M. Biotechnology

Course Curriculum

Department of Biotechnology
All India Institute of Medical Sciences (AIIMS)
New Delhi-110029

M. Biotechnology (2018-2020)
AIIMS, NEW DELHI

Course Curriculum

Course No.	Title	Credits*	Semester	Page #
1	Cell & Developmental Biology (CDB)	2.0	I	3
	Lab 1 - Cell & Developmental Biology	3.0	I	4
2	Genetics (GEN)	2.5	I	5
	Lab 2 - Genetics	3.0	I	7
3	Biochemistry (HBC)	4.0	I	8
	Lab 3 - Biochemistry	1.5	I	9
4	Microbiology	1.0	I	10
7	Medical Microbiology and Infection Biology (MMI)	2.0	I	11
	Lab 4 - Medical Microbiology and Infection Biology	3.5	II	12
5	Immunology & Immunotechnology (IMM)	3.0	I	13
	Lab 5 - Immunology & Immunotechnology	6.5	I, II	14
6	Molecular Biology (MB)	3.0	I, II	15
	Lab 6 - Molecular Biology and Gene Technology	6.5	I	16
8	Genetic Engineering & Gene Technology (GT)	2.0	I, II	17
9	NMR and Structural Biology	2.5	II	19
10	Bioinformatics (BI)	2.5	II	20
	Lab 7 - Bioinformatics	3.5	II	21
11	OMICS: Genomics, Transcriptomics, Proteomics and Metabolomics (OMI)	1.5	II	22
12	Biostatistics (STAT)	2.0	III	24
13	Seminars in Laboratory Techniques: Principles & Instrumentation	1.0	III	25
14	Medical Biotechnology: Journal Club & Communication Skills	2.0	IV	25
15	Seminars in Molecular Medicine & Biotechnology	2.0	IV	25
16	M. Biotech Dissertation	51.0	III, IV	26
	Total: Theory – 33.0, Total Practical –78.5 credits			
	TOTAL CREDITS	**111.5**		

*Theory 1 credit = 14 Lecture hours and Lab 1 credit = 28 laboratory hours.

Course 1 : CELL AND DEVELOPMENTAL BIOLOGY (CDB)
Credits: 2.0

Course Objectives

The major emphasis of this course will be on imparting both basic and translational aspects of Cell and Developmental Biology.

Student Learning Outcomes

Understanding major ideas in cell and developmental biology.Gain an understanding of the basics and applied aspects and how they are correlated and applied to specific clinical problems in cell and developmental biology.

Syllabus

Unit - 1: Introduction and working with Cell
Introduction to Cell Theory and methods of Cell Study; Structure & Function of Bio-membranes; Transport of Ions & Macromolecules.

Unit - 2: Cellular Architecture
Cellular Junctions, Adhesion and Extracellular Matrix; Nucleus Structure & Function; Mitochondria – Structure, Origin & Evolution; Endoplasmic Reticulum, Golgi complex and Lysosomes; Cytoskeletal Structure and Function

Unit - 3: Cellular Physiology
Cell cycle and Check points; Protein Synthesis and Degradation; Vesicular trafficking: Protein Sorting, Transport and Secretion, Endocytosis; Mechanisms of Cell Death: Autophagy, Apoptosis, Necrosis; Signal Transduction Pathways.

Unit - 4: Developmental Biology
Introduction to Development and Pattern formation; Developmental processes in Drosophila; Developmental processes in C. elegans; Overview of Homeotic Genes; Pre- and Post- embryonic development in Humans; Developmental defects and disorders.

Unit - 5: Cells in their Social Context
Basic concepts in Stem Cell Biology; Applications of Stem Cells in Regenerative Medicine; The Developmental Biology of Cancer.

Lab 1 : CELL AND DEVELOPMENTAL BIOLOGY
Credits: 3.0

Course Objectives

The objective of this course is to provide a practical understanding of the cellular organization and developmental biology.

Learning outcomes

The structure of this course is to bring together students from diverse backgrounds and start with basic cell and developmental biology techniques. Basic as well as advanced methods will be taught to study subcellular and developmental process. The students would be able to apply their knowledge and design experiments to demonstrate the increased use of cell developmental biology in therapy.

Syllabus

1. Transmission Electron Microscopy (TEM).
2. Scanning Electron Microscopy (SEM).
3. Immuno-electron Microscopy (IEM).
4. Localization of cellular organelles.
5. Study of sections of testes and ovary of mammals.
6. Study of pre-embryonic developmental stages: Cumulus Oocyte Complex, 2-celled, 4-celled, 8-celled embryo, Morulae and Blastocyst.
7. Observation and study of Sperm parameters (Sperm count, Motility, Viability and Morphology) in physiological saline
8. Window preparation to study development in chick embryo in eggs and study of development of chick embryos from permanent slide mounts

Course 2 : GENETICS (GEN)

Credits: 2.5

Course Objectives

The objectives of this course are to take the students through the basics of genetics encompassing prokaryotic/phage genetics to yeast and higher eukaryotic domains and will cover all concepts of Mendelian genetics. It will also take the students through the basics of human genetics and disease gene mapping.

Student Learning Outcomes

On successful completion of this course, students should be able to: describe the fundamental molecular principles of genetics; fundamentals of bacterial genetics, bacteriophage life cycle and yeast genetics; mechanisms of gene transfer in bacteria; understand the relationship between phenotype and genotype in human genetic traits; describe the basics of genetic mapping; understand how gene expression is regulated; human genetics and approaches to molecular diagnostics of genetic disorders

Syllabus

Unit - 1: Genetics of bacteria and bacteriophages
Genetic complementation and other genetic crosses using phenotypic markers; phenotype to genotype connectivity prior to DNA-based understanding of a gene; Bacterial mutants: nomenclature; classification; recessive/dominant; dominant negative mutation; polar mutation; suppressor tRNA; mutant detection and isolation; conditional mutant; silent mutation; Ames test; Restriction modification system in Bacteria; F factor and conjugation; Transformation; Transduction; Bacteriophages: Nomenclature, structure and assay; Bacteriophages lambda: life cycle and gene regulation; Biology of filamentous phage's; Transposable elements; Genetic analysis of bacteria using reporter technology.

Unit - 2: Yeast genetics
Meiotic crosses, tetrad analyses, non-Mendelian and Mendelian ratios, gene conversion, models of genetic recombination, yeast mating type switch; dominant and recessive genes/mutations, suppressor or modifier screens, complementation groups, transposon mutagenesis, synthetic lethality, genetic epistasis.

Unit - 3: Human genetics
Human genetics- background and history, types of genetic diseases and clinical impact, Genetic variations – mutation, genome polymorphism, diversity, DNA and genetic markers, Mendelian Inheritance, Non-Mendelian inheritance patterns- mitochondrial inheritance, genome imprinting, isodisomy and epigenetics, Genetics of metabolic disorders, Multifactorial inheritance – common diseases and Analysis of qualitative & quantitative traits, Genetic susceptibility in multifactorial disorders.

Unit - 4: Cytogenetics, immunogenetics and developmental genetics
Cytogenetics and karyotype analysis, Molecular cytogenetics approaches such as: FISH, CGH etc, Immunogenetics, Developmental genetics –Genetic mediators of

development, Genetics of Surgical specialties- craniofacial, cardiothoracic, neurosurgical disorders

Unit - 5: Medical Genetics and applications

Traditional and modern approaches of Gene Mapping and Genomics, Haemoglobinopathies, Cancer Genetics, Reproductive Genetics, Prenatal Diagnosis and genetic counselling and ethical issues, Molecular Diagnosis of genetic Disorders, Gene therapy.

Unit - 6: Population Genetics

Evolutionary and Population Genetics, Speciation and Population Divergence, Quantitative Trait loci and Genetic Determinants and population traits.

Lab 2 : GENETICS
Credits: 3.0

Course Objectives

The objective of this course is to provide a practical understanding of the genetic analysis techniques of bacterial and human systems.

Learning outcomes

This course will enable students to acquire basic microbiology and cell culturing principles and understand the basics of fundamental molecular principles of genetics. Students will get a first-hand experience of the horizontal gene transfer methods and bacterial defense systems that will coincide with the basics taught during the theory lectures. They will also obtain an understanding of the relationship between phenotype and genotype in human genetic traits with specific examples of diseases.

Syllabus

1. Bacteriophage titer estimation by plaque assay
2. Study of bacterial Restriction and Modification system
3. To understand the process of bacterial conjugation through transfer of genes and to predict the order of genes on chromosome (Hfr mating technique).
4. Identify mutations in DNA by Sanger sequencing
5. To study the phenomena of genetic linkage.
6. Introduction to molecular cytogenetic techniques: Fluorescence in situ hybridization.
7. Culture of Peripheral blood mononuclear cells and karyotype analysis.

Course 3 : BIOCHEMISTRY (HBC)
Credits: 4.0

Course Objectives

The objectives of this course are to build upon the knowledge of biochemical principles. The course shall make the students aware of various disease pathologies with the context of each topic.

Student Learning Outcomes

On completion of this course, students should be able to gain fundamental knowledge in biochemistry, such as the structure/function of biomolecules, metabolic pathways, bioenergetics, regulation of processes and biochemical techniques. They will be able understand the molecular basis of various pathological conditions from the perspective of biochemical reactions.

Syllabus

Unit - 1: Macromolecules: structure and function
Amino Acids- Classification & metabolism; Carbohydrate-structure, Classification & Properties; Proteins: Structure and Function; Protein Structure and diseases associated with structural derangement; Lipids- structure, Classification & Properties; Haemoglobin and oxygen dissociation curve; Techniques to study receptor-ligand binding; Enzyme kinetics including techniques.

Unit - 2: Metabolism
Carbohydrate Metabolism; Amino Acid Metabolism and Urea cycle; Lipid Metabolism; Nucleotide Metabolism; Integration of Metabolism; Analytical methods in metabolism.

Unit - 3: Bioenergetics
Electron Transport Chain and Mitochondria; Metabolism during exercise, starvation and diabetes; Modern approaches to study Bioenergetics.

Unit - 4: Biochemical techniques
Quality Control in Research; Lab Safety and Etiquette; Biomedical Waste Management; pH, buffer systems and pH meter; Balances and Photometric methods; Microscopy: Fluorescence, Confocal, Electron and Atomic Force Microscopes; Centrifugation: Types of centrifuges; Western blotting, ELISA and IHC; Protein purification and electrophoresis; Chromatography; Evaluation of methods; Spectroscopic techniques including NMR and CD spectroscopy.

Unit - 5: Methods in Clinical Biochemistry
Nutritional Assessment and Monitoring; Vitamins and their deficiencies; Pregnancy: Maternal and Foetal Health Assessment; Tumor markers; Liposomes and drug delivery systems; Therapeutic Drug Monitoring; Inherited Diseases: Clinical Laboratory Assessment; Nucleic Acid Techniques in Diagnostics; Automation in Clinical Chemistry; Radio isotopes and their uses in biochemistry; PET and its applications in research; Approaches to Biomarker Discovery.

Lab 3 : BIOCHEMISTRY
Credits: 1.5

Course Objectives

The objective of this course is to introduce students to experimental methodologies and analytical techniques in Biochemistry.

Learning outcomes

In this module the students will gain proficiency in basic laboratory techniques in biochemistry, such as the Estimation of macromolecules in serum/plasma; Studying the effect of pH, temperature and inhibitors on enzyme kinetics and the Purification of proteins by chromatography.

Syllabus

1. To prepare phosphate buffer and validate Henderson-Hasselbach equation using pH meter.
2. To estimate the concentration of protein in given sample by Bradford's assay.
3. To estimate the concentration of glucose in blood serum using o-Toluidine method.
4. To estimate the concentration of vitamin C in blood plasma using Folin's reagent.
5. To study the effect of various parameters on the kinetics of lactate dehydrogenase enzyme:
    - Temperature
    - pH
    - Inhibitors
6. To learn the basics and practical application of:
    - Gel-filtration chromatography
    - Ion exchange chromatography

Course 4 : MICROBIOLOGY
Credits: 1.0

Course Objectives

This course will provide a perspective and exposure to basic and clinical aspects of bacteriology, virology, mycology and parasitology

Student Learning Outcomes

Students will obtain a perspective and exposure to aspects of infectious diseases and parasitic and fungal infections.

Syllabus

Unit 1: Bacteriology
Introduction to microbiology and its role in medicine, Introduction to bacteriology: classification and structure, Bacterial staining and Cultivation, Common culture media, Common tests for bacterial identification and their principles, Antibiotic susceptibility testing.

Unit 2: Mycology
Introduction to Mycology, structure and classification, laboratory tests and diagnosis of fungal infections.

Unit 3: Virology
Introduction to virology, structure and classification, Laboratory diagnosis of viral infections, viral genetics.

Unit 4: Medical Parasitology
Introduction to parasitology and classification of parasites, laboratory diagnosis of parasitic infections.

Course 7 : MEDICAL MICROBIOLOGY AND INFECTION BIOLOGY (MMI)
Credits: 2.0

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.

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.

Syllabus

Unit - 1: Bacterial infections, Antibiotic action and Drug Resistance

Concepts of microbial sterilization and disinfection, Bacterial growth and measurements, Principles of Microbial Pathogenesis, Molecular basis of infection and pathogenesis of *Bacillus anthracis, Mycobacterium* spp., Mechanism of bacterial persistence and survival, Immunological response of Mycobacterial infection, Antibiotic action and resistance mechanisms, Drug resistance - origin (genetic and non-genetic), mechanisms.

Unit - 2: Viral Infections

Molecular basis of viral infections, pathogenesis and therapeutics: molecular basis of Dengue infection and pathogenesis, molecular biology of Hepatitis infection and virulence, molecular Biology of HIV Infection, Anti-viral chemotherapy and viral vaccines.

Unit – 3: Fungal and protozoan infections

Protozoan diseases: Molecular basis of Leishmaniasis and Malaria, invasion and evasion mechanism, diagnosis and treatment, Strategies for malaria control, Molecular basis of fungal infections, therapeutics and control.

Unit – 4: Basis of various diseases; Zoonotic infections and Molecular basis of their pathogenesis, Plant diseases, Microbiome and infectious diseases.

Lab 4 : MEDICAL MICROBIOLOGY AND INFECTION BIOLOGY Credits: 3.5

Course Objectives

The aim of introducing the practicals of Medical Microbiology and Infection Biology is to acquaint the students with various microbiological techniques. This course is intended to provide hands on training and make students of diverse backgrounds proficient in microbiological techniques and application of these techniques to study aspects of microbial growth and antibiotic susceptibility.

Learning outcomes

It is expected that by the end of this course, students will have good understanding of different aspects of microbial physiology, and should be able to safely handle different bacterial species, differentiate those species on the basis of gram phenotype, and understand the basis of growth, different factors impacting microbial growth and should be able to score for anti-microbial susceptibility and drug resistance in bacteria.

Syllabus

1. Introduction to the basic techniques of microbiology and safe handling of bacterial cultures (Gram positive, Gram Negative, Acid-fast bacteria), isolate pure cultures from a mix population using selective media and determine their purity using different staining techniques (Gram staining and Acid-fast staining).
2. Understanding the factors impacting bacterial growth: As different bacteria have different growth requirements, the students will be taught
    - Concept of minimal media
    - The effect of different carbon sources such as glucose, acetate, and lactose
    - The effect of different minerals such as copper and iron
3. Drug susceptibility testing of bacterial strains using different methods
    - Disc-diffusion method
    - Redox activity-based assays using REMA (resazurin) assay.
    - CFU.
4. Visit to Medical Microbiology laboratories at AIIMS with an emphasis on:
    - Slide and microscopic analysis of various microbes.
    - Understanding the throughput machines and auto-analyzers used for clinical microbiology diagnosis.

Course 5 : IMMUNOLOGY & IMMUNOTECHNOLOGY (IMM)
Credits: 3.0

Course Objectives

The objectives of this course are to learn about structural features of components of immune system as well as their function. The major emphasis of this course is on the study of development of immune system and mechanisms by which human body elicits immune response.

Student Learning Outcomes

On completion of this course, students should be able to: apply their knowledge and design experiments to study innate, humoral or cytotoxic T lymphocyte responses.

Syllabus

Unit - 1: Components of the Immune System
Cardinal Principles of Immunity; Cells, Tissues and Organs of the Immune system; Evolution of Immune System; Innate Immunity; Humoral Immunity; Biology of: Macrophages; Dendritic Cells; NK Cells; Antigens, Antigenicity and Immunogenicity; Antibody: Structure and Function; Antigen-Antibody Interaction ;Complement Biology

Unit - 2: Modes of Immune Response
T cell development; T cell Receptors and activation; T cell differentiation and effector functions; B cell development; B cell receptor; VDJ recombination and Class switching; Immunoglobulin gene rearrangement and expression; Biology of Chemokines and Cytokine, receptors & signalling; Cell migration and homing; MHC molecules and MHC gene organization; MHC gene expression and regulation; Antigen processing and presentation: MHC-I and MHC-II; Cell and antibody mediated cytotoxicity.

Unit - 3: Defence against Infectious Agents
Immunity to Viruses; Immunity to Bacteria; Immunity to Protozoans; Immunity to Worms

Unit - 4: Immunomodulation
Primary and Secondary Immunodeficiencies; Immunosuppression; Hypersensitivity type I, II, III and IV; Vaccine Biology; Immunological tolerance; Mucosal Immunity

Unit - 5: Immune Responses against Tissues
Autoimmunity and Diseases; Transplantation and Rejection; Immunity to Cancers

Unit - 6: Immunotechnology
Flow Cytometry: Principles and Applications; Monoclonal antibodies & Hybridoma Technology; Applications of Monoclonal Antibodies; New generation Antibodies; Immunoassays - ELISA, ELISPOT; Assay Precision and Accuracy; Preparation of Radioactive Tracers; Electro chemiluminometric assay; Vaccine Biology.

Lab 5 : IMMUNOLOGY & IMMUNOTECHNOLOGY
Credits: 6.5

Course Objectives

The objective of this course is to develop a practical understanding of the components and functioning immune system, as well as introduce the students to immunological techniques.

Learning outcomes

This practical module is designed to provide students training in basic to advanced immunology techniques routinely applicable in clinical, laboratory or industrial settings. The primary objective of this course is to help students develop the necessary skills to experimentally evaluate innate, humoral or cell mediated immune responses. The course will enable students to independently design and establish assays, using peripheral blood cells, cell lines or mouse-derived cells. This training will enable students to handle and dissect mice, prepare organ single cell suspension, establish cellular assays and apply immunological techniques. In addition to experiment implementation, the students will acquire skills for the interpretation of cell-assays and flowcytometry data and will be able to evaluate the usefulness of immunology in different clinical settings.

Syllabus

1. To estimate the total and differential leukocyte counts in peripheral blood.
2. To learn the technique of Ficoll^{TM} density gradient centrifugation for isolation of peripheral blood mononuclear cells (PBMCs).
3. To study the proportion of T cells in PBMCs by Immunofluorescence assay.
4. To learn the techniques of cell culture: cell revival, passage and cryopreservation.
5. To assess the viability of cells by MTT assay
6. To study the phenomena of phagocytosis in macrophages using fluorescent latex beads.
7. To study the phenomena of apoptosis by flowcytometry
8. Demonstration of handling of mouse, dissection (identify organs of immune system), study of routes of antigen administration and routes of blood withdrawal.
9. To isolate and culture mouse splenocytes and study the frequency of various T cell subpopulations (T_H1, T_H2, T_H17, T_{reg}).
10. To study T cell proliferation and cytokine secretion following *in vitro* stimulation of mouse splenocytes.
11. To study the different forms of enzyme-linked immunosorbent assay (ELISA)
12. To learn the technique of single radial immunodiffusion assay (RIA).

Course 6 : MOLECULAR BIOLOGY (MB)
Credits: 3.0

Course Objectives

The aim of this course is to obtain and understand fundamental knowledge of various concepts of biological processes at the molecular level.

Student Learning Outcomes

After completion of course, students should be able to describe and explain molecular processes and principles of DNA replication, Transcription and Tanslation in both prokaryotes and eukaryotes.

Syllabus

Unit - 1: DNA Structure and Genome organization
DNA structure and function: Central dogma, DNA as genetic material, DNA supercoiling, gyrases, topoisomerases; Physical properties of nucleic acids: Chromatin structure; Chromatin remodelling and its functional significance.

Unit - 2: DNA Replication repair and Recombination
DNA replication: Mechanism of Prokaryotic and Eukaryotic DNA replication; DNA damaging agents; DNA repair - Components and pathways; DNA recombination- Components and pathways.

Unit – 3: RNA structure and function
Structure and function: mRNA, tRNA and rRNA.

Unit – 4: RNA transcription, processing and regulation in prokaryotes
Transcription in prokaryotes; Processing and degradation of bacterial RNA; Prokaryotic gene regulation: Lac operon, Attenuation and antitermination, DNA recombination, small RNAs, riboswitch, Control of ribosome, rRNA and r-protein synthesis.

Unit – 5: RNA transcription, processing and regulation in Eukaryotes
Interaction of transcription factors with DNA; Families of DNA binding transcription factors: Role of histone modifications and chromatin remodeling in regulation of transcription; Eukaryotic transcription; Post-transcriptional modification: mRNA capping, polyadenylation and regulation; pre-mRNA splicing and regulation; mRNA surveillance mechanisms; tRNA processing; Catalytic RNAs; Non-coding RNA: LncRNAs, circRNAs, piRNAs etc; RNA interference.

Unit - 6: Protein translation, posttranslational modification
Genetic code, protein biosynthesis; Components of Translation; Mechanism of Prokaryotic and Eukaryotic Translation; Post-translational modifications of proteins.

Lab 6 : MOLECULAR BIOLOGY & GENE TECHNOLOGY
Credits: 6.5

Course Objectives

The objective of this course is to provide a practical understanding of the molecular biology and genetic engineering techniques to students.

Learning outcomes

In this module the students will gain proficiency in basic techniques that are employed in any molecular biology and genetic engineering laboratory or industry. The students will gain hands-on experience in gene manipulation and cloning, protein expression and purification, and evaluation of gene expression and regulation. The students will be able to independently design and establish assays and communicate the results with scientific rigor. Above all, this module will boost the overall student confidence and enable them to get good positions in reputed institutes in India and abroad in future.

Syllabus

1. To isolate and purify high molecular weight genomic DNA from *E. coli*.
2. To learn the technique of TA-cloning
   - To learn primer designing and amplify the gene of interest by PCR
   - To prepare the competent cells of *E. coli* and estimate their viability and transformation efficiency.
   - To A-tail and ligate the DNA of gene of interest in pGEM®-T easy vector
   - Transformation of *E. coli*
   - Screening of recombinant colonies by Blue/white screening and colony PCR
   - Isolation of recombinant plasmid DNA by alkaline lysis method.
3. To induce the expression of recombinant protein in *E. coli*
4. To analyze the expression level of induced protein by SDS PAGE and Western blot techniques.
5. Purification of His_6-tagged protein by non-denaturing Ni-NTA affinity chromatography and demonstration of FPLC.
6. Estimation of Limit of Detection (LOD) of aptamers by ALISA.
7. To isolate RNA from mammalian cells and analyze gene expression by qRT-PCR.
8. Reporter assay:
   - Assessment of *lacZ* promoter activity by β-galactosidase reporter assay.
   - Demonstration of GFP and luciferase reporters

Course 8 : GENETIC ENGINEERING AND GENE TECHNOLOGY (GT)
Credits: 2.0

Course Objectives

The objective of the course is to familiarize the students with the basic concepts in genetic engineering, with versatile tools and techniques employed in genetic engineering and recombinant DNA technology and about applications of genetic engineering in the fields of therapeutics, diagnosis and medicine.

Student Learning Outcomes

The students will have knowledge of tools and strategies used in genetic engineering. They will obtain an understanding of the applications of recombinant DNA technology and genetic engineering from medical perspective. The students can then use and apply the knowledge of genetic engineering in problem solving and in practice.

Syllabus

Unit -1: Introduction and tools for genetic engineering
Methodologies, enzymes and small nucleic acid molecules in genome engineering: restriction endonucleases and methylases; DNA ligase, Klenow enzyme, T4 DNA polymerase, polynucleotide kinase, alkaline phosphatase; cohesive and blunt end ligation; linkers; adaptors; homopolymer tailing; labelling of DNA: nick translation, random priming.

Unit - 2: Gene cloning and expression systems
Bacterial plasmids and expression systems; Bacteriophages: M13mp vectors; pUC19 and pBluescript vectors, phagemids, Lambda vectors; Insertion and Replacement vectors; Cosmids; Artificial chromosome vectors (YACs; BACs); Principles for maximizing gene expression vectors; pMal; GST; pET-based vectors; Protein purification methods including inclusion bodies and methodologies to reduce formation of inclusion bodies; Eukaryotic and viral vector systems. Advanced cloning methods: Gateway, Golden gateway and Gibson cloning.

Unit - 3: DNA and cDNA analysis
Introduction of foreign DNA into host cells; transformation, electroporation, transfection; construction of genomic and cDNA libraries, phage display; strategies for library screening; radioactive and non-radioactive probes; hybridization techniques: Northern, Southern, South-western and Far-western and colony hybridization, fluorescence in situ hybridization.

Unit - 4: Gene silencing and genome editing technologies
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; Transgenics - gene replacement; gene targeting; creation of transgenic and knock-out mice; disease model; introduction to genome editing: zinc finger nucleases, Meganucleases, TALEN; CRISPR/Cas with specific emphasis on Chinese and American clinical trials;

Applications of CRISPR/cas9 technology. Applications gene therapy/gene editing-antiviral strategies, cancer immunotherapy, hematologic disorders, liver-targeted gene editing; nucleic-acid therapeutics; Methods of epigenetic analysis: diagnostic and therapeutic applications.

Unit 5: Study of interaction of bio-molecules

Methods to study DNA/ RNA-protein interactions, biochemical methods to study protein-protein interactions.

Course 9 : NMR AND STRUCTURAL BIOLOGY
Credits: 2.5

Course Objectives

The course is designed to provide a broad exposure to all basic techniques (Biochemical & Biophysical) used in current Modern Biology research. The goal is to impart basic conceptual understanding of principles of these techniques and emphasize on their utility.

Student Learning Outcomes

On completion of this course, students should have the knowledge and clear understanding of various biophysical techniques and their applications in biomedical research.

Syllabus

Unit-1: Nucleic acids and Protein structure
Introduction to DNA-Structure: historic and current view; Polymorphism of DNA; Introduction to Circular Dichroism; Introduction to protein structure: Chemistry of building blocks (amino acids); Peptide bonds, conformation and dihedral angles; hydrophilicity, hydrophobicity and amphipathicity in proteins; Various structural organization in proteins: Secondary and Tertiary structure (with example of a globular protein); Homologous sequences and structures; Structural features of protein-DNA and drug-DNA complexes.

Unit-2: Basic principles, instrumentation and applications of NMR
Basic principles of NMR - Classical picture, resonance condition, etc; concept of spin, quantum description, etc; Pulse Fourier NMR & relaxation phenomena; Chemical shift and coupling constants; Basic principles of 2D NMR; Analysis of NMR spectra, proton decoupling, NOE, etc; 2D NMR methods like COSY, TCOSY, J-resolved, etc; Proton MR of amino acids and peptides; NMR of other nuclei, editing etc; Other applications of NMR to study of biomolecules; In vivo NMR spectroscopy and metabolomics; Practical Demonstration in NMR; Magnetic Resonance Imaging – MRI.

Unit-3: Chromatography, Mass spectroscopy and spectrophotometry
Introduction to HPLC; Introduction to Tandem Mass Spectroscopy: QQQ-LC/MS. Basic principles, instrumentation and applications of UV-visible, IR, fluorimetry, atomic absorption and emission spectrophotometry: US-VIS Absorption Spectroscopy; Fluorescence Spectroscopy; Infrared spectroscopy and application to biomolecular studies; Infrared imaging and role in diagnosis; Atomic Absorption Spectroscopy in biomedical research.

Unit-5: Protein engineering
An Introduction to existing methods for *in silico* protein engineering.

Course 10 : BIOINFORMATICS (BI)
Credits: 2.5

Course Objectives

This course covers all basic aspects of Bioinformatics starting from sequence comparison tools to genome annotation to protein structure prediction methods. The course also touches upon *in silico* methods of biological networks to drug designs, thus giving a comprehensive understanding of the most of the bioinformatics and computational analyses.

Student Learning Outcomes

On completion of this course, students should be able to: develop an understanding of the basic theory and working knowledge of computational tools; able to understand in depth and ask question regarding contemporary biology; critically analyse and interpret the outcome of their study.

Syllabus

Unit - 1: Bioinformatics Basics
Introduction to Bioinformatics; Web Sites, Servers and Databases, Database Architecture, Introduction to SQL, Nucleic Acid Sequence Databases, Introduction to Genomics, Human Genes & Genomes, Genomic Databases

Unit - 2: Multiple sequence alignment tools
Pairwise Sequence Comparison and Alignment, Hidden Markov Models etc., Taxonomy and Systematics, Multiple Sequence Alignment, Introduction to Molecular Phylogenetics: Alignment and alignment editing, Deciding on a data model, Tree building and evaluation

Unit - 3: Computational biology in drug designing
Quantitative Structure Activity Relationship (QSAR), Introduction to Systems Biology, Preclinical drug development strategies and official requirements, Methods for Drug Target Identification: Application of Hidden Markov Models, Virtual Screening for drug discovery, Clinical benefits of analyzing 3D structure of proteins-basis of disease and therapy.

Unit-4: Protein structure prediction and modelling
Predictive methods using Protein Sequences. Predicting 2D and 3D structures, My Pattern Finder/Spectral Repeat Finder, MoPP (Motif Prediction Program), Gene Prediction Methods, Protein Sequence Motifs and Patterns /Protein Structure Databases.

Unit-5: Functional genomics and transcriptomics
Functional Genomics: High throughput Biology and Microarrays, Analysis of Microarray Data, Sequence Polymorphisms, Promoter Analysis: Characterization and Prediction; Sequence Assembly and Finishing methods.

Lab 7: BIOINFORMATICS
Credits: 3.5

Course Objectives

The aim of this practical course is to provide hands on training to students in bioinformatics methods and acquaint them with various analytical tools available to a biologist handling high-throughput data.

Learning outcomes

This course will equip students to apply computational tools and retrieve data from online repositories to answer fundamental biological questions. The course aims at providing training in accessing major public databases, sequence alignment tools, protein modelling, analysis of big data and learning basics of programming languages. It is envisioned that this *in silico* module will enable students to undertake advanced research and develop skills necessary for large-scale data management and analysis, preparing them for addressing complex issues in systems biology.

Syllabus

1. To understand working with biological databases with references to NCBI, INDSC and Expasy.
2. To utilize primer designing tools and perform *in silico* PCR.
3. To perform pair-wise and multiple sequence alignments using online tools such as BLAST, CLUSTALW etc.
4. To learn the basics of mass spectrometry with practical demonstration.
5. To learn how to display and manipulate protein structures using online softwares.
6. To study protein ligand interactions and docking using Glide, Autodock
7. To predict tertiary protein structures from primary sequence.
8. To perform homology modeling of proteins
9. Introduction to MS access and Linux
10. To learn the computer languages:
    - R-package
    - SQL
    - Perl
11. To work with Gene prediction software.
12. To analyze and identify single nucleotide polymorphisms.
13. To learn the analysis of microarray data.

Course 11 : OMICS - Genomics, Transcriptomics, Proteomics Metabolomics (OMI)
Credits: 1.5

Course Objectives

The objective of this course is to give an introduction to global Omics technologies, theory and practical aspects and its applications in biology. The student should be able to gain working knowledge of these technologies and appreciate their ability to impart a global understanding of biological systems and processes in health and disease.

Student Learning Outcomes

On completion of this course, students will have an understanding the basis of Genome sequencing approaches and assembly strategies; understand how high-throughput sequencing (HST) technologies can be used to explore changes in gene expression, detect genome variation in population; identify disease causing genetic variants; understand how Genome-wide association study (GWAS) can detect disease associated markers in multifactorial diseases; understand the concepts in Pharmogenomics and Personalized medicine.

Syllabus

Unit – 1: Introduction to Genomics and Sequencing technologies
Sequencing Technologies: Overview, methods and applications: Maxam and Gilbert method, Sanger Sequencing techniques and applications; Introduction to Next Generation sequencing (NGS): NGS platform overview and comparison; types of NGS (Whole genome sequencing, ChIP sequencing, transcriptome sequencing, small RNA and non-coding RNA sequencing.

Unit – 2: Whole Genome Sequencing: Approaches, Strategies and Applications
Whole genome sequencing approaches for prokaryotes and eukaryotic organisms, Construction of libraries (Plasmid based and BAC libraries), Shotgun sequencing, Assembly and Data analysis, concepts of contig and scaffolds; Comparative genomics and pathogenomics, metagenomic approaches, analysis of human microbiome; methods, analysis and applications including pharmogenomics and personalized medicine.

Unit – 3: Microarray technology and Transcriptome Analysis
Introduction, Basic principles and design, cDNA and oligonucleotide arrays, DNA microarray, protein array, comparative Genomic Hybridization (CGH) arrays, Resequencing arrays; Instrumentation; Designing a microarray experiment, Data Processing and Normalization; databases – NCBI-GEO, ArrayExpres; Whole transcriptome sequencing; Data Processing and Analysis, Genome assembly and mapping to reference genomes using mapping tools (bowtie, maq etc), Sequence Alignment formats, Functional Analysis and enrichment tools; Pathway analysis; Application of different sequencing techniques, methylomics, *in vivo* protein binding, genome wide association studies (GWAS), Applications for human health and

diseases, Comparison of Microarray technology and RNA sequencing technologies, case studies.

Unit – 4: Proteomics and Metabolomics

Metabolomics : preclinical and Clinical: Basics of Metabolomics, Tools for studying Metabolomics, NMR and LC-MS Based metabolomics, Metabolomics in Biomarker Discovery, Flux Analysis, Metabolomics in Drug profiling and Drug Target Identification, Metabolic databases, Genomic basis of Construction and analysis of Metabolic Pathways. Basics of Proteomics, Proteomic Profiling and fingerprinting for precision medicine, therapeutic drug re-positioning using personalized proteomics.

Course 12 : Biostatistics (STAT)

Credits: 2.0

Course Objectives

The objective of this course is to give an overview of basics of statistics and its application in biomedical research.

Student Learning Outcomes

On completion of this course, students will be able to collect, analyse and infer from various research and clinical data. They will gain working knowledge of various analysis tools and software's for the same.

Syllabus

Unit – 1: Introduction, Central Tendency & Dispersion
Introduction to Biostatistics – scope & need for the application of statistical methods in medical and biological. Presentation of data – statistical tables, diagrams and graphs. Needs for reduction of data – measures of average and location. Measures of dispersion –Range, quartile deviation, mean deviation and standard deviation, coefficient of variation.

Unit – 2: Sample Size and Sampling Methods
Concepts of statistical population and sample, sample size. Methods of sampling. KAP studies: sample size.

Unit – 3: Concept of Probability & Distribution
Probability: Basics concepts and theorems of probability – conditional probability – Baye's theorem. Binomial distribution, Poisson distribution and their application. Statistical methods in Diagnostic Test studies. Normal distribution and its application.

Unit – 4: Statistical Inference
Logic of statistical inference, standard error, estimation and testing the statistical significance. Test of significance: Normal deviate test (Z test); Student's t tests; Chi-Squared tests. F – Test and one way analysis of variance, multiple range tests. Non – Parametric statistical methods of hypothesis testing.

Unit – 5: Correlation, Regression, Data Management and Analysis using SPSS
Correlation and Regression- definition and application. Questionnaire development and data management. Data Analysis using SPSS.

Course 13 : Seminars in Laboratory Techniques - Principles and Instrumentation
Credits: 1.0

Course Objectives

To provide a clear understanding of various modern techniques through student seminars.

Student Learning Outcomes

At the end of the seminars, the students should have a thorough understanding of the principles and instrumentation used in various biomedical researches.

Course 14 : Medical Biotechnology - Journal Club and Communication Skills
Credits: 2.0

Course Objectives

To provide training in the critical analysis of publications in emerging areas of Medical Biotechnology.

Student Learning Outcomes

At the end of the seminar presentations, the students will acquire the skills to critically analyse, interpret and present biomedical research data.

Course 15 : Seminars in Molecular Medicine & Biotechnology
Credits: 2.0

Course Objectives

To provide students with an exposure to ongoing research activities in Molecular Medicine and Biotechnology through invited lectures by experts.

Student Learning Outcomes

The students will learn how to identify, develop and execute a research programme that is in sync with healthcare priorities of India.

Course 16 : M. Biotech Dissertation
Credits: 51.0

Course Objectives

To provide students with a first-hand experience of identifying, developing and executing a small research project in Medical Biotechnology.

Student Learning Outcomes

The students will learn all the essential steps required to implement a research project: review of literature, lacunae in field, hypothesis development, preparation and submission of ethics documents (human, animal, recombinant DNA/ biosafety), experiment design and execution, data analysis, compilation and presentation, documentation and preparation of dissertation.