PROJECTS FOR THE HIGH SCHOOL BIOLOGY STUDENT

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PREFACE

Wherever and whenever they are used, student projects do much toward providing for a more realistic and meaningful approach to the teaching of high school biology. The purpose of this report is to provide information which may prove beneficial to the teacher of high school biology who wishes to become better acquainted with the use of projects, and also to provide a nucleus around which a larger store of project materials might be built.

This report is limited to a discussion of information concerning projects, and to giving examples of types of projects, suitable for the high school biology student.

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CHAPTER I

INTRODUCTION

Statement of the Problem and Limitations of the Study

For many years there has been a need in high school biology classrooms for a more realistic and meaningful approach to the teaching of biology. One method that has done much toward meeting this need is the use of student projects. Many teachers do not employ this method, however, because they do not have at hand enough materials or because they lack knowledge of the subject.

The purpose of this report is to provide information of a nature which might help overcome this lack of knowledge, and at the same time to provide a nucleus around which a larger store of project materials might be built. It is hoped that this information will be of benefit to the teacher of high school biology and to his students.

This report shall be limited to a discussion of information concerning projects, and to giving examples of types of projects, suitable for the high school biology student.

Clarification of Terms

The word "project" has been so widely and diversely used in educational and scientific circles that its meaning has grown vague. It has

been applied by educators to almost any sort of division of subject matter without regard to its organization or treatment. Among practicing scientists, a project is simply a study of something.

A student project, as it shall be used in this report, is an extended problem or series of related problems that are the outgrowth of the students own interests and endeavors.

Projects may be individual in nature, or they may be undertaken by small groups of students. The project may involve a variety of activities, and may last over a period of several weeks or months. These activities are generally carried on in a natural manner, and with a spirit of purpose to accomplish a definite and attainable goal. Usually the project involves some physical outcome, such as a product, display, written report, or a combination of these.

CHAPTER II

GENERAL INFORMATION

Uses of Projects

The project method was originally used in connection with the teaching of agriculture and home economics. Teachers of these courses, realizing the need for extending the scope of their subjects beyond the textbook and the meager laboratory equipment and materials available, assigned supplementary exercises for students to do on their farms and in their homes.

At the present time, the method of teaching by projects is widely accepted in the biology classroom, as well as in the other sciences, where many of the materials used are of the laboratory or field varieties. Project work is now thought of as being appropriate to class and laboratory time as well as out-of-school time.

Projects are generally used by biology teachers in one of two ways. The first way is to organize the course or part of the course by projects, while carrying out other parts of the course by some other method. The second is to make use of projects which will provide for individual interests or needs.

When projects become a part of the biology course they provide opportunity for the teacher and the students to work together in a spirit that may not be found in conventional laboratory work. While the use of

a project approach in the laboratory may prevent "covering the ground," the student growth that results from project work justifies a less formal program of study than the ordinary pattern of recitation and laboratory work.

There is some evidence that our modern schools do not challenge sufficiently the students of higher than average ability. The project has the potential of continuous stimulation; it supplies avenues for study and investigation that will lead the gifted student as far as his ability and time permit.

There is no reason to think, however, that the use of projects should be limited to students of above-average ability. Because projects are, by their nature, individualized, they provide readily for differences in ability and achievement, as well as interest. The average and the less able students are thus provided for in ways that avoid those types of laboratory procedures that tend not to stimulate students, regardless of ability.

Characteristics of a Good Project

A worthwhile project possesses the following desirable characteristics: (1) The project is a natural and lifelike learning experience complete within itself. Its unity depends upon pupil purpose rather than upon the logical arrangement of subject matter. (2) The project is a willingly accepted task growing out of the student's interests. Along with the acceptance of the challenge of the task goes the acceptance of responsibility for "following through" until its completion. (3) The student plans and directs his own activities. (4) The student has freedom

to pursue his purpose unrestrained by the barriers of subject matter or teacher domination. (5) The project invokes a whole-hearted effort on the part of the student to achieve a definite, desirable, and attainable goal. (6) The project leads to goals which are recognizable by the student, thereby enabling him to evaluate his own progress. (7) The project is of a nature enabling the student to carry on his work, as much as possible, during out-of-school hours.

Values of Projects

Projects, when used properly, have many values. The most important value of project work is that it provides an opportunity for the complete act of thinking by the student. The student defines his problem, plans his work, finds appropriate resources, carries out his plans, and draws his conclusions. In short, the student learns by doing. Other worthwhile values are: the development of an open-minded and tolerant attitude, the development of a scientific way of thinking, and the acquisition of certain skills and techniques.

Opposition to Projects

Although most biology teachers realize the importance of project work, there is still much opposition to it. The three most common objections are: (1) it is more work on the part of the teacher; (2) there are not enough materials available; and (3) there is not enough space available.

Although projects do require a little more effort, planning, technical knowledge and skill on the part of the teacher, the results obtained more than compensate for the extra work involved.

The necessity for more materials and equipment is generally an excuse rather than a reason for not having students work on projects. The problems and experiments can be so devised that they will require nothing that cannot be had at little or no expense to the student. The providing of these materials should be the student's responsibility, and not the teacher's.

Often projects are not done for lack of space. A corner in the laboratory, the demonstration table in the preparation room, a cellar in the house of one of the students, a table in a garage--anywhere where the facilities needed are present will do. Usually space will be made available by civic-minded individuals, businesses and organizations.

Creating Interest in Projects

Interested students present no problem, for they will ask for special project work to be done "on their own." So the first concern of the teacher is to arouse the interest of the less willing students and to get them to select a special project of some kind. Following are a few ideas that help the teacher to create and hold interest in projects:

A list of fifty or more projects might be prepared so that each student is free to make his own selection. In this way almost every student will find something to suit his interests. A suggested list of projects suitable for the high school biology student will be found in Appendix A of this report. Complete dependence upon such a list of titles is not too satisfactory, however, because it does not provide sufficient detail. It is more practical for the teacher to prepare his own resource descriptions of projects as he goes about his reading and his work. To ideas from published lists he can add the essential or suggestive detail. A

card should be prepared for each project. The title of the project may be entered on the card as well as some description of its nature. Details may be added concerning such things as materials, construction, organization, further reading, and presentation.

At the beginning of the school year, students should be shown projects undertaken by former students. A discussion of the problems involved, methods of construction, working ability, potential value, and other pertinent facts may suggest new projects.

Early in the school year, the teacher should hold conferences with students to find out any interests which can be directed toward project activities.

A working calendar should be established. For example, the project should be selected by October, progress reports in November, final sketches of plans by January, and tentative presentation of the project by March. On the calendar might also be included the dates of science fairs and award programs, and a listing of local scientific meetings to be held.

The teacher should allow for frequent and informal buzz sessions that will allow students to predict, analyze, and solve problems related to their projects.

Students appreciate seeing their projects used as teaching aids in regular classroom instruction.

There might be some question as to the classification of a biology club as a project for students, yet it seems that this classification is quite logical because it takes time and effort to carry on a successful club, and by far the greatest amount of this time and effort is that of the students. There is no doubt that the formation of a biology club is an important factor in creating and holding interest in project work.

One of the best sources of information on how to organize a biology club is the Sponsor's Handbook, published by Science Clubs of America, a division of Science Service. For advice on constitutional organization, the club should draw from its local library a copy of Robert's Rules of Order. This book is the accepted authority on such subjects, and it is best that the club vote to govern itself by these rules.

The organization of the club, however, is not as important as arranging a varied, interesting, and educational program. The number of projects a club might carry out is practically unlimited. The following are a few which certainly have merit: maintaining live material for the laboratory, maintaining a school museum, planting flowers and shrubs around the school, developing a "natural garden" on school grounds, keeping bird migration records, aiding in reforestation and other conservation practices, preparing exhibits, taking field trips, giving assembly programs, sponsoring social activities, and most important of all, sponsoring a local science fair.

The primary purpose of the science fair is to give students an opportunity to exhibit their projects. This in itself does much toward creating more interest in project work.

Ordinarily, science fairs are judged on scientific thought, creative ability, thoroughness, clarity, and technical skill. These points should be borne in mind by the student during the preparation of his project.

The booklets, Science Fair Manual for Students and Teachers, prepared by the science department of Northwestern State College, Alva, Oklahoma, and A Manual for Science Fairs, prepared by the Educational Section of the American Museum of Atomic Energy, and published by Science Service, give many practical suggestions for the organization of and participation in

science fairs.

Selection of the Project

The first problem encountered in project work is the selection of the project. In the selection of a project, the student makes decisions about what is to be done--that is, what the nature and goals of the project should be. Student interests, needs, and abilities must be carefully considered in directing this step. In the selection of projects the following criteria should be applied to each project which is proposed: (1) As a result of the study of the project, will the student acquire understandings, skills, ideals, and attitudes which will contribute to his knowledge of biology? (2) In terms of the time and facilities available, how does the project compare with alternative learning activities? (3) Are the learning activities involved in the study of the project appropriate to the maturity of the pupil? (4) Are the probable outcomes of the project in keeping with the objectives of the course? (5) Is the learning involved in the project of such a nature as to lead on to further important learnings? And, (6) does the project illustrate a biological principle or concept?

Ideas for projects may come from: science teachers or other persons having scientific backgrounds or interests; reading in the various journals such as, Scientific American, Science News Letter, The Science Teacher, The American Biology Teacher, Science World; reading abstracts of scientific papers on biological problems and projects, and subsequently corresponding with the scientist who is working on the problem; and from publications by organizations such as Future Scientists of America, Science Clubs of America, Science Service, and National Science Teachers Association.

Planning the Project

The second major problem is planning the project. This step is very important for proper development of the project. Students who turn out good projects usually make use of the system used by professional scientists who save time and money by "thinking through" their experiments before doing them. Their thinking is compounded of their own training and experience, plus that of their associates and colleagues; and the study of books, technical papers and magazines which contain references to their problems. Often the planning and thinking take longer than the actual doing. This painstaking care in predetermining various possibilities assures better results than helter-skelter, time and money-consuming dabbling.

The booklet, If You Want to Do a Science Project, published by the National Science Teachers Association, gives many valuable suggestions to the student for planning his project work.

Executing the Project

The third major problem is executing the project. In this phase, the teacher should guide the students in carrying out the plans which they formulated during the planning period. The teacher's chief function, however, is to aid in providing favorable working conditions.

This period of execution is also the period in which the results of the project are organized for final evaluation and presentation. These results should be in the form of a report in which the student should ask himself the following questions: (1) What was done? What hypothesis was explored? Where did the idea for the hypothesis come from? Was the hypothesis stated in a way which lead into the project?

(2) Why was this particular approach taken? What about those things which were tried but didn't work out successfully? (These are very important for others who might hope later to reproduce the project.) (3) How successful was the student? What did he find out? Was his work accurate? What did he learn from this project which would allow him to design a more successful project in the future? (4) What good has come from the student's work on this project? Has it opened up any new fields of interest for him? Did it help him see the importance of biology, and how effectively the methods of science can be used to solve problems?

Evaluation of the Project

The last real problem encountered is the judging or evaluation of the project. The final evaluation should be made by the student. He should consider his plans and procedures in the light of his experiences with the project. He should also consider what modifications might have been desirable, whether his source materials were adequate, and the value of the outcomes in relation to the time and effort involved.

Aid from Outside Agencies

Several agencies that are interested in and concerned with schools provide help and encouragement for students working on projects. This aid is in the form of advice, prizes and awards. Science teachers can profitably familiarize themselves with the publications and services of each.

The National Science Fair provides a total of more than $3500 in awards in the form of scientific equipment. It is limited to students in grades ten through twelve who have participated in a local science

fair conducted by Science Service. Science Clubs of America may be contacted for further information.

Science Achievement Awards for Students, sponsored by the American Society for Metals, provides awards worth $14,000 to students in grades seven through twelve. There are eleven geographical regions, with separate awards in each region and in each grade division. Future Scientists of America Foundation should be contacted for further information.

Westinghouse Science Talent Search, co-sponsored by Westinghouse Electric Corporation and Science Service, provides $34,500 in scholarships and forty all-expense trips to a five day Science Talent Institute in Washington, D. C. It is limited to graduating high school seniors who score high on a competitive examination and complete a project report.

Addresses of these and other organizations offering advice and help to the high school student interested in project work may be found in Appendix B of this report.

General Suggestions

Some general suggestions which may help the biology teacher direct project work are as follows: (1) Projects should be reasonably adapted to the student's ability and background. This becomes doubly important when it is remembered that the purpose of these projects is to encourage interest in science. There are few things which will dull interest more completely than repeated failures. (2) Be sure that there is sufficient time for the project. This is especially important in those projects requiring living organisms or cultures which may die and need to be replaced. (3) Try to direct the student into a project for which all or at least a large part of the material may be obtained locally. (4) Do

not try to make each student produce a prize-winning project. Some superior students will of course do excellent work, but the majority should be encouraged to do relatively simple projects. (5) Start the student working on the method of presentation early. Good work is useless if it cannot be reported in a clear concise manner. (6) Try to impress on the student that a simple collection of plants or animals does not constitute a good project nor does the reproduction of some textbook diagram no matter how elaborate. (7) Try to remain patient and receptive to new ideas.

CHAPTER III

TYPES OF BIOLOGY PROJECTS

This section shall be devoted to giving examples of various types of projects suitable for high school biology students. The examples given were chosen because of their great range, as far as student interest and ability is concerned, and serve as a cross-sampling of the many projects available for students.

For the sake of convenience, the projects are placed in one of six different groups. These groups are: (1) General Projects; (2) Collections; (3) Surveys; (4) Experiments with Living Things; (5) Collecting and Caring for Living Things; and (6) Human Health, Physiology and Anatomy. These divisions are quite artificial in nature because any one project chosen will likely overlap into one or more other divisions.

No one may care to follow any one of these projects exactly as outlined (any project given might be used as outlined with good results, but this would be defeating the actual purpose of project work, which is to stimulate original thinking on the part of the student), but a careful examination of them should suggest the many possibilities that lie in the use of student projects.

Group I

General Projects

This first group deals with projects concerned with making things, and with photography and microscopy. Many projects belonging in this group may be safely untertaken by those students who are not of above-average ability, while other will challenge even the most highly advanced students. Seven examples are given.

Making a Model Plant Cell

This is a good example of a simple project that might be made with a little imagination and a few common household articles. A cell model of this type is valuable in understanding the depth and thickness of a cell, a characteristic which is often difficult to see under the microscope.

Materials for this project consist of : pieces of transparent plastic (about the thickness of cardboard); clear cellophane; glass wool; green modeling clay; ping pong ball; and plastic cement.

Construct a rectangular box, about ten inches long, five inches wide, and three inches deep, using transparent plastic. Cement the bottom, sides, and ends with plastic cement. This portion of the model represents the cell wall. Line the bottom, sides and ends with clear cellophane to represent the plasma membrane. General cytoplasm may be represented by the

glass wool. Press this out against the celophane and extend strands through the center, leaving spaces as vacuoles. Construct model chloroplasts from green modeling clay. Make them oval in shape and about one-half inch long. Distribute these through the glass wool cytoplasm. Represent the nucleus with a ping pong ball. Shade the ball with a pencil to represent nucleoplasm and draw in a nucleolus. Chromatin may be represented with netted lines. Embed the ping pong ball in the glass wool. Lay a sheet of cellophane over the top side of the model cell, then add the final plastic wall.

Models of Soil Conservation Practices

A project of this type is very useful to those students who are interested in conservation, particularly those with agricultural tendencies.

Materials needed are: soil boxes eighteen to twenty-four inches square and six inches deep (one for each conservation practice shown); straws or other devices for representing crops; and twigs for representing trees and logs.

To show strip cropping and contour farming, prepare a gentle slope in the soil box, sloping the soil from back to front about half the length of the box and from right to left in the left half of the box. This will form a low hill with slopes in two directions. Divide the slope into strips, each about two inches wide. These strips must follow the contour of the hill. Crops may be indicated with pieces of straw painted different colors with poster paint. Or other devices may be used to represent crops of different kinds. In "planting" the crops, row crops (with straws set in straight rows one-quarter to one-half inch apart)

should be used in some of the strips and cover crops (with straws scattered through the strip) in others. Row crops and cover crops should be alternated. The system used in identifying various crops must be indicated. This may be done by means of a colored key.

To show terracing, prepare a steep slope from the back to the front of a soil box. Cut the slope into level terraces by digging into the hill at the back of the terrace and moving dirt forward at the front of the terrace. Using straws or other devices, plant the terraces with crops. A drainage ditch should be prepared at the foot of each terrace and joined to a ditch running down the hill at the end of the terrace. The banks between terraces should be sloped and planted with small straws representing grass.

To illustrate gully control, a deep, winding gully is made in a sand box. A check dam is then constructed in the lower end of the gully with twigs representing logs. A large gully must be provided with a series of check dams. The banks should be planted with branching twigs representing trees or with pieces of spruce or fir twigs to represent coniferous trees.

Making Skeletons

The making of skeletons is a very worthy project, particularly if a systematic study is made of the comparative skeletal structure of several classes of vertebrates. Skeletons always make a valuable addition to the school museum also.

Necessary materials are: a dissecting set; wire; crocks; thread; hand brush; toothbrush; rubber gloves; strips of wood or cork; hydrogen peroxide; trisodium phosphate; chlorinated lime; and carbon tetrachloride.

High school students usually have less trouble preparing dry ligamentous skeletons than other types. The advantage of making skeletons with their natural ligaments left in place becomes quite evident when mounting the preparation. A minimum of wiring is all that is needed to hold the various parts together and there is no tedious gluing or cementing of small loose bones. The net result being ease of handling, strength, and durability.

A really good skeleton is characterized not only by clean, white bones, but also by a correct and natural arrangement of bones, and a type of mounting that is sturdy enough to withstand much handling without deterioration. In order to have a good skeleton, a large, perfectly formed, mature specimen must be selected. One which has fully developed teeth and bones that are entirely ossified. Only fresh or salted specimens should be used.

The fleshing of an animal is one of the most important operations in preparing a skeleton. Assuming that a fresh specimen is at hand proceed by first removing the skin. If the animal has clavicles of the false type (cat) these should be dissected out first. In case of true clavicles, dismember at the coracoid process and leave attached at the episternum. Remove the four limbs intact and remove the skull by carefully cutting between the atlas and condyles. Carefully dissect out the tongue bones. This series of bones lie in the muscles of the throat and should be removed as a unit with their connective ligaments intact. Flesh the skull carefully, removing the eyes and as much muscle as possible. Separate the mandible from the rest of the skull. Remove the brain with a brain spoon or similar instrument. Next remove the flesh from the limbs, but leave the ligaments that hold the bones together. Use special care when

fleshing the hind limbs. Do not cut away the knee bones as these are floaters connected to the tibia by a ligament. Remove the fatty tissue in behind the patellae. Eviscerate the body of the animal and proceed to flesh it out, again being careful to leave the ligaments that hold the bones together. Flesh out the tail. In working on the trunk be careful not to cut through the costal cartilages of the sternum. Now go back over the various bones to be sure that all of the flesh has been removed thoroughly. If too much flesh has been left on the skeleton the maceration takes place too fast and in a short time the bones will become separated. Pass a stiff piece of wire through the neural canal, moving the wire back and forth so as to clean out the spinal cord. The skeleton is now ready for maceration.

Place the bones in a glass or earthenware container of suitable size and cover the bones with cold tap-water. Do not allow any foreign substance to get into the maceration jar as this will discolor the bones. The maceration jar should be kept at room temperature and the water should be changed daily, replacing it with fresh water each time. At first a great deal of blood will be evident at each change, but will gradually diminish as it is extracted from the bones. This will take two to three days depending on the temperature of the room. During this time rotting is taking place on the flesh that was left adhering to the bones. When the bath becomes clear, pour off and place the bones in a solution made up of one ounce of trisodium phosphate to each gallon of water. Stir well until the trisodium is dissolved, and leave the bones in this solution from twelve to twenty-four hours. This bath serves a dual purpose in that it halts the maceration and also swells and loosens the tissue remaining on the bones.

Remove the bones from the trisodium phosphate solution and let drain. Secure a hand brush with stiff bristles, an old toothbrush, chlorinated lime and lots of hot water. Dip the brush in hot water, then into the chlorinated lime; now brush the bones using short rapid strokes (use the toothbrush in small hard to get places and on small bones). The action of the hot water and lime plus the friction of the brush creates a burning action upon the tissues adhering to the bones and these virtually disappear as the liming progresses. The use of rubber gloves in this operation is recommended to protect the hands. Rinse in cold water frequently, watching the removal of the flesh carefully. Brush or lime until the flesh has been removed entirely but not the ligaments holding the bones in place. Rinse thoroughly in cold water and lay out to dry at room temperature.

When the bones are thoroughly dry they will contain a certain amount of grease which is objectionable in a good skeletal preparation and should be removed. Place the dried skeleton back into the jar used for maceration and cover with carbon tetrachloride. This chemical has dangerous fumes so prolonged breathing of the fumes should be avoided. Cover the layer with water which will float on top. This prevents excessive evaporation. Cover the container and let the skeleton remain in this degreasing fluid at least a week. Remove and let air dry.

For bleaching, place the bones in a 3% solution of hydrogen peroxide for ten to twelve hours. Remove from the bleach and rinse in cold water.

The skeleton is now ready for drying in its natural position preparatory to mounting. Run a stiff, straight wire pointed at one end down the neural canal and anchor it securely in the sacrum. Bend the vertebral column into its natural shape. While the skeleton is still wet, space

the ribs equally by stringing with thread. Pin out the limbs in shape on a soft piece of wood or cork and allow to dry.

Mounting is largely a matter of experience and skill. Anyone with a little mechanical ability can work out methods of mounting on stiff wires or (in the case of large skeletons) on metal rods and tubes. In general it is wise to enclose small skeletons in glass cases, as this protects them from dust and careless handling.

Making a Photographic Record

There are always a few students interested in photography. This type of project has special appeal to them. Equipment may range from the cheapest box camera on up to the most expensive.

Necessary materials are: a camera; any special equipment necessary, depending upon the needs of the particular project; paper for mounting pictures; and a folder for keeping the record.

Keeping a photographic record of some phase of biology can become a lifetime hobby or even a profitable business. In either case, it can provide the student with an enjoyable addition to his study of biology. Photographic needs will depend upon the type of record made. For a study of environments almost any inexpensive camera will do. Photographs of deciduous forests, evergreen forests, deserts, swamps, ponds, lakes, streams or any number of other environments may be made. Pictures should be given titles and put into a folder. On a page following the picture animals and plants found in such a place should be listed. Pictures of specific animals and plants should also be taken. In this way the student can assemble pictures and facts about a particular community or about several types of communities near his home, and in so doing make a careful

study of them and their inhabitants.

Simplified Photomicrography

Photomicrography offers many interesting opportunities to the student interested in the study of microscopic life and cellular structure. Equipment can be very elementary. Any kind of microscope and almost any kind of lamp can be made to work. However, the quality of the picture will always be dependent on the accuracy of applied technique and cannot exceed the degree of optical perfection in the microscope. The optical qualities of all the microscope powers, in turn, cannot be utilized fully without a suitable light source equipped with condenser and iris diaphragm.

Necessary equipment includes: a camera; a microscope; lamp; block of wood; tissue paper; ground glass; two or three-power magnifier; and cardboard tubes.

The miscroscope is capable of projecting a magnified real image. Photomicrography consists essentially of timed micro-projection to a light-sensitive photographic emulsion held in place within a non-reflecting light-tight container (the camera). The treatment of films or plates and printing procedures are simply a matter of ordinary photographic technique; such information is generally furnished by the manufacturer of photographic supplies.

The first problem is that of adapting the camera at hand for photomicrography. With the common roll-film camera some way of focusing the individual pictures must be found. Most 35 mm. non-reflex roll-film cameras can be adapted directly to the microscope with apparatus which provides a means of focusing each frame. Such equipment generally uses the camera without lens. It is, however, possible to employ the complete

camera for photomicrography by the following technique:

To align the camera and microscope horizontally, mount either the camera or microscope and lamp on a block of wood of such height that the center of the camera lens will occupy the same level as the center of the microscope ocular. Focus the microscope on a transparent object or stage micrometer lines. Illuminate strongly. Remove the back of the camera and replace the camera in the line-up with the shutter closed. The degree of centration may be gauged by observing the position of the microscope eye-point on the front lens of the camera. (The rays of light, on leaving the microscope, converge to a focus above the eyepiece and form the eye-point--an image of the objective's aperture. A piece of tissue paper, moved slowly from a point close to the ocular, will reveal the location of the circular eyepoint disc.) Make any necessary adjustments to obtain mutual centration. The camera distance from the microscope is correct when the image of the microscope eyepoint is smallest on the camera lens. (If the camera is brought closer than this to the microscope, some loss of definition will result. Should the camera distance be increased beyond the critical plane, aberrations are introduced and the edge of the camera lens will act as a field diaphragm, cutting off some of the image. The iris diaphragm of the camera has no useful function in photomicrography and must be kept open at all times.)

Next open the shutter of the camera and set the lens at infinity. View the microscope image on a piece of ground glass placed in the film plane. Center the image and recheck the centration of the eyepoint on the camera lens. If now a series of guide stops are set up which will always restore this exact alignment of camera and microscope (whenever the camera is replaced after direct viewing through the microscope) it will be

possible to proceed with photomicrography after the loaded camera is brought into light-tight connection with the microscope provided the operator is able to focus the microscope at infinity. Since few observers can be certain of viewing the virtual microscope image at infinity, it is suggested that a special viewer be made which eliminates all uncertainty. It consists of any kind of two or three power magnifier mounted in one end of a cardboard tube and a piece of ground glass (ground side outward) attached to a second tube which is capable of telescoping into the first. (The total length of the finished product can be determined in advance by noting the magnifier-to-ground-glass distance when a distant out-of-doors object is brought to a focus on the ground glass.) Fix the relative positions of both tubes permanently with tape when the infinity setting has been verified. By holding the viewer over the eyepiece of the focused microscope, with the lens end of the viewer near the eyepoint, an image can be seen on the ground glass of the instrument. When the image on the ground glass of the viewer is brought into critical focus by a turn of the fine adjustment on the microscope, the infinity focus has been attained and the microscope may then be coupled with any camera which is also focused for infinity. If it is desired to examine the microscope image on the gound glass of the viewer in more detail, the focusing instrument may be converted into a telescope by ruling two crossing pencil lines on the center of the ground glass, placing a drop of Canada balsam on the area and covering it with a cover glass. The mounted portion will then become clear except for the pencil lines. (The ground glass focusing plate of the photomicrographic camera may be converted for clear glass focusing in the same way.) The microscope image may now be focused in the plane of the crossing lines with a second magnifier. The latter can, for convenience,

be mounted in a tube sliding over the original viewer. However, if a box camera or camera of fixed focus is used with its lens, then an object about twenty-five feet distant should be brought into focus on the ground glass viewing device--to match the focus of the camera. When these adjustments are completed, exposure of the film can be effected in the usual way with the camera shutter. The photomicrographic unit may also be set up vertically if a sturdy ring stand or improvised apparatus can be provided. However it is accomplished, alignment is made in the same manner as for the horizontal set-up. Troublesome vibration can be eliminated to a great extent by mounting all the apparatus on a single board and setting the latter on several thick pads of sponge rubber.

Preparing Microscope Slides of Simple Objects

Permanent mounts suitable for study under the microscope may be made without the expenditure of a great deal of time or the purchase of expensive apparatus. The necessary materials can be obtained easily or are such as can be found readily in the high school laboratory.

Materials should include: a pair of fine pointed forceps; dissecting scissors; a few dissecting needles; a small camel's hair brush; a section lifter; plain glass slides; cover glasses; a dozen or so small flat dishes with covers; a few bottles for reagents; and a good razor or one of the simpler microtomes.

Absolute alcohol and 95% alcohol are also necessary for slide preparation. Iso-propanol may be substituted for ethyl alcohol in all microtechnique procedures with quite satisfactory results. The 95% alcohol is usually denatured ethyl. A series of varying strength alcohol solutions should be made up from the 95% alcohol. These are 35%, 50%, 70%, 83%, and

95%. Make as follows: For 70%, use 70 units of 95% and add distilled water to 95 units. Others are made similarly. Other necessary chemicals are Canada balsam, xylol, clove oil, oil of wintergreen, formalin, and acetic acid. The most useful stains for routine work are hematoxylin, eosin, safranin, methylene blue, and light green.

Objects for microscope study are of three general types--whole mounts, teased mounts, and sectioned objects.

Whole mounts are preparations of the object entire. Many interesting and instructive mounts may be made without cutting sections, and without staining. Fibres of cotton, wool, silk, hairs from mammals, feathers, scales, wings and antennae of insects, and other like objects may be dropped directly into xylol, placed in a drop of balsam on a slide, and covered. The microscope slide and cover glass should be cleaned thoroughly with alcohol. After the cover glass is placed over the balsam, the balsam should spread into a thin film under the glass. The excess balsam may be wiped from the edge of the cover glass with a cloth dipped in xylol. The slide should be set aside until the balsam hardens. Many very beautiful and interesting mounts can be made in this way. Plant materials which may be given the same treatment include fern sporanges, the fruiting heads of mosses (if dried first) and pine pollen. Some objects need to be dehydrated before being mounted, but need not be stained. Small insects, after killing with chloroform, may be dropped into aniline oil or into carbol-xylol (carbolic acid crystals one part--xylol two parts) and, after half an hour or more, transferred to pure xylol. Fleas taken from various animals make very nice mounts. Usually there will be a different kind of flea on each kind of mammal. The mouth parts of various insects may also be used, such as the honey bee, butterfly, true bug, house fly, horse fly,

or grasshopper. The legs of the honey bee have many interesting structures, such as the pollen basket, pollen comb and wax shears on the third leg, and the antenna cleaner and eye brush on the first leg. These, as well as the legs of many other insects, are easily mounted by the above method. With a little imagination and patience a nice series of slides of this nature may be developed.

Teasing means dividing or separating the component parts of an organism or other object. Thus one may remove part of an organism, separate it by means of needles, tearing or shredding until it can finally be pressed out on the slide, and stained if desired. After the cover glass is attached a great many details can be studied that were invisible when the object was observed as a whole. This method is applicable to parts of insects, parts of smaller animals, and as well to some plants.

Teasing is often combined with maceration in the preparation of microscope slides. In this latter process the tissue is first placed in a fluid which will cause softening of the cement substance which holds the elements together. It may then be teased, stained and mounted (following the schedules outlined on the next page for plant or animal sections). Macerating fluids include 30% alcohol, 0.5% formalin and weak acids.

Free-hand sections of cork, pith and the stems and roots of many of the common plants may be cut. For this purpose the razor must be sharp and free from nicks. Better results will be obtained if the object can be clamped into a simple microtome. In any case only the thinnest sections should be used, and they may be placed into 95% alcohol as cut. Sections of a few animal tissues can be prepared in this manner also, especially such objects as liver, kidney and muscle, but the tissue must first be thoroughly hardened in 10% formalin, having been removed from a freshly

killed animal and placed directly into formalin. After cutting, sections of animal tissue must be placed first in water. Subsequent treatment of sections in each case is as follows. Handling should be as gentle as possible, using section lifter and camel's hair brush.

Plant sections

1. 95% alcohol--½ hour.
2. 50% alcohol--5 minutes.
3. Water--5 minutes.
4. Safranin--4 hours (1% solution in water).
5. Water to rinse.
6. 50% alcohol--5 minutes.
7. 95% alcohol--5 minutes.
8. Absolute alcohol--5 minutes.
9. 95% alcohol containing 1% Light Green--1-3 minutes.
10. Absolute alcohol--3-5 minutes.
11. Clove oil--5 minutes.
12. Xylol--indefinite.
13. Mount in balsam; cover with cover glass.

Animal sections

1. Water.
2. Delafield's hematoxylin--5-10 minutes.
3. Water to rinse.
4. 35% alcohol containing ½% of hydrochlocic acid. (Watch section and remove from acid when light red.)
5. Tap water until section becomes blue.
6. Eosin (½% solution in water)--1-2 minutes.
7. 35% alcohol--1 minute.
8. 50% alcohol--1 minute.
9. 70% alcohol--1 minute.
10. 83% alcohol--1 minute.
11. 95% alcohol--1 minute.
12. Absolute alcohol--1 minute.
13. Absolute alcohol and xylol (half and half)--1 minute.
14. Xylol--indefinite.
15. Mount in balsam; cover with cover glass.

The time values given are approximate, and will vary somewhat with the kind of section being stained. Practice on a few sections will teach the student whether to hasten or retard the process.

There are numerous animal and plant materials from which successful permanent mounts cannot be made by the methods given above. They require special fixations of various sorts, long and complicated processes of

dehydration and clearing, embedding in special media such as paraffine or celloidin, and sectioning on one of the larger automatic microtomes. Slides of such materials cannot be successfully prepared in the high school laboratory unless special equipment is available and the instructor has previous training and experience in making slides. Many good books are available on microtechnique and several should be present in the laboratory for reference.

Preparing Bacteria Smears

The preparation of bacteria smears makes an excellent project if some specific group of bacteria is studied, for example bacteria of the mouth. This is a project that should interest most students with tendencies toward becoming doctors, nurses or dentists.

Materials necessary are: bacteria colonies growing on culture media; inoculating loop; burner; methylene blue stain; slides and cover glasses; distilled water; alcohol; and a microscope.

The smear may be made either on a cover glass or on a slide. Dip both slide and cover glass in alcohol, then wipe dry. Hold the end of the slide between thumb and forefinger, then invert it. Pass the center of the slide through a flame three times. Lay the slide down, flamed side up. Sterilize an inoculating loop in the flame, then dip it into a beaker of distilled water. Transfer a loopful of water to the slide. Sterilize the loop again, allow it to cool a few seconds, then touch it to a colony of bacteria. Transfer the bacteria to the drop of water and spread them into a thin, uniform film, slightly smaller in diameter than a cover glass. Sterilize the loop again. Allow the film to dry completely in the air. Then, turn the slide with the smear down and flame

it three times, being sure that the flame reaches the smear. Flood the smear with methylene blue for two or three minutes, then wash off the excess stain in a jar of water or a gentle stream from a faucet. Wipe off the excess water around the smear and on the lower side of the slide. Place a cover glass over the smear, adding a small quantity of water if necessary and examine under high power.

Group II

Collections

Collections are possibly the most misused of all projects. In order to be worthy of the time consumed, a collection must be specific enough to be a true contribution to science. For instance, a collection of "Flowers" could hardly be considered worthy, while a collection of "Spring Flowers of Hidalgo County, Texas" would be a contribution indeed. This difference between a good and bad collection should be impressed upon the student at the very first. Collections, of course, vary with the student and his interests. Three examples are given here out of the hundreds that are possible.

Preparing a Herbarium Collection

Not only is a herbarium collection a good project, it is also an excellent teaching aid that may be used year after year. The equipment of the amateur collector need not be too elaborate, but there are several items which each collector of botanical specimens should have.

Necessary equipment includes: trowel or pick; vasculum or portfolio type field plant press; heavy laboratory plant press; blotting paper or plant driers; corrugated cardboard; collecting sheets; mounting sheets;

glue; gummed tape; labels; notebook; waterproof ink; and pen or pencil.

As plants are procured they are placed in collecting sheets in which they are kept until they have been pressed and are ready for the mounting sheets. These collecting sheets are folders of unglazed paper. The most practical size is 16½ by 23 inches; when folded 11½ by 16½ inches. Old newspapers cut to the sizes mentioned above make good collecting sheets and a supply of them should be included in every collector's equipment.

A plant consists of many parts, often repeated in numbers such as roots, stems, leaves, flowers and fruit. A good herbarium specimen should have each of the parts represented. It is also desirable to have these arranged on the finished sheets in such a way that their relation to each other can be shown. It is impossible in the case of many of the larger plants to collect the entire plant, but this should be done whenever possible. Herbaceous plants two feet high or less should be collected entire, including a portion of the root, and bent in a V or N shape, when necessary, to get them on the collecting sheet. In taking parts of shrubs or trees, a branch about a foot long should be collected which contains representative leaves, flowers, and fruits, when possible. It is desirable to have both the staminate and pistillate flowers, although these may be found on different parts of the same plant or on different plants. In addition to the leaves which are found on the terminal branches, leaves should be collected from the lower parts of the branches. Also a portion of the bark should be taken.

If the fruits of woody plants are too thick to be placed in a press they should be put in a separate container and labeled for future reference. In the case of large herbs where the entire plant cannot be folded to the size of a herbarium sheet it is advisable to collect the flowering and

fruiting portions, leaves from the upper and lower parts of the stem and representative portions of the root.

When it is necessary to bend the stem of a plant to make it fit the collecting sheet a small piece of cardboard about the weight of a postal card may be used for holding the stem in position and keeping it from springing outward and extending beyond the edge of the collecting sheet. The card is prepared by cutting a small slit in it. Place the V-shaped bent portion of the stem through this slit. In this way a neat specimen is obtained.

Many delicate flowers will collapse even if perfectly fresh when placed in the press; but it is possible to get perfect specimens of these by applying bits of moist paper to the fresh flower and spreading the petals when the plants are placed in the portfolio. It will be found that some parts of herbaceous plants are too thick to be placed in the collecting sheet in the usual way. In cases of this kind it is best to split these before placing them in the collecting sheet. After this is done they may be dried in the usual manner.

Many of the fine-leaved water plants will collapse entirely if dried by the usual method. These should be rolled up in wet paper when in the field and brought back in this condition. After they are in the laboratory they should be placed in water and floated out on sheets of white paper. These sheets should then be carefully taken from the water so that the fine divisions of the leaves do not cohere. These white sheets may then be placed in the collecting sheets and the specimens treated exactly as others.

Marine algae and some kinds of fresh-water algae can be floated onto the herbarium mounting paper in a shallow pan and arranged while under

water. The paper is then carefully removed from the water and the specimen is allowed to dry without pressing. Most algae will stick to the paper.

After the specimen has been collected and placed in the collecting sheet, put it in the collecting press. For the proper arrangement in the portfolio, first place a sheet of blotting paper or plant drier in the portfolio, then the collecting sheet, then another plant drier and so on. As soon as the portfolio is full it is strapped up and if several are used each one should be numbered.

It is of first importance in making a herbarium collection, that notes regarding the date, locality, habitat, height, method of branching, color of flower parts, and common name, should be kept. These should either be keyed with the collecting sheet or placed in the collecting sheet with the specimen until they can be recorded on the permanent record. It is also highly desirable to secure photographs of individual plants if possible. These are not only interesting, but are also of great value for future reference to the herbarium collection.

It is necessary to make some arrangements whereby the specimen may be associated with its data. This may easily be done by giving the herbarium specimen a number and placing this number in the field notebook opposite the information regarding that particular specimen. This number should appear with all of the parts of the plant in question. After the specimen is permanently mounted on a herbarium sheet this information can be rewritten and possibly added to, to a certain extent, and placed in the permanent notebook.

Specimens should be taken from the portfolio and placed in the laboratory press as soon as possible after returning from the collecting trip.

This is particularly true in hot weather. In some cases they may be left over night if the portfolio is strapped up tightly, but as a rule, this is inadvisable.

In pressing the herbarium specimen it must be remembered that the specimen will be the same size and shape when it comes out of the press as it is when it goes in. This means that the specimen must be neatly arranged in the proper position within a space which will allow it to be fastened to the herbarium sheet after it has been dried. Both sides of leaves of plants should be shown when the specimen is mounted. This may be arranged when the specimens are limp after their first day in the press.

For arranging the specimens in the press use first a blotting sheet, then the collecting sheet with specimen in place, then a sheet of strawboard, another drier, and so on. The clamps or straps of the press should then be tightened and the press placed in a warm, well-aired place to dry. After the specimens have been in the press for twenty-four hours they should be examined and new driers exchanged for the old ones. The damp driers may be laid out in the sun to dry.

For average specimens two or three changes of driers should be sufficient if plenty of strawboards have been used. It should not take more than a week or ten days for the average specimens to dry, while very thin or delicate ones, such as fine grasses and ferns, will be dry in two or three days. Very fleshy plants will require a longer period of time.

When dried, the specimens in their collecting sheets should be neatly stacked and each specimen identified. The specimen is then ready for mounting.

The standard size mounting paper is $11\frac{1}{2}$ by $16\frac{1}{2}$ inches. This paper should be of a good weight and not too flexible. It should also be of a

quality which will not readily turn yellow with age.

Three methods are used for attaching the specimens to the herbarium sheets. (1) The glue is spread on a glass plate, the specimen laid on the glue and lifted as soon as all parts have come in contact with the glue, then transferred to the paper. (2) The specimen is inverted on the paper where it has been stored, painted with glue by means of a brush and then transferred to the mounting sheet. (3) The specimen is laid on the mounting sheet and fastened there by means of small strips of gummed tape which are used in large enough quantity to securely affix the stems, petioles, flower stocks, leaf tips and other parts. If either of the first two methods are used it is also well to use some of the small strips of gummed tape to fasten the specimen more securely to the sheet.

After the specimens are mounted, the label should be placed in the lower right-hand corner. This label should contain as much information as possible regarding the specimen, including the common, scientific and family names, name of collector, collector's number, date, place collected, and remarks.

The teaching collection of herbarium mounts is usually handled quite frequently and is subjected to a certain amount of abuse. For this reason cellophane is often used in covering the mounted specimen. Cellophane sheets, $12\frac{1}{4}$ by $17\frac{1}{2}$ inches, are placed over the specimen and the upper edge folded under the edge of the herbarium sheet. This upper edge, about one inch, is glued to the backside of the herbarium sheet. The lower corners of the cellophane are then fastened to the corners of the herbarium sheets by means of paper clips. This makes it possible for the cellophane to be rolled back should it become necessary.

After the specimens have been mounted they should be arranged according

to their classification so that any specimen may readily be located when reference is made to it in class work. In doing this it is well to divide the specimens into groups according to species, genus or family. Each group should then be placed in a folder slightly larger than the herbarium sheets--12 by 17 inches. Each folder should be labeled in one of the lower corners where the label may be seen without removing the folder from the file.

The collection may be stored in any kind of case that will keep out dust and moisture. Herbarium specimens are often attacked by museum pests. To guard against the collection being ruined by these pests, the specimens should be fumigated with carbon bisulphide three or four times per year.

Leaf Prints

Making leaf prints is an excellent way to show leaf shape, margin types and venation. There are several methods of making the prints, four of which shall be discussed here.

Materials include: carbon paper; blueprint paper; photographic printing paper; ordinary white paper; old newspapers; electric iron; glass plates; rubber bands; printer's roller; and printer's ink.

To make leaf prints with carbon paper, a fresh leaf is placed on a sheet of newspaper. A piece of carbon paper is layed over the leaf with the carbon side next to the leaf. It is then covered with another sheet of neswpaper and pressed firmly with a hot electric iron. The leaf is removed and placed, carboned side down, on a sheet of paper to receive the print. The leaf is covered with newspaper again and pressed with the hot iron. This transfers the carbon print to the sheet of paper.

In using blueprint paper for making leaf prints, a dark room must be

used. Cut a sheet of blueprint paper to the size wanted and place the sensitive side up on a sheet of heavy cardboard of the same size. Lay the leaf or leaves to be printed on the paper, then cover with a sheet of glass. Fasten the layers firmly with rubber bands or twine. Expose the mount to bright sunlight for eight to ten minutes. Return to the dark room and put the paper into a pan of cold water until the blue color is solid. If the color is splotchy, the exposure was not long enough. Lay the paper between sheets of newspaper to dry, and put a weight on it to keep it flat. If spaces to label each leaf print are desired, clip pieces of cardboard to the blueprint paper in suitable places before it is exposed to the light. This will leave white labeling spaces after the paper is developed. This method is good to show leaf shape and margins, but will not show venation.

To make a photographic print, technique similar to the above is used. Comparatively thin leaves should be used for this type of print. In a dark room, place the leaf in the center of a piece of photographic printing paper. Place both between two pieces of glass to flatten out. Expose to light the same as for ordinary contact prints and develop the same as ordinary printing paper.

Making leaf prints with printer's ink is a little messy, but with some practice good results may be obtained. Using a printer's roller, spread some printer's ink onto a pane of glass or other smooth surface. Place the leaf on this surface, veined side down, and press. Now place the leaf on a white piece of paper and press down with a roller or with the fingers. Remove the leaf slowly, being careful not to smudge. The ink print of the leaf veins will show very nicely on the paper.

Making an Insect Collection

Insect collections are among the most challenging of all collections due to the vast numbers of insects and the multitude of biological principles and concepts which may be illustrated by them. For example, collections may be made to show types of harmful insects, beneficial insects, social insects, protective adaptations in insects, mimicry, orders of insects, and so forth. The possibilities are endless, and a good collection is a welcome addition to the school museum.

Materials include: water and sweep nets; pint jars; killing jars; vials; 80% alcohol; 95% alcohol; carbon tetrachloride; insect pins; cigar boxes; corrugated cardboard; forceps; labels; white shellac; India ink; coffee can; spreading board; and Paradichlorobenzene.

All specimens in the collection should be in first-class condition, correctly mounted, and each should bear a locality label indicating where and when it was collected, as well as a label showing the scientific name of the insect.

Aquatic collecting should be done with a water dip net. The net may be worked from side to side in the water or swept over the water weeds. In swift-flowing streams the net may be held against the bottom of the stream while the stones just above the net are moved. Many forms will float with the current and be caught in the net. Other aquatic insects may be picked from the undersides of stones and sticks. Practically all aquatic insects may be preserved in 80% alcohol. Wide-mouth pint jars make ideal insect-collecting jars.

Terrestrial collecting may be divided into various types. Many insects, particularly beetles, can be captured by moving stones or sticks or by digging them out of rotten logs. Others can best be collected by

"sweeping." This is a term applied to collecting insects by passing the net rapidly back and forth over the grass and weeds. If much of this type of collecting is to be done, a sturdily made net should be used. For collecting butterflies and for other light insect work a lighter net may be used.

Insects are killed in a "killing jar." These jars may be of various sizes and the killing agents also vary. One of the easiest to make and safest to use is the carbon tetrachloride killing jar. Refrigerator jars of the peanut butter or pickle variety are excellent. About a fourth of an inch to half an inch of cotton should be placed in the bottom of the jar. A round piece of cardboard punched full of small holes is then placed over the cotton. The cardboard should fit the jar snugly. Carbon tetrachloride may be added to the jar as needed. Usually a few drops will charge a jar for several hours. Care should be taken not to inhale the fumes or spill the liquid on the skin. A piece of paper toweling wadded up and placed in the jar will keep the more delicate insects from injuring themselves before they are killed. The lid should be kept tightly closed at all times.

In addition to the killing jars it is well to take several bottles of 80% alcohol into the field. This is for the softer bodied insects, insect larvae and foul-smelling carrion bettles. Spiders should be collected and preserved in 95% alcohol. Immature stages of most insects, and the adults of a few orders, are preserved in 80% alcohol. Most adults, since they have a chitinous exoskeleton, can be collected and killed and then mounted on insect pins.

Night collecting is one of the best ways to obtain moths and many other nocturnal insects. One of the simplest and most effective ways to

do this is to hang up a white sheet and focus on this white surface the rays of a strong flashlight or a portable spotlight. Many night-flying insects will be attracted to this illuminated white area, where they are easily collected. Lighted insect traps of many kinds can also be used for this purpose. Warm, calm evenings are best for insect collecting.

Insects collected in the field should be mounted the day after they are collected. If they are kept in a tightly closed jar they will remain relaxed for a day.

Cigar boxes make excellent pinning boxes. These may be prepared by securely fastening in the bottom of each a layer of soft balsa wood or pressed cork. Corrugated cardboard may also be used. Four large cigar boxes furnish ample space for starting the collection.

Special insect pins should always be used in mounting. Number three is the most satisfactory size for most common species. Insects should always be handled with forceps to avoid breakage. Fine-pointed curved forceps are probably the best for most work.

Labels can be made from good quality paper. Order and family labels should be outlined with black, while genus and species labels should be plain. The black border may be made by means of a drawing pen and India ink. Order labels should be $2\frac{1}{4}$ inches long by $\frac{1}{2}$ inch wide; family labels $1\frac{1}{2}$ inches by $\frac{1}{2}$ inch. Genus and species labels should be the same as family labels.

The method of mounting differs in different orders of insects. Orthoptera, Neuroptera, Ephemerida, Odonata, Homoptera, Mecoptera, Trichoptera, Lepidoptera, and Hymenoptera should be pinned through the middle of the thorax. In some cases it is best to put the pin a little to the right of the center in order not to destroy any structure which may be in the median

dorsal line.

Hemiptera should be pinned through the thorax (scutellum). Coleoptera should be pinned through the right wing, a little less than one-fourth of the length of the wing from its base. Diptera should be pinned through the mesothorax, a little to the right of the center.

Many small Hemiptera, Homoptera, Coleoptera, and Hymenoptera must be mounted on cardboard points. The points should be triangular in shape and about one-fourth inch long and one-eighth inch wide at the base. These are conveniently made with a punch or cut from heavy glazed paper. The specimen is attached to the point with white shellac. It should be on the left side of the pin when the head end is directed forward.

Small Lepidoptera are pinned on minute pins fastened to a small piece of balsa wood or cork. The insect pin is then thrust through the wood and the specimen adjusted to the proper height. Small Diptera may be mounted like small Lepidoptera or they may be glued to the pin by their right side at the same height they would have been if the pin were thrust through the body. Diptera should never be mounted on paper points.

In small collections, the larger butterflies and moths are best pinned with the wings slightly spread, which avoids the spreading of the wings on spreading boards. It is impossible for the average collector to identify many butterflies further than the family, so spreading is not usually necessary.

In the case of large specimens it is well to place a drop of shellac where the pin enters the insect. This holds the insect firmly on the pin and prevents the danger of its swinging around and breaking adjacent specimens.

The locality label should be as small as possible and should contain

the following information: the locality, date, and name of collector. This lable should be on the pin below the specimen, leaving about the same amount of space between the label and insect as there is between the insect and the head of the pin.

For specimens in vials the same information should be written with India ink on a small bond-paper label, which is placed inside the vial.

The scientific name of the insect should appear on a label which lies against the floor of the box or case. It should be pinned in place by the pin which carries the insect. When more than one specimen of the same species is to be mounted, each insect may be labelled or only the first.

The order and family labels are placed on the bottom of the case ahead of the insects belonging to that group.

In many cases it is not practical to pin out and dry Lepidoptera in the field while intent on collecting. These specimens may be dried with their wings folded and then placed in envelopes or triangles. This facilitates the storing and handling of the specimens until they are wanted for mounting and added to a collection.

The method of relaxing these dried and folded specimens is as follows: Secure a wide-mouth jar or can with a tight-fitting cover (a coffee-can is good for this). Into this container pour one or two inches of clean sand; saturate the sand with water to which has been added a few drops of carbolic acid. This keeps mold from forming. Cover the sand with cardboard so that the insects do not come in contact with the wet sand. Place the insects to be relaxed on the cardboard and close the container tight. Do not place the jar or can where it will be exposed to excessive heat, as this will cause the container to sweat on the inside.

Ordinary room temperature will do. In one to three days (depending on size) the specimens are relaxed and can be pinned out as readily as a fresh insect.

Remove the specimen from the relaxing can, spread the wings and insert a pin through the thorax. If a spreading board is available pin the specimen in the groove of the board at its proper height; with a pair of forceps pull the forward wing up until its hind margin is at right angles to the body. Now place a thin transparent strip of paper (wax paper is excellent) over the wing and pin in place. Do not put pins through the wing. Now bring the hind wing forward until its front margin just touches the hind margin of the forward wing. Bring the paper strip down over the hind wing and pin in place. Another strip may now be added if necessary. Proceed the same way with the other side.

If no spreading board is available, a piece of cork, balsa wood or other suitable pinning material may be used. Proceed as follows: Instead of putting the pin through the dorsal portion of the thorax put it in from the ventral side. Now spread the wings and pin the specimen securely on its back. Proceed to pin out the wings in the same manner as with a pinning board. The only other requirement in this method is to remove the pin when the specimen is dry and reverse it, putting it through the same hole from the dorsal side. Fasten the insect to the pin with a small drop of shellac or cement to prevent the specimen from turning. A pinning board is best to use since the less butterflies are handled the better the specimen will be.

After a collection of pinned insects has been made it must be examined three or four times a year for museum pests. Their presence is readily detected by sawdust-like material around the bases of some of the pins.

These pests can be killed by pouring a tablespoon of carbon bisulphide, benzene, or carbon tetrachloride and keeping the box tightly closed. Since carbon bisulphide is highly inflammable, extreme care should be taken to keep all fires away.

The best general protecting agent or fumigant is Paradichlorobenzene which is harmless to humans, but will successfully kill museum pests.

Group III

Surveys

One of the most interesting projects for a biology class, and one in which any number of students may participate, is the making of a biological survey of the region in which the school is located. Field work of this kind, when properly directed, has the tremendous advantage of making students realize that biology is "at their doorstep." They cease considering biology as a dry textbook subject when they recognize the fact that countless new and interesting happenings in the plant and animal kingdoms are taking place all around their school. In making a neighborhood biological survey, one student or a small group of students should specialize on one particular part of the survey. Records should be kept of the species identified and when possible specimens should be brought to the laboratory. Surveys may range from making a local bird study to showing the distribution of bacteria in the hallways. Three examples of survey projects are given here.

Bird Study

Certain phases of biology apply especially to field study. Among these, bird study is probably most ideal. The bird population is much

more abundant than a casual observation might indicate. Bird study combines the thrills of exploring different habitats with scientific observation, preparation of field notes, and classification. It is an ideal combination of scientific research in the field and a thoroughly enjoyable hobby. About all that is needed in the way of equipment is a good pair of binoculars (7 x 50 is best) and a field guide on birds.

The scientific study of bird life will take the student into a variety of habitats. Birds occupy specific surroundings. Certain species will be found in the open fields, others in the trees and shrubs of deep woods, on the forest floor, along the shores of rivers and lakes, in the open water, or in the tall grasses and cattails of a marsh. In the course of the bird study, the student should visit each of the different habitats in his region several times. The study should be conducted over a period of several months, including more than one season. Over such a period, one might observe permanent residents, winter residents, summer residents, and migratory birds during the fall and spring.

Most of the students' success in observing birds will depend on their skill in the field. Work should be conducted individually or in small groups, never in large groups. A habitat must be approached with as little noise and confusion as possible. In some cases, it is better to conceal one's self behind a cover or in a temporary blind and wait for the birds to come to the observer. This is especially true in the case of shore and water birds.

The student's ability to identify a bird or to make observations which will lead to its correct identification will improve with practice in the field. The first observation should concern size. Since an estimate of the length of the bird in inches is very difficult, bird students

often compare a bird with three common examples: the English sparrow, robin and crow. The general color, both above and below, should be noted. In addition, special markings, such as wing bars, stripes on the crown, rump spots, and cheek bars, are extremely important. Other points to look for include top knots, length of tail, general shape of the body, shape and color of the beak and, in some cases, form and color of the feet. In most cases, a pair of field glasses or binoculars are essential for making these close observations. Those having a magnification of about seven or eight and an objective of 50 mm. are usually considered best for this type of field work.

Each bird studied should be entered under a number in a field notebook. In the case of a bird whose identification is positive, only its name and the date seen should be entered. In the case of those birds which have not been positively identified, include data pertaining to the place observed, date seen, type of habitat, size and color, distinctive markings, and any other peculiarities noted.

The most satisfactory method of identifying an unknown bird from field notes is by the use of special field keys and identification books. Pocket-sized editions of these are available and should be consulted at the time the field study is being made. Peterson's guides are usually considered to best for identification in the field. Larger bird books may be consulted at school or at home, provided field notes are accurate and complete.

A record of the season's observations should be kept in the form of a bird calendar, on which data may be recorded from the field notebook. The calendar may be constructed on a large piece of white cardboard. The following data might be included: observation number, date seen, name of

bird, place observed, habitat, and type of resident.

In the spring and fall a migration calendar might be kept, showing the following data: name of bird, date first seen, locality, date of greatest abundance, date last seen, and remarks.

Other worthwhile phases of bird study include the stydying of nests in winter, making and putting up bird houses, maintaining feeding stations during the winter, study of structure and anatomy, study of distribution and nesting, study of foods, study of plumage and song, banding photography, study of ecological associations, and the keeping of a local list.

The student who becomes skilled in observing and identifying birds in the field usually finds that his interest does not cease with the completion of his course work, but instead may continue throughout life.

Survey of Stream Pollution

A survey of this type should be made along a local river or stream, preferably one of some economic importance to the community. A township map will show the course of the stream through the region.

Necessary materials are: collecting jars; funnel; ring stand; filter-paper; scales; and a township map of the region.

The study should be started at a point upstream from the community in question. The stream should be followed for a mile or more, if possible, making a careful check on the condition of the water, the presence of fish, crayfish and other aquatic animal life, and the condition of aquatic plants in the stream. Evidence of pollution in the form of dark coloration of the water, odor and dead aquatic animals, should be watched for. If possible, the sources of pollution such as entrances of sewers and drains from factories or refineries should be determined.

Quart samples of water should be collected at certain points along the stream. This may be analyzed in the laboratory as follows: Set a funnel on a ring stand and line it with filter paper which has been weighed previously. Filter the quart sample. Mud, silt, and other residue will collect in the filter paper. Dry the paper overnight. Use care not to lose any of the sediment. Weigh the filter paper and the sediment the next day. By substracting the original weight of the paper, the exact weight of the sediment in one quart of water may be determined. Calculate the sediment per gallon.

Using a map of the region indicate the following: conditions of the water along the course of the stream, sources of pollution, results of samples taken at intervals, and any other factors which may influence the life in the stream. The scope of this project may be increased by sampling several streams, rivers or lakes in the locality.

Tree Survey on City Streets

This project concerns the tree plantings along city streets and in yards close to the street. It makes a nice survey for students living in towns or cities who have no way to get to the country. The only equipment needed is a plant press and a yard stick.

Select a square block in the community where the survey will be conducted. An area with a considerable number of trees should be selected. A diagram of the block, showing the streets bordering it, alleys, and other significant features, should be made previous to the survey. In conducting the survey, each tree growing between the sidewalk and the street, or in a yard with the branches overhanging the sidewalk, should be numbered. After numbering each tree, its location should be shown on the diagram. Collect

a leaf from each tree and place it in a plant press for pressing. Using a yardstick, measure the diameter of each tree four and one-half feet above the ground. Record this figure by the number of the tree in a notebook. If the name of the tree is known, it should be recorded with the diameter. Judge the general condition of the tree as vigorous, damaged, diseased, attacked by insects, or dead. Record this in the notebook also.

Group IV

Experiments with Living Things

Of all the types of biology projects, those belonging to this group are the most challenging. Each year, biology students doing projects of this nature discover things that were hitherto unknown. This is a type of project where the student's initiative, knowledge and imagination are the main limiting factors. Nine examples of projects dealing with experiments with living things are given here.

Vitamin Deficiency Diets

Feeding experiments involving diets sufficient and deficient in specific vitamins are among the most striking and interesting investigations in the biology laboratory. Directions must be followed closely and observations must be very accurate. Usually, the symptoms of vitamin deficiency appear gradually, but are very pronounced by the end of four weeks of special feeding.

Materials needed include: young white rats (less than four weeks old and litter mates if possible); two animal cages for each vitamin to be investigated; commercially prepared vitamin-sufficient and deficient diets (in the case of vitamin A the diet may be "home-made"); food and

water dishes; and scales that weigh in grams.

Two rats, or better still two groups of rats, are involved in the experiment. One serves as the control, and is fed a normal diet containing the vitamin in question. The other is fed an identical diet except for a complete lack of the vitamin. Thus the vitamin is the only variable in the experiment. If a group of rats are used in the experiment, they should be of the same age and size. Individual members, both of the control and the experimental group, should be marked because weight records of each specimen will be kept throughout the experiment.

Best results are obtained by using commercially prepared vitamin-sufficient and deficient diets. These diets, prepared for vitamins A, B, C, D, and G, are available at biological supply houses, and it is usually cheaper in the long run to buy them prepared. About one and one-half pounds of food will be required for each pair of rats, fed for four weeks. In certain cases, the student can prepare a diet suitable for a vitamin experiment. Such a diet is outlined below in connection with vitamin A sufficiency and deficiency.

Place one or more rats in a cage and mark it "control." (Due to cannibalistic tendencies, it is best to have only one rat per cage if at all possible.) Place another rat or group of rats of the same age and size as the control group in a cage marked "experimental." Provide both groups with water at all times. If possible, obtain a vitamin A sufficient and deficient diet already prepared. Otherwise, prepare a vitamin A sufficient diet of cooked lean meat, bread, cream, and carrots. Use cooked meat and bread as the vitamin A deficient diet. Feed the diet containing sufficient vitamin A to the control animals and the deficient diet to the experimental animals.

Continue the experiment for at least four weeks, or until unmistakable symptoms of vitamin A deficiency appear in the experimental animals. During this period, weigh the animals regularly (Monday, Wednesday, and Friday) and watch for symptoms of deficiency disease, especially in the eyes of the experimental group. Record weight changes, condition of general health, and condition of the eyes on a chart prepared for each animal used in the experiment. Record the data in columns marked as follows: date, weight, general health, and condition of eyes. At the end of the experiment, place the experimental animals, showing vitamin A deficiency disease, on a diet containing the lacking vitamin. Continue the records, making note of improvement and the length of time necessary to restore the animals to health.

Using other white rats, repeat the experiment, using vitamin D, B, C, or G sufficient and deficient diets.

During the vitamin D experiment, both the control and experimental cages must be removed from the sun, as sunlight stimulates the body to produce the vitamin. The experimental animals, receiving the vitamin D deficient diet should show symptoms of rickets and malnutrition within four weeks. These symptoms include lack of growth, weak bones, general weakness and wobbliness, and bent limbs. Again, make regular weighings and examinations of the subjects and keep complete records of observations. At the close of the experiment, place the experimental animals on a sufficiency diet and make close observations of changes which occur.

Artificial Induction of Breeding in Frogs

A project of this type is well suited to those students who are interested in scientific research methods. This process of obtaining

frog eggs for use in the laboratory during the winter months is relatively simple and excellent results can be obtained if a few precautions are taken. Mature frogs obtained either at the end of the summer feeding season or from hibernation must be used because the ovaries are ripe at this time. In the normal life history of the frog, the increase of pituitary secretion in the spring months stimulates the release of eggs into the uterus. Artificial increase of the pituitary level is produced in the laboratory by the injection of whole frog pituitaries. This causes normal ovulation and the eggs can be stripped from the uterus with mechanical pressure.

Materials needed include: heavy scissors; fine scissors; regular forceps; fine forceps; dissecting needles; steel section lifter; 2 cc. syringe; No. 18 needle; isolation jar for frog ($\frac{1}{2}$ pint wide mouth canning jar); culture dishes (at least twenty); medicine droppers; five female frogs; and one male frog.

Certain precautions must be undertaken. All glassware and instruments coming into contact with the eggs should be biologically clean. It is suggested that glassware be washed in hot soapy water and then be thoroughly rinsed for at least two hours in running water. Chromium plated instruments should not come into contact with living eggs. The water used for culturing should be allowed to come to room temperature before use. Pond water which is known to support frog eggs should be used. Tap water usually contains chlorine which is harmful to the embryos and distilled water does not contain the salts necessary for normal development.

Select the largest of the female frogs and place in a separate container. When the eggs are ready for release from the ovary the lower abdomen will bulge out on either side as the finger is pressed on the

upper abdomen.

The pituitaries are dissected from the remaining females, and placed in a small dish of clean pond water until they are to be injected. They are dissected as follows: The upper jaw and head of the frog are cut off along the angle of a line drawn from the corner of the mouth around the back of the head to the other corner of the mouth, the line passing about one-fourth inch behind the tympanum. This removes the cranium with the brain and part of the medulla intact. Insert a fine pair of scissors into the back of the cranial cavity keeping as far as possible to the right side so as to avoid damage to the brain, then cut through the bone. Withdraw the scissors and then cut along the left side in the same manner. This should release the floor of the cranial cavity as a small flap. Pull up the flap with a pair of forceps to expose the pituitary. It will be found either adhering to the flap or lying on the ventral surface of the brain. It is a small kidney-shaped organ which is usually imbedded in very white lymphatic tissue which should be removed if possible. Do not injure the gland since it will lose much of its effectiveness if broken.

When all of the pituitaries are removed draw them up into the syringe with a small amount of water. Place a No. 18 needle on the syringe. Inject the pituitaries intraperitoneally. It is best to inject through the wall of the lower abdomen directing the needle anteriorly. Avoid damage to the ventral abdominal and lateral veins and the internal organs. Return the injected female to the container and leave for forty-eight hours.

At the end of forty-eight hours, pith the male frog and dissect out the testes. These will be fairly large oval yellowish bodies on the dorsal surface of the peritoneal cavity at the posterior end of the kidneys. The testes should be torn up thoroughly in about 10 cc. of pond water. The

sperm will become active in about five minutes after they are released.

The female frog is then held firmly in the palm of the hand while the legs are held in the other hand. A firm pressure with a milking motion is applied to the abdomen with the thumb to force the eggs from the uterus. With a firm steady milking motion, the eggs will soon appear. They should be milked into a clean culture dish into which the sperm suspension has been placed. It is usually necessary to pipette the sperm suspension over the eggs so that they will all be exposed. Allow the inseminated eggs to stand for about five minutes, and then add about 50 cc. of pond water. In twenty minutes change the water, adding just enough to cover the eggs. If the eggs are fertilized, they will rotate so that the dark side is up in about one hour.

As soon as the jelly membrane has enlarged, the eggs may be separated. They should be distributed into culture dishes in lots of about twenty-five to thirty eggs each. This should be done before the first cleavage. If they are too crowded abnormalities will begin to appear at about the third cleavage.

Tadpoles may be maintained after hatching if placed in an aquarium and fed boiled lettuce daily. Be sure to remove uneaten lettuce one or two hours after feeding, or the water will become fouled.

Reducing the Growth of Bacteria

This type of project is especially good for those students interested in medicine or public health work.

Materials needed are: three 250 cc. Erlenmeyer flasks; bouillon cube; absorbent cotton; pressure cooker; sixteen petri dishes; pipette; dehydrated nutrient agar; source of heat; inoculating needle; samples of

twelve common antiseptics and disinfectants; paper punchings; cultures of Bacillus subtilis, Penicillium notatum, and Streptomyces griseus; thermometer; forceps; gummed labels; and a glass marking pencil.

To determine the effects of antiseptics and disinfectants upon the growth of bacteria the following procedure should be followed: Sterilize sixteen Petri dishes in a hot air overn at 350° F. for two hours, or in a pressure cooker at fifteen pounds for twenty minutes. Add a bouillon cube to 250 cc. water in an Erlenmeyer flask. Heat the water and shake at intervals until the cube is completely dissolved. Plug the flask with cotton and boil the broth for ten minutes to destroy most of the bacteria present. Using a flamed inoculating needle, transfer some Bacillus subtilis organisms from a stock culture to the broth. Incubate or set in a warm place for 24 to 48 hours.

Prepare 500 cc. nutrient agar by adding distilled water to dehydrated media. Divide the cooked media between two 250 cc. Erlenmeyer flasks. Plug the media flasks and sterilize at fifteen pounds for twenty minutes in a pressure cooker. After sterilization, place the flasks in a pan of hot water containing a thermometer. Allow the water and media to cool to 45° C. (113° F.). Using a sterilized pipette or dropper, place about one cc. of broth containing the bacteria (after incubation) in the center of a sterilized Petri dish. Pour agar (cooled to 45° C.) into the dish until it has reached a depth of about one-quarter of an inch. Shake the warm media slowly back and forth to mix the broth and bacteria with the agar. In shaking, do not raise the dish from the table and do not splash media against the cover. Repeat the process for sixteen dishes. Work fast or the media will gel. If this happens, it will require re-boiling and re-cooling.

After pouring and shaking the inoculated plates, allow them to gel and cool before proceeding. Reserve four dishes for the second part of the experiment.

Place the antiseptics and disinfectants to be tested in a row. Label a Petri dish with the name of each antiseptic. Secure some paper punchings or small squares of paper (about one-quarter inch). Using clean forceps, dip a square of paper into a bottle of antiseptic. Allow any excess fluid to drip off, then place the punching on the surface of the upper region of a poured plate. Repeat, using three more punchings and arranging them in the relative positions of 12, 3, 6 and 9 on the face of a clock, about one-half inch from the edge of the dish. Prepare similar dishes for the remaining antiseptics. Incubate the dishes for forty-eight hours.

The effects of the antiseptics and disinfectants used will be shown by a clear zone around each of the punchings or squares. This zone of inhibition marks the area in which bacteria have not grown. The rest of the plate should show evidence of growth. You should find considerable variation in the width of the zones of inhibition. Measure the zones produced by the various antiseptics and disinfectants and record the results in a table.

To show the effects of penicillin and streptomycin on bacterial growth the following procedures should be followed: Incubate the four inoculated and poured dishes for forty-eight hours and observe the bacterial growth at the end of this period. Using a sterilized inoculating needle, transfer some mold fragments and spores from a culture of Penicillium notatum to the edge of the dish. Inoculate a second dish in a like manner, and two dishes with Streptomyces griseus. Allow the molds

to grow over the surface of the media at room temperature. Observations should be made daily. A record of any visible effects of the mold upon the bacteria should be kept.

Effects of Light on Growing Plants

This makes a very interesting project concerning the physiology of plants. Students interested in gardening or farming should find this project to their liking.

For this project the following materials are needed: two mature plants, such as geraniums or sunflowers; three young seedlings, such as tomatoes or sunflowers; three large paper cartons; two squares of glass eight by ten inches (one square red, the other blue), or sheets of red and blue cellophane and squares of clear glass will do; a sharp knife; adhesive tape; alcohol; and iodine.

To find the effect of total darkness on the growth of a plant, place one of the mature plants in good sunlight and the other plant under a paper carton or in any other totally dark place so that all light is excluded. Water the plants regularly and allow them to grow for a week. Compare the two plants at the end of this time as to color of leaves, presence of starch in leaves, and other differences. Two questions which might present themselves at this point are: What happened to the chlorophyll in the plant placed in darkness, and what is the relationship of light to the development of chlorophyll?

To find the effect of filtered light on the growth of plants, place one of the seedling plants in strong sunlight. The other two seedling plants should be placed in two paper cartons. Cut a window in the side of each carton with a sharp knife so that it is about seven inches wide

and nine inches high. Fasten the colored glasses to each of these windows so that one carton admits the red rays of the sun and the other admits the blue rays. Use strips of adhesive tape to hold the filters securely in place and to exclude all other light. Water the plants regularly and allow them to grow for a week. Then examine the plants to see how they differ as to size, condition of leaves, condition of stem, amount of chlorophyll present, and presence of starch.

Growing Plants with Chemicals

The original idea of growing plants in nutrient solutions dates back many decades to those investigations which gave us our fundamental knowledge as to the food requirements of plants. Contemporary scientists have revived this interesting study, have applied it to the growth of plants of economic importance and are now carrying on extensive research in this field. Many high school biology students find it equally interesting.

The following materials will be required: two young plants (tomatoes, sunflowers, or other rapidly growing plants about six inches high); two eight inch flower pots; melted paraffin and brush; washed gravel the size of marbles; two ring stands with rings; two funnels; two rubber stoppers with holes; two pinch clamps; rubber and glass tubing; two gallon jars; flat board twelve by twenty-four inches with a large hole drilled at each end; three bricks; two gummed labels; distilled water; commercial mixture of chemicals for growing plants, or 2.8 g. potassium nitrate, 1.4 g. monocalcium phosphate, 2.2 g. magnesium sulfate, and 9.4 g. iron sulfate dissolved in one-half gallon of distilled water. An even more complete mixture of nutrients are as follows: 10 ml. 1M calcium nitrate; 10 ml.

1M potassium nitrate; 4 ml. 1M magnesium sulfate; 2ml. 1M potassium acid phosphate; 2 ml. FeEDTA; and 2 ml. micronutrients (to one liter of distilled water add 2.86 g. boric acid, 1.81 g. manganese chloride, 0.11 g. zinc chloride, 0.05 g. copper chloride, and 0.025 g. sodium molybdate). All the above nutrients are then added to one-half gallon distilled water.

Wash the flower pots thoroughly. If they are not glazed on the outside, coat the outer surfaces with melted paraffin to seal the pores. Insert a rubber stopper with a glass tube through the hole in the bottom of each pot, and attach a rubber tube to the outside. Elevate the board so that when the flower pots are placed over the holes in it the rubber tubes can extend from under the flower pots to a level even with the top of each pot. Fill each pot with washed gravel. Gently remove the two young plants from the growing bed and wash the soil from the roots. Place the plants firmly in the gravel in each pot. Label one pot "water" and the other "nutrients." Insert a funnel into each of the rubber tubes, and support the funnel on a ring stand so that the top of the funnel is even with the top of the pot.

Into the first funnel pour distilled water until the level of water is an inch below the rim of the flower pot. In the second funnel pour the chemical solution to the same level. Leave the liquid in the pot for fifteen minutes. Remove the funnels and drain the fluids back into their respective jars. Place a pinch clamp on each rubber tube to prevent seepage. Repeat this "feeding" three times daily. If the room is unusually dry or the plants show signs of wilting, increase the number of "feedings" to five per day. Keep the plants in the sunlight and make up a new solution of chemicals each week. Observe the growth of the plants and record the results, showing development of roots, stem, leaves, and flowers of

each plant at one week intervals for a period of four weeks. This project might be carried much farther by finding what effects come about from leaving out certain of the nutrient materials.

Responses of Roots

An interesting part of the study of plant tropisms is the study of responses of roots to their environment.

Materials needed are: a small wood box; sawdust; funnel; two glass plates three by four inches; blotter; adhesive tape; sponge; beans and radish or wheat seeds; ruler; fine pen; and India ink.

To observe responses of a root to water, plant several soaked bean seeds in one end of a box filled with moist sawdust. In the other end of the box place a funnel in the sawdust so that only a small area of the box will receive water. After the beans have developed their primary root systems, water daily through the funnel so that the end of the box away from the beans is kept continually moist. Allow these seedlings to grow for a week. Then, pull up the plants and examine their root systems to find out in what direction the main growth of the roots has taken place.

To observe responses of a root to gravity, cut a blotter so that there are several thicknesses to fit between the two plates of glass. Place the pieces of blotter on one glass and moisten them thoroughly. Distribute a row of radish or wheat seeds across the middle of the blotter about a quarter of an inch apart. Place the other glass over the seeds and fasten the two glasses together with adhesive tape all around the edges. Stand the mount on an edge and allow the seeds to germinate for several days. After about an inch of root has developed, turn the plates so that they are standing on the opposite edge. Allow the seeds to

continue their growth for another three or four days. Observe the growth of the root systems under these conditions.

To compare the relative effects of hydrotropism and geotropism, sprinkle some radish or wheat seeds on one side of a moist sponge. Hang the sponge so that all the seeds are on the underside. Keep the sponge moist and allow the seeds to germinate several days. Examine the seedlings and observe the direction of the growth of the roots to see if one tropism seems to have a greater effect than the other.

Effects of Insecticides on Insects

This project has many possibilities for expansion, as well as being quite practical in nature.

Materials needed include: twenty adult grasshoppers; eighteen adult houseflies; arsentate of lead; arsenate of calcium; Paris gree; water solution of chlordane; fly spray containing DDT; fly spray without DDT; four quart jars with perforated lids; two pint jars with tight lids; small twigs with green leaves; gummed labels; and absorbent cotton.

To make a comparison of stomach poisons, obtain twenty grasshoppers and put them in a clean jar or insect cage for a few days without food. Dust three twigs with each of the powdered insecticides and spray the fourth twig with chlordane solution. Suspend the twigs each in a different quart jar so that the bottom leaf is about an inch above the bottom of the jar. Label each jar POISON and write the name of the insecticide used on the label. Since these insecticides are deadly poisons, wash hands thoroughly after preparing the jars. Place five grasshoppers in each jar and fasten the perforated lid. Observe them carefully and note the time necessary for the effects of the different poisons to be felt.

Record the results in a table which shows the following: time first grasshopper began feeding, time first grasshopper dropped from twig, time last grasshopper dropped from twig, shortest killing time, longest killing time, average killing time, and a rating of the poisons used. Do this for each of the insecticides to be tested (lead arsenate, calcium arsenate, Paris green, and chlordane).

To compare contact poisons, obtain eighteen houseflies and keep them in a jar with food. Soak a piece of cotton in the fly spray containing DDT and place it in a pint jar. Soak a second piece of cotton in the fly spray without DDT and put it in a second pint jar. Label each of the jars with the spray used. Put three flies in each jar and fasten the lid securely. Note the time necessary for the flies to be killed by the action of the insecticide. Remove the dead flies and leave the jars uncovered twenty-four hours. Put three more flies in each jar at the end of that time and fasten the lids securely. Note the time necessary to kill the flies again. Remove the dead flies and once more leave the jars for twenty-four hours uncovered. Place three more flies in each jar and note the time necessary for the effects of the insecticide to be felt. Record the results in a table showing: time necessary to kill flies immediately after using spray, time necessary to kill flies twenty-four hours after using spray, time necessary to kill flies forty-eight hours after using spray, and the effectiveness of the sprays.

Responses in the Earthworm

Among the most interesting experiments with living organisms are those dealing with responses to environment. The earthworm makes an ideal subject to study in this respect.

Materials needed for this project are: one or more earthworms; newspaper or paper toweling; dissecting needle; flashlight; wires; 1½-volt dry cell; absorbent cotton; toothpicks; vinegar; ammonia; strong salt solution; and sugar water.

To test for response to touch, touch the earthworm at various parts of the body with the point of the needle. Observe the responses of the worm.

To test for response to light, keep the earthworm in the dark for one or more hours. Have a flashlight ready. Uncover the earthworm and direct a beam of light at the front end of the worm. Observe the movements of the worm.

To test for response to electricity, attach wires to the terminals of a 1½-volt dry cell battery. Touch the positive wire to the front end of the worm and the negative wire to the rear end of the worm. The worm should respond by becoming shorter. Now reverse the wires, touching the positive wire to the rear end of the worm and the negative wire to the front end. The worm should now respond by becoming longer.

To test for response to toxic substances, place some absorbent cotton on one end of several toothpicks. Dip the cotton on one toothpick into some vinegar and bring it to within one inch of the head of the worm. Observe any reactions. Repeat with other toothpicks, using household ammonia, strong salt solution, and sugar water.

Crossing Fruit Flies to Study Heredity

A project of this type should be of great interest to those students interested in heredity or in the applied aspects of plant or animal breeding.

Materials include: a number of ½-pint glass milk bottles; cotton plugs for the jars; sterilizing equipment; hand magnifier or binocular microscope; ether; yeast cake; and whole bananas which are not overripe and which have not had the skin broken.

The main problem in growing successful cultures is that molds frequently appear which destroy the flies. Therefore, it is necessary to wash the nands and sterilize all jars, cotton plugs, and utensils used in preparing a culture. Wash the jars thoroughly in soap and water and allow to dry. Roll a cotton plug for each jar so that it fits tightly. Plug the jars and heat in a sterilizer or in a moderate oven for half an hour or longer. Sterilize the other instruments to be used at the same time. Do not remove the cotton plugs until the jars are ready to be used. Select a ripe banana that has a whole, firm skin. Wash the outside with cold water and peel carefully, being careful to avoid touching the inside with the fingers. Cut about two inches of the peeled banana into small pieces, dropping these directly into each sterilized jar. Crush the banana with a sterilized rod and add two or three drops of yeast culture. (Mix a small amount of yeast cake in water for this culture.) Be sure to place the cotton plugs on a clean piece of paper while the food is being prepared so that the plugs will not be contaminated. Replace the plugs in each jar, and the jars are now ready to rear the flies.

Flies introduced into these culture jars will lay eggs on the banana, and in four or five days small larvae will be seen. In about eight days the pupae will appear as small, yellow-brown cases, about the size of small seeds. The new generation of adult flies will begin to emerge from the pupa cases in about a week. The whole life cycle is completed in twelve to sixteen days. The cultures can be kept alive by adding small

amounts of banana every week or so. Rearing bottles must be kept at temperatures of about 75° F., and should not be exposed to bright light. Cultures will grow all right during the winter months.

A number of wild flies may be obtained by leaving an open culture jar near a fruit stand. The flies will be attracted by the mashed banana. When a number are in the jar, replace the plug. Pure-strain cultures, showing various mutations, may be obtained from biological supply houses. Interesting crosses may be observed, however, by using ordinary wild fruit flies.

The flies may be transferred from one jar to another by first tapping the jar gently on a table to dislodge the flies from near the mouth of the jar, then removing the cotton plugs from each jar, and holding the open ends together. A gentle tap on the bottom of the upper jar, or shining a bright light on that end, will cause the flies to move to the lower jar. When a sufficient number have been transferred, replace the cotton plugs.

In order to observe the flies to study their traits, the flies must be etherized. Use a sterile jar with no food for this purpose. Fit a cork for this jar and attach a small wad of cotton to the under surface of the cork. Place a few drops of ether on the cotton, but do not place the cork in the jar until the required number of flies has been transferred. Ether is highly inflammable so it should be kept away from open flames. As soon as the flies are in the etherizing jar, observe them carefully. The flies will "go under" in about ten seconds. In fifteen seconds the flies should be motionless. Remove the cork and tap the jar to pour the flies on a clean, white card at once. If flies are etherized for a longer period, their wings bend upward, their bodies curl, and they die. It is well to practice this step with a few wild flies so that the

technique will become familiar.

Etherized flies will recover in about five minutes. Hence, any study and sorting must be done quickly. Use only a few flies at a time so that each one can be observed carefully. A small pocket knife can be used to move the flies up and down on the card as each characteristic is seen. Care should be taken not to work under a strong light, as overheating will harm the flies. It is well to be accurate in examining and sorting the flies, as a second etherization will usually render the flies sterile and will make them useless for further study. After the flies are sorted, each group is placed in a fresh culture jar for further experimentation.

The first step in identification is to learn how to separate the male and the female flies. The males may be distinguished by their black-tipped, blunt posterior ends; the female is lighter in color and has a more pointed posterior. Positive identification may be made under a magnifier by examining the first pair of legs for the "sex comb" of dark bristles present on the legs of the males. The major characteristics to learn to identify are the size and shape of the wings, the color and shading of the body, and the color of the eyes. The normal wing in the fruit fly is dominant over other characteristics, such as stubby wings, curved wings, spread wings, and raised wings. The tan-gray body color is dominant over black or yellow. The red-colored eyes are dominant over brown, black, or colorless eyes. It is a good idea to first etherize a few wild fruit flies collected near a fruit stand and to study these with a magnifier to become familiar with the dominant characteristics named above.

To cross the fruit flies, a culture bottle in which a number of new adult flies are emerging should be selected. Remove all the adult flies

and discard them or transfer them to another jar. Make a note on the jar as to when this was done. Examine the jar again in ten to twelve hours to see if new adults have hatched. These flies have not had time to mate and can be used in crossing experiments. Female flies older than twelve hours should not be used as it must be considered that they have already mated, and once mated, a female fly may not be successfully mated with another male.

Transfer these unmated flies into the etherizing jar and etherize them for examination. Sort out the males and females and place two females and two males in each new culture jar. Mark the date, and the type of flies used on each jar. The flies will mate soon after recovering from the ether, and the females should begin producing eggs in thirty-six hours. In order to make sure that the experiment will be successful, it is wise to prepare three culture jars for each cross. After a few days, when a number of larvae are seen in the jars, remove the parent flies and discard them. Be sure to add bits of banana from time to time to keep these culture jars alive.

About twelve days after the original mating, the F_1 hybrid generation of flies should begin to emerge. Usually the females begin to hatch out first and the males appear soon after. All of these flies should be hybrids, showing the wild strain characteristics. Now place about six males and six females of the F_1 generation in each of three fresh culture jars. Mark the date again and the type of the original cross. Each of these jars should develop into about 250 flies of the F_2 generation. The adult flies should be removed from the three jars in about a week. Keep the cultures alive by adding bits of banana from time to time. The F_2 jars should be examined every other day for about a week. As each batch of

flies emerges, transfer them to an etherizing jar, then place them on a card, and examine with a magnifier for variations in the wing structure, body shading, and eye color. Keep track of the number of each characteristic seen in the flies. After examination, the F_2 generation flies may be discarded. Do not return them to the original jar as this will throw the count off. Compute the ratio for each variation and see if it agrees with the ratios given in text books.

Any number of further crosses may be attempted by the student. New mutations may come about by treating with X rays, so this opens up another avenue of study for the student. The main limiting factor is time.

Group V

Collecting and Caring for Living Things

Not only does this type of project prove to be quite interesting to most students, it also may be of great help to the teacher insofar as providing a source of living material to be used throughout the year as instructional material. For instance, rearing insects in the school laboratory makes a good project as well as enabling the entire class to see all the stages of the life cycles living under approximately natural conditions. The maintaining of aquaria and terraria in the laboratory is of utmost importance, and this might also become project material. Eight examples of projects dealing with collecting and caring for living things are given here.

Growing Fresh-Water Algae in the Laboratory

Fresh-water algae are of very general distribution and are found in nearly every type of damp and aquatic habitat. Ditches, ponds, rivers,

lakes and marshes of any locality abound in a great many forms, both of the unicellular and multicellular types. Some algae grow on damp earth or rocks and some kinds make up the greenish covering which appears on the bark of trees. Most of the more common forms are independent, either free floating or attached to the substratum, but there are also epiphytic and endophytic species. Many of the smaller forms are attached to other water plants or are found in the loose sediment and debris of the substratum.

Many kinds of algae are easily cultured in the laboratory. Some kinds will grow well if brought indoors and placed in containers of pond water or in a balanced aquarium; other field-collected algae are more difficult to culture and can be maintained over long periods only by the use of nutrient solutions, and the pure-culture technique. For most kinds of algae, large finger bowls or battery jars are good culture containers; some robust forms such as Spirogyra and Cladophora are best grown in aquarium tanks.

When possible, use the water in which the algae were growing, since sudden changes in kinds of water are injurious. Where additional water is needed use distilled water, or let the tap water run for several minutes before filling the jar, since water standing in pipes, or other metal containers, is harmful to most algae. Add new water occasionally, to compensate for evaporation.

Do not put too much material into a jar. Actively growing material will increase and gradually accommodate itself to conditions. The excess in an overabundant supply will be choked off and the consequent decay will cause fermentation and general fouling of the culture, utimately killing the whole. Likewise, luxuriant forms such as Cladophora may soon

overstock themselves and the excess material must be removed from time to time.

Cultures may be started at any time of the year. In winter, bring in some mud over which the desired form was growing the previous season. Sticks or stones may also be used. Place it in a jar and add some tap water, distilled water, or rain water as the case may require. Cultures of some forms, such as Chara and Nitella, have been obtained from mud collected several years before. Cultures started in this manner usually yield a variety of material.

Do not throw out a healthy culture if the alga should disappear. Seasons of dormancy occur in nature—look in the bottom of the jar for spores. Let the water evaporate, cover the jar for protection, and set it aside. After a period of a month or two add more water and very likely there will be alga present again. Lengths of dormancy periods vary. In Volvox it is quite long; in Oscillatoria it is only a couple of months; in Cladophora there may be no dormancy. Warm weather makes it difficult to keep some cultures of algae in good condition. Refrigeration, after collection, will make it possible to study the material for a week or more instead of just a day or two.

The method used in collecting algae is different from that used for aquatic zoological specimens. Care must be taken to avoid metal containers, especially on long, warm trips. Ample water must be used if the containers are to be sealed. However, forms like Spirogyra, Zygnema, Cladophora, and Vaucheria may be rolled in wet newspapers or magazines and carried thus.

Oscillatoria is readily found in stagnant water, watering-troughs, damp earth, flower pots, and in many other habitats. It is recognized

by its dark blue-green, or blackish color. Oscillatoria is one of the easiest of plants to keep in the laboratory. Place a little of the material in a container partly filled with water; cover with a lid to avoid unnecessary contamination from dust. By adding water from time to time to compensate for evaporation, the cultures should keep indefinitely.

Nostoc is frequently found in lakes and also occurs in damp earth. It is easy to maintain in laboratory cultures, and will keep in good condition for a month or more if kept under refrigeration at a temperature of about 40° F.

Spirogyra is found in the field in the quiet waters of ponds, ditches and lagoons, where it often forms large green mats covering the surface of the water. Occasionally it occurs in running water, while still less commonly it is found attached to rocks and piles. Spirogyra is not easily cultured for prolonged periods, but it will sometimes grow well in a balanced aquarium, and some species live and increase nicely in nutrient solutions. Solutions made of distilled water containing minute quantities of dissolved commercial fertilizers (such as "Vigoro") often work well. Spirogyra cultures should receive plenty of daylight and at least some direct sunlight. The cultures should be started with distilled or natural pond water, as city water treated with chlorine or other chemicals is very destructive to Spirogyra.

Cladophora is found growing attached to sticks and stones in quiet or running water. For cultures, select the forms found in quiet water, and place in one or two-gallon aquaria. In larger aquaria Cladophora is likely to grow too luxuriantly; even with the smaller containers, care must be taken to remove the excess material from time to time. This is one of the easiest algae to culture. It can be maintained with so little

trouble that pure cultures in nutrient solutions are not usually worth bothering with.

Pleurococcus makes good material for study and can be secured by collecting pieces of tree bark on which it is growing. Such material may be stored dry; if placed in a moist chamber for twenty-four hours, the Pleurococcus will begin active growth and is then in good condition to study. It is also possible to grow Pleurococcus on nutrient agar slants.

Chara and Nitella are both fairly easy to grow in large containers. Prepare a good-sized battery jar or an aquarium tank with a couple inches of sand and pond mud in the bottom and fill with natural pond water. Plant a small amount of freshly-collected material in this and place the container where it will get plenty of light and some direct sunlight.

Mud taken from the bottom of ponds in which Chara and Nitella are known to grow (even when the ponds have completely dried up) will often produce excellent new growths.

Diatoms are usually found in large quantities around springs and in pools and ponds, clinging in great numbers to filamentous algae, or forming gelatinous masses on various submerged plants. The surface mud of a pond, ditch or lagoon will always yield some forms. Fresh-water diatoms appear in greatest abundance in the spring, are comparatively scarce in summer, but reappear again in the autumn.

Diatoms show their characteristic movements best when transferred from cooler to warmer water. This phase of motility is therefore well illustrated shortly after being brought into the warm laboratory.

The following formula is well suited for the growth of many of the fresh-water algae: ammonium nitrate (NH_4NO_3), 0.5 g.; potassium phosphate

(KH_2PO_4), 0.2 g.; magnesium sulphate ($MgSO_4.7H_2O$), 0.2 g.; calcium chloride ($CaCl_2.2H_2O$), 0.1 g.; distilled water, 1000 cc.; and 1% ferric chloride solution, 0.1 cc.

Collecting Bryophytes

A collection of mosses and liverworts is interesting to maintain in the laboratory. Field trips to deep woods, shaded hillsides, and ravines should yield an interesting assortment of these plants. Look on decaying stumps and logs, among the leaf litter of the forest floor, along the banks of streams, and in other places where conditions are moist and where the light is reduced. Collect clumps of different species, especially those which are bearing stalks and capsules on the leafy shoots. Plant them in the laboratory terrarium, where they should thrive for many months.

If possible, move mosses and liverworts to the collection with them still on the rock, soil or bark to which they were attached.

The planting base for mosses should consist of a mixture of two parts humus, one part peat moss, one part loam and one part mixture of pebbles and sand for drainage. Mix together thoroughly, add enough water to moisten, and fill the terrarium to one-fourth of its height. Place the moss on the soil, press down firmly, sprinkle a little water on top, cover the case with a glass top (not too tightly), and place in a cool airy location. A north window is the best all year-round location. Water from time to time, but not excessively, because mold may develop.

The soil mixture for the liverworts should consist of one part cinders, one part sand, one part loam and one part peat moss--all thoroughly moistened. Place the liverworts on top of this mixture and cover the terrarium with glass. Keep in a cool place out of direct sunlight. Add

water from time to time.

Germinating Fern Prothalli

This project might be carried on in conjunction with the preceeding one. Materials needed are: a flower pot saucer; bell jar; small amount of humus; and a fern frond with mature sori.

Place a thin layer of humus ($\frac{1}{4}$ to $\frac{1}{2}$ inch thick) in a four or five-inch flowerpot saucer. Dust some fern spores from mature sori (on the lower side of a frond) into the soil. It may be necessary to brush the spores out with the tip of a dissecting needle. After the spores are sown, cover the saucer with a bell jar. Keep the soil moist while the spores are germinating and the prothalli are growing, but do not allow water to stand on the soil. Watch for the appearance of slender, green filaments which broaden at the tips to form the heart-shaped prothalli of the fern. This may require several weeks. When the prothalli have developed, they may be mounted on a microscope slide and examined under low power for rhizoids, archegonia, and antheridia.

Spores may also be sown on agar or liquid media with very good results. The culture medium surface should be evenly covered with spores rather than spotted with large masses. Cultures should be kept at room temperature, and should be kept in a window or other suitable lighted area which will provide eight to ten hours of combined direct and indirect sunlight.

Culturing Protozoa

Any student interested in microscopic life will certainly be interested in this project.

Materials needed are: stacking dishes; battery jars; dropping pipettes; wheat and rice kernels; timothy hay; and distilled water.

Directions will be given for the culturing of amebae, paramecia, and euglenae. The same general techniques apply to culturing most other protozoans.

Culture dishes are best for amebae because of the large amount of surface for the depth of the water. Fill the dish with distilled water and add three to five wheat kernels and ten pieces of timothy hay, about one inch long, which have been boiled in water five minutes. Cover the culture and set it aside for about two weeks. If stacking dishes are used, several cultures can be prepared and one set on the other , with a cover on the top dish. During the two-week period, bacteria will multiply and provide a source of food for the amebae. If a culture of amebae is available (one used for laboratory study), transfer a few animals to each of the new cultures with a clean pipette. If it is necessary to collect amebae from a pond, bring in several bottles of pond water containing decaying water plants. Transfer this to culture dishes and examine the sediment under the microscope each day after about one week. If amebae are present, they should appear in considerable numbers within two weeks. They may then be transferred to the new cultures with a clean pipette. Keep the cultures at room temperature and in reduced light.

Either stacked culture dishes or battery jars are excellent for culturing paramecia. If a culture dish is used, fill it with distilled water, then add six kernels of wheat or rice and fifteen pieces of timothy hay which have been boiled five minutes. Cover the dish (several may be stacked) and set it aside for five days to one week. It is then time to add the paramecia. These may be taken from a stock culture, or

taken from a pond culture. If freshly collected paramecia are used, they should be transferred with a clean pipette, using care not to introduce other protozoans which might contaminate the culture. A micropipette is best, since a single organism can be picked up. A micropipette may be prepared by heating a piece of glass tubing in a burner and pulling out the tube from both ends while the glass is hot. After the tube cools, break the narrow, pulled section to produce a small-bore, tapering pipette of the desired length. Fasten a pipette bulb on the large end. Paramecia should multiply rapidly in the culture for about two weeks. It is then necessary to add more food, or better still, to transfer animals to a new culture. Maintain paramecia at room temperature or in a warm place (80 to 85° F.) and in a north window where they do not receive direct sunlight.

To culture euglenae, fill a six-inch battery jar with distilled water and add about one hundred kernels of wheat or rice, which have been boiled five minutes. Set the culture in a window for about one week, then add euglenae from a stock culture. Or they may be collected from a pond and transferred to the culture. In collecting euglenae, look for small pools of water which are greenish in color. Examine the water with a microscope and, if euglenae are present (other organisms give water this color also), transfer them to the culture with a clean pipette or micro-pipette. The culture should last at least one month. Add food every two weeks as long as the culture is maintained. When the euglenae begin to die out, transfer some to a new culture.

A Demonstration Ant Colony

Many kinds of living insects can be studied in the laboratory, but

none is more interesting than a healthy colony of ants. Ants are cosmopolitan in nature, are obtainable at nearly all seasons, thrive for long periods of time and require relatively little care.

Many kinds of artificial containers for ant colonies are possible. The chief requirement is that the nest be so constructed that the ants can be observed at will. By referring to the literature, one will find a number of patterns for observation ant nests. The most practical type is one about twelve inches long, eight inches wide, and one inch deep. The object of the flat type nest is to obviate the use of dirt which merely interferes with close study. The nest might be divided into two or three rooms, with interconnections. The top should be covered with glass plates which have been cut to size. If a piece of red glass is used, the ants will behave as though they were in total darkness.

Most of the mound-building ants are well suited to classroom use. They are large in size and among the most intelligent of the ant family. The collector should be equipped with the following items: a garden trowel; a muslin cloth about forty inches square; a pair of tweezers; and some string.

In opening a mound, first use spade or trowel, digging around in a circle of about fourteen inches diameter on the top. After prying it up somewhat with the spade, the hardened top disc can be lifted back like a manhole cover, and if sufficiently intact will offer a nice study in nest construction. In digging down further into the nest, try to follow the structure of the galleries since most of the ants will be found massed in the enlarged chambers. Avoid taking too many workers. After placing the trowels full of earth containing workers on the center of the cloth, catch up the corners and tie it tightly into a bag. In digging,

look carefully for the wingless (fertilized) queen. In most species this individual is larger and usually shinier than the workers. Try also to get a quantity of eggs, larvae, and pupae, and possibly also the nest parasites. These can be carried in a small bottle, and introduced to the artificial nest after the workers are well established.

After finishing the excavating and making observations and notes, replace as much of the earth as possible and then replace the top disc, trying to help this much in reestablishing the nest.

Ants seem to do better when allowed to enter a laboratory nest by themselves. As for care and handling, certain general suggestions are worth making, but the special details vary greatly with the species used, the type of work and the type of individual who is attending the ants. For moisture, in a flat earthless nest, two thin flakes of sponge can be used interchangeably. As one dries out, the ants will spend less time near it, thus permitting its removal, or the partly dried flake of sponge may be resoaked. The ants should be provided with a balanced ration, in no greater quantity than they can dispose of readily. The routine of timing administering of food and water and the regulating of quantity should be carefully worked out according to the number of ants in the nest. A balanced diet which may be used for most of our common species consists of watered honey (two parts water to one of honey) or of sugar solution (the carbohydrate), into which is mixed one part of egg white (protein) and one part of melted butter (fat). To prevent fermentation, this mixture should be prepared in fresh supply each time the ants are fed. This may be supplemented with dead insects; or honey water and dead insects may be used exclusively.

In handling the nest, whether to introduce food or water, or to

clean, or in making observations on nest life, it pays to move deliberately and carefully. The nest must not be jarred, since this excites the ants and greatly interferes with satisfactory observation.

Observations may be made of general nest activities, recognition of nestmates, mixed colony reactions, effects of temperature, special behavior, individual behavior, foraging, parasites, and many other phases of ant life.

Construction of Habitats

This is a very worthwhile project because it affords an opportunity to study and arrange an unlimited variety of plant and animal groups. Native flora and fauna may be represented or interrelationships of such environmental factors as food habits, temperature and moisture upon plant and animal associations may be shown.

Materials needed are: terraria with glass covers, or large glass jars; various kinds of soil; and assorted plants and animals (depending on the type of habitat constructed).

Ordinarily, a large terrarium is easier to plant and maintain than a small one. The terrarium should hold water for it may be desirable to include bog and semi-aquatic plant groups in it. It should have a glass cover so as to maintain the proper humidity, to avoid sudden changes of temperature, and to protect the plants from drafts.

In general, the terrarium should receive plenty of light. The north window is an ideal location for woodland groups consisting of ferns, mosses, newts and tree frogs because direct sunlight is not essential. On the other hand, cacti, alligators, lizards, turtles and most snakes require sun and will not thrive unless they can be kept in a sunny location.

The bog, woodland and semi-aquatic terraria should be kept at a temperature of 65° to 72° F. for this is conducive to the normal growth of both plants and animals. The plants and animals of the desert terrarium will thrive if the temperature is maintained at from 80° to 90° F. It is sometimes necessary to suspend an electric light over the terrarium in order to maintain this temperature.

In the preliminary preparation of the terrarium for planting, the foundation or base material is of utmost importance. Plants require air at their roots as well as around their leaves. Sufficient coarse material should be included in the base to allow for good drainage and aeration. The base material should be slightly moistened and may be arranged in any desired manner. It should cover the entire bottom to one-fourth the height of the container. An attractive terrarium is one in which the soil is arranged unevenly with hills and hollows, high in back and low in front and a few pieces of stone and wood placed here and there to vary the scene.

The simplest group to assemble and one which requires the least amount of attention is the desert terrarium. The tank bottom may be covered over with one and one-half inches of coarse sand and topped with a mixture of equal parts of loam and sand about two inches deep. A few stones and a shallow pan of drinking water for desert animals may be added--the pan should be partially buried in the sand so that its top edge is even with the surface. The lower layer of coarse sand should be moistened slightly when it is placed in the tank; but the top layer should be kept reasonably dry.

Plant small cacti in the sand, being sure to spread the roots. The roots should be moistened before planting and, after all plants are

satisfactorily placed, sprinkle the surface about them. One or more desert animals add considerable life to the desert scene; horned "toads," collared lizards, and small snakes (common desert species), as well as certain Arthropods, will live in the terrarium with a minimum amount of attention. The top of the terrarium should be covered with wire screen rather than the glass plate. Most of the animals may be fed earthworms or mealworms. Water the desert about once a week, but keep a supply of water for the animals at all times.

The woodland terrarium offers an unlimited number of possibilities and may include a variety of combinations representing various kinds of habitats. The foundation layer should be coarse gravel. Over this should be a mixture of one part sand and three parts humus several inches thick. The soil mixture should be moistened sufficiently so that it will cling loosely together without caking. After the soil mixture has been properly arranged, dampen the roots of the plants, and group them to achieve the desired scenic effect. Those plants whose leaves have a tendency to spread should be centered so that their leaves may grow without touching the sides of the tank, or obstructing a part of the group from view. Mosses, liverworts, lichens, club mosses, and wood ferns go well in a habitat of this nature. Overcrowding should be avoided; but, if the space is available, the addition of evergreen plants is worthwhile and attractive. After the planting is completed, the plants should be trimmed, all broken stems and leaves as well as debris removed, and the glass given a final cleaning. The entire group should be sprinkled with a fine spray, the glass cover placed on top, and the terrarium placed in a cool location. Water the habitat every two to three days, keeping the soil moist but not wet.

Animals may be included in the woodland habitat; among those to be considered are the common newts and salamanders, toads, tree frogs, chameleons, small snakes, snails, slugs, beetles, and many others. A stout stemmed plant should be included as one of the plants to offer sufficient support for the climbing animals.

The plants in a bog terrarium require acid conditions and the foundation must supply this need if the plants are to grow and develop normally. A layer of gravel should cover the bottom for drainage and this should be covered with a soil mixture of one part Sphagnum and two parts acid soil. The bog should be thoroughly soaked, with an excess of moisture being allowed to remain in the gravel layer. The roots of the bog plants should be wrapped with Sphagnum; the Venus' flytrap and Pitcher Plant should be planted deep, Sundew is a shallow-rooted plant and should be planted accordingly. The bog terrarium should be covered with a glass top and placed in a cool location. The bog should be watered frequently, so that the layer of Sphagnum is moist but not submerged. A small pool of open water may be constructed near the center by removing dirt and covering the banks with Sphagnum.

Any animals which prefer moist surroundings will do well in a bog terrarium. This group includes newts, toads, salamanders, frogs and turtles.

For the construction of a tropical habitat, place a two-inch layer of rich loam mixed with humus over a layer of coarse gravel. Plant the habitat with small ferns and vines, such as Philadendron. The leafy top cut from a pineapple will often grow roots and give a tropical atmosphere. Small orchids may be included (they may be perched on limbs) if the terrarium is kept warm, especially at night. Animals in this

environment may include chameleons and small snakes. Many other types of habitats are possible, all of them being quite interesting.

Balancing an Aquarium

Balancing and maintaining an aquarium may prove to be a life-long hobby as well as being a worthy project. Many ecological principles may be illustrated with a balanced aquarium. Food chains, carbon dioxide-oxygen cycles, and many other plant-animal-environment relationships are readily seen. Balancing an aquarium is largely a matter of establishing the proper kinds and numbers of plants and animals in the right amount of clean water under suitable light and temperature conditions.

Materials that are necessary include: an aquarium, battery jar or other large glass container; clean sand and garden soil; aquatic plants; and aquatic animals.

The aquarium used is a matter of choice. It may be from five to fifty gallon capacity. Be sure it is clean. It is also a good idea to test for leakage by filling with water and letting it stand overnight.

A layer of garden soil at least one-half inch deep should first be placed in the tank. Cover this with a layer of clean, washed sand to a depth of from one to two inches. The sand may be shallow toward the front so that sediment and waste material will have a tendency to collect in this area and thus be siphoned away.

Add water slowly, pouring onto a piece of clean paper or glass. This will prevent needless stirring of the sand. Use clean pond-water, spring-water, or aerated tap-water. If chlorinated water must be used, it should be previously treated by boiling vigorously, letting stand overnight, and pouring back and forth to introduce air.

Establish the plants first. It is generally best to place rooted plants such as Vallisneria and Sagittaria toward the back corners of the tank in the deeper sand. Place deep enough so that roots are below sand but do not cover leaves as this may kill the plants. Other plants, such as Elodea, Cabomba, Myriophyllum, and Ceratophyllum, may be placed toward the center and front. Although these plants may not have roots when obtained, they will eventually become rooted and so should be planted in the sand. If they tend to float free they should be anchored in the sand by means of pebbles or small lead strips. Lemna (duckweed), Azolla, Salvinia, Riccia, Limnobium, Pistia (water lettuce), and water hyacinths are all floating plants. Chara and Nitella may either float free or be anchored basal end downward. Use all plants sparingly. Too few plants are better than too many. Overstocking may cause all to die.

If the water is not reasonably clear, wait until suspended matter has settled before adding animals. Use snails, mussels, tadpoles, and fish. Aquatic newts and small turtles can be introduced if a floating platform is provided. Avoid animals which are not compatible. Do not use too many animals. One inch of live fish to each gallon of water is a good rule.

Generally, a well-lighted place where sunshine is available at some time during the day is desirable. However, the latitude and the season of the year are important factors. In a northern latitude, a southern exposure during winter months may be desirable, whereas the same exposure during spring and summer may require some shading. When possible, allow light to strike the aquarium from above. Do not permit more than two hours of direct sunlight per day, especially during warm weather or if there is a tendency for the water to become warm. A glass plate should

cover the aquarium to prevent sudden changes of temperature, to prevent evaporation, and to shield against dust and other foreign matter.

To protect the aquarium from sudden changes in temperature, it may be best to place the tank a short distance back from the window. Do not place near a radiator. An ideal temperature is between 40° and 60° F. Do not allow freezing. A large tank of water will have a slower fluctuation of temperature than a small one.

Since a newly established aquarium may lack sufficient food, a good start is to place a small amount of boiled lettuce leaves in the water for snails and tadpoles. Use the lettuce sparingly and do not leave for more than a few hours, since decaying material will easily contaminate the water. Fish may be fed a small amount of tiny worms, Daphnia, or prepared fish food. Fish will do well on very little food. After the aquarium has become established very little added food is needed. If soil is used under the sand, the plants should require very little extra food. If the plants do not develop a healthy green color, the addition of a small amount of fully soluble plant food should be of value. Use sparingly until the results are obvious. Dead material should be removed promptly. A cloudy appearance may be the result of overstocking or an unfavorable plant-animal ratio. An abundance of algae on the glass may be due to too much light and may require shading with a green cloth. Excess algae should be scraped away with a razor blade and siphoned off. A balanced aquarium does not need running water.

Remember that the important factors are light, temperature, oxygen, and nutrients. Frequent observation and a sustained interest in the project help one to profit from each day's experience and to achieve final success.

Development of Frog Eggs

This is a seasonal project, since frog or toad eggs are usually available only in the spring. They are easily found in April, May and June, the exact time depending on the locality, weather, and species. Hunt for them in quiet pools or ponds, floating in masses or clinging to water plants. Transfer the egg mass to a bucket with a dipper or with the cupped hands. Do not pour the eggs or disturb them unnecessarily. They should be brought to the laboratory in the pond water in which they are found.

Allow the eggs to remain in the bucket until the temperature of the water is the same as that of the water in a previously filled aquarium, battery jar, or shallow pan. Then dip the eggs into the new container. A mass of frog eggs the size of the hand requires about five gallons of water for proper aeration. The container should contain several washed aquatic plants to aid in aeration.

The rate of development of the frog egg is determined almost entirely by the temperature of the water. A temperature range of 50° to 60° F. is satisfactory, but a water temperature of 70° to 75° will cause more rapid development especially where the food supply is adequate. Overheating of the water will cause the eggs to die.

Place several eggs in a watch glass and examine them with a hand lens. The eggs should be floating with the dark spot (containing the protoplasm and developing tadpole) up and the white yolk down. If development has started, the dark area will have become oval in shape. Study changes in the eggs during development each day. Young tadpoles should hatch from the eggs in eight to twenty days, depending on the temperature of the temperature of the water. When the tadpoles hatch, add more plants to

the container. Watch the changes that occur for several weeks. It may be desirable to preserve stages in the development of the eggs and of the tadpole. This can be done easily by dropping specimens into a small bottle containing 8% formaldehyde.

At first the tadpoles have no mouths, only sucking discs by which they cling to the algae or sides of the aquarium. They do not require any feeding during their early stage as they rely, to a large extent, upon the stored up nourishment in their abdominal yolk mass.

In order that the tadpoles have food available, the aquarium should be well planted containing aquatic plants, plentiful algal growth and bottom debris from a pond or stream. The tadpoles will eat some artificial fish food, but it is essential that they have access to dead and living algae and an abundance of bottom debris. Keep all fishes and aquatic insect larvae out of the tank because they will prey on the tadpoles. Large grassfrog and bullfrog tadpoles will eat finely chopped lean meat, cornmeal, cooked oatmeal, cooked spinach and powdered egg yolk.

Metamorphosis in the grassfrog, spring peeper and common toad occurs during the first summer. Tadpoles of the green-frog, Rana clamitans, metamorphose the second summer, while the bullfrog remains in the tadpole stage from two to four years.

An interesting experiment with frog tadpoles is to watch the effect feedings of thyroid has on them. Other interesting experiments may be devised by the student.

Group VI

Human Health, Physiology and Anatomy

The average biology class usually has several members who have their

minds set on becoming doctors, nurses, laboratory technicians, or other workers in the field of public health. Projects of this nature are of especial interest to this group of students, as well as being interesting to most students who do not intend to pursue public health as a career. Three examples of projects belonging to this group are given here.

Chemistry of Digestion

Materials necessary for this project are: fifteen test tubes; test-tube racks; glass marking pencil; funnel; filter paper; commercial pepsin; commercial pancreatin; dilute hydrochloric acid; sodium carbonate; litmus paper; crackers; hard-boiled egg white; lard or butter; iodine solution; and Fehling's A and B solutions.

For showing salivary digestion, number six test tubes consecutively with the glass-marking pencil. Place them in a rack. In test tubes number one and two put a small piece of cracker and add enough water to fill the tubes one-third full. Shake the tubes and allow them to stand for a few minutes. Test for the presence of starch with iodine in test tube number one, and for the presence of a simple sugar with Fehling's solution in test tube number two. Record the results in a table. Now rinse the mouth with water and place a small quantity of saliva in test tubes number three and four. Test for the presence of starch in number three and simple sugar in number four. Record the results in the table. Chew a piece of cracker thoroughly, but do not swallow it. When the cracker is chewed into a soft mass, place part of it in test tubes number five and six. Test for starch in number five and for simple sugar in number six. Record the results.

To show gastric digestion, make a solution of artificial gastric

fluid by mixing two grams of commercial pepsin in 0.4% hydrochloric acid solution to make 100 cc. of the fluid. Filter this to obtain a clear fluid. Test the resulting solution with litmus paper. Number six test tubes consecutively and place them in a rack. Put a few pieces of cracker in test tube number one, a small piece of hard-boiled egg white in number two, and a small quantity of lard or butter in number three. Fill each tube one-third full of the artificial gastric fluid. In test tubes number four, five and six, put a piece of each of the same foods, but fill each tube one-third full of plain water.

Allow the six test tubes to stand overnight in a warm place, as near body temperature as possible. Then check the degree of digestion by noting the cloudiness of each solution. Fill in a table to show the results.

To show intestinal digestion, make up a solution of artificial pancreatic fluid by mixing two grams of commercial pancreatin and distilled water to make 100 cc. of the fluid. Add a pinch of sodium carbonate. Stir and filter to get a clear liquid. Test the solution with litmus paper. Number three test tubes and place them in a rack. Put a few pieces of cracker in test tube number one, a small piece of hard-boiled egg white in test tube number two, and a small amount of lard or butter in test tube number three. Fill each tube one-third full with artificial pancreatic fluid.

Allow these tubes to stand in a warm place overnight also. Then check the degree of digestion by noting the extent to which each of the foods has been dissolved. Fill in a table to show the results of digestion. This project may be varied to meet different needs.

Testing Common Remedies

With the increased influence of television commercials on the American public, this project certainly has much merit. An almost endless variety of tests present themselves to the student who is interested enough to perform them. To make a start, the following materials may be used: several brands of aspirin; common stomach (indigestion) remedies; beakers; stopwatch; dilute hydrochloric acid; limewater; laxative remedies; grain alcohol; and ammonia.

Solubility of various brands of aspirin may be tested by placing 50 cc. of water in each of several beakers. Since all aspirin is the same chemical compound, the main difference in the speed with which it acts in the body is the rate at which it dissolves and is absorbed into the blood. Using a stopwatch to time the speed with which each tablet goes into solution, drop each tablet separately into a beaker of water and time the rate of dissolving. Keep a record of these solubility rates. Report to the class on the results of all tests. Since aspirin must be absorbed into the blood to be effective, do not jump to the conclusion that the brand that dissolves fastest is necessarily the best pain reliever.

To test for the presence of sodium bicarbonate in stomach remedies, place a pinch of the powder, or a piece of the tablet, in a test tube and add dilute hydrochloric acid. If the substance fizzes the test indicates the presence of carbon dioxide. Bubbling this gas through limewater will give a positive test if the solution turns milky. Test several of these remedies for the presence of a carbonate. Record the results of these tests in a table, and report to the class.

Many laxatives contain phenolphthalein. A test for this drug may be made by adding 2 cc. of the liquid (or one gram of the powdered solid)

to 15 cc. of grain alcohol in a test tube. Shake vigorously and allow the test tube to stand for five to ten minutes. Pour the clear liquid carefully from the top of the test tube into another test tube. Add a few drops of ammonia carefully to this solution. If the substance turns a reddish-purple color, the presence of phenolphthalein is indicated. Test several of the laxative remedies for this substance.

Distribution of Bacteria

For this project the following materials will be needed: thirty-four Petri dishes containing sterile nutrient agar; incubator; glass-marking pencil; and a pressure cooker.

Prepare the nutrient agar as follows: Measure out 500 cc. of water, two grams of beef extract, two and one-half grams of peptone, seven and one-half grams of agar, and one-eighth gram of sodium chloride, into a large beaker. Boil until completely dissolved, stirring constantly. As soon as the medium is dissolved, pour it into the sterile Petri dishes until each dish is about one-fourth full. Cover the dishes immediately and place them in the pressure cooker. Build up the pressure to twelve pounds and leave for about twenty minutes. Remove the dishes immediately and set them aside to cool and solidify. Be careful not to remove the covers.

To test the bacterial content of the air in various places in the school building, two sterile dishes should be exposed in each of at least eight places, leaving one dish for a control. To make an exposure, open the dish for exactly ten minutes in all cases. Mark each dish after exposure, indicating the place of inoculation. Incubate for forty-eight hours. After incubation, the bacteria which fell on the agar will have

multiplied to form visible colonies. By counting the colonies, a determination can be made of the exact number which fell in ten minutes. Comparison of the various areas tested will provide valuable data which should be recorded in a table. Good places to run the tests are in the biology laboratory, gymnasium, crowded hallway, cafeteria, auditorium, study hall, locker room, and out-of-doors.

Using seventeen sterile Petri dishes, make contact plates of the following: lipstick, powder puff, chewed pencil eraser, unchewed pencil eraser, door knob, spout of drinking fountain, drinking-fountain handle, penny, nickel, dime, dollar bill, fingers of washed hand, fingers of unwashed hand, eye glasses, handkerchief, and fountain pen. Certain of these inoculations may be made by touching the object to the surface of the media. In some cases it is best to touch the media to the object. Cover the dish at once, label, and incubate for forty-eight hours. Use an uninoculated dish as a control. The amount of growth will indicate the relative abundance of bacteria which were present on each object. Record the results in a table.

SUMMARY

No matter what a persons vocation is, there is always the need for improvement. There has been for many years, and there still is, a need for improvement in the teaching of high school biology. During the last few years, the increasing use of student projects has done much to alleviate this situation. But, there still those who do not make use of student projects, either because they do not have at hand the proper materials or because they lack fundamental knowledge of the subject.

It is the writer's opinion that any biology teacher can, with a little effort and determination, become familiar enough with the techniques involved in directing project work to successfully guide his students, and can also build up a very complete file of resource materials in a fairly short time.

Since the primary aim of science teaching in high school is to build up a keen interest in science, we as science teachers should strive toward those things which tend to accomplish this aim. It is the opinion of the writer that the use of biology projects in the high school biology course will do more to create interest in biology and related sciences than any other single thing.

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APPENDIX A

A SUGGESTED LIST OF PROJECTS SUITABLE FOR THE HIGH SCHOOL BIOLOGY STUDENT

I. GENERAL PROJECTS

A. Making Things

1. Relief maps to show drainage areas of rivers of U. S.
2. Relief maps to show effect of mountains and prevailing winds on rainfall in U. S.
3. Dioramas of special habitats, prehistoric life, etc.
4. Clay models of animals, ancient or modern
5. Soap carvings of animals
6. Model beaver dam and lodge
7. Model plant or animal cells
8. Models of common protozoa
9. Models of representatives from each plant or animal phylum
10. Models of vertebrate hearts
11. Model of human heart
12. Models of vertebrate brains
13. Model of human brain
14. Model of human digestive system
15. Model of human tooth
16. Model of human eye
17. Model of reflex arc
18. Models of puffball life cycle
19. Models of moss or liverwort life cycle
20. Models of fern life cycle
21. Models of plant organs or structures
22. Skeletons of vertebrates
23. Electrical model of mental activity
24. Materials to test color perception
25. Apparatus to demonstrate heart beat in frog
26. Microprojector
27. Animal cages
28. Aquarium or terrarium
29. Incubator (egg or bacteriological)
30. Bird Feeding stations, bird houses, bird baths

31. Plastic imbedding (insects, small animals, plants)
32. Dry mounts of insects, reptiles, etc.
33. Plaster of Paris casts of animals or animal foot prints
34. Insect observation box
35. Glass-fronted germination box
36. Plant press

B. Charts

1. Island formation from coral
2. Cave formation
3. Young, mature, and old rivers
4. Profile of a river
5. Formation and stages in life of Great Lakes
6. Glaciers
7. Life Zones
8. Cell types
9. Mitosis and meiosis
10. Evolution of horse, man or fish
11. Balance of nature
12. Oxygen-carbon dioxide cycle
13. Nitrogen cycle
14. Energy cycle
15. Food chains
16. Plant and animal phylum trees
17. Life history of a representative from each major phylum of plant or animal
18. Insect parts
19. Systems of man
20. Human body as a machine
21. Life cycles of parasitic worms
22. Hibernation
23. Protective adaptations in plants, animals and insects
24. Enemies of one particular organism
25. Photosynthesis as a factory
26. Circulation in man before and after birth
27. Composition of blood
28. Proteins and amino acids
29. End products of food digestion
30. Heredity of hybrid corn strains, Drosophila, etc.
31. Causes of varied amount of rainfall in U. S.
32. Plant formations of U. S.
33. Forest regions of U. S.
34. Bird migration calendar
35. Map showing national parks, national forests, game refuges in the U. S.
36. Map of your state showing soil types, mineral abundance, etc.
37. Map of your community, locating as many different habitats as possible

C. Photography

1. Monocot and dicot flowers

2. Spring flowers, summer flowers or fall flowers
3. Composite flowers
4. Pollen grains
5. Types of fungi
6. Types of algae
7. Mosses
8. Ferns
9. Slime molds
10. Diatoms
11. Native weeds
12. Tree leaves
13. Types of roots
14. Types of stems
15. Kinds of trees
16. Types of tree branching
17. Time-lapse pictures showing germination of seeds, growth of seedling, opening of flowers, etc.
18. Photographic record of habitat types
19. Kinds of protozoa
20. Kinds of crustaceans
21. Kinds of insects
22. Kinds of butterflies
23. Life histories of insects
24. Toads, frogs and salamanders
25. Life history of frog
26. Snakes, lizards and turtles
27. Birds
28. Bird nests
29. Bird eggs
30. Variation in butterflies
31. Variation in leaves
32. Spider webs
33. Life in a drop of water
34. Reproduction in protozoa
35. Embryology of snail
36. Organisms in their natural environments
37. Flood control, irrigation, and soil conservation projects in your community
38. General photomicrography
39. Lantern slides
40. Collection of biological illustrations from magazines, supply catalogs, etc., which can be used in an opaque projector, displayed on bulletin board, etc.
41. Learn operation of projection equipment

D. Microscopy

1. Stain slides of plant or animal tissues
2. Whole mounts of simple objects
3. Bacteria smears
4. Nitrogen-fixing bacteria
5. Slime molds
6. Molds

7. Spores
8. Pollen grains
9. Protozoa
10. Effects of chemicals on protozoans
11. Adjustment of protozoans to environmental changes
12. Effects of hormones upon protozoans
13. Effect of hormones on division rate of Paramecium
14. Reactions of certain protozoans to others
15. Effects of X ray upon protozoans
16. Tiny crustaceans and insects
17. Chromosomes and cell division
18. Normal and abnormal cells
19. Blood typing
20. Common foods

E. Conservation

1. Soil profile
2. Capillary movement of water in soils
3. Models of soil conservation practices
4. Factors in the rate of evaporation
5. Sediment content of streams
6. School tree nursery
7. Stream pollution
8. Wildlife destruction on roads and highways
9. Diagram of an area of soil erosion, showing kind and extent of erosion and methods to be used in checking it
10. Study of river bed and flood plain, including report on flood danger, extent of cultivation, location of houses, presence or absence of levees and other control measures, etc.
11. Survey of homes of wildlife in a given area, including a report on best methods for conserving each type studied
12. Local methods of water conservation, including survey of flood control, hydro-electric plants, irrigation, etc.
13. Local or state forest conservation methods and practices
14. Local or state fish and game conservation programs and practices
15. Survey of common fish found in local waters, including record of their food

II. COLLECTIONS

1. Demonstration and display materials
2. Types of algae
3. Types of fungi
4. Mushrooms
5. Lichens
6. Types of mosses
7. Life cycle of moss
8. Types of ferns
9. Types of evergreens
10. Spring, summer or fall flowers
11. Composites

12. Seeds
13. Nuts
14. Seed dispersal
15. Grains
16. Fruits
17. Herbarium collection of native plants
18. Root types
19. Stem types
20. Winter twigs
21. Wood
22. Climbing stems
23. Tree leaves
24. Leaf prints
25. Leaves of same tree to show variation
26. Native weeds
27. Harmful plants
28. Types of vegetative reproduction
29. Stages of growth of one plant
30. Protective adaptation in plants
31. Cactus
32. Drugs
33. Examples of symbiosis
34. Fossils of plants or animals
35. Insect groups
36. Mosquito life histories
37. Honey bee life history
38. Beneficial insects
39. Harmful insects
40. Protective coloration in insects
41. Insects from your back yard
42. Galls
43. Life cycle of grasshopper
44. Moth cocoons
45. Mimics and their models
46. Butterflies of one species to show variation
47. Kinds of spiders
48. Spider webs
49. Life cycle of Daphnia
50. Local invertebrates
51. Invertebrates of the seashore
52. Arthropods of the seashore
53. Seaweeds
54. Mollusks of the seashore
55. Variation among Coquina shells
56. Life cycles of vertebrates or invertebrates
57. Small vertebrates
58. Comparative study of vertebrate embryos
59. Protective adaptation in animals
60. Kinds of fish
61. Kinds of amphibians
62. Kinds of reptiles
63. Turtle shells
64. Bird or mammal skins (must consult game laws)
65. Bird nests (during winter only)

66. Owl pellets
67. Skeletons of vertebrates (comparative)
68. Mammal skeletons or skulls

III. SURVEYS

1. Life in a small pond
2. Life in a small section of a stream
3. Life in a weed patch
4. Life in a small wood lot
5. Soil analysis and relationship to plant growth
6. Winter study of small pond or stream
7. Study of subterranean organisms
8. Ants, bees, and wasps in habitat
9. Types of trees in the community
10. Tree survey on city streets
11. Survey of reptiles and amphibians in water meter holes
12. Survey of trees and shrubs on school or home grounds for evidence of insect pests and diseases
13. Types of wildflowers in the community
14. Seasonal growth of algae
15. Distribution of bacteria on the school campus
16. Bird survey (permanent, summer, winter or transient)
17. Location and prevalence of pests, weeds, or vermin
18. Ecological study, such as study of a food chain

IV. EXPERIMENTS WITH LIVING THINGS

1. Types of vegetative reproduction in plants
2. Budding and grafting
3. Seed germination
4. Testing seed viability
5. Germination of pollen grains
6. Plant breeding
7. Conjugation in bread mold
8. Plant culture with artificial light
9. Effects of light on growing plants
10. Effect of vitamin B_1 on plant growth
11. Effects of hormones on plant growth
12. Growth of seedlings fed with various types of fertilizers
13. Effects of lack of one mineral on plant growth
14. Use of "tracer" elements to determine physiological processes in plants
15. Products of photosynthesis
16. Growth of plants in nutrient media
17. Study of factors affecting photosynthesis
18. Conduction of water in stems
19. Amount of water given off by various plants
20. Growth of moss on agar
21. Growth of algae on agar
22. Extraction of leaf pigments
23. Extraction of plant enzymes

24. Effect of temperature on rate of fermentation by yeast
25. Tropisms in plants
26. Responses of roots
27. Effects of environment on water plants
28. Effect of X ray on radishes, oats, etc.
29. Plant and animal respiration
30. Plant and animal hormone experiments
31. Experiments with bacteria
32. Reducing growth of bacteria
33. Regeneration in hydra, planaria, or earthworms
34. Regeneration in protozoans
35. Artificial induction of breeding in frogs
36. Metamorphosis in grasshopper or butterfly
37. Embryonic development of vertebrates
38. Embryology of snail
39. Experiments with sow bugs
40. Dietary experiments with rats or mice
41. Vitamin deficiency experiments with rats or mice
42. Effect of X ray on guppies
43. Effect of too much of any hormone upon growth of chickens
44. Effects of insecticides on insects
45. Gain and loss of water by frogs
46. Observing heart beat of frog
47. Observing capillaries in frog or fish
48. Simple responses in protozoans
49. Responses in earthworms
50. Responses in crayfish
51. Responses in insects
52. Learning in fish, frogs, chickens, or mice
53. Behavior of a pet dog or cat
54. Study of human senses
55. Responses in plants
56. Planting corn lacking chlorophyll to study inheritance
57. Crossing flowers to study color inheritance
58. Blending in four-o'clocks
59. Effect of X ray upon plant inheritance
60. Family likenesses in seeds of related plants
61. Family likenesses in pollen of related plants
62. Crossing fruit flies to study heredity
63. Variations in color and markings of a single species of butterfly
64. Crossing parakeets to study color
65. Effect of X ray upon mice
66. Crossing in hogs or cattle
67. Crossing mice to study coat color
68. Variations in size of living things
69. Variations in coquina shells
70. Tracing an outstanding family characteristic
71. Study of twins
72. Study of identical twins by a third person
73. Inheritance of taste shown with phenylthiocarbamide
74. Study of inherited traits using corn, peas, tobacco, etc.
75. Study of inherited traits using Mormoniella (wasp)

76. Demonstration of the various systems of frogs, rats, rabbits
77. Demonstration of the comparative anatomy of the classes of vertebrates

V. COLLECTING AND CARING FOR LIVING THINGS

1. Preparation of swamp, bog, woodland or desert terraria
2. Preparation of miniature sealed biomes
3. Balanced aquarium
4. Balanced terrarium
5. Comparison of two environments
6. Pond life
7. Marine aquarium
8. Demonstration ant colony
9. Observation colony of bees
10. Culture protozoans
11. Culture flatworms
12. Culture Hydra or Planaria
13. Culture and study life history of Daphnia, Cyclops or brine shrimp
14. Raise insects
15. Raise earthworms
16. Raise mealworms and study life history
17. Raise silk worm moth and study life history
18. Raise cockroach and study life history
19. Care for frog eggs, observe and record life history
20. Raise mice, rats, guinea pigs, hamsters, etc.
21. Raise frogs, toads, snakes, lizards, turtles, etc.
22. Raise tropical fish, native fish, goldfish, etc.
23. Culture pure strains of bacteria
24. Culture molds
25. Culture fresh-water algae
26. Raise insectivorous plants
27. Show alternation of generations in moss or ferns
28. Germinate fern prothalli
29. Prepare a moss garden
30. Collect and culture molds from soil
31. Start bulbs in green house
32. Start seeds for spring garden planting
33. Start a "natural garden" of native plants on the school campus

VI. PROJECTS ON HUMAN HEALTH, PHYSIOLOGY AND ANATOMY

1. Study of human skull
2. Chemistry of digestion
3. Effect of temperature on protein digestion
4. Changes in the teeth with age
5. Comparative dental anatomy
6. Amylase activity of saliva
7. Acid production in the mouth
8. Survey of oral bacteria

9. Record of weight and height of class members for school year
10. Study of human senses
11. Effect of oxygen on blood
12. Blood typing
13. Blood counts
14. Testing common remedies
15. Investigation of leading causes of death
16. Separating tars in tobacco
17. Spot maps showing locations of communicable diseases in the community
18. Survey of local sanitation and health conditions
19. Accident survey
20. Control of mosquitoes, flies, and other insect pests in the community
21. Maps showing breeding places of animal pests in the community

APPENDIX B

SOURCES OF FREE OR INEXPENSIVE

PROJECT AIDS AND MATERIALS

American Biology Teacher
Interstate Press
19 N. Jackson St.
Danville, Illinois

American Dental Association
Chicago, Illinois

American Museum of Atomic Energy
Educational Section
P. O. Box 117
Oak Ridge, Tennessee

American Museum of Natural History
New York City, New York

Carolina Biological Supply Co.
Elon College, North Carolina

Future Scientists of America
Foundation
1201 16th St., N. W.
Washington 6, D. C.

Henry Holt and Co.
383 Madison Ave.
New York 17, New York

National Audubon Society
1000 Fifth Avenue
New York City, New York

National Science Teachers Assn.
1201 16th St., N. W.
Washington 6, D. C.

Science Clubs of America
1719 N St., N. W.
Washington 6, D. C.

Science Service
1719 N St., N. W.
Washington 6, D. C.

The Science Teacher
Publication-Sales Section
National Education Association
1201 Sixteenth St., N. W.
Washington 6, D. C.

Turtox Service Department
General Biological Supply House
8200 South Hoyne Ave.
Chicago 20, Illinois

United States Department of
Agriculture
Washington, D. C.

W. M. Welch Scientific Co.
1515 Sedgwick St.
Chicago 10, Illinois

VITA

Lavon Bradley Ellis

Candidate for the Degree of

Master of Science

Report: PROJECTS FOR THE HIGH SCHOOL BIOLOGY STUDENT

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Professional experience: Began teaching science in September, 1956; have taught biology in high school past three years; am at present a member of Texas State Teachers Association and National Science Teachers Association; have been a member of Summer Biology Institute at Oklahoma State University for past two summers.