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Alisa G. Woods
Costel C. Darie *Editors*

Advancements of Mass Spectrometry in Biomedical Research

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Editors

Advancements of Mass Spectrometry in Biomedical Research

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To Professor Dr. Vlad Artenie
(University of Iasi, Romania)
and the late Professor Dr. Wolfgang Haehnel
(University of Freiburg, Germany),
as well as the many supporters
of the Darie Lab. Also to Kenneth R. Woods, PhD, an inspirational biochemist

Foreword

The use of mass spectrometry is becoming increasingly important for biomedical research and for clinical applications. Detection of exogenous substances, such as toxins, can be performed by mass spectrometry and was perhaps the initial use of this technology in biomedicine. However, identification of endogenous, disease-related molecules is also possible. With the advent of the genetic revolution, the proteomic revolution has followed in close succession. Mass spectrometers are essential for proteomic discovery, and other "omic" fields (such as glycomics, metabolomics, lipidomics, and many more) are exploding with new information. The sensitivity of mass spectrometers and increasingly more sophisticated bioinformatics tools are opening up this field to untold possibilities for biomedical researchers and clinicians.

In the spirit of this revolution in biomedicine, we have assembled this comprehensive work, which largely focuses on the application of mass spectrometry to "omics" analysis in biomedicine. We start with broad descriptions of the field, definitions of the machinery, and then delve into the various methods and approaches that can be utilized. We consider aspects of molecular analysis and then discuss how mass spectrometry can be applied to our understanding of specific diseases and disorders.

Numerous MS-based methodologies are now available to researchers, and this text reviews many cutting-edge and relevant technologies. Dudley focuses on the application of matrix-assisted laser desorption/ionization (MALDI) in biomedicine, while colleagues from Waters Corporation examine how peak capacity can be best maximized. Brown describes quantitative shotgun proteomics with data-independent acquisition and traveling wave ion mobility spectrometry as a versatile life-science tool, while Hoedt and coworkers explore the use of stable isotope labeling by amino acids in cell culture (SILAC) for protein quantification using mass spectrometry. A chapter by Roy and coworkers discusses the complementary use of computational structural biology in mass spectrometry.

Different aspects of proteins and other molecules can be studied using MS. Petre explores protein structures and interactions, while Zamfir has tackled the use of mass spectrometry to understand gangliosides. Ngounou Wetie et al. examine the

analysis of protein posttranslational modifications and protein–protein interactions, and Samways focuses on mass spectrometry-based analysis of ion channel structure and function. Budayeva and Cristea further expand upon how protein–protein interactions can be understood via mass spectrometry, while Ferguson et al. focus on its use for understanding small molecules. Ckless elucidates how redox proteomics can be studied in the lab, for eventual application to clinical uses, and Luck looks at fluorinated proteins. Baral et al. explore mass spectrometry analysis using a model animal system, the zebrafish, while Miller and Spellman describe the workflow for biomarker discovery in a pharmaceutical company.

Focusing on health promotion, Andrei and colleagues examine how mass spectrometry can be used to analyze biomedically relevant stilbenes from wine. With regard to increased understanding of diseases and disorders, chapters by Monien, Schneider, Patel, and Sandu describe the use of mass spectrometry for analysis in cancer. Topics covered include quantifying DNA adducts, analysis of breast milk, apoptosis and cancer secretome analysis as identification of heat shock response. In the field of infectious disease, Branza-Nichita and colleagues use mass spectrometry to study the HBV life cycle, while Marrakchi et al. look at oxidative stress and antibiotic resistance in bacterial pathogens.

Deinhardt examines how mass spectrometry can be used to understand neuronal signaling, which could apply to numerous neurological and psychiatric conditions, while Woods et al. study how mass spectrometry can facilitate the understanding of a novel central nervous system protein. We then explore more applied uses of mass spectrometry in the central nervous system: specifically, how biomarker discovery may be directly performed for neurodevelopmental disorders and be used to understand and potentially diagnose depression.

In the realm of therapeutics, Heckman and coworkers have used mass spectrometry to localize and analyze the efficacy of nanoceria, a potential delivery system for a variety of medical conditions. We finally end with a particularly important chapter on bottlenecks in proteomics, topics that are encountered by almost all researchers but that are almost never discussed in publications.

We thus present to the reader a comprehensive text, examining the many uses of mass spectrometry in biomedicine, with the hope that this will be useful to both researchers and clinicians. As this exciting field further expands, so will the potential applications for using mass spectrometry to understand medical issues and to address them through exploration, as well as eventual clinical prognosis, diagnosis, and monitoring. We look forward to an exciting era of MS-based discovery and application.

Potsdam, NY

Alisa G. Woods
Costel C. Darie

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Chapter 1
Mass Spectrometry for Proteomics-Based Investigation

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Abstract Within the past years, we have witnessed a great improvement in mass spectrometry (MS) and proteomics approaches in terms of instrumentation, protein fractionation, and bioinformatics. With the current technology, protein identification alone is no longer sufficient. Both scientists and clinicians want not only to identify proteins but also to identify the protein's posttranslational modifications (PTMs), protein isoforms, protein truncation, protein–protein interaction (PPI), and protein quantitation. Here, we describe the principle of MS and proteomics and strategies to identify proteins, protein's PTMs, protein isoforms, protein truncation, PPIs, and protein quantitation. We also discuss the strengths and weaknesses within this field. Finally, in our concluding remarks we assess the role of mass spectrometry and proteomics in scientific and clinical settings in the near future. This chapter provides an introduction and overview for subsequent chapters that will discuss specific MS proteomic methodologies and their application to specific medical conditions. Other chapters will also touch upon areas that expand beyond proteomics, such as lipidomics and metabolomics.

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Abbreviations

BN-PAGE	Blue native PAGE
CI	Chemical ionization
CN-PAGE	Colorless native PAGE
DIGE	Differential gel electrophoresis
EI	Electron ionization
ESI	Electrospray ionization
ESI-MS	Electrospray ionization mass spectrometry
FT	Fourier transform
IT	Ion trap
LC–MS/MS	Liquid chromatography–mass spectrometry
m/*z*	Mass/charge
MALDI	Matrix-assisted laser desorption ionization
MALDI-MS	MALDI mass spectrometry
MS	Mass spectrometry
Mw	Molecular weight
Q	Quadrupole
SDS-PAGE	Sodium dodecyl sulfate-polyacrylamide gel electrophoresis
TIC	Total ion current/chromatogram
TOF	Time of flight

1.1 Introduction

Proteomics is the large scale study of the protein complement, also known as the proteome. Proteomics is studied through mass spectrometry (MS) [1–8]. MS can be used to investigate a large variety of chemical and biological molecules, including products of chemical synthesis or degradation, biological molecules such as proteins, nucleic acids, lipids, or glycans, or various natural compounds of either large or small molecular mass. Depending on what type of molecule is being analyzed, there are various types of MS focus, such as small-molecule MS, large-molecule MS, and biological MS (when the molecules investigated are biomolecules). Within biological MS, there are also different MS subfields, such as proteomics, lipidomics, glycomics, and metabolomics. The focus of proteomics is to analyze proteins and protein derivatives (such as glycoproteins), peptides, posttranslational modifications (PTMs) within proteins, or protein–protein interactions (PPIs).

The standard workflow in a proteomics experiment starts with sample fractionation, involving the separation of proteins prior to their analysis by MS [9–17]. This can be done by one or more biochemical fractionation methods. For example, a one-dimensional separation can be achieved by sodium dodecyl sulfate-polyacrylamide gel electrophoresis (SDS-PAGE); a two-dimensional separation can be performed by two-dimensional electrophoresis or by affinity purification followed by

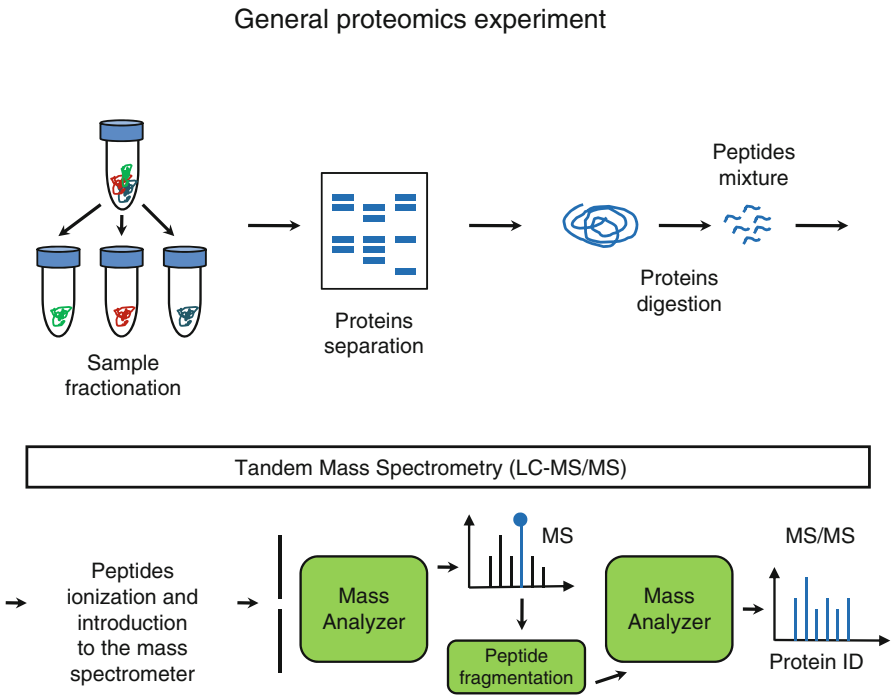

Fig. 1.1 General proteomic experiment workflow schematic. Reprinted and adapted with permission from the *Australian Journal of Chemistry CSIRO Publishing* http://www.publish.csiro.au/?paper=CH13137 [15]

SDS-PAGE. Biochemical fractionation is then followed by enzymatic digestion (usually trypsin), peptide extraction, and peptide fractionation by HPLC and MS analysis [1]. Data analysis leads to identification of one or more proteins and further simultaneous investigation or re-investigation of the results can extract additional information from the same MS experiment, such as PTMs and interaction partners of some proteins (PPIs) [18–26]. A schematic of a proteomics workflow is shown in Fig. 1.1 and a schematic of a proteomics experiment is shown in Fig. 1.2a.

Proteomic analysis can be performed using samples from various sources such as supracellular, subcellular, intracellular, or extracellular, as well as at the peptide level (peptidomics), protein (regular proteomics), PTMs ("PTM-omics"), or protein complex level (interactomics). Proteomics can also be classified as classical or functional, when one analyzes protein samples from two different conditions (for example, normal and cancer), and targeted proteomics, when one focuses on a particular sub-proteome, such as phosphoproteomics or glycoproteomics. Proteomics can also be classified based on the protein complement from a set of samples that is being analyzed such as proteomes (i.e., all proteins) or sub-proteomes (i.e., just the nuclei or mitochondria). A schematic of such classification is shown in Fig. 1.2b.

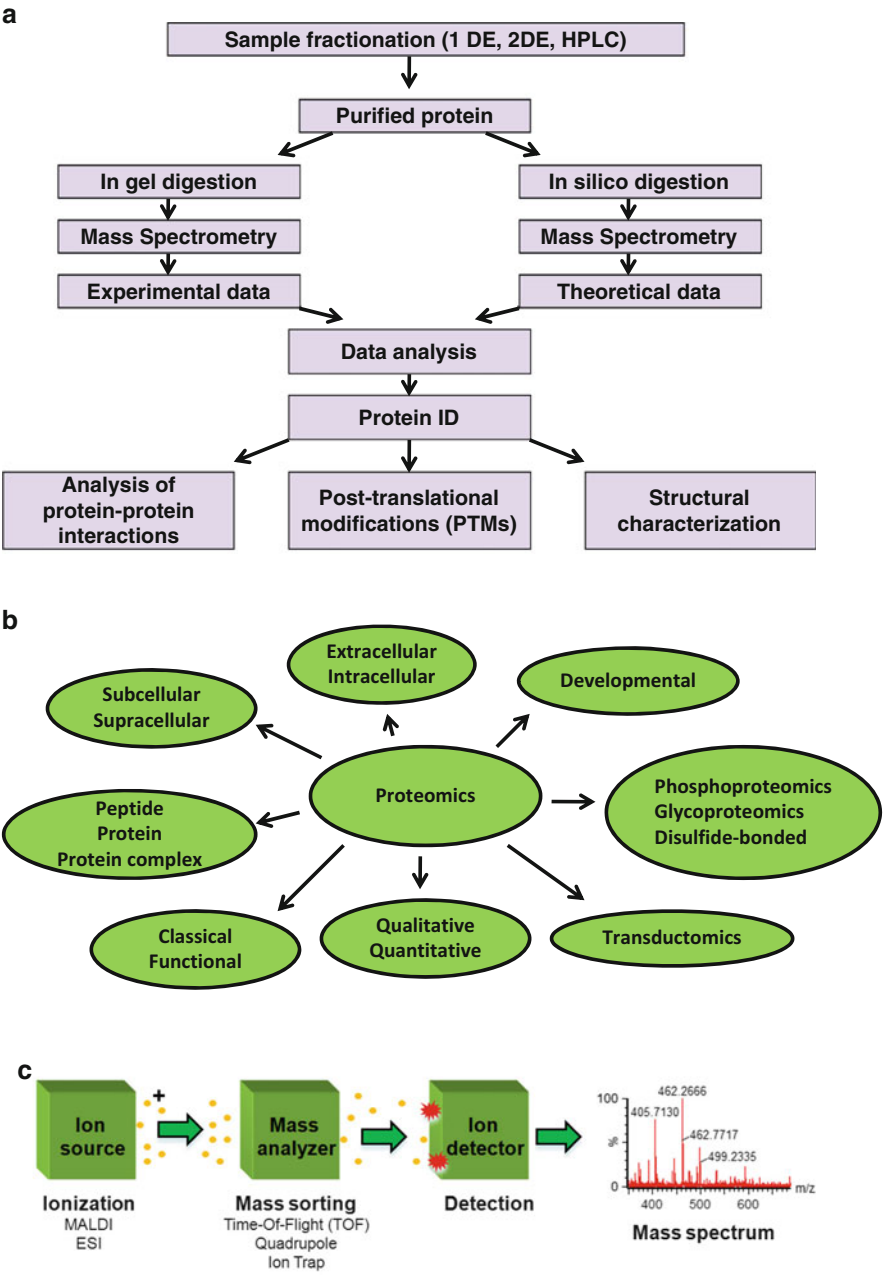

Fig. 1.2 General proteomics experiment. (**a**) Proteomics experiment workflow schematic. (**b**) Proteomics and applications schematic. (**c**) Mass spectrometer schematic. Reprinted and adapted with permission from the *Oxidative Stress*: *Diagnostics*, *Prevention*, *and Therapy*, S. Andreescu and M. Hepel, Editors. 2011, American Chemical Society: Washington, D.C [16]

Proteomic analysis can also focus on quality such as for protein identification, or the determination of protein amounts by quantitative proteomics. These analyses are usually performed using a mass spectrometer, the "workhorse" in a proteomics experiment. A mass spectrometer has three main components: the ionization source, a mass analyzer, and a detector (Fig. 1.2c). There are primarily two types of ionization sources on mass spectrometers: matrix-assisted laser desorption ionization (MALDI) and electrospray ionization (ESI). The mass spectrometers are consequently named MALDI mass spectrometry (MALDI-MS) and electrospray ionization mass spectrometry (ESI-MS). Here, we describe a proteomics experiment, specifically how proteins and peptides are analyzed by MS. We also describe the type of information that can be obtained from such an experiment.

1.2 Biochemical Fractionation

The first step in a proteomics experiment is biochemical fractionation, in which various proteins are separated from each other using their physicochemical properties. Biochemical fractionation usually depends on the goal of the experiment and it is perhaps the most important step in a proteomics experiment. A good sample fractionation usually leads to a good experimental outcome. A proteomics experiment can still be performed without biochemical fractionation, for example, when one analyzes the full proteome of a cell at once. However, without biochemical fractionation, the results in a proteomics experiment may not necessarily be optimal.

The physicochemical properties of proteins (or compounds of interest) that are used to achieve biochemical fractionation are, among others, molecular mass, isoelectric point, charge at various pH, and the protein's affinity to other compounds. These properties of the proteins are well exploited by biochemical fractionations such as electrophoresis, centrifugation, and chromatography. Types of chromatography can include affinity chromatography, ion exchange chromatography, and size-exclusion chromatography.

To give one example, proteins can be separated by electrophoresis, usually SDS-PAGE, reduced and denatured, and then separated according to their molecular mass. If the reduction step is not used, the disulfide bridges in a protein or between proteins remain intact, thus providing an additional fractionation principle: two proteins with low molecular mass (such as haptoglobin subunits) are kept together through disulfide bridges and are separated under SDS-PAGE under nonreducing conditions as a heterotetramer with a high molecular mass. In a different variant of SDS-PAGE, but not using the detergent (SDS), one may separate proteins under native conditions. Therefore, simply by adding one reagent (for example, SDS) or two (SDS and a reducing agent like dithiothreitol or DTT), separation of these proteins may have a totally different outcome. A variant of SDS-PAGE is tricine-PAGE [27, 28], which has a principle of separation similar to the SDS-PAGE, but it has the highest separating resolution in the low molecular weight (Mw) proteins and peptides (2–20 kDa), where SDS-PAGE has poor or very poor resolution. Therefore, SDS-PAGE and tricine-PAGE complement each other.

Other types of electrophoresis are blue native PAGE (BN-PAGE), colorless native PAGE (CN-PAGE), and detergent-less SDS-PAGE (native PAGE) [1, 4, 6, 18–22, 29–34], all native electrophoresis. BN-PAGE separates protein complexes by using the external charge induced by Coomassie dye; thus, the complexes will have the same charge and will separate according to their molecular weight. If the Coomassie dye is not used, the external charge is not induced and the separation does not take place according to the molecular weight of the complexes, but rather according to the internal charge of the protein complexes. This method, a variant on BN-PAGE, is named CN-PAGE. CN-PAGE is particularly useful when two protein complexes with identical mass must be separated from each other.

In addition to the techniques mentioned for biochemical fractionation, hyphenated techniques may also be used. The classical example is two-dimensional electrophoresis (2D-PAGE), which includes separation of proteins by isoelectric focusing and by SDS-PAGE [3, 7, 35–45], still used in some proteomics labs. In fact, a variant of 2D-PAGE is differential gel electrophoresis (DIGE), a powerful method for gel-based proteomics. Other fractionation methods such as pre-coated chips, centrifugal filters, and magnetic beads are also possible [46, 47].

1.3 Mass Spectrometry

A mass spectrometer has three main parts: an ion source, a mass analyzer, and a detector. Initially, the sample is ionized and the ions produced by MALDI or ESI source are separated in the mass analyzer based on their mass-to-charge (m/z) ratio. The ions are then detected by the detector. The end product is a mass spectrum, which is a plot of ion abundance versus m/z.

Ionization sources. Ionization of peptides is dependent on the electrical potential at the ion source and on the pH at which they are analyzed. At low pH, the peptides are protonated through the amino-containing amino acids such as Arg or Lys, while at high pH, the peptides are de-protonated through the carboxyl-containing amino acids such as Asp or Glu. When the electrical potential at the ion source is positive, ionization is in positive ion mode. Conversely, when the electrical potential is negative, ionization is in negative ion mode. Therefore, there are two types of ionization: positive, when peptides are analyzed at low pH and the Arg, Lys, and His are protonated, and negative ionization, when peptides are analyzed at high pH and the Asp and Glu are de-protonated. In the current chapter, we will focus only on positive ionization, because it is one of the most used ionization modes for analyzing peptides and proteins. In addition, the enzyme that is the most widely used in proteomics is trypsin which cleaves conveniently at the C-terminus of Arg and Lys and produces peptides that are, upon ionization, at least doubly charged (the peptide and the C-terminal amino acid) and produces a y product ion series upon collision-induced fragmentation (described later).

In addition to ESI and MALDI, there are several additional ionization methods, such as chemical ionization (CI), electron ionization (EI), or atmospheric pressure

chemical ionization (APCI) [48, 49]. EI is used for analysis of organic compounds and can be used for all volatile compounds with a mass smaller than 1,000 Da. EI provides good structural information derived from fragmentation. However, molecular mass determination is rather poor (poor signal or the absence of M^+ ions) [50]. Chemical ionization is the opposite: it is very good for the determination of the molecular mass of molecules, but it is not very good in providing structural information due to reduced fragmentation in comparison to EI. Therefore CI and EI could complement each other. In CI experiments, ionized species are formed when the gaseous molecules to be analyzed collide with primary ions present in the source under a high vacuum [51]. A variant of CI is negative CI used only for volatile analytes with a mass of less than 1,000 Da [52, 53]. Another ionization technique, APCI, is an alternative for analysis of compounds that do not ionize in ESI. During APCI, generally only singly charged ions are formed and it is usually applied to compounds with a molecular weight of less than 1,500 Da [54].

Mass analyzers. There are three main types of mass analyzers used for proteomics experiments: trapping type instruments (quadrupole ion trap—QIT, linear ion trap—LIT, Fourier transform ion cyclotron resonance—FT-ICR, and Orbitrap), quadrupole (Q), and time of flight (TOF) instruments.

Trapping type instruments first accumulate ions and then allow for mass measurement. The ion trap analyzers first capture ions in three-dimensional space (trap), and then electrostatic gate pulses to inject ions into the ion trap. The ion trap-based analyzers are relatively inexpensive, sensitive, and robust. They have been extensively used in proteomic analysis. However, a problem with these instruments is their accuracy for both precursor and product ions, partially overcome by an FT-ICR. Unfortunately, this instrument is not very often used in proteomics research because peptides do not fragment well and the instrument is expensive [55, 56].

In quadrupole mass analyzers, ions constantly enter the analyzers, which are separated based on their trajectory in the electric field applied to two pairs of charged cylindrical rods. There is an electric potential between each pair of rods drawing the ions towards one rod. These instruments provide good reproducibility and low cost, but their resolution and accuracy are limited [49, 57].

Instruments with TOF mass analyzers are popular for sample analysis in proteomics due to their high resolution and relatively low cost, speed of measurements, and high mass accuracy [49, 57]. In TOF mass analyzers, ions are accelerated by a known electric field and then travel from the ion source to the detector. The instrument measures the time it takes for ions with different masses to travel from the ion source to detector,

Mass spectrometers can have stand-alone analyzers or in combination, usually two or three analyzers within one instrument, thus taking advantage of the strength of all combined analyzers simultaneously. Examples of such instruments are Q-Trap, QQQ, Q-TOF, TOF–TOF, QQ-LIT; these instruments are also called hybrid mass spectrometers, and are highly sensitive and also have a high resolution [1, 57–59].

MS detectors. The MS detectors are usually electron multipliers, photodiode arrays, microchannel plates, or image current detectors.

1.4 MALDI-TOF MS

MALDI-TOF MS or MALDI-MS (Fig. 1.3a) is mostly used for determination of the mass of a peptide or protein and for identification of a protein using peptide mass fingerprinting. In MALDI-MS, the peptide mixture is co-crystallized under acidic conditions with a UV-absorbing matrix (for example, dihydrobenzoic acid, sinapinic acid, alpha-hydroxycinnamic acid) and spotted on a plate. A laser beam (usually nitrogen; 337 nm) then ionizes the matrix and peptides, which desorb and start to fly under an electrical field. The matrix molecules transfer a proton to peptides, which then become ionized, fly through the TOF tube, and are detected in the detector as a mass spectrum. Charged peptides fly through the mass analyzer as ions according to their mass-to-charge ratio (m/z) and to the formula: $[M+zH]/z$, where M is the mass of the peptide and z is the charge of the peptide; H is the mass of hydrogen (1.007825035 atomic mass units). In MALDI-MS analysis, the charge of peptides is almost always +1 and the peptides are mostly observed as singly charged; the formula is then $[M+1\times1]/1$ or $[M+1]/1$ or $[M+1]$. Therefore, the peptides are mostly detected as singly charged peaks or [MH]+ peaks (Fig. 1.3b).

In the MALDI-MS mass spectrum, one peak corresponds to one peptide and many peaks correspond to many peptides, either from one protein or from more proteins. Database search of the MALDI-MS spectra usually identifies that single protein or those proteins through a process named peptide mass fingerprinting (Fig. 1.3c).

1.5 ESI-MS

In contrast to MALDI-MS, in which peptides are ionized with the help of a matrix (and are in the solid phase), in ESI-MS (Fig. 1.4a) peptides are ionized in the liquid phase, under high electrical current. Also, while in MALDI-MS peptides are mostly singly charged, in ESI-MS peptides are mostly double or multiple charged. Regarding the ionization method, peptides fly as ions according to m/z and calculation of the molecular mass of the peptide is performed according to the same $[M+z]/z$ formula, where z is again the charge (z is 2 for doubly charged peptides, 3 for triply charged peptides, etc.).

When a peptide mixture is injected into the mass spectrometer, all or most peptides that ionize under the experimental conditions are detected as ions in an MS

Fig. 1.3 (continued) peptide mixture is analyzed by MALDI-MS and a spectrum is collected. A similar experiment is performed in silico (a theoretical experiment in computer), but the cleavage is performed in all proteins from a database. During the database search, the best match between the theoretical and the experimental spectra then lead to identification of a protein. Reprinted and adapted with permission from the *Oxidative Stress*: *Diagnostics*, *Prevention*, *and Therapy*, S. Andreescu and M. Hepel, Editors. 2011, American Chemical Society: Washington, D.C [16]

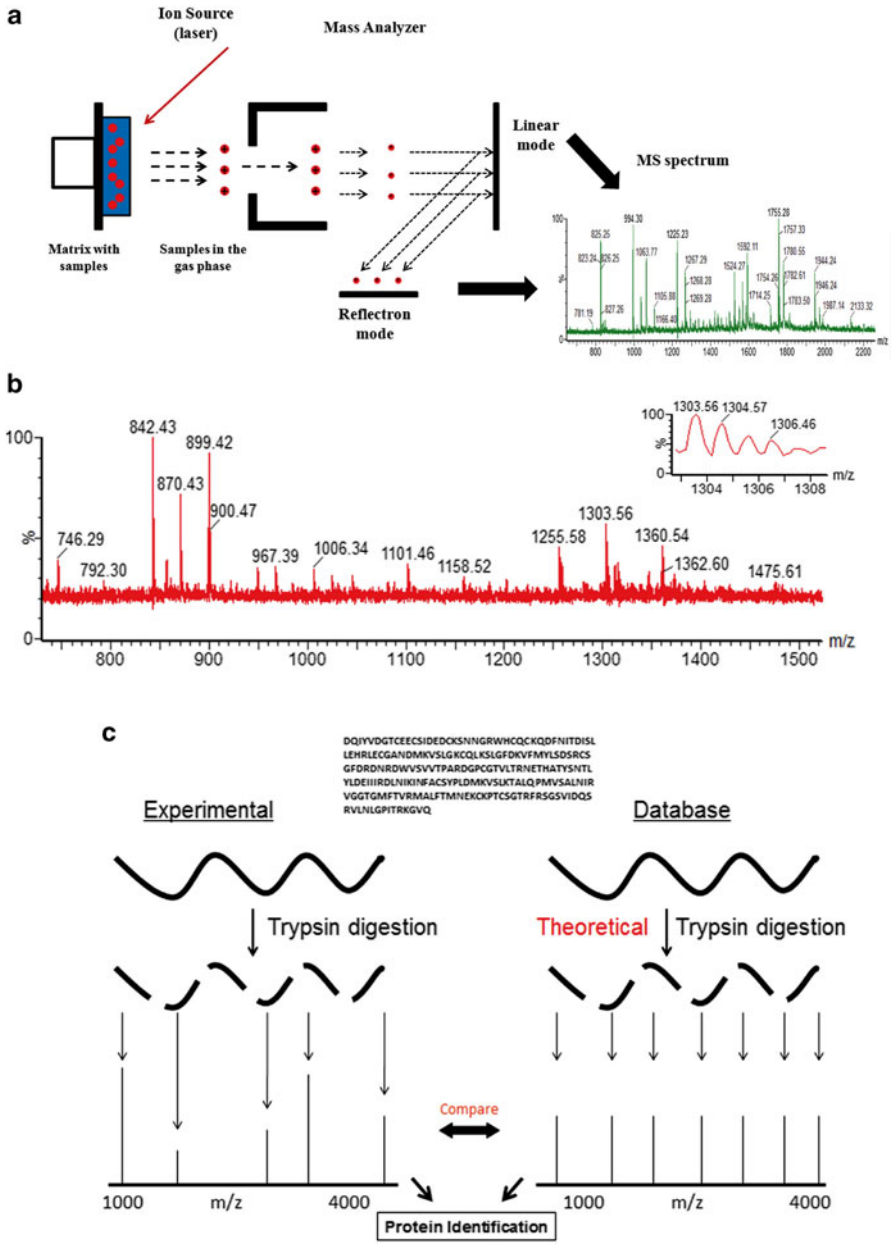

Fig. 1.3 MALDI-TOF MS. (**a**) MALDI-TOF mass spectrometer principle. An ion source, a mass analyzer, and detector are present on the instrument. At the detector the mass spectrum is detected/recorded. The mass analyzer is a TOF and can be used in linear mode or reflective mode. (**b**) A MALDI-MS spectrum primarily contains singly charged peaks; one example is shown (enlarged) to reveal the peak's charged state (single charged or +1). (**c**) Protein identification via MALDI-MS and peptide mass fingerprinting (PMF). A protein is digested into peptides using trypsin and the

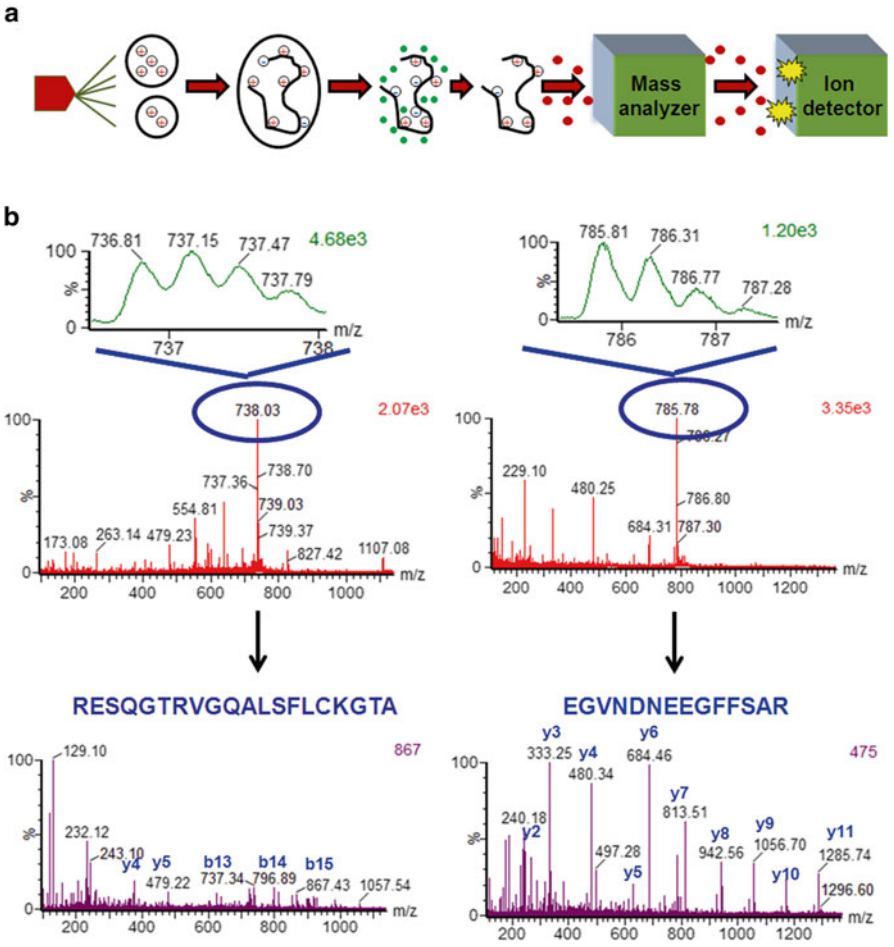

Fig. 1.4 ESI-MS of peptides. (**a**) An ESI-MS mass spectrometer. The ESI-MS has an ion source, in which the ions are ionized, a mass analyzer that ions travel through, as well as an ion detector, which records the mass spectrum. In ESI-MS, the sample is liquid, under high temperature and high electric current. The sample dehydrates and becomes protonated for positive ionization. (**b**) TOF MS spectra example, in which two different peaks, one triply charged peak with *m/z* of 736.81 (*left*) and one double charged with *m/z* of 785.81 (*right*, both circled and zoomed in), are selected for fragmentation and produce the MS/MS spectra whose data analysis led to identification of peptides with the amino acid sequence RESQGTRVGQALSFCKGTA (*left*) and EGVNDNEEGFFSAR (*right*). Note that when the protonation site (R) is on the N-terminus of the peptide, the quality of the MS/MS spectrum is not great and analysis of the b and y ions produced by the MS/MS fragmentation is difficult to interpret. However, when the protonation site is on the C-terminus of the peptide, the fragmentation produces a nice y ion series and the analysis of these ions can easily identify the amino acid sequence of the peptide. Reprinted and adapted with permission from the *Oxidative Stress*: *Diagnostics*, *Prevention*, *and Therapy*, S. Andreescu and M. Hepel, Editors. 2011, American Chemical Society: Washington, D.C [16]

spectrum in a process called direct infusion (ESI-MS mode). For example, if one has 10 peptides in an Eppendorf tube, one can identify all 10 peptides in one spectrum. However, in the MS one identifies only the masses of the peptides. In order to identify the sequence information about one particular peptide, one must isolate one peak that corresponds to one of the 10 peptides (precursor ion), fragment it in the collision cell using a neutral gas (for example, Argon gas), and record a spectrum (a sum of spectra) of the product ions that resulted from fragmentation of the precursor ion called MS/MS (ESI-MS/MS mode). Data analysis of the MS and MS/MS spectra usually leads to identification of the mass and sequence information about the peptide of interest. Examples of ESI-MS and ESI-MS/MS spectra are shown in Fig. 1.4b. As observed, the quality of the MS/MS spectra is directly dependent on the amino acid sequence, but more important, by the position of the proton-trapping amino acid (R, H, or K, in this case, R). For example, if the proton-trapping amino acid is on the N-terminus, low intensity b and y ions are observed (Fig. 1.4b, left). However, when the proton-trapping amino acid is located on the C-terminus, the fragments produced are almost always y ions of high quality. This is also the main reason for which most proteomics experiments use trypsin as an enzyme, since it cleaves the C-termini of R and K and produces peptides with an R or a K at the C-terminus.

Sometimes, when a peptide has more than one proton accepting amino acid such as Arg or Lys, the peptide may be protonated by more than two or three protons. Therefore, the same peptide may be identified with more than two or three charges. The advantage for these peptides is that if the precursor ion in a charge state of, e.g., 2+ does not fragment well in MS/MS, then the peak that corresponds to the same peptide but in a different charge state (e.g., 3+ or 4+) may fragment very well. One drawback for the multiply charged peptides is that they are usually longer (2,500–3,000 Da) than the regular peptides analyzed by MS (800–2,500 Da) and data analysis for these peptides may be more difficult than for regular peptides. However, overall, fragmentation of more than one peak corresponding to the same peptide but with different charge states may help in obtaining additional information about that peptide.

ESI-MS can be used not only for peptides but also for investigation of proteins and the information is particularly useful for determining the molecular mass of those proteins, of their potential PTMs, and of their conformation. In addition, the high molecular mass proteins can also be analyzed by ESI-MS in either positive mode (protonated) or negative mode (de-protonated), thus providing distinct, yet complementary, information regarding the distribution of charges on the surface of the protein investigated. Examples of MS spectra of a 16.9 kDa protein investigated by ESI-MS in both positive and negative mode are shown in Fig. 1.5.

1.6 LC–MS/MS

Analysis of peptide mixtures by ESI-MS for determination of the molecular mass of the peptides is usually a quick procedure. However if one wants to investigate the sequence information of more than one peptide, it is not the method of choice,

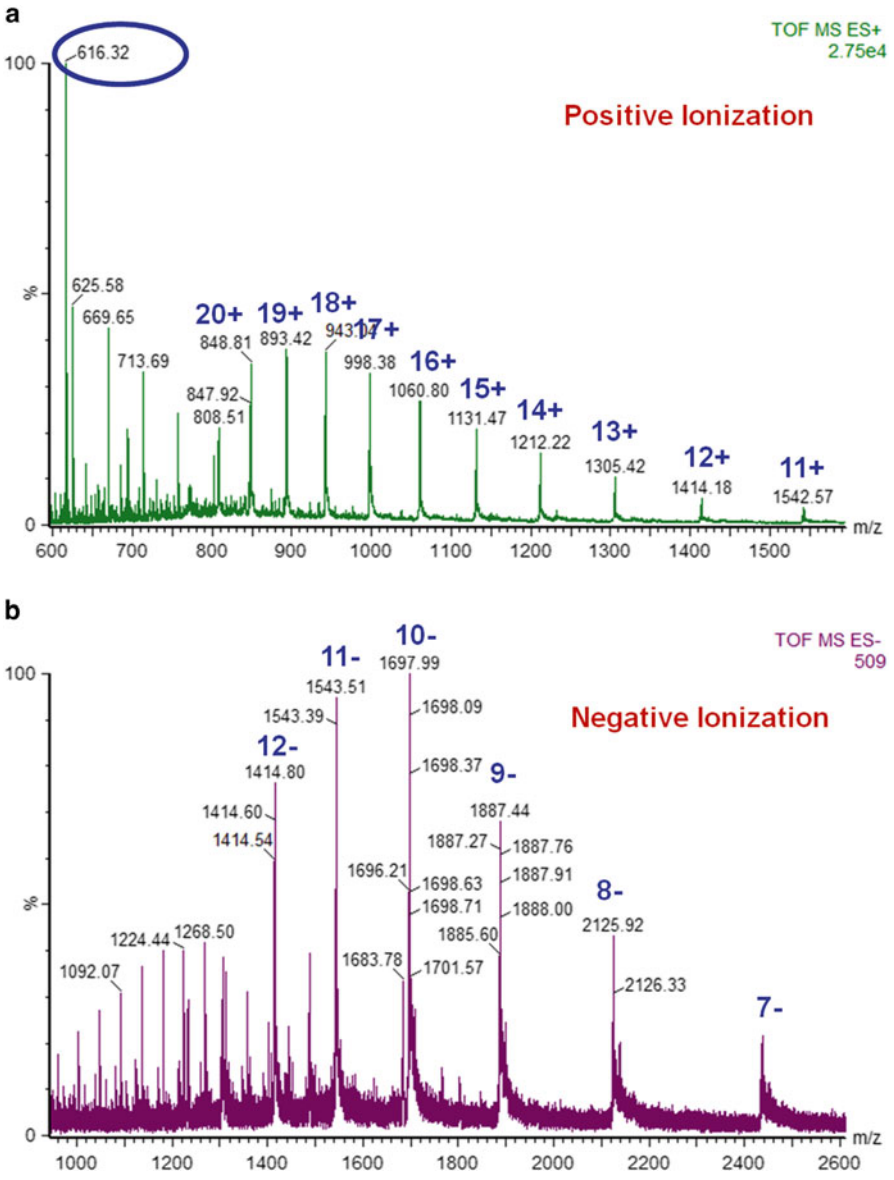

Fig. 1.5 ESI-MS proteins: ESI-MS spectra of intact 17 kDa protein, myoglobin, analyzed under acidic conditions (pH ~ 2). (**a**) MS spectrum in positive ionization; (**b**) MS spectrum analyzed in negative ionization. The positive (A) and negative (−) charges are indicated. The peak with *m*/*z* of 616.32 (1+) corresponds to the heme group, which is the prosthetic group of myoglobin. Reprinted and adapted with permission from the *Australian Journal of Chemistry CSIRO Publishing* http://www.publish.csiro.au/?paper=CH13137 [15]

since fragmentation of the ions that correspond to peptides happens manually; one peptide at the time. For example, if one has 4 peptides in a mixture, we can determine the molecular mass of all peptides in minutes, but to determine their amino acid sequence, the peptides must be selected for fragmentation one at the time. Therefore, to automate this process, an alternative approach is necessary. One option is to fractionate the peptides by column chromatography coupled to an HPLC, i.e., reversed phase-based HPLC (reversed phase columns are particularly compatible with MS). The combination of HPLC and ESI-MS is named HPLC–ESI-MS or LC–MS. In this setting, the peptides are fractionated by HPLC prior to MS analysis. They can also be selected for fragmentation and then fragmented by MS/MS. In a process called data-dependent analysis (DDA), usually 3–4 precursor peaks (which correspond to peptides) are selected for fragmentation from one MS scan and fragmented by MS/MS in a process called LC–MS/MS. In LC–MS/MS, the mass spectrometer analyzes fewer peptides per unit of time as compared with ESI-MS, simply because the HPLC fractionates the peptide mixture over a longer period of time (such as a 60 min gradient) and gives the mass spectrometer more time to analyze more peptides. A schematic of the LC–MS/MS is shown in Fig. 1.6a.

Various types of improvements can be done to increase the number of MS/MS spectra with high quality data which can lead to identification of additional proteins. One is at the flow rate of the HPLC. On a high flow rate, the mass spectrometer will have less time to analyze the peptide mixtures, as compared with lower flow rate. On a longer HPLC gradient (such as 120 min), the mass spectrometer will have more time to analyze more peptides, as compared with a shorted gradient. The number of MS/MS may also influence the number of peptides fragmented per minute. For example, a mass spectrometer has usually one MS survey followed by several MS/MS, for example, between 3 and 10 channels for MS/MS (newer instruments can be up to 30 MS/MS). If the method is set to have one MS survey scan and then to do MS/MS of the two most intense peaks, then the instrument will work as follows: one second MS survey, one second MS/MS (Peak 1), one second MS/MS (Peak 2), and then again one second MS survey (Fig. 1.6a).

Assuming that a mass spectrometer has a cycle of one MS and two MS/MS (such as 0.1 s for an MS survey followed by selection of two precursor peaks for fragmentation by MS/MS; 3 s per MS/MS), this means that in 1 min, the MS instrument can perform ~30 MS/MS that can lead to identification of ~15 proteins. In a 120 min gradient, the possible number of proteins that can be identified is $\sim 15 \times 120 = 1{,}800$ proteins, but keeping in mind that the real length of a 120 min gradient is about 90 min (the rest of 30 min in washing with organic), this means that an MS run can identify $\sim 15 \times 90 = 1{,}350$ proteins. If the length of an MS/MS decreases from 3 to 1 s and the number of precursors selected within MS survey for MS/MS increases to 6, then the number of proteins identified increases by sixfold ($\sim 1{,}350 \times 6 = 8{,}100$ proteins). Assuming that these results are at a flow rate of 0.5 μL/min, if we reduce the flow rate by ½, the number of proteins that can be identified increases by a factor of 2 (i.e., $8{,}100 \times 2 = 16{,}200$).

However, when we calculate the number of these proteins that can be identified, our assumption is that all the steps mentioned work perfectly. In practice, this is often