Hochschule
Bonn-Rhein-Sieg
University of Applied Sciences

Module Handbook
Biomedical Sciences
Master of Science (M.Sc.)

Department of Natural Sciences
Hochschule Bonn-Rhein-Sieg

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Compulsory Courses:

Electives (Selection):

Module:	**Monitoring of Clinical Trials**
Semester:	1st Semester
Course coordinator:	Prof. Martin Sieber
Lecturer:	Prof. Martin Sieber
Language:	English
Assignment in Curriculum:	**Compulsory Course in the 1st Semester Biomedical Sciences**
Course units/Credit hours:	Lecture: 2 SWS Seminar: 2 SWS, Exercise: 2 SWS; group size: max. 16
Student workload:	Contact hours / Self study L: 30 / 30 E: 30 / 60 P: 30 / 60 Sum: 90 / 150 **Sum total: 240 hours**
Credits:	8 ECTS
Prerequisites according to examination regulations:	None
Recommendation:	Basic Immunology, Molecular Biology, Cell Biology
Learning outcomes:	Students are able to • Learn the international standards for clinical research (such a ICH-E6-GCP) • Learn the regulatory environment for clinical trials esp. clinical trials for medicinal products • know the laws, rules, regulations, essential document requirements and guidelines relevenat to trial design, implementation, evaluation and reporting. By means of understanding • international standards and regulations on study planning, site selection & activation, study management and monitoring in order to • do a design on planning and evaluation of clincial trials.
Content:	Lecture: • Fundamentals and rational of trial design • Medicinal Product and medical device development • Good Clinical Practice including, informed consent, data protection in clinical trials • Approval of Clincal Trials. • Planning and Conduct of Clinical Trials; • Project Management & Study Management • Data Managemnt and Drug Supply • Audits and inspections • Reporting of Clinical Trials: CSR, Publication Guidelines, Evidence based Medicine

	Seminar: • Details on Trail Design and Protocoll Development (Hypotheses Testing, Biases Confounding Factors) • Site Selection and Actication • Details of Monitoring of Clinical Trials such as Source Date verification and Review, Safety Reporting, Drug Accountability • Documentation in clinical trials: Data capuring, Trial Master file & Investigator Site File • Site Close out Excercise: • Practical Excersises on Study Planning, Site Selection & Activation and Monitoring (Saftey reporting, Drug Accountability, Source Data Verification & Review)
Assessment:	Written exam – 100%, 120 min
Teaching formats:	L/E: Power Point Presentation, legal texts, exercises, student presentations. P: Clinical trials simulations,
Literature**:**	• International Council for Harmonisation of Technical Requirements for Pharmaceuticals for Human Use • A Clinical Trials Manual From The Duke Clinical Research Institute: Lessons from a Horse Named Jim • Fundamentals of Clinical Trials

Module:	**Pharmacology & Toxicology**
Semester:	1st Semester
Course coordinator:	Prof. Dr. Ulrike Bartz
Lecturer:	Prof. Dr. Ulrike Bartz
Language:	English
Assignment in Curriculum:	**Compulsory Course in the 1st Semester Biomedical Sciences**
Course units/Credit hours:	L: 2 SWS E: 2 SWS, max. group size: 16 Lab work: 2 SWS; group size: max. 16
Student workload:	Contact hours / Self study L: 30 / 30 E: 30 / 60 P: 30 / 60 Sum: 90 / 150 **Sum total: 240 hours**
Credits:	8 ECTS
Prerequisites according to examination regulations:	None
Recommendation:	Biochemistry
Learning outcomes:	The students are able • to describe the characteristics of compounds in the body relating to Pharmacokinetics (PK) and Pharmacodynamics (PD) • to discuss and interpret PK profiles of different dosage forms; oral dosing (single/multiple doses), intravenous dosing (bolus and infusion) • to derive potential metabolites (urine/plasma) after exposure with a drug • to interpret analytical data from biological samples (plasma, urine) in the context of clinical trials by means of understanding • application principles of PK and PD • Phase I/II metabolisation reactions and their mechanisms in order to • apply bioanalytical methods and together with the relevant documentation • understand therapeutic interventions in diseases and approaches followed by pharmaceutical industry (e.g. prodrugs) and academic researchers
Content:	Lecture: drug actions, pharmacokinetics, (L)ADME, pharmacodynamics (mode of action), adverse effects, pharmaceutical interactions,

	toxicology, in particular: biotransformation, bioactivation, elimination mechanisms, medical terminology, pharmacokinetic models, cumulation, bioavailability, prodrug concept, enterohepatic circulation, pharmacogenetics, preclinical and clinical trials/bioequivalence studies/regulations for drug approval. Exercises/seminar: Exercises for calculations and data analysis; work on an EPAR (European Product Assessment Report) and a SmPC (Summary of Product Characteristics) of a chosen innovative drug. Laboratory course: Several experiments in small groups on pharmacokinetics (plasma samples) including mathematical analysis: One compartment model setup: oral intake, intravenous dose administration, infusion or multiple i.v. bolus administration, drug cumulation, calculation of the relevant pharmacokinetic parameters. Analysis of urinary data: analysis of metabolites (SPE/HPLC or SPE/GC-MS of a chosen urine sample) and/or enzyme based assay (mode of action of a drug; data analysis via Graph Pad Prism Software).
Assessment:	Oral exam (30 min) – 70% , Laboratory report 30%
Teaching formats:	L/E: power point presentation, overhead, whiteboard, textbooks P: practical course description, textbooks, supportive videos
Textbooks**:**	Drug actions - Basic Principles and therapeutic aspects E. Mutschler/H. Derendorf; MedPharm Scientific Publishers Pharmacokinetic Processes, mathematics and applications Peter G. Welling Wiley Science Applied Biopharmaceutics and Pharmacokinetics L. Shargel/A.Yu; McGraw-Hill Medical Publishing Division Pharmakokinetik kompakt: Grundlagen und Praxisrelevanz Hartmut Derendorf, Thomas Gramatte, Hans Günter Schäfer, Alexander Staab, Wissenschaftl. Verlagsgesellschaft Stuttgart Further literature and weblinks will be provided at the beginning of the module.

Module:	**Clinical Chemistry**
Semester:	1st Semester
Course coordinator:	Prof. Jörn Oliver Sass
Lecturer:	Prof. Jörn Oliver Sass
Language:	English
Assignment in Curriculum:	**Compulsory Course in the 1st Semester MSc Biomedical Sciences**
Course units/Credit hours:	The course comprises lectures, exercises/seminars and experiments. L: 3 SWS E: 1 SWS Lab work: 2 SWS; group size max.16 students (participation mandatory)
Student workload:	Contact hours / Self study L: 45 / 60 E: 15 / 45 P: 30 / 45 Sum: 90 / 150 **Sum total: 240 hours**
Credits:	8 ECTS
Prerequisites according to examination regulations:	None
Recommendation:	Basic General Chemistry and Biochemistry; Arithmetic
Learning outcomes:	Students are able to • read and plan routine laboratory tests • consider the effects of pre- and post-analytics as well as opportunities and limitations associated with certain laboratory techniques • prepare reagents for experiments and perform some manual tests by means of understanding • the basics of Clinical Chemistry and its role in a medical setting • a wide range of analytical methods • the pathobiochemical and pathophysical alterations present in exemplary diseases and the selection of biomarkers • fundamentals of quality control in a medical laboratory • concepts and elements of newborn screening • chemical calculations in order to • use the acquired skills and knowledge in related fields or to use them as a starting point for future work in Clinical Chemistry/ Analytical Biochemistry/ Laboratory Diagnostics
Content:	<u>Lecture:</u> • Lab safety (participation mandatory) and course standards; branches of diagnostic laboratories; literature and other helpful resources; sample materials and possible laboratory parameters • Preanalytics • Postanalytics, reference intervals, clinical decision limits • Pricipals of Measurement

	• Analytical Methods. including selected immunoassays • (Serum) Protein Electrophoresis • Enzymes in Laboratory Diagnostics • Biomarkers for diseases of liver, pancreas, kidney and heart • Quality Control Exercise: questions, selected repetitions, chemical calculations (buffers, substrates for photometric tests etc.), pointing to/ repetition of fundamentals of Biochemistry/ Organic Chemistry/ General Chemistry Lab course: • Pipet training. Enzyme activity tests. • Different types of creatinine assays. • Turbidimetry. Radial Immunodiffusion. • Radial Immunodiffusion. Immunochromatography. Urine analyses. Mandatory excursion to a medical laboratory.
Assessment:	Written exam (90 min) 100 %, practical part needs to be passed (based on regular and active participation and the submitted protocol)
Teaching formats:	L/E: Power Point Presentation, Blackboard/ Whiteboard, video animations, Textbooks/ scientific journal article, internet resources P: Practical work in the laboratory, guided by lecturer/ tutor and script, supported by literature.
Textbooks**:**	• Tietz Textbook of Clinical Chemistry and Molecular Diagnostics, Elsevier, 6th or later edition • Abbott Learning Guide Clinical Chemistry, latest edition: https://www.corelaboratory.abbott/sal/learningGuide/ADD-00061345_ClinChem_Learning_Guide.pdf • For background studies (if needed): various textbooks/ internet resources from biochemistry and general chemistry

Module:	**Virology**
Semester:	2nd Semester
Course coordinator:	Prof. Dr. Edda Tobiasch
Lecturer:	Prof. Dr. Edda Tobiasch
Language:	English
Assignment in Curriculum:	**Compulsory Course in the 2nd Semester Biomedical Sciences**
Course units/Credit hours:	L: 2 SWS E: 2 SWS, max. group size: 16 Lab work: 2 SWS; group size: max. 16
Student workload:	Contact hours / Self study L: 30 / 30 E: 30 / 60 P: 30 / 60 Sum: 90 / 150 **Sum total: 240 hours**
Credits:	8 ECTS
Prerequisites according to examination regulations:	None
Recommendation:	Previous knowledge in Molecular Genetics and Cell Culture from previous study courses General safety instructions for working in laboratories Safety instructions for working with S12 and L2 organisms
Learning outcomes:	Students are able to • understand virologic mechanisms of replication • can apply virologic knowledge to address questions related to viral diseases • apply knowledge to basic experiments • apply major virologic techniques by means of understanding • the basic properties of viruses • the principles of virus-cell interaction • rational of basic virologic experimental techniques • how to access and work with specific literature in order to • develop experimental workflows on an introductionary level for virological applications in corresponding laboratoires in science and development in pharmaceutical industries and universities (e.g. diagnostic laboriatories) • understand basic viral disease mechnanisms
Content:	<u>Lecture:</u> The couse focuses on human infecting viral families with respect to the following aspects

	• Worldwide infections, different cause of viral hepatitis, virus and cancer, viral childhold diseases, viruses and gene therapy, emerging viruses, bioterrorism, prion diseases • Occurrence and disemination of viral diseases • Adaption of the virus to the host: zoonosis, host change, "shift and drift", evolution • Prevention measurement and treatment of viral diseases • Taxonomy, history, morphology of virions, replication strategies (Baltimore classification), transmission, host and risk groups, symptoms diagnostic methods, viral protection against the immue system, specific immune reactions, acute and chonic diseases, complications • Cosomopolitan and endemic ocurrence of viral diseases with respect to climatic and socila factors of dissemination, epidemiology, erradication measures and prevention. <u>Tutorial:</u> the students have to work on their own and present a current scientific paper representing a defined specific question from virology. In the following discussion, the student has to defend this paper and the group has to understand the relation of this paper in the context of the already gained knowledge. <u>Lab course:</u> • Various methods (e.g. Western blot, ELISA, Hemmagglutinations test) for detecing viral proteins or antibodies of several viruses • Determination of viral antibody titers • Plaques assay to determine the viral titers • Cytopathic effects of defined viruses in cell culture • Viral popagation in cell culture and embryonated eggs
Assessment:	Written exam (120 min) 70% , Laboratory report 15%, Student talk in seminar 15%
Teaching formats:	L/E: Power Point Presentation, video animations, black board, textbooks, reviews and original paper P: Script, textbooks, lab videos
Textbooks**:**	• Fields Virology; D. M. Knipe and P. M. Howley, Lippincott Williams & Wilkins, latest edition. • Principles of Virus Molecular Biology, Pathogenesis and Control; S. J. Flint, L. W. Enquist, R. M. Krug, V. R. Racaniello, and A. M.-Skalka, ASM Press, latest edition. • Lexikon der Infektionskrankheiten des Menschen: Erreger, Symptome, Diagnose, Therapie und Prophylaxe; G. Darai, M. Handermann, E. Hinz und h.-G. Sonntag, Springer Verlag, latest edition.

Module:	**Neurobiology**
Semester	2nd Semester
Course Leader:	Prof. Dr. Mike Althaus
Lecturer:	Prof. Dr. Mike Althaus
Language:	English
Assignment in Curriculum:	**Compulsory Course in the 2nd Semester M.Sc. Biomedical Sciences**
Course Units/Credit hours:	The module consists of Lectures with integrated Exercises, and Practical Classes. Lectures: 3 credit hours Exercises: 1 credit hour Practical classes: 2 credit hours; max. group size 15
Student workload:	Contact hours / Private study time (hours) Lectures: 45 / 45 Exercises: 15 / 45 Practical classes: 30 / 60 Sum: 90 / 150 **Total sum: 240**
Credits:	8 ECTS
Prerequisites according to examination regulations:	None
Recommendations:	Basic knowledge in human or animal physiology acquired in a preceding study programme
Learning outcomes:	Student are able to • describe and explain neurobiological processes with correct terminology, • explain pathophysiological observations, • perform, obtain, analyse, interpret and communicate data derived from neurobiological experiments, • communicate neurobiological concepts, by means of understanding • learning neurobiological concepts in lectures and by applying their knowledge to answer neurobiological questions in exercises and practical classes, • applying neurobiological concepts to answer questions related to pathophysiological mechanisms in lectures and exercises, • performing neurobiological experiments (supervised in small groups) and delivering adequate experimental analyses, interpretation and documentation, • preparing educational posters on current concepts in neurobiology, in order to • apply their knowledge in neurobiology concepts to understand and interpret specialist literature in Neurobiology.

	• apply their knowlede in neurobiology to understand and interpret specialist literature in Pathophysiology and Biomedicine. • apply their skills to interprete and document experimental data derived from neurobiological experiments. • apply their skills to communicate complex scientific findings to an educated lay audience.
Summary of indicative content:	Lectures: The module teaches current concepts in neurobiology: • Membrane physiology • Structure and function of neurons • Electrophysiological techniques to study neuron function • Functional organisation of nervous systems • Neurobiology of vision • Biological clocks • Mechanical senses • Chemical senses • Learning and memory Exercises: • Analysis and interpretation of data from electrophysiological experiments • Design of experiments to address neurobiological research questions • Preparation and presentation of an educational poster on current concepts in neurobiology Practical classes: Students will perform neurobiological experiments, analyse, interpret, and document experimental data: • Recording of action potentials. • Advanced electrophysiological techniques: current clamp & voltage clamp
Assessment during module:	Successful participation in practical classes Succsessful poster presentation Successful Lab report (must be passed in order to pass module)
Assessment at the end of module:	Module examination with marks. Written exam (90 min) 100 %
Teaching style:	Lectures/Exercises: PowerPoint, Blackboard/Whiteboard, digital content (e.g. Videos), written Exercises, Textbooks Practical classes: written instructions incl. theoretical background, digital teaching formats (virtual Physiology), Other
Literature:	– Eric R. Kandel, James H. Schwartz, Thomas M. Jessell, Steven A. Siegelbaum, A. J. Hudspeth: Principles of Neural Science. 6th ed., McGraw-Hill Education, 2021. – Dale Purves, George J. Augustine, David Fitzpatrick et al.: Neuroscience. International 6th ed., Sinauer/Oxford University Press, 2018. – Jeremy M. Wolfe, Keith R. Kluender, Dennis M. Levi, et al.: Sensation & Perception, 5th ed., Sinauer/Oxford University Press, 2019.

Module:	**Medical Proteomics**
Semester:	2nd Semester
Course coordinator:	Prof. Angelika Muscate-Magnussen
Lecturer:	Prof. Angelika Muscate-Magnussen
Language:	English
Assignment in Curriculum:	**Compulsary Course in the 2nd Semester Biomedical Sciences**
Course units/Credit hours:	L: 2 SWS E: 2 SWS, max. group size: 16 Lab work: 2 SWS; group size: max. 16
Student workload:	Contact hours / Self study L: 30 / 30 E: 30 / 60 P: 30 / 30 Sum: 90 / 120 **Sum total: 210 hours**
Credits:	8 ECTS
Prerequisites according to examination regulations:	None
Recommendation:	Basic Organic Chemistry, Instrumental Analysis, Biochemistry, Clinical Chemistry
Learning outcomes:	Students are able to • find suitable research articles and select necessary information in publications for state of the art sample preparation and electrophoretic, chromatographic and mass spectrometric analyses • write detailed lab scripts for sample preparation based on published procedures in the literature • design reasonable proteomics work-flows for selected entry level applications in the field of mining and differential proteomics • decide on reasonable methods for protein purification • deduce basic analytical parameters from mass spectra • explain typical current proteomics strategies and applications to a novice in the field by means of understanding • the physiochemical properties of the major analyte classes in the biomedical field • the principles of analyte separation and detetction • the strategies of qualitative and quantitative analysis • the concepts of workflow design in order to • develop and optimize sample purification and quantification workflows on an introductionary level for biomedical applications

	• perform sample preparation of the major clinical sample types for qualitative and quantitative analyses by mass spectrometry
Content:	Lecture: • Sample preparation: sampling, sample handling, cell lysis methods, laser microdissection, diverse prefractionation techniques (like i.e. affinity and gel filtration chromatography, protein depletion, ultracentrifugation), protein digestion, protein quantification, clinical requirements • Separation techniques: 1- and 2D gel electrophoresis, DIGE, preparative IEF, capillary electrophoresis, HPLC and UPLC (employing Reversed Phase and Ion Exchange), multidimensional LC • Mass spectrometry: general principles, MALDI and ESI ionization, TOF, quadrupole, FT-ICR/ orbitrap, ion trap, tandem and hybrid instrumentation, CID, ECD/ETD, LC-ESI, SELDI, MALDI imaging • Proteomics strategies: bottom up, middle down, top down, discovery vs. hypothesis based, identification based on PMF and peptide sequencing, label and label free quantification approaches like SRM/MRM, SILAC, iTRAQ, ICAT, database analysis, search algorithm • Applications: mining, differential like biomarker discovery, protein-protein interactions, posttranslational modifications, glycomics, imaging MALDI, infectious diseases Exercise: Data analysis and evaluation based on problem sets and case studies of the primary literature, biochemical calculations Lab course: • Isolation and purification of Benzoylformate decarboxylase from P. putida, overexpressed in E. coli. Determination of the purity, activity and overall yield. • 2D-gel electrophoresis analysis of E. coli cell lysates, analyzing differences of the bacterial proteome in response to overexpression induction in cell culture
Assessment:	Written exam (120min) – 100% of overall grade
Teaching formats:	L/E: Power Point Presentation, self-assessment tools like Kahoot, Quizacademy, LEA quizzes, eboards like padlet, video animations, video lectures, textbooks P: Script, textbook, lab videos
Textbooks**:**	• Lovric, J. (2011): Introducing Proteomics from concepts to sample preparation, mass spectrometry and data analysis, Wiley-Blackwell • Westermeier, R. (2016): Electrophoresis in practice : a guide to methods and applications of DNA and protein separations, Wiley-VCH • Greaves, J., Roboz, J. (2014): Mass spectrometry for the novice, CRC Press https://ebookcentral.proquest.com/lib/hukb/detail.action?docID=1375545 • Lottspeich, F and Engels, J. (2018): Bioanalytics, Wiley-VCH

Module:	**Pathopysiology**
Semester	3rd Semester
Course Leader:	Prof. Dr. Mike Althaus
Lecturer:	Prof. Dr. Mike Althaus
Language:	English
Assignment in Curriculum:	**Compulsory Course in the** 3rd Semester **M.Sc. Biomedical Sciences**
Course Units/Credit hours:	The module consists of Lectures with integrated Exercises, and Practical Classes. Lectures: 3 credit hours Exercises: 1 credit hour Practical classes: 2 credit hours; max. group size 15
Student workload:	Contact hours / Private study time (hours) Lectures: 45 / 45 Exercises: 15 / 45 Practical classes: 30 / 60 Sum: 90 / 150 **Total sum: 240**
Credits:	8 ECTS
Prerequisites according to examination regulations:	None
Recommendations:	Basic knowledge in human or animal physiology acquired in a preceding study programme
Learning outcomes:	Student are able to • describe and explain pathophysiological processes with correct terminology, • explain pathophysiological observations, • perform, obtain, analyse, interpret and communicate data derived from physiological/pathophysiological experiments, by means of understanding • learning pathophysiological concepts in lectures and by applying their knowledge to answer pathophysiological questions in exercises and practical classes, • applying basic physiological concepts to answer questions related to pathophysiological mechanisms in lectures and exercises, • performing physiological/pathophysiological experiments (supervised in small groups) and delivering adequate experimental analyses, interpretation and documentation, in order to • apply their knowledge in pathophysiological concepts to understand and interpret specialist literature in Pathophysiology and Biomedicine. • apply their knowlede in physiological concepts to understand and interpret specialist literature in Pathophysiology and Biomedicine.

	• apply their skills to interprete and document experimental data derived from physiological/pathophyssiological experiments.
Summary of indicative content:	Lectures/Exercises: The module teaches current concepts in Pathophysiology with a focus on the molecular mechanisms of diseases: • Cellular adaptations and response to stress • Homeostasis • Inflammation and tissue repair • Cancer • Lung diseases and their mechanisms • Kidney diseases and their mechanisms • Heart diseases and their mechanisms • Cardiovascular diseases and their mechanisms Practical classes: Students will perform physiological/pathophysiological experiments, analyse, interpret, and document experimental data: • Analysis of protein content and composition of urine samples representing pathophysiological conditions (LMW proteinuria and nephrotic syndrome) with Bradford Assays and SDS-PAGE. • Electrophysiological measurement of epithelial sodium channel (ENaC) activation by nephrotic urine: Heterologous gene expression in Xenopus oocytes and Two-Electrode Voltage-Clamp recordings.
Assessment during module:	Successful participation in practical classes Successful Lab report (must be passed in order to pass module)
Assessment at the end of module:	Module examination with marks. Written exam 100 % (90 min)
Teaching style:	Lectures/Exercises: PowerPoint, Blackboard/Whiteboard, digital content (e.g. Videos), written Exercises, Textbooks Practical classes: written instructions incl. theoretical background, digital teaching formats (virtual Physiology), Other
Literature:	– Current research literature (Peer-reviewed Journals) in Pathophysiology – Braun, CA and Anderson, CM: Applied Pathophysiology - A conceptual approach to the mechanisms of disease. 3rd ed., Wolter Kluwer, 2017. – Norris, T: Porth's Pathophysiology - Concepts of Altered Health States. 10th ed, Wolters Kluwer, 2019. – Silverthorn: Human Physiology - An Integrated Approach. 8th ed., Pearson, 2019.

Module:	**Human Genetics**
Semester:	5. Semester MSc Biomedical Sciences
Course coordinator:	Prof. Dr. Alexander Glassmann/ Dr. Barbara Roitzheim
Lecturer:	Prof. Dr. Alexander Glassmann/ Dr. Barbara Roitzheim
Language:	Englisch
Assignment in Curriculum:	Compulsory Course in 3rd Semester MSc Biomedical Sciences
Course units/Credit hours:	Lecture: 2 credit hours Exercise: 2 credit hours; Group size: 16 Lab work: 2 credit hours; Group size: 16
Student workload:	Contact hours Self study L: 30 30 E: 30 60 P: 30 60 Sum: 90 150 **Sum total: 240 Stunden**
Credits:	8 ECTS
Prerequisites according to examination regulations:	none
Recommendation:	Basic knowledge in Molecular Biology/Genetics from a former BSc (or other basic) curriculum
Learning outcomes:	Students are able to • identify human gene variations using methods of molecular genetic analysis • apply hybridization techniques in tumor diagnosis • analyze food products for their genetic properties • employ chromosome analyses in prenatal and tumor diagnosis • carry out genetic analyses in forensic applications by means of understanding • in the lecture the theoretical basics of human genetics, the correlated disorders, methods to diagnose and to analyze these disorders • in the exercises the theoretical background of analytical investigations to determine human genetic disorders and the description and discussion of human genetic disorders • in the lab course the preparation and banding of chromosomes, fluorescence in situ hybridization (FISH), sex chromatin identification in order to • develop experimental workflows on an introductory level for human genetics applications • understand disease mechanisms based on human genetics disorders • diagnose human genetic disorders
Content:	Lecture: • Mendelian Genetics, extensions to and exceptions from Mendelian Genetics

	• sexual development , sex chromosomes, X-chromosome inactivation, influence of sex on genetic properties, genomic imprinting, • multifactorial traits, behavioral genetics, population genetics, human evolution, genetics of human cancer, • gene therapy and genetic consulting, reproductive technologies, cloning of mammals, ethical issues • selected topics of human genetic research. Exercises: • last lectures related questions • discussion of human genetic based diseases • discussion of theoretical experiments. Practical course: • Cytogenetic and other human genetic analysis methods • karyotype analyses • identification of sex chromosomes, • fluorescence in situ hybridization (FISH) • Analysis of karyotypes in unknown genetic and tumor samples • Experimental analysis of genetic polymorphisms.
Summary indicative content:	Participation in the practical class
Assessment:	Module exam – graded Written test (90 min): 50%; written practical course report 50%
Teaching formats:	L/E: Power Point Presentation, video animations, textbooks P: Script, textbook, lab videos
Textbooks:	• Human Genetics by Ricky Lewis, Mc Graw Hill, 2018 ISBN10: 1260240894 • In Situ Hybridization Protocols, 5th Ed. Edited by Boye Schnack Nielsen Julia Jones Methods in Molecular Biology (2148) ISBN 978-1-0716-0622-3 • Molecular cloning: a laboratory Manual., Vol. 1,2 and 3, 4th Ed, Sambrook, Fritsch., Maniatis, Cold Spring Harbor Laboratory Press, ISBN 978-1-936113-41-5 • Current Protocols in Human Genetics https://currentprotocols.onlinelibrary.wiley.com/journal/19348258?tabActivePane=undefined

Module:	**Advanced and Clinical Immunology (ACI)**
Semester:	3rd Semester
Course coordinator:	Prof. Harald Illges
Lecturer:	Prof. Harald Illges
Language:	English
Assignment in Curriculum:	**Compulsary Course in the 3rd Semester Biomedical Sciences**
Course units/Credit hours:	L: 2 SWS E: 2 SWS, max. group size: 16 Lab work: 2 SWS; group size: max. 16
Student workload:	Contact hours / Self study L: 30 / 30 E: 30 / 60 P: 30 / 60 Sum: 90 / 150 **Sum total: 240 hours**
Credits:	8 ECTS
Prerequisites according to examination regulations:	None
Recommendation:	Basic Immunology, Molecular Biology, Cell Biology
Learning outcomes:	Students are able to • understand immunological mechanisms • can apply immunological knowledge to address questions related to immunological diseases • can apply knowledge to basic experiments • can apply major immunological techniques • are able to analyze basic FACS data by means of understanding • the basic properties of immunological cells • the principles of cell-cell interaction in the immune system • the strategies of cellular analysis • rational of basic immunological experimental techniques in order to • develop experimetal workflows on an introductionary level for immunological applications • understand basic disease mechnanisms in comparison the healthy condition
Content:	<u>Lecture:</u> • FACS analysis of cells at various activation and differentiation stages, cell death, apoptosis.Basics of FACS and MACS technologies, applications thereof. • B cell biology. Differentiation, activation, germinal center reaction. Recombinant antibody production, hybridoma technology. Vaccine development, vaccination. Antibody structure, somatic mutation, epitop, affinity, avidity.

	• T cell immunology, development, cytotoxic, helper, regulatory T cells. Activation, MHC restriction, CAR T cells, tissue culture of T cells, manipulation in tissue culture. • Complement system, complement receptors, immune complexes, immune complex disease, vasculitis, SLE • Allergies, basophil allergen test, vaccine development, antiobody production, purification, immunodeficiennies, gene targetting, knock-out, -in, transgenic mice. Animal research. • Autoimmunity, rheumatoid arthritis, SLE, MS <u>Exercise:</u> questions related to last lectures, discussion of theoretical experiments. <u>Lab course:</u> • Basophil activation test using dust mite and grass pollen allergens. • Sera test using ochterlony technology with immunodeficient sera. • ELISA for glucose 6 phosphate isomerase specific antibodies from a mouse model of rheumatoid arthritis.
Assessment:	Written exam (120 min) – 60% , Laboratory report 40%
Teaching formats:	L/E: Power Point Presentation, video animations, textbooks P: Script, textbook, lab videos
Textbooks**:**	• Janeway Immunobiology, Garland, latest edition. • Case studies in Immunology, Garland, latest edition

Module:	**Final Thesis**
Semester:	**4th Semester MSc Biomedical Sciences**
Course Leader:	Course leaders of the department
Lecturer:	Course leaders of the department
Language:	English
Assignment in Curriculum	**Compulsory course in 4th Sem. MSc Biomedical Sciences**
Course Units/Credit hours	The Master thesis is done either in research groups of the department or in national or international research groups, which offer research activities which match the focus of the study program. During the master thesis, the students are supervised by at least one professor from the department, who also evaluates the final thesis. Details can be found in the examination regulations.
Students workload:	Contact hours: 17,5 weeks, with a work load of 40 hours/week Private study (writing of thesis, preparing of oral presentation, learning for final exam): 5 weeks, with a work load of 40 hours/week **Total Sum: 900 hours**
Credits	30 ECTS
Prerequisites according to examination regulations:	Admission to Master thesis, if not more than two compulsory courses have not been passed. Admission to the Master thesis is regulated under §14 in the Examination Regulations.
Recommendations:	none
Learning outcomes:	The students are able to • solve independently and in a given time complex scientific questions in their special fields of work. • to present their results both literally and orally in an adequate manner. by means of understanding • the practical and theoretical background of the thesis work. • methods to evaluate their scientific data. In order to • demonstrate the ability for independent scientific work • the competence to use theoretical and analytical abilities for the solution of specific scientific questions.
Summary indicative content:	Theoretical and practical work to solve research-related questions using scientific methods. Practical application of knowledge and skills gained during the Master studies, and their accentuation in specific topics. The results have to be summarized in a scientific document, i.e. the Master thesis. The students present their results in a defined time frame, and defend their results in a final oral examination.
Assessment:	Master-Thesis: marked Oral examination: marked
Teaching style:	According to need.
Indicative Bibliography/Sources:	According to need.

Electives

Module:	**Inborn Errors of Metabolism**
Semester:	1st Semester
Course coordinator:	Prof. Jörn Oliver Sass
Lecturer:	Prof. Jörn Oliver Sass
Language:	English
Assignment in Curriculum:	**Elective Course in the 1st Semester MSc Biomedical Sciences**
Course units/Credit hours:	The course comprises lectures and seminar-like sessions/ exercises. L: 2 SWS E: 1 SWS Group size max.16 students.
Student workload:	Contact hours / Self study L: 30 / 15 E: 15 / 30 P: 0 / 0 Sum: 45 / 45 **Sum total: 90 hours**
Credits:	3 ECTS
Prerequisites according to examination regulations:	None
Recommendation:	Clinical Chemistry should have been passed.
Learning outcomes:	Students are able to • understand and use literature and other information on inborn errors of metabolism (IEM) by means of understanding • fundamentals of inborn errors of metabolism in regard of clinical features and underlying pathobiochemistry • analytical methods and strategies in laboratory diagnostics of IEM in order to • use the acquired skills and knowledge as a starting point for work in specialized laboratory diagnostics and for related research
Content:	Concepts of inborn errors of metabolism. Study of processes, exemplary diseases, bioanalytical strategies and methods. Pathobiochemistry.
Assessment:	Not graded, but students need to participate regularly and actively, provide a presentation on a disease/ group of diseases and a handout, pass a quiz.
Teaching formats:	L/E: Power Point presentations, blackboard/ whiteboard, video animations, scientific journal articles, case studies, internet resources, student presentations
Textbooks**:**	• Zschocke/Hoffmann, Vademecum Metabolicum, Geotg Thieme Verlag, recent English edition

	• www.vademetab.org, www.omim.org • various articles from scientific journals

Module:	**Practical Elective: Complement Receptor**
Semester:	**1st semester MSc in Biomedical Sciences**
Course leader:	**Prof. Dr. Harald Illges**
Lecturer:	**Prof. Dr. Harald Illges**
Language:	**English**
Assignment to curriculum:	**Practical elective, 1st MSc in Biomedical Sciences**
Course units / Hours per week:	Laboratory work: 6 credit hours
Student workload:	Contact hours Self study P: 45 45 Sum: 45 45 **Sum total: 90 hours**
Credits:	3 ECTS
Prerequisites according to examination regulations:	None
Recommendations:	None
Learning outcomes:	The students are able to • explain the immunology and molecular biology of complement receptors. by means of understanding • shedding of membrane bound molecules • experimental techniques to analyze CD21 shedding based on actual literature. In order to • perform tissue culture, ELISA, FACS to analyze expression and shedding.
Summary indicative content:	**Laboratory course:** Basics of complement receptor biology/immunology. Redox regulation of shedding, current research questions about CD21-
Assessment:	Active participation demonstrated by protocol.
Teaching style:	Combination of theory and practical work in the laboratory.
Indicative bibliography/Sources:	Current scientific literature

Module:	**Advanced Bioinformatics**
Semester:	**1st Semester MSc Biomedical Sciences**
Course leader:	**Dr. Kurt Stüber**
dito	**Dr. Kurt Stüber**
Language:	**English**
Assignment to curriculum:	**Practical Elective 1st Semester MSc Biomedical Sciences**
Course units / Hours per week:	Laboratory work: 6 credit hours
Student workload:	Contact hours / Private study Lab work: 45 / 45 **Total study hours: 90 hours**
Credits:	3 ECTS
Prerequisites according to examination regulations:	None
Recommendations:	None
Learning outcomes:	The students are able • Learn basic principles of data analysis • Get an insight in data bases and commonly used software By means of understanding • data base searches • methods of sequence comparisons • pattern recognition • They are familiar with and know secondary data bases (for instance gene ontology, biochemical pathways and taxonomical data). In order to • Construct evolutionary trees • Do gene prediction, • To perform prediction of secondary and tertiary structure of proteins and nucleic acids.
Summary indicative content:	The course will take place in one of the computer rooms of the institutes. Students have access to the internet and are able to use preinstalled software on the institute servers. Only non-commercial free software will be used.
Assessment:	The course is not graded. There is a written exam (60 min) at the end of the semester.
Teaching style:	PowerPoint presentations, live computer demonstrations, blackboard.
Indicative bibliography/Sources:	Introduction to Bioinformatics. Arthur M. Lesk (2006) Oxford University Press or comparable text books

Module:	**Practical Elective: Fundamentals of Membrane Transport**
Semester:	**1. Semester MSc Biomedical Sciences**
Course leader:	**Prof. Dr. Christopher Volk**
Lecturer:	**Prof. Dr. Christopher Volk**
Language:	**English**
Assignment to curriculum:	**Practical Elective 1st Semester MSc Biomedical Sciences**
Course units / Hours per week:	Laboratory work: 6 credit hours
Student workload:	Contact hours / Private study Lab work: 45 / 45 **Total study hours: 90 hours**
Credits:	3 ECTS
Prerequisites according to examination regulations:	none
Recommendations:	none
Learning outcomes:	The students are able to • understand the principle mechanisms of membrane transport and recognize the different types of transporters and channels by means of understanding • oocytes of Xenopus laevis as a tool for the expression of heterologous proteins in order to • perform measurements of membrane potentials and membrane currents using a two-electrode voltage-clamp system
Summary indicative content:	The students will inject RNA of membrane transport proteins into Xenopus oocytes to express the proteins. Subsequently, they will use these oocytes to perform voltage-clamp measurements of substrate-induced currents and analyze the obtained data. In an accompanying seminar the theoretical background will be presented by the lecturer and short presentations of scientific papers will be given by the students.
Assessment:	The mode of examination is announced by the beginning of the semester. No grading in this course.
Teaching style:	The unit consists of a 6 SWS practical course, including an accompanying theoretical seminar.
Indicative bibliography/Sources:	The Axon Guide http://www.culturacientifica.org/textosudc/Axon_Guide.pdf

Module:	**Next Generation Sequencing**
Semester:	2nd Semester
Course coordinator:	Prof. Dr. Harald Illges / Dr. Julia Holtel
Lecturer:	Dr. Julia Holtel
Language:	English
Assignment in Curriculum:	**Elective Course in the 2nd Semester Biomedical Sciences**
Course units/Credit hours:	3 SWS, max. group size: 8
Student workload:	Contact hours / Self study L: 15 / 15 E: 15 / 15 P: 15 / 15 Sum: 45 / 45 **Sum total: 90 hours**
Credits:	3 ECTS
Prerequisites according to examination regulations:	None
Recommendation:	Molecular Biology, Microbiology, Human Biology
Learning outcomes:	Students are able to • differentiate different types of NGS methods • analyze NGS metagenomics sequence files by means of understanding • understand fundamental principles of NGS in order to • design a basic 16S-rRNA metagenomics experiment
Content:	Lectures: • Fundamentals of NGS: historical background of NGS, key principes of NGS (library, flow cell, library immobilisation and bridge amplification, clusters, reversibly modified nucleotides, sequencing by sythesis principle), Illumina's NGS workflow, single-end and paired-end sequencing, Miseq applications (metagenomics, small genome sequencing, amplicon sequencing), NGS library preparation for metagenomics • Assessing qualty of sequence runs and sequence data. Programs pre-installed on Miseq: Miseq control software (MCS), real-time analysis software, sequence analysis viewer (SAV) and Miseq reporter, run folder structure, MCS for primary quality assessement, Sequence Analysis Viewer (SAV) for detailed quality assessment (SAV files, SAV terminology, SAV interface and SAV functionalities, typical problems with Miseq runs (under- and overclustering, low diversity libraries, diagnosis and troubleshooting) • 16S-rRNA metagenomic sequencing: data analysis in Geneious. Geneious metagenomic workflow, pre-processing metagenomic data, FASTAQ files, Paired-end reads and importing, trimming, Merging, Length correction, operational taxonomic units (OTUs), OTU clustering, parameters for OTU clustering, BLAST, classification of

	results, parameters for classification, stringent and less stringent conditions • 16S-rRNA metagenomic sequencing: data analysis in CLC Genomics. CLC Genomics metagenomics workflow, comparison with the one of Geneious, pre-processing metagenomic data, importing, trimming, OUT reference database, OTU clustering, parameters for OTU clustering, alpha diversity (species richness and phylogenetic diversity) and beta diversity (Bray-Curtis, Unifrac) • Shotgun metagenomic sequencing: data analysis in CLC Genomics. Limitations of 16S-rRNA metageomics, definition of shotgun metagenomics, library preparation aspects, the mechanism of tagmentation reaction, two strategies for data analysis (taxonomic, functional), workflow for shotgun sequence taxonomy analysis, genome reference database, taxonomic report, abundance and merged abundance tables. • Whole genome sequencing. WGS definition, applications, library preparation aspects, WGS data analysis, reference assembly, de novo assembly, estimating quality of assembly, N50, coverage, approaches for sequence assembly (overlap and de Brujn graphs) • Whole genome sequencing. WGS in clinical microbiology, evolution of pathogenicity in microbial pathogens, outbreak definition, outbreak investigation, traditional typing and phenotyping (MLST, PFGE, antibiograms), Center for Genomic Epidemiology (CGE) tools, WGS-based typing and phenotyping (cgMLST, SNP-based typing, in silico antibiograms) • Oral presentations of CGE tools: NDtree, PathogenFinder, PHASTER, ResFinder, SSCMecFinder, VirulenceFinder, PlasmidFinder Exercises: • Comparison of hypervariable regions of the 16S-rRNA gene in Geneious • Evaluation of NGS runs with Sequence Analysis Viewer • Analysis of 16S-rRNA metagenomics data in Geneious • Analysis of 16S-rRNA metagenomics data in CLC Genomics • Analysis of shotgun metagenomics data in CLC Genomics • Analysis of whole genome sequencing data in Geneious • Analysis of WGS data with CGE tools: in silico MLST, cgMLST, wgMLST Lab course: • 16S-rRNA amplification via qPCR • Index PCR and library purification with magnetic beads • Library quantification, normalization & pooling • Library denaturation, dilution, Miseq run
Assessment:	Oral presentation 20 min - 30% , homeworks – 30%, Lab course report 40%
Teaching formats:	L/E: Power Point Presentations, exercise scripts P: lab course script

Module:	**Stem Cells**
Semester:	**3rd Semester MSc Biomedical Sciences**
Course leader:	**Prof. Dr. Edda Tobiasch**
Lecturer:	**Prof. Dr. Edda Tobiasch**
Language:	**English**
Assignment to curriculum:	**Special Field 3rd Semester MSc Biomedical Sciences**
Course units / Hours per week:	Lecture: 2 credit hours Tutorial: 2 credit hours Laboratory work: 2 credit hours
Student workload:	Contact hours / Private study Lecture: 15 / 15 Tutorial: 15 / 15 Lab work: 15 / 15 Total: 45 / 45 **Total study hours: 90**
Credits:	3 ECTS
Prerequisites according to examination regulations:	none
Recommendations:	Knowledge in cell culture from previous study courses General Safety Instructions for working in laboratories Safety Instructions for working with S1 organisms
Learning outcomes:	Lecture: students are able • discriminate between embryonic and adult stem cells • evaluate the sources of stem cells • learn about ethical aspects of working with stem cells by means of understanding • markers for differentiation of lines • detection methods of various differentiation lines In order to • To perform work in laboratories of industries and universities, which focus on the development of stem cell therapies with only a short training period for adjustments.
Summary indicative content:	Lecture: The lecture focuses on the following questions and aspects: stem cells vs. progenitor cells, embryonic vs. adult stem cells: advantages and disadvantages, sources for stem cells, iPS, plasticity and potency, differentiation and transdifferentiation, isolation and purification, differentiation lines and markers, line-specific staining Tutorial: Each student has to present a paper which is related to his/her specific project and discuss the content with respect to their own current data and adjust the project, if applicable. Ethical aspects will be discussed. The data will be presented at an international or national conference if enough scientific results can be achieved.

	Laboratory course: Each student will have an own part of a scientific project to work on. This subproject will be part of a doctoral thesis or will partially overlap with the other subprojects. All components together will be a complete scientific project.
Assessment:	The module is graded. The mode of assessment will be announced by the beginning of the module. Active participation and attendance is required to pass.
Teaching style:	Lecture: PowerPoint, Overhead, black board Tutorial: paper, PowerPoint, black board
Indicative bibliography/Sources:	Turksen, Kursad: Adult stem cells, Humana Press Sell, Stewart: Stem cells handbook, Humana Press Chiu, Arlene Y.: Human embryonic stem cells, Humana Press Artmann G.M., Hescheler J., Minger S.: Stem Cell Engineering, Springerverlag Paolo Di Nardo: Adult Stem Cell Standardization, River Publishers Kasper, Cornelia; Witte, Frank; Pörtner, Ralf: Tissue Engineering III: Cell – Surface interactions for Tissue Culture, Springer-Verlag

Module:	**Introduction to Flow Cytometry**
Semester:	**3rd semester MSc Biomedical Sciences**
Course leader:	**Prof. Dr. Harald Illges**
Lecturer:	**Prof. Dr. Harald Illges**
Language:	**English**
Assignment to curriculum:	**Practical elective, 3rd semester MSc Biomedical Sciences**
Course units / Hours per week:	Laboratory work: 6 credit hours
Student workload:	Contact hours Private study Lab work: 45 45 **Total study hours: 90 hours**
Credits:	3 ECTS
Prerequisites according to examination regulations:	none
Recommendations:	none
Learning outcomes:	Laboratory course: The students are able to • Get hands-on experience with FACS machines • Run routines and use specific software to run FACS machines by means of understanding • statistical evaluation of FACS data • to evaluate and organize FACS data in order to • run maintenance routines for the FACS machine (basics) • perform FACS measurements independently
Summary indicative content:	Laboratory course: The students will be introduced to the theoretical basis of the FACS technology. They will understand the basis of the FACS machines and technology and the used principles of fluidics, optics/lasers and electronics, understand to run the machines and apply the CellQuest program (used for setup and steering the machine), calibration, recording of data, multiparameter analysis of 4 color experiments statistical evaluation of single and multicolor stained cell, dotplot and histogram evaluation of data, DNA/cell cycle experiments.
Assessment:	Active participation demonstrated by protocol.
Teaching style:	Combination of theory and practical work in the laboratory.
Indicative bibliography/Sources:	Handbook of flow cytometry methods, Robinson (Editor) A guide to Fluorescent Probes and Labeling Technologies, 10. Edition, Molecular Probes

Module:	**Digital Imaging and Analysis – Principles and Applications of Confocal Microscopy**
Semester:	3rd Semester
Course coordinator:	Prof. Dr. Harald Illges / Dr Zhanlu Ma-Högemeier
Lecturer:	Dr. Zhanlu Ma-Högemeier
Language:	English
Assignment in Curriculum:	**Elective Course in the 3rd Semester Biomedical Sciences**
Course units/Credit hours:	3 SWS, max. group size: 6 (Students from international partner universities may join virtually, max. group size: 6)
Student workload:	Contact hours / Self study L: 10 / 10 E: 10 / 10 P: 25 / 25 Sum: 45 / 45 **Sum total: 90 hours**
Credits:	3 ECTS
Prerequisites according to examination regulations:	None
Recommendation:	Microscopy, Confocal Microscopy, Cell Biology, Molecular Biology, Biochemistry
Learning outcomes:	Students are able to • learn fundamental principles of confocal microscopy • learn fundamental principles of digital imaging • learn fundamental principles of basic and advanced imaging analysis by means of understanding • application method and evaluation of images in order to • apply basic and advanced digital imaging • apply digital imaging for digital analysis purposes • perform basic and advanced imaging analysis • perform mammalian cell culture maintenance and transient transfections
Content:	Confocal microscopy offers advantages over conventional wide field optical microscopy and becomes a powerful tool in life science research. This course introduces principles and applications of the confocal microscopy. In combination with live fluorescent proteins and synthetic fluorescent probes, basic and advanced digital imaging (live cell imaging and fixed cell imaging) would be performed. All participants will have intensive hands-on training with cell culture and digital imaging processing, as well as basic and advanced imaging analysis. <u>Lectures:</u> • Fundamentals of confocal microscopy

	• Fundamentals of digital imaging • Cell culture and transfection • Fluorescent proteins and sythetic fluorescent probes • Basic digital imaging processing and analysis • Advanced digital imaging processing and analysis <u>Exercises:</u> • Oral presentations on selected peer-reviewd publications • Perform basic and advanced imaging analysis <u>Lab course:</u> • Mammalian cell culture maintenance • Perform transient transfection of mammalian cells in vitro aiming to the overexpression of exogenous proteins in mammalian cells • Select and utilize live fluorescent proteins as well as synthetic fluorescent probes according to diverse purposes • Apply basic digital imaging, including single channel imaging, simultaneous and sequential dual channel imaging, 3D imaging • Apply advanced digital imaging, including sequential multi-channel imaging, 3D reconstruction, time lapse imaging • Apply digital imaging for analysis purposes, for example colocalization analysis, protein-protein interaction analysis
Assessment:	Lab course performance - 40%, Oral presentation - 30% , Lab course report - 30%
Teaching formats:	L/E: Power Point Presentations, exercise materials P: lab course script