ICE IGERT Lab Modules, Summer 2008

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)

Maximum Class Size: 8

Dates: July 10 – 11, from 8:30 am – 4:30 pm

Location: July 10: LGRT 201; July 11: E-Lab II, Rm 108

Polymerase Chain Reaction (PCR)

This two-day module will cover the basics of how PCR works, primer design, trouble-shooting, 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 (Vet & Animal Sci)

Maximum Class Size: 8

Dates/Times: July 14 – 15, from 9:00 am -5:00 pm

Location: Meet in Paige Lab 404

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)

Maximum Class Size: 12

Dates: July 16 - July 17, from 8:30 am – 4:30 pm

Location: Hasbrouck 310/309

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)

Dates/Times: August 6 – 7, from 8:30 am – 4:30 pm

Location: Meet in LGRT 201

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)

Maximum Class Size: 8

Dates/Times: August 25, from 12:00pm – 3:00pm; August 26, from 12:00-5:00pm; August 27, from 12:00-5:00pm; August 28, from 12:00pm – 3:00 pm.

Location: Meet in Morrill 351

ICE IGERT Lab Modules, Summer 2009

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 (BMB), Prof. John Burand (PSIS), & Genzyme guest lecturer

Maximum Class Size: 10

Dates: July 13 from 9:00 am – 2:00 pm; July 14-16 from 9:00 am – 11:00 am

Location: 7^{th} Floor LGRT (TBD)

Automating Plant Transformations

Students will be exposed to scaling up a relatively low through-put process in order to make it high through-put. Using a moss model system, students will isolate DNA, transform plants, and analyze phenotypes resulting from the transformation. The transformation will be done using a semi-automated procedure.

Instructors: Prof. Magdalena Bezanilla (Biology) and Shu-Zon Wu (TA)

Maximum Class Size: 6

Dates/Times: August 18-19, from 1:00–5:00 PM; August 26, from 9:00-5:00pm

Location: Meet in Morrill 122

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)

Maximum Class Size: 10

Dates/Times: July 27-29, and July 31, 9 am – 1 pm

Location: LGRT

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)

Maximum Class Size: 8

Dates/Times: Aug 24-25, 27-28, 9:00 am – 1:00 pm

Location: Meet in Microbiology Conference Room, N202 Morrill IV

ICE IGERT Lab Modules, Summer 2010

Introduction to Genome Expression Analysis

Genome expression analysis provide a beautiful perspective of the transcriptional dynamics of a cell. This workshop will introduce students to microarray and next-generation sequence 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 genome expression analysis.

Instructors: Prof. Jeffrey Blanchard (Microbiology)

Maximum Class Size: 12

Dates: June 1-4, from 8:30 am – 12:30 pm

Location: Integrated Sciences Building (ISB), 145/143 (report to Room 145 on June 1st)

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, Prof. Susan Roberts, and Marty Kolewe (Chemical Engineering)

Dates/Times: June 9-10, from 8:30 am – 4:30 pm

Location: Report to Kellogg Conference Room (E-Lab II, Room 118). Group will be escorted to labs.

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)

Maximum Class Size: 6

Dates/Times: June 15-16, from 9 am – 5 pm

Location: Integrated Sciences Building, 143/363

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)

Maximum Class Size: 6

Dates/Times: July 29-30, 8:30 am – 4:30 pm

Location: Du Bois Library, Room 720 (Computer Lab, 7th Floor – allow 5 minutes to get up elevators!)

ICE IGERT Lab Modules, Summer 2011

Bioreactors and Protein Analysis

This module will teach students how to grow bacterial cultures in a bioreactor system. Students will measure cell concentrations and metabolites, harvest and lyse cells, separate the soluble and insoluble protein fractions, determine protein concentration, run SDS-PAGE gels and enzyme activity assays, and process proteins through an ion exchange column to demonstrate protein chromatography.

Instructor: Prof. Jim Holden (Microbiology) — **Maximum Class Size:** 10

Dates/Times: June 13-14, from 9 AM – 5 PM — **Location:** N211 Morrill IV N

Optical Trapping

In this module, students will learn to set up and use an "optical tweezer" for force and displacement measurements. Optical tweezers are a particular configuration of optical trap that is widely used in biophysical experiments since it can be used on almost any micron-sized dielectric particle in suspension, including microbes. Applications include binding assays based on force, the study of molecular motors and enzymatic dynamics, the study of protein folding and unfolding pathways, and the manipulation of biological structures both inside a cell and *ex-vivo* (see flyer).

Instructors: Prof. Lori Goldner & Ben Gamari (Physics) — **Maximum Class Size:** 6

Dates/Times: Rescheduled to August 18-19, from 9 am – 5 pm — **Location:** LGRT 201, 432

Quantitative Fluorescence Microscopy and Image Analysis

This module will teach students quantitative fluorescence microscopy of live and fixed samples, with emphasis on image processing and quantitative analysis. Time-lapse image acquisition will be demonstrated and quantitative spatiotemporal analysis will be performed to provide data suitable for mathematical modeling.

Instructors: Prof. Neil Forbes & Charles Swofford (Chemical Engineering) — **Maximum Class Size:** 10

Dates/Times: August 1-2, from 9 AM – 5 PM — **Location:** 1667 Du Bois Library, E-Lab II

Bioimaging

In this module, students will learn high efficiency cloning technologies, generate DNA constructs encoding for a fluorescent fusion protein, transfect constructs into animal cells for localization studies, and image cells expressing these constructs. Hands-on microscopy training will cover resolution, magnification, fluorescence and phase contrast.

Instructors: Prof. Magdalena Bezanilla & Prof. Pat Wadsworth (Biology) — **Maximum Class Size:** 6

Dates/Times: August 8-11, 9AM – 4 PM (Note, this is a 2 cr. Lab Module) — **Location:** TBD

Patch Clamp Electrophysiology

This module will teach students basic patch clamp electrophysiology on mammalian cells expressing an exogenous protein. Introductory work will be done on a Shaker voltage-gated ion channel, with the potential for more advanced photochemical-based manipulation of ligand-gated ion channels.

Instructors: Prof. Jim Chambers (Chemistry) & Kathryne Medeiros (MCB) — **Maximum Class Size:** 8

Dates/Times: August 10-11, from 10 AM – 3 PM; August 12, from 10 AM – 4 PM — **Location:** 609 LGRT

Structural Characterization of Biomaterials using Scattering

This two-day module will cover the use of advanced scattering (neutron, x-ray, and light) techniques to characterize the structure of soft materials and biomaterials. The lecture portion will cover basic background and theory of small-angle scattering and dynamic light scattering. Examples of applying these techniques to hydrogels and drug delivery vehicles will be presented. Students will also receive hands-on instruction and practice in the use of modern software packages to fit small-angle scattering data to various models to obtain information about morphology and interactions.

Instructors: Prof. Surita Bhatia and Erika Saffer (Chemical Engineering) — **Maximum Class Size:** 8

Dates/Times: August 24-25, from 9 am – 5 pm — **Location:** LGRT 201

ICE IGERT Lab Modules, Summer 2012

Structural Characterization of Biomaterials using Scattering

This two-day module will cover the use of advanced scattering (neutron, x-ray, and light) techniques to characterize the structure of soft materials and biomaterials. The lecture portion will cover basic background and theory of small-angle scattering and dynamic light scattering. Examples of applying these techniques to hydrogels and drug delivery vehicles will be presented. Students will also receive hands-on instruction and practice in the use of modern software packages to fit small-angle scattering data to various models to obtain information about morphology and interactions.

Instructors: Prof. Surita Bhatia and Erika Saffer (Chemical Engineering) **Maximum Class Size:** 8
Dates/Times: July 10, from 1:00 – 5:00 PM and July 11, from 9:00 AM – 5:00 PM **Location:** LGRT 201

Skills for Scientific Communication

This lab module will teach students how to prepare effective images for scientific communication including papers, dissertations and oral presentations. We will provide hands-on training with sessions on Pymol, Adobe Illustrator & Photoshop, Chemdraw, Endnote and other relevant programs.

Instructor: Prof. Jeanne Hardy (Chemistry), Scott Garman (BMB) and others **Maximum Class Size:** 24
Dates/Times: July 23 – 24, times TBD **Location:** ISB 321

Introduction to Biomaterials and their Applications

This lab module will explore common biomaterials and their applications to cell culture through a combination of short lectures and hands-on activities. The module will define and familiarize students with the term “biomaterial,” discuss the application of biomaterials *in vitro* and *in vivo*, and provide hands-on experience working with natural and synthetic biopolymers widely used in the field of tissue engineering. Instructors will teach methods for growing cells in and around these materials. Some familiarity with mammalian cell culture is preferred, but the module is designed for those with little to no familiarity with biomaterials.

Instructors: Prof. Shelly Peyton and Whitney Stoppel (Chemical Engineering) **Maximum Class Size:** 8
Dates/Times: July 30 – August 1, from 9:30 – 11:30 AM and 1:00 – 4:30 PM **Location:** E-Lab II, Room 118

NMR Spectroscopy

This module will introduce several NMR techniques to characterize the structure of complex molecules. Following a short lecture on the potential applications of NMR, each student will select one or more compounds in his/her current research projects to investigate using NMR. Through the investigation, students will learn how to run basic NMR experiments and how to understand the spectra. Students might have to design additional advanced experiments in order to probe more aspects of the molecules. At the end, students should be able to “connect the puzzle pieces” and obtain useful information about their molecules.

Instructor: Dr. Weiguo Hu (Polymer Science & Engineering) **Maximum Class Size:** 6
Dates/Times: July 31 and August 2, from 9:30 AM – 11:30 AM **Location:** LGRT 29
August 6 and 8, from 9:00 AM – 12:00 PM (4 sessions total)

Monoclonal Antibody Production

This lab module will teach students how to make their own monoclonal antibody. The module will combine lectures about basic immunological processes with hands-on demonstration of laboratory techniques. The module will cover immunization, fusion, and observation and testing of resulting hybridomas.

Instructor: Prof. Dominique Alfandari (Veterinary & Animal Sciences) **Maximum Class Size:** 6
Dates/Times: Tentatively: August 9/10 and 23/24 (requires 2 weeks in between sessions) **Location:** TBD

Characterization of Bioconjugates and other Materials by Mass Spectrometry (MS)

This module will introduce students to a variety of mass spectrometry (MS) techniques currently employed for the characterization of biologically relevant materials ranging from small drug molecules to large polymers and bioconjugates. Through a combination of lectures, lab demonstrations and hands-on activities, students will learn some of the myriad MS technologies currently available, and how they can be applied to questions in the field of cellular engineering. A hands-on example of producing a biopharmaceutical by conjugating a protein target with a synthetic polymer to increase its biocompatibility will be combined with purification and subsequent characterization by mass spectrometry. Students will gain a useful insight into the power of MS in the biomaterials arena.

Instructor: Dr. Stephen Eyles (UMass Mass Spectrometry Center) **Maximum Class Size:** 8
Dates/Times: Tentatively: August 13-15, from 9:30 AM – 2 PM **Location:** Conte B-162

ICE IGERT Lab Modules, Summer 2013

Intro to Fermentation and High-Throughput Screening

This lab module aims to introduce students to the concepts of laboratory scale fermentation and high-throughput screening techniques through lectures and hands-on experience. These concepts are often applied in industrial development and production of pharmaceuticals, and students will have the opportunity to gain bench-scale experience in this field using the equipment from the Bioprocess Engineering Laboratory (Chem-Eng 590A). Lab work will consist of preparation of a fermentation reaction and an 8 hour fermentation run, which will allow students to experiment with the interplay between growth and enzyme production through modulation of culture conditions. Demonstrations and discussion will focus on laboratory scale high-throughput screening, automation platforms and considerations for their application in industry.

Instructors: Whitney Stoppel and Lauren Jansen (Chemical Engineering); Prof. Lou Roberts (Biochemistry)
Dates/Times: June 10 (9:30am – Noon), June 11 (9am – 5pm), June 12 (9am – Noon)
Maximum Class Size: 8
Location: ISB

Polymer Analysis and Characterization: Applications for Engineered Biomaterials

This module will introduce students to classical polymer characterization methods with a focus on size and molecular weight determination. Lecture portion will target fundamental concepts relating to polymer solutions (molecular weight distributions, polydispersity, and hydrodynamic size). Several methods (DLS, Static Light Scattering, Turbidity, Viscosity, Zeta Potential, and SEC) will be explained in detail prior to hands-on demonstrations (Light Scattering via Brookhaven Instruments DLS/SLS, and Zeta Potential via Malvern ZS). This module aims to provide introductory knowledge regarding the application of polymer characterization methods to polymeric or colloid based biomaterials, and participation by life scientists is encouraged!

Instructors: Prof. Paul Dubin and Daniel Seeman (Chemistry)
Dates/Times: June 24 (1pm-5 pm), June 25 (9am – 5pm), June 26 (10:30am – 4:30pm)
Maximum Class Size: 10
Location: LGRT 201 (day 1)

Multi-Color Total Internal Reflection Fluorescence and Super-Resolution Imaging

This module will introduce students to the fluorescence microscopy techniques of total internal reflection fluorescence imaging and super-resolution techniques available at UMass, STORM and PALM. The module will utilize the PALM/STORM user facility microscope to visualize live cells via TIRF and fixed cell samples via STORM. Students are encouraged to bring their own samples for either live cell or STORM imaging.

Instructors: Profs. Jennifer Ross (Physics) and Pat Wadsworth (Biology)
Dates/Times: July 15 – 16, 9am – 5 pm
Maximum Class Size: 8
Location: TBD

IPython and the Systems Biology Knowledgebase (KBase)

Python is a popular programming language for scientists and engineers. IPython notebooks have been recently developed for creating and sharing reproducible data analyses using Python, Perl, R and shell. This 4-day module will provide a primer for learning Python using IPython notebooks. Special guest, Dr. Folker Meyer, a computational biologist at Argonne National Laboratory and the Systems Biology Knowledgebase (KBase) Science Lead, will present the final session focusing on the use of IPython notebooks for the analysis of genomic and systems biology data in KBase.

Instructors: Prof. Jeff Blanchard (Microbiology) and Dr. Folker Meyer (Argonne Nat'l Lab)
Dates/Times: August 12-15, 9am – 1 pm
Maximum Class Size: 40
Location: LGRT 201

Signal Multiplexing with the MAGPix Platform

This lab module will introduce students to the xMAP technology used by the Peyton Lab's MAGPIX multiplex system. Instructors will present the principles behind the technology, compare/contrast it to traditional methods (i.e. Western blots, ELISAs), discuss practical application and cost, and describe the process behind making custom xMAP beads. Students will be given samples (or may bring their own) to lyse and prepare for analysis on the MAGPIX instrument; the next day, plates will be read in the MAGPIX instrument and data will be analyzed. At the end of the module, students will understand when it is appropriate to use this technology, how to estimate associated costs, and how to work with it.

Instructors: Prof. Shelly Peyton and Will Herrick (Chemical Engineering)
Dates/Times: August 19 – 21, 9am – 3pm
Maximum Class Size: 10
Location: TBD