



## 1.1.2

### List of Employability/ Entrepreneurship/ Skill Development Courses with Course Contents

Colour Codes		
Name of the Subjects	Yellow	
Employability Contents	Green	
Entrepreneurship Contents	Light Blue	
Skill Development Contents	Pink	



**List of Courses Focus on Employability/ Entrepreneurship/  
Skill Development**

**Department : *Biotechnology***

**Programme Name : *M.Sc.***

**Academic Year : *2020-2021***

***List of Courses Focus on Employability/ Entrepreneurship/Skill Development***

Sr. No.	Course Code	Name of the Course
1.	MBT 103T	Plant and Animal Biotechnology
2.	MBT 104T	Microbiology
3.	MBT 106T	Biostatistics
4.	MBT 107L	Biochemistry and Analytical Techniques
5.	MBT 108L	Microbiology
6.	MBT 109L	Plant and Animal Biotechnology
7.	MBT 201 T	Genetic Engineering
8.	MBT 203T	Bioinformatics
9.	MBT 204T	Genomics and Proteomics
10.	MBT 205T	Molecular Diagnostics
11.	MBT 206T	Research Methodology and Scientific Communication Skills
12.	MBT 207T	Environmental Biotechnology
13.	MBT 208T	BIOLOGICAL IMAGING
14.	MBT 209T	Nanobiotechnology
15.	MBT 301 T	Bioprocess Engineering and Technology
16.	MBT 302T	Emerging Technologies
17.	MBT 304T	Bioentrepreneurship
18.	MBT 305T	Intellectual Property Rights, Biosafety and Bioethics
19.	MBT 306T	Project Proposal Preparation and Presentation



20.	MBT 308T	<b>Microbial Technology</b>
21.	MBT 309 T	<b>Animal Biotechnology</b>
22.	MBT 310 T	<b>Computational Biology</b>
23.	MBT 311 T	<b>Drug Discovery and Development</b>
24.	MBT 312 T	<b>Vaccines</b>
25.	MBT 313 T	<b>Protein Engineering</b>
26.	MBT 314 T	<b>Medical Microbiology and Infection Biology</b>
27.	MBT 316L	<b>Laboratory VI: Bioprocess Engineering and Technology</b>
28.	MBT 317 L	<b>Laboratory VII: Bioinformatics</b>
29.	MBT 401	Dissertation

*@shatt*

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### Syllabus for M.Sc program in Biotechnology 2020-21

M.Sc Biotechnology PG Semester I					
code	Course opted	Subjects	Hours/ semester	Hours/ week	Credits
MBT 101 T	Core -1	Biochemistry	48	03	3
MBT 102T	Core -2	Cell and Molecular Biology	48	03	3
MBT 103T	Core -3	Plant and Animal Biotechnology	48	03	3
MBT 104T	Core -4	Microbiology	32	02	2
MBT 105T	Core-5	Genetics	32	02	2
MBT 106T	Core-6	Biostatistics	48	03	3
		Laboratory			
MBT 107L	Lab 01	Biochemistry and Analytical Techniques	128	08	4
MBT 108L	Lab 02	Microbiology	64	04	2
MBT 109L	Lab 03	Plant and Animal Biotechnology	64	04	2
		Total	512	32	24
M.Sc Biotechnology PG Semester II					
Code	Course opted	Subjects	Hours/ semester	Hours/ week	Credits
MBT 201 T	Core -1	Genetic Engineering	48	03	3
MBT 202T	Core -2	Immunology	48	03	3
MBT 203T	Core -3	Bioinformatics	48	03	3
MBT 204T	Core-4	Genomics and Proteomics	32	02	2
MBT 205T	Core -5	Molecular Diagnostics	32	02	2
MBT 206T	Core -6	Research Methodology and Scientific Communication Skills	32	02	2
MBT 207T	Elective-1	Environmental Biotechnology	32	02	2
MBT 208T	Elective-1	Biological Imaging			
MBT 209T	Elective-1	Nanobiotechnology			
*MBT 210S	Elective	MOOCs course to be selected/opted from SWAYAM portal (SWAYAM-BIOTECH-1)			
		Laboratory			
MBT 211L	Lab 01	Molecular Biology and Genetic Engineering	128	08	4
MBT 212 L	Lab 02	Immunology	96	06	3
		Total	496	31	24
M.Sc Biotechnology PG Semester III					
Code	Course opted	Subjects	Hours/ semester	Hours/ week	Credits





MBT 301 T	Core -1	Bioprocess Engineering and Technology	48	03	3			
MBT 302T	Core -2	Emerging Technologies	32	02	2			
MBT 303T	Core -3	Critical Analysis of Classical Papers	32	02	2			
MBT 304T	Core-4	Bioentrepreneurship	32	02	2			
MBT 305T	Core -5	Intellectual Property Rights, Biosafety and Bioethics	32	02	2			
MBT 306T	Core -6	Project Proposal Preparation and Presentation	32	02	2			
MBT 307T	Core -7	Research Seminar	32	02	2			
MBT 308T	Elective	Microbial Technology	48	03	3			
MBT 309 T	Elective	Animal Biotechnology						
MBT 310 T	Elective	Computational Biology						
MBT 311 T	Elective	Drug Discovery and Development						
MBT 312 T	Elective	Vaccines						
MBT 313 T	Elective	Protein Engineering						
MBT 314 T	Elective	Medical Microbiology and Infection Biology						
*MBT 315T	Elective	MOOCs course to be selected/opted from SWAYAM portal (SWAYAM-BIOTECH-1) Laboratory						
MBT 316L	Lab 01	Laboratory VI: Bioprocess Engineering and Technology				128	08	4
MBT 317 L	Lab 02	Laboratory VII: Bioinformatics				64	04	2
Total			480	30	24			
<b>M.Sc Biotechnology PG Semester IV</b>								
<b>Code</b>	<b>Course opted</b>	<b>Subjects</b>	<b>Hours/ semester</b>	<b>Hours/ week</b>	<b>Credits</b>			
MBT 401	Core -1	Dissertation	512	32	22			
Total			512	32	22			
				Total	94			
Credits								

\*M.Sc. Biotechnology students will select Massive Open Online Course (MOOCs)-SWAYAM course in the II and III semester available at <http://ugcmoocs.inflibnet.ac.in/courses.php> in consultation with Coordinator.



## Plant and Animal Biotechnology

Credits



### Course Objectives

The objectives of this course are to introduce students to the principles, practices and application of animal biotechnology, plant tissue culture, plant and animal genomics, genetic transformation and molecular breeding of plants and animals.

### Student Learning Outcomes

Students should be able to gain fundamental knowledge in animal and plant biotechnology and their applications.

### Unit I

#### Plant tissue culture and animal cell culture 10 lectures

Plant tissue culture: historical perspective; totipotency; organogenesis; Somatic embryogenesis; establishment of cultures – callus culture, cell suspension culture, media preparation – nutrients and plant hormones; sterilization techniques; applications of tissue culture - micropropagation; somaclonal variation; androgenesis and its applications in genetics and plant breeding; germplasm conservation and cryopreservation; synthetic seed production; protoplast culture and somatic hybridization - protoplast isolation; culture and usage; somatic hybridization - methods and applications; cybrids and somatic cell genetics; plant cell cultures for secondary metabolite production.  
Animal cell culture: brief history of animal cell culture; cell culture media and reagents; culture of mammalian cells, tissues and organs; primary culture, secondary culture, continuous cell lines, suspension cultures; application of animal cell culture for virus isolation and *in vitro* testing of drugs, testing of toxicity of environmental pollutants in cell culture, application of cell culture technology in production of human and animal viral vaccines and pharmaceutical proteins.

### Unit II

#### Plant genetic manipulation 10 lectures

Genetic engineering: *Agrobacterium*-plant interaction; virulence; Ti and Ri plasmids; opines and their significance; T-DNA transfer; disarmed Ti plasmid; Genetic transformation - *Agrobacterium*-mediated gene delivery; cointegrate and binary vectors and their utility; direct gene transfer - PEG-mediated, electroporation, particle bombardment and alternative methods; screenable and selectable markers; characterization of transgenics; chloroplast transformation; marker-free methodologies; advanced methodologies - cisgenesis, intragenesis and genome editing; molecular pharming - concept of plants as biofactories, production of industrial enzymes and pharmaceutically important compounds.

### Unit III

#### Animal reproductive biotechnology and vaccinology 8 lectures

Animal reproductive biotechnology: structure of sperms and ovum; cryopreservation of sperms and ova of livestock; artificial insemination; super ovulation, embryo recovery and *in vitro* fertilization; culture of embryos; cryopreservation of embryos; embryo transfer technology; transgenic manipulation of animal embryos; applications of transgenic animal technology; animal cloning - basic concept, cloning for conservation for conservation endangered species; Vaccinology: history of development of vaccines, introduction to the concept of vaccines, conventional methods of animal vaccine production, recombinant approaches to vaccine production, modern vaccines.

### Unit IV

#### Plant and animal genomics 4 lectures

Overview of genomics – definition, complexity and classification; need for genomics level analysis; methods of analyzing genome at various levels – DNA, RNA, protein, metabolites and phenotype; genome projects and bioinformatics resources for genome research – databases; overview of forward and reverse genetics for assigning function for genes.

### Unit V

#### Molecular mapping and marker assisted selection 8 lectures

Molecular markers - hybridization and PCR based markers RFLP, RAPD, STS, SSR, AFLP, SNP markers; DNA fingerprinting-principles and applications; introduction to mapping of genes/QTLs; marker-assisted selection - strategies for introducing genes of biotic and abiotic stress resistance in plants; genetic basis for disease resistance in animals; molecular diagnostics of pathogens in plants and animals; detection of meat adulteration using DNA based methods.





## Microbiology

Credits

2

### Course Objectives

The objectives of this course are to introduce field of microbiology with special emphasis on microbial diversity, morphology, physiology and nutrition; methods for control of microbes and host-microbe interactions.

### Student Learning Outcomes

Students should be able to:

- Identify major categories of microorganisms and analyze their classification, diversity, and ubiquity;
- Identify and demonstrate structural, physiological, genetic similarities and differences of major categories of microorganisms;
- Identify and demonstrate how to control microbial growth;
- Demonstrate and evaluate interactions between microbes, hosts and environment.

### Unit I

#### Microbial characteristics

6 lectures

Introduction to microbiology and microbes, history & scope of microbiology, morphology, structure, growth and nutrition of bacteria, bacterial growth curve, bacterial culture methods; bacterial genetics: mutation and recombination in bacteria, plasmids, transformation, transduction and conjugation; antimicrobial resistance.

### Unit II

#### Microbial diversity

9 lectures

Microbial taxonomy and evolution of diversity, classification of microorganisms, criteria for classification; classification of bacteria; Cyanobacteria, acetic acid bacteria, Pseudomonads, lactic and propionic acid bacteria, endospore forming bacteria,

Mycobacteria and Mycoplasma. Archaea: Halophiles, Methanogens, Hyperthermophilic archae, Thermoplasm; eukarya: algae, fungi, slime molds and protozoa; extremophiles and unculturable microbes.

### Unit III

#### Control of microorganisms

3 lectures

Sterilization, disinfection and antisepsis: physical and chemical methods for control of microorganisms, antibiotics, antiviral and antifungal drugs, biological control of microorganisms.

### Unit IV

#### Virology

5 lectures

Virus and bacteriophages, general properties of viruses, viral structure, taxonomy of virus, viral replication, cultivation and identification of viruses; sub-viral particles – viroids and prions.

### Unit V

#### Host-microbes interaction

5 lectures

Host-pathogen interaction, ecological impact of microbes; symbiosis (Nitrogen fixation and ruminant symbiosis); microbes and nutrient cycles; microbial communication system; bacterial quorum sensing; microbial fuel cells; prebiotics and probiotics.



## Bio-Statistics

Credits



### Course Objectives

The objective of this course is to give conceptual exposure of statistics, error analysis, hypothesis testing, and design of experiments in biological systems

### Student Learning Outcomes

On completion of this course, students should be able to:

- Understand how to summarise statistical data;
- Apply appropriate statistical tests based on an understanding of study question, type of study and type of data;
- Interpret results of statistical tests and application in biological systems.

Unit I

### Introduction

5 lectures

Types of biological data (ordinal scale, nominal scale, continuous and discrete logical systems data), frequency distribution and graphical representations (bar graph, histogram, box plot and frequency polygon), cumulative frequency distribution, populations, samples, simple random, stratified and systematic sampling.

Unit II

### Descriptive statistics, Probability and distribution

10 lectures

Measures of Location, Properties of Arithmetic Mean, median, mode, range, Properties of the Variance and Standard Deviation, Coefficient of Variation, Grouped Data, Graphic Methods, Obtaining Descriptive Statistics on the Computer, Case study. Introduction to probability and laws of probability, Random Events, Events-exhaustive, Mutually exclusive and equally likely (with simple exercises), Definition and properties of binomial distribution, Poisson distribution and normal distribution.

Unit III

### Correlation and regression analysis, Statistical hypothesis

10 lectures

Correlation, Covariance, calculation of covariance and correlation, Correlation coefficient from ungrouped data, Spearson's Rank Correlation Coefficient, scatter and dot diagram, General Concepts of regression, Fitting Regression Lines, regression coefficient, properties of Regression Coefficients, Standard error of estimate. Making assumption, Null and alternate hypothesis, error in hypothesis testing, confidence interval, one-tailed and two-tailed testing, decision making. Making assumption, Null and alternate hypothesis, error in hypothesis testing, confidence interval, one-tailed and two-tailed testing, decision making.

Unit IV

### Tests of significance

8 lectures

Steps in testing statistical significance, selection and computation of test of significance and interpretation of results; Sampling distribution of mean and standard error, Large sample tests (test for an assumed mean and equality of two population means with known S.D.), z-test; Small sample tests (t-test for an assumed mean and equality of means of two populations when sample observations are independent); parametric and Non parametric tests (Mann-Whitney test); paired and unpaired t-test, chi square test.

Unit V

### Experimental designs

Introduction to study designs: Longitudinal, cross-sectional, retrospective and prospective study, Principles of experimental designs, Randomized block, and Simple factorial designs, Analysis of variance (ANOVA) and its use in analysis of RBD, introduction to meta-analysis and systematic reviews, ethics in statistics.





## Laboratory I: Biochemistry & Analytical Techniques

Credits



### Course Objectives

The objective of this laboratory course is to introduce students to experiments in biochemistry. The course is designed to teach students the utility of set of experimental methods in biochemistry in a problem oriented manner.

### Student Learning Outcomes

On completion of this course, students should be able to:

- To elaborate concepts of biochemistry with easy to run experiments;
- To familiarize with basic laboratory instruments and understand the principle of measurements using those instruments with experiments in biochemistry.

1. Preparing various stock solutions and working solutions that will be needed for the course.
2. To prepare an Acetic-Na Acetate Buffer and validate the Henderson-Hasselbach equation.
3. To determine an unknown protein concentration by plotting a standard graph of BSA using UV-Vis Spectrophotometer and validating the Beer-Lambert's Law.
4. Titration of Amino Acids and separation of aliphatic, aromatic and polar amino acids by thin layer chromatography.
5. Purification and characterization of an enzyme from a recombinant source (such as Alkaline Phosphatase or Lactate Dehydrogenase or any enzyme of the institution's choice).
  - a) Preparation of cell-free lysates
  - b) Ammonium Sulfate precipitation
  - c) Ion-exchange Chromatography
  - d) Gel Filtration
  - e) Affinity Chromatography
  - f) Dialysis of the purified protein solution against 60% glycerol as a demonstration of storage method
  - g) Generating a Purification Table (protein concentration, amount of total protein; Computing specific activity of the enzyme preparation at each stage of purification)
  - h) Assessing purity of samples from each step of purification by SDS-PAGE Gel Electrophoresis
  - i) Enzyme Kinetic Parameters:  $K_m$ ,  $V_{max}$  and  $K_{cat}$ .
6. Experimental verification that absorption at  $OD_{260}$  is more for denatured DNA as compared to native double stranded DNA. reversal of the same following DNA renaturation. Kinetics of DNA renaturation as a function of DNA size.
7. Identification of an unknown sample as DNA, RNA or protein using available laboratory tools. (Optional Experiments)
8. Biophysical methods (Circular Dichroism Spectroscopy, Fluorescence Spectroscopy).
9. Determination of mass of small molecules and fragmentation patterns by Mass Spectrometry.
10. Preparing various stock solutions and working solutions that will be needed for the course.





## Laboratory II: Microbiology

Credits



### Course Objectives

The objective of this laboratory course is to provide practical skills on basic microbiological techniques.

### Student Learning Outcomes

Students should be able to:

- Isolate, characterize and identify common bacterial organisms;
- Determine bacterial load of different samples;
- Perform antimicrobial sensitivity tests;
- Preserve bacterial cultures.

## Syllabus

1. Sterilization, disinfection and safety in microbiological laboratory.
2. Preparation of media for cultivation of bacteria.
3. Isolation of bacteria in pure culture by streak plate method.
4. Study of colony and growth characteristics of some common bacteria:  
*Bacillus, E. coli, Staphylococcus, Streptococcus, etc.*
5. Preparation of bacterial smear and Gram's staining.
6. Enumeration of bacteria: standard plate count.
7. Antimicrobial sensitivity test and demonstration of drug resistance.
8. Maintenance of stock cultures: slants, stabs and glycerol stock cultures
9. Determination of phenol co-efficient of antimicrobial agents.
10. Determination of Minimum Inhibitory Concentration (MIC)
11. Isolation and identification of bacteria from soil/water samples.



## Laboratory III: Plant and Animal Biotechnology

Credits



### Course Objectives

The objectives of this course are to provide hands-on training in basic experiments of plant and animal biotechnology.

### Student Learning Outcomes

On completion of course, students should be able to gain basic skills in plant and animal biotechnology.

### Syllabus

#### Plant Biotechnology

1. Prepare culture media with various supplements for plant tissue culture.
2. Prepare explants of *Vallerianawallichii* for inoculation under aseptic conditions.
3. Attempt *in vitro* andro and gyno genesis in plants (*Daturastramonium*).
4. Isolate plant protoplast by enzymatic and mechanical methods and attempt fusion by PEG (available material).
5. Culture *Agrobacterium tumefaciens* and attempt transformation of any dicot species.
6. Generate an RAPD and ISSR profile of *Eremurus persicus* and *Vallerianawallichii*.
7. Prepare karyotypes and study the morphology of somatic chromosomes of *Allium cepa*, *A. sativum*, *A. tuberosum* and compare them on the basis of karyotypes.
8. Pollen mother cell meiosis and recombination index of select species (one achiasmate, and the other chiasmate) and correlate with generation of variation.
9. Undertake plant genomic DNA isolation by CTAB method and its quantitation by visual as well as spectrophotometric methods.
10. Perform PCR amplification of n' number of genotypes of a species for studying the genetic variation among the individuals of a species using random primers.
11. Study genetic fingerprinting profiles of plants and calculate polymorphic information content.



## Semester Two

### Genetic Engineering

Credits

3

#### Course Objectives

The objectives of this course are to teach students with various approaches to conducting genetic engineering and their applications in biological research as well as in biotechnology industries. Genetic engineering is a technology that has been developed based on our fundamental understanding of the principles of molecular biology and this is reflected in the contents of this course.

#### Student Learning Outcomes

Given the impact of genetic engineering in modern society, the students should be endowed with strong theoretical knowledge of this technology. In conjunction with the practicals in molecular biology & genetic engineering, the students should be able to take up biological research as well as placement in the relevant biotech industry.

#### Unit I

#### Introduction and tools for genetic engineering

6 lectures

Impact of genetic engineering in modern society; general requirements for performing a genetic engineering experiment; restriction endonucleases and methylases; DNA ligase, Klenow enzyme, T4 DNA polymerase, polynucleotide kinase, alkaline phosphatase; cohesive and blunt end ligation; linkers; adaptors; homopolymeric tailing; labelling of DNA: nick translation, random priming, radioactive and non-radioactive probes, hybridization techniques: northern, southern, south-western and far-western and colony hybridization, fluorescence *in situ* hybridization.

#### Unit II

#### Different types of vectors

7 lectures

Plasmids; Bacteriophages; M13 mp vectors; PUC19 and Bluescript vectors, hagemids; Lambda vectors; Insertion and Replacement vectors; Cosmids; Artificial chromosome vectors (YACs; BACs); Principles for maximizing gene expression expression vectors; pMal; GST; pET-based vectors; Protein purification; His-tag; GST-tag; MBP-tag *etc.*; Intein-based vectors; Inclusion bodies; methodologies to reduce formation of inclusion bodies; mammalian expression and replicating vectors; Baculovirus and *Pichia* vectors system, plant based vectors, Ti and Ri as vectors, yeast vectors, shuttle vectors.

#### Unit III

#### Different types of PCR techniques

7 lectures

Principles of PCR: primer design; fidelity of thermostable enzymes; DNA polymerases; types of PCR – multiplex, nested; reverse-transcription PCR, real time PCR, touchdown PCR, hot start PCR, colony PCR, asymmetric PCR, cloning of PCR products; T-vectors; proof reading enzymes; PCR based site specific mutagenesis; PCR in molecular diagnostics; viral and bacterial detection; sequencing methods; enzymatic DNA sequencing; chemical sequencing of DNA; automated DNA sequencing; RNA sequencing; chemical synthesis of oligonucleotides; mutation detection: SSCP, DGGE, RFLP.

#### Unit IV

#### Gene manipulation and protein-DNA interaction

7 lectures

Insertion of foreign DNA into host cells; transformation, electroporation, transfection; construction of libraries; isolation of mRNA and total RNA; reverse transcriptase and cDNA synthesis; cDNA and genomic libraries; construction of microarrays – genomic arrays, cDNA arrays and oligo arrays; study of protein-DNA interactions: electrophoretic mobility shift assay; DNase footprinting; methyl interference assay, chromatin immunoprecipitation; protein-protein interactions using yeast two-hybrid system; phage display.

#### Unit V

#### Gene silencing and genome editing technologies

13 lectures

Gene silencing techniques; introduction to siRNA; siRNA technology; Micro RNA; construction of siRNA vectors; principle and application of gene silencing; gene knockouts and gene therapy; creation of transgenic plants; debate over GM crops; introduction to methods of genetic manipulation in different model systems *e.g.* fruit flies

(*Drosophila*), worms (*C. elegans*), frogs (*Xenopus*), fish (zebra fish) and chick; Transgenics - gene replacement; gene targeting; creation of transgenic and knock-out mice; disease model; introduction to genome editing by CRISPR-CAS with specific emphasis on Chinese and American clinical trials.





## Bioinformatics

Credits



### Course Objectives

The objectives of this course are to provide theory and practical experience of the use of common computational tools and databases which facilitate investigation of molecular biology and evolution-related concepts.

### Student Learning Outcomes

Student should be able to :

- Develop an understanding of basic theory of these computational tools;
- Gain working knowledge of these computational tools and methods;
- Appreciate their relevance for investigating specific contemporary biological questions;
- Critically analyse and interpret results of their study.

### Unit I Bioinformatics basics 5 lectures

Bioinformatics basics: Computers in biology and medicine; Introduction to Unix and Linux systems and basic commands; Database concepts; Protein and nucleic acid databases; Structural databases; Biological XML/DTD's; pattern matching algorithm basics; databases and search tools: biological background for sequence analysis; Identification of protein sequence from DNA sequence; searching of databases similar sequence; NCBI; publicly available tools; resources at EBI; resources on web; database mining tools.

### Unit II DNA sequence analysis 5 lectures

DNA sequence analysis: gene bank sequence database; submitting DNA sequences to databases and database searching; sequence alignment; pairwise alignment techniques; motif discovery and gene prediction; local structural variants of DNA, their relevance in molecular level processes, and their identification; assembly of data from genome sequencing.

### Unit III Multiple sequence analysis 5 lectures

Multiple sequence analysis: multiple sequence alignment; flexible sequence similarity searching with the FASTA3 program package; use of CLUSTALW and CLUSTALX for multiple sequence alignment; submitting DNA/protein sequence to databases; where and how to submit, SEQUIN, genome centres; submitting aligned sets of sequences, updating submitted sequences, methods of phylogenetic analysis.

### Unit IV Protein modelling 5 lectures

Protein modelling: introduction; force field methods; energy, buried and exposed residues; side chains and neighbours; fixed regions; hydrogen bonds; mapping properties onto surfaces; fitting monomers; RMS fit of conformers; assigning secondary structures; sequence alignment- methods, evaluation, scoring; protein completion: backbone construction and side chain addition; small peptide methodology; software accessibility; building peptides; protein displays; substructure manipulations, annealing.

### Unit V Protein structure prediction and virtual library 6 lectures

Protein structure prediction: protein folding and model generation; secondary structure prediction; analyzing secondary structures; protein loop searching; loop generating methods; homology modelling: potential applications, description, methodology, homologous sequence identification; align structures, align model sequence; construction of variable and conserved regions; threading techniques; topology fingerprint approach for prediction; evaluation of alternate models; structure prediction on a mystery sequence; structure aided sequence techniques of structure prediction; structural profiles, alignment algorithms, mutation tables, prediction, validation, sequence based methods of structure prediction, prediction using inverse folding, fold prediction; significance analysis, scoring techniques, sequence-sequence scoring; protein function prediction; elements of in silico drug design; Virtual library: Searching PubMed, current content, science citation index and current awareness services, electronic journals, grants and funding information.



## Genomics and Proteomics

Credits

2

### Course Objectives

The objectives of this course is to provide introductory knowledge concerning genomics, proteomics and their applications.

### Student Learning Outcomes

Students should be able to acquire knowledge and understanding of fundamentals of genomics and proteomics, transcriptomics and metabolomics and their applications in various applied areas of biology.

Unit I <b>Basics of genomics and proteomics</b> 3 lectures	Brief overview of prokaryotic and eukaryotic genome organization; extra-chromosomal DNA: bacterial plasmids, mitochondria and chloroplast.
Unit II <b>Genome mapping</b> 4 lectures	Genetic and physical maps; markers for genetic mapping; methods and techniques used for gene mapping, physical mapping, linkage analysis, cytogenetic techniques, FISH technique in gene mapping, somatic cell hybridization, radiation hybrid maps, <i>in situ</i> hybridization, comparative gene mapping.
Unit III <b>Genome sequencing projects</b> 3 lectures	Human Genome Project, genome sequencing projects for microbes, plants and animals, accessing and retrieving genome project information from the web.
Unit IV <b>Comparative genomics</b> 5 lectures	Identification and classification of organisms using molecular markers- 16S rRNA typing/sequencing, SNPs; use of genomes to understand evolution of eukaryotes, track emerging diseases and design new drugs; determining gene location in genome sequence.
Unit V <b>Proteomics</b> 5 lectures	Aims, strategies and challenges in proteomics; proteomics technologies: 2D-PAGE, isoelectric focusing, mass spectrometry, MALDI-TOF, yeast 2-hybrid system, proteome databases.
Unit VI <b>Functional genomics and proteomics</b> 8 lectures	Transcriptome analysis for identification and functional annotation of gene, Contig assembly, chromosome walking and characterization of chromosomes, mining functional genes in genome, gene function- forward and reverse genetics, gene ethics; protein-protein and protein-DNA interactions; protein chips and functional proteomics; clinical and biomedical applications of proteomics; introduction to metabolomics, lipidomics, metagenomics and systems biology.





## Molecular Diagnostics

Credits



### Course Objectives

The objectives of this course are to sensitize students about recent advances in molecular biology and various facets of molecular medicine which has potential to profoundly alter many aspects of modern medicine including pre- or post-natal analysis of genetic diseases and identification of individuals predisposed to disease ranging from common cold to cancer.

### Student Learning Outcomes

Students should be able to understand various facets of molecular procedures and basics of genomics, proteomics and metabolomics that could be employed in early diagnosis and prognosis of human diseases.

### Unit I

#### Genome biology in health and disease

4 lectures

DNA, RNA, Protein: An overview; chromosomal structure & mutations; DNA polymorphism: human identity; clinical variability and genetically determined adverse reactions to drugs.

### Unit II

#### Genome: resolution, detection & analysis

5 lectures

PCR: Real-time; ARMS; Multiplex; ISH; FISH; ISA; RFLP; DHPLC; DGGE; CSCE; SSCP; Nucleic acid sequencing: new generations of automated sequencers; Microarray chips; EST; SAGE; microarray data normalization & analysis; molecular markers: 16S rRNA typing; Diagnostic proteomics: SELDI-TOF-MS; Bioinformatics data acquisition & analysis.

### Unit III

#### Diagnostic metabolomics

2 lectures

Metabolite profile for biomarker detection the body fluids/tissues in various metabolic disorders by making using LCMS & NMR technological platforms.

### Unit IV

#### Detection and identity of microbial diseases

4 lectures

Direct detection and identification of pathogenic-organisms that are slow growing or currently lacking a system of *in vitro* cultivation as well as genotypic markers of microbial resistance to specific antibiotics.

### Unit V

#### Detection of inherited diseases

4 lectures

Exemplified by two inherited diseases for which molecular diagnosis has provided a dramatic improvement of quality of medical care: Fragile X Syndrome: Paradigm of new mutational mechanism of unstable triplet repeats, von-Hippel Lindau disease: recent acquisition in growing number of familial cancersyndromes.

### Unit VI

#### Molecular oncology

5 lectures

Detection of recognized genetic aberrations in clinical samples from cancer patients; types of cancer-causing alterations revealed by next-generation sequencing of clinical isolates; predictive biomarkers for personalized onco-therapy of human diseases such as chronic myeloid leukemia, colon, breast, lung cancer and melanoma as well as matching targeted therapies with patients and preventing toxicity of standard systemic therapies.

### Unit VII

#### Quality assurance and control

1 lecture

Quality oversight; regulations and approved testing.



# Research Methodology and Scientific Communication Skills

Credits



## Course Objectives

The objectives of this course are to give background on history of science, emphasizing methodologies used to do research, use framework of these methodologies for understanding effective lab practices and scientific communication and appreciate scientific ethics.

## Student Learning Outcomes

Students should be able to:

- Understand history and methodology of scientific research, applying these to recent published papers;
- Understand and practice scientific reading, writing and presentations;
- Appreciate scientific ethics through case studies.

Unit I  
**History of science and science methodologies**  
8 lectures

Empirical science; scientific method; manipulative experiments and controls; deductive and inductive reasoning; descriptive science; reductionist vs holistic biology.

Unit II  
**Preparation for research**  
2 lectures

Choosing a mentor, lab and research question; maintaining a lab notebook.

Unit III  
**Process of communication**  
5 lectures

Concept of effective communication- setting clear goals for communication; determining outcomes and results; initiating communication; avoiding breakdowns while communicating; creating value in conversation; barriers to effective communication; non-verbal communication- interpreting non-verbal cues; importance of body language, power of effective listening; recognizing cultural differences; Presentation skills - formal presentation skills; preparing and presenting using over-head projector, PowerPoint; defending interrogation; scientific poster preparation & presentation; participating in group discussions; Computing skills for scientific research - web browsing for information search; search engines and their mechanism of searching; hidden Web and its importance in scientific research; internet as a medium of interaction between scientists; effective email strategy using the right tone and conciseness.

Unit IV  
**Scientific communication**  
9 lectures

Technical writing skills - types of reports; layout of a formal report; scientific writing skills - importance of communicating science; problems while writing a scientific document; plagiarism, software for plagiarism; scientific publication writing; elements of a scientific paper including abstract, introduction, materials & methods, results, discussion, references; drafting titles and framing abstracts; publishing scientific papers - peer review process and problems, recent developments such as open access and non-blind review; plagiarism; characteristics of effective technical communication; scientific presentations; ethical issues; scientific misconduct.





## Environmental Biotechnology

Credits

2

### Course Objectives

This course aims to introduce fundamentals of Environmental Biotechnology. The course will introduce major groups of microorganisms- tools in biotechnology and their most important environmental applications. The environmental applications of biotechnology will be presented in detail and will be supported by examples from the national and international literature.

### Student Learning Outcomes

On completion of course, students will be able to understand use of basic microbiological, molecular and analytical methods, which are extensively used in environmental biotechnology.

### Unit I

#### Introduction to environment

6 lectures

Introduction to environment; pollution and its control; pollution indicators; waste management: domestic, industrial, solid and hazardous wastes; strain improvement; Biodiversity and its conservation; Role of microorganisms in geochemical cycles; microbial energy metabolism, microbial growth kinetics and elementary chemostat theory, relevant microbiological processes, microbial ecology.

### Unit II

#### Bioremediation

6 lectures

Bioremediation: Fundamentals, methods and strategies of application (biostimulation, bioaugmentation) – examples, bioremediation of metals (Cr, As, Se, Hg), radionuclides (U, Te), organic pollutants (PAHs, PCBs, Pesticides, TNT *etc.*), technological aspects of bioremediation (*in situ, ex situ*).

### Unit III

#### Role of microorganisms in bioremediation

6 lectures

Application of bacteria and fungi in bioremediation: White rot fungi vs specialized degrading bacteria: examples, uses and advantages vs disadvantages; Phytoremediation: Fundamentals and description of major methods of application (phytoaccumulation, phytovolatilization, rhizofiltration phytostabilization).

### Unit IV

#### Biotechnology and agriculture

11 lectures

Bioinsecticides: *Bacillus thuringiensis*, Baculoviruses, uses, genetic modifications and aspects of safety in their use; Biofungicides: Description of mode of actions and mechanisms (*e.g. Trichoderma, Pseudomonas fluorescens*); Biofertilizers: Symbiotic systems between plants – microorganisms (nitrogen fixing symbiosis, mycorrhiza fungi symbiosis), Plant growth promoting rhizobacteria (PGPR) – uses, practical aspects and problems in application.

### Unit V

#### Biofuels

11 lectures

Environmental Biotechnology and biofuels: biogas; bioethanol; biodiesel; biohydrogen; Description of the industrial processes involved, microorganisms and biotechnological interventions for optimization of production; Microbiologically enhanced oil recovery (MEOR); Bioleaching of metals; Production of bioplastics; Production of biosurfactants: bioemulsifiers; Paper production: use of xylanases and white rot fungi.



## Bioprocess Engineering & Technology

Credits

3

### Course Objectives

The objectives of this course are to educate students about the fundamental concepts of bioprocess technology and its related applications, thus preparing them to meet the challenges of the new and emerging areas of biotechnology industry.

### Student Learning Outcomes

Students should be able to:

- Appreciate relevance of microorganisms from industrial context;
- Carry out stoichiometric calculations and specify models of their growth;
- Give an account of design and operations of various fermenters;
- Present unit operations together with the fundamental principles for basic methods in production technique for bio-based products;
- Calculate yield and production rates in a biological production process, and also interpret data;
- Calculate the need for oxygen and oxygen transfer;
- Critically analyze any bioprocess from market point of view;
- Give an account of important microbial/enzymatic industrial processes in food and fuel industry.

Abhatt

Bohri 09/11/2020

Unit I <b>Basic principles of biochemical engineering</b> 4 lectures	Isolation, screening and maintenance of industrially important microbes; microbial growth and death kinetics (an example from each group, particularly with reference to industrially useful microorganisms); strain improvement for increased yield and other desirable characteristics.
Unit II <b>Stoichiometry and models of microbial growth</b> 4 lectures	Elemental balance equations; metabolic coupling – ATP and NAD <sup>+</sup> ; yield coefficients; unstructured models of microbial growth; structured models of microbial growth.
Unit III <b>Bioreactor design and analysis</b> 8 lectures	Batch and continuous fermenters; modifying batch and continuous reactors: chemostat with recycle, multistage chemostat systems, fed-batch operations; conventional fermentation v/s biotransformation; immobilized cell systems; large scale animal and plant cell cultivation; fermentation economics; upstream processing: media formulation and optimization; sterilization; aeration, agitation and heat transfer in bioprocess; scale up and scale down; measurement and control of bioprocess parameters.
Unit IV <b>Downstream processing and product recovery</b> 8 lectures	Separation of insoluble products - filtration, centrifugation, sedimentation, flocculation; Cell disruption; separation of soluble products: liquid-liquid extraction, precipitation, chromatographic techniques, reverse osmosis, ultra and micro filtration, electrophoresis; final purification: drying; crystallization; storage and packaging.
Unit V <b>Fermentation economics</b> 4 lectures	Isolation of micro-organisms of potential industrial interest; strain improvement; market analysis; equipment and plant costs; media; sterilization, heating and cooling; aeration and agitation; bath-process cycle times and continuous cultures; recovery costs; water usage and recycling; effluent treatment and disposal.
Unit VI <b>Applications of enzyme technology in food processing</b> 4 lectures	Mechanism of enzyme function and reactions in process techniques; enzymatic bioconversions e.g. starch and sugar conversion processes; high-fructose corn syrup; interesterified fat; hydrolyzed protein etc. and their downstream processing; baking by amylases, deoxygenation and desugaring by glucoses oxidase, beer mashing and chill proofing; cheese making by proteases and various other enzyme catalytic actions in food processing.
Unit VII <b>Applications of microbial technology in food process operations and production, biofuels and biorefinery</b> 4 lectures	Fermented foods and beverages; food ingredients and additives prepared by fermentation and their purification; fermentation as a method of preparing and preserving foods; microbes and their use in pickling, producing colours and flavours, alcoholic beverages and other products; process wastes-whey, molasses, starch substrates and other food wastes for bioconversion to useful products; bacteriocins from lactic acid bacteria – production and applications in food preservation; biofuels and biorefinery





## Emerging Technologies

Credits

2

### Course Objectives

This course is broad-based in nature encompassing several new technologies that current experimental researchers are employing to probe complex system biology questions in life-sciences. The objectives of this course are to teach basics of the new principles to students so as to appreciate current-day research tool-kit better.

### Student Learning Outcomes

Students should be to learn history, theoretical basis and basic understanding of latest technologies in area of biotechnology. They should also be able to learn about various applications of these technologies. The students may also learn one application in depth through an assignment and/or seminar.

### Unit I

#### Optical microscopy methods

8 lectures

**Basic Microscopy:** Light Microscopy: lenses and microscopes, resolution: Rayleigh's Approach, Darkfield; Phase Contrast; Differential Interference Contrast; fluorescence and fluorescence microscopy: what is fluorescence, what makes a molecule fluorescent, fluorescence microscope; optical arrangement, light source; filter sets: excitation filter, dichroic mirror, and barrier, optical layout for image capture; CCD cameras; back illumination, binning; recording color; three CCD elements with dichroic beamsplitters, boosting the signal.

### Unit II

#### Mass spectroscopy

4 lectures

Ionization techniques; mass analyzers/overview MS; FT-ICR and Orbitrap, fragmentation of peptides; proteomics, nano LC-MS; Phosphoproteomics; interaction proteomics, mass spectroscopy in structural biology; imaging mass spectrometry.

### Unit III

#### Systems biology

3 lectures

High throughput screens in cellular systems, target identification, validation of experimental methods to generate the omics data, bioinformatics analyses, mathematical modeling and designing testable predictions.

### Unit IV

#### Structural biology

3 lectures

X-ray diffraction methods, solution & solid-state NMR, cryo-electron microscopy, small-angle X-ray scattering, Atomic force microscopy.

### Unit V

#### CRISPR-CAS

6 lectures

History of its discovery, elucidation of the mechanism including introduction to all the molecular players, development of applications for *in vivo* genome engineering for genetic studies, promise of the technology as a next generation therapeutic method.

### Unit VI

#### Nanobodies

4 lectures

Introduction to nanobodies, combining nanobody with phage-display method for development of antibody against native proteins, nanobody as a tool for protein structure-function studies, use of nanobodies for molecular imaging, catabolic antibodies using nanobodies.





## Bioentrepreneurship

Credits



### Course Objectives

Research and business belong together and both are needed. In a rapidly developing life science industry, there is an urgent need for people who combine business knowledge with the understanding of science & technology. Bio-entrepreneurship, an interdisciplinary course, revolves around the central theme of how to manage and develop life science companies and projects. The objectives of this course are to teach students about concepts of entrepreneurship including identifying a winning business opportunity, gathering funding and launching a business, growing and nurturing the organization and harvesting the rewards.

### Student Learning Outcomes

Students should be able to gain entrepreneurial skills, understand the various operations involved in venture creation, identify scope for entrepreneurship in biosciences and utilize the schemes promoted through knowledge centres and various agencies. The knowledge pertaining to management should also help students to be able to build up a strong network within the industry.

### Unit I

#### Innovation and entrepreneurship in bio-business

8 lectures

Introduction and scope in Bio-entrepreneurship, Types of bio-industries and competitive dynamics between the sub-industries of the bio-sector (e.g. pharmaceuticals vs. Industrial biotech), Strategy and operations of bio-sector firms: Factors shaping opportunities for innovation and entrepreneurship in bio-sectors, and the business implications of those opportunities, Alternatives faced by emerging bio-firms and the relevant tools for strategic decision, Entrepreneurship development programs of public and private agencies (MSME, DBT, BIRAC, Make In India), strategic dimensions of patenting & commercialization strategies.

### Unit II

#### Bio markets - business strategy and marketing

8 lectures

Negotiating the road from lab to the market (strategies and processes of negotiation with financiers, government and regulatory authorities), Pricing strategy, Challenges in marketing in bio business (market conditions & segments; developing distribution channels, the nature, analysis and management of customer needs), Basic contract principles, different types of agreement and contract terms typically found in joint venture and development agreements, Dispute resolution skills.

### Unit III

#### Finance and accounting

8 lectures

Business plan preparation including statutory and legal requirements, Business feasibility study, financial management issues of procurement of capital and management of costs, Collaborations & partnership, Information technology.

### Unit IV

#### Technology management

8 lectures

Technology - assessment, development & upgradation, Managing technology transfer, Quality control & transfer of foreign technologies, Knowledge centers and Technology transfer agencies, Understanding of regulatory compliances and procedures (CDSCO, NBA, GCP, GLA, GMP).



## Intellectual Property Rights, Biosafety and Bioethics

Credits

2

### Course Objectives

The objectives of this course are:

- To provide basic knowledge on intellectual property rights and their implications in biological research and product development;
- To become familiar with India's IPR Policy;
- To learn biosafety and risk assessment of products derived from biotechnology and regulation of such products;
- To become familiar with ethical issues in biological research. This course will focus on consequences of biomedical research technologies such as cloning of whole organisms, genetic modifications, DNA testing.

### Student Learning

**Outcomes On completion of this course, students should be able to:**

- Understand the rationale for and against IPR and especially patents;
- Understand why India has adopted an IPR Policy and be familiar with broad outline of patent regulations;
- Understand different types of intellectual property rights in general and protection of products derived from biotechnology research and issues related to application and obtaining patents;
- Gain knowledge of biosafety and risk assessment of products derived from recombinant DNA research and environmental release of genetically modified organisms, national and international regulations;
- Understand ethical aspects related to biological, biomedical, health care and biotechnology research.

### Unit I Introduction to IPR 5 lectures

Introduction to intellectual property; types of IP: patents, trademarks, copyright & related rights, industrial design, traditional knowledge, geographical indications, protection of new GMOs; International framework for the protection of IP; IP as a factor in R&D; IPs of relevance to biotechnology and few case studies; introduction to history of GATT, WTO, WIPO and TRIPS; plant variety protection and farmers rights act; concept of 'prior art': invention in context of "prior art"; patent databases - country-wise patent searches (USPTO, EPO, India); analysis and report formation.

### Unit II Patenting 5 lectures

Basics of patents: types of patents; Indian Patent Act 1970; recent amendments; WIPO Treaties; Budapest Treaty; Patent Cooperation Treaty (PCT) and implications; procedure for filing a PCT application; role of a Country Patent Office; filing of a patent application; precautions before patenting-disclosure/non-disclosure - patent application-forms and guidelines including those of National Bio-diversity Authority (NBA) and other regulatory bodies, fee structure, time frames; types of patent applications: provisional and complete specifications; PCT and conventional patent applications; international patenting-requirement, procedures and costs; financial assistance for patenting-introduction to existing schemes; publication of patents-gazette of India, status in Europe and US; patent infringement- meaning, scope, litigation, case studies and examples; commercialization of patented innovations; licensing - outright sale, licensing, royalty; patenting by research students and scientists-university/organizational rules in India and abroad, collaborative research - backward and forward IP; benefit/credit sharing among parties/community, commercial (financial) and non-commercial incentives.

### Unit III Biosafety 5 lectures

Biosafety and Biosecurity - introduction; historical background; introduction to biological safety cabinets; primary containment for biohazards; biosafety levels; GRAS organisms, biosafety levels of specific microorganisms; recommended biosafety levels for infectious agents and infected animals; definition of GMOs & LMOs; principles of safety assessment of transgenic plants - sequential steps in risk assessment; concepts of familiarity and substantial equivalence; risk - environmental risk assessment and food and feed safety assessment; problem formulation - protection goals, compilation of relevant information, risk characterization and development of analysis plan; risk assessment of transgenic crops vs cisgenic plants or products derived from RNAi, genome editing tools.

### Unit IV National and international regulations 5 lectures

International regulations - Cartagena protocol, OECD consensus documents and Codex Alimentarius; Indian regulations - EPA act and rules, guidance documents, regulatory framework - RCGM, GEAC, IBSC and other regulatory bodies; Draft bill of Biotechnology Regulatory authority of India - containments - biosafety levels and category of rDNA experiments; field trials - biosafety research trials - standard operating procedures - guidelines of state governments; GM labeling - Food Safety and Standards Authority of India (FSSAI).

### Unit V Bioethics 5 lectures

Introduction, ethical conflicts in biological sciences - interference with nature, bioethics in health care - patient confidentiality, informed consent, euthanasia, artificial reproductive technologies, prenatal diagnosis, genetic screening, gene therapy, transplantation. Bioethics in research - cloning and stem cell research, Human and animal experimentation, animal rights/welfare, Agricultural biotechnology - Genetically engineered food, environmental risk, labeling and public opinion. Sharing benefits and protecting future generations - Protection of environment and biodiversity - biopiracy.





## Project Proposal Preparation & Presentation

Credits



The purpose of this course is to help students organize ideas, material and objectives for their dissertation and to begin development of communication skills and to prepare the students to present their topic of research and explain its importance to their fellow classmates and teachers.

Students should be able to demonstrate the following abilities:

- Formulate a scientific question;
- Present scientific approach to solve the problem;
- Interpret, discuss and communicate scientific results in written form;
- Gain experience in writing a scientific proposal;
- Learn how to present and explain their research findings to the audience effectively.

### Syllabus Project Proposal Preparation

**Selection of research lab and research topic:** Students should first select a lab wherein they would like to pursue their dissertation. The supervisor or senior researchers should be able to help the students to read papers in the areas of interest of the lab and help them select a topic for their project. The topic of the research should be hypothesis driven.

**Review of literature:** Students should engage in systematic and critical review of appropriate and relevant information sources and appropriately apply qualitative and/or quantitative evaluation processes to original data; keeping in mind ethical standards of conduct in the collection and evaluation of data and other resources.

**Writing Research Proposal:** With the help of the senior researchers, students should be able to discuss the research questions, goals, approach, methodology, data collection, *etc.* Students should be able to construct a logical outline for the project including analysis steps and expected outcomes and prepare a complete proposal in scientific proposal format for dissertation.

### Syllabus Poster Presentation

Students will have to present the topic of their project proposal after few months of their selection of the topic. They should be able to explain the novelty and importance of their research topic.

### Syllabus Oral Presentation

At the end of their project, presentation will have to be given by the students to explain work done by them in detail. Along with summarizing their findings they should also be able to discuss the future expected outcome of their work.



## Laboratory VI: Bioprocess Engineering & Technology

Credits



### Course Objectives

The objectives of this laboratory course are to provide hands-on training to students in upstream and downstream unit operations.

### Student Learning Outcomes

Students should be able to:

- Investigate, design and conduct experiments, analyze and interpret data, and apply the laboratory skills to solve complex bioprocess engineering problems;
- Apply skills and knowledge gained will be useful in solving problems typical of bio industries and research.

### Syllabus

- Basic Microbiology techniques
  - Scale up from frozen vial to agar plate to shake flask culture.
  - Instrumentation: Microplate reader, spectrophotometer, microscopy.
  - Isolation of microorganisms from soil samples.
- Experimental set-up
  - Assembly of bioreactor and sterilization.
  - Growth kinetics.
  - Substrate and product inhibitions.
  - Measurement of residual substrates.
- Data Analysis
  - Introduction to Metabolic Flux Analysis (MFA).
- Fermentation
  - Batch.
  - Fed-batch.
  - Continuous.
- Unit operations
  - Microfiltrations: Separation of cells from broth.
  - Bioseparations: Various chromatographic techniques and extractions.
- Bioanalytics
  - Analytical techniques like HPLC, FPLC, GC, GC-MS *etc.* for measurement of amounts of products/substrates.



## Laboratory VII: Bioinformatics

Credits



### Course Objectives

The aim of this course is to provide practical training in bioinformatic methods including accessing major public sequence databases, use of different computational tools to find sequences, analysis of protein and nucleic acid sequences by various software packages.

### Student Learning Outcomes

On completion of this course, students should be able to:

- Describe contents and properties of most important bioinformatics databases;
- Perform text- and sequence-based searches and analyze and discuss results in light of molecular biological knowledge;
- Explain major steps in pairwise and multiple sequence alignment, explain principle and execute pairwise sequence alignment by dynamic programming;
- Predict secondary and tertiary structures of protein sequences.

### Syllabus

1. Using NCBI and Uniprot web resources.
2. Introduction and use of various genome databases.
3. Sequence information resource: Using NCBI, EMBL, Genbank, Entrez, Swissprot/ TrEMBL, UniProt.
4. Similarity searches using tools like BLAST and interpretation of results.
5. Multiple sequence alignment using ClustalW.
6. Phylogenetic analysis of protein and nucleotide sequences.
7. Use of gene prediction methods (GRAIL, Genscan, Glimmer).
8. Using RNA structure prediction tools.
9. Use of various primer designing and restriction site prediction tools.
10. Use of different protein structure prediction databases (PDB, SCOP, CATH).
11. Construction and study of protein structures using Deepview/PyMol.
12. Homology modelling of proteins.
13. Use of tools for mutation and analysis of the energy minimization of protein structures.
14. Use of miRNA prediction, designing and target prediction tools.





## Semester Four

### Dissertation

Credits



(Semester III: 4 Credits;  
Semester IV: 20 Credits)

#### Course Objectives

The objectives of this course are to prepare the students to adapt to the research environment and understand how projects are executed in a research laboratory. It will also enable students to learn practical aspects of research and train students in the art of analysis and thesis writing.

#### Student Learning Outcomes

Students should be able to learn how to select and defend a topic of their research, how to effectively plan, execute, evaluate and discuss their experiments. Students should be able to demonstrate considerable improvement in the following areas:

- In-depth knowledge of the chosen area of research.
- Capability to critically and systematically integrate knowledge to identify issues that must be addressed within framework of specific thesis.
- Competence in research design

*Abhatt*

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and planning.

- Capability to create, analyse and critically evaluate different technical solutions.
- Ability to conduct research independently.
- Ability to perform analytical techniques/experimental methods.
- Project management skills.
- Report writing skills.
- Problem solving skills.
- Communication and interpersonal skills.

Syllabus

#### Planning & performing experiments

Based on the project proposal submitted in earlier semester, students should be able to plan, and engage in, an independent and sustained critical investigation and evaluate a chosen research topic relevant to biological sciences and society. They should be able to systematically identify relevant theory and concepts, relate these to appropriate methodologies and evidence, apply appropriate techniques and draw appropriate conclusions. Senior researchers should be able to train the students such that they can work independently and are able to understand the aim of each experiment performed by them. They should also be able to understand the possible outcomes of each experiment.

Syllabus

#### Thesis writing

At the end of their project, thesis has to be written giving all the details such as aim, methodology, results, discussion and future work related to their project. Students may aim to get their research findings published in a peer-reviewed journal. If the research findings have application-oriented outcomes, the students may file patent application.



## Recommended Electives

### Biological Imaging

Credits



#### Course Objectives

The objectives of this course are to provide complete overview of state-of-art live-cell imaging techniques using microscopes currently available in literature. Live-cell imaging techniques allow real-time examination of almost every aspect of cellular function under normal and experimental conditions. With live-cell imaging experiments, main challenges are to keep cells alive and healthy over a period of time. The growing number of live-cell imaging techniques means one can obtain greater amounts of information without stressing out cells.

#### Student Learning Outcomes

On completion of this course, students shall be able to gain a complete overview of super-resolution field from fundamentals to state-of-art methods and applications in biomedical research. The students shall learn the comparative advantages and disadvantages of each technique, covers all key techniques in field of biomedical science. The students shall also learn how to use new tools to increase resolution in sub-nanometer-scale images of living cells and tissue, which leads to new information about molecules, pathways and dynamics and state-of-the-art examples of applications using microscopes.

capture images and the epi-fluorescence illumination source can be a mercury lamp, xenon lamp, LED's, etc. Each of light sources require carefully matched interference filters for specific excitation and emission wavelengths of your fluorophore of interest. With widefield microscopy, your specimen is only exposed to excitation light for relatively short time periods as the full aperture of emission light is collected by the objectives. Widefield fluorescence microscopy can be used in combination with other common contrast techniques such as phase contrast and differential interference contrast (DIC) microscopy. This combination is useful when performing live-cell imaging to examine general cell morphology or viability while also imaging regions of interest within cells.

#### Unit II Confocal laser scanning microscopy (CLSM) 3 lectures

CLSM has ability to eliminate out-of-focus light and information. It is also possible to obtain optical serial sections from thicker specimens. A conjugate pinhole in optical path of confocal microscope prevents fluorescence from outside of focal plane from being collected by photomultiplier detector or imaged by camera. In CLSM, a single pinhole (and single focused laser spot) is scanned across specimen by scanning system. This spot forms a reflected epi-fluorescence image back on original pinhole. When specimen is in focus, fluorescent light from it passes through pinhole to detector. Any out-of-focus light is defocused at pinhole and very little of this signal passes through to detector meaning that background fluorescence is greatly reduced. The pinhole acts as a spatial filter for emission light from the specimen.

#### Unit III Spinning disc confocal microscopy (SDCM) 2 lectures

This method utilises a "Nipkow Disc" which is a mechanical opaque disc which has a series of thousands of drilled or etched pinholes arranged in a spiral pattern. Each illuminated pinhole on disc is imaged by microscope objective to a diffraction-limited spot on region of interest on specimen. The emission from fluorophores passes back through Nipkow disc pinholes and can be observed and captured by a CCD camera. The effect of spinning disc is that many thousands of points on specimen are simultaneously illuminated. Using SDCM to examine a specimen means that real-time imaging (30-frames-per-second or faster) can be achieved, which is extremely useful if you are looking at dynamic changes within living cells over a wide spectrum of time-scales.

#### Unit IV Light-sheet fluorescence microscopy (LSFM, or SPIM) 2 lectures

This method enables one to perform live-cell imaging on whole embryos, tissues and cells spheroids *in vivo* in a gentle manner with high temporal resolution and in three dimensions. One is able to track cell movement over extended periods of time and follow development of organs and tissues on a cellular level. The next evolution of light-sheet fluorescence microscopy, termed lattice light-sheet microscopy as developed by Eric Betzig (Nobel Prize Laureate 2014 for PALM super-resolution microscopy) will even allow live-cell imaging with super-resolved *in vivo* cellular localization capabilities.

#### Unit V Super-resolved fluorescence microscopy 8 lectures

Super-Resolution in a Standard Microscope: From Fast Fluorescence Imaging to Molecular Diffusion Laws in Live Cells; Photoswitching Fluorophores in Super-Resolution Fluorescence Microscopy; Image Analysis for Single-Molecule Localization Microscopy Deconvolution of Nanoscopic Images; Super-Resolution Fluorescence Microscopy of the Nanoscale Organization in cells; Correlative Live-Cell and Super-Resolution Microscopy and Its Biological Applications; SAX Microscopy and Its Application to Imaging of 3D-Cultured Cells; Quantitative Super-Resolution Microscopy for Cancer Biology and Medicine.





## Computational Biology

Credits



### Course Objectives

The objective of this course is to provide students with theory and practical experience of essentials to aid for genomic, proteomic and metabolomics courses and drug design program.

### Student Learning Outcomes

On completion of this course, the students are expected to:

- Develop an understanding of the basic theory of these computational tools;
- Develop required database extraction, integration, coding for computational tools and methods necessary for all Omics;
- Create hypothesis for investigating specific contemporary biological questions, provide help to experiment with or develop appropriate tools;
- Critically analyze and interpret results of their study with respect to whole systems.

### Unit I

#### Introduction to computational biology basics and biological databases

4 lectures

Computers in biology and medicine; Overview of biological databases, nucleic acid & protein databases, primary, secondary, functional, composite, structural classification database, Sequence formats & storage, Access databases, Extract and create sub databases, limitations of existing databases.

### Unit II

#### Pairwise and multiple sequence alignments

5 lectures

Local alignment, Global alignment, Scoring matrices - PAM, BLOSUM, Gaps and penalties, Dot plots. Dynamic programming approach: Needleman and Wunsch Algorithm, Smith and Waterman Algorithm, Hidden Markov Model: Viterbi Algorithm. Heuristic approach: BLAST, FASTA. Building Profiles, Profile based functional identification.

### Unit III

#### Genome analysis

6 lectures

Polymorphisms in DNA sequence, Introduction to Next Generation Sequencing technologies, Whole Genome Assembly and challenges, Sequencing and analysis of large genomes, Gene prediction, Functional annotation, Comparative genomics, Probabilistic functional gene networks, Human genome project, Genomics and crop improvement. Study available GWAS, ENCODE, HUGO projects, extract and build sub databases; Visualization tools including Artemis and Vista for genome comparison; Functional genomics case studies.

### Unit IV

#### Structure visualization

3 lectures

Retrieving and drawing structures, Macromolecule viewing platforms, Structure validation and correction, Structure optimization, Analysis of ligand-protein interactions; Tools such as PyMol or VMD.

### Unit V

#### Molecular modelling

6 lectures

Significance and need, force field methods, energy, buried and exposed residues; side chains and neighbours; fixed regions; hydrogen bonds; mapping properties onto surfaces; RMS fit of conformers and protein chains, assigning secondary structures; sequence alignment: methods, evaluation, scoring; protein curation: backbone construction and side chain addition; different types of protein chain modelling: ab initio, homology, hybrid, loop; Template recognition and alignments; Modelling parameters and considerations; Model analysis and validation; Model optimization; Substructure manipulations, annealing, protein folding and model generation; loop generating methods; loop analysis; Analysis of active sites using different methods in studying protein-protein interactions.

### Unit VI

#### Structure-based drug development

6 lectures

Molecular docking: Types and principles, Semi-flexible docking, Flexible docking; Ligand and protein preparation, Macromolecule and ligand optimization, Ligand conformations, Clustering, Analysis of docking results and validation with known information. Extra-precision docking platforms, Use of Small-molecule libraries, Natural compound libraries for virtual high throughput screenings.

### Unit VII

#### Ligand-based drug development

6 lectures

Quantitative structure activity relationships; Introduction to chemical descriptors like 2D, 3D and Group-based; Radar plots and contribution plots and Activity predictions, Pharmacophore modeling, Pharmacophore-based screenings of compound library, analysis and experimental validation.





## Drug Discovery and Development

Credits



This course will give a broad overview of research and development carried out in industrial setup towards drug discovery.

On completion of this course, students should be able to understand basics of R&D in drug discovery and should be able to apply knowledge gained in respective fields of pharmaceutical industry.

### Unit I Target identification and molecular modelling 7 lectures

Identification of target or drug leads associated with a particular disease by a number of different techniques including combinations of molecular modeling, combinatorial libraries and high-throughput screening (HTS); Conceptualizing the automation of the HTS process and the importance of bioinformatics and data processing in identification of lead compounds; Rational drug design, based on understanding the three-dimensional

structures and physicochemical properties of drugs and receptors; Modelling drug/receptor interactions with the emphasis on molecular mechanisms, molecular dynamics simulations and homology modelling; Conformational sampling, macromolecular folding, structural bioinformatics, receptor-based and ligand-based design and docking methods, in silico screening of libraries, semi-empirical and ab-initio methods, QSAR methods, molecular diversity, design of combinatorial libraries of drug-like molecules, macromolecular and chemical databases.

### Unit II Lead optimization 5 lectures

Identification of relevant groups on a molecule that interact with a receptor and are responsible for biological activity; Understanding structure activity relationship; Structure modification to increase potency and therapeutic index; Concept of quantitative drug design using Quantitative structure-activity relationship models (QSAR models) based on the fact that the biological properties of a compound are a function of its physicochemical parameters such as solubility, lipophilicity, electronic effects, ionization, stereochemistry, etc.; Bioanalytical assay development in support of *in vitro* and *in vivo* studies (LC/MS/MS, GC/MS and ELISA).

### Unit III Preclinical development 5 lectures

Principles of drug absorption, drug metabolism and distribution - intestinal absorption, metabolic stability, drug-drug interactions, plasma protein binding assays, metabolite profile studies, Principles of toxicology, Experimental design for preclinical and clinical PK/PD/TK studies, Selection of animal model; Regulatory guidelines for preclinical PK/PD/TK studies; Scope of GLP, SOP for conduct of clinical & non clinical testing, control on animal house, report preparation and documentation Integration of non-clinical and preclinical data to aid design of clinical studies.

### Unit IV Drug manufacturing 4 lectures

Requirements of GMP implementation, Documentation of GMP practices, CoA, Regulatory certification of GMP, Quality control and Quality assurance, concept and philosophy of TQM, ICH and ISO 9000; ICH guidelines for Manufacturing, Understanding Impurity Qualification Data, Stability Studies.

### Unit V Clinical trial design 4 lectures

Objectives of Phase I, II, III and IV clinical studies, Clinical study design, enrollment, sites and documentation, Clinical safety studies; Adverse events and adverse drug reactions, Clinical PK, pharmacology, drug-drug interaction studies, Statistical analysis and documentation.

### Unit VI Fundamentals of regulatory affairs and bioethics 4 lectures

Global Regulatory Affairs and different steps involved, Regulatory Objectives, Regulatory Agencies; FDA guidelines on IND and NDA submissions, Studies required for IND and NDA submissions for oncology, HIV, cardiovascular indications, On-label vs. off-label drug use GCP and Requirements of GCP Compliance, Ethical issues and Compliance to current ethical guidelines, Ethical Committees and their set up, Animal Ethical issues and compliance.



## Protein Engineering

Credits

2

### Course Objectives

The aim of this course is to introduce methods and strategies commonly used in protein engineering.

### Student Learning Outcomes

On completion of this course, students should be able to:

- Analyse structure and construction of proteins by computer-based methods;
- Describe structure and classification of proteins;
- Analyse purity and stability of proteins and explain how to store them in best way;
- Explain how proteins can be used for different industrial and academic purposes such as structure determination, organic synthesis and drug design.

### Unit I

#### Introduction to protein engineering 5 lectures

Protein engineering – definition, applications; Features or characteristics of proteins that can be engineered (definition and methods of study) – affinity and specificity; Spectroscopic properties; Stability to changes in parameters as pH, temperature and amino acid sequence, aggregation propensities, etc. Protein engineering with unnatural amino acids and its applications.

### Unit II

#### Stability of protein structure 5 lectures

Methods of measuring stability of a protein; Spectroscopic methods to study physicochemical properties of proteins: far-UV and near-UV CD; Fluorescence; UV absorbance; ORD; Hydrodynamic properties–viscosity, hydrogen-deuterium exchange; Brief introduction to NMR spectroscopy – emphasis on parameters that can be measured/obtained from NMR and their interpretation.

### Unit III

#### Applications 5 lectures

Forces stabilizing proteins – Van der Waals, electrostatic, hydrogen bonding and weakly polar interactions, hydrophobic effects; Entropy – enthalpy compensation; Experimental methods of protein engineering: directed evolution like gene site saturation mutagenesis; Module shuffling; Guided protein recombination, etc., Optimization and high throughput screening methodologies like GigaMetrix, High throughput microplate screens etc., Application to devices with bacteriorhodopsin as an example; Engineering antibody affinity by yeast surface display; Applications to vaccines, Peptidomimetics and its use in drug discovery.

### Unit IV

#### Computational approaches 5 lectures

Computational approaches to protein engineering: sequence and 3D structure analysis, Data mining, Ramachandran map, Mechanism of stabilization of proteins from psychrophiles and thermophiles vis-à-vis those from mesophiles; Protein design, Directed evolution for protein engineering and its potential.

### Unit V

#### Case studies 1 lecture

Case Studies.





## Vaccines

Credits



### Course Objectives

This course will provide students with an overview of current developments in different areas of vaccines.

### Student Learning Outcomes

By the end of this course, students should be able to:

- Understand fundamental concepts of human immune system and basic immunology;
- Differentiate and understand immune responses in relation to infection and vaccination;
- Understand requirement and designing of different types of vaccines;
- Understand importance of conventional and new emerging vaccine technologies.

#### Unit I

### Fundamentals of immune system

6 lectures

Overview of Immune system; Human Immune system: Effectors of immune system; Innate & Adaptive Immunity; Activation of the Innate Immunity; Adaptive Immunity; T and B cells in adaptive immunity; Immune response in infection; Correlates of protection.

#### Unit II

### Immune response to infection

9 lectures

Protective immune response in bacterial; viral and parasitic infections; Primary and Secondary immune responses during infection; Antigen presentation and Role of Antigen presenting cells; Dendritic cells in immune response; Innate immune response; Humoral (antibody mediated) responses; Cell mediated responses: role of CD4+ and CD8+ T cells; Memory responses: Memory and effector T and B cells, Generation and Maintenance of memory T and B cells.

#### Unit III

### Immune response to vaccination

8 lectures

Vaccination and immune response; Adjuvants in Vaccination; Modulation of immune responses: Induction of Th1 and Th2 responses by using appropriate adjuvants and antigen delivery systems - Microbial adjuvants, Liposomal and Microparticles as delivery systems; Chemokines and cytokines; Role of soluble mediators in vaccination; Oral immunization and Mucosal Immunity.

#### Unit IV

### Vaccine types & design

3 lectures

History of vaccines, Conventional vaccines; Bacterial vaccines; Viral Vaccines; Vaccines based on routes of administration: parenteral, oral, mucosal; Live attenuated and inactivated vaccine; Subunit Vaccines and Toxoids; Peptide Vaccine.

#### Unit V

### Vaccine technologies

4 lectures

New Vaccine Technologies; Rationally designed Vaccines; DNA Vaccination; Mucosal vaccination; New approaches for vaccine delivery; Engineering virus vectors for vaccination; Vaccines for targeted delivery (Vaccine Delivery systems); Disease specific vaccine design: Tuberculosis Vaccine; Malaria Vaccine; HIV/AIDS vaccine; New emerging diseases and vaccine needs (Ebola, Zika).



## Medical Microbiology and Infection Biology

Credits



### Course Objectives

This course will provide a perspective and exposure to medical aspects of bacteriology, virology, mycology, parasitology and infectious diseases along with concepts of symptoms, pathogenesis, transmission, prophylaxis and control, a conceptual understanding of host – pathogen interactions using well characterized systems as examples. The student should have a good grasp of disease causing microbes and their interactions with host.

### Student Learning Outcomes

On completion of this course, students should be able to:

- Compare and contrast different microbial diseases, including properties of different types of pathogens, and mechanisms of pathogenesis;
- Summarize role of host in infectious disease, including natural barriers to infection, innate and acquired immune responses to infection, and inflammation;
- Compare and contrast experimental approaches for identifying virulence genes and advantages/disadvantages of each approach for specific pathogens.

### Unit I Bacterial diseases 8 lectures

Normal microflora (microbiome) of human body and its role – Skin, mouth and respiratory tract, intestinal tract, urogenital tract; Pathogenesis and virulence factors - Koch's postulates, Adherence and invasion, Toxins, Enzymes, Antiphagocytic factors, Antigenic heterogeneity, Iron acquisition; *Bacillus anthracis*, *Clostridium* spp., *Corynebacterium diphtheriae*; *E. coli*, *Vibrio cholerae*, *Helicobacter pylori*, *Salmonella typhi* and *paratyphi*, *Shigella dysenteriae*; *Listeria monocytogenes*, *Mycobacterium* spp., Rickettsial diseases; *Haemophilus influenzae*, *Bordetella pertussis*, Brucellosis, Streptococcal and Staphylococcal infections; Antibacterial chemotherapy (with examples of antibiotics) - Inhibition of cell wall synthesis, inhibition of cell membrane function, inhibition of protein and nucleic acid synthesis, antimetabolites; Drug resistance - origin (genetic and non-genetic), mechanisms, antimicrobial activity *in vitro* and *in vivo*, Multi-drug resistance and its mechanisms e.g. MDR-TB.

### Unit II Viral diseases 7 lectures

Viral Pathogenesis - Routes of entry, Viral spread (local and systemic infection), Viral persistence (chronic and latent infection); Polio, Chicken pox, Mumps, Measles, Rubella; Viral hemorrhagic fever, viral encephalitis, Dengue and Yellow fever; Influenza virus infection (emphasis on Avian and swine flu), Rabies and Prion diseases; Hepatitis and Human Cancer viruses; Emerging viral diseases – Ebola, Marburg, SARS, Hanta, Chikungunya, Zika, Chandipura; Antiviral chemotherapy and Viral vaccines; Nucleotide and nucleoside analogs, Reverse transcriptase inhibitor, protease inhibitor, fusion inhibitor etc., Interferons, Killed and attenuated vaccines.

### Unit III Fungal and protozoan infections 7 lectures

Types of Mycoses (with specific example of causative fungi) – Superficial, Cutaneous, Sub-cutaneous; Types of Mycoses (with specific example of causative fungi) – Endemic and Opportunistic; Mycotoxins and Antifungal chemotherapy – Mycetismus, Aflatoxins, classes of currently available drugs and new inhibitors in the pipeline; Protozoan diseases - Giardiasis, Amoebiasis; Leishmaniasis, African sleeping sickness; Malaria, Cryptosporidiosis; Infection by Helminths – Nematodes, Trematodes, Cestodes.

### Unit IV Sexually transmitted diseases and congenital infections 6 lectures

Syphilis and Gonorrhoeal infections; AIDS and Lentiviral infection; Herpes infections; Chlamydial infections (*Chlamydia trachomatis*); Mycoplasma and Ureaplasma infection; Toxoplasmosis; Congenital viral infections – Cytomegalovirus, Varicella zoster, HBV, Enterovirus, Parvovirus B19 etc.

### Unit V Host-pathogen interaction 6 lectures

Intracellular and extracellular pathogens, Principles of microbial pathogenesis, host damage, inflammatory responses, adaptation strategies of pathogen- impact of host and pathogen metabolism on immunity and pathogen survival; Chronic pathogens and mechanisms of persistence; Evasion mechanisms of pathogens; Bacterial – host interaction- *Mycobacterium tuberculosis*, *Borrelia burgdorferi*; Viruses – host interaction: HIV, Influenza; Protozoan – host interaction: *Plasmodium* spp., *Leishmania major*.





## Course: Animal Biotechnology

Course Code: MBT 309T

Course Credit: 3

### Unit I

Introduction to the balanced salt solutions and simple growth medium. Brief discussion on the chemical, physical and metabolic functions of different constituents of culture medium, Serum & protein free defined media and their applications.

### Unit II

Primary and secondary cell culture, Development of cell lines, Biology and characterization of the cultured cells. Basic techniques of mammalian cell cultures in vitro.

### Unit III

Maintenance of cell culture, Cell Passaging, Measuring parameters of growth, Measurement of viability and cytotoxicity.

### Unit IV

Cell synchronization, Cell transformation, Apoptosis, Cryopreservation, Common cell culture contaminants.

### Unit V

Applications of animal cell culture: cell culture based products, vaccines, Hybridoma technology and monoclonal antibodies, stem cells and their applications, Animal cloning, IVF technology, Organ, organotypic and histotypic cultures.

### Suggested Readings

1. Culture of Animal Cells: Freshney
2. Animal Cell Culture: John RW Masters
3. Animal Cell Culture Techniques: Martin Clynes
4. Transgenic Animals: Generation and Use: Louis-Marie Houdebine