BENHA UNIVERSITY,

FACULTY OF SCIENCE,

ENTOMOLOGY DEPARTMENT.

Field training

(491E)

Prepared by

Prof. Abdelwahab A. Ibrahim

Overview:

1. This course serves as a good introduction to the scientific method.
2. Students will develop their own hypotheses and experimental design, as well as analyze data to draw conclusions.
3. The course covers all the steps of the scientific method and allows students to think like scientists and answer the questions.
4. Unlike many classroom experiments where teachers have an idea of the "correct" results, in this course each student or class might come up with his or her own design and all data is meaningful.

General aim of the course:

This course will introduce students with a background in basic field entomology. The course will include insect survey and collecting methods, observation and experimentation on insect ecology and behavior, large and small-group research projects, scientific writing, and discussion of a set of research papers on the biology of insects.

Aims of the course:

1. Introduce students to insect field survey methods and insect biodiversity.
2. Develop skills in insect natural history observation.
3. Formulate project questions and hypotheses; collect ecological and behavioral field data to analyze a question/hypothesis.
4. Apply basic statistical methods as applied to field ecological and behavioral studies.
5. Explain/present scientific results orally and in writing.
6. Interpret, analyze, and discuss scientific literature on insect biology and ecology.

Learning outcomes

On successful completion of the course, the student should be able to:

1. Define the techniques, designs and methods used in entomology under experimental field conditions.
2. Describe the methods and tools used in modeling, sampling, and surveying insect population.
3. Mention the safety precautions needed during field work.
4. Formulate and test hypotheses using appropriate experimental design and statistical analysis of data.
5. Design plans for making entomological experiments in the field.
6. Select the proper method for trapping, surveying and sampling insect populations.
7. Make field experiments, keeping the safety of workers and environment.
8. Use laboratory and field–based methods to generate data.
9. Conducts insect surveys to determine insect population in different habitats.
10. Communicate with others; work in team and involvement in group discussion.
11. Present data and results orally and in written form.
12. Practice self-learning.

***Overall structure of the course*:**

A. methods of insects collection:

- Including trapping, black-lighting, and aerial netting, as well as insect diversity through lectures and slide shows on insect field identification to the family level.
- We will expect students to make a small insect collection based on their exploration of the areas visited.

B. Insect behavior and natural history:

- We request that students to observe and document photographically insects in the field.
- Things to consider when making these observations:
  - ✓ What is the insect feeding on?
  - ✓ What kinds of behaviors is the insect engaged in?
  - ✓ Are there multiple individuals of the same species interacting with each other and what is the nature of these interactions?
  - ✓ What sort of interspecific interactions do you observe (e.g., predation, herbivore, parasitism, symbiosis, etc.)?
  - ✓ What order/family do you think the insect is in?

C. Designing field experiments:

- Based on behavioral and natural history observations made above, we will develop both a large group as well as small group projects on field ecology behavior and control.
- What predictions might you make about the ecology, behavior, or natural history of insects based on your observations?

- What sort of hypotheses might you develop based on your observations? How would you test this hypothesis? What data might you collect and how would you analyze these data?
- What statistical tests would you conduct to analyze your data?
- Write down specific null and alternative hypotheses that you would use to frame your question.
- Will you have a sufficient sample size to analyze your question?

Field trips

Be prepared for the field.

General tips: no sandals, wear light-weight, light colored clothing, long pants & short sleeves, sunscreen, insect repellent, and bring some sort of bag/pack to carry equipment/supplies. We will provide all the collecting equipment.

Note: Students are encouraged to photograph interesting insect specimens in the field and to share these images with the group.

Before making experiments:

1. Scientific research may begin by generating new scientific questions that can be answered through replicable scientific investigations that are logically developed and conducted systematically.

2. Scientific conclusions and explanations result from careful analysis of empirical evidence and the use of logical reasoning.

3. Results from investigations are communicated in reports that are scrutinized through a peer review process.

4. All experiments have certain things in common, so designing an experiment usually includes the following steps:

5. You must decide what question you want to have answered. This is the goal, or objective, of the experiment.

6. The goal of the experiment will dictate what to include in the experiment to help you answer your question.

7. The individual things that you wish to test in your experiment are called “treatments” and the physical areas to which the treatments are applied are called “plots.

8. Then you need to decide how the treatments should be physically arranged in the field. Technically, this is what is called the “experimental design.

9. Experiments answer your original question by allowing you to make unbiased comparisons among the treatments you selected.

10. You will need some way to evaluate how well each treatment worked to make comparisons among treatments.

11. The information you collect to help you make those comparisons (such as yield, insect counts, or disease severity) is called "data."

12. Finally, you need an objective way to evaluate the data. This is usually done through statistical analysis.

Biodiversity

1. Insects and other invertebrates are essential for any healthy ecosystem. Invertebrates aerate the soil, eat plant pests, pollinate flowers, and provide food for other organisms such as birds and small mammals.

2. Ecosystems with high invertebrate diversity may be more stable over time and able to support a greater diversity of plants and animals. High diversity in an ecosystem can also affect ecosystem services.

3. Ecosystem services are resources and processes generated by ecosystems that benefit humans. For example, food, water, energy, air quality, water purification, disease control, nutrient cycling, seed dispersal, and recreation are all ecosystem services.

Surveying & Sampling Insect Populations

1. **Obtaining data:** For understanding pest activities, life cycle, long-range migration, local movement, feeding and reproduction.

2. **Sampling program:** Determines the timing of life cycle to search for, where to set up traps, how to trap, etc.

3. **Sampling technique:** Is the specific method used- how the insects are counted and what tools are used.

- ✓ **In Situ sampling technique:** Direct Counts- good for large, conspicuous insects, mostly on plants, insects are not necessarily removed from the field.
- ✓ **Knockdown/extraction technique**: insects removed from habit via jarring, chemical knockdown, Berlese funnel (light/heat extraction) and soil extraction.

- ✓ **Indirect sampling methods:** sample the amount of injury rather than insect's numbers and requires research base relating injury to population levels.

Main sampling methods

1. **Scouting:** The general occurrence and abundance of invertebrates within each plot is assessed with scouting. Scouting consists of detailed inspection of soil and individual plants for insects within a 0.25 x 0.25m area of the plot.

2. **Sweep netting:** Sweep netting can also provide data on general occurrence and abundance of invertebrates that fly and/or perch on plants.

3. **Sticky traps:** Detailed sampling for flying insects is performed with sticky traps. One sticky trap is placed into each plot for one week prior to collecting.

4. **Pitfall traps:** Detailed sampling for ground-dwelling insects and other invertebrates is performed with pitfall traps. One pitfall trap is placed into each plot for one week prior to collecting.

SCOUTING PROCEDURE

Materials

1. Invertebrate Guide
2. 0.25m x 0.25m frame
3. Camera to photograph unknown invertebrates

Instructions

1. Place frame at random location in the plot using the 'Randomization Procedure' protocol.

- ✓ Sides of the frame should be parallel to the plot borders.
- ✓ Place the center of the frame on the location identified from the 'Randomization' protocol.

Note: If disturbance (walking path, animal burrow, previous biomass sampling, etc.) is present in the area, repeat the 'Randomization Procedure' to select a new sampling location.

2. Search the ground and plants inside the frame and use the Guide to identify organisms by order.

3. Count and record the number of individuals of each order. Record your results on the Invertebrate Biodiversity Data Sheet.

4. Photograph and describe any invertebrate you cannot identify.

- ✓ Identify this invertebrate later.
- ✓ List as: Unknown Invert 1, Unknown Invert 2, etc.

5. Repeat for each plot.

<u>SWEEP NETTING PROCEDURE</u>

Materials

• Invertebrate Guide

• Sweep nets

• White paper or trays

• Camera to photograph unknown invertebrates

Instructions

1. For each sweep, swing net through tops of plants in one.

2. Take 3 sweeps of each plot.

3. After the sweeps, quickly move the net through the air to move all of the invertebrates to the bottom of the net bag.
4. Empty invertebrates onto white paper or tray.
5. Identify each invertebrate to order using the Invertebrate Guide.
6. Count and record the number of individuals of each order. Record your results on the Invertebrate Biodiversity Data Sheet.
7. Photograph and describe any invertebrate you cannot identify. Identify this invertebrate later. List as Unknown Invert 1, Unknown Invert 2, etc. for each invertebrate you cannot identify.
8. Repeat for each plot.

Note: Sweep netting is most successful when plants are at least 6 inches tall.

Entomology Traps

1. Insect traps are the most efficient way to effectively trap a large area within a limited amount of time, with limited staff and resources.
2. Each of the insect surveys conducted utilizes one or many trapping methods to passively collect the targeted insect species.

Sticky Traps

1. Sticky traps are used as one of the effective IPM strategy for monitoring different types of the insects.
2. They provide an easy method for estimation of pest population density and require low cost and less skilled labor and are helpful for developing an eco-friendly control strategy.

Pitfall Traps

1. There are many variations of pitfall traps, but in its most basic form, a pitfall trap consists of some type of cup or other container (gallon bucket, for example) that is submerged in the soil and partially filled with a preservative.

2. Insects and other organisms crawling about on the ground simply walk into the container and then cannot get out. Pitfalls can be covered to help prevent excessive rain from overflowing the cup, they can have guide vanes that may help guide organisms into the cup, and they may be baited to capture more specific types of insects.

Tree Band Trap

1. Tree bands are made from weather resistant paper and are coated with a light adhesive.

2. Insects that walk up and down trees become entangled in the glue and are collected.

Jug or Bottle Trap

1. A Jug Trap, sometimes referred to as a Sugar-Bait Trap, is a plastic container hung from a tree or post and baited with a pheromone lure, volatile plant compounds, sugar or other sweetener, or a combination of any of these, along with a dispatching agent and/or preservative, such as propylene glycol or ethyl alcohol.

2. A new survey, using a smaller version called a bottle trap.

3. The bottle will be suspended in a tree and filled with fermenting fruit juice or other sweetened liquid.

<u>The Pan Trap, or Bee Bowl</u>

1. Is used in collecting visually-oriented flying insects, such as bees.

2. Visually-oriented insects are often attracted by color, so traps encountered may be found in white, yellow, blue, or, possibly, purple.

3. The bowl or pan is variable in size, and is most often filled with water and a mild, biodegradable detergent, which is used to break the water's surface tension.

4. These traps are set out in series of one or more bowls of each color in open areas near wildflowers, and remain up for 8 to 24 hours at a time.


<u>Pheromone and Kairomone traps</u>

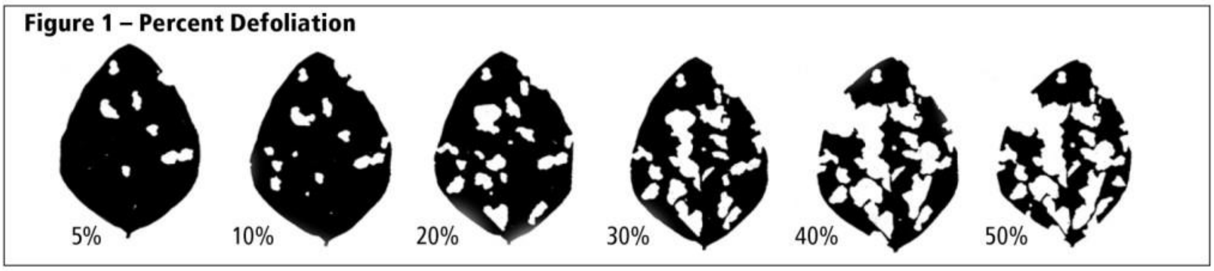
Pheromone trap   (Mimic insect pheromones)

Kairomone trap (Mimic of host plant odor)

1. To detect, understand population density and phenological information.
2. Semiochecmicals are used in intraspecific communication - sex, aggregation, alarm, trail marking

Indirect sampling methods

1. Look at leaf defoliation



2. Use root damage model

Surveying aquatic habitats

Surveying aquatic habitats provide information on aquatic insect species, their distribution, density, bionomics and susceptibility/resistance to insecticides for vector control.

Potential breeding sites of mosquito larvae include:

1. Small rain pools, hoof-prints, drains and ditches (entire water surface should be examined for larvae).
2. Brackish water (where fresh water and salt water mix).
3. Streams (larvae may be found at the edges where there is vegetation and the water moves slowly).
4. Ponds, lakes, swamps and marshes.
5. Special sites, such as wells and water containers made of cement.

Aquatic Sampling requirements

1. Vehicle to get around.
2. Good topographical map or at least a city map.
3. Data sheets.
4. Boots.
5. Dipper, collecting jars, small type net.
6. Water proof marking pens, tape etc.
7. Carrying box or container, ice chest etc.
8. Data logger, GPS

Aquatic Sampling Equipment's

The correct equipment should be used according to the breeding place

1. Dipper.
2. Pipette.
3. Aspirator.
4. Flashlight Aspirator.
5. Larval Container.
6. Bag for extra vials, pens, labeling tape, etc.

Larval collection techniques

1. Dipping.
2. Larval collection by pipetting.
3. Larval collection by pulling up aquatic plants.

Once larvae and pupae are collected they should be:

• Transported as live larvae and pupae to laboratories

• Killed using hot water between 50 – 70 0 C

• Preserved using 70% alcohol

Mosquito Adult Sampling

1. Using mouth aspirator for resting mosquitoes.
2. This aspirator is kept near the resting mosquito and is sucked.
3. After collecting in the aspirator, mosquitoes are kept at the paper cup covered with net.

Window Trap Collection (Exit Traps)

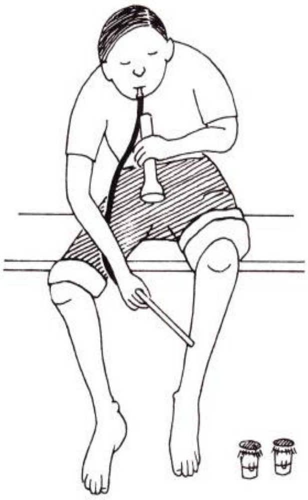
Window trap collection is done for those mosquitoes which bites indoor and goes outside for the rest.

Human and animal landing catches

1. In this type of collection technique, humans are directed to keep up their pants or trousers up to the knee and the shirt up to their shoulder. Mosquito is allowed to bite to human.

2. Immediately at the instant the biting mosquito is sucked through the mouth aspirator.

3. Since biting of mosquito may spread the disease if it is carrying organism responsible for disease, so nowadays it is not used.



Collection techniques for sand fly

Mainly adult sand flies are collected during surveys.

Methods

1. Resting adults can be aspirated directly or trapped after disturbing them.
2. Trapped using baited animal's traps, CO_2 and light traps.
3. Some sand-fly genera of the Phlebotominae subfamily are the primary vectors of leishmaniasis and pappataci fever.

Sticky trap for Sandflies

1. Sticky paper is placed on the land when sand fly jumps over it, it gets stick on the paper.

Population dynamics

Three Key Features of Populations

1. **Size: (**number of individuals in an area)
2. **Density:** measurement of population per unit area or unit volume
3. **Population Density =** number of individuals ÷ unit of space

- **Dispersion of individuals may be:**

  (Clumped, even/uniform, random).

Factors that affect density:

1. **Immigration:** movement of individuals into a population

2. **Emigration:** movement of individuals out of a population

3. **Density-dependent factors:** Biotic factors in the environment that have an increasing effect as population size increases

   E.g.: diseases, competition, parasites, and parasitoids.

4. **Density-independent factors:** abiotic factors in the environment that affect populations regardless of their density E.g.:

   E.g.: temperature, storms, habitat destruction, and drought.

Factors affecting insects in the environment

Environment of an insect population consists of:

A. Physical factors (Abiotic factors):

- Climatic factors (temperature, humidity, light)
- Edaphic factors (medium as soil)

B. Biological factors (Biotic factors):

- Members of the same species (homo-typal effects)
- Members of other species (hetero-typal effects)
- Nourishment, food source

C. Anthropogenic factors:

- Impacts of human activities

A. Abiotic environmental factors

Temperature

1. The bulk of insects die on 40-60°C (killing effect depends on the duration of high temperature)
2. Owing to their ecological plasticity insects are able to adapt and become acclimatized to the changing environment enlarging the boundaries of the tolerable temperature zone.
3. All the activities of insects require an adequate temperature (feeding, development, reproduction)
4. Developmental rate of insects is highly influenced by the temperature, it can come off only above the biological zero point or developmental threshold temperature.

humidity

1. It is obvious that all insect species have a concrete claim to water as well.

2. Humidity can also affect on the behavior of insects e.g. insects overwintering as adults emerge earlier in case of dry soil/litter; wireworms can move slower in wet soil.

3. Horizontal and vertical movement of terricol (soil inhabitant) pest is mostly influenced by the soil moisture.

4. Many insects drink water.

5. In case of many insects, embryonic development can start only after the egg absorbed some water.

6. Liquid water can be dangerous to insects (e.g.: young larvae of potato beetle can show 60% mortality because of a thunder-shower) interesting opposite that potato beetle adults can survive till two weeks on the surface of water

7. Humidity (together with temperature) effect on the developmental rate and reproduction.

Light

- Energy of natural biotopes derives almost entirely from the radiating sun.
- Light can affect insects in many ways:

intensity

- ✓ current position of the Sun in the sky
- ✓ wave length (color)
- ✓ polarization
- ✓ photoperiod

MEDIUM (AIR)

- Only a temporary space where insects may occur.
- Tiny wingless insects utilizing the ascending warm air currents were caught in surprising heights (4500m)
- Air pressure changes influence the behavior of insects too, generally all insects hide in windy weather
- Insects which are capable of flying, can generally fly against the breeze and can be carried by the wind.

MEDIUM (SOIL)

- All physical and chemical parameters of soils have effects on terricol (soil-inhabitant) insects.
- Soil organic matter content, CO_2, and pH are important for insects especially in their early larval stages.
- Its physical characteristics influence species composition of the fauna.

B. Biotic environmental factors

(FOOD)

Nutritional modes of life of insects:

- Hylophagous (eats dead organic matter)
- Necrophagous (eats dead zoogenic matter)
- Saprophagous (eats dead phytogenic matter)
- Coprophagous (eats the dung of higher animals)

• Biophagous (eats living organic matter)

• Phytophagous (herbivor) (eats living phytogenic matter)

• Zoophagous (carnivor) (eats living zoogenic matter)

• Parasite (lives on higher animal but not killing it)

• Parasitoid (lives in higher species and finally kill it)

• Episite or predator (immediately kills other species)

Other factors

Experimental design

SELECTING TREATMENTS

1. The objective, or purpose, of the study will determine the treatments included in an experiment.

2. A test may have more than one objective, although multiple objectives should be closely related.

3. if the purpose were to determine which of five fungicides works the best, then the treatments would include all five of those fungicides. If the purpose were to determine if any of the five fungicides works better than your current choice, then the treatments would include the five fungicides plus the fungicide you currently use.

4. Accurately stating the purpose of the test before the treatments are applied in the field is critical. After the treatments have begun, it will be too late to add other treatments to answer the question you really wanted to address.

5. “checks” or “controls.” Without the proper controls, you will not be able to say that the new nematicide worked better than the currently used nematicide or even that the new nematicide worked better than no nematicide!
6. The questions you wish the experiment to answer should indicate what treatments should be included as controls
7. It is frequently desirable to have both a positive and a negative control in an experiment.
8. The negative control helps you determine if the treatments being tested work better than some minimal treatment (or nothing) and positive controls help you determine if the treatments being tested work better than the current standard practice.
9. You may have several control treatments in an experiment if you currently have several viable options from which to choose.
10. For example, if you currently can choose either of two fungicides to control a leaf spot problem, you may wish to include them both as controls in your experiment when you test new products.

<u>REPLICATION</u>

1. The purpose of replication is to allow you to make a more accurate estimate of how each treatment performed even though there is uncontrolled variation in the experiment.
2. In an experiment, replication means that individual treatments (such as each of the five pesticides being tested in an experiment) have been applied to more than one plot.

3. Replication is necessary because all test plots are not identical, and that leads to variation in the data you collect;

4. you will not get the same results from two plots that received the same treatment.

5. You can take steps to minimize the effect of variation if it has an identifiable cause, but there will always be some variation among plots that cannot be controlled.

6. In statistical terms, uncontrolled variation is called experimental error.

RANDOMIZATION

1. Randomization in an experiment means that the treatments are assigned to plots with no discernable pattern to the assignments.

2. The reason randomization is important is that the positioning of treatments within the block may affect their performance.

3. One example of this is an experiment testing five corn hybrids (labeled 1 through 5) in which you plant the hybrids in the same order in each block: 1, 2, 3, 4, then 5.

4. If hybrid 2 is naturally much taller than the others, it can slightly shade the hybrids planted next to it (hybrids 1 and 3) and unfairly make them look a little bit worse than they would look if they were not planted next to hybrid 2.

5. Because you cannot anticipate all the influences that may introduce bias into a test, ALL experiments should be randomized.

	1	2	3	4	5
	1	2	3	4	5
	1	2	3	4	5
	1	2	3	4	5
	1	2	3	4	5

Completely Randomized Design

1. The completely randomized design is the simplest experimental design.
2. In this design, treatments are replicated but not blocked, which means that the treatments are assigned to plots in a completely random manner.
3. This design is appropriate if the entire test area is homogeneous (uniform in every way that can influence the results).
4. Unfortunately, it is rare that you can ever be confident of a test site's uniformity, so a completely randomized design is rarely used in field tests.
5. The completely randomized design is used more commonly in places like greenhouses.

A	**B**	**C**	**A**
A	**B**	C	B
C	A	B	C
B	A	C	B

<u>Randomized Complete Block Design</u>

1. The randomized complete block design is the most commonly used design in agricultural field research.

2. In this design, treatments are both replicated and blocked, which means that plots are arranged into blocks and then treatments are assigned to plots within a block in a random manner.

3. This design is most effective if you can identify the patterns of non-uniformity in a field such as changing soil types, drainage patterns, fertility gradients, direction of insect migration into a field, etc.

4. If you cannot identify the potential sources of variation, you should still use this design for field research but make your blocks as square as possible.

This usually will keep plots within a block as uniform as possible even if you cannot predict the variation among plots.

A	**B**	**C**	**A**
A	**B**	C	B
C	A	B	C
B	A	C	B
B LOOK 1	**BLOOK 2**	**BLOOK 3**	**BLOOK 4**

Randomized Complete Block Design

1. Blocking refers to physically grouping treatments together in an experiment to minimize unexplained variation in the data you collect (referred to as experimental error).

2. This allows the statistical analysis to identify treatment differences that would otherwise be obscured by too much unexplained variation in the experiment.

3. Variation in an experiment can be divided into two types: variation for which you can account in the statistical analysis and variation that is unexplained.

4. The goal in blocking is to allow you to measure the variation among blocks and then remove that variation from the statistical comparison of treatment means.

5. If you can anticipate causes of variation, you can block the treatments to minimize variation within each block and remove some variation from the statistical analysis.

A	**B**	**C**	**A**
A	**B**	**C**	**B**
C	**A**	**B**	**C**
B	**A**	**C**	**B**
B LOOK 1	**BLOOK 2**	**BLOOK 3**	**BLOOK 4**

Randomized Complete Block Design

1. In the most common experimental designs, a block will contain one plot of each treatment in the experiment.

2. If an experiment has five treatments, then each block will contain five plots, with each plot receiving a different treatment. When a block contains one plot of each treatment, then each block represents one replication of each treatment.

3. For this reason, blocks are frequently referred to as “replications” or “reps,” but the concept of blocking should not be confused with the concept of replication; replication and blocking serve different purposes.

4. In agricultural research, field plots are almost always blocked even when no obvious differences are present in the field.

It is much better to block when you did not really need to than not to block when you should have blocked.

A	**B**	**C**	**A**
A	**B**	**C**	**B**
C	**A**	**B**	**C**
B	**A**	**C**	**B**
B LOOK **1**	**BLOOK** **2**	**BLOOK** **3**	**BLOOK** **4**

<u>Randomized Complete Block Design</u>

1. The process of blocking follows a logical sequence.

2. First, you determine that there is something (weeds, drainage, sun/shadow, water, soil type, etc.) that is not uniform throughout the experimental area (field, greenhouse, etc.) that may influence whatever you are measuring (yield, plant height, etc.).

3. Then you can arrange your treatments into blocks so that the area within each block is as uniform as possible (see figure 2).

4. Though the area within a block should be relatively uniform, there may be large differences among the blocks, but that is what makes blocking effective.

5. Your goal is to maximize the differences among blocks while minimizing the differences within a block.

6. If the field is wide enough, an easy way to arrange blocks is to place them side-by-side, also blocks may be scattered through the field in any way that is convenient for you.

A	**B**	**C**	**A**
A	**B**	**C**	**B**
C	**A**	**B**	**C**
B	**A**	**C**	**B**
B LOOK 1	**BLOOK 2**	**BLOOK 3**	**BLOOK 4**

<u>DATA COLLECTION</u>

1. Collect only useful data.

2. For example, if the objective is "to evaluate the ability of five fungicides to reduce leaf spot incidence and severity," then collecting data on leaf spot incidence and severity and plant yield should seem obvious.

3. Collecting data on rainfall and temperature, which strongly influence leaf spots, can help you explain your results.

4. But collecting data on soil physical properties does not seem to be related to the objective.

5. You also must decide when to collect that data and if you need to collect data many times.

6. For example, in an insecticide trial, you must collect data before treatment and may be several times post treatment. The biology of the organisms involved will determine when and how frequently data should be collected.

7. You should take photographs of any differences among treatments that are easily visible.

COLLECTING UNBIASED DATA

1. It is critically important to collect unbiased data.

2. The only way to ensure this is to collect data without knowing what the treatment was in that plot.

3. It is beneficial to use some type of code on the plot stakes so that you must decode the stake number to determine what the treatment was.

4. If you do not collect unbiased data, you cannot be certain that your conclusions are correct.

STATISTICAL CALCULATIONS

1. After collecting data from a properly designed experiment, you will usually need to analyze the data with appropriate statistical calculations.

2. Statistical analysis may not be necessary if treatment differences are very large and consistent; treatment means may then be sufficient.

3. It is probably best for you to seek help in making statistical calculations.

4. If your experiment was properly designed, Extension specialists and other scientists may be willing to help you with the statistics if you involve them early in the process.

5. They can also check your proposed design for flaws and omissions.

6. If you want to do the work yourself some simple statistics can be calculated by hand, but most people will make the calculations with the help of computer software.

Specialized statistical software is available, but most spreadsheet software can calculate simple statistics.

Safety in the field

1. There are two forms for fieldwork:
2. The **local form** is for fieldwork that is relatively close to the University - for example fieldwork project that includes visiting multiple sites across the city of Benha and surrounding villages.
3. The **non-regional form** is for fieldwork that takes place in a far-off location - for example, trip to other governorates.
4. An off-campus field activity presents hazards that many students are not familiar with.
5. In planning such an activity, the supervisor must assess the precautions that are necessary to protect the health and safety of all participants.

Role of supervisor

1. 1.1. Identify the appropriate mode(s) of transportation [e.g. University-owned, leased or rented car or van, privately owned vehicle (POV)].
2. If a vehicle owned, leased or rented by ACC is not available, the College may allow the use of POVs provided that the driver(s) have a valid driver license, have proof of insurance, and assume liability for the use of their vehicle.
3. If POVs are used, students must make their own arrangements to share vehicles.
4. Any requirement for students to arrange their own transportation for field activities must be included in the course syllabus.

5. 1.2. Arrange for the appropriate personal protective equipment PPE and clothing.

6. 1.3. Establish safety policies and procedures for the conduct of the activity. This shall include, but not be limited to, the evaluation of circumstances such as terrain, road and water conditions, length of trip, and anticipated weather conditions. Driving between midnight and 6:00 AM is strongly discouraged except in cases of an emergency.

7. Obtain any required collecting permits or permissions to enter restricted areas.

8. Ensure that they have a current list of emergency telephone numbers for the area to be visited.

9. On the day prior to and day of the field activity listen to radio, television or to the National Oceanographic and Atmospheric Administration weather/marine forecast for information on temperature, precipitation, severe weather.

10. Forecast conditions shall be used to determine the necessity of safety briefings on weather or water hazards such as lightning, flash floods, and other, temperature conditions, and water currents.

11. Trip participants are briefed on general safety hazards prior to departure for the field activity and on specific hazards at each stop. Participants must also be briefed on the location of first aid kit(s) and, if applicable, fire extinguisher(s) and water safety devices.

12. Establish procedures to notify participants if a field activity has been cancelled.

Equipment and Supplies

1. Prior to a field activity, the supervisor must purchase, test and assemble appropriate equipment and supplies.
2. The supervisor must ensure that they will have the appropriate quantity of PPE for all trip participants.
3. If appropriate, the supervisor should ensure that they have a functional megaphone, water lifesaving equipment
4. All off-campus field activities must have at least one appropriately stocked first aid kit.

Standard Policies for All Field Activities

1. You must:

1. Follow instructions of person(s) in charge.
2. Wear appropriate clothing and footwear for the field activity. This will be explained to you in advance by your leader or described on a checklist provided to you.
3. Be on time for the field activity or you will be left behind and miss the activity.
4. Wear seatbelts in equipped vehicles whenever they are in operation.
5. Report any unsafe conditions to an activity leader as soon as possible. o If this involves fatigue or sleepiness of the driver of your vehicle, then you must alert other passengers and immediately request that the driver take a break.

6. o Any resistance to such a request must be reported to the professor, or if the resistance is from the leader, then you must make a confidential report to head of the department.

1. You must not:

1. Leave the group at any time without first informing the person(s) in charge of the group
2. Endanger the safety of another group member - if you do so you will be dismissed immediately and may be subsequently disqualified from participation in other ACC activities.
3. Smoke or improperly dispose of cigarette butts which might cause a fire hazard.

3. If you:

1. Damage or lose Department property you may be required to reimburse the College for repair or replacement of the loss.
2. Are stopped or arrested for illegal activities during the field activity you will be dismissed immediately
3. Fail to observe these policies and procedures you will be disciplined as specified in the ACC Student Handbook.

Each participant in an off-campus field activity must

1. Wear a seat belt whenever their vehicle is in operation.
2. Report any unsafe conditions to the RCO as soon as possible. If this involves fatigue or sleepiness of the driver of their vehicle, then they must alert other passengers and immediately request that

the driver take a break. Any resistance to such a request must be reported to the RMO.

3. Provide and administer any medications that they take orally or by injection for chronic medical conditions.

4. Any accidents involving vehicles used for off-campus field activities must be reported immediately to local law enforcement authorities, the RCO, the RMO, and, if applicable, the rental company.

5. Any accidents or incidents during a field activity that require the attention of a medical professional shall be reported to the RCO and to the RMO as soon as possible.

Insects Biodiversity Data Sheet

1. **Names:____**

2. **District:_________________________**

   Instructor/Fellow:______________________

3. **Date:_________________________________ Time:________________**

   Weather:________________________

4. **Location Name:**

5. **Collection method (Sticky Trap, Pitfall Trap, Scouting, Sweep Net) ___________________**

All insect orders are listed in a table

Insect Order	Insect Name	Number of individuals

Sample field sheet

Survey of scale insects and mealy bugs infesting citrus trees

Leaf	Tree 1		Tree 2		Tree 3	
	Scale	Bugs	Scale	Bugs	Scale	Bugs

Sample field sheet

Distribution of infestation on plant directions

Name

Date

Site Location

Site Description

INSECT SPECIES:

	NUMBER OF INSECTS COLLECTED					
	SIDE	NORTH	SOUTH	EAST	WEST	TOTAL
TREE1	LEAF 1					
	LEAF 2					
	LEAF 3					
	LEAF 4					
	LEAF 5					
TREE2	LEAF 1					
	LEAF 2					
	LEAF 3					
	LEAF 4					
	LEAF 5					
TREE3	LEAF 1					
	LEAF 2					
	LEAF 3					
	LEAF 4					
	LEAF 5					
TREE4	LEAF 1					
	LEAF 2					
	LEAF 3					
	LEAF 4					
	LEAF 5					
TREE5	LEAF 1					
	LEAF 2					
	LEAF 3					
	LEAF 4					
	LEAF 5					
TREE6	LEAF 1					
	LEAF 2					
	LEAF 3					
	LEAF 4					
	LEAF 5					

Population Dynamics   Report

Purpose:

- Analyze graphs to determine the population size of two aquatic species.
- Explain how predation, birth, and death rates impact aquatic populations.
- Describe how biotic and abiotic factors influence aquatic populations.

Research:

Prey Initial Size: The starting number of prey. (Assume that the prey have an unlimited amount of food.)

Prey Growth Rate: The birth rate of the prey. The larger the growth rate, the faster the prey population will increase.

Predator Initial Size: The starting number of predators.

Predator Death Rate: The death rate of the predators. A larger death rate means predators will die out quicker.

Capture Efficiency: This number represents the ability of the predator to capture the prey over some time interval. A larger value for this parameter means that the predators have a better chance of capturing prey.

Hypotheses:

Prediction 1: If the starting population of the prey is higher than the predators, the resulting population of the prey will be **higher** than the population of the predator.

Prediction 2: If the starting population of the predator is higher than the prey, the resulting population of the whales will be **higher** than the population of the prey.

Prediction 3: If the prey growth rate increases, the carrying capacity of the prey will be **higher** than the carrying capacity of the predator. (Both starting populations set to 25.)

Prediction 4: If the death rate of the predator increases, the carrying capacity of the prey will be **higher** than the carrying capacity of the predator. (Both starting populations set to 25.)

Materials:

1. The Population Dynamics Virtual Lab Activity
2. Population Dynamics Lab Report

Procedures:

The procedures are listed in Population Dynamics Virtual Lab Activity. You do not need to include them here.

Data and Observations

Table 1: Predation and Carrying Capacity

	Starting prey population	Starting predator population	Highest prey Population	Highest predator Population
Trial 1	25	25	49	52
Trial 2	50	25	72	80
Trial 3	25	200	160	179

Table 2: Growth Rate and Capture Efficiency

	Prey birth rate	Predator birth rate	Highest prey Population	Highest predator Population
Trial 1	0.05	0.005	49	52
Trial 2	0.09	0.005	52	73

Table 3: Death Rate and Capture Efficiency

	Prey death rate	Predator capture efficiency	Highest prey Population	Highest predator Population
Trial 1	0.05	0.005	49	52
Trial 2	0.09	0.005	68	57

Analysis and Conclusion:

Be sure to answer the following reflection questions in the conclusion of your lab report:

1. Using Table 1, which of the three trials produced the highest population for both the predator and the prey? Why do you think this trial had the best outcome for the predator and the prey populations?

   In the charts above, trail 3 has the highest amount of carrying capacity for both predator and prey because the predator population was a lot higher than the prey population.

2. Using Table 2 and 3, explain how the birth rates of the prey and the death rates of the predator affected the population for both predator and the prey.

   If the death rate of predator is higher, then the prey population will rise. This means that the carrying capacity of the prey will be higher than that of the predator.

3. Climate change (an abiotic factor) has slowly decreased habitat for the prey. Predict how a reduction in habitat could change the capture efficiency of the predator. How would this affect the carrying capacities for both the predator and the prey?

   Taking away their habitat of the prey will limit their population and as a result, predator will find it harder to find food. This can affect the carrying capacity for

both the predator and the prey. The prey' carrying capacity will lower because they won't have much amount of lad and they also have a low population.

4. If another source of prey were available to the predator, what changes in population size would you expect for the predator and the prey?

   If there was another type of prey the predator could eat, the prey' population might slightly increase while the predator' population will increase exponentially.

5. In this lesson, you learned about two patterns of population growth and decline, the sigmoid and peak phenomena. Does the predator and the prey relationship represent a sigmoid or peak phenomenon? Please provide supporting details.

   The predator and the prey relationship could represent peak phenomena. This is because when the prey population is high, the predator population will increase as well and due to this, the prey population will decrease.

6. What are the limitations of the population dynamics lab? Is the lab activity a realistic representation of an aquatic ecosystem? Note: Please provide detailed support for your opinion.

   This lab was based on nature, but it is not so accurate nor realistic as real nature because we didn't take environment changes, diseases, and other types of predators. As well as that predators have other food sources they could get instead of this prey.

SAMPLE REPORT

Perception of Different Sugars by Blowflies

by

Field training 491E

October 24, 2017

Supervisor

ABSTRACT

To feed on materials that are healthy for them, flies (order Diptera) use taste receptors on their tarsi to find sugars to ingest. We examined the ability of blowflies to taste monosaccharide and disaccharide sugars as well as saccharin. To do this, we attached flies to the ends of sticks and lowered their feet into solutions with different concentrations of these sugars. We counted a positive response when they lowered their proboscis to feed.

The flies responded to sucrose at a lower concentration than they did of glucose, and they didn't respond to saccharin at all. Our results show that they taste larger sugar molecules more readily than they do smaller ones.

They didn’t feed on saccharin because the saccharin we use is the sodium salt of saccharin, and they reject salt solutions.

Overall, our results show that flies can taste and choose foods that are good for them.

INTRODUCTION

All animals rely on senses of taste and smell to find acceptable food for survival.

Chemoreceptors are found in the taste buds on the tongue in humans (Campbell, 2008), for example, for tasting food. Studies of sensory physiology have often used insects as experimental subjects because insects can be manipulated with ease and because their sensory-response system is relatively simple (E. Williams, personal communication). Flies

are able to taste food by walking on it (Dethier, 1963). Hollow hairs around the proboscis and tarsi contain receptor neurons that can distinguish among water, salts, and sugars, and flies can distinguish among different sugars (Dethier, 1976). These traits enable them to find necessary nutrition.

In this experiment we tested the ability of the blowfly *Sarcophaga bullata* to taste different sugars and a sugar substitute, saccharin. Because sucrose is so sweet to people, I expected the flies to taste lower concentrations of sucrose than they would of maltose and glucose, sugars that are less sweet to people. Because saccharin is also sweet tasting to people, I expected the flies to respond positively and feed on it as well.

METHODS

We stuck flies to popsickle sticks by pushing their wings into a sticky wax we rubbed on the sticks. Then we made a dilution series of glucose, maltose, and sucrose in one-half log molar steps (0.003M, 0.01M, 0.03M, 0.1M, 0.3M, and 1M) from the 1M concentrations of the sugars we were given. We tested the flies' sensory perception by giving each fly the chance to feed from each sugar, starting with the lowest concentration and working up. We rinsed the flies between tests by swishing their feet in distilled water.

We counted a positive response whenever a fly lowered its proboscis. To ensure that positive responses were to sugars and not to water, we let them drink distilled water before each test. See the lab handout Taste Reception in Flies (Biology Department, 2000) for details.

RESULTS

Flies responded to high concentrations (1M) of sugar by lowering their proboscis and feeding. The threshold concentration required to elicit a positive response from at least 50% of the flies was lowest for sucrose, while the threshold concentration was highest for glucose (Fig. 1). Hardly any flies responded to saccharin. Based on the results from all the lab groups together, there was a major difference in the response of flies to the sugars and to saccharin (Table 1). When all the sugars were considered together, this difference was significant ($t = 10.46$, $df = 8$, $p < .05$). Also, the response of two flies to saccharin was not statistically different from zero ($t = 1.12$, $df = 8$, n.s.).

DISCUSSION

The results supported my first hypothesis that sucrose would be the most easily detectable sugar by the flies. Flies show a selectivity of response to sugars based on molecular size and structure. Glucose, the smallest of the three sugars, is a monosaccharide. The threshold value of glucose was the highest in this experiment because a higher concentration of this small sugar was needed to elicit a positive response.

Maltose and sucrose are both disaccharides but not with the same molecular weight or composition. It has been shown that flies respond better to alpha-glucosidase derivatives than to beta-glucosidase derivatives (Dethier 1975). Because sucrose is an alpha glucosidase derivative, it makes sense that the threshold value for sucrose occurs at a lower concentration than that for maltose. This might also be the reason why sucrose tastes so sweet to people.

My other hypothesis was not supported, however, because the flies did not respond positively to saccharin. The sweetener people use is actually the sodium salt of saccharic acid (Budavari, 1989). Even though it tastes 300 to 500 times as sweet as sucrose to people (Budavari, 1989), flies taste the sodium and so reject saccharin as a salt. Two flies did respond positively to saccharin, but the response of only two flies is not significant, and the lab group that got the positive responses to saccharin may not have rinsed the flies off properly before the test.

Flies taste food with specific cells on their tarsal hairs. Each hair has, in addition to a mechanoreceptor, five distinct cells – alcohol, oil, water, salt, and sugar – that determine its acceptance or rejection of the food (Dethier, 1975). The membranes located on the tarsi are the actual functional receptors since it is their depolarization that propagates the stimulus to the fly (Dethier, 1975). Of the five cells, stimulation of the water and sugar

cells induce feeding, while stimulation of the salt, alcohol, and oil receptors inhibit feeding. More specifically, a fly will reject food if the substrate fails to stimulate the sugar or water receptors, stimulates a salt receptor, or causes a different message from normal (e.g., salt and sugar receptors stimulated concurrently) (Dethier 1963).

Flies accept sugars and reject salts as well as unpalatable compounds like alkaloids (Dethier & Bowdan, 1989). This selectivity is a valuable asset to a fly because it helps the fly recognize potentially toxic substances as well as valuable nutrients (H. Cramer, personal communication). Substances such as alcohols and salts could dehydrate the fly and have other harmful effects on its homeostasis (Dethier, 1976). Thus, flies are well adapted to finding food for their own survival.

ACKNOWLEDGMENTS

I thank Prof. --------------- for help with the t-test and my lab partners for helping me conduct and understand this experiment.

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Table 1. The average number of flies in each lab group that fed from 0.3M concentrations of each chemical tested. The mean ± standard deviation is shown.

chemical tested	number of 10 flies responding
glucose	3.2 ± 1.5
maltose	7.8 ± 2.3
sucrose	8.6 ± 2.1
saccharin	0.2 ± 0.5

Fig. 1. Taste response curves of flies to different concentrations of the sugars glucose, maltose, and sucrose.

Draw the data in table using Excel program