

## Lab Modules (*Sample offerings, to date*)

### **Anaerobic Systems Microbiology**

This module will expose students to the practical laboratory methods used in anaerobic microbiology. Students will learn about the physiology of anaerobic microbes with a particular focus on biofuels organisms. They will get hands on training to culture live cells using an anaerobic gassing station. Students will also have the opportunity to take physiological measurements, extract RNA for microarray analysis, and learn to analyze genomic data.

**Instructors:** Prof. Jeffrey Blanchard and Supratim Mukherjee (Microbiology)

### **Analysis of DNA Methylation Using Pyrosequencing**

In this two-day module, students will learn how to create and run pyrosequencing assays for assessing DNA methylation. Pyrosequencing, based on the sequencing by synthesis principle, relies on detection of pyrophosphate release upon nucleotide incorporation, and provides accurate quantification of the percent methylated DNA. This two-day module will cover bisulfite-conversion of DNA, primer design of bisulfite-modified DNA for pyrosequencing, polymerase chain reaction for pyrosequencing, and analysis of DNA methylation using a 24-well pyrosequencer. Module time will be divided among a computer lab to design pyrosequencing assays, lectures on different aspects of pyrosequencing, and hands-on lab time.

**Instructors:** Prof. Kathleen Arcaro and Eva Browne (Veterinary & Animal Sciences)

### **Building a Microscope**

This module will expose students to optics theory and experiment by building a transmitted light microscope. Lecture will include basic ray optics and the path of a modern microscope. Lab work will include the construction of a microscope, leading to the imaging of several specimens.

**Instructors:** Prof. Jennifer Ross (Physics)

### **Drug Delivery**

This two-day module will cover key topics of drug delivery through lecture and laboratory instruction. Students will learn the fundamentals of hydrogel design and synthesis, and see how hydrogel design impacts the release profile in delivery applications. Students will explore how the structural properties of delivery vehicles can influence drug release rates and biodistribution of drugs in vivo, and how mechanical properties can be important factors in thermoreversible delivery systems. Scattering techniques, rheometry, microscopy, and UV techniques will be demonstrated.

**Instructors:** Prof. S. Thayumanavan (Chemistry) and Prof. Surita Bhatia (Chemical Engineering)

### **Introduction to Cell Culture**

Students will set up and passage insect cells, and learn sterile technique for large (lab-scale) cell culture. Protein expression experiments will be: stable insect cell line, baculovirus-infected cell line (both in shake flasks) vs. stable insect cell line in the Wave bioreactor. Students will run activity assays to determine expression levels and count cells using a hemacytometer and a Cedex analyzer.

**Instructors:** Nat Clark and Prof. Scott Garman (Biochemistry & Molecular Biology), Prof. John Burand (Plant, Soil & Insect Sciences), Genzyme guest lecturer

### **Introduction to Microarray Analysis**

Microarrays provide a beautiful perspective of the transcriptional dynamics of a cell. This module will introduce students to microarray technology, experimental design, and data analysis. Students will learn how to use the statistical programming language R, MEV, NCBI's GEO database and other bioinformatic tools for microarray analysis.

**Instructors:** Prof. Jeffrey Blanchard (Microbiology)

### **Kinesin Motor Protein Purification and Biophysical Assessment**

This two-day module will provide hands-on experience, which includes working with bacteria to express protein, performing GFP-kinesin purification using an affinity tag and column, and performing optical microscopy using biophysical assays to assess the protein produced.

**Instructor:** Prof. Jennifer Ross (Physics)

### **Quantitative Reconstruction of Three-Dimensional Fluorescence Images**

Students will learn to reconstruct 3D stacked confocal fluorescence images. Students will prepare yeast culture expressing YFP/GFP-tagged proteins, acquire confocal fluorescence images, perform deconvolution, and compare and analyze the results.

**Instructor:** Prof. Wei-Lih Lee (Biology)

### **Modeling Cellular Metabolism and Processes**

This module will describe how modeling can be effectively used to interpret and guide cellular-based experiments and processes including fermentation, cell culture and cancer therapeutics. Students will first learn about mathematical modeling of cellular metabolism and heterogeneity. Students will have the opportunity to perform yeast fermentation experiments in a controlled parallel reactor system and to measure relevant metabolic outputs such as biomass, glucose, glycerol and ethanol concentrations. Students will also gain experience in measuring cell population properties using a state-of-the-art Coulter Counter system. Data will be incorporated into models and predictions will be compared to experimental data.

**Instructor:** Prof. Mike Henson and Prof. Susan Roberts (Chemical Engineering)

### **Topics on Fluorescence Spectroscopy: Fluorescence Lifetime Determinations**

The fluorescence lifetime is one of the most important characteristics of a fluorescent probe because it defines the time window of observation of dynamic phenomena (FRET, quenching, anisotropy). We will introduce the principles for pulse fluorometry and phase-modulation fluorometry, and learn to determine the lifetime of fluorophores in biological samples.

**Instructors:** Prof. Alejandro Heuck (Biochemistry & Molecular Biology); Fabian B. Romano & Benjamin Johnson (TAs)

### **Polymerase Chain Reaction (PCR)**

This two-day module will cover the basics of how PCR works, primer design, troubleshooting, and applications of PCR, such as PCR-based cloning, site-directed mutagenesis, and reverse-transcriptase PCR. Students will practice designing PCR primers, setting up and running PCR reactions, and analyzing PCR products on an agarose gel.

**Instructor:** Prof. Janice Telfer (Veterinary & Animal Sciences)

### **Hands-on X-ray Diffraction**

This workshop covers crystal growth, preparation and diffraction, data collection and processing, and model building. After participating in a morning lecture, each participant will have the hands-on experience of growing and freezing protein crystals, mounting them in the X-ray beam, watching data collection proceed, and building a model into electron density using a program called O. The course is intended for anyone with an interest in practical aspects of structural biology,

**Instructors:** Prof. Scott Garman (BMB), Jeanne Hardy, and Karsten Theis (Chem)