



ANDHRA KESARI UNIVERSITY ::ONGOLE

**Model Syllabus for 4-Year UG Honours in B.Sc. (Biotechnology) as Major
in consonance with Curriculum framework w.e.f. AY 2025-26**

COURSE STRUCTURE (for Semester I to VI)

Year	Semester	Course	Title of the Course	No. of Hrs /Week	No. of Credits
I	I	1	Introduction to Cell Biology and Genetics	3	3
			Introduction to Cell Biology and Genetics-Practical	2	1
		2	Biological Chemistry	3	3
			Biological Chemistry-Practical	2	1
	II	3	Microbiology	3	3
			Microbiology-Practical	2	1
		4	Basic Immunology	3	3
			Basic Immunology-Practical	2	1
II	III	5	Biophysical Techniques	3	3
			Biophysical Techniques-Practical	2	1
		6	Basic Molecular Biology	3	3
			Basic Molecular Biology-Practical	2	1
		7	Genetic Engineering	3	3
			Genetic Engineering-Practical	2	1
	IV	8	Bioinformatics, Biostatistics & Bioethics	3	3
			Bioinformatics, Biostatistics & Bioethics-Practical	2	1
		9	Basics of Plant Biotechnology	3	3
			Basics of Plant Biotechnology-Practical	2	1

Year	Semester	Course	Title of the Course	No. of Hrs /Week	No. of Credits
III		10	Basics of Animal Biotechnology	3	3
			Basics of Animal Biotechnology-Practical	2	1
	V	11	Industrial Biotechnology	3	3
			Industrial Biotechnology-Practical	2	1
		12 A	Medical Biotechnology	3	3
			Medical Biotechnology-Practical	2	1
		OR			
		12 B	Marine Biotechnology	3	3
			Marine Biotechnology-Practical	2	1
		13 A	Nano Biotechnology	3	3
			Nano Biotechnology-Practical	2	1
		OR			
		13 B	Biofertilizers and Biopesticides Production	3	3
			Biofertilizers and Biopesticides Production-Practical	2	1
	VI	14 A	Pharmaceutical Biotechnology	3	3
			Pharmaceutical Biotechnology-Practical	2	1
		OR			
		14 B	Food and Nutritional Biotechnology	3	3
			Food and Nutritional Biotechnology-Practical	2	1
		15 A	Genomics & Proteomics	3	3
			Genomics & Proteomics-Practical	2	1
		OR			
		15 B	Environmental Biotechnology	3	3
			Environmental Biotechnology-Practical	2	1

Note: In the III Year (during the V and VI Semesters), students are required to select a pair of electives from one of the Two specified domains. For example: if set 'A' is chosen, courses 12 to 15 to be chosen as 12 A, 13 A, 14 A and 15 A or if set 'B' is chosen, It is to be chosen as 12 B, 13 B, 14 B and 15 B to ensure in-depth understanding and skill development in the chosen domain, students must continue with the same domain electives in both the V and VI Semesters.

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SEMESTER - III

COURSE 5: BIOPHYSICAL TECHNIQUES

Theory

Credits: 3

3 hrs/week

Course Objectives:

1. To introduce the principles and applications of spectrophotometry in biological research.
2. To explain the theory, instrumentation, and uses of different types of chromatography.
3. To familiarize students with the principles and applications of electrophoresis techniques.
4. To provide knowledge about centrifugation methods, rotors, centrifuge types, and their applications.
5. To describe tracer techniques, radioactivity measurements, autoradiography, and the use of non-radioactive labels.

Learning Outcomes:

After completing this course, students will be able to:

1. Understand the principles of light absorption and apply UV-visible spectrophotometry.
2. Differentiate between various chromatographic techniques and apply them for biomolecule separation and analysis.
3. Perform and interpret results from gel electrophoresis, SDS-PAGE, isoelectric focusing, and related techniques.
4. Apply centrifugation methods for separation and analysis of cellular and molecular components.
5. Explain the principles of tracer techniques, radioactivity measurement, and non-radioactive labeling methods in biotechnology.

Syllabus

UNIT I

Spectrophotometry: Spectrum of light, absorption of electromagnetic radiations, Beer-Lambert's law: derivation and deviations, extinction coefficient, colorimeter, instrumentation of UV and visible spectrophotometry, double beam spectrometer, dual-wavelength spectrometer, applications of UV and visible spectrophotometry.

UNIT II

Chromatography: Principle, instrumentation and applications of paper chromatography, thin layer chromatography, gel filtration chromatography, ion-exchange chromatography, affinity chromatography, high performance liquid chromatography (HPLC).

UNIT III

Electrophoresis: Migration of ions in electric field, factors affecting electrophoretic mobility, gel electrophoresis: types of gels, agarose gel electrophoresis, SDS-PAGE and Native-PAGE electrophoresis and their applications, isoelectric focusing, two-dimensional electrophoresis, pulsed-field gel electrophoresis.

UNIT IV

Centrifugation: Basic principles of centrifugation, types of rotors, types of centrifuges: clinical, high-speed and ultracentrifuges, preparative centrifugation: differential and density gradient centrifugation, analytical centrifugation and their applications.

UNIT V

Tracer Techniques: Radioactive and stable isotopes, rate of radioactive decay, units of radioactivity, measurement of radioactivity: Geiger-Muller counter, autoradiography, non-radioactive compounds: fluorescein, biotin, digoxigenin and their applications in biotechnology.

Reference books:

- Keith Wilson and John Walker – *Principles and Techniques of Biochemistry and Molecular Biology* – Cambridge University Press.
- R. K. Murray et al. – *Harper's Illustrated Biochemistry* – McGraw Hill (for basics of spectrophotometry and biochemistry techniques).
- David J. Holme and Hazel Peck – *Analytical Biochemistry* – Pearson Education.
- Boyer, R. – *Modern Experimental Biochemistry* – Pearson.
- Plummer, D.T. – *An Introduction to Practical Biochemistry* – Tata McGraw Hill.
- S. K. Sawhney and R. Singh – *Introductory Practical Biochemistry* – Narosa Publishing House.
- Ghosh, T.K. – *Biophysical Chemistry* – Books and Allied (for tracer techniques and centrifugation).

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SEMESTER - III

COURSE 5: BIOPHYSICAL TECHNIQUES

Practical

Credits: 1

2 hrs/week

Practical Component:

1. Separation of plant pigments and amino acids by paper chromatography
2. Separation of lipids of TLC
3. Quantification of DNA using Spectrophotometer
4. Quantification of RNA using Spectrophotometer
5. Agarose gel electrophoresis

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SEMESTER - III

COURSE 6: BASIC MOLECULAR BIOLOGY

Theory

Credits: 3

3 hrs/week

Course Objectives:

1. To understand the experimental proof that DNA is the genetic material.
2. To study genome organization in prokaryotes and eukaryotes.
3. To explain the processes of DNA replication, transcription, and translation.
4. To understand the regulation of gene expression in prokaryotic and eukaryotic systems.
5. To develop a basic understanding of molecular mechanisms underlying heredity and protein synthesis.

Learning Outcomes:

After completing this course, students will be able to:

1. Describe experiments that established DNA as the genetic material.
2. Compare the genome structure of prokaryotes and eukaryotes.
3. Explain the mechanism of DNA replication, transcription, and translation.
4. Discuss the regulation of gene expression using operon models.
5. Apply knowledge of molecular biology to understand genetic processes in living organisms.

Syllabus

Unit I

Genome Structure: Experiments to prove DNA as genetic material (Griffith experiment, Hershey- Chase experiment). Genome Organization: Prokaryotic and Eukaryotic genomes. Concept of Gene, Genome and Genome size with respect to Prokaryotic and Eukaryotic genomes.

Unit II

DNA Replication: Proof of semiconservative replication. Enzymes Involved in DNA Replication: DNA polymerase I, II and III, Helicases, Topoisomerases, single strand binding proteins, primase. Mechanism of DNA Replication (Prokaryotic and Eukaryotic). M13 Viral DNA Replication (Rolling circle method).

Unit III

Transcription: Basic features of transcription, the structure of prokaryotic RNA polymerases – Prokaryotic and Eukaryotic. Concept of core enzyme, holo enzyme, and sigma factor. Concept of promoter – TATA Box, -10 and -35 sequences). Transcription Mechanisms: Prokaryotic and Eukaryotic. Post Transcriptional Modifications in Eukaryotes. Concept of Reverse Transcription.

Unit IV

Translation: Genetic code: Features of genetic code, Structure of mRNA, brief structure of tRNA. Codon-anticodon interaction - the Wobble Hypothesis. Mechanism of Translation - Prokaryotic and Eukaryotic. Initiation, elongation, termination protein.

Unit V

Gene Regulation: Clustered genes the operon concepts - Negative and positive control of the Lac Operon, trp operon, Control of gene expression. Poly and Mono cistronic mRNA.

Reference books:

- Watson, J.D., Baker, T.A., Bell, S.P., Gann, A., Levine, M. & Losick, R. (2017). *Molecular Biology of the Gene*. 7th Edition, Pearson Education.
- Lodish, H., Berk, A., Kaiser, C.A., Krieger, M., Bretscher, A., Ploegh, H., Amon, A. & Martin, K. (2021). *Molecular Cell Biology*. 9th Edition, W.H. Freeman and Company.
- Krebs, J.E., Goldstein, E.S. & Kilpatrick, S.T. (2017). *Lewin's Genes XII*. Jones & Bartlett Learning.
- Alberts, B., Johnson, A., Lewis, J., Morgan, D., Raff, M., Roberts, K. & Walter, P. (2014). *Molecular Biology of the Cell*. 6th Edition, Garland Science.
- Primrose, S.B. & Twyman, R.M. (2006). *Principles of Gene Manipulation and Genomics*. 7th Edition, Blackwell Publishing.
- Berg, J.M., Tymoczko, J.L., Gatto, G.J. & Stryer, L. (2019). *Biochemistry*. 9th Edition, W.H. Freeman and Company.
- Weaver, R.F. (2018). *Molecular Biology*. 6th Edition, McGraw Hill Education.

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SEMESTER - III

COURSE 6: BASIC MOLECULAR BIOLOGY

Practical

Credits: 1

2 hrs/week

Practical Component:

1. Effect of UV radiations on the growth of microorganisms.
2. Determination of absorption maxima of DNA and RNA and their quantification
3. Quantitative estimation of RNA
4. Quantitative estimation of DNA
5. Isolation of plasmid DNA from bacteria
6. Isolation of genomic DNA from *E. coli*
7. Isolation of DNA from sheep liver
8. Isolation of DNA from plant leaves (Rice or Tobacco or any other plant)
9. Separation of DNA by Agarose gel Electrophoresis
10. Purity analysis of the Nucleic acids

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SEMESTER - III

COURSE 7: GENETIC ENGINEERING

Theory

Credits: 3

3 hrs/week

Course Objectives:

1. To introduce the principles, scope, and applications of genetic engineering.
2. To study molecular tools such as enzymes, cloning vectors, and transformation methods.
3. To understand hybridization techniques, PCR, and their applications in research.
4. To learn strategies for construction of genomic/cDNA libraries and protein expression.
5. To explore advanced genetic manipulation techniques such as CRISPR, site-directed mutagenesis, and next-generation sequencing.

Learning Outcomes:

After completion of this course, students will be able to:

1. Explain the history, scope, and tools used in genetic engineering.
2. Describe cloning vectors and transformation techniques in bacteria, yeast, plants, and animals.
3. Apply hybridization and PCR techniques for molecular analysis.
4. Construct and analyze genomic and cDNA libraries for gene studies.
5. Evaluate modern techniques such as CRISPR-Cas9, gene silencing, and next-generation sequencing.

Syllabus

UNIT – I

History, scope, and recent developments in Genetic Engineering. Molecular tools in genetic engineering – Restriction enzymes: Endonucleases and Exonucleases. DNA Modifying enzymes. Ligation (cohesive-end and blunt-end ligation), use of linkers and adaptors. Guidelines and strategies in plant and animal genetic engineering.

UNIT – II

Cloning vectors: Definition, properties and types. Plasmid vectors – pUC19 and pBR322. Phage vectors – λ (lambda) and M13. Cosmid vectors, shuttle vectors, and expression vectors. YAC vectors (*Saccharomyces cerevisiae* as a model) and BAC vectors (*E. coli*). Bacterial transformation. Screening and selection of recombinants – GUS and GFP assisted selection.

UNIT – III

Hybridization techniques: Probes (radioactive and non-radioactive) and their labelling. DNA labelling methods – Nick translation, random priming, and primer extension. Southern blotting and Northern blotting – principles and applications. Polymerase Chain Reaction (PCR): Principle, types of PCR, and applications.

UNIT – IV

Construction of genomic and cDNA libraries. Vector engineering – role of promoters. Codon optimization and *in vitro* translation. Strategies for protein overexpression in bacteria, yeast, insects, plants, and mammalian cell lines.

UNIT – V

Chromosome engineering and targeted gene replacement. Gene editing – CRISPR-Cas9. Manipulation of gene regulation and gene silencing. Site-directed mutagenesis. DNA sequencing methods – Maxam-Gilbert (chemical method), Sanger's method, and Next Generation Sequencing (NGS) with applications.

Reference books:

1. Brown, T.A. (2016). *Gene Cloning and DNA Analysis: An Introduction*. 7th Edition, Wiley-Blackwell.
2. Primrose, S.B. & Twyman, R.M. (2006). *Principles of Gene Manipulation and Genomics*. 7th Edition, Blackwell Publishing.
3. Watson, J.D., Baker, T.A., Bell, S.P., Gann, A., Levine, M. & Losick, R. (2017). *Molecular Biology of the Gene*. 7th Edition, Pearson Education.
4. Glick, B.R., Pasternak, J.J. & Patten, C.L. (2010). *Molecular Biotechnology: Principles and Applications of Recombinant DNA*. 4th Edition, ASM Press.
5. Alberts, B., Johnson, A., Lewis, J., Morgan, D., Raff, M., Roberts, K. & Walter, P. (2014). *Molecular Biology of the Cell*. 6th Edition, Garland Science.
6. Weaver, R.F. (2018). *Molecular Biology*. 6th Edition, McGraw Hill Education.
7. Channarayappa, C. (2014). *Molecular Biotechnology: Principles and Practices*. Universities Press, India.

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SEMESTER - III

COURSE 7: GENETIC ENGINEERING

Practical

Credits: 1

2 hrs/week

Practical Component:

1. Problem solving related to restriction enzymes sites in Genetic engineering.
2. Transformation in Bacteria using plasmid
3. Restriction digestion of DNA and its electrophoretic separation.
4. Ligation of DNA molecules and their testing using electrophoresis.
5. Activity of DNAase and RNAse on DNA and RNA.
6. Isolation of Plasmid DNA
7. Demonstration of PCR

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SEMESTER - IV

COURSE 8: BIOINFORMATICS, BIOSTATISTICS & BIOETHICS

Theory

Credits: 3

3 hrs/week

Course Objectives:

1. To introduce the scope of bioinformatics and its role in biological research.
2. To provide knowledge of genomics, proteomics, and systems biology concepts.
3. To familiarize students with databases, BLAST, drug design, and phylogenetic tools.
4. To train students in statistical methods for data collection, analysis, and interpretation.
5. To create awareness about bioethics, biosafety, and intellectual property rights in biotechnology.

Learning Outcomes:

After successful completion of this course, students will be able to:

1. Explain the fundamentals of bioinformatics and its applications.
2. Use biological databases, BLAST, and computational methods in biotechnology research.
3. Apply statistical tools like central tendency, probability, and hypothesis testing.
4. Perform tests such as t-test, ANOVA, chi-square, correlation, and regression for biological data.
5. Demonstrate understanding of bioethical issues, biosafety practices, and intellectual property rights.

Syllabus

Unit – I

Scope of computers in biological research. Introduction to Bioinformatics: definition, nature and scope of bioinformatics. Bioinformatics versus computational biology. Concept of genomics and proteomics. Branches of Bioinformatics.

Unit – II

Basic concepts of systems biology. Types of databases and their applications in biotechnology. Sequence similarity search using BLAST. Overview of computer-aided drug design. Computational phylogenetics (PHYlip) and its applications.

Unit – III

Data collection and measurement of central tendency (mean, median and mode). Dispersion (standard error and standard deviation). Probability and distribution (Poisson and binomial distributions. Normal distribution).

Unit – IV

Population and sampling, test of significance, and test of hypothesis. Student t-test for small samples. ANOVA and chi-square test for analysis. Correlation and regression.

Unit – V

Bioethics in human and animal experimentation, cloning and stem cell research. Animal rights and welfare. Biosafety -Introduction to biological safety cabinets and biosafety levels. Primary containment for biohazards and strategies for their management (GLP, GMP). Intellectual Property Rights (IPRs).

Reference Books

1. Mount, D.W. (2004). *Bioinformatics: Sequence and Genome Analysis*. 2nd Edition, Cold Spring Harbor Laboratory Press.
2. Lesk, A.M. (2017). *Introduction to Bioinformatics*. 5th Edition, Oxford University Press.
3. Rastogi, S.C., Mendiratta, N. & Rastogi, P. (2019). *Bioinformatics: Methods and Applications*. 5th Edition, PHI Learning Pvt. Ltd.
4. Gupta, S.P. (2012). *Statistical Methods*. 43rd Edition, Sultan Chand & Sons.
5. Bailey, N.T.J. (1995). *Statistical Methods in Biology*. 3rd Edition, Cambridge University Press.
6. Beauchamp, T.L. & Walters, L. (2008). *Contemporary Issues in Bioethics*. 7th Edition, Cengage Learning.
7. Singh, B.D. (2012). *Biotechnology: Expanding Horizons*. Kalyani Publishers.

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SEMESTER - IV

COURSE 8: BIOINFORMATICS, BIOSTATISTICS & BIOETHICS

Practical

Credits: 1

2 hrs/week

Practical Component:

1. Mean, Median, Mode
2. Standard deviation, Standard error
3. Testing of hypotheses regarding population mean
4. Testing of hypotheses about the difference between population means
5. Chi-square test
6. Testing of Correlation Coefficient
7. Fitting of simple linear regression
8. ANOVA
9. Sequence retrieval (protein and gene) from NCBI
10. Similarity search using BLASTN, BLASTP
11. Structure download (protein and DNA) from PDB

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SEMESTER - IV

COURSE 9: BASICS OF PLANT BIOTECHNOLOGY

Theory

Credits: 3

3 hrs/week

Course Objectives:

1. To understand the principles of plant tissue culture and its applications.
2. To study specialized tissue culture techniques such as micropropagation, haploid culture, and protoplast fusion.
3. To learn different methods of genetic transformation in plants.
4. To explore the development and applications of transgenic plants.
5. To examine the use of plant biotechnology in production of valuable compounds, biofuels, and in sustainable agriculture, along with ethical and biosafety considerations.

Learning Outcomes:

After completing this course, students will be able to:

1. Explain the basic principles of plant tissue culture and its significance.
2. Apply micropropagation, haploid culture, and protoplast fusion techniques in plant improvement.
3. Differentiate between direct and indirect methods of plant genetic transformation.
4. Analyze the role of transgenic plants in agriculture, health, and environment.
5. Discuss applications of plant biotechnology in molecular farming, biofuels, biocontrol, and biofertilizers, and evaluate related ethical and biosafety issues.

Syllabus

Unit I

Plant Tissue Culture: Principles of plant tissue culture. Totipotency, differentiation, dedifferentiation, redifferentiation. Sterilization techniques, Types of culture media and composition of MS medium. Callus culture, organogenesis, somatic embryogenesis

Unit II

Specialized Plant Tissue Culture Techniques: Micropropagation and its applications. Haploid culture – anther and pollen culture, doubled haploids and their applications. Protoplast culture and fusion – somatic hybridization, cybrids. Cryopreservation and germplasm conservation

Unit III

Plant Genetic Transformation Methods: Direct gene transfer methods – particle bombardment, electroporation, microinjection. Indirect Gene Transfer Method - Agrobacterium tumefaciens and Ti plasmid, Ri plasmid. Agrobacterium-mediated gene transfer

Unit IV

Transgenic Plants and Applications: Transgenic plants for insect resistance (Bt crops). Transgenic plants for herbicide tolerance. Transgenic plants for disease resistance and stress tolerance. GMO Foods, Ecological impact of transgenic plants

Unit V

Applications of Plant Biotechnology: Therapeutic proteins, Edible vaccines, biofortification, and molecular farming. Phytochemical products from plants- Secondary Metabolites (Alkaloids, Flavonoids, Terpenoids). Bioethanol and biodiesel production. Biocontrol and Biofertilizers. Ethical issues and biosafety regulations in Plant biotechnology

Reference books:

1. Bhojwani, S.S. & Razdan, M.K. (1996). *Plant Tissue Culture: Theory and Practice*. Elsevier.
2. Gamborg, O.L. & Phillips, G.C. (1995). *Plant Cell, Tissue and Organ Culture: Fundamental Methods*. Springer.
3. Chawla, H.S. (2009). *Introduction to Plant Biotechnology*. 3rd Edition, Oxford & IBH Publishing.
4. Slater, A., Scott, N. & Fowler, M. (2008). *Plant Biotechnology: The Genetic Manipulation of Plants*. 2nd Edition, Oxford University Press.
5. Singh, B.D. (2015). *Biotechnology: Expanding Horizons*. Kalyani Publishers.
6. Glick, B.R., Pasternak, J.J. & Patten, C.L. (2010). *Molecular Biotechnology: Principles and Applications of Recombinant DNA*. 4th Edition, ASM Press.
7. Razdan, M.K. (2003). *Introduction to Plant Tissue Culture*. Science Publishers.

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SEMESTER - IV

COURSE 9: BASICS OF PLANT BIOTECHNOLOGY

Practical

Credits: 1

2 hrs/week

Practical Component:

1. Plant culture media and composition of MS media
2. Raising of aseptic seedlings
3. Induction of callus from different explants
4. Plant propagation through Tissue culture (shoot tip and Nodal culture)
5. Establishing a plant cell culture (both in solid and liquid media)
6. Suspension cell culture

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SEMESTER - IV

COURSE 10: BASICS OF ANIMAL BIOTECHNOLOGY

Theory

Credits: 3

3 hrs/week

Course Objectives:

1. To provide knowledge about the scope, importance, and laboratory requirements for animal cell culture.
2. To introduce students to various animal cell culture techniques and their applications.
3. To understand the role of stem cells and tissue engineering in regenerative medicine and organ culture.
4. To study transgenic animal production methods, cloning, and reproductive biotechnologies.
5. To explore applications of animal biotechnology in molecular farming, pharmaceuticals, and disease models, while addressing ethical and biosafety issues.

Learning Outcomes:

After completing this course, students will be able to:

1. Explain the principles of animal cell culture, media composition, and growth requirements.
2. Perform and differentiate between primary, secondary, continuous, and suspension cell cultures, including preservation and contamination control.
3. Analyze the applications of stem cells, scaffolds, and bioreactors in tissue engineering.
4. Describe the production of transgenic animals, animal cloning techniques, and reproductive biotechnology methods.
5. Evaluate the applications of animal biotechnology in health, pharmaceuticals, livestock improvement, and discuss related ethical and biosafety concerns.

Syllabus

Unit I

Animal Cell Culture: Scope and importance of animal biotechnology. Laboratory organization, and Equipment required. Composition and Types of Animal Cell Culture Media (BSS, RPMI-1640). Role of carbon dioxide, serum, and growth factors in cell culture. Application of antibiotics and growth supplements

Unit II

Cell Culture Techniques: Primary culture, secondary culture, continuous cell lines, and suspension cultures. Sub-culturing, cytotoxicity and cell viability assays. Contamination in cell culture and control measures, cryopreservation methods

Unit III

Stem Cells and Tissue Engineering: Embryonic and adult stem cell culture and its application. Biocompatible Materials and their applications for Scaffolds preparation used in Organ culture and 3D culture models. Bioreactors for large-scale animal cell culture

Unit IV

Transgenic Technology: Transgenic animal production methods – microinjection, viral vectors. Animal cloning techniques – nuclear transfer, case studies (e.g., Dolly). Invitro fertilization in livestock. Gene targeting, gene silencing, and knock-out techniques

Unit V

Applications of Animal Biotechnology: Molecular Farming: Hormone production in livestock, Production of vaccines and pharmaceutical compounds. Role of animal models in disease diagnosis and treatment. Ethical, regulatory and biosafety issues in animal biotechnology

Reference books:

1. Freshney, R.I. (2015). *Culture of Animal Cells: A Manual of Basic Technique and Specialized Applications*. 7th Edition, Wiley-Blackwell.
2. Masters, J.R.W. (2000). *Animal Cell Culture: Practical Approach*. 3rd Edition, Oxford University Press.
3. Butler, M. (2004). *Animal Cell Culture and Technology*. 2nd Edition, Taylor & Francis.
4. Davis, J.M. (2011). *Animal Cell Culture: Essential Methods*. Wiley-Blackwell.
5. Glick, B.R., Pasternak, J.J. & Patten, C.L. (2010). *Molecular Biotechnology: Principles and Applications of Recombinant DNA*. 4th Edition, ASM Press.
6. Singh, B.D. (2015). *Biotechnology: Expanding Horizons*. Kalyani Publishers.
7. Portner, R. (2007). *Animal Cell Biotechnology: Methods and Protocols*. Humana Press.

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SEMESTER - IV

COURSE 10: BASICS OF ANIMAL BIOTECHNOLOGY

Practical

Credits: 3

3 hrs/week

Practical Component:

1. Cell count by haemocytometer.
2. Establishing primary cell culture of chicken embryo fibroblasts.
3. Animal tissue culture– maintenance of established cell lines.
4. Animal tissue culture– virus cultivation.
5. Estimation of cell viability by dye exclusion (Trypan blue).

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