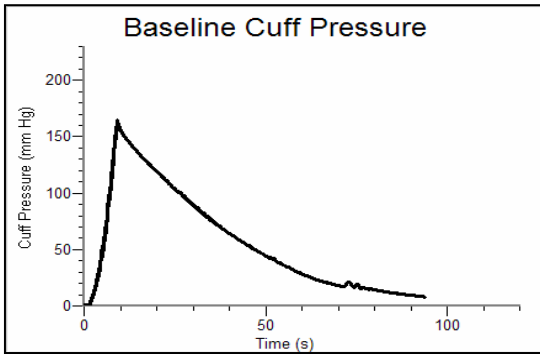
TEACHER INFORMATION

Blood Pressure as a Vital Sign

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Blood pressure may vary from reading to reading for a variety of reasons. It is normal for blood pressure to vary with the time of day or to be elevated in times of stress. For this reason, single blood pressure readings are not adequate for a diagnosis of abnormal blood pressure. It is normal for health care professionals to repeat blood pressure measurements over a period of several weeks before diagnosing high blood pressure.
3. Alert the students to be sure to secure the cuff around the subjects' arms very snugly. A loosely secured cuff will return erratic data.
4. The duration of the experiment is set for 120 s in order to accommodate individual differences in the determination of blood pressure. When the blood pressure readings have stabilized (after the pressure drops to 50 mm Hg), data collection can be terminated and pressure released from the cuff.
5. If the pressure does not reach 50 mm Hg by the time data collection ends, adjust the exhaust rate of the pressure release valve according to the directions listed under "Adjusting the Pressure Release Valve" in the Blood Pressure Sensor booklet that accompanies the sensor. This sensor booklet is also available on the Vernier web site, www.vernier.com/probes.
6. Graphs of oscillatory amplitude are included to aid in assuring that the pressure sensor is sending enough data for Logger *Pro* to correctly calculate blood pressure.

Experiment 7

SAMPLE DATA



Baseline and response to cold data for cuff pressure and oscillatory amplitude

Table 1–Baseline Blood Pressure			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (beats/minute)
105	69	82	77

Table 2–Blood Pressure Response to Cold			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (beats/minute)
112	73	88	74

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Cold exposure caused discomfort or mild pain, which led to activation of the sympathetic nervous system. Sympathetic nervous system activation increases heart rate and peripheral vascular resistance (by constricting arterioles), which leads to an elevation of systolic, diastolic and mean arterial blood pressures.

2. Sudden activation of the parasympathetic nervous system has the opposite effect of sympathetic activation. The heart rate may slow significantly. Dilatation of peripheral blood vessels leads to pooling of blood in the legs and lack of blood return to the heart. These two factors together cause a precipitous fall in blood pressure, lack of adequate blood supply to the brain, and fainting.
3. A discussion of the general health and well-being of the students in the class (as relates to blood pressure) would be an appropriate conclusion to this experiment.

Blood Pressure and Exercise

Constant pressure is required to pump blood through the circulatory system. This ensures the delivery of oxygen and nutrients to and the removal of carbon dioxide and waste products from tissues. Positive pressure is created by forceful contraction of the left ventricle of the heart, measured as *systole*. It is maintained during relaxation of the ventricle by closure of the aortic valve and recoil of arteries, measured as *diastole* (see Figure 1).

Mean arterial pressure (MAP) is a useful measure of the adequacy of tissue perfusion. It is not a simple average of systolic and diastolic blood pressures. This is because diastole continues for twice as long as systole. MAP can be reasonably approximated using the equation:

$$\frac{(\text{systole} + 2(\text{diastole}))}{3} = \text{MAP}$$

The mean arterial pressure is directly proportional to *cardiac output* and inversely proportional to *total peripheral resistance*, where:

Cardiac output is the amount of blood pumped out of the heart with each beat (called the *stroke volume*) multiplied by the number of beats per minute.

Total peripheral resistance depends on blood viscosity, length of the arterial system, diameter and elasticity of the blood vessels, and the pressure entering versus leaving the arterial system (systolic pressure minus the pressure in the venous system).

In this experiment, you will examine your blood pressure using the Vernier Blood Pressure Sensor. You will compare blood pressure readings taken before and after exercise and measure changes in systolic, diastolic, and mean arterial pressures. You will also consider the effect that exercise has on cardiac output and peripheral vascular resistance.

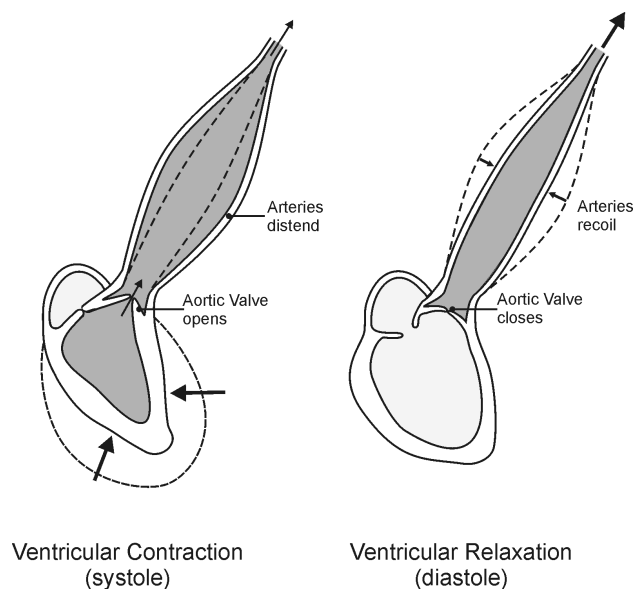


Figure 1

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of blood pressure.
- Compare changes in systolic, diastolic, and mean arterial pressures with exercise.
- Use blood pressure readings and pulse to infer changes in cardiac output and peripheral vascular resistance with exercise.

MATERIALS

computer
Vernier computer interface
Logger Pro

Vernier Blood Pressure Sensor
watch with second hand

PROCEDURE

Select one person from your lab group to be the subject.

Part I Baseline Blood Pressure

1. Connect the Blood Pressure Sensor to the Vernier computer interface. Open the file “08 Blood Pressure Exercise” from the *Human Physiology with Vernier* folder.
2. Attach the Blood Pressure Sensor to the blood pressure cuff if it is not already attached. There are two rubber tubes connected to the cuff. One tube has a black Luer-lock connector at the end and the other tube has a bulb pump attached. Connect the Luer-lock connector to the stem on the Blood Pressure Sensor with a gentle half turn.
3. Attach the Blood Pressure cuff to the upper arm, approximately 2 cm above the elbow. The two rubber hoses from the cuff should be positioned over the biceps muscle (brachial artery) and not under the arm (see Figure 2).
4. Have the subject sit quietly in a chair and avoid moving his or her arm or hand during blood pressure measurements.
5. Click to begin data collection. Immediately begin to pump until the cuff pressure reaches at least 160 mm Hg. Stop pumping.
6. Immediately feel for the radial pulse (place two or three fingers over the radial artery in the wrist proximal to the thumb) in the arm without the Blood Pressure cuff. Count the beats over 20 s. Multiply by 3 to obtain the number of beats per minute. Record this value in Table 1.
7. During this time the systolic, diastolic, and mean arterial pressures will be calculated by the software. These values will be displayed on the computer screen. When the blood pressure readings have stabilized (after the pressure drops to 50 mm Hg), the program will stop calculating blood pressure. At this point, you can terminate data collection by clicking . Release the pressure from the cuff, but do not remove it.
8. Store your data by choosing Store Latest Run from the Experiment menu.
9. Enter the systolic, diastolic, and mean arterial pressures in Table 1.

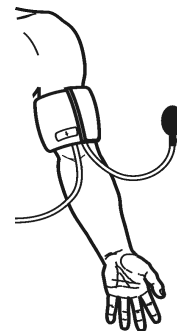


Figure 2

Part II Blood Pressure After Exercise

10. With the blood pressure cuff still attached to his/her arm, the subject from Part I of the experiment should run in place for 2 minutes. At the end of 2 minutes, the subject should sit down, placing his or her arm on the table surface.
11. Click to begin data collection. Immediately begin to pump until the cuff pressure reaches at least 170 mm Hg and then stop pumping.
12. Immediately feel for the radial pulse (place two or three fingers over the radial artery in the wrist proximal to the thumb) in the arm without the Blood Pressure cuff. Count the beats over 20 s. Multiply by 3 to obtain the number of beats per minute. Record this value in Table 2.
13. During this time the systolic, diastolic, and mean arterial pressures will be calculated by the software. These values will be displayed on the computer screen. When the blood pressure readings have stabilized (after the pressure drops to 50 mm Hg), the program will stop calculating blood pressure. At this point, you can terminate data collection by clicking . Release the pressure from the cuff, and remove the cuff from the subject's arm.
14. Enter the systolic, diastolic, and mean arterial pressures in Table 2.

DATA

Table 1–Baseline Blood Pressure			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (bpm)

Table 2–Blood Pressure After Exercise			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (bpm)

DATA ANALYSIS

1. Describe the trends that occurred in the systolic pressure, diastolic pressure, mean arterial pressure, and pulse with exercise. Assume that the stroke volume increased from 75 mL/beat to 100 mL/beat. Use this information and the change in pulse with exercise to calculate the change in *cardiac output* (stroke volume × heart rate) that occurred per minute.

Computer 8

2. *Pulse pressure* is the difference between systolic pressure (peak pressure during active contraction of the ventricles) and diastolic pressure (the pressure that is maintained even while the left ventricle is relaxing). Describe the change in pulse pressure seen with exercise. Which component of the blood pressure is most responsible for this change?

3. A change in pulse pressure can be seen in a variety of medical conditions. What would you expect to happen to the pulse pressure in the following examples?
 - (a) In atherosclerosis there is a hardening of the arterial walls.

 - (b) A damaged aortic valve does not seal properly and allows blood to flow back into the ventricle during diastole.

EXTENSIONS

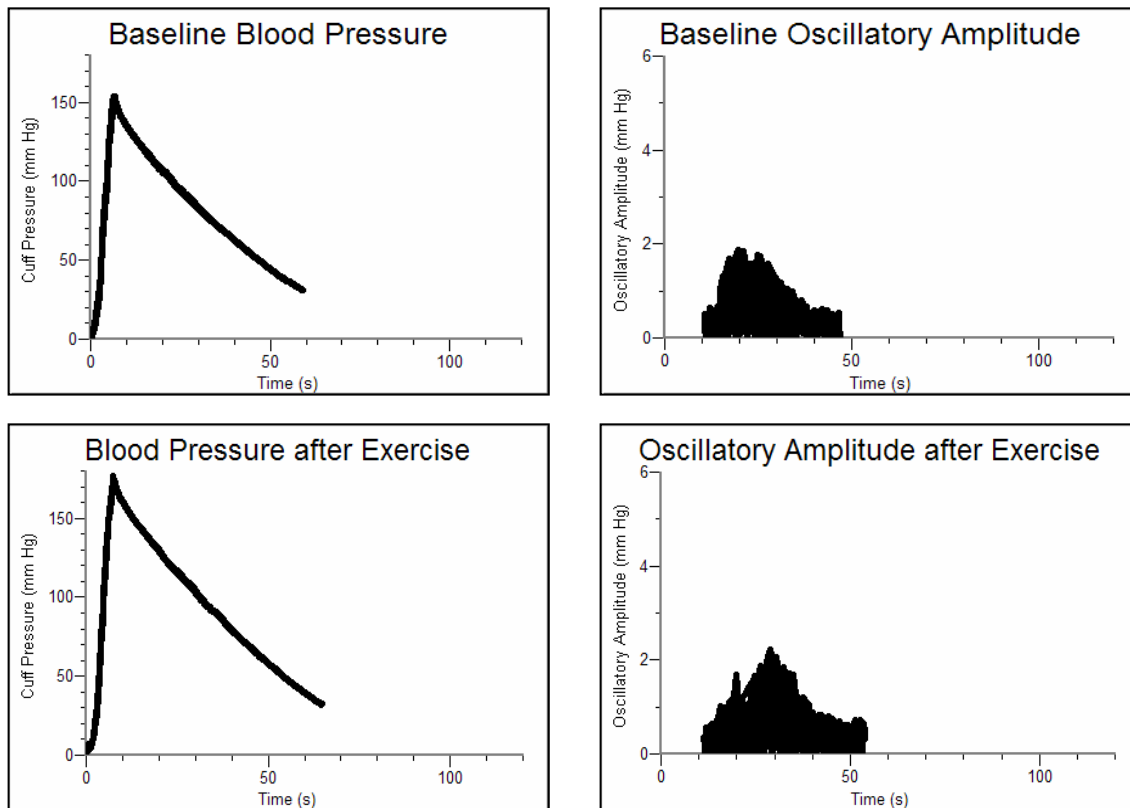
1. Exercise for varying lengths of time and use Page 2 of the experiment file to compare the blood pressure readings taken after each exercise period.
2. Measure blood pressure 5, 10, and 15 minutes after stopping exercise, and use Page 2 of the experiment file to compare readings.
3. Measure blood pressure while lifting a weight with the opposite arm.

TEACHER INFORMATION

Blood Pressure and Exercise

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Since blood pressure will be elevated after exercise, the cuff should be inflated to 170–190 mm Hg (rather than to 160 mm Hg) in Part II of the experiment.
3. The pulse displayed in the meter at the lower left of the screen is an average pulse taken over the time during which the blood pressure is measured. Thus, it is accurate at rest but cannot be used to determine the pulse rate immediately after exercise. To do this, the students are instructed to feel for the radial pulse and count the beats over 20 s in Part II of the experiment.
4. Graphs of oscillatory amplitude are included to aid in assuring that the pressure sensor is sending enough data for Logger *Pro* to correctly calculate blood pressure.

SAMPLE DATA



Baseline and post-exercise data for cuff pressure and oscillatory amplitude

Experiment 8

Table 1–Baseline Blood Pressure			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (bpm)
121	89	109	95

Table 2–Blood Pressure After Exercise			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (bpm)
133	87	108	112

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Most students should report that the systolic pressure increased with exercise. Diastolic pressure may increase, decrease or remain the same. Mean arterial pressure should be higher as should the pulse.

Resting cardiac output = (75 mL/beat) (95 beats/min) = 7125 mL/min = 7.1 L/min

Cardiac output immediately after exercise = (100 mL/beat) (112 beats/min) = 11200 mL/min = 11 L/min

2. Students should see an increase in pulse pressure because of a rise in systolic pressure. This is due to the increase in cardiac output resulting from exercise.
3. (a) Pulse pressure depends on *distensibility* of arterial walls, as well as the cardiac output. In atherosclerosis there is a hardening of arterial walls, making them less distensible. This causes a higher systolic pressure and a higher pulse pressure.
(b) Diastolic pressure is maintained in part by closure of the aortic valve. When this valve fails to close completely, there is a backflow of blood into the left ventricle, reducing the accompanying increase in systolic pressure generated by the extra volume of blood pumped with each contraction of the left ventricle (blood from the left atrium plus regurgitant blood from the aorta).

Diurnal Blood Pressure Variation

Blood pressure is determined by the output of the heart and the resistance to flow in the vascular system. These, in turn, are affected by factors such as the levels of certain hormones (renin, angiotensin II, aldosterone, cortisol, epinephrine, and norepinephrine), physical activity, emotional or physical stress, salt intake, and smoking. Blood pressure usually reaches a high at 6 am and a low between 2 and 4 a.m. Small peaks and valleys occur every one to two hours throughout the day. For this reason, health care providers measure blood pressure at random times on multiple occasions before diagnosing *hypertension*, or elevated blood pressure.

Nearly one in three American adults has high blood pressure, and of those who have high blood pressure one-third do not know that they have it. Uncontrolled hypertension increases the risk of stroke, mental impairment, heart disease and kidney disease. The following table shows the guidelines set forth by the Joint National Committee on Prevention, Detection, Evaluation, and Treatment of High Blood Pressure in 2003.

Blood Pressure	Category
140/90 or higher	High
120–139/80–89	Pre-hypertension
119/79 or below	Normal

When hypertension is diagnosed, a clinician may prescribe lifestyle changes (e.g., diet, exercise, stress reduction). If these are ineffective, medication is recommended. Different categories of medication act on the heart, peripheral vascular system, central or peripheral nervous system, or hormonal system to lower the blood pressure.

In this experiment, you will take multiple blood pressure readings over the course of two or more days and analyze the variability in these readings.

Important: The equipment used in this experiment is for educational purposes only and should not be used to diagnose blood pressure disorders.

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of blood pressure measured at different times of the day.
- Analyze the variability of blood pressure readings individually and as a class.
- Correlate your findings with variables that may have influenced your blood pressure.

MATERIALS

computer
Vernier computer interface

Logger Pro
Vernier Blood Pressure Sensor

PROCEDURE

1. Connect the Blood Pressure Sensor to the Vernier computer interface. Open the file "09 Diurnal Blood Pressure" from the *Human Physiology with Vernier* folder.
2. Attach the Blood Pressure Sensor to the blood pressure cuff if it is not already attached. There are two rubber tubes connected to the cuff. One tube has a black Luer-lock connector at the end and the other tube has a bulb pump attached. Connect the Luer-lock connector to the stem on the Blood Pressure Sensor with a gentle half turn.
3. Attach the blood pressure cuff to the upper arm, approximately 2 cm above the elbow. The two rubber hoses from the cuff should be positioned over the biceps muscle (brachial artery) and not under the arm (see Figure 1).
4. Have the subject sit quietly in a chair and avoid moving his or her arm or hand during blood pressure measurements.
5. Click to begin data collection. Immediately begin to pump until the cuff pressure reaches at least 160 mm Hg. Stop pumping. The cuff will slowly deflate and the pressure will fall. During this time the pulse and the systolic, diastolic, and mean arterial pressures will be calculated by the software. These will be displayed on the computer screen. When the blood pressure readings have stabilized (after cuff pressure drops below 50 mm Hg), click to end data collection and release the pressure from the cuff.
6. Store your data by choosing Store Latest Run from the Experiment menu.
7. Choose Text Annotation from the Insert menu. Label the run with the date and time it was taken.
8. Enter the pulse and the systolic, diastolic, and mean arterial pressures in Table 1. These values are also stored on Page 2 of the experiment file.
9. Save the experiment file as instructed by your teacher.
10. To collect additional data, open your experiment file and repeat Steps 2–9. Take at least 6 readings over 2 or more days. The readings taken during any one day should be separated by at least 3–4 hours. Different times of the day should be represented in your data (i.e., early morning, late morning, afternoon).
11. Complete Table 2 using either the data that was stored on Page 2 of your experiment file or the data you recorded in Table 1.
12. Compile class data to complete Table 3.



Figure 1

DATA

Table 1—Individual Data					
Run	Time of day (hour: minute)	Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	MAP (mm Hg)	Pulse (bpm)

Table 2—Individual Data			
Blood pressure reading	Maximum	Minimum	Δ Blood pressure
Systolic (mm Hg)			
Diastolic (mm Hg)			
MAP (mm Hg)			
Pulse (bpm)			

Table 3—Class Average Data				
Time of day	Systolic (mm Hg)	Diastolic (mm Hg)	MAP (mm Hg)	Pulse (bpm)
Early morning (6–8 a.m.)				
Mid-morning (8–10 a.m.)				
Late morning (10–12 p.m.)				
Early afternoon (12–2 p.m.)				
Mid-afternoon (2–4 p.m.)				
Late afternoon (4–5 p.m.)				

DATA ANALYSIS

1. At what time of day did you record your highest blood pressure? Lowest? How much variation was there between these two readings? It is common to see a higher blood pressure in the early morning hours. Do your findings agree with this? Do the class average readings agree with this?
2. List any factors that may have affected your blood pressure at the various times at which it was measured (i.e., relation to lunch, physical activity, stress, smoking).
3. According to your data, does it appear that the blood pressure is dependent on pulse? Explain your findings based on the factors that determine blood pressure, as listed in the Introduction to this experiment.

4. By what percent did your systolic blood pressure vary? Use the following equation to calculate this value:

$$\frac{\Delta\text{Blood Pressure}}{\text{Minimum Blood Pressure}} \times 100\%$$

5. Find the percent change in your diastolic pressure, MAP, and pulse using similar equations.
6. Calculate the percent change from the upper limit of normal for systolic and diastolic blood pressures (seen in the chart on the first page of this experiment) and the lower limit of hypertension.
7. Your patient, Mrs. Jones, is visiting you for her annual physical. Her blood pressure is mildly elevated. What should you recommend to her as a course of action?

8. Adrenalin (epinephrine) is used to treat acute asthma attacks, severe allergic reactions, and shock. What would you anticipate would happen to blood pressure and pulse after a shot of epinephrine has been given?

9. Blood pressure is directly related to (1) heart rate, (2) resistance in the peripheral blood vessels, (3) blood volume, and (4) levels of stress hormones, among other factors. For each of the following classes of medicines, describe which of these four factors is affected:
 - a. calcium channel blockers dilate arterioles

 - b. diuretics cause a loss of fluid from the body

 - c. angiotensin-converting enzyme inhibitors stop conversion of angiotensin I to angiotensin II, which is a potent constrictor of blood vessels

 - d. beta blockers inhibit the sympathetic nervous system

EXTENSIONS

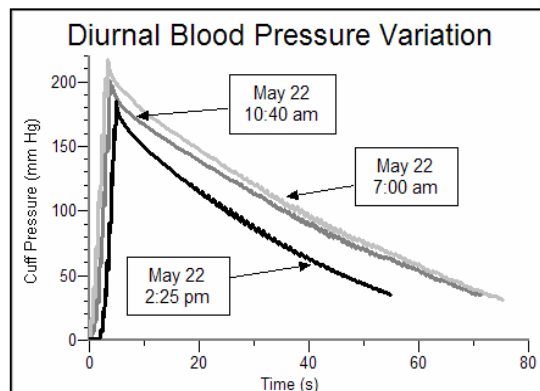
1. Continue blood pressure measurements beyond the hours prescribed in this experiment (i.e. between every class period/hourly/late at night, etc.).
2. Measure blood pressure before and after playing a fast-paced video game for 10 minutes.

TEACHER INFORMATION

Diurnal Blood Pressure Variation

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. The Blood Pressure cuff functions best when the rubber tubes are centered over the brachial artery.
3. While most people will exhibit variation in blood pressure throughout a 24-hour period, the amount of variation may differ. Some students may have a more consistent blood pressure, and some will experience wider fluctuations.
4. In Logger *Pro*, as additional blood pressure readings are obtained, direct students' attention to Page 2 of the experiment file, where individual readings are compared graphically. LabQuest users can enter data in the Data Table to create a graph.

SAMPLE DATA



Sample data for the first day of data collection

Run	Time of day (hour: minute)	Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	MAP (mm Hg)	Pulse (bpm)
1	8:00 p.m.	118	81	101	70
2	7:00 a.m.	130	76	102	92
3	10:40 a.m.	114	62	84	75
4	2:15 p.m.	105	71	88	78
5	7:30 a.m.	125	76	95	76
6	11:30 a.m.	122	70	87	79

Experiment 9

Table 2—Individual Data			
Blood pressure reading	Maximum	Minimum	Δ Blood pressure
Systolic (mm Hg)	130	105	25
Diastolic (mm Hg)	81	62	19
MAP (mm Hg)	102	84	18
Pulse (bpm)	91	70	21

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Answers will vary.
2. Answers will vary. Exercise, smoking, exam-anxiety or other stressors will usually result in blood pressure variation.
3. Most students will find no clear relationship between pulse and blood pressure. While cardiac output is directly related to pulse, blood pressure is determined by a variety of other factors including circulating hormones, which affect resistance in the vascular system.
4. Answers will vary. Using the sample data gathered for this experiment:
Systolic Pressure readings varied by 24%
5. Answers will vary. Using the sample data gathered for this experiment:
Diastolic Pressure readings varied by 31%, MAP readings varied by 21%, Pulse readings varied by 30%
6. There is a difference of 18% between the normal and high categories of systolic blood pressure as defined by the Joint National Committee on Prevention, Detection, Evaluation, and Treatment of High Blood Pressure. For diastolic pressure, the difference between these two categories is 14%.
7. Students should understand from this experiment that a one-time measurement of blood pressure is not sufficient to make a diagnosis of hypertension. Mrs. Jones should either return for multiple measurements of her blood pressure, or she should purchase a blood pressure cuff and collect this data on her own, as is recommended by many physicians.
8. Epinephrine (adrenaline) is the main “fight or flight” hormone, and is released during periods of physical or emotional stress. This leads to an increase in heart rate and blood pressure, preparing the body for a sudden burst of muscular activity.
9.
 - a. (2) Calcium channel blockers dilate arterioles, leading to a decrease in peripheral vascular resistance.
 - b. (3) Diuretics increase fluid output from the kidneys, which lowers blood volume.
 - c. (2) Angiotensin-converting enzyme inhibitors inhibit the formation of the vasoconstrictor Angiotensin II, leading to a reduction in vascular tone and a decrease in peripheral vascular resistance.
 - d. (1) Beta blockers inhibit the sympathetic nervous system, leading to a reduction of heart stimulation, heart rate, and blood pressure.

Heart Rate and Blood Pressure as Vital Signs

Since the earliest days of medicine heart rate has been recognized as a *vital sign*—an indicator of health, disease, excitement, and stress. Medical personnel use the heart rate to provide clues as to the presence of many medical conditions. Reflex changes in heart rate are one of the body's most basic mechanisms for maintaining proper perfusion to the brain and other tissues. Low blood volume caused by bleeding or dehydration results in the heart beating faster as it attempts to maintain adequate blood pressure. Excitement, stress, and anxiety activate the nervous system, which may also speed the heart rate and raise blood pressure.

By the second half of the 19th century a non-invasive method for measuring blood pressure had been invented. Called a *sphygmomanometer*, this instrument is still in use today allowing us to measure this important vital sign.

Blood pressure is a measure of the changing fluid pressure within the circulatory system. It varies from a peak pressure produced by contraction of the left ventricle, to a low pressure, which is maintained by closure of the aortic valve and elastic recoil of the arterial system. The peak pressure is called *systole*, and the pressure that is maintained even while the left ventricle is relaxing is called *diastole*.

Blood pressure and heart rate are interrelated, and both are influenced by the sympathetic and parasympathetic nervous systems. Sympathetic activation raises blood pressure in addition to pulse. After an initial activation of the sympathetic nervous system, the increase in blood pressure stretches nerve fibers in the baroreceptors (see Figure 1). This results in a reflex activation of the parasympathetic nervous system, which, through actions opposite to those of the sympathetic nervous system, helps to restore homeostasis.

In this experiment, you will observe how the heart and circulatory system respond to cold stimulus applied peripherally. Cold will act as a noxious stimulus, activating the “fight or flight” response through the sympathetic nervous system.

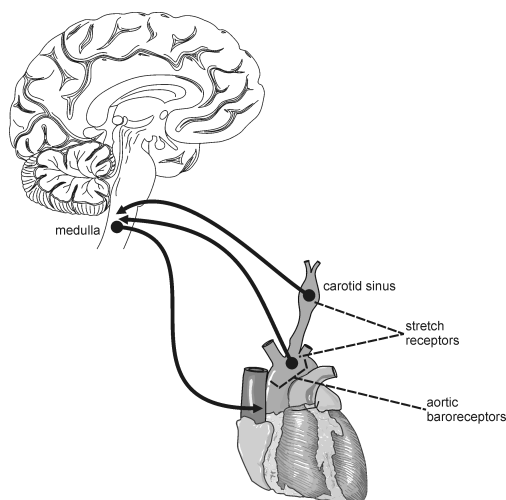


Figure 1

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of heart rate and blood pressure.
- Compare heart rate and blood pressure before and after exposure to cold stimulus.
- Observe an example of sympathetic nervous system activation (“fight or flight response”).

MATERIALS

computer	Vernier Blood Pressure Sensor
Vernier computer interface	ice water bath
Logger Pro	towel
Vernier Hand-Grip Heart Rate Monitor or	saline solution in dropper bottle
Vernier Exercise Heart Rate Monitor	(only for use with Exercise HR Monitor)

PROCEDURE

Part I Baseline Blood Pressure Determination

1. Connect the Blood Pressure Sensor to Channel 1 of the Vernier computer interface. There are two rubber tubes connected to the pressure cuff. One tube has a black Luer-lock connector at the end and the other tube has a bulb pump attached. Connect the Luer-lock connector to the stem on the Blood Pressure Sensor with a gentle half turn if it is not already attached.
2. Open the file “10a Heart Rate and BP” from the *Human Physiology with Vernier* folder.
3. Attach the Blood Pressure cuff firmly around the upper arm, approximately 2 cm above the elbow. The two rubber hoses from the cuff should be positioned over the biceps muscle (brachial artery) and not under the arm (see Figure 2).
4. Have the subject sit quietly in a chair with forearms resting on his/her lap, or on a table surface. *The person having his or her blood pressure measured must remain still during data collection; there should be no movement of the arm or hand during measurements.*
5. Click to begin data collection. Immediately begin to pump until the cuff pressure reaches at least 160 mm Hg. Stop pumping. The cuff will slowly deflate and the pressure will fall. During this time, the systolic, diastolic, and mean arterial pressures and the pulse will be calculated by the software. These values will be displayed on the computer screen. When the cuff pressure drops below 50 mm Hg, the program will stop calculating blood pressure. At this point, you can terminate data collection by clicking . Release the pressure from the cuff, but do not remove it.
6. Enter the systolic, diastolic, and mean arterial pressures in Table 1.



Figure 2

Part II Heart Rate and Blood Pressure Response to Cold

7. Connect the receiver module of the Heart Rate Monitor to Channel 2 of the Vernier computer interface. Open the file “10b Heart Rate and BP” from the *Human Physiology with Vernier* folder.

8. Set an ice water bath on the floor, next to the subject's feet.
9. Prepare to collect data.
 - a. Sit in a chair.
 - b. Prepare to submerge your foot in the ice water bath by removing your shoe and sock.
 - c. Position your foot adjacent to the ice water bath, but do not put it in the bath yet.
10. Set up the Heart Rate Monitor. Follow the directions for your type of Heart Rate Monitor.

Using a Hand-Grip Heart Rate Monitor

- a. The receiver and one of the handles are marked with a white alignment arrow as shown in Figure 3. Locate these two arrows.
- b. Have the subject grasp the handles of the Hand-Grip Heart Rate Monitor so that their fingers are in the reference areas indicated in Figure 4. Hold the handles vertically.
- c. Have someone else hold the receiver near the handles so that the two alignment arrows are pointing in the same direction and are at approximately the same height as shown in Figure 3. **Note:** The receiver must stay within 60 cm of the handles during data collection.

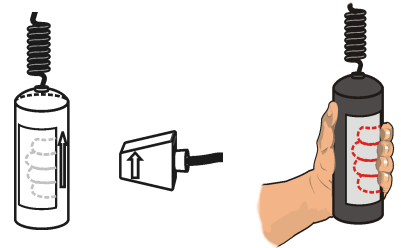


Figure 3

Figure 4

Using an Exercise Heart Rate Monitor

- a. Depending upon your size, select a small- or large-size elastic strap. Secure one of the plastic ends of the elastic strap to the transmitter belt. It is important that the strap provide a snug fit of the transmitter belt.
- b. Wet each of the electrodes (the two textured oval areas on the underside of the transmitter belt) with 3 drops of saline solution.
- c. Secure the transmitter belt against the skin directly over the base of the rib cage (see Figure 5). The POLAR logo on the front of the belt should be centered. Adjust the elastic strap to ensure a tight fit.
- d. Affix the receiver module of the Heart Rate Monitor to a ring stand or table within 80 cm of the transmitter in the Heart Rate Monitor belt.

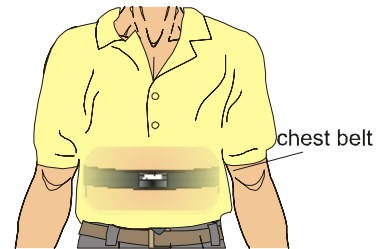



Figure 5

11. With the subject sitting quietly, click to begin data collection.
 - a. At 40 s, instruct the subject to submerge his/her foot in the ice water bath.
 - b. Immediately pump the bulb pump of the Blood Pressure Sensor until the cuff pressure reaches at least 160 mm Hg. Stop pumping.
 - c. At 70 s instruct the subject to remove his/her foot from the ice water bath.
 - d. As data collection continues, the cuff will slowly deflate and the pressure will fall. During this time, the systolic, diastolic, and mean arterial pressures will be calculated by the software. When the cuff pressure drops below 50 mm Hg, the program will stop calculating blood pressure.
 - e. The subject should remain seated and allow data collection to continue for the full 240 s data-collection period.

12. Enter the systolic, diastolic, and mean arterial pressures in Table 2.
13. Click and drag over the area of the heart rate graph where the resting (“baseline”) heart rate is displayed (15–40 s). Click the Statistics button, . The Statistics box will appear with the statistics calculated for the selected region. Record the mean resting heart rate, to the nearest whole number, in Table 3.
14. Move the statistics brackets to highlight the region of the graph beginning at 40 s (when the foot was immersed in the ice water bath) and ending at the first peak (see Figure 6). Record the maximum heart rate value to the nearest whole number in Table 3. In the corresponding Time column record (to the nearest whole number) the Δx value displayed at the lower left corner of the graph.

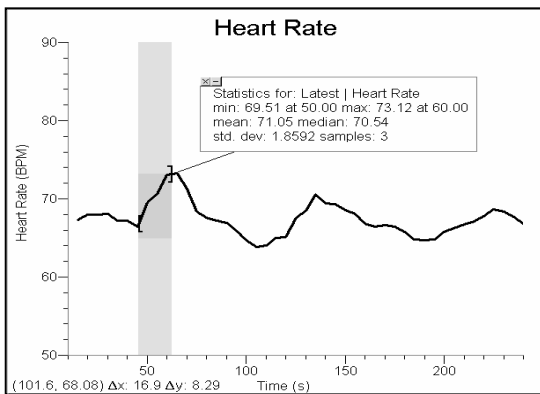


Figure 6

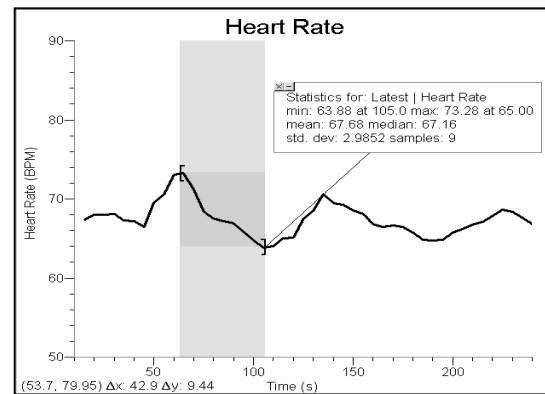


Figure 7

15. Move the Statistics brackets to enclose the region of the graph beginning at the first peak and ending at the lowest point in the valley that follows (see Figure 7). Record the minimum heart rate value to the nearest whole number as the Rebound heart rate in Table 3. Record the Δx value in the corresponding Time column.

DATA

Table 1–Baseline Blood Pressure		
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)

Table 2–Blood Pressure Response to Cold		
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)

Table 3		
Condition	Heart rate (bpm)	Time (s)
Resting heart rate		
Maximum heart rate		
Rebound heart rate		

DATA ANALYSIS

1. Describe the trends that occurred in the systolic pressure, diastolic pressure, mean arterial pressure, and heart rate with cold stimulus. How might these responses be useful in a “fight or flight” situation?

2. As a vital sign, blood pressure is an indicator of general health. A high blood pressure (140/90 or higher) increases the risk of cardiovascular disease and strokes. Collect the systolic and diastolic pressures for the class and calculate the average for each. Rate the class average blood pressure using the follow scale:

Blood Pressure	Category
140/90 or higher	High
120–139/80–89	Pre-hypertension
119/79 or below	Normal

Computer 10

3. How long after immersion did your heart rate reach its maximum value? Explain the physiologic mechanism that led to this change in heart rate.

4. Describe the changes in heart rate that occurred after the maximum value. How can you explain the minimum heart rate value? How would you explain the heart rate variations seen in the remainder of the experiment?

5. How long after the maximum heart rate did it take to arrive at your rebound heart rate? What can you say about the relative speed of physiologic response to a stimulus *vs.* the speed of mechanisms that are designed to maintain homeostasis?

6. If the heart rate is too slow there is inadequate blood pressure to maintain perfusion to the brain. This can lead to loss of consciousness (fainting). Keeping in mind the autonomic nervous system responses that you observed in this experiment, explain the sequence of events that results in a severely frightened person fainting.

TEACHER INFORMATION

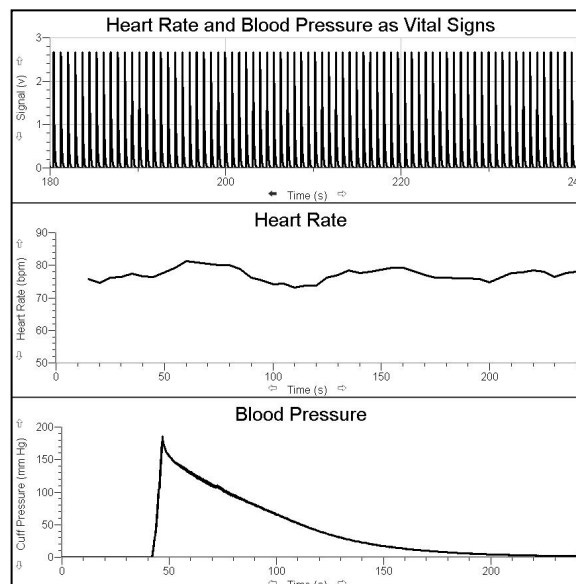
Heart Rate and Blood Pressure as Vital Signs

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. This lab exercise can be done with either of two types of heart rate monitors: the Hand-Grip Heart Rate Monitor and the Exercise Heart Rate Monitor. The Hand-Grip Heart Rate Monitor, which consists of two hand grips and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through the hand grips and relaying the information to the plug-in receiver. The Exercise Heart Rate Monitor, which consists of a chest belt and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through a belt worn around the chest and relaying the information to the plug-in receiver.
3. When using either the Hand-Grip Heart Rate Monitor or the Exercise Heart Rate Monitor with the Logger *Pro* software, there will be a 15 s delay while data are collected before the first point is plotted on the upper graph. Thereafter, a point will be plotted every 5 s.
4. Computer monitors can be a source of electrical interference for both designs of the Heart Rate Monitors. To ensure that this is not a factor in your students' data-collection activities, move the receiver module as far away from the computer monitor as possible.
5. The receiver module of both designs of the Heart Rate Monitors will receive signals from the closest transmitter source. This may result in an apparent doubling of the subjects' heart rates. To avoid confusion or erroneous readings, have test subjects from different lab teams maintain a distance of at least 2 m from each other.
6. When using the Hand-Grip Heart Rate Monitor, it is important that students closely follow the instructions for holding the handles. The hand grips are marked for the right or left hand, each showing where the individuals' fingers and palms should be placed. In addition, the left hand grip and the receiver are both marked with alignment arrows. When collecting data, it is important that the arrow labels on each of these devices be in alignment (see figures in the Student Version of the experiment). Failure to follow these instructions will result in unreliable data.
7. It is important to have good contact between the transmitter belt and the test subject when using the Exercise Heart Rate Monitor. It is very important that the belt fit snugly. Both electrodes should be wetted with either saline solution or contact lens solution. A 5% salt solution works well and can be prepared by adding 5 g per 100 mL of solution. Typical symptoms of inadequate contact with the electrodes are a noisy signal with erroneous peaks, missing heart beat readings, or a flat-line display. If the students receive a flat reading with no heart rate detected, have them move the transmitter and receiver closer together. The range of the transmitter in the chest belt is 60–80 cm.

Experiment 10

8. The pulse value displayed in the meter on the screen will not agree with the heart rate measured by the Heart Rate Monitor. This is because the pulse calculated by the blood pressure software is calculated over the entire time of data collection, as opposed to the continuously-averaged reading displayed by the heart rate software.
9. Blood pressure may vary from reading to reading for a variety of reasons. It is normal for blood pressure to vary with the time of day or to be elevated in times of stress. For this reason, single blood pressure readings are not adequate for a diagnosis of abnormal blood pressure. It is normal for health care professionals to repeat blood pressure measurements over a period of several weeks before diagnosing high blood pressure.
10. Alert the students to be sure to secure the Blood Pressure cuff around the subjects' arms very snugly. A loosely secured cuff will return erratic data.
11. If the blood pressure tracing does not reach 50 mm Hg by the time data collection ends, adjust the exhaust rate of the pressure release valve according to the directions listed under "Adjusting the Pressure Release Valve" in the Blood Pressure Sensor booklet that accompanies the sensor. This sensor booklet is also available on the Vernier web site, www.vernier.com.
12. Individual variability will be more pronounced in this experiment than in many of the other experiments in this book. This is because the balance between the sympathetic and parasympathetic nervous systems varies from individual to individual and, within the same individual, at different times of the day. It might be instructive to ask students with unexpected results to offer explanations for these results (i.e. are they taking medication, have excessive amounts of caffeine been ingested, etc.).

SAMPLE DATA



Sample data

Table 1–Baseline Blood Pressure		
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)
132	80	97

Table 2–Blood Pressure Response to Cold		
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)
146	95	117

Table 3		
Condition	Heart rate (bpm)	Time (s)
Resting heart rate	76	
Maximum heart rate	81	18
Rebound heart rate	73	51

ANSWERS TO DATA ANALYSIS QUESTIONS

1. Cold exposure caused discomfort or mild pain, which led to activation of the sympathetic nervous system. Sympathetic nervous system activation increases heart rate and peripheral vascular resistance (by constricting arterioles), which leads to an elevation of systolic, diastolic and mean arterial blood pressures.
2. A discussion of the general health and well-being of the students in the class (as relates to blood pressure) would be an appropriate conclusion to this experiment.
3. Answers will vary, but should be in the range of 30–40 s. Factors such as weight, regular exercise, health, medications, and tolerance to cold, may affect individual response. Exposure to cold causes severe enough discomfort to activate the sympathetic nervous system, which acts to increase the heart rate.
4. Following the maximum value, most students will report a slowing of the heart to a rate below the resting heart rate. The initial increase in heart rate (and blood pressure) activates baroreceptors which in turn cause parasympathetic activation and a slowing of the heart rate. In this case, the slowing briefly “overshoots” the resting heart rate. Some students may even report a “see-sawing” effect, where the heart rate increases and decreases in an almost sinusoidal pattern while homeostasis is being restored.

Experiment 10

5. Answers will vary, but should be in the range of 30–50 s. The initial physiologic response to stimuli is more rapid than compensatory responses. There are obvious benefits to a rapid response to danger. A more measured return to homeostasis lessens the see-sawing effect as the sympathetic and parasympathetic systems are achieving new balance.
6. When a person is severely frightened, there is an overwhelming sympathetic response. This increases heart rate and blood pressure dramatically. The compensatory parasympathetic response is likewise much greater than normal, causing marked slowing of the heart rate and a drop in blood pressure and circulation to the brain. Sudden activation of the parasympathetic nervous system has the opposite effect of sympathetic activation. The heart rate may slow significantly. Dilatation of peripheral blood vessels leads to pooling of blood in the legs and lack of blood return to the heart. These two factors together cause a precipitous fall in blood pressure, lack of adequate blood supply to the brain, and fainting.

Heart Rate, Blood Pressure, and Exercise

The adaptability of the heart can be observed during exercise, when the metabolic activity of skeletal muscles increases. The cardiovascular system, consisting of the heart and blood vessels, responds to exercise with an increase in heart rate and strength of contraction with each beat, resulting in a higher *cardiac output* (*cardiac output* = quantity of blood pumped through the heart per unit of time) and blood pressure. Positive pressure is created by forceful contraction of the left ventricle of the heart, measured as *systole*. It is maintained during relaxation of the ventricle by closure of the aortic valve and recoil of arteries, measured as *diastole* (see Figure 1).

Mean arterial pressure (MAP) is a useful measure of the adequacy of tissue perfusion, and is not a simple average of systolic and diastolic blood pressures. This is because diastole continues for twice as long as systole. MAP can be reasonably approximated using the equation:

$$\frac{(\text{systole} + 2(\text{diastole}))}{3} = \text{MAP}$$

The mean arterial pressure is directly proportional to *cardiac output* and inversely proportional to *total peripheral resistance*, where:

Cardiac output is the amount of blood pumped out of the heart with each beat (called the *stroke volume*), multiplied by the number of beats per minute.

Total peripheral resistance depends on blood viscosity, length of the arterial system, diameter and elasticity of the blood vessels, and the pressure entering versus leaving the arterial system (systolic pressure minus the pressure in the venous system).

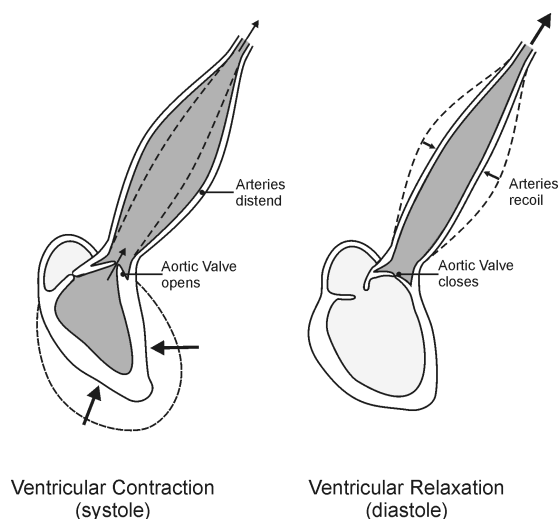


Figure 1

Computer 11

In this experiment, you will observe how the heart responds to the increased metabolic demand of muscles during exercise. You will compare heart rate and blood pressure readings taken before and after exercise and measure changes in systolic, diastolic and mean arterial pressures. You will also consider the effect that exercise has on cardiac output and peripheral vascular resistance.

Important: Do not attempt this experiment if physical exertion will aggravate a health problem. Inform your instructor of any possible health problems that might be exacerbated if you participate in this exercise.

OBJECTIVES

In this experiment, you will

- Obtain graphic representation of heart rate and blood pressure.
- Determine the effect of exercise on heart rate, and systolic, diastolic and mean arterial pressures.
- Use blood pressure readings and pulse to infer changes in cardiac output and peripheral vascular resistance with exercise.
- Correlate the fitness level of individuals with amount of daily exercise.

MATERIALS

computer
Vernier computer interface
Logger Pro
Vernier Blood Pressure Sensor

Vernier Hand-Grip Heart Rate Monitor **or**
Vernier Exercise Heart Rate Monitor
saline solution in dropper bottle
(only for use with the Exercise HR Monitor)

PROCEDURE

Part I Baseline Blood Pressure

1. Connect the Blood Pressure Sensor to Channel 1 of the Vernier computer interface. Open the file "11a Heart Rate BP Exercise" from the *Human Physiology with Vernier* folder.
2. Attach the Blood Pressure Sensor to the blood pressure cuff if it is not already attached. There are two rubber tubes connected to the cuff. One tube has a black Luer-lock connector at the end and the other tube has a bulb pump attached. Connect the Luer-lock connector to the stem on the Blood pressure Sensor with a gentle half turn.
3. Attach the Blood Pressure cuff to the upper arm, approximately 2 cm above the elbow. The two rubber hoses from the cuff should be positioned over the biceps muscle (brachial artery) and not under the arm (see Figure 2).
4. The subject should sit quietly in a chair and avoid moving his or her arm or hand during blood pressure measurements.

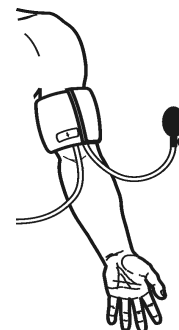


Figure 2

- Click **Collect** to begin data collection. Immediately begin to pump until the cuff pressure reaches at least 160 mm Hg. Stop pumping.
- During this time the systolic, diastolic, and mean arterial pressures will be calculated by the software. These values will be displayed on the computer screen. When the blood pressure readings have stabilized (after the pressure drops to 50 mm Hg), the program will stop calculating blood pressure. At this point, you can terminate data collection by clicking **Stop**. Release the pressure from the cuff, but do not remove it.
- Enter the pulse and the systolic, diastolic, and mean arterial pressures in Table 1.

Part II Heart Rate and Blood Pressure after Exercise

- Connect the receiver module of the Heart Rate Monitor to Channel 2 of the Vernier computer interface. Open the file “11b Heart Rate BP Exercise” from the *Human Physiology with Vernier* folder.
- Set up the Heart Rate Monitor. Follow the directions for your type of Heart Rate Monitor.

Using a Hand-Grip Heart Rate Monitor

- The receiver and one of the handles are marked with a white alignment arrow as shown in Figure 3. Locate these two arrows.
- Have the subject grasp the handles of the Hand-Grip Heart Rate Monitor so that their fingers are in the reference areas indicated in Figure 4. Hold the handles vertically.
- Have someone else hold the receiver near the handles so that the two alignment arrows are pointing in the same direction and are at approximately the same height as shown in Figure 3. **Note:** The receiver must stay within 60 cm of the handles during data collection.

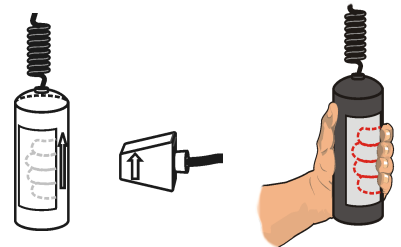


Figure 3

Figure 4

Using an Exercise Heart Rate Monitor

- Depending upon your size, select a small or large size elastic strap. Secure one of the plastic ends of the elastic strap to the transmitter belt. It is important that the strap provide a snug fit of the transmitter belt.
- Wet each of the electrodes (the two textured oval areas on the underside of the transmitter belt) with 3 drops of saline solution.
- Secure the transmitter belt against the skin directly over the base of the rib cage (see Figure 5). The POLAR logo on the front of the belt should be centered. Adjust the elastic strap to ensure a tight fit.
- Take the receiver module of the Heart Rate Monitor in your right hand. Remember that the receiver must be within 80 cm of the transmitter in the Heart Rate Monitor belt.

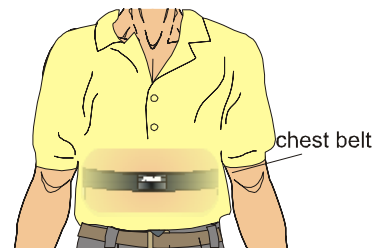






Figure 5

- Stand quietly facing your table or lab bench.

Computer 11

11. To determine that everything is set up correctly, click  to begin monitoring heart rate. Note that there may be up to a 30 second delay before data are seen. The readings should be within the normal range of the individual, usually between 55 and 80 beats per minute. Click  when you have determined that the equipment is operating properly, and proceed to Step 12.
12. Click  to begin data collection. If the baseline appears stable, begin to run in place at 40 s. Continue data collection while running in place for the next 2 minutes.
13. At approximately 160 s, stop running. Stand still. Do not move during blood pressure measurement.
14. Immediately begin to pump the blood pressure cuff until the cuff pressure reaches at least 160 mm Hg. Stop pumping.
15. During this time the systolic, diastolic, and mean arterial pressures will be calculated by the software. These values will be displayed on the computer screen. When the blood pressure readings have stabilized (after the pressure drops to 50 mm Hg), the program will stop calculating blood pressure. At this point, release the pressure from the cuff.
16. Enter the systolic, diastolic, and mean arterial pressures in Table 2.
17. The subject should continue to stand in place while his/her heart rate slows toward its resting pre-exercise value. Data will be collected for 280 s.
18. Click and drag over the area of the graph where the resting heart rate is displayed (from 0 to approximately 40 s). This will highlight the region of interest.
19. Click the Statistics button, . Record the mean resting heart rate in Table 3.
20. Drag the right hand bracket to the right edge of the graph, until all the data points are highlighted. The values in the Statistics box will be adjusted based on the data within the brackets. Record the maximum heart rate in Table 2 (under “pulse”) and in Table 3.
21. Move the statistics brackets to highlight the area of the graph beginning with the maximum heart rate and ending with the first data point that matches the initial baseline value (or the last point graphed, if baseline is not achieved). Record the Δx value displayed at the lower left corner of the graph as the recovery time in Table 3.

DATA

Table 1–Baseline Blood Pressure			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (bpm)

Table 2–Blood Pressure After Exercise			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (bpm)

Table 3–Heart Rate	
Condition	
Resting heart rate (bpm)	
Maximum heart rate (bpm)	
Recovery time (s)	

DATA ANALYSIS

1. Describe the trends that occurred in the systolic pressure, diastolic pressure, mean arterial pressure and pulse with exercise. Assume that the stroke volume increased from 75 mL/beat to 100 mL/beat. Use this information and the change in pulse with exercise to calculate the change in *cardiac output* (stroke volume \times heart rate) that occurred per minute.

2. *Pulse pressure* is the difference between systolic pressure (peak pressure during active contraction of the ventricles) and diastolic pressure (the pressure that is maintained even while the left ventricle is relaxing). Describe the change in pulse pressure seen with exercise. Which component of the blood pressure is most responsible for this change?

Computer 11

3. A change in pulse pressure can be seen in a variety of medical conditions. What would you expect to happen to the pulse pressure in the following examples?
 - (a) In atherosclerosis there is a hardening of the arterial walls.

 - (b) A damaged aortic valve does not seal properly and allows blood to flow back into the ventricle during diastole.

4. Normal resting heart rates range from 55–100 beats per minute. What was your/the subject's resting heart rate? How much did your/the subject's heart rate increase above resting rate with exercise? What percent increase was this?

5. How does your/the subject's maximum heart rate compare with other students in your group/class? Is this what you expected?

6. Recovery time has been shown to correlate with degree of physical fitness. How does your/the subject's recovery rate compare to that of your classmates? Is this what you expected?

7. Congestive heart failure is a condition in which the strength of contraction with each beat may be significantly reduced. For example, the ventricle may pump only half the usual volume of blood with each beat. Would you expect a person with congestive heart failure to have a faster or slower heart rate at rest? With exercise?

8. Medications are available which can slow the heart or speed it up. If a patient complains of feeling poorly and has a heart rate of 120 beats per minute, should you administer a medicine to slow the rate?

EXTENSIONS

1. Exercise for varying lengths of time and use page 2 of the experiment file to compare the heart rate and blood pressure readings taken after each exercise period.
2. Measure heart rate and blood pressure 5, 10, and 15 minutes after stopping exercise, and use Page 2 of the experiment file to compare readings.
3. Measure heart rate and blood pressure while lifting a weight with the opposite arm.
4. Design an anonymous survey to be taken by each member of your class. In the survey ask questions that you think might influence the test results (examples might include: gender, age, exercise regimen, coffee drinking within 2 hours of experiment, smoking status). Compare and contrast resting heart rates and recovery rates after exercise within and among these groups.

TEACHER INFORMATION

Heart Rate, Blood Pressure, and Exercise

The privacy of your students needs to be respected in this exercise. We suggest that the test subjects be completely voluntary. No student should be required to perform the test if he/she is uncomfortable sharing data with others. In cases where questions refer to personal data and comparison to class data, this should be collected anonymously and compiled by the teacher.

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Since blood pressure will be elevated after exercise, the cuff should be inflated to 170–190 mm Hg (rather than to 160 mm Hg) in Part II of the experiment.
3. The pulse displayed on the screen is an average pulse taken over the time during which the blood pressure is measured. Thus, it is accurate at rest, but cannot be used to determine the pulse rate immediately after exercise. To do this, the students are instructed to feel for the radial pulse and count the beats over 20 s in Part II of the experiment.
4. This lab exercise can be done with either of two types of heart rate monitors: the Hand-Grip Heart Rate Monitor and the Exercise Heart Rate Monitor. The Hand-Grip Heart Rate Monitor, which consists of two hand grips and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through the hand grips and relaying the information to the plug-in receiver. The Exercise Heart Rate Monitor, which consists of a chest belt and a plug-in receiver, measures heart rate by detecting the electrical impulses of the heart through a belt worn around the chest and relaying the information to the plug-in receiver.
5. When using either the Hand-Grip Heart Rate Monitor or the Exercise Heart Rate Monitor, there will be a 15 s delay while data are collected before the first point is plotted on the upper graph. Thereafter, a point will be plotted every 5 s.
6. Computer monitors can be a source of electrical interference for both designs of the Heart Rate Monitors. To ensure that this is not a factor in your students' data-collection activities, move the receiver module as far away from the computer monitor as possible.
7. The receiver module of both designs of the Heart Rate Monitors will receive signals from the closest transmitter source. This may result in an apparent doubling of the subjects' heart rates. To avoid confusion or erroneous readings, have test subjects from different lab teams maintain a distance of at least 2 m from each other.
8. When using the Hand-Grip Heart Rate Monitor, it is important that students closely follow the instructions for holding the handles. The hand grips are marked for the right or left hand, each showing where the individuals' fingers and palms should be placed. In addition, the left hand grip and the receiver are both marked with alignment arrows. When collecting data, it is important that the arrow labels on each of these devices be in alignment (see figures in the Student Version of the experiment). Failure to follow these instructions will result in unreliable data.

Experiment 11

9. It is important to have good contact between the transmitter belt and the test subject when using the Exercise Heart Rate Monitor. It is very important that the belt fit snugly. Both electrodes should be wetted with either saline solution or contact lens solution. A 5% salt solution works well and can be prepared by adding 5 g per 100 mL of solution. Typical symptoms of inadequate contact with the electrodes are a noisy signal with erroneous peaks, missing heart beat readings, or a flat-line display. If the students receive a flat reading with no heart rate detected, have them move the transmitter and receiver closer together. The range of the transmitter in the chest belt is 60–80 cm.

SAMPLE DATA

Table 1–Baseline Blood Pressure			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (bpm)
108	67	84	73

Table 2–Blood Pressure After Exercise			
Systolic pressure (mm Hg)	Diastolic pressure (mm Hg)	Mean arterial pressure (mm Hg)	Pulse (bpm)
142	86	91	140

Table 3–Heart Rate	
Condition	
Resting heart rate (bpm)	89
Maximum heart rate (bpm)	140
Recovery time (s)	124

ANSWERS TO DATA ANALYSIS QUESTIONS

1. Most students should report that the systolic pressure increased with exercise. Diastolic pressure may increase, decrease or remain the same. Mean arterial pressure should be higher as should the pulse.

$$\text{Resting cardiac output} = (75 \text{ mL/beat}) (95 \text{ beats/min}) = 7125 \text{ mL/min} = 7.1 \text{ L/min}$$

$$\text{Cardiac output immediately after exercise} = (100 \text{ mL/beat}) (112 \text{ beats/min}) = 11200 \text{ mL/min} = 11 \text{ L/min}$$

2. Students should see an increase in pulse pressure because of a rise in systolic pressure. This is due to the increase in cardiac output resulting from exercise.
3. (a) Pulse pressure depends on *distensibility* of arterial walls, as well as the cardiac output. In atherosclerosis there is a hardening of arterial walls, making them less distensible. This causes a higher systolic pressure and a higher pulse pressure.

(b) Diastolic pressure is maintained in part by closure of the aortic valve. When this valve fails to close completely, there is a backflow of blood into the left ventricle, reducing the diastolic pressure. In fact, the pulse pressure may be increased dramatically because of an accompanying increase in systolic pressure generated by the extra volume of blood pumped with each contraction of the left ventricle (blood from the left atrium plus regurgitant blood from the aorta).
4. Answers will vary. Factors such as weight, regular exercise, health, medications, etc., may play a part in determining the resting and exercise heart rate.
5. Answers will vary. Factors such as effort, weight, regular exercise, health, medications, etc., may play a part in determining the maximum heart rate achieved by students in this experiment.
6. Answers will vary. Factors such as weight, regular exercise, health, medications, etc., may play a part in determining the resting and exercise heart rate.
7. A person with congestive heart failure will tend to have a more rapid heart rate at rest and with exercise. If less blood is pumped during each heart beat, the heart will attempt to compensate by beating more rapidly.
8. Students should be able to answer “no” to this question, based on their understanding of the heart rate as a homeostatic mechanism. The heart may beat rapidly to compensate for other problems, such as blood loss and low blood pressure. In this case, slowing the heart with medication may lead to a precipitous and fatal fall in blood pressure. One should not attempt to manipulate the heart rate before ascertaining the underlying cause of the abnormality.

Analyzing the Heart with EKG

An electrocardiogram (ECG or EKG) is a graphical recording of the electrical events occurring within the heart. In a healthy heart there is a natural pacemaker in the right atrium (the *sinoatrial node*) which initiates an electrical sequence. This impulse then passes down natural conduction pathways between the atria to the atrioventricular node and from there to both ventricles. The natural conduction pathways facilitate orderly spread of the impulse and coordinated contraction of first the atria and then the ventricles. The electrical journey creates unique deflections in the EKG that tell a story about heart function and health (Figure 1). Even more information is obtained by looking at the story from different angles, which is accomplished by placing electrodes in various positions on the chest and extremities. A positive deflection in an EKG tracing represents electrical activity moving toward the active lead (the green lead in this experiment).

Five components of a single beat are traditionally recognized and labeled P, Q, R, S, and T. The P wave represents the start of the electrical journey as the impulse spreads from the sinoatrial node downward from the atria through the atrioventricular node and to the ventricles. Ventricular activation is represented by the QRS complex. The T wave results from ventricular repolarization, which is a recovery of the ventricular muscle tissue to its resting state. By looking at several beats you can also calculate the rate for each component.

Doctors and other trained personnel can look at an EKG tracing and see evidence for disorders of the heart such as abnormal slowing, speeding, irregular rhythms, injury to muscle tissue (*angina*), and death of muscle tissue (*myocardial infarction*). The length of an interval indicates whether an impulse is following its normal pathway. A long interval reveals that an impulse has been slowed or has taken a longer route. A short interval reflects an impulse which followed a shorter route. If a complex is absent, the electrical impulse did not rise normally, or was blocked at that part of the heart. Lack of normal depolarization of the atria leads to an absent P wave. An absent QRS complex after a normal P wave indicates the electrical impulse was blocked before it reached the ventricles. Abnormally shaped complexes result from abnormal spread of the impulse through the muscle tissue, such as in myocardial infarction where the impulse cannot follow its normal pathway because of tissue death or injury. Electrical patterns may also be changed by metabolic abnormalities and by various medicines.

In this experiment, you will use the EKG sensor to make a five second graphical recording of your heart's electrical activity, and then switch the red and green leads to simulate the change in electrical activity that can occur with a myocardial infarction (heart attack). You will identify the different components of the waveforms and use them to determine your heart rate. You will also determine the direction of electrical activity for the QRS complex.

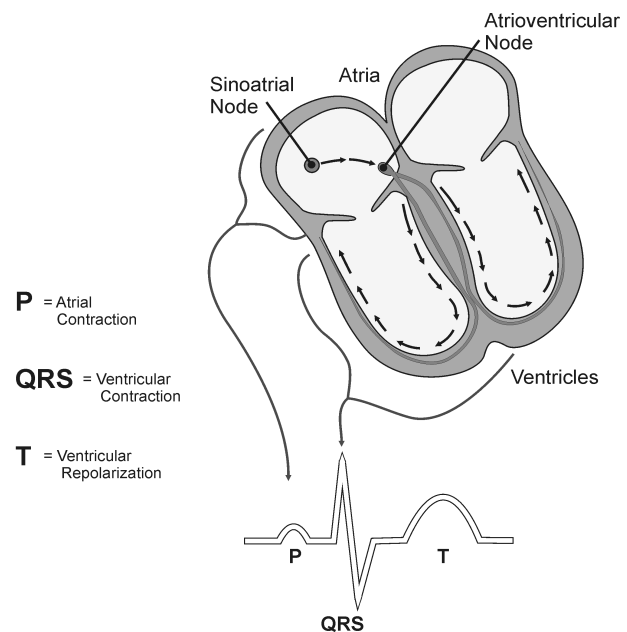


Figure 1

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of the electrical activity of the heart over a period of time.
- Learn to recognize the different wave forms seen in an EKG, and associate these wave forms with activity of the heart.
- Determine the heart rate by determining the rate of individual wave forms in the EKG.
- Compare wave forms generated by alternate EKG lead placements.

MATERIALS

computer
Vernier computer interface
Logger Pro

Vernier EKG Sensor
electrode tabs

PROCEDURE

Part I Standard limb lead EKG

1. Connect the EKG Sensor to the Vernier computer interface. Open the file “12 Analyzing Heart EKG” from the *Human Physiology with Vernier* folder.
2. Attach three electrode tabs to your arms, as shown in Figure 2. Place a single patch on the inside of the right wrist, on the inside of the right upper forearm (distal to the elbow), and on the inside of the left upper forearm (distal to elbow).
3. Connect the EKG clips to the electrode tabs as shown in Figure 2. Sit in a relaxed position in a chair, with your forearms resting on your legs or on the arms of the chair. When you are properly positioned, have someone click **Collect** to begin data collection.
4. Once data collection is finished, click and drag to highlight each interval listed in Table 1. Use Figure 3 as your guide when determining these intervals. Enter the Δx value of each highlighted area to the nearest 0.01 s in Table 1. This value can be found in the lower left corner of the graph.
5. Calculate the heart rate in beats/min using the EKG data. Record the heart rate to the nearest whole number in Table 1.
6. Store this run by choosing Store Latest Run from the Experiment menu.

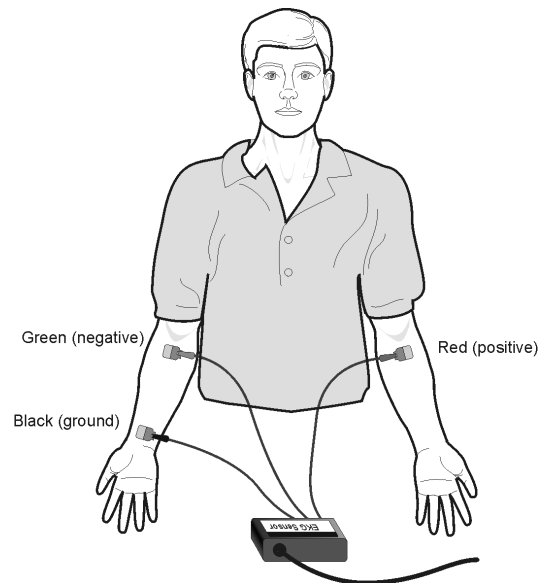


Figure 2

Part II Alternate limb lead EKG

7. Exchange the red and green EKG clips so that the green clip is now attached to the electrode tab on the left arm and the red clip is on the right arm. Sit in a relaxed position in a chair, with your forearms resting on your legs or on the arms of the chair. When you are properly positioned, have someone click **Collect** to begin data collection.

8. Print or sketch the tracing for alternate limb lead placement only.

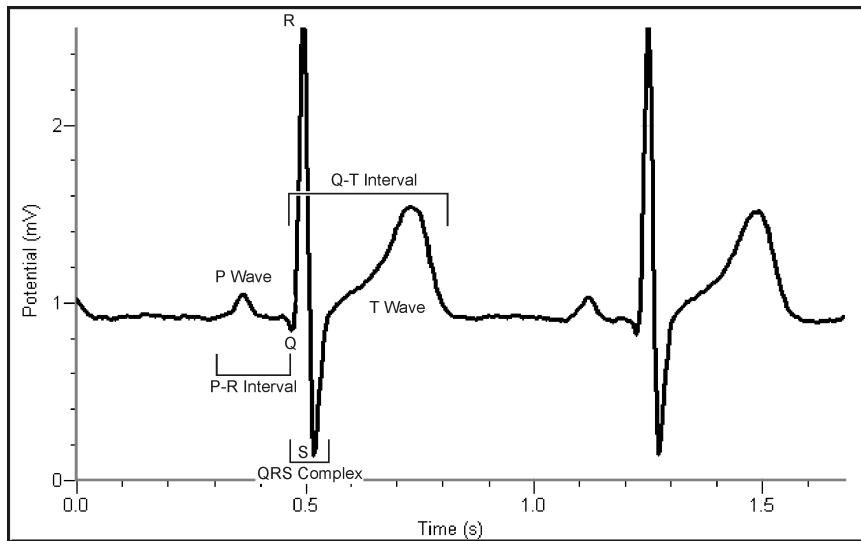


Figure 3

- **P-R interval:** time from the beginning of P wave to the start of the QRS complex
- **QRS complex:** time from Q deflection to S deflection
- **Q-T interval:** time from Q deflection to the end of the T

DATA

Table 1	
Interval	Time (s)
P-R	
QRS	
Q-T	
R-R	

Heart Rate (bpm)	
------------------	--

Table 2	
Standard Resting Electrocardiogram Interval Times	
P-R interval	0.12 to 0.20 s
QRS interval	less than 0.12 s
Q-T interval	0.30 to 0.40 s

DATA ANALYSIS

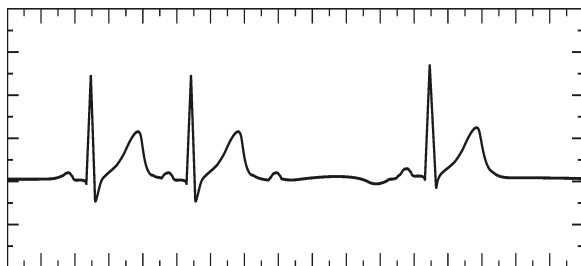
1. Remember that a positive deflection indicates electrical activity moving toward the green EKG lead. Examine the two major deflections of a single QRS complex (R wave and S wave) in your EKG tracing from Part I of this experiment. According to this data, does ventricular depolarization proceed from right to left or left to right? How does your tracing from Part II confirm your answer?

2. Health-care professionals ask the following questions when interpreting an EKG:

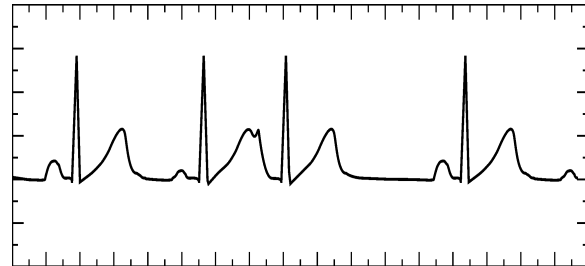
- Can all components be identified in each beat?
- Are the intervals between each component and each complex consistent?
- Are there clear abnormalities of any of the wave components?

Using these questions as guides, analyze each of the following three-beat EKG tracings and record your conclusions in Table 3 (indicate presence or absence of the P wave, and whether other intervals and/or shapes are normal or abnormal). The first analysis (a) is done for you.

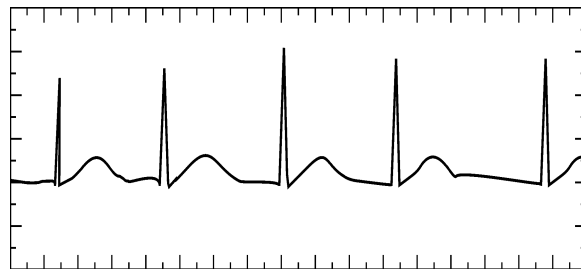
a.



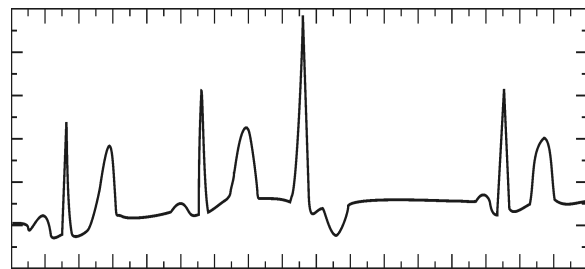
b.



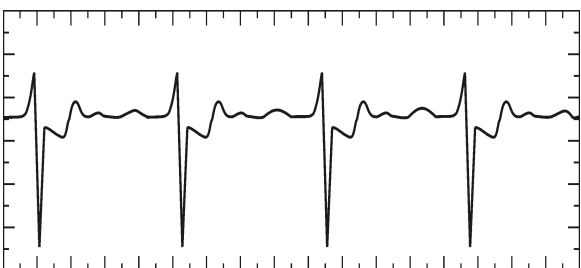
c.



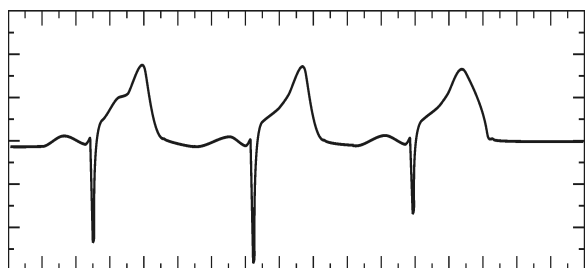
d.



e.



f.



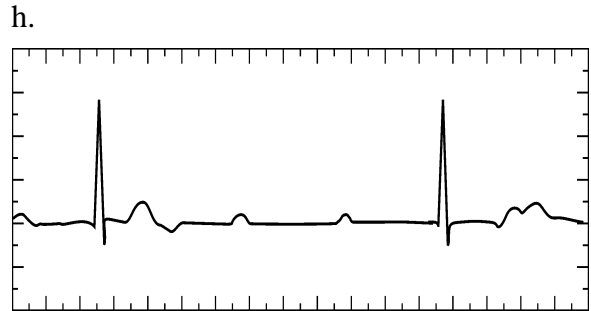
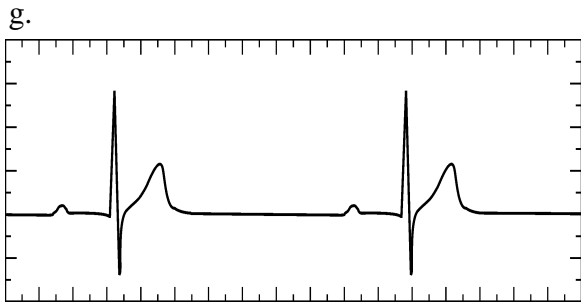


Table 3

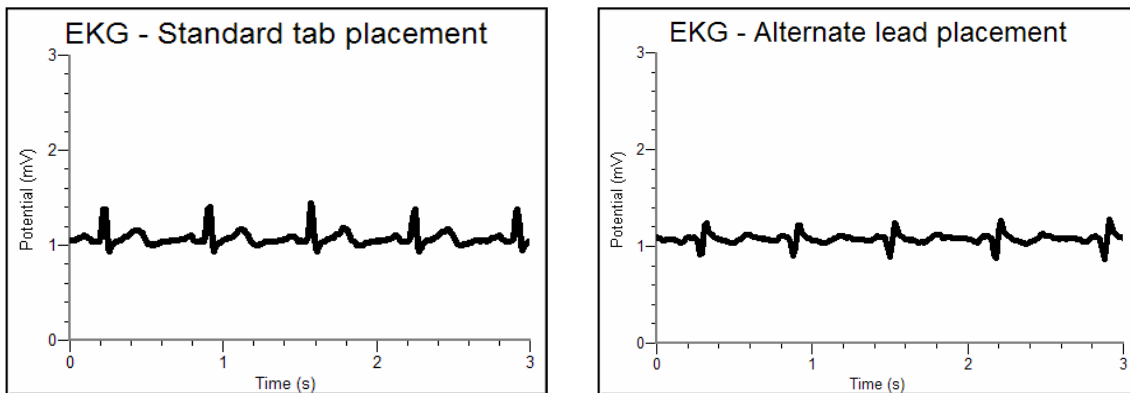
ECG	Beat	P Wave		PR Interval		QRS Interval		QRS Shape		T Wave Shape	
		Pres.	Abs.	Nml.	Abs./Abn.	Nml.	Abs./Abn.	Nml.	Abn.	Nml.	Abs./Abn.
a	1	X		X		X		X		X	
	2	X		X		X		X		X	
	3	X			X		X		X		X
b	1										
	2										
	3										
c	1										
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e	1										
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f	1										
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	3										
g	1										
	2										
	3										
h	1										
	2										
	3										

TEACHER INFORMATION

Analyzing the Heart with EKG

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. If the test subject's skin is oily or a lotion has been applied, scrub the area of skin where the electrode is to be attached with soap and water prior to applying the electrode.
3. Students will find that not all EKGs look alike. They should not be alarmed. Individual differences can be caused by a variety of benign conditions. Two common benign conditions that may be seen in class are premature atrial beats (as in Question 1b) and premature ventricular beats (as in Question 1d). Waveform amplitudes and shapes may also vary from student to student.
4. Table 3 is provided to allow students to methodically analyze each EKG tracing. However, in practice, reading EKGs is a visual experience. Encourage students to review the EKG tracings and be able to describe unique attributes of each tracing.
5. A cleaner signal may be obtained by disconnecting LabQuest from AC power.

SAMPLE DATA



Sample EKG data with standard and alternate limb lead placements

Table 1	
Interval	Time (s)
P-R	0.17
QRS	0.11
Q-T	0.41
R-R	0.67
Heart Rate (bpm)	90

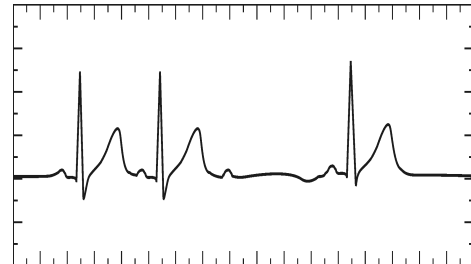
ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. The initial deflection of the QRS complex in Part I of the experiment is positive, with the next deflection negative. This indicates that the initial depolarization then proceeds to the left, as shown by the negative S wave.

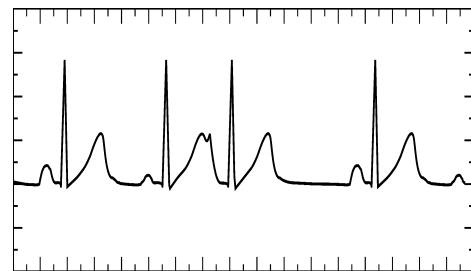
In Part II, the leads are switched. The initial QRS wave is negative because its depolarization is moving toward the right side of the body, this time *away* from the green lead. This confirms the finding in Part I of the experiment.

2. Following are details about each of the tracings presented in the Questions section of the student version of this lab exercise:

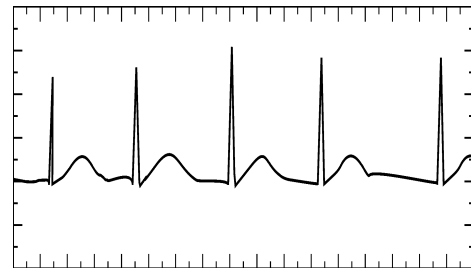
- a. Second degree heart block: This is a term given to the sudden dropping of a ventricular beat (QRS complex). A normal P wave is generated, but at periodic intervals the impulse fails to reach the ventricles. This can be caused by scarring in conduction pathways or the acute injury which occurs in myocardial infarction. If this condition persists implantation of an artificial pacemaker may be considered.



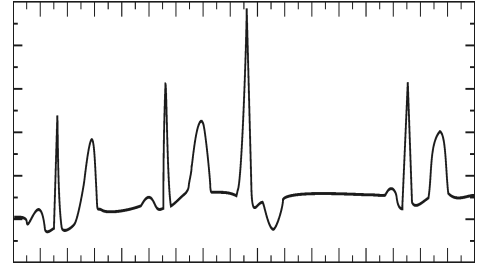
- b. Premature atrial contraction: The P wave of the third complex occurs earlier than expected and has an abnormal shape. This beat arose somewhere below the sinoatrial node and had a shorter distance to travel to the ventricles. This condition is benign and can occur more frequently in the presence of stimulants such as caffeine.



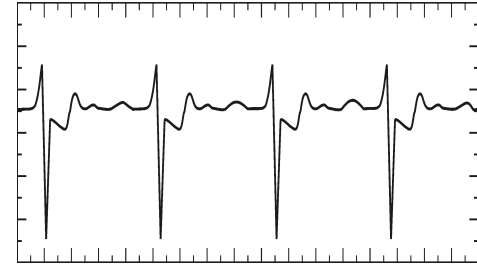
- c. Atrial fibrillation: There are no P waves seen in this tracing, indicating that the sinoatrial node is not generating electrical impulses. Instead, electrical impulses are flowing in random directions all over the atria and random impulses make their way through the atrial ventricular node down to the ventricles. This results in an irregular heartbeat, but the ventricles are contracting normally. This is a common condition at all ages but is even more common in the elderly. Since electrical activity in the atria is flowing in random directions, the atria are not contracting. This reduces the efficiency of the heart because blood must flow passively from the atria to the ventricles. The lack of contraction of the atria leads to a stagnation of blood along its walls and a higher incidence of blood clots in certain individuals. Aspirin or warfarin, a more potent anticoagulant, are used to thin the blood in many of these patients. Others may be treated by electrical cardioversion, where a shock is administered to the heart causing depolarization of all of the muscle, allowing the sinoatrial node to resume its role as a natural pacemaker.



- d. Premature ventricular contraction (PVC): The wide QRS complex of beat 3 occurred as the result of an impulse arising in the ventricles. Since the impulse began in the ventricle there is no P wave, and the QRS complex is lengthened because the electricity is not following the usual conduction pathways. This condition is most often benign and may be observed in some of the students. As in premature atrial contractions, these may occur more frequently in the presence of stimulants.



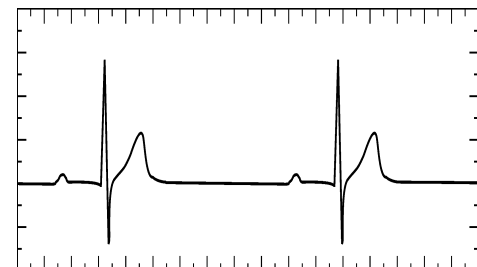
- e. Ischemia: In this tracing, the T wave is inverted as a result of abnormal repolarization of ventricular tissue. This most commonly occurs when heart muscle is not being properly supplied with blood and hence oxygen. This is the pattern often seen in patients with angina (chest pain), where partial blockages in the coronary arteries result in temporary muscle pain. After treatment with nitroglycerin or other medication the T wave will normalize.



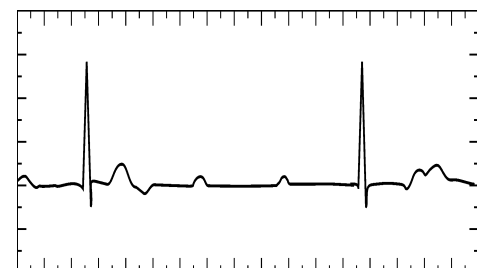
- f. Myocardial infarction: This condition, commonly known as a heart attack, results in muscle tissue damage which is severe enough to block depolarization in that tissue. Electrical forces follow an alternate pathway and can cause a deep Q wave and a grossly elevated and abnormal T wave.



- g. First degree heart block: The prolonged P-R interval is caused by abnormalities in the pathway from the sinoatrial node to the ventricles. This commonly occurs in the vicinity of the atrioventricular node. This condition is usually benign.



- h. Complete heart block: P waves and QRS complexes appear at normal intervals, but they are not attached to each other. In this condition the electrical impulse is completely blocked before it reaches the ventricles. The atrioventricular node takes over as the heart's pacemaker, but its intrinsic rate is slower than that of the sinoatrial node. As with second degree heart block, this can be caused by scarring in conduction pathways or the acute injury which occurs in myocardial infarction. In this case, a pacemaker is required.



Experiment 12

Table 3											
ECG	Beat	P Wave		PR Interval		QRS Interval		QRS Shape		T Wave Shape	
		Pres.	Abs.	Nml.	Abs./Abn.	Nml.	Abs./Abn.	Nml.	Abn.	Nml.	Abs./Abn.
a	1	X		X		X		X		X	
	2	X		X		X		X		X	
	3	X			X		X		X		X
b	1	X		X		X		X		X	
	2	X		X		X		X			X
	3	X			X	X		X		X	
c	1		X		X	X		X		X	
	2		X		X	X		X		X	
	3		X		X	X		X		X	
d	1	X		X		X		X		X	
	2	X		X		X		X		X	
	3		X		X		X		X		X
e	1	X		X		X		X			X
	2	X		X		X		X			X
	3	X		X		X		X			X
f	1	X		X			X		X		X
	2	X		X			X		X		X
	3	X		X			X		X		X
g	1	X			X	X		X		X	
	2	X			X	X		X		X	
	3	X			X	X		X		X	
h	1	X			X	X		X			X
	2	X			X	X		X			X
	3	X			X	X		X			X

Introduction to EMG

An electromyogram, or EMG, is a graphical recording of electrical activity within muscles. Activation of muscles by nerves results in changes in ion flow across cell membranes, which generates electrical activity. This can be measured using surface electrodes placed on the skin over the muscle of interest.

Electrical activity correlates with strength of muscle contraction, and is dependent on the quantity of nerve impulses which are sent to the muscle. This is easily visible in large muscles such as the biceps muscle in the arm and the quadriceps muscle in the leg, but can also be demonstrated in smaller, less visible muscles, such as the masseter muscle in the jaw.

Temporomandibular Disorders, TMD, result from problems in the temporomandibular (jaw) joint, and affect jaw action (chewing of food, talking, playing the trumpet). In this experiment, you will examine the electrical activity generated by chewing and see how food texture influences the strength of contraction in the masseter muscle of the jaw (see Figure 1).

Important: Do not attempt this experiment if you suffer from pain in or around the jaw. Inform your instructor of any possible health problems that might be exacerbated if you participate in this exercise.

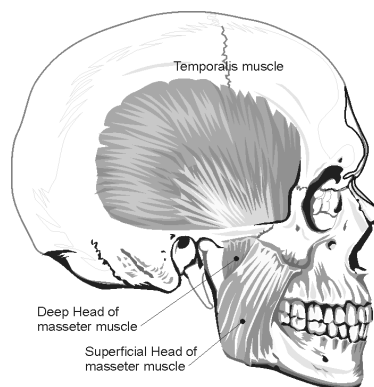


Figure 1

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of the electrical activity of a muscle.
- Associate amount of electrical activity with strength of muscle contraction.
- Compare masseter muscle function during different types of chewing activity.

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier EKG Sensor
electrode tabs

chewing gum
raw carrot
hard-boiled egg
soap or rubbing alcohol

PROCEDURE

Part I Conscious Clenching of the Jaw

Select one person from your lab group to be the subject.

1. Connect the EKG Sensor to the Vernier computer interface. Open the file “13 Introduction to EMG” from the *Human Physiology with Vernier* folder.
2. Instruct the subject to be seated. Remove excess oil from the skin with soap and water or alcohol to improve the adhesion of the electrode tabs to the skin. Position the upper electrode tab facing the ear so that the electrode wire may be looped over the ear (see Figure 2). Position the lower tab so it faces downward and the wire hangs down. Attach the EKG electrodes to the tabs; in this experiment red and green leads are interchangeable. Place a third electrode tab on some other area of the body, such as the left or right forearm, and attach the black EKG electrode to this tab.

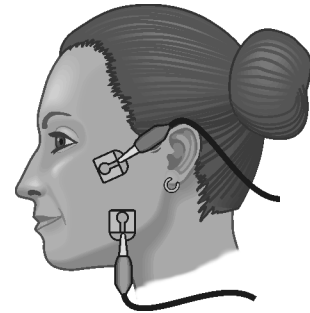


Figure 2

3. Have the student sit with his or her jaw relaxed. Click to begin data collection. If your graph has a stable baseline for 5 s (see Figure 3), click and continue to Step 4. If your graph has an unstable baseline, click and try again until you have a stable baseline for 5 s.
4. Click . After recording 5 s of stable baseline with the jaw relaxed, instruct the subject to clench his/her jaw for 5 s, then relax. Repeat this process of clenching for 5 second and relaxing for 5 s to obtain several events. Data collection will end after 30 s.

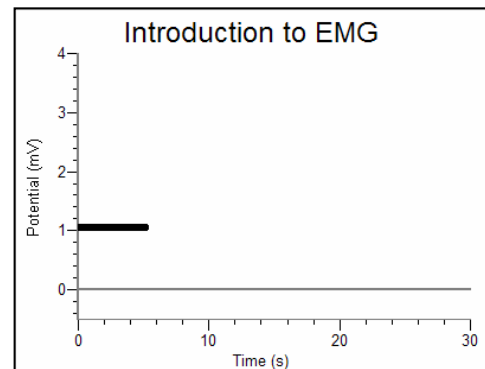



Figure 3

5. Click and drag to highlight the first period during which the subject's jaw was relaxed (approximately 0–5 s). Click the Statistics button, . Record the minimum and maximum values in Table 1, rounding to the nearest 0.01 mV.
6. Move the Statistics brackets to frame the next 5 s interval (5–10 s), during which the subject was clenching his/her jaw. Record the minimum and maximum values in Table 1, rounding to the nearest 0.01 mV. To close the Statistics box, click the × in the corner of the box.

Part II Comparison of Muscle Action in the Chewing of Different Foods

7. Click to begin data collection. If your graph has a stable baseline for 5 s (see Figure 3), click and continue to Step 8. If your graph has an unstable baseline, click and try again until you have a stable baseline for 5 s.
8. Click to begin data collection. After recording 5 s of stable baseline, instruct the subject to take a bite of hard-boiled egg and chew for the next 15–20 s. After chewing and swallowing has been accomplished, have the subject relax his/her jaw to return to baseline for the last 5–10 s of data collection.

9. Click and drag to highlight the first period during which the subject’s jaw was relaxed (approximately 0–5 s). Click the Statistics button, . Record the minimum and maximum values in Table 1, rounding to the nearest 0.01 mV.
10. Move the brackets to frame the data recorded during the chewing interval of Run 2, and record the minimum and maximum values for this interval in Table 1, rounding to the nearest 0.01 mV. To close the Statistics box, click the × in the corner of the box.
11. Repeat Steps 7–10 with the subject chewing on a raw carrot. If chewing and swallowing has not been completed by 25 s, subject should cease chewing and relax the jaw to return to baseline for the final 5 s of data collection. Be sure to select the correct run when you are obtaining statistics for the data.
12. Repeat Steps 7–10 with the subject chewing on a piece of gum. The subject should cease chewing and relax the jaw to return to baseline for the final 5 s of data collection.
13. Calculate the difference between each minimum and maximum value and record this value in the data table under the column marked Δ mV.

DATA

Table 1				
Condition	Interval	Minimum mV	Maximum mV	Δ mV
Jaw clenching	0–5 s			
	5–10 s			
Chewing hard-boiled egg	0–5 s			
	chewing interval			
Chewing raw carrot	0–5 s			
	chewing interval			
Chewing gum	0–5 s			
	chewing interval			

DATA ANALYSIS

1. Rank, in order (from greatest to least), the amplitude of EMG electrical activity for each of the items tested: carrot, egg, chewing gum.
2. Compare the frequency of muscle activation during mastication (chewing) of the three food items tested. Is there a significant difference in the number of similar spikes generated during a 5 s interval of data collection for each of the items tested?
3. Compare rates of chewing within your lab group/class. Are there significant differences?
4. On the basis of the findings in this experiment what recommendation would you make to a friend with a temporomandibular disorder (TMD) regarding his/her food choices?
5. The “Iron Jaw Trick” is a popular circus act in which a performer (or two performers) hang from a trapeze by his/her teeth. What exercises might someone do to strengthen the masseter muscles of the jaw so that a trick of this type could be performed successfully?
6. Chronic headaches and temporomandibular disorders (TMD) may be the result of unconscious clenching of the jaw. What are some ways to reduce/prevent jaw clenching?

EXTENSION

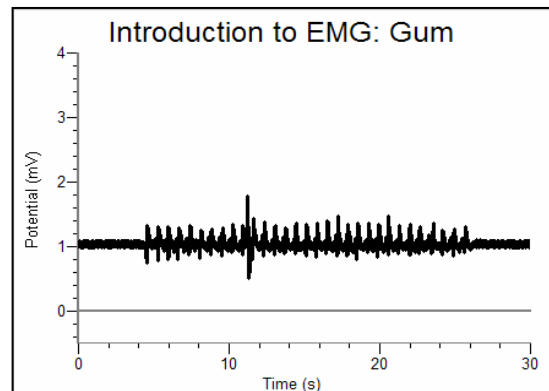
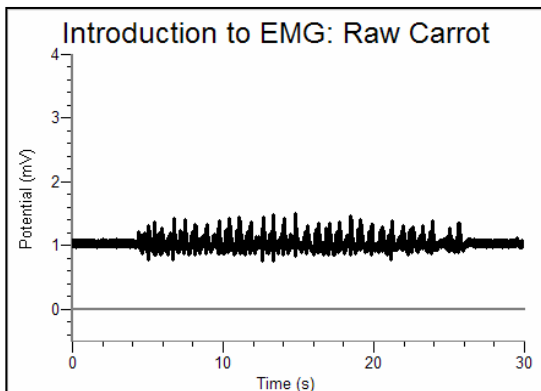
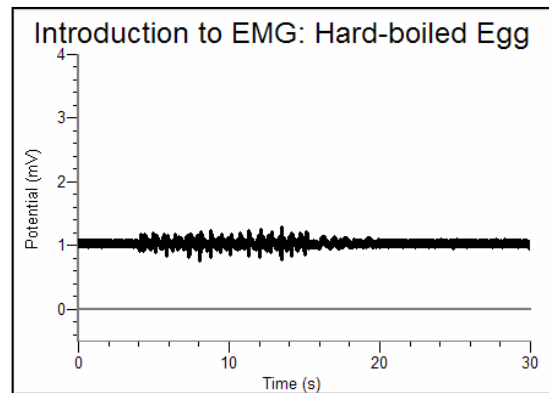
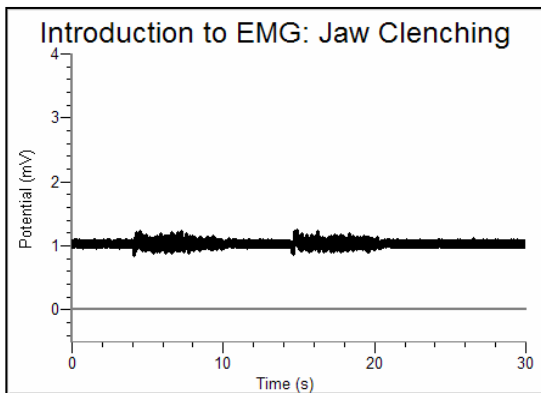
Test other food items, such as beef jerky, gummi bears, pudding, various meats, various fruits, etc.

TEACHER INFORMATION

Introduction to EMG

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Make sure that all hazardous materials are completely removed from any area in the lab in which food is going to be chewed.
3. A cleaner signal may be obtained by disconnecting LabQuest from AC power.

SAMPLE DATA



Sample data showing response of masseter muscle to chewing different types of food

Experiment 13

Table 1				
Condition	Interval	Minimum mV	Maximum mV	Δ mV
Jaw clenching	0–5 s	0.97	1.10	0.13
	5–10 s	0.84	1.23	0.39
Chewing hard-boiled egg	0–5 s	0.95	1.09	0.14
	chewing interval	0.77	1.28	0.51
Chewing raw carrot	0–5 s	0.97	1.10	0.13
	chewing interval	0.76	1.50	0.74
Chewing gum	0–5 s	0.97	1.09	0.12
	chewing interval	0.50	1.77	1.27

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Electrical activity (and muscle contraction) was greatest for chewing gum and least for egg.
2. For most people, the rate of chewing is fairly constant, regardless of the texture of the food. There may be significant differences between chewing rates of different students, which may merit further discussion (i.e., Do those with a faster chewing rate normally eat meals quickly? Do those with a faster rate of chewing also chew gum regularly?)
3. Answers will vary. Chewing rates differ between individuals.
4. A person with TMD would probably experience less pain in the jaw if he/she selected soft foods and avoided gum chewing.
5. Gum chewing would be a way to exercise the masseter muscles throughout the day.
6. Some of your students may have experienced TMD and will be able to answer this question from personal experience. Doctors and dentists will often prescribe relaxation techniques, physical therapy and mouth appliances for patients who have TMD.

Neuromuscular Reflexes

The automatic response of a muscle to a stimulus is called a reflex. The patellar reflex results from tapping the patellar tendon below the knee with a reflex hammer. This causes contraction of the quadriceps muscle and extension of the leg. Stretching of the muscle activates nerve impulses which travel to the spinal cord. Here the incoming impulses activate motor neurons, which travel back to the muscle and result in muscle contraction. This reflex arc is primarily a spinal reflex, but is influenced by other pathways to and from the brain (Figure 1).

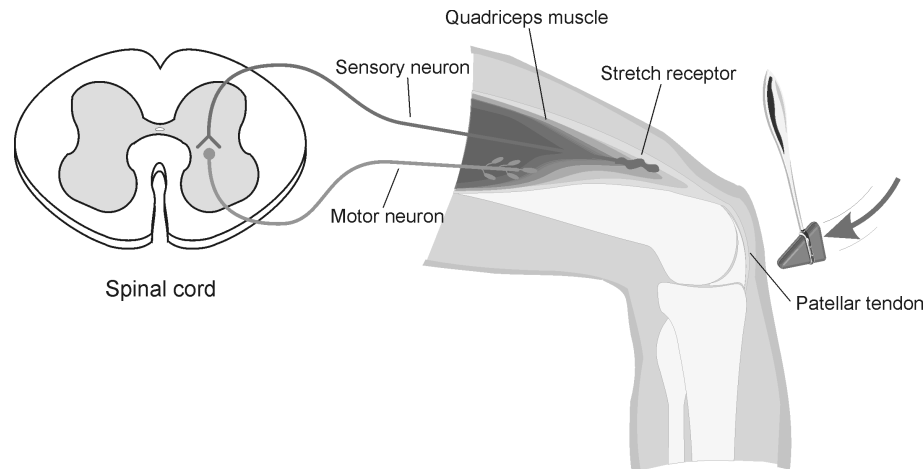


Figure 1

A reflex may be *reinforced* (a term used by neurologists) by slight voluntary contraction of muscles other than the one being tested. For example, voluntary activation of arm muscles by motor neurons in the central nervous system “spills over” to cause a slight activation of the leg muscles as well. This results in the enhancement of the patellar reflex. There are other examples of central nervous system influences on reflexes. Health care professionals use knowledge of these influences to aid in diagnosis of conditions such as acute stroke and herniated lumbar disk, where reflexes may be absent; and spinal cord injury and multiple sclerosis, which may result in exuberant reflexes.

In this experiment, you will use an EKG Sensor to compare the speed of a voluntary *vs.* a reflex muscle action and to measure the relative strength (amplitude) of the impulse generated by a stimulus with and without reinforcement. You will make a rough calculation of nerve impulse speed using data generated by an Accelerometer used in conjunction with the EKG sensor.

Important: Do not attempt this experiment if you have pain in or around the knee. Inform your instructor of any possible health problems that might be exacerbated if you participate in this exercise.

OBJECTIVES

In this experiment, you will

- Graph the electrical activity of a muscle activated by a reflex arc through nerves to and from the spinal cord.
- Compare the relative speeds of voluntary and reflex muscle activation.
- Associate muscle activity with involuntary activation.
- Observe the effect of central nervous system influence on reflex amplitude.
- Calculate the approximate speed of a nerve impulse.
- Compare reflex response and electrical amplitude in different subjects.

MATERIALS

computer
Vernier computer interface
Logger Pro
Vernier EKG Sensor
Vernier 25-g Accelerometer

electrode tabs
reflex hammer
cable tie, 10 cm long
cloth tape measure
pen

PROCEDURE

Each person in the group will take turns being subject and tester.

Part I Voluntary Activation of the Quadriceps Muscle

1. Connect the EKG Sensor and the Accelerometer to the Vernier computer interface.
2. Open the file “14A Reflexes with ACC” from the *Human Physiology with Vernier* folder.
3. Connect the Accelerometer to the reflex hammer using the cable tie, as shown in Figure 2. Place the Accelerometer cable toward the back of the hammer so it does not get in the way.

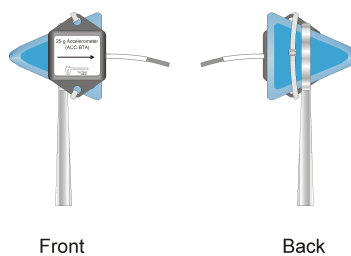


Figure 2

4. Have the subject sit comfortably in a chair that is high enough to allow his/her legs to dangle freely above the floor.
5. Attach two electrode tabs above one knee along the line of the quadriceps muscle between the knee and the hip. The tabs should be 5 cm and 13 cm from the middle of the patella (see Figure 3). Place a third electrode tab on the lower leg.

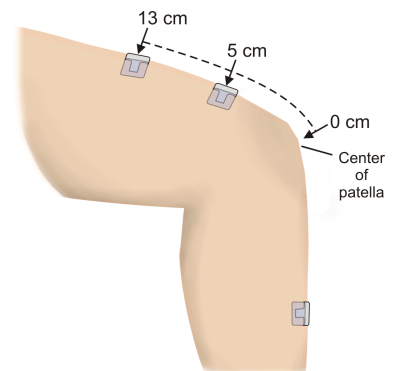


Figure 3

6. Attach the red and green leads to the electrode tabs above the knee with the red electrode closest to the knee. Attach the black lead (ground) to the electrode tab on the lower leg.
7. Click to begin data collection. If the graph has a stable baseline as shown in Figure 4, click and continue to Step 8. If your graph has an unstable baseline, click and collect a new set of data by clicking again. Repeat data collection until you have obtained a stable baseline for 5 s.
8. Collect voluntary activation data. **Note:** Read the entire step before collecting data to familiarize yourself with the procedure.
 - a. Have the subject close his/her eyes, or avert them from the screen.
 - b. Click .
 - c. After recording 5 s of stable baseline, swing the reflex hammer briskly to contact the table or other surface that generates a sound.
 - d. The subject should kick his/her leg out immediately upon hearing the sound.
 - e. Continue obtaining reflexes (repeat parts c and d of this step) so that you record 5–10 kicks during the data-collection period.

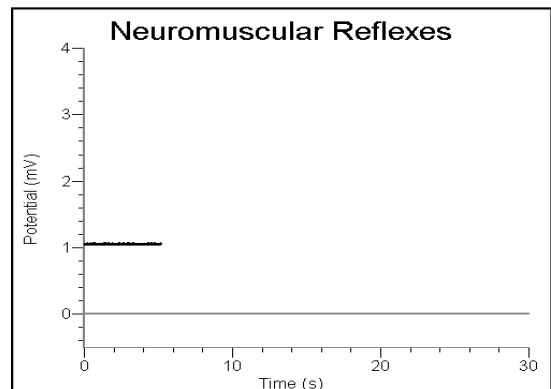


Figure 4

9. Determine the time elapsed between striking the table surface with the reflex hammer and the contraction of the quadriceps muscle.
 - a. Click the Examine button, , and place your cursor somewhere on the Acceleration graph.
 - b. Align the Examine line with the first high peak (which corresponds to the first kick) in the Accelerometer graph (Figure 5). This peak indicates the time at which the table surface was struck. Record this time in Table 1.
 - c. Move the cursor to the EMG graph and align the Examine line with the first high peak (Kick 1) in the graph. This peak indicates the time at which the quadriceps muscle contracted. Record this time in Table 1.
 - d. Repeat this process of determining the time of the hammer strike and reflex for a total of five stimulus-kick pairs.
 - e. Close the Examine box by clicking the × in the corner of the box.
 - f. Calculate the change in time between the hammer strike and reflex for the five stimulus-kick pairs and then calculate the average change in time for all five pairs. Record the values in Table 1.

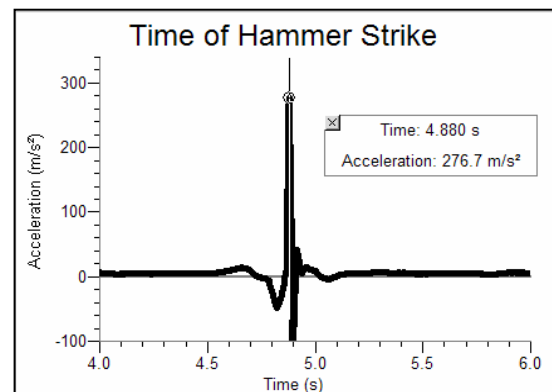


Figure 5

Part II Patellar Reflex

10. Locate the subject's patellar tendon by feeling for the narrow band of tissue that connects the lower aspect of the patella to the tibia. Place a pen mark in the center of the tendon, which can be identified by its softness compared with the bones above and below (see Figure 6).
11. Click to begin data collection. If your graph has a stable baseline as shown in Figure 4, click and continue to Step 12. If your graph has an unstable baseline, click and repeat data collection until you have obtained a stable baseline for 5 s.
12. Collect patellar reflex data. **Note:** Read the entire step before collecting data to familiarize yourself with the procedure.
 - a. Have the subject close his/her eyes, or avert them from the screen.
 - b. Click .
 - c. After recording 5 s of stable baseline, swing the reflex hammer briskly to contact the mark on the subject's tendon. If this does not result in a visible reflex, aim toward other areas of the tendon until the reflex is obtained.
 - d. Continue obtaining reflexes so that you record 5–10 reflexes during the collection period.
13. Determine the time elapsed between striking the patellar tendon with the reflex hammer and the contraction of the quadriceps muscle.
 - a. Click the Examine button, and place your cursor somewhere on the Acceleration graph.
 - b. Align the Examine line with the first high peak (which corresponds to the first kick) in the Accelerometer graph (Figure 5). This peak indicates the time at which the tendon was struck. Record this time in Table 2.
 - c. Move the cursor to the EMG graph and align the Examine line with the first high peak (Kick 1) in the graph. This peak indicates the time at which the quadriceps muscle contracted. Record this time in Table 2.
 - d. Repeat this process of determining the time of the hammer strike and reflex for a total of five stimulus-kick pairs.
 - e. Close the Examine box by clicking the X in the corner of the box.
 - f. Calculate the change in time between the hammer strike and reflex for the five stimulus-kick pairs and then calculate the average change in time for all five pairs. Record the values in Table 2.

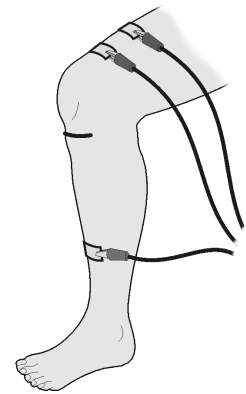


Figure 6

Part III Reflex Reinforcement

14. With the subject sitting comfortably in a chair, click to begin data collection. If your graph has a stable baseline, click and continue to Step 15. If your graph has an unstable baseline, click and repeat data collection until you have obtained a stable baseline for 5 s.
15. Collect patellar reflex data without and with reinforcement. **Note:** Read the entire step before collecting data to familiarize yourself with the procedure.
 - a. Have the subject close his/her eyes, or avert them from the screen.
 - b. Click .
 - c. After recording a stable baseline for 5 s, swing the reflex hammer briskly to contact the mark on the subject's tendon. If this does not result in a visible reflex, aim toward other areas of the tendon until the reflex is obtained.

- d. After 5 or 6 successful reflexes have been obtained, have the subject *reinforce* the reflex by hooking together his/her flexed fingers and pulling apart at chest level, with elbows extending outward (see Figure 7).
- e. Continue obtaining reflexes until data collection is completed at 30 s. A total of 10–15 reflexes should appear on the graph.

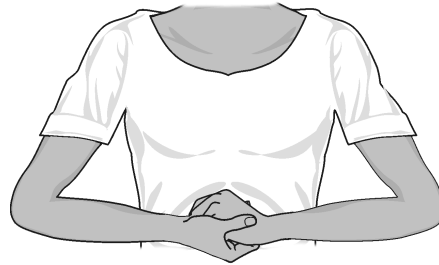



Figure 7

16. Click the Statistics button, . Move the brackets to frame the first area of increased amplitude (depolarization) in this run (see Figure 8). Record the minimum, maximum and Δy value (amplitude) for this depolarization in Table 3, rounding to the nearest 0.01 mV.
17. Repeat this process for each of five *unreinforced* and five *reinforced* depolarization events, using the Accelerometer to identify each primary reflex. Ignore rebound responses. Record the appropriate values in Table 3.
18. Determine the average amplitude of the *reinforced* and *unreinforced* depolarization events examined. Record these values in Table 3.

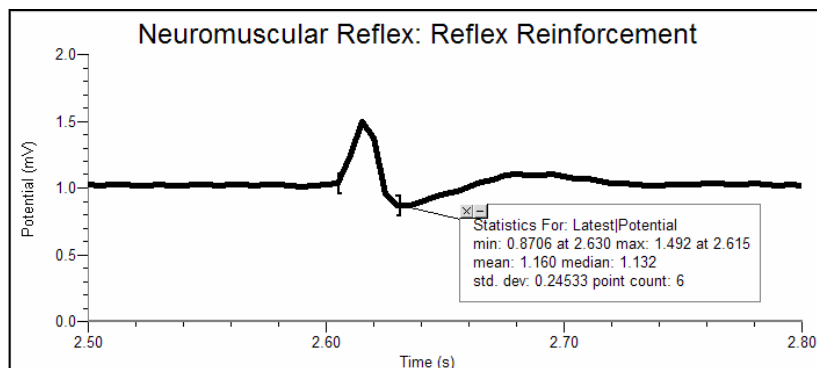


Figure 8

DATA

Table 1						
	Kick 1	Kick 2	Kick 3	Kick 4	Kick 5	Average
Time of muscle contraction (s)						
Time of stimulus (s)						
Δt (s)						

Table 2						
	Reflex 1	Reflex 2	Reflex 3	Reflex 4	Reflex 5	Average
Time of muscle contraction (s)						
Time of stimulus (s)						
Δt (s)						

Table 3						
	Reflex without reinforcement			Reflex with reinforcement		
Reflex response	Max (mV)	Min (mV)	ΔmV	Max (mV)	Min (mV)	ΔmV
1						
2						
3						
4						
5						
Average values						

DATA ANALYSIS

1. Compare the reaction times for voluntary *vs.* involuntary activation of the quadriceps muscle. What might account for the observed differences in reaction times?

- Using data from Table 2, calculate speed at which a stimulus traveled from the patellar tendon to the spinal cord and back to the quadriceps muscle (a complete reflex arc). To do this, you must estimate the distance traveled. Using a cloth tape measure, measure the distance in cm from the mark on the patellar tendon to the spinal cord at waist level (straight across from the anterior-superior iliac spine—see Figure 9). Multiply the distance by two to obtain the total distance traveled in the reflex arc. Once this value has been obtained, divide by the average Δt from Table 2 and divide by 100 to obtain the speed, in m/s, at which the stimulus traveled.

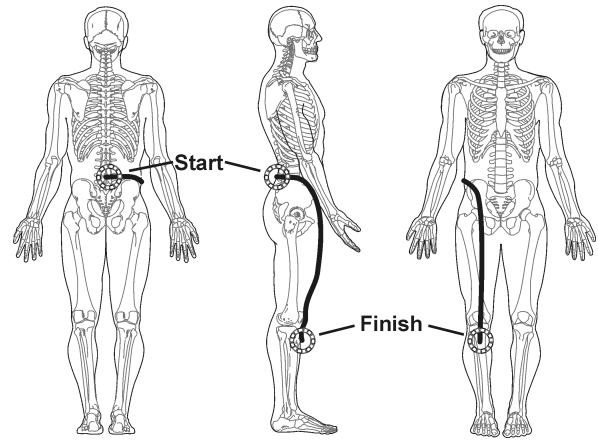


Figure 9

- Nerve impulses have been found to travel as fast as 100 m/s. What could account for the difference between your answer to Question 2 and this value obtained by researchers?
- Assume the speed of a nerve impulse is 100 m/s. How does this compare to the speed of electricity in a copper wire (approx. 3.00×10^8 m/s)?
- Compare the data you obtained in this experiment with other members of your group/class. Can individual differences be attributed to any physical differences (body shape/size, muscle mass, physical fitness level)?

EXTENSION

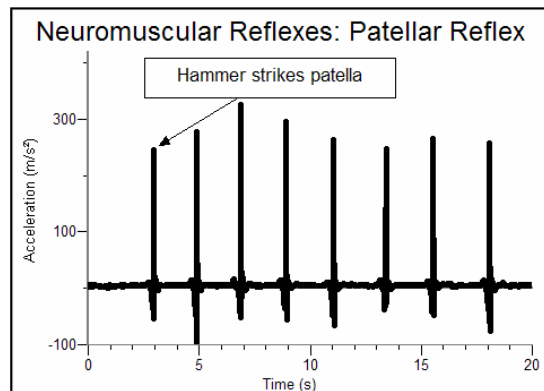
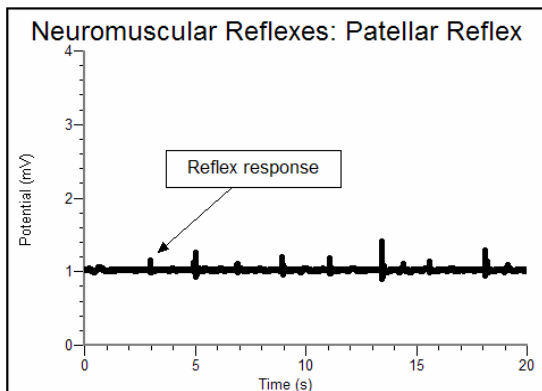
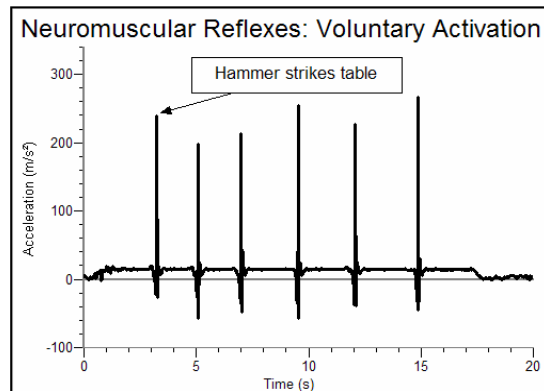
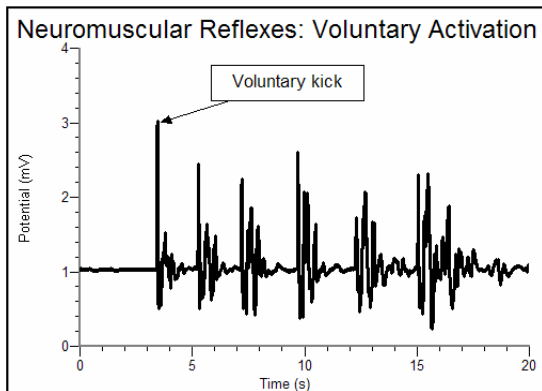
- Explore the ankle and elbow reflexes.
- Perform the experiment with the subject watching the reflex hammer as it hits the patellar tendon. Compare this data to data gathered while the subject is focusing on an object elsewhere in the room.

TEACHER INFORMATION

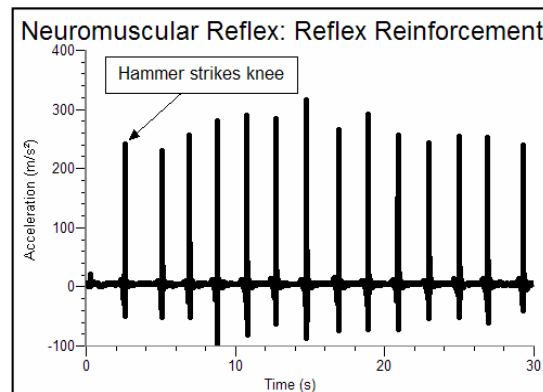
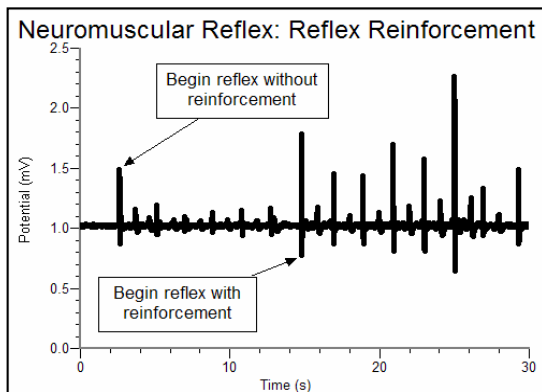
Neuromuscular Reflexes

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. You will want to warn students to dress appropriately for the lab period(s) during which this experiment is performed. In order to place the electrode tabs above the knee, short pants are recommended.
3. Make sure to ask students whether they are experiencing pain in or around the knee before allowing them to proceed with the experiment.
4. In Part III of the experiment, students will need to identify the initial stimulus. Be sure to warn them not to measure rebound responses that appear after each primary reflex.
5. Some students may not have measurable reflexes. Reassure them that this is part of biologic variability.
6. To obtain good reflex responses, the subject should be relaxed. This may require that they lean against a back support. This is particularly important in Part III of the experiment, where students should be warned to pull their arms apart isometrically without allowing this action to cause tension in other areas of their bodies.
7. A cleaner signal may be obtained by disconnecting LabQuest from AC power.

SAMPLE DATA



Sample data collected during voluntary and reflexive response to the strike of the reflex hammer



Sample data collected while examining the effect of reinforcing a reflex

Table 1						
	Kick 1	Kick 2	Kick 3	Kick 4	Kick 5	Average
Time of muscle contraction (s)	3.45	5.30	7.18	9.72	12.29	
Time of stimulus (s)	3.23	5.07	7.00	9.52	12.07	
Δt (s)	0.22	0.23	0.18	0.20	0.22	0.21

	Reflex 1	Reflex 2	Reflex 3	Reflex 4	Reflex 5	Average
Time of muscle contraction (s)	2.98	5.02	6.90	8.84	11.08	
Time of stimulus (s)	2.94	4.87	6.87	8.90	11.05	
Δt (s)	0.04	0.13	0.03	0.04	0.03	0.05

Reflex response	Reflex without reinforcement			Reflex with reinforcement		
	Max (mV)	Min (mV)	ΔmV	Max (mV)	Min (mV)	ΔmV
1	1.49	0.87	0.62	1.78	0.77	1.01
2	1.20	0.95	0.25	1.46	0.87	0.59
3	1.10	0.98	0.12	1.44	0.87	0.57
4	1.13	0.97	0.16	1.70	0.81	0.89
5	1.15	0.95	0.20	1.58	0.81	0.77
Average values			0.27			0.77

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. In the case of involuntary activation of the quadriceps muscle, the reflex arc is short—from patella to spinal cord and back. In the case of voluntary activation, the sound of the reflex hammer must travel to the ear and from the ear to the brain. The brain must then process the sound and send motor impulses back to the muscle.
2. Answers will vary, but should be in the range of 3050 m/s. In the sample data, the speed is 30.50 m/s.
3. The 100 m/s results found by researchers were measured under ideal conditions, using isolated nerve fibers.
4. Electricity travels through copper wire at a speed of 3.00×10^8 m/sec. This is approximately 3 million times faster than the speed of a nerve impulse.
5. Contrary to what might be intuitive for your students, fitness level, body type, etc., have no bearing on the individual differences seen in this experiment.

Neuromuscular Reflexes

The automatic response of a muscle to a stimulus is called a reflex. The patellar reflex results from tapping the patellar tendon below the knee with a reflex hammer. This causes contraction of the quadriceps muscle and extension of the leg. Stretching of the muscle activates nerve impulses which travel to the spinal cord. Here the incoming impulses activate motor neurons, which travel back to the muscle and result in muscle contraction. This reflex arc is primarily a spinal reflex, but is influenced by other pathways to and from the brain (Figure 1).

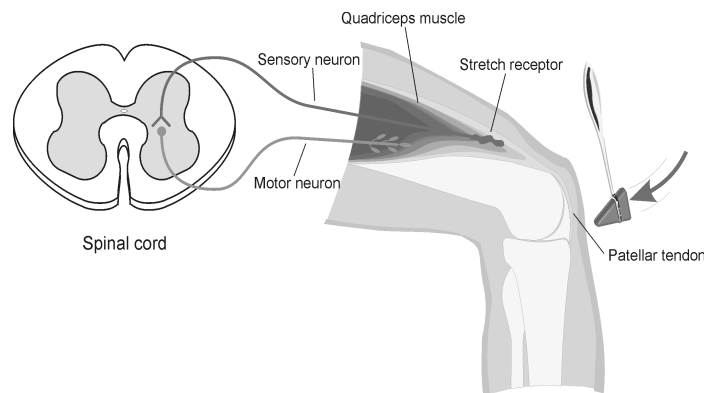


Figure 1

A reflex may be *reinforced* (a term used by neurologists to mean *enhanced*) by slight voluntary contraction of muscles other than the one being tested. For example, voluntary activation of arm muscles by motor neurons in the central nervous system “spills over” to cause a slight activation of the leg muscles as well. This results in the enhancement of the patellar reflex. There are other examples of central nervous system influences on reflexes. Health care professionals use knowledge of these influences to aid in diagnosis of conditions such as acute stroke and herniated lumbar disk, where reflexes may be absent; and spinal cord injury and multiple sclerosis, which may result in exuberant reflexes.

In this experiment, you will use an EKG Sensor to measure the relative strength (amplitude) of the impulse generated by a stimulus with and without reinforcement. This sensor, developed to measure the electrical events occurring within the heart, can be used to measure the electrical activity resulting from the contraction of other muscles, as well.

Important: Do not attempt this experiment if you have pain in or around the knee. Inform your instructor of any possible health problems that might be exacerbated if you participate in this exercise.

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of the electrical activity of a muscle activated by a reflex arc through nerves to and from the spinal cord.
- Associate muscle activity with involuntary activation.
- Observe the effect of central nervous system influence on reflex amplitude.

MATERIALS

computer
Vernier computer interface
Logger Pro
Vernier EKG Sensor

electrode tabs
reflex hammer
pen

PROCEDURE

Each person in the group will take turns being subject and tester.

1. Connect the EKG Sensor to the Vernier computer interface.
2. Open the file “14B Reflexes without ACC” from the *Human Physiology with Vernier* folder.
3. Have the subject sit comfortably in a chair that is high enough to allow his/her legs to dangle freely above the floor.
4. Attach two electrode tabs above one knee along the line of the quadriceps muscle between the knee and the hip. The tabs should be 5 and 13 cm, from the middle of the patella (see Figure 2). A third electrode tab should be placed on the lower leg.
5. Locate the subject’s patellar tendon by feeling for the narrow band of tissue that connects the lower aspect of the patella to the tibia. Place a pen mark in the center of the tendon, which can be identified by its softness compared with the bones above and below (see Figure 3).
6. Attach the red and green leads to the electrode tabs above the knee with the red electrode closest to the knee. Attach the black lead (ground) to the electrode tab on the lower leg.

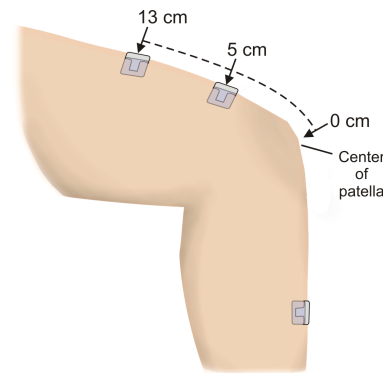


Figure 2

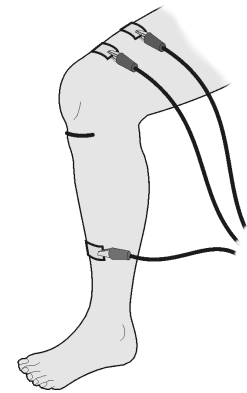


Figure 3

7. Click to begin data collection. If your graph has a stable baseline as shown in Figure 4, click and continue to Step 8. If your graph has an unstable baseline, click and repeat data collection until you have obtained a stable baseline for 5 s.

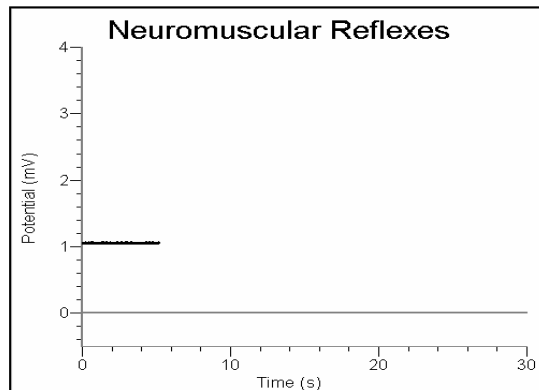


Figure 4

8. Collect patellar reflex data without and with reinforcement. **Note:** Read this entire step before collecting data to familiarize yourself with the process.
- Have the subject close his/her eyes, or avert them from the screen.
 - Click .
 - After recording a stable baseline for 5 s, swing the reflex hammer briskly to contact the mark on the subject's tendon. If this does not result in a visible reflex, aim toward other areas of the tendon until the reflex is obtained.
 - After 5 or 6 successful reflexes have been obtained, have the subject *reinforce* the reflex by hooking together his/her flexed fingers and pulling apart at chest level, with elbows extending outward (see Figure 5).
 - Continue obtaining reflexes until data collection is completed at 30 s. A total of 10–15 reflexes should appear on the graph.

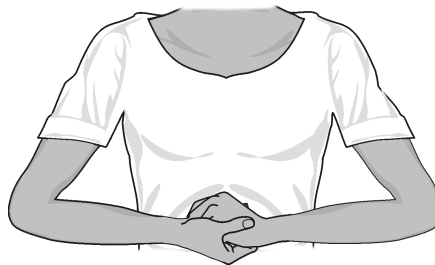


Figure 5

9. Determine the average amplitude of the *reinforced* and *unreinforced* reflexes.
- Click the Statistics button, .
 - Move the brackets to frame the first area of increased amplitude (depolarization) in this run (see Figure 6).
 - Record the minimum, maximum and Δy value (amplitude) for this depolarization in Table 1, rounding to the nearest 0.01 mV.
 - Close the Statistics box by clicking on the \times in the corner of the box.

- e. Repeat this process for 5 *unreinforced* and 5 *reinforced* depolarization events. Ignore rebound responses. Record the appropriate values in Table 1.
- f. Determine the average amplitude of the *reinforced* and *unreinforced* depolarization events examined. Record these values in Table 1.

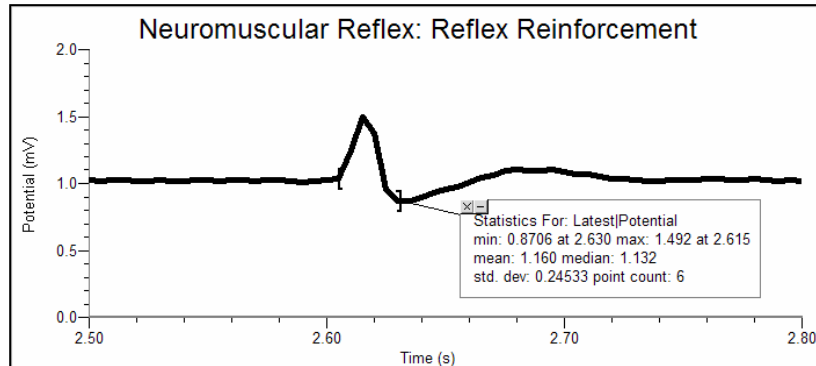


Figure 6

DATA

	Reflex without reinforcement			Reflex with reinforcement		
Reflex response	Max (mV)	Min (mV)	Δ mV	Max (mV)	Min (mV)	Δ mV
1						
2						
3						
4						
5						
Average values						

DATA ANALYSIS

1. Assume that there are nerve fibers that travel from the brain to the motor neurons in the spinal cord involved in this reflex arc. Some nerve tracts from the brain stimulate the spinal neurons, and some inhibit the same neurons. Speculate on the mechanism that would result in the differences you observed in the Δ mV values between reflexes with and without reinforcement.

2. Describe the differences between the wave forms resulting from a reflex stimulus with and without reinforcement. What can you hypothesize about the number of nerve fibers involved in a reinforced reflex as compared to one without reinforcement?
3. Compare the data you obtained in this experiment with other members of your group/class. Can individual differences be attributed to any physical differences (body shape/size, muscle mass, physical fitness level)?

EXTENSIONS

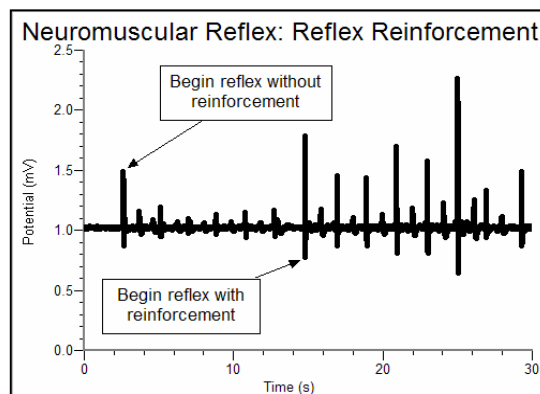
1. Explore the ankle and elbow reflexes.
2. Perform the experiment with the subject watching the reflex hammer as it hits the patellar tendon. Compare this data to data gathered while the subject is focusing on an object elsewhere in the room.
3. Research how various diseases (such as those mentioned in the Introduction to this experiment) can affect reflexes.

TEACHER INFORMATION

Neuromuscular Reflexes

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. You will want to warn students to dress appropriately for the lab period(s) during which this experiment is performed. In order to place the electrode tabs above the knee, short pants are recommended.
3. Make sure to ask students whether they are experiencing pain in or around the knee before allowing them to proceed with the experiment.
4. Some students may not have measurable reflexes. Reassure them that this is part of biologic variability.
5. To obtain good reflex responses, the subject should be relaxed. This may require that they lean against a back support. This is particularly important during reinforcement, where students should be warned to pull their arms apart isometrically without allowing this action to cause tension in other areas of their bodies.
6. A cleaner signal may be obtained by disconnecting LabQuest from AC power.

SAMPLE DATA



Sample data collected while examining the effect of reinforcing a reflex

Experiment 14B

Reflex response	Reflex without reinforcement			Reflex with reinforcement		
	Max (mV)	Min (mV)	Δ mV	Max (mV)	Min (mV)	Δ mV
1	1.49	0.87	0.62	1.78	0.77	1.01
2	1.20	0.95	0.25	1.46	0.87	0.59
3	1.10	0.98	0.12	1.44	0.87	0.57
4	1.13	0.97	0.16	1.70	0.81	0.89
5	1.15	0.95	0.20	1.58	0.81	0.77
Average values			0.27			0.77

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Possible explanations include:

- (1) There is a constant inhibitory impulse from the brain which is removed by the distraction of contracting a distant muscle group.
- (2) Reinforcement causes an increase in stimulatory impulses from the brain to other areas of the spine, resulting in a more pronounced reflex.

The mechanism by which reinforcement increases a reflex response remains uncertain.

2. The reinforced reflex is longer in duration and has a larger amplitude than the unreinforced reflex. Students may also point out additional spikes in the reinforced reflex, resulting from “rebound” contractions. Larger amplitude muscle activity indicates involvement of a larger number of nerve fibers.
3. Student answers will vary. Physical fitness does not have a direct influence on the size of the reflex. However, the size of a leg and the amount of subcutaneous tissue may render the small reflex hammer ineffective in eliciting a reflex response. It may also be difficult to accurately locate the patellar tendon.

Muscle Function Analysis

Muscle tissues maintain electrical imbalances, or *potentials*, across cell membranes by concentrating positive or negative charges on opposite sides of those membranes. These potentials are a form of stored energy. With activation (such as from a nerve impulse), the ions are allowed to cross the muscle cell membranes, generating electrical activity and resulting in muscle contraction.

An electromyogram, or EMG, is a graphical recording of electrical activity within muscles. It is useful in the diagnosis of disorders affecting muscles and the nerves that supply them. Inherited and acquired disorders of muscles (such as the muscular dystrophies), and disorders of the central and peripheral nervous systems (such as Huntington's disease and diabetic neuropathy) result in abnormal EMG readings.

EMG studies are also useful for investigating normal muscle function. In this experiment, you will analyze electrical activity in the extensor muscles of the forearm. These muscles originate in tendons of the proximal dorsal forearm in the region of the lateral epicondyle and attach distally to tendons which control extension of the hand and fingers (see Figure 1). Inflammation of the tendons at the elbow is common and can result from repetitive motions used in sports, hobbies, and in the workplace. This experiment will demonstrate the rationale behind treating tennis elbow with a wrist splint.

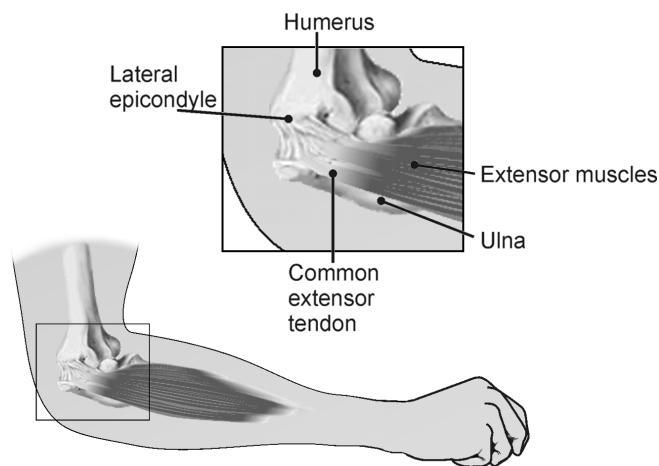


Figure 1

Important: Do not attempt this experiment if you suffer from tennis elbow or other disorders of the arm or hand. Inform your instructor of any possible health problems that might be exacerbated if you participate in this exercise.

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of the electrical activity of a muscle.
- Associate muscle activity with movement of joints.
- Correlate muscle activity with injury.

MATERIALS

computer
Vernier computer interface
Logger Pro
Vernier EKG Sensor

electrode tabs
ruler
1–2 kg (3 lb) weight (barbell or similar)

PROCEDURE

Select one person from your lab group to be the subject.

1. Connect the EKG Sensor to the Vernier computer interface. Open the file “15 Muscle Funct Analysis” from the *Human Physiology with Vernier* folder.
2. Attach three electrode tabs to your dominant arm, as shown in Figure 2. Two tabs should be placed on the dorsal forearm, 5 cm and 10 cm from the lateral epicondyle along an imaginary line connecting the epicondyle and the middle finger. A third electrode tab should be placed on the upper arm.
3. Connect the EKG clips to the electrode tabs as shown in Figure 2. The red and green leads are interchangeable for this experiment. Stand facing your table or lab bench so that your arm is angled down toward the surface (Figure 2).

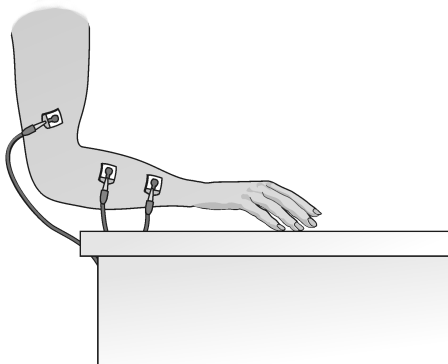


Figure 2

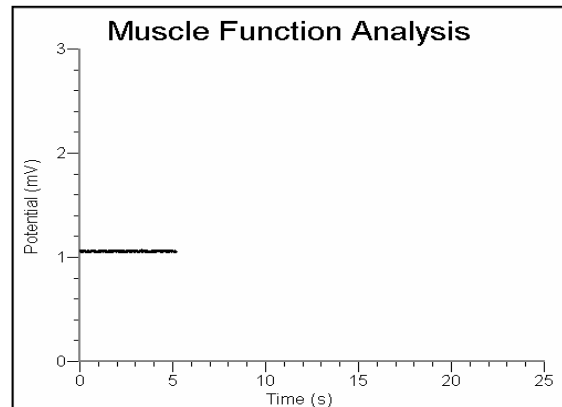


Figure 3

4. Rest your fingertips on the table, with your palm above the surface and in line with your forearm, as shown in Figure 2. Click to begin data collection. If your graph has a stable baseline (see Figure 3), click and continue to Step 5. If your graph has an unstable baseline, click and try again until you have a stable baseline for 5 s.

5. Collect data while flexing and relaxing the extensor muscle.
 - a. Click .
 - b. After recording a stable baseline for 5 s, gently cock your hand back as far as you can (Figure 4). Hold this position for 5 s.
 - c. Return your hand to its relaxed position, with fingertips resting on the table surface, for a period of 5 s.
 - d. Repeat the extension and relaxation action for one more full cycle—5 s with the hand cocked, followed by 5 s with the hand in a resting position. Data will be collected for 25 s.
6. Store this run by choosing Store Latest Run from the Experiment menu.

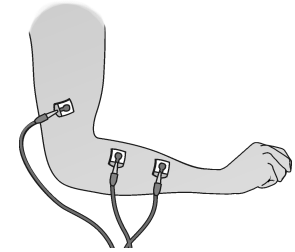


Figure 4

7. Click and drag to highlight the first 5 s of data collected. Click the Statistics button, . Record the minimum and maximum values displayed in Table 1, rounding to the nearest 0.01 mV.
8. Drag the Statistics brackets to highlight each of the next 5 s intervals until you reach 20 s (5–10 s, 10–15 s, 15–20 s). Record the minimum and maximum values displayed in the Statistics boxes in Table 1. When you are finished, close the Statistics box by clicking the × in the corner of the box. **Note:** You may have to slightly adjust the time parameters for the data selected, as flexion and relaxation times may not conform strictly to the 5-second time periods.

Ergonomics and Muscle Protection

9. Obtain a 1–2 kg (3 lb) weight (barbell or similar) from your instructor.
10. Place your relaxed hand on the weight, with your palm facing downward. Click to begin data collection. If your graph has a stable baseline, click and continue to Step 11. If your graph has an unstable baseline, click and repeat data collection until you have obtained a stable baseline for approximately 5 s.
11. Collect data to examine the effect of lifting a weight using your extensor muscles:

- a. Click .
- b. After recording a stable baseline for 5 s, grip the weight with your hand. Using only the action of your wrist, lift the weight to a height of 5–10 cm off the surface of the table (Figure 5). Hold this position for 5 s.
- c. Gently lower the weight onto the table and rest for 5 s.
- d. Repeat the extension and relaxation action for one more full cycle—5 s with the hand cocked, followed by 5 s with the hand in a resting position. Data will be collected for 25 s.

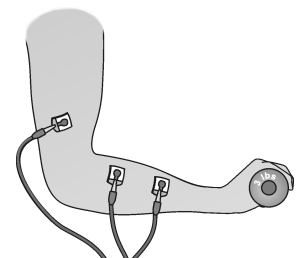


Figure 5

12. Store this run by choosing Store Latest Run from the Experiment menu.
13. Click and drag to highlight the first 5 s of data collected. Click the Statistics button, . Record the minimum and maximum values displayed in Table 1, rounding to the nearest 0.01 mV.
14. Drag the Statistics brackets to highlight each of the next 5 s intervals until you reach 20 s (5–10 s, 10–15 s, 15–20 s). Record the minimum and maximum values displayed in the Statistics boxes in Table 1. When you are done, close the Statistics box by clicking the × in

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the corner of the box. Note: You may have to slightly adjust the time parameters for the data selected, as flexion and relaxation times may not conform strictly to the 5-second time periods.

15. Place your hand, palm facing upwards, under the weight. Lightly grip the weight. Click to begin data collection. If your graph has a stable baseline, click and continue to Step 16. If your graph has an unstable baseline, click and repeat data collection until you have obtained a stable baseline for approximately 5 s.
16. Collect data while lifting a weight using your flexor muscles:
 - a. Click .
 - b. After recording a stable baseline for 5 s, using only the action of your wrist, lift the weight to a height of 5–10 cm off the surface of the table (see Figure 6). Hold this position for 5 s.
 - c. Gently lower the weight onto the table and rest for 5 s.
 - d. Repeat the flexation and relaxation action for one more full cycle—5 s of flexing upward with weight, followed by 5 s with the hand in a resting position.
17. Click and drag to highlight the first 5 s of data. Click the Statistics button, . Record the minimum and maximum values in Table 1, rounding to the nearest 0.01 mV.
18. Drag the Statistics brackets to highlight each of the 5-second intervals until you reach 20 s (5–10 s, 10–15 s, 15–20 s). Record the minimum and maximum values in Table 1. Note: You may have to slightly adjust the time parameters for the data selected.
19. Calculate the difference between each minimum and maximum value and record this value in the data table.

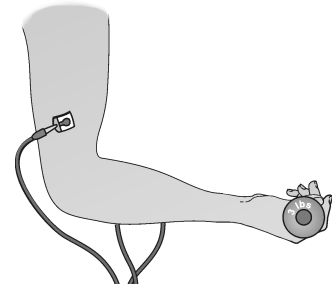


Figure 6

DATA

Table 1				
Condition	Interval (approximate)	Minimum (mV)	Maximum (mV)	Δ mV
Fingers resting on table	0–5 s			
Hand cocked back	5–10 s			
Fingers resting on table	10–15 s			
Hand cocked back	15–20 s			
Fingers resting on weight–overhand	0–5 s			
Hand cocked back with weight	5–10 s			
Fingers resting on weight–overhand	10–15 s			
Hand cocked back with weight	15–20 s			
Fingers loosely gripping weight–underhand	0–5 s			
Hand flexed upward with weight	5–10 s			
Fingers loosely gripping weight–underhand	10–15 s			
Hand flexed upward with weight	15–20 s			

DATA ANALYSIS

1. Reviewing your data, what conclusion can you reach regarding the relationship between the strength of muscle contraction and the amplitude of electrical activity generated?
2. What do the results in Table 1 show about the relationship between hand position and muscle activity when lifting an object?
3. What advice would you give to a friend with tennis elbow about how she should lift a frying pan onto the stove or a can of paint onto a work bench?

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4. Using what you have learned in this experiment, explain why a wrist brace is effective treatment for tennis elbow.

5. Some people keyboard with their wrists resting on the table or on a wrist support. In light of what you have learned from this experiment, would you recommend this practice as a way of preventing injury to the extensor muscles of the forearm? Why or why not?

6. Based on this experiment, what advice would you give to a beginning piano player about proper hand position?

7. Pitcher's elbow is a repetitive use injury of the medial epicondyle. What do the muscles that attach to the medial epicondyle control? How would you adapt this experiment to test these muscles?

EXTENSION

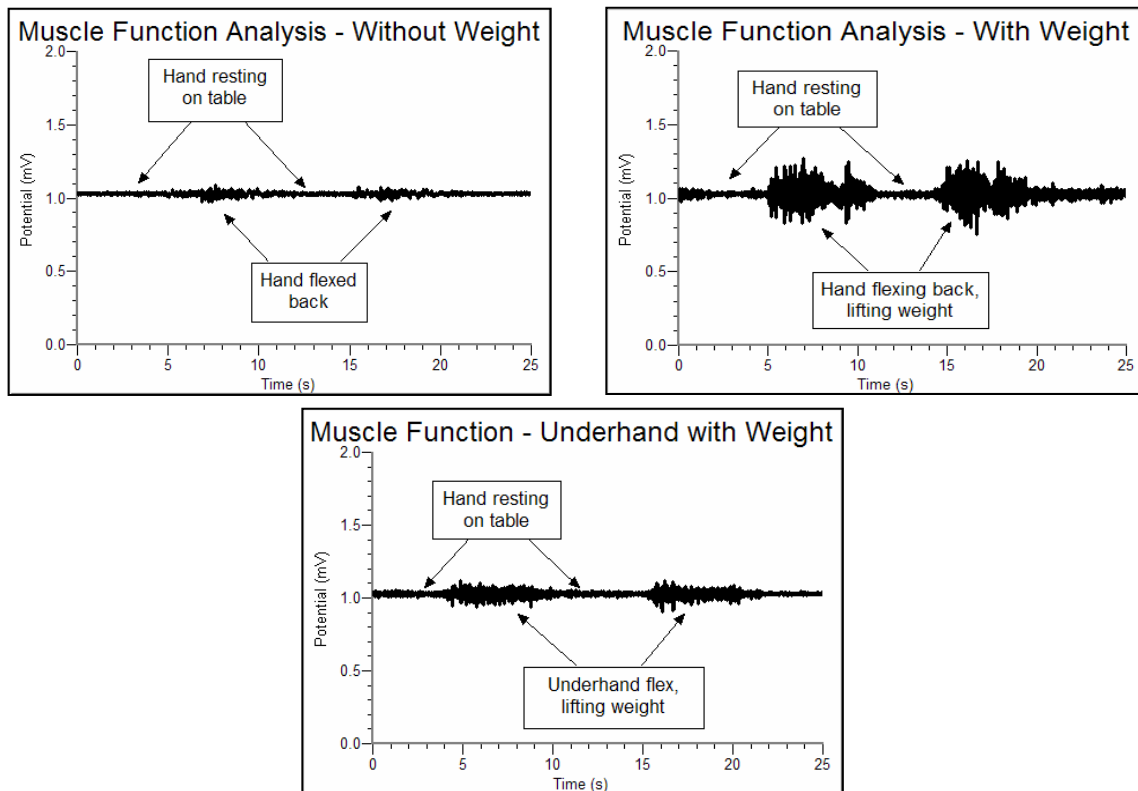
In Step 2, the leads may be placed on the ventral forearm instead of the dorsal forearm (Figure 6). The remaining steps can then be carried out without change. Explain the resulting EMG tracing.

TEACHER INFORMATION

Muscle Function Analysis

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. The position of the red and green leads at 5 and 10 cm from the lateral epicondyle is a general guideline. Students may experiment with different distances to optimize the baseline graph.
3. The leads should be placed on the dominant arm to allow for easier lifting of the weight. The positioning of the green and red leads on the forearm is interchangeable, and the ground (black) lead may be placed on the same or opposite upper arm.
4. Any object weighing 3–5 lb (large can of soup, full water bottle, etc.) can be substituted for the barbell-type weight called for in the Materials list in the student version of the experiment.
5. Make sure to ask students whether they are experiencing pain in the fingers, wrist, forearm, or elbow before allowing them to proceed with the experiment.
6. A cleaner signal may be obtained by disconnecting LabQuest from AC power.

SAMPLE DATA



Data collected while performing this activity

Experiment 15

Condition	Interval (approximate)	Minimum (mV)	Maximum (mV)	Δ mV
Fingers resting on table	0–5 s	1.00	1.04	0.04
Hand cocked back	5–10 s	0.97	1.09	0.12
Fingers resting on table	10–15 s	1.00	1.05	0.05
Hand cocked back	15–20 s	0.98	1.08	0.10
Fingers resting on weight–overhand	0–5 s	0.97	1.07	0.10
Hand cocked back with weight	5–10 s	0.83	1.27	0.44
Fingers resting on weight–overhand	10–15 s	0.92	1.12	0.20
Hand cocked back with weight	15–20 s	0.75	1.25	0.50
Fingers loosely gripping weight–underhand	0–5 s	0.95	1.12	0.17
Hand flexed upward with weight	5–10 s	0.93	1.11	0.18
Fingers loosely gripping weight–underhand	10–15 s	1.00	1.05	0.05
Hand flexed upward with weight	15–20 s	0.90	1.12	0.22

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. The greater the strength of contraction, the greater the amplitude of electrical activity.
2. Cocking the wrist requires contraction of the extensor muscles. This contraction is seen graphically as an increase in amplitude over the baseline electrical activity of this muscle group. Lifting the weight with the palm up accomplishes the same task without using the extensor muscles. Remind students that there are many instances of opposing muscle groups in the human body. When one group is flexed, the opposite group is completely relaxed.
3. She should lift either object with her palm facing upward. This will utilize the flexor muscles which attach at the medial epicondyle and will allow the lateral epicondyle to rest. Repetitive use of the forearm flexors may cause “pitcher’s elbow.”
4. Injured tendons (such as in tennis elbow) are best healed through rest. A wrist splint is the most effective way to immobilize the muscles in the forearm, allowing them to rest. Tennis elbow straps are useful if work or sports activities must be continued but are less effective.
5. This practice can result in greater injury depending upon the position of the wrist during typing. Typing with your wrist on a rest usually involves cocking the wrist, which requires use of the extensor muscles to keep the fingers elevated off of the keyboard. Wrist rests are meant to be used to rest the hands while not in the act of typing.
6. The most relaxed hand position is one in which the wrist is level with the hand as the fingers rest on the piano keys.
7. The muscles that attach to the medial epicondyle control flexion of the hand and fingers. The leads may be placed on the flexor muscles similar to how they were placed on the extensor muscles in this experiment. The results obtained using the weight will be opposite to those obtained in this experiment.

Grip Strength Comparison

The importance of hand strength and function is evident in all aspects of our daily living, from eating and maintaining personal hygiene to keyboarding at the computer, performing brain surgery, or playing tennis or the piano. People suffering from arthritis or hand injury quickly appreciate the difficulty of performing even the most mundane tasks with reduced grip strength.

Testing of hand grip strength is used by orthopedic surgeons and physical therapists to evaluate the extent of an injury and the progress of recovery. Grip strength can also be used to diagnose neuromuscular problems such as stroke, herniated disks in the neck, carpal tunnel syndrome, and elbow tendonitis. Athletes are interested in grip strength because it relates to performance in many sports, such as tennis, golf, baseball, football, gymnastics, and rock climbing.

Pinch strength is a way for occupational therapists to measure loss of fine-motor strength in the thumb, fingers, and forearm. It is useful for analyzing the extent of an injury and the outcome from surgery or therapy.

In Part I of this experiment, you will measure and compare grip strength in your right and left hands. You will also correlate grip strength with gender, handedness, and height. In Part II you will analyze the pinch strength of each of your four fingers.

Important: Do not attempt this exercise if you have arthritis, carpal tunnel syndrome, or any ailment that might be exacerbated by using the muscles of your arm and hand.



Figure 1

OBJECTIVES

In this experiment, you will

- Measure and compare grip strength of your right and left hands.
- Correlate grip strength with gender and certain physical characteristics.
- Compare the pinch strengths of the individual fingers of the dominant hand.

MATERIALS

computer
Vernier computer interface

Logger Pro
Vernier Hand Dynamometer

PROCEDURE

Each person in the group will take turns being subject and tester.

Part I Hand Grip Strength

1. Connect the Hand Dynamometer to the Vernier computer interface. Open the file “16a Compare Grip Strength” from the *Human Physiology with Vernier* folder.
2. Zero the readings for the Hand Dynamometer.
 - a. Hold the Hand Dynamometer along the sides, in an upright position (see Figure 2). Do not put any force on the pads of the Hand Dynamometer.
 - b. Click the Zero button, .
3. Have the subject sit with his or her back straight and feet flat on the floor. The Hand Dynamometer should be held in the right hand. The elbow should be at a 90° angle, with the arm unsupported (see Figure 1).
4. Have the subject close his or her eyes, or avert them from the screen.
5. Click to begin data collection. After collecting 2 s of baseline data, instruct the subject to grip the sensor with full strength for the next 8 s. Data will be collected for 10 s.
6. Store this run by choosing Store Latest Run from the Experiment menu.
7. Repeat Step 2–5 with the left hand.
8. Determine the maximum and mean force exerted by your hands during a portion of the data collection period.
 - a. Place the cursor over your graph at 4 s and click and drag to highlight both runs from 4 s to 8 s.
 - b. Click the Statistics button, , to see the Statistics box.
 - c. Check the boxes in front of Run 1 and Latest and click .
 - d. Record the maximum and mean force for each run in Table 1.
 - e. Close the Statistics box by clicking the × in the corner of the box.

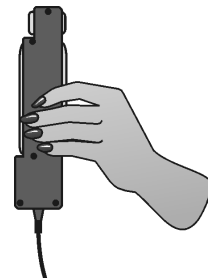





Figure 2

- Work with your classmates to complete Tables 2–4. **Note:** In Table 4, round height to the nearest inch.

Part II Pinch Strength

- Open the file “16b Compare Grip Strength” from the *Human Physiology with Vernier* folder.
- Have the subject sit with his or her back straight and feet flat on the floor, holding the Hand Dynamometer along the sides in the non-dominant hand (see Figure 3). **Note:** No additional force should be placed on the sensor by this hand.
- Have the subject close his or her eyes, or avert them from the screen.
- Zero the readings for the Hand Dynamometer.
 - Hold the Hand Dynamometer along the sides, in an upright position. Do not put any force on the gray pads of the Hand Dynamometer.
 - Click the Zero button, .
- Click  to begin data collection. Instruct the subject to immediately pinch the end of the sensor between the pads of the thumb and forefinger of his or her dominant hand, and hold for 5 s.
- Instruct the subject to switch to successive fingers every 5 s. Data collection will stop after 20 s.
- Determine the mean force applied during each pinch.
 - Click and drag the cursor over the first plateau on the graph, representing the pinch strength of the thumb and index finger (see Figure 4).
 - Click the Statistics button, , and record the mean pinch strength to the nearest 0.1 N in Table 5.
 - Move the brackets to obtain statistics for the second plateau, representing the pinch strength of the thumb and middle finger. As you move the brackets, the statistics in the Statistics box will be updated based on the data between the brackets.
 - Record the mean pinch strength to the nearest 0.1 N in Table 5.
 - Repeat this process to obtain statistics for the remaining two pinch strengths.
 - Close the Statistics box by clicking the × in the corner of the box.

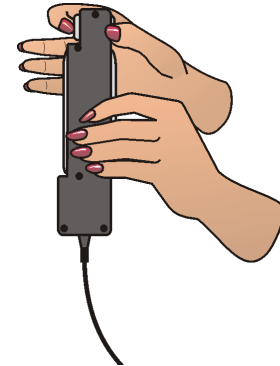


Figure 3

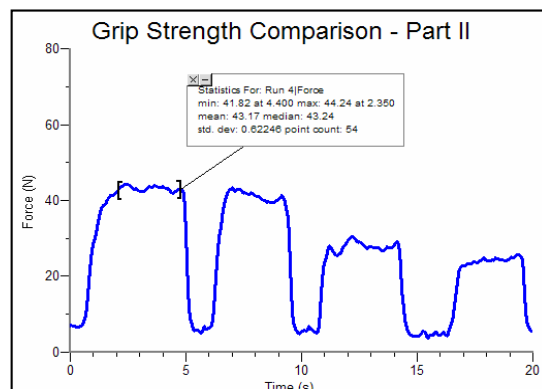


Figure 4

DATA

Table 1–Individual Grip Strength Data		
	Maximum force (N)	Mean force (N)
Right hand grip strength		
Left hand grip strength		

Table 2–Class Grip Strength Data	
	Average mean force (N)
Males (dominant hand grip strength)	
Females (dominant hand grip strength)	

Table 3–Class Grip Strength Data		
	Average mean force (N)	
	Right hand	Left hand
Right-handed individuals		
Left-handed individuals		

Table 4–Class Grip Strength Data	
Height (rounded to nearest inch)	Average mean grip strength of dominant hand (N)
1.52 m (5') or below	
1.55–1.63 m (5'1"–5'4")	
1.65–1.73 m (5'5"–5'8")	
1.75–1.83 m (5'9"–6')	
1.85 m (6'1") and above	

Table 5–Individual Pinch Strength Data	
	Mean force (N)
Dominant hand index finger	
Dominant hand middle finger	
Dominant hand ring finger	
Dominant hand little finger	

DATA ANALYSIS

1. Is there a difference in grip strength in your dominant and non-dominant hands? Are you surprised by the result?
2. Examining the data in Table 3, does there appear to be a correlation between “handedness” and grip strength? Are the results similar for right-handed and left-handed people?
3. Is there a difference between the grip strengths in the different categories of height for which data was collected in Table 4? What conclusion can you draw about the relationship between height and grip strength?
4. Does gender play a more significant role in grip strength than height? than “handedness?”
5. Using the pinch strength data in Table 5, describe the difference in strength between fingers. Where is the difference the largest?

6. List at least two possible reasons for the differences you see between the pinch strength of the first two fingers and the second two fingers. In your answer consider actions of the hand and musculature (Use an anatomy textbook or atlas to view the muscles of the forearm and hand).

EXTENSIONS

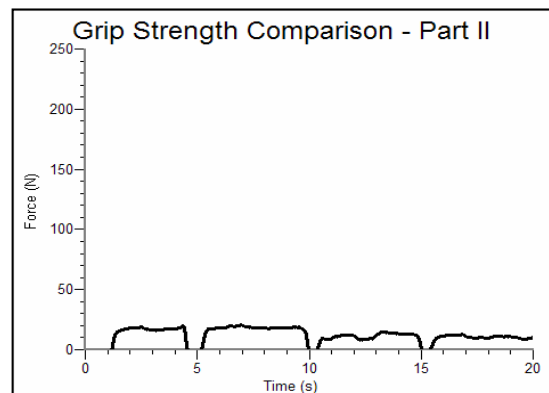
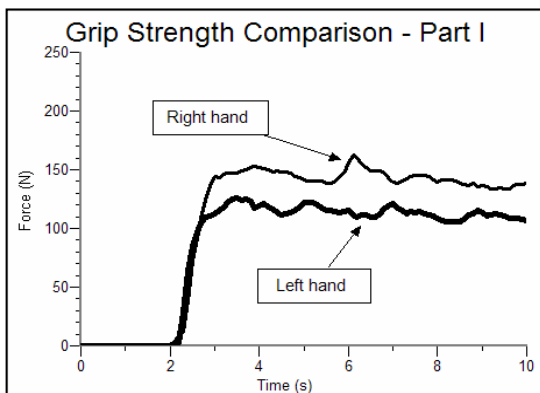
1. Plot a graph of the maximum and average grip strengths for each participant in each category. Do the results correspond with what you would expect in a human population?
2. Perform daily hand-strengthening exercises to increase your grip and/or pinch strength (such as squeezing a rubber ball). Measure your grip and/or pinch strength after two weeks and after four weeks. Compare the results with your original data.

TEACHER INFORMATION

Grip Strength Comparison

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Students should work in pairs or groups for this exercise. Because it is important that the subject not look at the data as it is being collected, one person should be responsible for data collection while the other is performing the experiment.
3. The person who is collecting the data should act as a “coach” for the subject, encouraging him/her to exert maximum effort throughout the duration of data collection.
4. Class averages are easiest to calculate if each lab group averages the data and shares it with the class. If Extension 1 is done, students will need to devise data-collection tables to collect class data. This can be done by manually entering data into the data table.

SAMPLE DATA



*Gripping strength of a female subject’s hands (Part I)
and fingers of the dominant hand (Part II)*

Table 1-Individual Grip Strength Data		
	Maximum force (N)	Mean force (N)
Right hand grip strength	162.6	145.9
Left hand grip strength	122.3	115.0

Experiment 16

Table 2–Class Grip Strength Data	
	Average mean force (N)
Males (dominant hand grip strength)	273.0
Females (dominant hand grip strength)	175.7

Table 3–Class Grip Strength Data		
	Average mean force (N)	
	Right hand	Left hand
Right-handed individuals	226.2	198.9
Left-handed individuals	204.5	215.2

Table 4–Class Grip Strength Data	
Height (rounded to nearest inch)	Average mean grip strength of dominant hand (N)
1.52 m (5') or below	
1.55–1.63 m (5'1"–5'4")	112.1
1.65–1.73 m (5'5"–5'8")	162.1
1.75–1.83 m (5'9"–6')	291.7
1.85 m (6'1") and above	483.4

Table 5–Individual Pinch Strength Data	
	Mean force (N)
Dominant hand index finger	17.2
Dominant hand middle finger	18.5
Dominant hand ring finger	11.8
Dominant hand little finger	10.8

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Right-handed students should see greater grip strength in their right hand than in their left; the corollary may not hold true for left-handed students (i.e., the left, “dominant,” hand may not be stronger than the right hand).
2. For right-handed people, grip strength is greater in the right hand. This correlation is not seen in left-handed people.
3. Height is correlated with grip strength. In most cases, the taller you are, the stronger your grip.
4. The average grip strength of females is less than the average grip strength of males. The difference is much larger than the differences related to height and handedness.
5. Answers will vary. For most students there is a significant drop in pinch strength between the middle and ring fingers. For students who have unexpected results, explore possible reasons (i.e., Do they play musical instruments, or a sport that would explain a greater finger strength than expected? Have they injured a hand in the past?).
6. Students may describe the mechanical disadvantage of the thumb reaching across the palm to meet the fourth and fifth fingers as compared with the first and second fingers without naming the muscles involved. Using an anatomy reference, students may describe attachments of the smaller muscles of the hand, including the *Opponens pollicis*, *Adductor pollicis obliquus*; *Adductor pollicis transversus* and their actions. They may also note the alignment of the *flexor digitorum profundus* and *superficialis* muscles and their mechanical advantage or disadvantage for pinching. The forefinger and middle finger are used more frequently in daily activities, leading to greater strength through exercise.

Grip Strength and Muscle Fatigue

Skeletal muscle is composed of bundles of individual muscle fibers (see Figure 1) and has unique properties which allow it to respond to stimuli by contracting. Individual muscle fibers respond to a stimulus (e.g., nerve impulse) with an all or none response, meaning the muscle fiber contracts to its maximum potential or not at all. Once a muscle has contracted, relaxation must occur before it can contract again. There are three basic types of muscle fibers: *slow fibers*, *fast fibers*, and *intermediate fibers*. Fast fibers contract quickly but for a relatively short duration. Slow fibers respond less rapidly, but are capable of a more sustained contraction. The strength of contraction of a whole muscle is dependent on the number of muscle fibers involved.

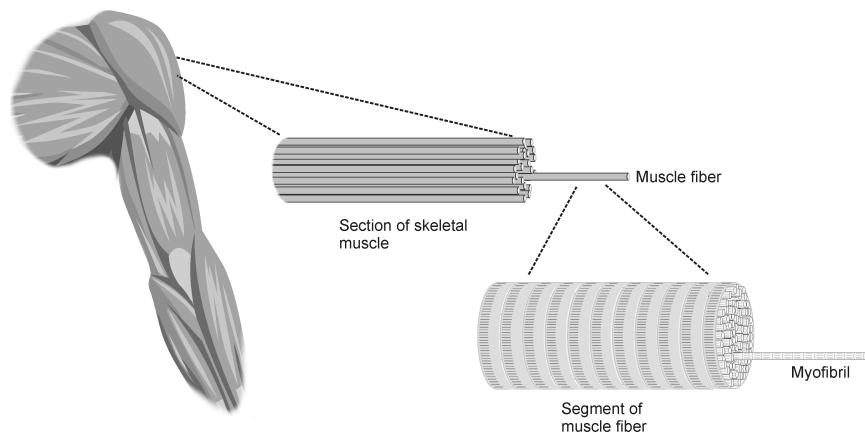


Figure 1

Muscle fatigue occurs with prolonged or repetitive use of a muscle group, and is familiar to anyone who has ever carried a heavy suitcase or walked up a long flight of stairs. With fatigue, there is a sense of weakness and even discomfort, which eventually leads one to discontinue the activity that is causing it. The mechanism of fatigue is multifactorial and not fully understood, but is felt to involve the central nervous system, peripheral nervous system, muscle units and individual muscle fibers. At the level of muscle cells, depletion of energy stores may be important.

Regular exercise improves muscular function and delays the onset of fatigue, thus increasing the amount and duration of work that can be performed. Exercise is important for optimal athletic performance, prevention of injury in athletes and non-athletes, and the maintenance of good general health.

In this experiment, you will examine the effect of fatigue on muscle action by performing sustained and repetitive isometric contractions of muscles of the arm and hand using a Vernier Hand Dynamometer.

Important: Do not attempt this experiment if you have arthritis, or other conditions of the hand, wrist, forearm, or elbow. Inform your instructor of any possible health problems that might be exacerbated if you participate in this exercise.

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of the force exerted by your hand while gripping.
- Observe the change in hand strength during a continuous grip over time.
- Observe the change in hand strength during rapid, repetitive gripping.

MATERIALS




computer
Vernier computer interface

Logger Pro
Vernier Hand Dynamometer

PROCEDURE

Select one person from your lab group to be the subject.

Part I Muscle Strength with Continuous Grip

1. Connect the Hand Dynamometer to the Vernier computer interface. Open the file “17a Grip Strength Fatigue” from the *Human Physiology with Vernier* folder.
2. Zero the readings for the Hand Dynamometer.
 - a. Hold the Hand Dynamometer along the sides, in an upright position (see Figure 2). Do not put any force on the pads of the Hand Dynamometer.
 - b. Click the Zero button, .
3. Have the subject sit with his/her back straight and feet flat on the floor. The Hand Dynamometer should be held in the dominant hand. The elbow should be at a 90° angle, with the arm unsupported (see Figure 3).
4. Have the subject close his/her eyes, or avert them from the screen.
5. Instruct the subject to grip the sensor with full strength and click  to begin data collection. The subject should exert maximum effort with each grip throughout the duration of the experiment.
6. At 90 s, the lab partner(s) should encourage the subject to grip even harder. Data will be collected for 100 s.
7. Determine the maximum force exerted during different time intervals.
 - a. Position the cursor at 0 s and click and drag to highlight 0–10 s on the graph.
 - b. Click the Statistics button, , to see the Statistics box.
 - c. Record the maximum force during the interval in Table 1, rounding to the nearest 0.1 N.
 - d. Move the brackets to highlight the 20–30 s period on the graph. As you move the brackets, the statistics in the Statistics box will be updated based on the data between the brackets.
 - e. Record the maximum force during this interval in Table 1, rounding to the nearest 0.1 N.
 - f. Repeat this process for the time intervals: 40–50 s, 60–70 s, and 80–90 s.
 - g. Close the Statistics box by clicking the × in the corner of the box.

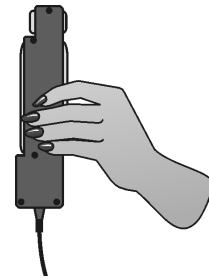







Figure 2



Figure 3

8. Calculate the difference between each maximum value and the next and record these values in Table 1.
9. Position the cursor at 0 s. Click and drag to highlight 0–90 s on the graph. Click the Linear fit button, , and record the slope (round to the nearest 0.01) in Table 3.

Part II Muscle Strength with Repetitive Grip

10. Open the file “17b Grip Strength Fatigue” from the *Human Physiology with Vernier* folder.
11. Have the subject sit with his/her back straight and feet flat on the floor. The Hand Dynamometer should be held in the dominant hand. The elbow should be at a 90° angle, with the arm unsupported (see Figure 2).
12. Have the subject close his/her eyes, or avert them from the screen.
13. Zero the readings for the Hand Dynamometer.
 - a. Hold the Hand Dynamometer along the sides, in an upright position. Do not put any force on the gray pads of the Hand Dynamometer.
 - b. Click the Zero button, .
14. Instruct the subject to rapidly grip and relax his/her grip on the sensor (approximately twice per second). Click  to begin data collection. The subject should exert maximum effort throughout the duration of the experiment.
15. At 90 s, the lab partner(s) should encourage the subject to grip even harder. Data will be collected for 100 s.
16. Determine the maximum force exerted during different time intervals.
 - a. Position the cursor at 0 s and click and drag to highlight 0–10 s on the graph.
 - b. Click the Statistics button, , and record the maximum force during that interval in Table 2, rounding to the nearest 0.1 N.
 - c. Move the brackets to highlight the 20–30 s period on the graph. As you move the brackets, the statistics in the Statistics box will be updated based on the data between the brackets.
 - d. Record the maximum force during this interval in Table 1, rounding to the nearest 0.1 N.
 - e. Repeat this process for the time intervals: 40–50 s, 60–70 s, and 80–90 s and then close the Statistics box by clicking the × in the corner of the box.
17. Calculate the difference between each maximum value and the next and record these values in Table 2.
18. Position the cursor at 0 s. Click and drag to highlight 0–90 s on the graph. Click the Linear fit button, , and record the slope in Table 3.

DATA

Table 1—Continuous Grip		
Time interval	Maximum force (N)	Δ Maximum force (N)
0–10 s		
20–30 s		
40–50 s		
60–70 s		
80–90 s		

Table 2—Repetitive Grip		
Time interval	Maximum force (N)	Δ Maximum force (N)
0–10 s		
20–30 s		
40–50 s		
60–70 s		
80–90 s		

Table 3	
	Slope
Part I—Continuous gripping	
Part II—Repetitive gripping	

DATA ANALYSIS

1. Examine your graph and the data in Table 1. What conclusion can you draw about the number of individual muscle fibers that are firing in the last 10 s compared with the first 10 s?

2. Is the change in number of muscle fibers that contract occurring at a constant rate?

3. Use your knowledge of fast, slow, and intermediate skeletal muscle fibers to hypothesize which fibers are contracting in the first, third, and final 10 s intervals.

4. How might you explain the subject's response to coaching? This should be evident in the last 10 s of data for Parts I and II of the exercise. Discuss the possible involvement of the central nervous system, in addition to the muscle fibers.

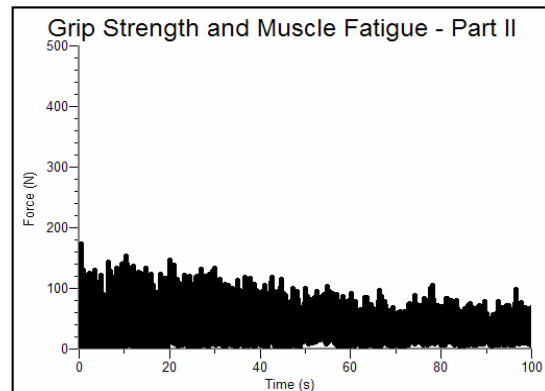
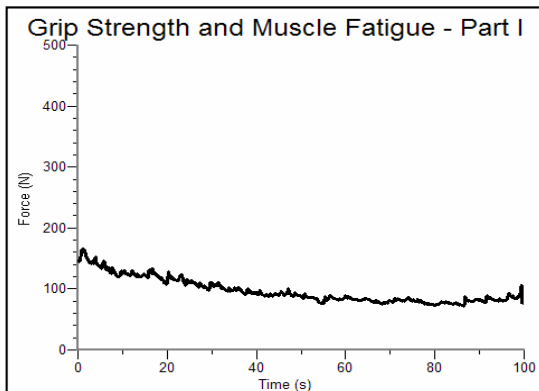
5. Compare the slopes recorded in Table 3. Give a possible explanation for the difference, if any, in muscle fatigue rates seen in continuous versus repetitive gripping.

TEACHER INFORMATION

Grip Strength and Muscle Fatigue

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Students may prefer to perform Parts I and II of this experiment on separate days to allow for full recovery of fatigued muscles.
3. Students should work in pairs or groups for this exercise. Because it is important that the subject not look at the data as they are being collected, one person should be responsible for data collection while the other is performing the experiment.
4. The person who is collecting the data should act as a “coach” for the subject, encouraging him/her to exert maximum effort throughout the duration of data collection.

SAMPLE DATA



Data showing change in grip strength over time while continually gripping (Part I) and repetitively gripping (Part II)

Table 1—Continuous Grip		
Time interval	Maximum force (N)	Δ Maximum force (N)
0–10 s	166.2	
20–30 s	128.8	-37.4
40–50 s	100.3	-28.5
60–70 s	88.3	-12.0
80–90 s	87.3	-1.0

Experiment 17

Table 2—Repetitive Grip		
Time interval	Maximum force (N)	Δ Maximum force (N)
0–10 s	172.7	
20–30 s	146.0	-26.7
40–50 s	118.3	-27.7
60–70 s	96.0	-22.3
80–90 s	84.1	-11.9

Table 3	
	Slope
Part I—Continuous Gripping	-0.75 N/s
Part II—Repetitive Gripping	-0.75 N/s

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Individual muscle fibers have an “all or none” response to stimuli. As the experiment progresses, fewer individual muscle fibers are contracting.
2. The largest change occurs in the first 10 s of the experiment; the smallest change occurs in the final 10 s of the experiment. This indicates a decrease in the drop out rate of muscle fibers over time.
3. Fast, medium, and slow muscle fibers are contributing to muscle contraction in the first 10 s of the experiment. By the final 10 s, the slow muscle fibers predominate.
4. The final 10 s of data help demonstrate that muscle fatigue is complex, involving not only the action of local muscle groups, but also the powerful central nervous system influence of desire. Coaches, fans, friends, and family use this concept to encourage athletes to reach new heights.
5. There is a greater negative slope in continuous gripping than in repetitive gripping (i.e. fatigue occurs more quickly). In repetitive gripping there are brief moments of relaxation during which there is partial recovery of the muscle fibers (calcium balance is restored, ATP is regenerated, actin and myosin fibers can return to their resting state). At the same time, recovery of the central and peripheral nerve fibers which supply the muscles is occurring, and the neuromuscular junctions are recovering.

EMG and Muscle Fatigue

Voluntary muscle contraction is the result of communication between the brain and individual muscle fibers of the musculoskeletal system. A thought is transformed into electrical impulses which travel down motor neurons (in the spine and peripheral nerves) to the neuromuscular junctions that form a motor unit (see Figure 1).

The individual muscle fibers within each motor unit contract with an “all or none” response when stimulated, meaning the muscle fiber contracts to its maximum potential or not at all. The strength of contraction of a whole muscle depends on how many individual fibers are activated, and can be correlated with electrical activity measured over the muscle with an EMG sensor.

Regular exercise is important for maintaining muscle strength and conditioning. The most common form of non-aerobic exercise is *isotonic* (weight training). In isotonic exercise, the muscle changes length against a constant force. In *isometric* exercise the length of the muscle remains the same as greater demand is placed on it. An example of this is holding a barbell (or suitcase) in one position for an extended period of time. Muscle fatigue occurs with both forms of exercise.

In this experiment, you will use a Vernier Hand Dynamometer to measure maximum grip strength and correlate this with electrical activity of the muscles involved as measured using the Vernier EKG Sensor. You will see if electrical activity changes as a muscle fatigues during continuous maximal effort. Finally, you will observe the results of a conscious effort to overcome fatigue in the muscles being tested.

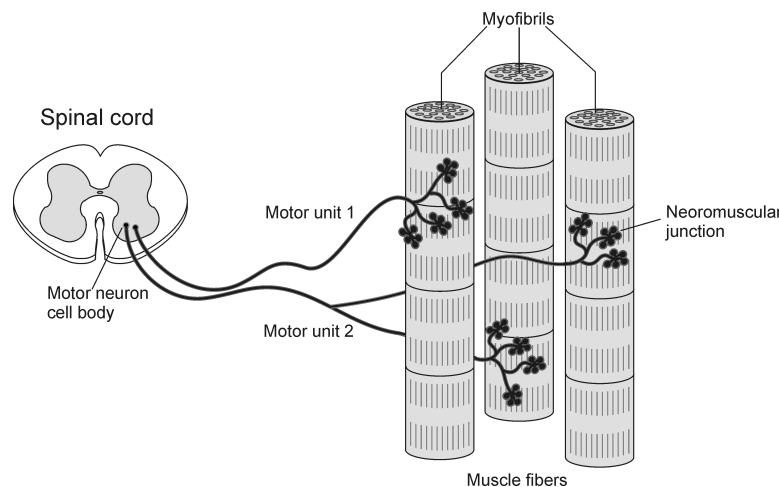


Figure 1

Important: Do not attempt this experiment if you suffer from arthritis, or other conditions of the hand, wrist, forearm, or elbow. Inform your instructor of any possible health problems that might be exacerbated if you participate in this exercise.

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of the electrical activity of a muscle.
- Correlate grip strength measurements with electrical activity data.
- Correlate measurements of grip strength and electrical activity with muscle fatigue.
- Observe the effect on grip strength of a conscious effort to overcome fatigue.

MATERIALS

computer
Vernier computer interface
Logger *Pro*

Vernier Hand Dynamometer
Vernier EKG Sensor
electrode tabs

PROCEDURE

Select one person from your lab group to be the subject.

Part I Grip Strength without Visual Feedback

1. Connect the Hand Dynamometer and the EKG Sensor to the Vernier computer interface. Open the file “18 EMG and Muscle Fatigue” from the *Human Physiology with Vernier* folder.

2. Zero the readings for the Hand Dynamometer.
 - a. Click the Zero button, .
 - b. Hold the Hand Dynamometer along the sides, in an upright position (see Figure 2). Do not put any force on the pads of the Hand Dynamometer.
 - c. Click the box in front of Hand Dynamometer to select it and click .

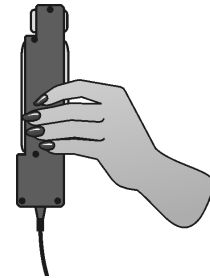


Figure 2

3. Attach three electrode tabs to on of your arms, as shown in Figure 3. Two tabs should be placed on the ventral forearm, 5 cm and 10 cm from the medial epicondyle along an imaginary line connecting the epicondyle and the middle finger.
4. Attach the green and red leads to the tabs on ventral forearm. For this activity, the green and red leads are interchangeable. Attach the black lead to the upper arm.

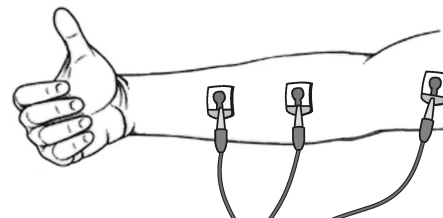







Figure 3

5. Have the subject sit with his/her back straight and feet flat on the floor. The elbow should be at a 90° angle, with the arm unsupported.
6. Have the subject close his/her eyes, or avert them from the screen.
7. Instruct the subject to grip the sensor with full strength and click to begin data collection. The subject should exert maximum effort throughout the data-collection period.
8. At 80 s, the lab partner(s) should encourage the subject to grip even harder. Data will be collected for 100 s.




9. Record statistical information about the grip strength data.
 - a. Position the cursor at 0 s on the Grip Strength graph (the top graph). Click and drag to highlight 0–20 s on the graph. Click the Statistics button, . Record the mean force during that interval in Table 1, rounding to the nearest 0.1 N.
 - b. Move the statistics brackets to highlight the time interval between 60 and 80 s on the same graph. Record the mean force during that interval in Table 1 (round to the nearest 0.1 N).
 - c. Move the statistics brackets to highlight the time interval between 80 and 100 s. Record the mean force during that interval in Table 1, rounding to the nearest 0.1 N.
10. Record statistical information about the EMG data.
 - a. Position the cursor at 0 s on the EMG graph (the bottom graph). Click and drag to highlight 0–20 s on the graph. Click the Statistics button, . Record the maximum and minimum mV during that interval in Table 1, rounding to the nearest 0.01 mV.
 - b. Move the statistics brackets to highlight the time interval between 60 and 80 s on the same graph. Record the maximum and minimum mV during that interval in Table 1, rounding to the nearest 0.01 mV.
 - c. Move the statistics brackets to highlight the time interval between 80 and 100 s on the EMG graph. Record the maximum and minimum mV during that interval in Table 1, rounding to the nearest 0.01 mV.
 - d. Calculate the difference between each minimum and maximum value and record this value in the Δ mV column in Table 1.

Part II Grip Strength with Visual Feedback

11. Have the subject sit with his/her back straight and feet flat on the floor. The Hand Dynamometer should be held in the same hand used in Part I of this experiment. Instruct the subject to position his/her elbow at a 90° angle, with the arm unsupported, and to close his/her eyes, or avert them from the screen.
12. Instruct the subject to grip the sensor with full strength and click  to begin data collection. The subject should exert near maximum effort throughout the duration of the experiment.
13. At 80 s, instruct the subject to watch the screen, and attempt to match his/her beginning grip strength (the level achieved in the first few seconds of the experiment) and to maintain this grip for the duration of the experiment. Data will be collected for 100 s.
14. Record statistical information about the grip strength data.
 - a. Position the cursor at 0 s on the Grip Strength graph (the top graph). Click and drag to highlight 0–20 s on the graph. Click on the Statistics button, . Record the mean force during that interval in Table 2, rounding to the nearest 0.1 N.
 - b. Move the statistics brackets to highlight the time interval between 60 and 80 s on the same graph. Record the mean force during that interval in Table 2, rounding to the nearest 0.1 N.
 - c. Move the statistics brackets to highlight the time interval between 80 and 100 s on the same graph. Record the mean force during that interval in Table 2, rounding to the nearest 0.1 N.

15. Record statistical information about the EMG data.
 - a. Position the cursor at 0 s on the EMG graph (the bottom graph). Click and drag to highlight 0–20 s on the graph. Click on the Statistics button, . Record the maximum and minimum mV during that interval in Table 2, rounding to the nearest 0.01 mV.
 - b. Move the statistics brackets to highlight the time interval between 60 and 80 s on the same graph. Record the maximum and minimum mV during that interval in Table 2, rounding to the nearest 0.01 mV.
 - c. Move the statistics brackets to highlight the time interval between 80 and 100 s on the same graph. Record the maximum and minimum mV during that interval in Table 2, rounding to the nearest 0.01 mV.
 - d. Calculate the difference between each minimum and maximum value and record this value in the Δ mV column in Table 2.

Part III Repetitive Grip Strength

16. Have the subject sit with his/her back straight and feet flat on the floor. The Grip Strength Sensor should be held in the same hand used in Parts I and II of this experiment. Instruct the subject to position his/her elbow at a 90° angle, with the arm unsupported, and to close his/her eyes, or avert them from the screen.
17. Instruct the subject to rapidly grip and relax his/her grip on the sensor (approximately twice per second). Click  to begin data collection. The subject should exert maximum effort throughout the duration of data collection.
18. At 80 s, the lab partner(s) should encourage the subject to grip even harder. Data will be collected for 100 s.
19. Record statistical information about the grip strength data.
 - a. Position the cursor at 0 s on the Grip Strength graph (the top graph). Click and drag to highlight 0–20 s on the graph. Click on the Statistics button, . Record the mean force during that interval in Table 3, rounding to the nearest 0.1 N.
 - b. Move the statistics brackets to highlight the time interval between 60 and 80 s on the same graph. Record the mean force during that interval in Table 3, rounding to the nearest 0.1 N.
 - c. Move the statistics brackets to highlight the time interval between 80 and 100 s on the same graph. Record the mean force during that interval in Table 3, rounding to the nearest 0.1 N.
20. Record statistical information about the EMG data.
 - a. Position the cursor at 0 s on the EMG graph (the bottom graph). Click and drag to highlight 0–20 s on the graph. Click on the Statistics button, . Record the maximum and minimum mV during that interval in Table 3, rounding to the nearest 0.01 mV.
 - b. Move the statistics brackets to highlight the time interval between 60 and 80 s on the same graph. Record the maximum and minimum mV during that interval in Table 3, rounding to the nearest 0.01 mV.
 - c. Move the statistics brackets to highlight the time interval between 80 and 100 s on the same graph. Record the maximum and minimum mV during that interval in Table 3, rounding to the nearest 0.01 mV.
 - d. Calculate the difference between each minimum and maximum value and record this value in the Δ mV column in Table 3.

DATA

Table 1–Continuous Grip Strength without Visual Feedback				
Time Interval	Mean grip strength (N)	EMG Data		
		Max (mV)	Min (mV)	Δ mV
0–20 s				
60–80 s				
80–100 s				

Table 2–Continuous Grip Strength with Visual Feedback				
Time Interval	Mean grip strength (N)	EMG data		
		Max (mV)	Min (mV)	Δ mV
0–20 s				
60–80 s				
80–100 s				

Table 3–Repetitive Grip Strength				
Time interval	Mean grip strength (N)	EMG data		
		Max (mV)	Min (mV)	Δ mV
0–20 s				
60–80 s				
80–100 s				

DATA ANALYSIS

1. Use the data in Table 1 to calculate the percent loss of grip strength that occurs between the 0–20 s and 60–80 s intervals. Describe a situation in which such a loss of grip strength is noticeable in your day-to-day life.

Computer 18

2. Use the data in Table 1 to calculate the percent change in amplitude (ΔmV) in electrical activity that occurs between the 0–20 s and 60–80 s intervals. Do the same for grip strength. What accounts for the difference in the percent change observed in grip strength and ΔmV for the two time intervals?

3. Compare mean grip strengths and ΔmV for the 0–20 s and 80–100 s in Table 1. Do your findings support or refute the practice of “coaching from the sidelines” at sporting events?

4. Use the graphs and your data from Table 1 to explain how our neuromuscular systems attempt to overcome fatigue during heavy work or exercise. How might fatigue increase the risk of musculoskeletal injury?

5. Compare the data in Tables 1 and 2. Explain any differences seen in the 80-100 s time intervals between the two tables. What does this tell you about the brain’s role in fatigue?

6. The mean grip strength is much less for repetitive gripping (Table 3) because repetitive relaxation of the hand is averaged into the calculation.
 - (a) Compare your mean grip strength during the 0–20 s and 80–100 s time intervals in Tables 1 and 3. Was there a difference in your ability to recover strength with coaching during continuous vs. repetitive gripping?

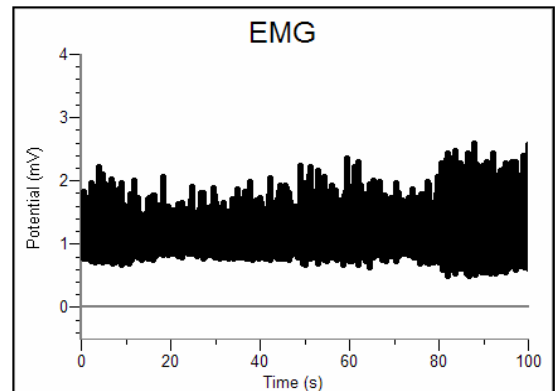
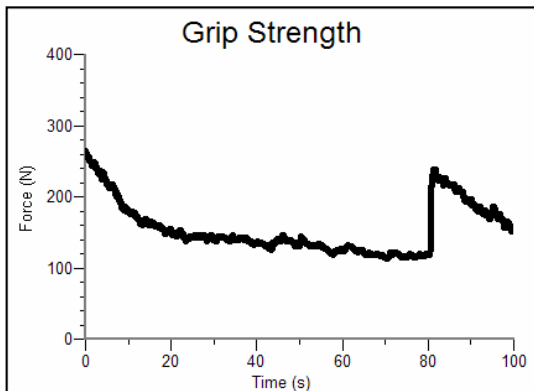
 - (b) Calculate the percent change in mean grip strength between the 1–20 s and 60–80 s time intervals in Tables 1 and 3. Do your answers support brief relaxation of muscles to delay fatigue?

TEACHER INFORMATION

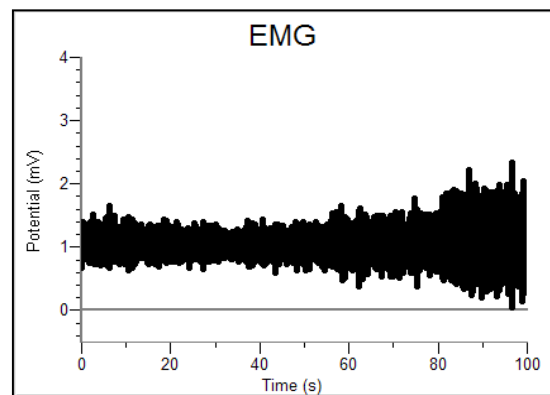
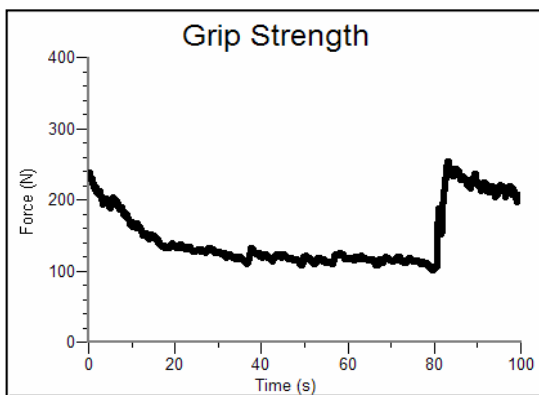
EMG and Muscle Fatigue

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. Students may prefer to perform different parts of this experiment on separate days to allow for full recovery of fatigued muscles.
3. Students should work in pairs or groups for this exercise. Because it is important that the subject not look at the data as they are being collected, one person should be responsible for data collection while the other is performing the experiment.
4. The person who is collecting the data should act as a “coach” for the subject, encouraging him/her to exert maximum effort, especially during the final 10 s of data collection.

SAMPLE RESULTS

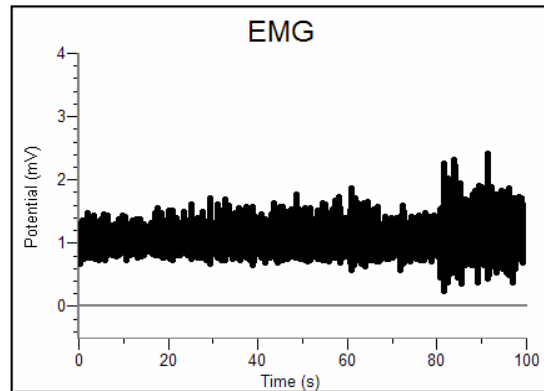
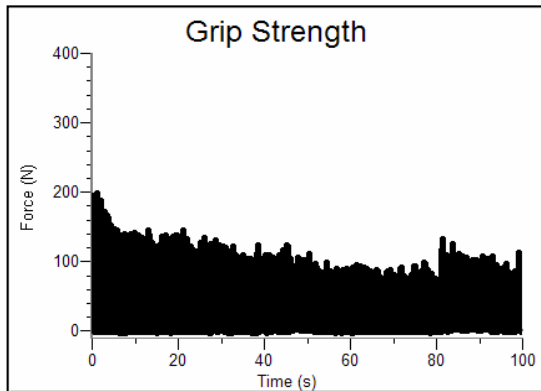


*Sample data from Part I of the student procedure:
Continuous grip strength without visual feedback*



*Sample data from Part II of the student procedure:
Continuous grip strength with visual feedback*

Experiment 18



Sample data from Part III of the student procedure:
Repetitive gripping without visual feedback

Table 1—Continuous Grip Strength without Visual Feedback				
Time interval	Mean grip strength (N)	EMG data		
		Max (mV)	Min (mV)	Δ mV
0–20 s	190.3	2.21	0.68	1.53
60–80 s	119.9	2.30	0.64	1.66
80–100 s	192.1	2.60	0.49	2.11

Table 2—Continuous Grip Strength with Visual Feedback				
Time Interval	Mean grip strength (N)	EMG data		
		Max (mV)	Min (mV)	Δ mV
0–20 s	172.0	1.65	0.64	1.01
60–80 s	113.9	1.77	0.38	1.39
80–100 s	216.8	2.33	0.05	2.28

Table 3—Repetitive Grip Strength				
Time interval	Mean grip strength (N)	EMG data		
		Max (mV)	Min (mV)	Δ mV
0–20 s	56.8	1.56	0.68	0.88
60–80 s	30.7	1.86	0.58	1.28
80–100 s	46.3	2.43	0.25	2.18

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Answers will vary. Students should see a fall in grip strength of approximately 30–40%. Daily examples include carrying a gallon of milk around a grocery store, carrying heavy backpacks or computer cases, or playing an instrument in a marching band.
2. Answers will vary. Students should report a fall in grip strength of approximately 30–40%, while ΔmV remains the same or increases by approximately 10%. Muscle fatigue accounts for this difference. The strength is less, despite unchanged or increased neuromuscular electrical activity.
3. The changes observed support our long tradition of cheering from the sidelines. Encouragement led to recovery of grip strength.
4. Despite loss of muscle power as a result of fatigue, students should see a 30–40% increase in ΔmV , which allowed them to regain mean grip strength. Tired muscles are more prone to giving way, leading to the use of less appropriate muscles. Bursts of muscle activity are less well-controlled and may cause tearing of muscle fibers or tendons.
5. Despite giving “every ounce of effort” during coaching, even greater strength was achieved with visual feedback. This indicates a central nervous system role in fatigue, mediated by visual feedback and conscious effort.
6. (a) Most students should find that mean grip strength was regained with coaching in Part I of the experiment. In Part III students will find that they fall short of regaining initial grip strength.
(b) Most students will find that grip strength fell by the same or greater proportion with repetitive gripping as with continuous gripping. This indicates that brief periods of relaxation do not restore optimal muscle function.

Lung Volumes and Capacities

Measurement of lung volumes provides a tool for understanding normal function of the lungs as well as disease states. The breathing cycle is initiated by expansion of the chest. Contraction of the diaphragm causes it to flatten downward. If chest muscles are used, the ribs expand outward. The resulting increase in chest volume creates a negative pressure that draws air in through the nose and mouth. Normal exhalation is passive, resulting from “recoil” of the chest wall, diaphragm, and lung tissue.

In normal breathing at rest, approximately one-tenth of the total lung capacity is used. Greater amounts are used as needed (i.e., with exercise). The following terms are used to describe lung volumes (see Figure 1):

<i>Tidal Volume (TV):</i>	The volume of air breathed in and out without conscious effort
<i>Inspiratory Reserve Volume (IRV):</i>	The additional volume of air that can be inhaled with maximum effort after a normal inspiration
<i>Expiratory Reserve Volume (ERV):</i>	The additional volume of air that can be forcibly exhaled after normal exhalation
<i>Vital Capacity (VC):</i>	The total volume of air that can be exhaled after a maximum inhalation: $VC = TV + IRV + ERV$
<i>Residual Volume (RV):</i>	The volume of air remaining in the lungs after maximum exhalation (the lungs can never be completely emptied)
<i>Total Lung Capacity (TLC):</i>	$= VC + RV$
<i>Minute Ventilation:</i>	The volume of air breathed in 1 minute: $(TV)(\text{breaths/minute})$

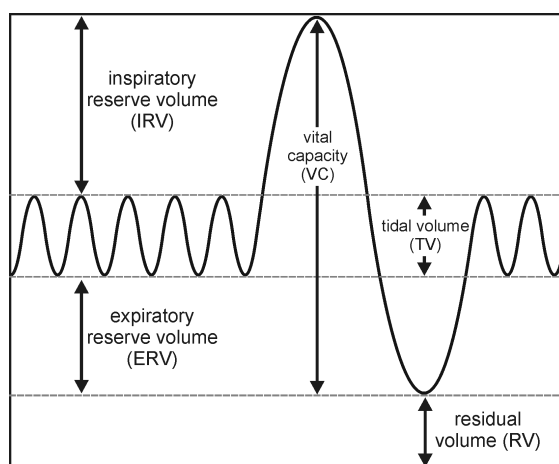


Figure 1

In this experiment, you will measure lung volumes during normal breathing and with maximum effort. You will correlate lung volumes with a variety of clinical scenarios.

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of lung capacities and volumes.
- Compare lung volumes between males and females.
- Correlate lung volumes with clinical conditions.

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier Spirometer

disposable mouthpiece
disposable bacterial filter
nose clip

PROCEDURE

Important: Do not attempt this experiment if you are currently suffering from a respiratory ailment such as the cold or flu.

1. Connect the Spirometer to the Vernier computer interface. Open the file “19 Lung Volumes” from the *Human Physiology with Vernier* folder.
2. Attach the larger diameter side of a bacterial filter to the “Inlet” side of the Spirometer. Attach a gray disposable mouthpiece to the other end of the bacterial filter (see Figure 2).

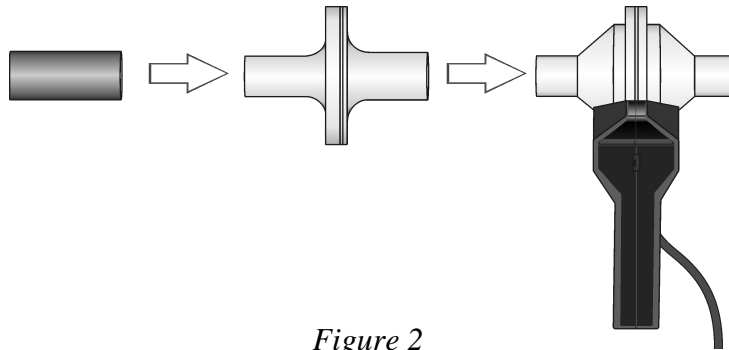



Figure 2

3. Hold the Spirometer in one or both hands. Brace your arm(s) against a solid surface, such as a table, and click to zero the sensor. **Note:** The Spirometer must be held straight up and down, as in Figure 2, and not moved during data collection.
4. Collect inhalation and exhalation data.
 - a. Put on the nose plug.
 - b. Click to begin data collection.
 - c. Taking normal breaths, begin data collection with an inhalation and continue to breathe in and out. After 4 cycles of normal inspirations and expirations fill your lungs as deeply as possible (maximum inspiration) and exhale as fully as possible (maximum expiration). *It is essential that maximum effort be expended when performing tests of lung volumes.*
 - d. Follow this with at least one additional recovery breath.
5. Click to end data collection.

- Click the Next Page button, , to see the lung volume data. If the baseline on your graph has drifted, use the Baseline Adjustment feature to bring the baseline volumes closer to zero, as in Figure 3.
- Select a representative peak and valley in the Tidal Volume portion of your graph. Place the cursor on the peak and click and drag down to the valley that follows it. Enter the Δy value displayed in the lower left corner of the graph to the nearest 0.1 L as Tidal Volume in Table 1.

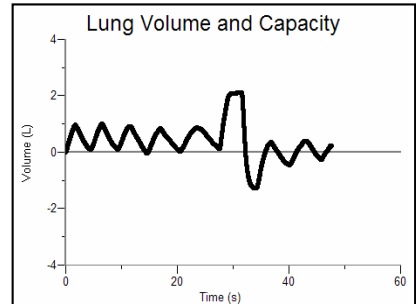


Figure 3

- Move the cursor to the peak that represents your maximum inspiration. Click and drag down the side of the peak until you reach the level of the peaks graphed during normal breathing. Enter the Δy value displayed in the lower left corner of the graph to the nearest 0.1 L as Inspiratory Reserve Volume in Table 1.
- Move the cursor to the valley that represents your maximum expiration. Click and drag up the side of the peak until you reach the level of the valleys graphed during normal breathing. Enter the Δy value displayed in the lower left corner of the graph to the nearest 0.1 L as Expiratory Reserve Volume in Table 1.
- Calculate the Vital Capacity and enter the total to the nearest 0.1 L in Table 1.

$$VC = TV + IRV + ERV$$

- Calculate the Total Lung Capacity and enter the total to the nearest 0.1 L in Table 1. (Use the value of 1.5 L for the RV.)

$$TLC = VC + RV$$

- Share your data with your classmates and complete the Class Average columns in Table 1.

DATA

Table 1			
Volume measurement (L)	Individual (L)	Class average (Male) (L)	Class average (Female) (L)
Tidal Volume (TV)			
Inspiratory Reserve (IRV)			
Expiratory Reserve (ERV)			
Vital Capacity (VC)			
Residual Volume (RV)	≈1.5	≈1.5	≈1.5
Total Lung Capacity (TLC)			

DATA ANALYSIS

1. What was your Tidal Volume (TV)? What would you expect your TV to be if you inhaled a foreign object which completely obstructed your right mainstem bronchus?

2. Describe the difference between lung volumes for males and females. What might account for this?

3. Calculate your Minute Volume at rest.

$$(\text{TV} \times \text{breaths/minute}) = \text{Minute Volume at rest}$$

If you are taking shallow breaths (TV = 0.20 L) to avoid severe pain from rib fractures, what respiratory rate will be required to achieve the same minute volume?

4. Exposure to occupational hazards such as coal dust, silica dust, and asbestos may lead to *fibrosis*, or scarring of lung tissue. With this condition, the lungs become stiff and have more “recoil.” What would happen to TLC and VC under these conditions?

5. In severe emphysema there is destruction of lung tissue and reduced recoil. What would you expect to happen to TLC and VC?

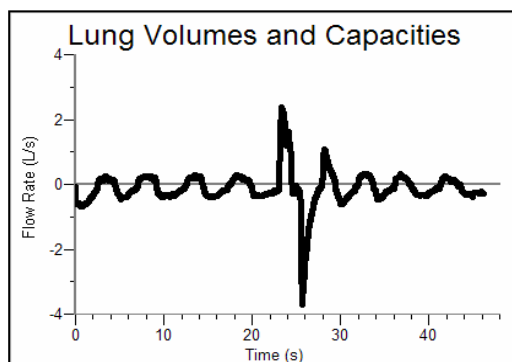
6. What would you expect to happen to your Expiratory Reserve Volume when you are treading water in a lake?

EXTENSION

Repeat the experiment with the chest or abdomen constricted (use a girdle or ace bandage).

TEACHER INFORMATION**Lung Volumes and Capacities**

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. The Spirometer should be held vertically and steadily. It is helpful to brace at least one arm on a hard surface, such as a table. Deviation from this position will affect data collection.
3. To maintain good hygiene, use the Spirometer with a MicroGard[®] bacterial filter and a disposable mouthpiece. In an ideal situation, every student would have his or her own bacterial filter and disposable mouthpiece. However, budget restraints may require you to share the bacterial filter among a few students. According to the manufacturer's specs, the MicroGard filters out more than 99% of bacterial and viral aerosols.
4. It is important for students to begin data collection with an inspiration as this is the trigger for data collection to begin.
5. The student procedure directs students to perform a baseline adjustment if their data drifted. The Baseline Adjustment feature was not present in the initial release of LabQuest App. It will be present in later versions of LabQuest App.
6. Residual volume cannot be measured. A value of ≈ 1.5 L is provided for the purposes of this experiment.
7. Expected ranges in Table 1 were gathered with clinically calibrated equipment and with professional coaching. Values in the classroom may vary.

SAMPLE DATA

Flow rate data representing regular breathing, a large inhale and exhale, and recovery

Experiment 19

Table 1				
Volume measurement (L)	Expected range (L)	Individual (L)	Class average (Male) (L)	Class average (Female) (L)
Tidal Volume (TV)	0.4–0.5	1.0		
Inspiratory Reserve (IRV)	2.5–3.5	1.3		
Expiratory Reserve (ERV)	1.0–2.0	1.3		
Vital Capacity (VC)	4.5–6.0	3.6		
Residual Volume (RV)	≈1.5	≈1.5	≈1.5	≈1.5
Total lung Capacity (TLC)	5.0–7.0	5.1		

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. Tidal volume answers will vary. If the right mainstem bronchus was totally obstructed, the TV would be reduced by approximately half.
2. Data should reveal that males have larger lung capacities than females. On average, male lungs are larger than female lungs, even when controlling for height.
3. Answers will vary. Using the sample data provided:

$$\text{Minute volume} = (0.5 \text{ L})(18 \text{ breaths/ minute}) = 9 \text{ L/minute}$$

If the tidal volume is 0.2 L, use the following equation to solve for respiratory rate:

$$(0.2 \text{ L})(x) = 9 \text{ L/minute}$$

$$x = 45 \text{ breaths/minute}$$

4. Total lung capacity and vital capacity are both reduced in conditions that result in an increase in stiffness or recoil of the lungs.
5. Total lung capacity and vital capacity are both increased in severe emphysema, where a loss of lung tissue leads to a reduction in stiffness and recoil.
6. Expiratory reserve volume is dependent on passive forces acting to compress the chest. When immersed in water there is a greater pressure exerted on the chest, causing a slightly greater expulsion of air with each exhalation. The expiratory reserve volume is reduced.

Respiratory Response to Physiologic Challenges

The respiratory cycle of inspiration and expiration is controlled by complex mechanisms involving neurons in the cerebral cortex, brain stem, and peripheral nervous system, as well as central and peripheral receptors. These receptors respond to a variety of stimuli including chemicals and pressure. Central respiratory control (*respiratory drive*) occurs in the pons and medulla, which respond directly to chemical influences. Other input is received from stretch receptors in the lungs and chemoreceptors located in the carotid and aortic bodies (see Figure 1). The chemoreceptors respond most sensitively and rapidly to carbon dioxide but also to oxygen and pH (acidity). Constant adjustments in the respiratory cycle occur throughout the day to allow gas exchange in the lungs to maintain a steady level of CO₂ in the bloodstream. An increase in the CO₂ level stimulates breathing, while a decrease inhibits it. If the deviation from the “set point” is large enough you may experience shortness of breath. The oxygen level can also influence the respiratory cycle, but larger deviations are required before its influence is felt.

At rest, the average adult male produces approximately 200 mL of CO₂ each minute, but this may increase to over 2000 mL with exercise or heavy work. Hyperventilation lowers CO₂ levels due to an increased opportunity for gas exchange in the lungs. Holding one’s breath or re-breathing air (such as breathing into a paper bag) raises CO₂ levels because there is less opportunity for gas exchange.

In this experiment, you will alter CO₂ levels by holding your breath (*hypoventilation*), rapid breathing (*hyperventilation*), and exercise. You will compare the respiratory rate, tidal volume, and minute ventilation that result from each physiologic challenge to homeostasis.

Important: Do not attempt this experiment if you are currently suffering from a respiratory ailment such as the cold or flu.

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of normal tidal volume.
- Compare tidal volumes generated by various physiologic challenges.
- Correlate your findings with real-life situations.

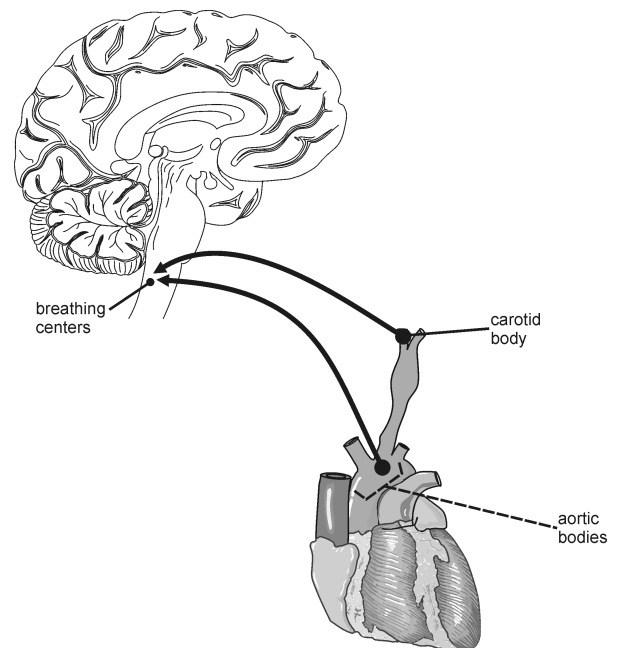


Figure 1

MATERIALS

computer
Vernier computer interface
Logger Pro
Vernier Spirometer

disposable mouthpiece
disposable bacterial filter
nose clip

PROCEDURE

Part I Tidal Volume Response to Breath Holding

1. Connect the Spirometer to the Vernier computer interface. Open the file “20 Respiratory Response” from the *Human Physiology with Vernier* folder.
2. Attach the larger diameter side of the disposable bacterial filter to the “Inlet” side of the Spirometer head. Attach a disposable Spirometer mouthpiece to the other end of the bacterial filter (Figure 2).

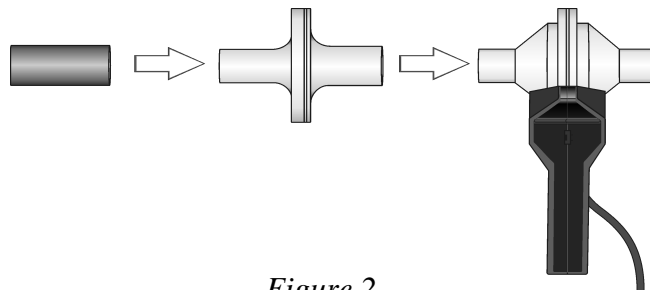


Figure 2

3. Hold the Spirometer in one or both hands. Brace your arm(s) against a solid surface, such as a table, and click to zero the sensor. **Note:** The Spirometer must be held straight up and down (as in Figure 2) during data collection.
4. Collect inhalation and exhalation data.
 - a. Put on the nose clip.
 - b. Click to begin data collection.
 - c. Taking normal breaths, begin data collection with an inhalation and continue to breathe in and out. After 4 cycles of normal inspirations and expirations fill your lungs as deeply as possible (maximum inspiration) and hold your breath for 40 s.
 - d. After 40 s of breath holding, resume normal breathing. Data will be collected for 120 s.
5. Click the Next Page button, , to see the volume data. If the baseline on your volume graph has drifted, use the Baseline Adjustment feature to bring the baseline volumes closer to zero, as in Figure 3.

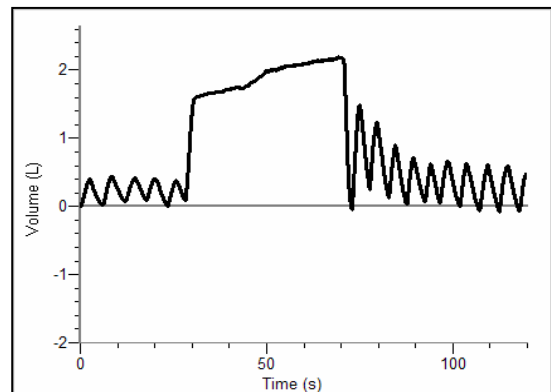


Figure 3

6. Select a representative peak and valley in the portion of your graph prior to the onset of breath holding. Place the cursor on the peak and click and drag down to the valley that follows it. Enter the Δy value displayed in the lower left corner of the graph to the nearest 0.1 L as the Before Challenge Tidal Volume in Table 1.
7. Select two adjacent peaks in the portion of your graph prior to the onset of breath holding. Click and drag the cursor from one peak to the next. Use the Δx value displayed in the lower left corner of the graph to calculate the respiratory rate in breaths/minute. Enter this value to the nearest 0.1 breaths/min as the Before Challenge Respiratory Rate in Table 1.
8. Repeat Steps 6 and 7, selecting regions in the portion of your graph after normal breathing had been resumed (between 60–80 s). Enter the values in the After Challenge section in Table 1.
9. Calculate the Minute Ventilation values for before and after the challenge and enter the results to the nearest 0.1 L in Table 1.

$$(\text{Tidal Volume})(\text{Respiration Rate}) = \text{Minute Ventilation}$$

Part II Tidal Volume Response to Rapid Breathing

10. Clear the data from Part I by choosing Clear All Data from the Data menu.
11. Hold the Spirometer in one or both hands. Brace your arm(s) against a solid surface, such as a table, and click . **Note:** The Spirometer must be held straight up and down (see Figure 2) during data collection.
12. Collect inhalation and exhalation data.
 - a. Put on the nose plug.
 - b. Click to begin data collection.
 - c. Taking normal breaths, begin data collection with an inhalation and continue to breathe in and out. After 4 cycles of normal inspirations and expirations, begin breathing deeply and rapidly for 40 s. **Note:** If you begin to feel faint, slow your breathing rate.
 - d. After 40 s of rapid breathing, resume normal breathing. Data will be collected for 120 s.
13. Click the Next Page button, , to see the volume data. If the baseline on your graph has drifted, use the Baseline Adjustment feature to bring the baseline volumes closer to zero.
14. Select a representative peak and valley in the portion of your graph prior to the onset of rapid breathing. Place the cursor on the peak and click and drag down to the valley that follows it. Enter the Δy value displayed in the lower left corner of the graph to the nearest 0.1 L as Before Challenge Tidal Volume in Table 1.
15. Select two adjacent peaks in the portion of your graph prior to the onset of rapid breathing. Click and drag the cursor from one peak to the next. Use the Δx value displayed in the lower left corner of the graph to calculate the respiratory rate in breaths/minute. Enter this value to the nearest 0.1 breaths/min as Before Challenge Respiratory Rate in Table 1.
16. Repeat Steps 14 and 15, selecting regions in the portion of your graph after normal breathing had been resumed (between 60–80 s). Enter the values in the After Challenge section in Table 1.
17. Calculate the Minute Ventilation values for before and after the challenge and enter the results to the nearest 0.1 L in Table 1. Return to Page 1 to prepare for Part III.

Part III Tidal Volume Response to Exercise

18. Clear the data from Part II by choosing Clear All Data from the Data menu.
19. Hold the Spirometer in one or both hands and click . **Note:** The Spirometer must be held straight up and down (see Figure 2) during data collection.
20. Collect inhalation and exhalation data.
 - a. Put on the nose plug.
 - b. Click to begin data collection.
 - c. Taking normal breaths, begin data collection with an inhalation and continue to breathe in and out. After 4 cycles of normal inspirations and expirations begin running in place for 40 s.
 - d. After 40 s of running in place, stand quietly. Continue to breathe into the Spirometer. Data will be collected for 120 s.
21. Click the Next Page button, , to see the volume data. If the baseline on your graph has drifted, use the Baseline Adjustment feature to bring the baseline volumes closer to zero.
22. Select a representative peak and valley in the portion of your graph prior to the onset of exercise. Place the cursor on the peak and click and drag down to the valley that follows it. Enter the Δy value displayed in the lower left corner of the graph to the nearest 0.1 L as Before Challenge Tidal Volume in Table 2.
23. Select two adjacent peaks in the portion of your graph prior to the onset of exercise. Click and drag the cursor from one peak to the next. Use the Δx value displayed in the lower left corner of the graph to calculate the respiratory rate in breaths/minute. Enter this value to the nearest 0.1 breaths/min as Initial Respiratory Rate in Table 2.
24. Repeat Steps 22 and 23 two times. The first time, select regions in the portion of your graph during the challenge and the second time select regions after normal breathing had been resumed. Enter the values in the During and After Challenge sections in Table 2.
25. Calculate the Minute Ventilation values for before, during, and after the challenge and enter the results to the nearest 0.1 L in Table 2.

DATA

Table 1		
	Holding Breath	Rapid Breathing
Before Challenge		
Tidal volume (L)		
Respiratory rate (breaths/min)		
Initial minute ventilation (L/min)		
After Challenge		
Tidal volume (L)		
Respiratory rate (breaths/min)		
Minute ventilation (L/min)		

Table 2—Exercise	
Before Challenge	
Tidal volume (L)	
Respiratory rate (breaths/min)	
Minute ventilation (L/min)	
During Challenge	
Tidal volume (L)	
Respiratory rate (breaths/min)	
Minute ventilation (L/min)	
After Challenge	
Tidal volume (L)	
Respiratory rate (breaths/min)	
Minute ventilation (L/min)	

DATA ANALYSIS

1. Describe the changes in respiratory rates, tidal volumes, and minute ventilations that occurred after each of the following physiologic challenges in terms of CO₂ levels and their effect on respiratory drive:
 - (a) breath holding
 - (b) rapid breathing
 - (c) exercise
2. Which challenge caused the greatest change in respiratory rate (pre-challenge vs. post challenge)? Tidal volume? Minute ventilation? Did respiratory rate or tidal volume change the most relative to its resting value?
3. How might breathing into a paper bag help someone who is extremely anxious and hyperventilating?
4. Some patients with severe emphysema have constant high levels of CO₂ because of inadequate ventilation. The central nervous system breathing center in these patients becomes insensitive to CO₂ and more dependent on the level of O₂, which is low. These patients are said to have “oxygen-dependent respiratory drive.” What might happen if you give such a person high levels of supplemental O₂?
5. Would breathing pure O₂ help the air hunger experienced by athletes who have just completed a race? Why or why not?

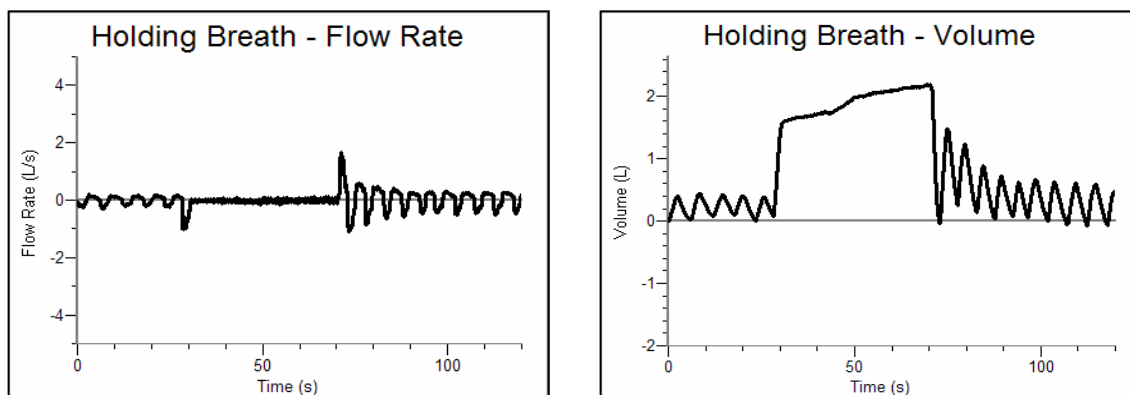
EXTENSION

Repeat the experiment, re-breathing (breathing in and out of a paper bag) from 20 to 60 s.

TEACHER INFORMATION

Respiratory Response to Physiologic Challenges

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. The Spirometer should be held vertically and steadily. It may be helpful to brace at least one arm on a hard surface, such as a table, when not exercising.
3. To maintain good hygiene, use the Spirometer with a MicroGard[®] bacterial filter and a disposable mouthpiece. In an ideal situation, every student would have his or her own bacterial filter and disposable mouthpiece. However, budget restraints may require you to share the bacterial filter among a few students. According to the manufacturer's specs, the MicroGard filters out more than 99% of bacterial and viral aerosols.
4. It is important for students to begin data collection with an inspiration, as this is the trigger for data collection to begin.
5. The student procedure directs students to perform a baseline adjustment if their data drifted. The Baseline Adjustment feature was not present in the initial release of LabQuest App. It will be present in later versions of LabQuest App. It is not necessary to have a perfectly level volume graph to obtain the data required for this experiment.

SAMPLE DATA

Sample flow rate and volume data for the first challenge of this activity

Experiment 20

Table 1		
	Holding Breath	Rapid Breathing
Before Challenge		
Tidal volume (L)	0.6	0.5
Respiratory rate (breaths/min)	12.5	14.3
Initial minute ventilation (L/min)	7.5	7.2
Post Challenge		
Tidal volume (L)	1.3	0.7
Respiratory rate (breaths/min)	19.4	11.5
Minute ventilation (L/min)	25.2	8.1

Table 2—Exercise	
Before Challenge	
Tidal volume (L)	0.6
Respiratory rate (breaths/min)	13.6
Minute ventilation (L/min)	8.2
During Challenge	
Tidal volume (L)	2.4
Respiratory rate (breaths/min)	24.0
Minute ventilation (L/min)	58.3
After Challenge	
Tidal volume (L)	1.0
Respiratory rate (breaths/min)	18.8
Minute ventilation (L/min)	18.8

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. (a) Breath holding results in a build-up of CO₂, which causes a subsequent increase in respiratory rate, tidal volume, and minute ventilation.
(b) Rapid breathing results in a reduction of CO₂, also referred to as *hyperventilation*, which causes a subsequent decrease in respiratory rate. Students may or may not see decreases in tidal volume and minute ventilation, depending on how rapidly they breathed.
(c) Exercise causes a marked increase in production of CO₂ by active muscles. This results in an increase in tidal volume, respiratory rate, and minute ventilation both during and immediately subsequent to exercise.
2. Answers will vary. In the sample data presented above, breath holding caused the greatest changes in respiratory rate, tidal volume and minute ventilation. Tidal volume increased 117% and respiratory rate increased by 56%.
3. Anxiety may stimulate the respiratory center directly, leading to an increase in respiratory rate and a fall in CO₂ level. This may result in dizziness, weakness, numbness, and tingling. Breathing in and out of a paper bag will raise the CO₂ level and help to alleviate these symptoms.
4. High levels of oxygen may be dangerous for people who have an oxygen-dependent respiratory drive. In these patients the urge to breathe is triggered by low levels of oxygen. Raising the oxygen levels too high will result in dangerous slowing of the respiratory rate. For this reason, patients with severe lung disease are usually instructed to keep their oxygen tanks set to 2 L/min or less.
5. The shortness of breath experienced during and after exertion is the result of increased metabolism of active muscles, the main product of which is CO₂. There is no shortage of O₂, and no physiologic benefit gained by adding it.

Analysis of Lung Function

Spirometry is a valuable tool for analyzing the flow rate of air passing into and out of the lungs. Flow rates vary over the course of a *respiratory cycle* (a single inspiration followed by a single expiration) and are dependent upon a variety of factors. Maximal inspiration results from contraction of the diaphragm downward and the movement of the ribs upward and outward, both of which expand the chest cavity. Forced expiration is the result of the rapid contraction of chest and abdominal muscles, as well as the relaxation of the diaphragm.

Lung flow rates are graphed as a *flow volume loop*, in which air flow is graphed as a function of volume. Because volume (graphed on the x axis) is increased with inspiration and decreased with expiration, the completed graph forms a loop (see Figure 1). As is demonstrated in the flow volume loop, in normal lungs air is rapidly expelled early in the process of forced expiration, with a tapering of flow rate as the lungs are emptying. This latter part of the expiration curve represents the emptying of small airways (*terminal bronchioles*). Inspiration shows a different pattern, with the maximum air flow occurring half-way through the cycle.

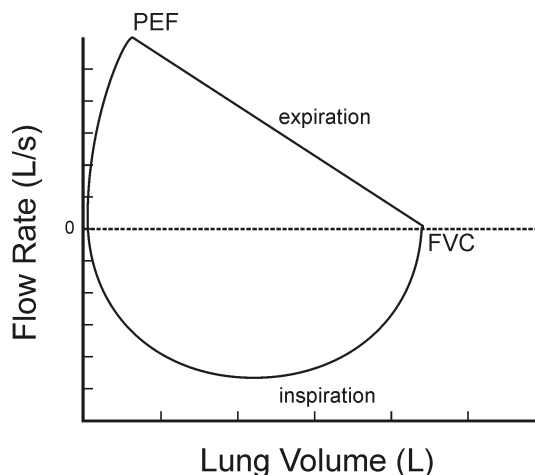


Figure 1

Information about disease states can be obtained through examination of flow volume loop graphs. Data that can be obtained or calculated from the graph include:

- *forced vital capacity* (FVC), the total exhaled air, from maximum inhalation to maximum exhalation;
- *forced expiratory volume* (FEV_1), the volume of air expelled in the first second of a forced exhalation;
- FEV_1/FVC , measured as a percentage;
- *peak expiratory flow* (PEF), the highest point on the exhalation graph.

Changes from expected values can be used to diagnose a variety of pulmonary conditions and to follow their progress over time.

Important: Do not attempt this experiment if you are currently suffering from a respiratory ailment such as the cold or flu.

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of a flow volume loop.
- Find the forced expiratory volume at 1 s (FEV_1) and the forced vital capacity (FVC).
- Calculate FEV_1/FVC .
- Find the peak expiratory flow rate (PEF).
- Create flow volume loops for several clinical scenarios.

MATERIALS

computer
Vernier computer interface
Vernier Spirometer
Logger *Pro*

disposable mouthpiece
disposable bacterial filter
nose clip

PROCEDURE

Select one person from your lab group to be the subject.

1. Connect the Spirometer to the Vernier computer interface. Open the file “21 Analyze Lung Function” from the *Human Physiology with Vernier* folder.
2. Attach the larger diameter side of a white bacterial filter to the side of the Spirometer head marked “Inlet.” Attach a disposable mouthpiece to the other end of the bacterial filter (see Figure 2).

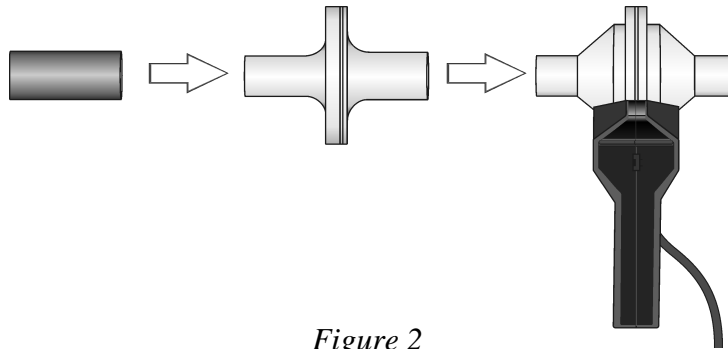




Figure 2

3. Hold the Spirometer in one or both hands. Brace your arm(s) against a solid surface, such as a table, and click . **Note:** The Spirometer must be held straight up and down (see Figure 2) and not moved during data collection.
4. Collect exhalation and inhalation data.
 - a. Put on the nose clip and put your mouth against the mouthpiece.
 - b. Click to begin data collection.
 - c. Take the deepest breath possible and exhale forcefully as quickly as possible through the Spirometer mouthpiece. **Important:** Force ALL of the air out of your lungs. Your exhalation will trigger data collection. Without removing your mouth from the mouthpiece, inhale as deeply as possible.

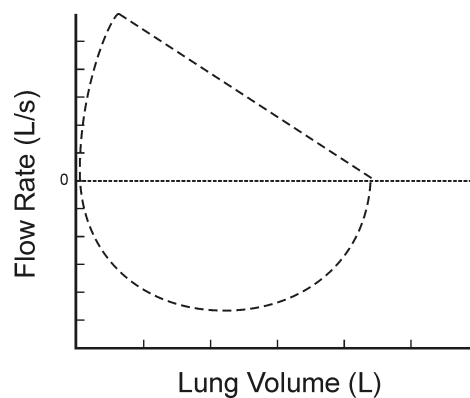
5. If your data does not approximate Figure 1, repeat Step 4. It is helpful to watch the screen while performing this test. **Important:** *It is essential that maximum effort be expended when performing tests of air flow.*
6. Find FEV₁ by scrolling down the data table to 1.00 s. Scroll to the right to see the volume. Record this value to the nearest 0.01 L in the data table.
7. Click the Examine button, . Move the examine line to the rightmost point of the flow volume loop. This is the FVC. Record the volume value to the nearest 0.01 L in the data table. Close the Statistics box by clicking the X in the corner of the box.
8. Find FEV₁/FVC, multiply by 100, and enter the calculated value in the data table.
9. Click the Statistics button, . Enter the maximum flow rate value to the nearest 0.01 L/s as *peak expiratory flow rate (PEF)* in the data table.

DATA

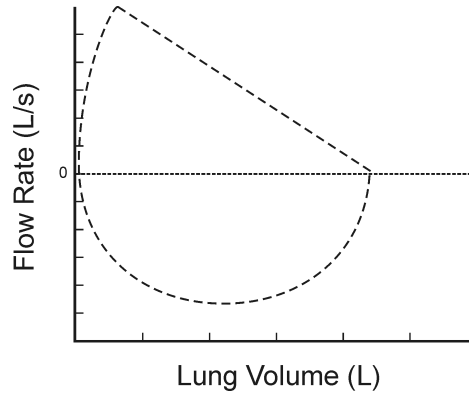
FEV ₁ (L)	FVC (L)	FEV ₁ /FVC (%)	PEF (L/s)

DATA ANALYSIS

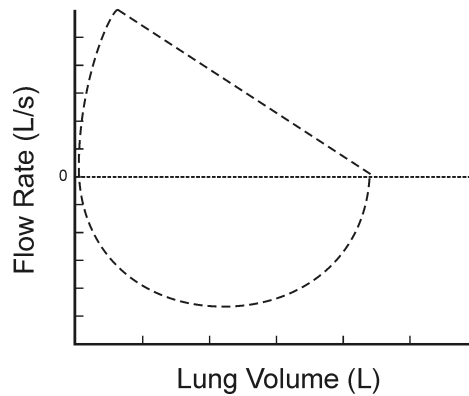
1. Using the flow volume loops pictured below as guides, superimpose loops for the following scenarios. In your answer consider whether the problem affects inspiration, expiration, or both; whether it is likely to affect the first half of the curve, the second half of the curve, or both. (Hint: It may be helpful to begin by estimating the maximum expected total lung capacity for each situation and drawing this point on the graph first.)
 - a. There is a narrowing of the trachea, causing blockage in this large airway.



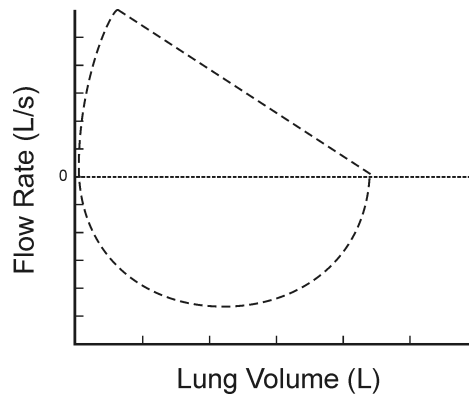
- b. A small grape is lodged in the right mainstem bronchus, completely blocking off the right lung.



- c. Emphysema reduces all flow rates, but has a greater effect on small airways, many of which are lost to the disease. (Acute asthma may show similar changes because of mucous obstruction of these same small airways.)



- d. Severe osteoporosis may result in *kyphoscoliosis*, in which there is a curvature of the upper spine that limits the normal ability of the ribs to expand with inspiration.



2. Lung disease is often divided into two broad categories: obstructive disease and restrictive disease. Examples of obstructive disease are emphysema, chronic bronchitis, and asthma. Examples of restrictive disease are abnormalities of the spine and chest and diseases within the lungs that make them less elastic (“stiffer”), such as pulmonary fibrosis. Calculate the FEV_1/FVC in the table below for obstructive and restrictive disease and compare to your values. How might these values be helpful diagnostically?

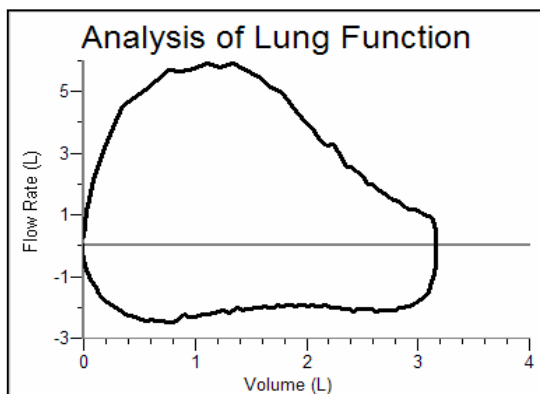
Volume (L)	Normal (your data)	Obstructive disease	Restrictive disease
FVC (L)		4	4
FEV_1 (L)		1.8	3.5
FEV_1/FVC (%)			

TEACHER INFORMATION

Analysis of Lung Function

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. It is extremely important that students exert maximum effort in this test. Anything less will result in data that are inaccurate. In order to insure that this occurs, it is helpful to coach and encourage the subject during both exhalation and inhalation. It may be necessary to perform the test several times to obtain optimum data and a normal-appearing flow volume loop.
3. Have the subject watch the screen while performing the test. This will help him/her to perform to maximum capacity.
4. The Spirometer should be held vertically and steadily. It is helpful to brace at least one arm on a hard surface, such as a table. Deviation from this position will affect data collection.
5. To maintain good hygiene, use the Spirometer with a MicroGard[®] bacterial filter and a disposable mouthpiece. In an ideal situation, every student would have his or her own bacterial filter and disposable mouthpiece. However, budget restraints may require you to share the bacterial filter among a few students. According to the manufacturer's specs, the MicroGard filters out more than 99% of bacterial and viral aerosols.

SAMPLE DATA

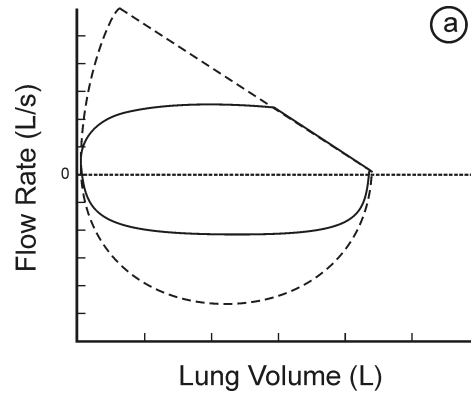


Sample flow volume loop

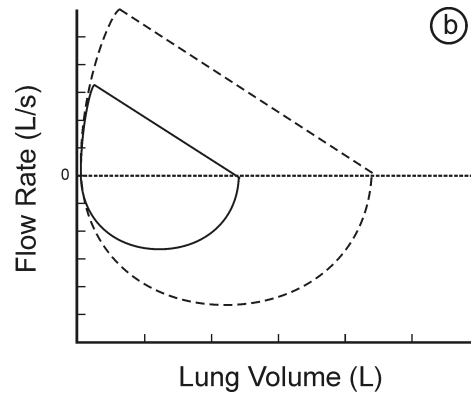
Table 1			
FEV ₁ (L)	FVC (L)	FEV ₁ /FVC (%)	PEF (L/s)
3.04	3.17	96	5.93

ANSWERS TO THE DATA ANALYSIS QUESTIONS

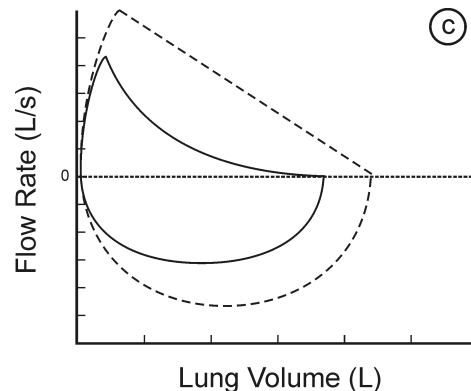
1. a. Upper airway obstruction impairs both inhalation and exhalation and therefore flattens both the upper and lower ends of the loop. It does not affect the vital capacity.



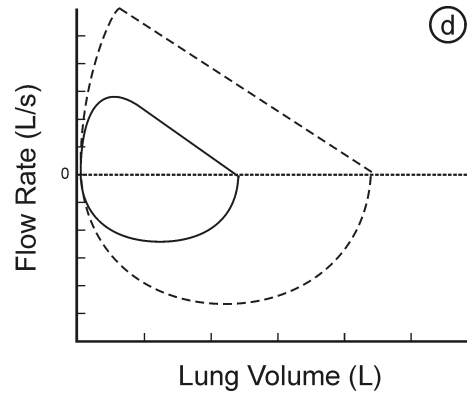
- b. Blocking off one lung entirely will lower vital capacity by about half but produce a normal appearing flow volume loop, just smaller.



- c. The peak flow is reduced but because of a greater effect on small airways, the right hand portion of the expiratory loop is reduced to a greater extent. Vital capacity may be close to normal or reduced.



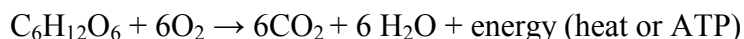
- d. With *kyphoscoliosis*, a form of restrictive disease, volumes are diminished but the shape of the loop is essentially the same.



2. Individual values for FEV_1 and FVC vary greatly between individuals. However, the ratio of FEV_1/FVC (%) holds true for different ages and body sizes, and allows medical personnel to see quickly whether an individual has evidence for obstructive or restrictive lung disease. The FEV_1/FVC (%) ratio is reduced in obstructive lung disease and normal or even increased in restrictive lung disease.

Oxygen and Aerobic Metabolism

Oxygen plays a key role in aerobic cellular metabolism, facilitating the conversion of glucose, protein, and lipids into usable energy. For every 6 molecules of oxygen used in the breakdown of glucose, 6 molecules of carbon dioxide are produced, along with water and adenosine triphosphate (ATP), according to the following equation:



The average person uses 200–250 mL of O₂ per minute at rest. This may increase to 2–3 L per minute during heavy exercise and to twice that amount in highly trained athletes. The increase of oxygen consumption is proportional to the amount of work performed up to a maximum level which is dependent on conditioning. At the start of exercise, anaerobic metabolism is used briefly, but this quickly changes to aerobic metabolism as blood flow to muscles increases.

As O₂ is consumed and CO₂ produced by muscle cells (and other cells), a pressure gradient is created between the cells, the interstitial fluid, and the bloodstream. A marked lowering of O₂ in interstitial fluid as it is used up by cells leads to O₂ diffusion from the bloodstream. While dissolved O₂ provides an immediate supply to replenish the interstitial fluid and cells, the majority of O₂ is carried on hemoglobin molecules. Oxyhemoglobin dissociates more readily as the oxygen concentration is lowered (and CO₂ concentration increased), rapidly replenishing the supply of dissolved O₂. CO₂ diffuses from active cells (where it is produced in high concentration) to the interstitial fluid and bloodstream, where it is transported to the lungs mainly as bicarbonate. In the lungs, the opposite is true. O₂ follows a pressure gradient from the alveoli into the bloodstream, and CO₂ from the bloodstream into the alveoli.

In this experiment, you will measure tidal volumes and oxygen concentrations of deeply inhaled and exhaled air at rest and after exercise. You will use these measurements to calculate the resulting differences in oxygen consumption.

Important: Do not attempt this experiment if you have pulmonary or musculoskeletal problems that might be aggravated by exercise.

OBJECTIVES

In this experiment, you will

- Obtain graphical representation of tidal volume and change in O₂ concentration with breathing at rest and after exercise.
- Calculate oxygen consumption at rest and after exercise.
- Correlate your findings with clinical situations.

MATERIALS

computer
 Vernier computer interface
 Logger Pro
 Vernier Spirometer
 Vernier O₂ Sensor

Vernier CO₂-O₂ Tee
 disposable bacterial filter
 nose clip
 4 cm (3/4 in.) PVC pipe

PROCEDURE

Part I Oxygen utilization at rest

1. Assemble the apparatus (see Figure 1).
 - a. Insert the PVC pipe into the larger end of the CO₂-O₂ Tee.
 - b. Attach the smaller diameter side of the bacterial filter to the free end of the PVC pipe. **Caution:** Do not puncture the filter material inside the bacterial filter.
 - c. Identify the side of the Spirometer head marked “Inlet” and insert it into the free end of the bacterial filter (the larger diameter side).
 - d. Insert the O₂ Gas Sensor into the vertical opening of the CO₂-O₂ Tee.
2. Connect the Spirometer and the O₂ Sensor to the Vernier computer interface. Open the file “22 Aerobic Metabolism” from the *Human Physiology with Vernier* folder.
3. Hold the apparatus in one or both hands. Brace your arm(s) against a solid surface, such as a table, and click . Check Spirometer in the pop-up box that appears. *Do not zero the O₂ Sensor.* **Note:** The Spirometer must be held straight up and down during data collection (see Figure 1).
4. Collect inhalation and exhalation data.
 - a. Put on the nose clip.
 - b. Click to begin data collection.
 - c. Taking deep breaths, begin data collection with an inhalation and continue to breathe in and out. Each respiratory cycle should last 4–6 s (2–3 s per inhalation and 2–3 s per exhalation). Continue breathing in this fashion for the duration of the experiment. Data collection will continue for 40 s.
5. Click the Next Page button, , to see the volume data. If the baseline on the top graph has drifted, use the Baseline Adjustment feature to bring the baseline volumes closer to zero, as in Figure 2.

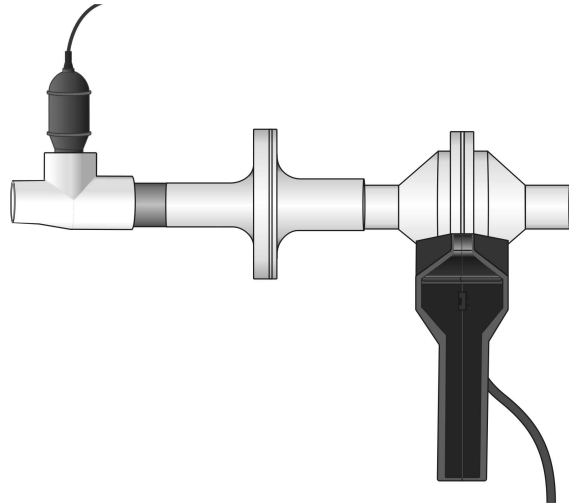


Figure 1

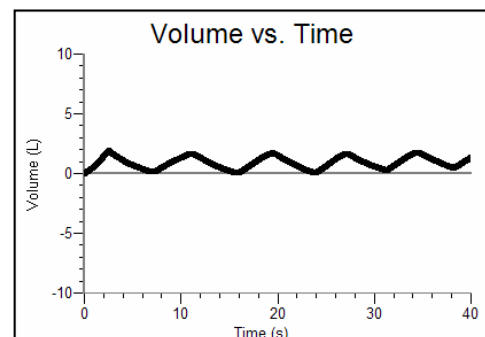


Figure 2

6. Place the cursor on the second peak of the volume graph. Click and drag down to the valley that follows it. Enter the Δy value displayed in the lower left corner of the graph to the nearest 0.1 L as Breath 1 Pre-exercise Tidal Volume in Table 1.
7. Repeat Step 6 for a total of 3 breaths, entering the Δy values for each breath in Table 1.
8. Place the cursor on the second peak of the oxygen gas vs. time graph. Click and drag down to the valley that follows it. Enter the Δy value displayed in the lower left corner of the graph to the nearest 0.01 % as Pre-exercise ΔO_2 Concentration in Table 1.
9. Repeat Step 8 for a total of 3 breaths, entering the Δy values for each breath in Table 1.

Part II Oxygen utilization during exercise

10. Clear your data by choosing Clear All Data from the Data menu. Return to Page 1.
11. Begin running in place for 2 minutes, moving arms as well as legs.
12. At the end of two minutes, pick up the apparatus in one or both hands. Brace your arm(s) against a solid surface, such as a table, and click . Check Spirometer in the pop-up box that appears. *Do not zero the O_2 Sensor.* **Note:** The Spirometer must be held straight up and down during data collection (see Figure 1).
13. Collect inhalation and exhalation data.
 - a. Put on the nose clip.
 - b. Click to begin data collection.
 - c. Taking deep breaths, begin data collection with an inhalation and continue to breathe in and out. Each respiratory cycle should last 4–6 s (2–3 s per inhalation and 2–3 s per exhalation). Continue breathing in this fashion for the duration of the experiment. Data collection will continue for 40 s.
14. Click the Next Page button, , to see the volume data. If the baseline on the top graph has drifted, use the Baseline Adjustment feature to bring the baseline volumes closer to zero, as in Figure 2.
15. Place the cursor on the second peak of the volume vs. time graph. Click and drag down to the valley that follows it. Enter the Δy value displayed in the lower left corner of the graph to the nearest 0.1 L as Breath 1 Post-exercise Tidal Volume in Table 1.
16. Repeat Step 15 for a total of 3 breaths, entering the Δy values for each breath in Table 1.
17. Place the cursor on the second peak of the Oxygen Gas vs. Time graph. Click and drag down to the valley that follows it. Enter the Δy value displayed in the lower left corner of the graph to the nearest 0.01% as Post-exercise ΔO_2 Concentration in Table 1.
18. Repeat Step 17 for a total of 3 breaths, entering the Δy values for each breath in Table 1.
19. Compute the average value for each of the parameters measured and enter those values in Table 1.

DATA

Table 1				
Breath	Tidal volume (L)		ΔO_2 Concentration (%)	
	Pre-exercise	Post-exercise	Pre-exercise	Post-exercise
1				
2				
3				
4				
Average				

DATA ANALYSIS

1. Compare the tidal volumes of the 4 breaths you analyzed both at rest and post-exercise. Is it important for this experiment that resting and post-exercise tidal volumes be similar?
2. Inhaled oxygen should have the same concentration at rest and post-exercise (approximately 21%), yet the peak oxygen concentration values in the post-exercise run never achieve this value. What aspect of the experimental design accounts for this finding?
3. Use the average values for Tidal Volume and ΔO_2 Concentration (%) from Table 1 to calculate the average O_2 consumed pre- and post-exercise per breath and over the combined 4 breaths:

$$\Delta O_2 \text{ Concentration (\%)} \times \text{Tidal Volume (L)} = O_2 \text{ consumed per breath}$$

$$O_2 \text{ consumed per breath} \times 4 \text{ breaths} = O_2 \text{ consumed over that time interval}$$

4. CO_2 was not measured. What would you expect the volume of exhaled CO_2 to be in this experiment at rest and after exercise?

EXTENSIONS

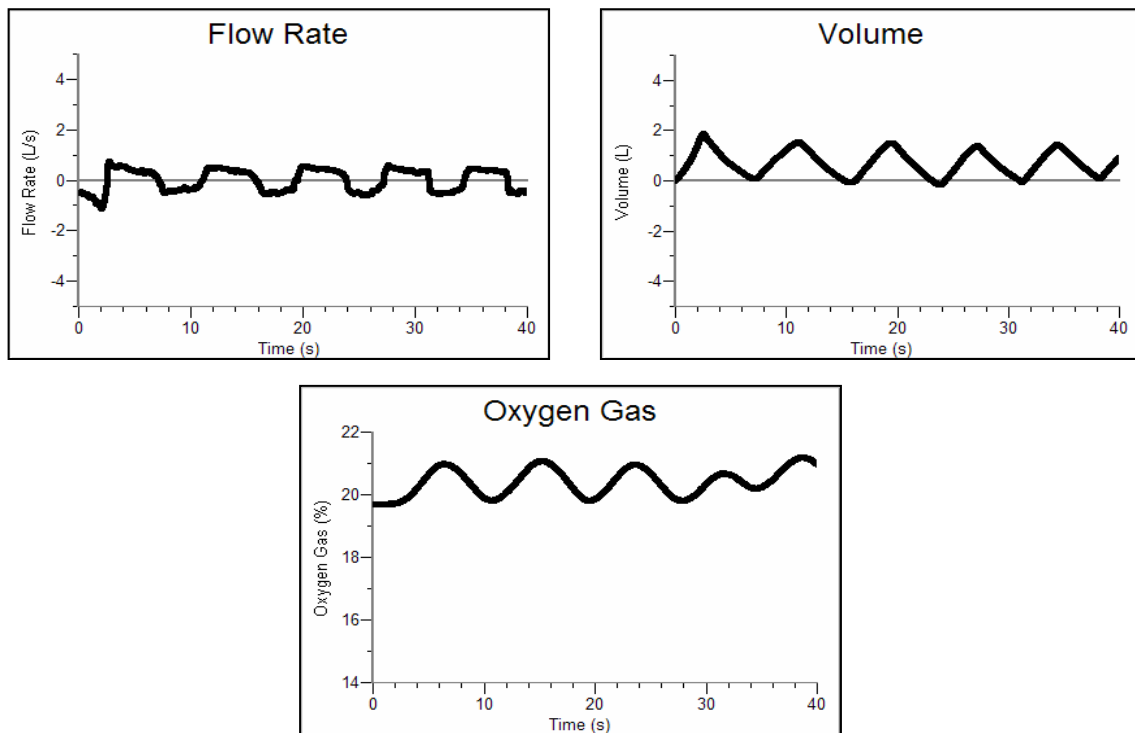
1. Oxygen consumption with exercise is directly proportional to the muscle mass being used. Demonstrate this principle by performing this experiment exercising with your legs only and/or with your arms only.
2. Perform this experiment after exercising for varying lengths of time (1 minute or 5 minutes).

TEACHER INFORMATION

Oxygen and Aerobic Exercise

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. The Spirometer should be held vertically and steadily. It is helpful to brace at least one arm on a hard surface, such as a table. Deviation from this position will affect data collection.
3. It is important for students to begin data collection with an inspiration, since this is the trigger for data collection to begin.
4. The student procedure directs students to perform a baseline adjustment if their data drifted. The Baseline Adjustment feature was not present in the initial release of LabQuest App. It will be present in later versions of LabQuest App.
5. The students are instructed to measure from the second peaks in the various runs, so that any anomalies in the first breath data are avoided.
6. To maintain good hygiene, use the Spirometer with a MicroGard[®] bacterial filter and a disposable mouthpiece. In an ideal situation, every student would have his or her own bacterial filter and disposable mouthpiece. However, budget restraints may require you to share the bacterial filter among a few students. According to the manufacturer's specs, the MicroGard filters out more than 99% of bacterial and viral aerosols.

SAMPLE DATA



Flow rate, volume, and percent oxygen gas graphs collected while breathing at rest

Experiment 22

Breath	Tidal Volume (L)		ΔO_2 Concentration (%)	
	Pre-exercise	Post-exercise	Pre-exercise	Post-exercise
1	4.2	4.2	1.11	2.40
2	4.0	4.3	1.11	2.58
3	4.5	4.6	1.33	2.40
Average	4.2	4.4	1.18	2.46

ANSWERS TO THE DATA ANALYSIS QUESTIONS

- Answers will vary. Students were asked to take deep breaths but the breaths were not standardized. This should not affect the outcome of the experiment, as O_2 uptake and utilization are dependent on tissue metabolism, not on the frequency or deepness of the breaths taken (with frequent shallow breaths less oxygen will be exchanged per breath).
- The entire curve is shifted downward in the post-exercise graph because excess CO_2 remains in the apparatus (*dead space*). The volume of air in the apparatus is re-breathed, leading to lesser O_2 concentrations overall. This does not affect the percent of O_2 extracted.
- Answers will vary. ΔO_2 Concentration (%) \times Tidal Volume (L) = O_2 consumed per breath.

Pre-exercise ΔO_2 Concentration = 1.2%

Pre-exercise Tidal Volume = 4.2 L

$(0.012)(4200\text{mL/breath}) = 50.4 \text{ mL/breath}$

$50.4 \text{ mL/breath} \times 4 \text{ breaths} = 202 \text{ mL}$

Post-exercise ΔO_2 Concentration = 2.5%

Post-exercise Tidal Volume = 4.4 L

$(.025)(4400\text{mL/breath}) = 110 \text{ mL/breath}$

$110\text{mL/breath} \times 4 \text{ breaths} = 440 \text{ mL}$

At rest, 202 mL of O_2 were consumed over the 30 second time interval. This is higher than what is found on precision medical equipment. O_2 consumption increased to 440 mL after exercise.

- The volume of exhaled CO_2 should be roughly equivalent to the volume of O_2 consumed.

O₂ Extraction by the Lungs

Oxygen is required for cell metabolism. During inhalation air is brought into the lungs, where oxygen is extracted. Oxygen passes into the bloodstream at the membrane between the alveoli and pulmonary capillaries (see Figure 1). The quantity of oxygen extracted from the air is dependent on the composition of the air and on the efficiency of the lungs. The percent concentration of oxygen in air on earth is almost uniformly 21% between sea level and the stratosphere. At sea level the density of the air (molecules/unit volume) is greater than it is at higher altitudes, allowing a greater number of molecules to be inhaled with each breath.

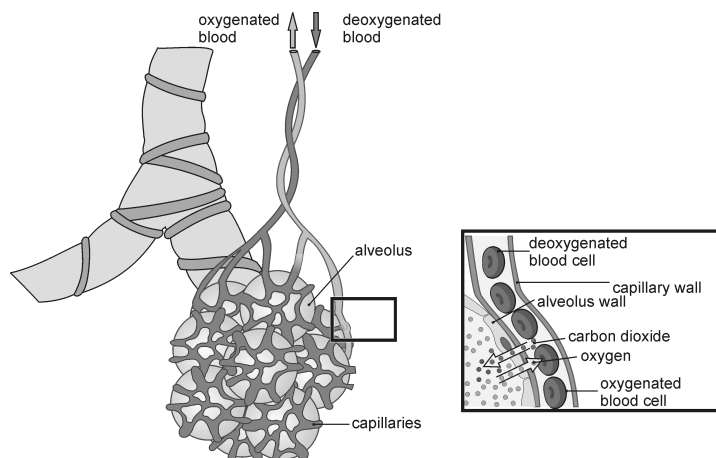


Figure 1

Diseases in which lung tissue is damaged reduce the ability of the lungs to absorb oxygen. Examples include *emphysema*, in which there are fewer total alveoli, and *pulmonary fibrosis*, in which scarring hinders oxygen transfer across a thickened membrane. Depending on the severity of the disease, people with lung disease may require supplemental oxygen. Oxygen may be supplied in concentrations anywhere from 21% to 100%. In less severe cases oxygen may not be required at sea level but needed at higher elevations or when traveling by air.

The majority of oxygen is carried in the blood by hemoglobin molecules. This is dependent upon the partial pressure of oxygen, but the binding of oxygen is efficient over a wide range of oxygen pressures. This relationship is demonstrated in the “Oxygen-Hemoglobin Dissociation Curve” (see Figure 2). In conditions where hemoglobin concentration is low (anemia), oxygen delivery to the tissues is proportionately decreased. Blood transfusions, rather than supplemental oxygen, will increase oxygen delivery to the tissues.

In this experiment, you will observe the quantity of oxygen that is absorbed from inhaled air by measuring the concentration of oxygen remaining in exhaled air. Successive breaths will further lower the oxygen concentration, allowing you to observe the efficiency of oxygen extraction by the lungs at lower oxygen concentrations.

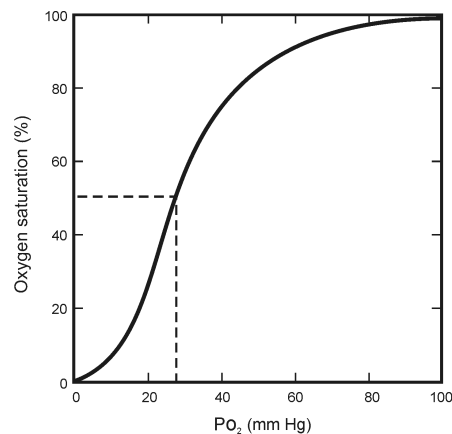


Figure 2

OBJECTIVES

In this experiment, you will

- Measure the concentration of exhaled oxygen.
- Observe the efficiency of oxygen extraction by the lungs as the inhaled oxygen concentration is reduced.

MATERIALS

computer
Vernier computer interface
Logger *Pro*
Vernier O₂ Gas Sensor
cardboard paper towel roll

rubber band
clear tape
gallon size plastic bag
Phillips screwdriver or pointed scissors
pen or pencil

PROCEDURE

1. Prepare the cardboard paper towel roll by cutting it to approximately 15 cm in length. Place the tube on a stable surface. Starting 3 cm from one end of the tube, mark 3 cm intervals to the other end of the tube. Draw four lines evenly spaced around the cylinder. Use a screwdriver or scissors to punch holes where indicated by the marks (see Figure 3).
2. Use clear tape to completely cover one end of the tube. On the opposite end, place tape around the circumference of the tube (Figure 3). This will prevent direct contact of the lips with the cardboard, which can result in the cardboard sticking to the lips.
3. Secure the bag to the O₂ Gas Sensor by cutting a small hole the size of a quarter in the top of the bag and feeding the shaft of the sensor through the hole. Use tape to seal the bag to the sensor (see Figure 4).
4. Connect the cardboard tube to the open end of the plastic bag with a rubber band. The tube should be *inside* the bag (see Figure 4).
5. Open the file “23 O₂ Extraction” from the *Human Physiology with Vernier* folder.
6. The subject should be seated comfortably. Hold the tube lightly around the rubber band to avoid blocking the holes. Use the opposite hand to squeeze all of the air out of the bag. Allow the sensor to hang upside down for the duration of the experiment.
7. Check the oxygen concentration in the meter to be sure that it reads 20–22% concentration. If it does not, remove the cardboard tube from the bag and expose the shaft of the sensor to the air by inverting the bag. When the meter reads correctly, reattach the tube and proceed with Step 8.

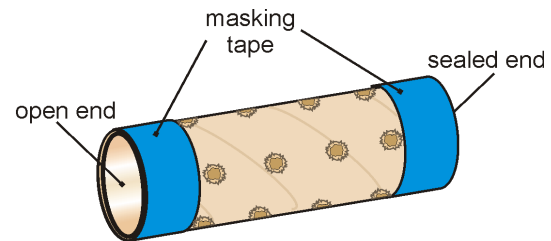


Figure 3

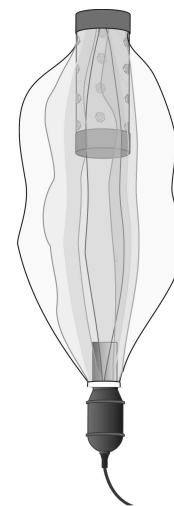



Figure 4

8. Click **Collect** to begin data collection. During the first 10 s take a large breath. At 10 s, exhale through the tube into the bag. Without removing your mouth from the tube, wait 10 s and inhale the air from the bag. Repeat this process alternately exhaling and inhaling every 10 s. *Stop immediately if you feel dizzy.* Data collection will end at 80 s.

9. Determine the mean O₂ concentration in the bag after each exhalation.
- Click and drag across the Breath 0 plateau (see Figure 5).
 - Once the area has been highlighted, click the Statistics button, , to display the Statistics box.
 - Record the mean O₂ concentration in Table 1 for Breath 0.
 - Move the brackets to highlight the Breath 1 exhalation plateau. As you move the brackets, the statistics in the Statistics box will be updated.
 - Record the mean O₂ concentration in Table 1 for this breath.
 - Repeat this process for each successive exhalation plateau.
 - Close the Statistics box by clicking the × in the corner of the box.

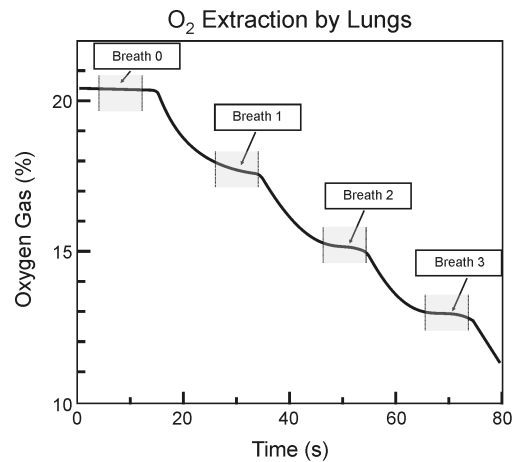


Figure 5

10. Determine the difference between each successive mean concentration of O₂. Enter your results in column 3 of Table 1.
11. Determine the percent of the existing O₂ that is removed with each successive breath.
- Divide the change in O₂ concentration value by the beginning concentration for that breath (i.e., Divide the number in Column 3 by the number in the previous row in Column 2).
 - Multiply the value by 100 to determine the percent change in O₂ concentration with each successive breath.
 - Enter your results in Column 4 of Table 1 (round to the nearest whole number).

DATA

Table 1			
Breath	Mean O ₂ concentration (%)	ΔO ₂ concentration (%)	Percent of existing O ₂ removed with each breath (%)
0			
1			
2			
3			

DATA ANALYSIS

1. Extrapolating from the graph, how many more breaths would it take to use up all of the oxygen in the bag?
2. Use the data from column 4 in Table 1 to decide whether there was any significant change in the efficiency of oxygen extraction by the lungs with successive breaths. Use your knowledge of oxygen binding to hemoglobin to explain your findings. Does this support the idea that lack of oxygen is what leads to the discomfort you may have experienced at the end of the experiment?
3. While the O₂ concentration in the bag declines with each breath, the volume of air breathed in remains the same. What is replacing the volume of oxygen that is being lost with each breath?
4. Air is humidified by the lungs such that the partial pressure of water (in the form of gas molecules) in the alveoli is 6.266 kPa (47 mm Hg). Use this information to explain why the windows in your car may fog up after you have been driving for 30 minutes.
5. Assume that the volume of air breathed in and out with normal breathing is 500 mL, or $\frac{1}{4}$ the volume breathed in and out during this experiment (1 gallon \approx 4 L). Also assume that the efficiency of oxygen extraction is the same for these smaller breaths. How long would it take without supplemental oxygen tanks to use up all of the oxygen in a Gemini space capsule (2-man space capsule used between 1964–1966 with 2.26 m³ of habitable space)?

6. Barometric or atmospheric pressure refers to the sum total of the pressure of all gases present. At sea level it is approximately 101.3 kPa (760 mm Hg). What is the oxygen pressure in the bag at the start of the experiment? What is it at the end of the experiment? (Perform these calculations using the current barometric pressure at your location, if available.)

7. Assume the barometric pressure at the top of Mt. Everest (8,850 m) is 32.7 kPa (245 mm Hg). What is the oxygen pressure there? What percent of oxygen would need to be breathed at the top of Mt. Everest, or in a commercial airliner flying at that altitude, to equal the pressure of oxygen breathed at sea level?

EXTENSION

Perform this experiment taking normal or shallow breaths. Compare the results of this to the results you obtained in this exercise, in which you were instructed to take very large breaths.

TEACHER INFORMATION**O₂ Extraction by the Lungs**

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. The results in the table below were obtained using a gallon-sized food and bread storage bag. Packages of these bags are easily obtained at any grocery store. An excessively large bag may result in inadequate mixing of the exhaled air.
3. To secure the bag to the O₂ Gas Sensor, cut a small hole the size of a quarter in the bottom of the bag 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. The O₂ Gas Sensor will be upside down during the duration of the experiment (80–100 s). This amount of time does not have any adverse effect on the sensor's ability to collect data.
5. **IMPORTANT:** Students should avoid placing their lips directly on the cardboard tube, which can result in the tube sticking to their lips. Be sure to surround the end of the tube with a layer of tape to protect against injury.
6. Students with respiratory ailments should exercise caution and not perform the entire experiment if they are feeling dizzy.
7. Warn students not to hold the cardboard tube in their hands while punching holes in it to avoid injury.

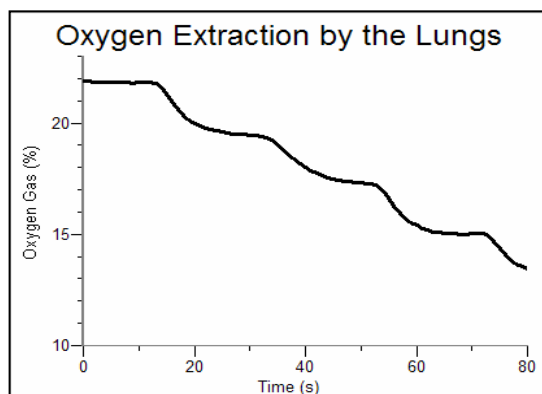
SAMPLE DATA*Sample data*

Table 1			
Breath	Mean O ₂ concentration (%)	ΔO ₂ concentration (%)	Percent of existing O ₂ removed with each breath (%)
0	21.8		
1	19.6	2.2	10
2	17.4	2.2	13
3	15.2	2.4	14

ANSWERS TO THE DATA ANALYSIS QUESTIONS

- The oxygen concentration will drop by approximately 8% after 3 breaths. Use the following algebraic manipulation to compute the answer:

$$\frac{7\%}{3 \text{ breaths}} = \frac{22\%}{x \text{ breaths}}$$

Therefore,

$$\frac{(22 \times 3)}{7} \approx 9 \text{ breaths}$$

Since 3 breaths have already been taken,

$$9 \text{ breaths} - 3 \text{ breaths} = 6 \text{ breaths}$$

The answer is 6 more breaths.

- There is no significant change in oxygen extraction by the lungs from one breath to the next. As demonstrated in Figure 2, the partial pressure of oxygen can drop significantly before the hemoglobin saturation drops. This does not support the idea that lack of oxygen is responsible for discomfort; in fact, it is the buildup of carbon dioxide that results in a feeling of “air hunger.”
- CO₂ and H₂O are added to the air in the alveoli and become part of the exhaled air mixture.
- Students may have noticed droplets of moisture in the bag at the end of the experiment. The lungs add water to each exhalation. If you are breathing in an enclosed space, the humidity will increase with time.

5. $2.26 \text{ m}^3 \times 10^6 \text{ mL/m}^3 = 2.26 \times 10^6 \text{ mL}$ is the volume of the space capsule.

In this experiment, students breathed approximately 4000 mL/breath. In question 1, above, it was demonstrated that 9 breaths would completely deplete the oxygen in a gallon sized (4000 mL) plastic bag. Breathing at 1/8 the volume (500 mL), it would take 72 breaths to deplete all the oxygen in the bag.

$$\frac{72 \text{ breaths}}{4 \times 10^3 \text{ mL}} = \frac{x \text{ breaths}}{2.26 \times 10^6 \text{ mL}}$$

$$x = 4.1 \times 10^4 \text{ breaths}$$

To discover how long it would take to breathe 4.1×10^4 times, assume a constant respiratory rate of 16 breaths/minute.

$$\frac{34.1 \times 10^4 \text{ breaths}}{16 \text{ breaths/min}} = 2.56 \times 10^3 \text{ min}$$

Convert minutes to hours

$$2.56 \times 10^3 \text{ min} \times \left(\frac{1 \text{ hour}}{60 \text{ min}} \right) = 42.7 \text{ hours}$$

If 2 people were occupying the capsule, the oxygen would be depleted in half this time, or 21.3 hours.

6. Answers will vary. To calculate the partial pressure of oxygen in the bag at the start of the experiment, use the % O₂ data from Breath 0 in Table 1 in the following computation:

at sea level: % O₂ (101.3 kPa) [or % O₂ (760 mm Hg)]

For the final calculation, use the % O₂ data from the last breath in the same equation.

7. The partial pressure of oxygen at sea level can be calculated as follows:

$$0.22 \times 101.3 \text{ kPa} = 22.3 \text{ kPa}$$

$$0.22 \times 760 \text{ mm Hg} = 167 \text{ mm Hg}$$

To compute the percent oxygen required at the top of Mt. Everest to deliver a partial pressure equivalent to that delivered at sea level

$$22.3 \text{ kPa} = (32.7 \text{ kPa}) x$$

$$167 \text{ mm Hg} = (245 \text{ mm Hg}) x$$

$$x = 0.68, \text{ or } 68\%$$

Effect of “Dead Space” on Oxygen Availability

In the average adult, approximately 150 mL of the air that is inhaled with each breath never reaches the alveoli. It fills the nose, mouth, larynx, trachea, bronchi, and bronchioles, a volume known as the “dead space.” This air is not available for gas mixing and exchange. It mixes with newly inhaled air and is “recycled” back to the alveoli. The relative size of the “dead space” as compared to functioning lung tissue impacts the efficiency of the respiratory system. Dead space is important in a variety of medical conditions such as asthma, pneumonia, and emphysema, and must be considered in treatments such as artificial ventilation in an intensive care unit. It is also important in physiologic challenges such as diving and high altitude activities.

In this experiment, you will use the O₂ Gas Sensor to analyze inhaled and exhaled air while artificially changing the size of the “dead space.” You will calculate the effect that dead space has on oxygen delivery to the lungs and correlate this with various physiologic challenges.

The majority of oxygen is carried in the blood by hemoglobin molecules. This is dependent upon the partial pressure of oxygen, but the binding of oxygen is efficient over a wide range of oxygen pressures. This relationship is demonstrated in the “Oxygen-Hemoglobin Dissociation Curve” (see Figure 1). In conditions where hemoglobin concentration is low (*anemias*), oxygen delivery to the tissues is proportionately decreased. Blood transfusions, rather than supplemental oxygen, will increase oxygen delivery to the tissues.

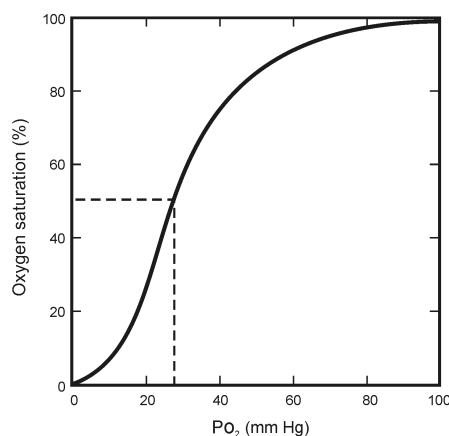


Figure 1

OBJECTIVES

In this experiment, you will

- Simulate different volumes of dead space.
- Measure the oxygen concentration within the dead space.
- Correlate dead space volume with a variety of physiologic challenges.

MATERIALS

computer
Vernier computer interface
Logger Pro
Vernier O₂ Gas Sensor

Vernier CO₂-O₂ Tee
15 cm (³/₄ in) PVC pipe
30 cm (³/₄ in) PVC pipe

PROCEDURE

Part I The Effect of Increasing Dead Space on Oxygen Concentration

1. Connect the O₂ Gas Sensor to Channel 1 of the Vernier computer interface. Open the file “24 Effect of Dead Space” from the *Human Physiology with Vernier* folder.
2. Insert the Vernier O₂ Gas Sensor into the CO₂-O₂ Tee as shown in Figure 2.
3. Click to begin data collection. Place your mouth on the smaller end of the CO₂-O₂

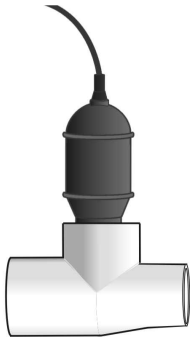


Figure 2

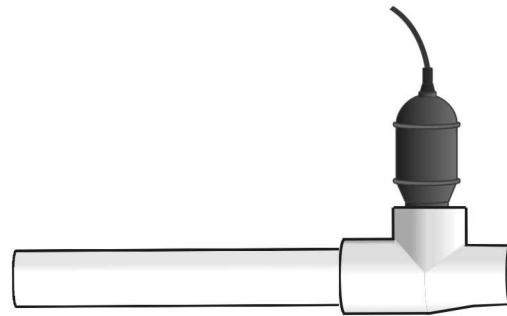


Figure 3

Tee and begin normal breathing. Data will be collected for 60 s.

4. Store this run by choosing Store Latest Run from the Experiment menu.
5. Remove the O₂ Gas Sensor from the CO₂-O₂ Tee and allow readings to return to ambient levels. Monitor oxygen levels by observing the readings on the screen. Once ambient levels have been reached, re-attach the sensor to the CO₂-O₂ Tee.
6. Attach the 15 cm PVC pipe to the larger end of the CO₂-O₂ Tee as shown in Figure 3.
7. Click to begin data collection. Place your mouth on the smaller end of the CO₂-O₂ Tee and begin normal breathing. Data will be collected for 60 s.
8. Store this run by choosing Store Latest Run from the Experiment menu.
9. Remove the O₂ Gas Sensor from the CO₂-O₂ Tee and allow readings to return to ambient levels. Monitor oxygen levels by observing the readings on the screen. Once ambient levels have been reached, re-attach the sensor to the CO₂-O₂ Tee.
10. Attach the 30 cm PVC pipe to the larger end of the CO₂-O₂ Tee as shown in Figure 2.

11. Click to begin data collection. Place your mouth on the smaller end of the CO₂-O₂ Tee and begin normal breathing. Data will be collected for 60 s.
12. Click and drag to highlight all data runs from 20–60 s. Click the Statistics button, . Click the boxes in front of Run 1, Run 2, and Latest and click . Record the mean values to the nearest 0.1% for all three runs in Table 1.

Part II The Effect of Increased Effort on Oxygen Concentration

13. Choose Clear All Data from the Data menu.
14. Remove the 30 cm PVC pipe from the CO₂-O₂ Tee and ensure that the Tee is well attached to the O₂ Gas Sensor.
15. Click to begin data collection. Place your mouth on the smaller end of the CO₂-O₂ Tee and begin deeply inhaling and exhaling. Allow 3 s for inhalation and 3 s for exhalation to prevent dizziness. Data will be collected for 60 s.
16. Store this run by choosing Store Latest Run from the Experiment Menu.
17. Remove the O₂ Gas Sensor from the CO₂-O₂ Tee and allow readings to return to ambient levels. Monitor oxygen levels by observing the readings in the Logger *Pro* tool bar. Once ambient levels have been reached, re-attach the sensor to the CO₂-O₂ Tee.
18. Attach the 15 cm PVC pipe to the larger end of the CO₂-O₂ Tee as shown in Figure 2.
19. Click to begin data collection. Place your mouth on the smaller end of the CO₂-O₂ Tee and begin deep breathing, as instructed in Step 16. Data will be collected for 60 s.
20. Store this run by choosing Store Latest Run from the Experiment Menu.
21. Remove the O₂ Gas Sensor from the CO₂-O₂ Tee and allow readings to return to ambient levels. Monitor oxygen levels by observing the readings in the Logger *Pro* tool bar. Once ambient levels have been reached, re-attach the sensor to the CO₂-O₂ Tee.
22. Attach the 30 cm PVC pipe to the larger end of the CO₂-O₂ Tee as shown in Figure 2.
23. Click to begin data collection. Place your mouth on the smaller end of the CO₂-O₂ Tee and begin deep breathing, as instructed in Step 16. Data will be collected for 60 s.
24. Click and drag to highlight all data runs from 20–60 s. Click the Statistics button, . Click the boxes in front of Run 1, Run 2, and Latest and click . Record the mean values for all three runs in Table 2.

DATA

Table 1–Normal Breathing		
Run	Description	Mean O ₂ concentration (%)
1	CO ₂ -O ₂ Tee alone	
2	CO ₂ -O ₂ Tee + 15 cm PVC pipe	
3	CO ₂ -O ₂ Tee + 30 cm PVC pipe	

Table 2–Deep Breathing		
Run	Description	Mean O ₂ concentration (%)
1	CO ₂ -O ₂ Tee alone	
2	CO ₂ -O ₂ Tee + 15 cm PVC pipe	
3	CO ₂ -O ₂ Tee + 30 cm PVC pipe	

DATA ANALYSIS

1. The varying lengths of PVC pipe act to increase the volume of inhaled air that is not available for gas mixing and exchange – the so-called “dead space.” Calculate the volume of air in the 15 and 30 cm lengths of PVC pipes ($\pi r^2 h$), respectively. To each value, add 30 mL to account for the volume of air present in the CO₂-O₂ Tee. Record your answers in the table below in the column labeled “Volume” (remember that cm³ = mL). Add these volumes to your anatomical dead space (150 mL), and enter the total values into the table.

Calculation of Dead Space		
Pipe length	Volume of PVC + tee (mL)	Anatomical + additional dead space (mL)
15 cm		
30 cm		

2. Using the table you completed in Question 1, estimate the proportion by which your anatomical dead space was artificially increased by
 - (a) the 15 cm tube.

 - (b) the 30 cm tube.

3. Describe the trend in mean oxygen concentration seen in the data entered in Table 1.

4. Air is humidified as it passes through the anatomical dead space, becoming fully saturated with water. If 6.3 kPa (47 mm Hg) of H₂O is added, it displaces the equivalent pressure of inhaled air. This must be considered when calculating the partial pressure of oxygen. Calculate (to the nearest whole number) the partial pressure of oxygen for each of the runs in Part I using the following formula (if you do not know the barometric pressure, use 101.3 kPa (760 mm Hg) or the normal barometric pressure for your location):
$$\text{Partial Pressure [O}_2\text{]} = (\text{Barometric Pressure} - 6.3 \text{ kPa})(\text{Mean Oxygen Concentration})$$
 - (a) Run 1

 - (b) Run 2

 - (c) Run 3

5. In normal breathing, the addition of CO₂ and H₂O to air in the alveoli reduces the partial pressure of oxygen to approximately 13.3 kPa (100 mm Hg). Subtract the partial pressure of O₂ that you calculated for Run 3 from the value you calculated for Run 1 in question 4. If the alveolar oxygen is reduced by the same amount does this affect the amount of oxygen bound to hemoglobin (see Figure 1).

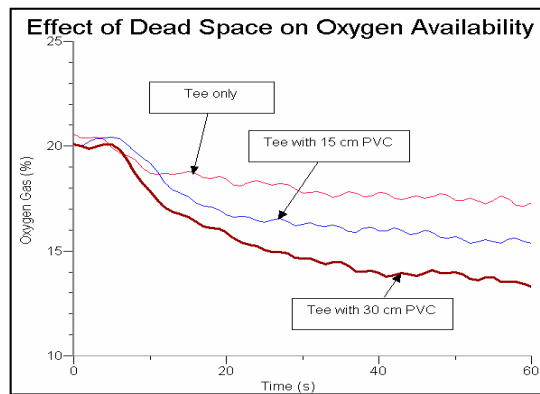
6. The average amount of air breathed in and out during normal respirations is approximately 500 mL (this is called *tidal volume*). The volume of air inhaled and exhaled during deep breaths is approximately 4000 mL (this is called *vital capacity*). Use these values to explain the difference in trends seen in Table 1 and Table 2.

TEACHER INFORMATION

Effect of Dead Space on Oxygen Availability

1. The student pages with complete instructions for data collection using LabQuest App and Logger *Pro* (computers) can be found on the CD that accompanies this book. See *Appendix A* for more information.
2. The CO₂-O₂ Tee should be disinfected prior to use by a different subject. Students with respiratory illness should be excused from acting as subjects in this experiment.
3. Most students should see significant differences between the three runs in Part I of the experiment.; for students who do not, taking shorter, shallower breaths may be helpful. This will cause the dead space to be a greater proportion of the tidal volume.

SAMPLE DATA



Sample data

Table 1–Normal Breathing		
Run	Description	Mean O ₂ concentration (%)
1	CO ₂ -O ₂ Tee alone	18.1
2	CO ₂ -O ₂ Tee + 15 cm PVC pipe	17.3
3	CO ₂ -O ₂ Tee + 30 cm PVC pipe	16.5

Table 2–Deep Breathing		
Run	Description	Mean O ₂ concentration (%)
1	CO ₂ -O ₂ Tee alone	19.7
2	CO ₂ -O ₂ Tee + 15 cm PVC pipe	19.8
3	CO ₂ -O ₂ Tee + 30 cm PVC pipe	19.9

ANSWERS TO THE DATA ANALYSIS QUESTIONS

1. For PVC tubing with a radius of 1.1 cm:

Calculation of Dead Space		
Pipe length	Volume of PVC + Tee (mL)	Anatomical + Additional Dead Space (mL)
15 cm	87	237
30 cm	144	294

2. (a) 58% increase in dead space (87 mL/150 mL)
 (b) 96% increase in dead space (144 mL/150 mL)
3. The greater the dead space, the lower the mean O₂ concentration. Students may calculate the exact percentage change represented by their data. Using the data above, increasing the dead space by 58% lowered the O₂ concentration by 4.4% [(18.1 %–17.3%)/18.1%]. Increasing the dead space by 96% lowered the O₂ concentration by 4.6%.
4. Partial pressure of O₂ for Run 1 (assuming a barometric pressure of 101.3 kPa (760 mm Hg)):
 17.2 kPa (129 mm Hg)

$$(101.3 \text{ kPa} - 6.3 \text{ kPa}) \times .181 = 17.2 \text{ kPa}$$

Partial pressure of O₂ for Run 2 (assuming a barometric pressure of 101.3 kPa (760 mm Hg)):
 16.4 kPa (123 mm Hg)


Partial pressure of O₂ for Run 3 (assuming a barometric pressure of 01.3 kPa (760 mm Hg)):
 15.7 kPa (118 mm Hg)


5. Partial pressure of O₂ Run 3 – Partial pressure of O₂ Run 1 = –1.5 kPa (–11 mm Hg)
 Reducing the partial pressure of O₂ in the alveoli by 1.5 kPa (11 mm Hg) results in a new partial pressure of 1.3 kPa (89 mm Hg). According to the Oxygen-Hemoglobin Dissociation Curve in Fig. 1, this does not result in a significant decline in oxygen saturation.
6. The oxygen concentration fell with increasing dead space in Part I of the experiment (Table 1). In part two, with deep breathing, the oxygen concentration remained steady (Table 2). With deep breathing, the dead space becomes much less significant compared to the total volume of air being breathed.
7. In order to maintain an identical dead space:tidal volume ratio, the diver needs to increase tidal volume to 980 mL (150 mL/500 mL = 294 mL/x, where x = the new tidal volume). It is actually an increase in CO₂ that is the main physiologic drive for increase in tidal volume or respiratory rate.


Using the Electronic Resources

The electronic resources, accessed through your account at www.vernier.com, contain the following:

Student

 **LabQuest**—Contains the LabQuest instructions for each of the student activities in this book.

 **Logger Pro**—Contains the Logger *Pro* instructions for each of the student activities in this book.

 **Teacher**—Contains teacher information pages for each of the experiments in this book.

Using the *Human Physiology with Vernier* Word-Processing Files

All file names begin with the experiment number, followed by an abbreviation of the title and then a name to indicate the equipment with which it should be used; e.g., 01 Warming Function is the file name for *Experiment 1, Warming Function of Nasal Passageways*. 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 *Human Physiology with Vernier*.

Vernier Products for Human Physiology

All software and laboratory interfacing hardware required for the experiments contained in this book are available from Vernier Software & Technology. The purchase of *Logger Pro* software includes a site license that permits you to make as many copies as you wish for use in your own school or college department. You may also use the programs on networks within your school at no extra cost. A table of order codes can be found in this appendix.

Vernier LabPro

Vernier LabPro provides a portable and versatile data-collection device for any class studying human physiology. A wide variety of Vernier probes and sensors can be connected to each of the four analog channels and two sonic/digital channels. LabPro is connected to a computer using a serial or USB port, to a TI graphing calculator, or to a Palm OS handheld. Data collection with LabPro and a computer is controlled by *Logger Pro* software.

Included with the purchase of a LabPro is a Voltage Probe, computer cables (USB and serial for Macintosh and Windows), calculator cradle, calculator link cable, user's manual, and AC power supply.

Vernier 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.

Data-Collection Software for Human Physiology 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
<i>Logger Pro</i>	(order code: LP)

LabQuest

LabQuest App is the data-collection application used to collect data when using LabQuest as a stand-alone device.

Vernier Products for Human Physiology with Vernier

Item	Order Code
Lab Manual	
<i>Human Physiology with Vernier</i>	HP-A
Interfaces	
LabPro interface	LABPRO
LabQuest interface	LABQ
Software	
Logger Pro 3 (for use with computers)	LP
Sensors	
EKG Sensor	EKG-BTA
Hand-Grip Heart Rate Monitor	HGH-BTA
Surface Temperature Sensor	STS-BTA
Blood Pressure Sensor	BPS-BTA
Hand Dynamometer	HD-BTA
Spirometer	SPR-BTA
O ₂ Gas Sensor	O2-BTA
25-g Accelometer (optional)	ACC-BTA

Optional Vernier Human Physiology Products

Respiration Monitor Belt (requires GPS-BTA)	RMB
Gas Pressure Sensor	GPS-BTA
Disposable Bacterial Filter (pkg of 10)	SPR-FIL
Disposable Mouthpiece (pkg of 30)	SPR-MP
Noseclip (pkg of 10)	SPR-NOSE
Spirometer Flow Head	SPR-FLOW
Small Blood Pressure Cuff	XSCUFF
EKG Electrode tabs	ELEC
Chest Belt Transmitter	CBT

Vernier Sensors for Human Physiology

Blood Pressure Sensor

This noninvasive sensor is designed to measure human blood pressure. It can be used to measure systolic and diastolic blood pressure utilizing the oscillometric technique. Range 20 mm Hg to 250 mm Hg

EKG Sensor

This sensor measure electrical signals produced by the heart. It uses three disposable electrodes. An EKG graph is displayed, demonstrating to students the contraction and repolarization of the heart's chambers. A package of 100 disposable electrodes is included with the sensor. The sensor can also be used to produce EMG recordings.

Hand-Grip Heart Rate Monitor

This sensor consists of wireless hand grips and a receiver. The hand grips sense the electrical signals generated by the heart, much like an ECG. For each pulse, a signal is transmitted to the receiver module, and the individual's pulse rate is calculated. You can use a Chest Belt Transmitter in conjunction with the Hand-Grip Heart Rate Monitor's receiver for a hands-free option of measuring heart rate.

Hand Dynamometer

This strain-gage based isometric dynamometer can be used to measure grip strength and to perform muscle fatigue studies. It can be used alone or in combination with EMG recordings for studies of muscular activity. Measurements can be made in newtons or kilograms. Range 0 to 200 N

O₂ Gas Sensor

This sensor measures oxygen concentration in air. It requires no special preparation or calibration. Included with the O₂ Gas Sensor is a 250 mL bottle. Range 0 to 27%

Spirometer

This sensor can be used to perform a variety of tests related to air flow and lung volume. The sensor includes a removable flow head for easy cleaning and sterilization and a differential pressure transducer. It is designed to make human respiratory measurements at rest and during moderate activity. The package includes 1 sensor handle, 1 flow head, 5 disposable mouthpieces, 1 disposable bacterial filter, and 1 nose clip. Range ± 300 L/min

Surface Temperature Sensor

This sensor features an exposed thermistor that results in an extremely rapid response time. It is ideal for taking skin temperature measurements. For use in air only. Range -25 to 125°C

25-g Accelerometer

This sensor measures acceleration along the line marked on the body of the sensor. It can be attached to objects, such as a reflex hammer, to examine acceleration over time. Range -25 g to $+25$ g

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 stations. The materials have been divided into **nonconsumables**, and **consumables**. Most consumables 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
barbell (or similar), 1.36 kg (3 lb)	8	15
bowl	8	3, 7
cable tie, 10 cm long	8	14A
paper towel roll (cardboard tube)	8	23
pen	8	14A, 14B, 23
PVC pipe, $\frac{3}{4}$ ", 4 cm long	8	22
PVC pipe, $\frac{3}{4}$ ", 15 cm long	8	24
PVC pipe, $\frac{3}{4}$ ", 30 cm long	8	24
reflex hammer	8	14A, 14B
ruler	8	15
screwdriver, Phillips (or pointed scissors)	8	23
tape measure, cloth	8	14A
watch (with second hand)	8	8

Consumables

Item	Amount	Experiment
bacterial filter, disposable	varies	19, 20, 21, 22
carrot	8	13
chewing gum	8 sticks	13
egg, hard-boiled	8	13
EKG electrode tabs	24	12, 14A, 14B, 15, 18
ice	varies	2, 3, 7, 10
mouthpiece, disposable	varies	19, 20, 21
nose clip	varies	19, 20, 21, 22
plastic bag, gallon size	8	23

Appendix C

rubber band	8	23
saline solution (for Exercise Heart Rate Monitor)	bottle	3, 4, 5, 6, 10, 11
soap or rubbing alcohol	bottle	2, 13
straw	30	1
tape, adhesive	1 roll	1, 2, 23
towel (paper or cloth)	8	2, 3, 7

Safety Information

While the experiments in this book are safe for the majority of students, certain health conditions may be exacerbated by participation in some of the activities. These are noted in the Introduction of each experiment and include warnings against the following:

- Serious skin conditions may be aggravated by the application of tape, EKG tabs, or ice to the skin.
- Heart or lung conditions may be worsened by exercise, coughing, or spirometry experiments.
- Musculoskeletal disorders may be adversely affected by exercise, squatting, cold immersion, weight lifting, or repetitive gripping.
- Temporomandibular disorders may worsen with chewing.
- Spirometry experiments may lead to shortness of breath or dizziness.
- Knee disorders may be worsened by reflex hammer contact.
- Migraines may be aggravated by application of ice to the cheek.
- Communicable diseases may be transmitted with improper attention to equipment disinfection.
- All Vernier products are designed for educational use only. Our equipment is not designed nor recommended for research or for any industrial or commercial process such as life support, patient diagnosis, control of a manufacturing process, or industrial testing of any kind.

Finally, it is important that lab benches or surfaces upon which physiology equipment rests be clean or covered to avoid contamination by chemicals, etc.

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