Role of pH level on the morphology and growth rate of myelin figures

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Abstract: The myelin figure (MF) is one of the basic structures of lipids, and the study of their formation and the effect of various parameters on their growth is useful in understanding several biological processes. In this paper, we address the influence of the pH degree of the surrounding medium on MF dynamics. We introduce a tunable shearing digital holographic microscopy arrangement to obtain quantitative and volumetric information about the complex growth of MFs. Our results show that (1) the time evolution of relative length and volume changes of MFs follows a power-law, (2) the acidity facilitates the growth rate, and (3) the acidic environment causes the formation of thicker MFs.

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1. Introduction

Cell membrane has an essential role in the evolution of life and acts as a barrier and a regulating mechanism for the movement of substances into and out of the cell [1]. Further, by the use of chemical and charge gradients, it provides energy to the cell, supplies the necessary substrates to use in signaling molecules, and facilitates information transduction in biosynthesis, and organizes enzyme activities [2]. Cell membranes are composed of several kinds of proteins, carbohydrates and lipids. Amongst, lipid bilayers are their most basic consisting structure [3–5]. Lipid species may differ depending on the configuration of their head group, the number of carbon-carbon bonds, molecular weight, and their whole structure [6,7]. In particular, the general lipid structure is determined by the polarity of the head group and the hydrophobicity of their tails [8]. Polymorphic structural characteristic of lipids plays a fundamental role in cellular function and is attributed to the tendency of the phospholipid molecules to minimize their contact with the surrounding water molecules [5,9]. Depending upon environmental parameters such as temperature, pH, ionic strength and hydration, different structures of lipids, such as micelles, liposomes, bilayer sheets, myelin figure (MF), etc. may be formed [10,11]. The formation of these structures is the consequence of the tendency of some of the membrane constituents to interact with small and soluble molecules and ions in their surrounding, which may happen in the presence of excess water [3].

MF is an important structure of lipids. They consists of a large number of concentric cylindrical bilayers separated by thin hydration layers which makes them as osmiophilic materials with lamellar bodies [12]. These structures were observed for the first time by Rudolf Virchow in 1854, and were thought to be created by a living tissue [13]. MFs may be derived from various types of membranes and organelles, such as endoplasmic reticulum, either in their normal or diseased shapes. It is known that they may play a role in interstitial lung diseases, in granular cell tumors, and in keratinisation in the epidermis [14,15]. In addition to the phospholipids as a main source for synthetic formation of MFs, there are other MF formation sources such as ionic...
and nonionic surfactants. It is also remarkable that beyond the biological importance of MFs, they can be considered as an important model for realization of artificial nanofluidic networks.

For these reasons, MFs have been extensively studied both theoretically and experimentally. The studies mainly include their detailed imaging, initial growth and retraction, coiling and uncoiling behavior, and the influence of external stimuli, stresses and factors on their dynamical behavior. Whatever the source that protrudes MFs, the structure and configuration of all MFs are very similar to what is formed via simple hydration of dried synthetic lipids. Therefore, it is usual to consider this simple in vitro experiment as a model to study the general features of natural MFs. MFs and their behavior under various circumstances are advantageous models for understanding the self-assemblies and self-organizations in biological membranes. In this direction, the growth of MF was first studied by Sakurai and Kawamura for egg-yolk phosphatidylcholine. Several research on MF formation and growth have been performed since then. It is known that MF formation starts from the sides of the initial lipid cakes once they start to interact with an aqueous medium. The rate of their growth depends on various parameters, which has been the subject of some of our previous research; for instance, we have shown that thermal gradient around the sample chamber facilitates the growth and increases its rate, while humidifying the initial lipid reservoir has a reverse effect on the growth rate even to the extent that can prevent MF growth. Despite an increasing interest in MF structure, currently for the formation of MF there is no explicit explanation. However, some hypotheses have been proposed that suggest swelling, diffusion, or mechanical instability may govern the growth of MFs. In this paper, we investigate the influence of another important parameter, namely the pH level of the surrounding buffer, on MFs formation and growth.

In order to visualize and investigate MFs several imaging and detection approaches have been considered. One high-resolution possibility is to incorporate electron microscopies. Scanning electron microscopy has been already applied to investigate the structure of MFs. However, it requires the sample to be dried or fixed before imaging. Furthermore, according to its scanning nature, it isn’t suitable for imaging of a dynamic specimen such as the growth of an MF. Bright-field microscopy can be useful to obtain general information on the size, shape, homogeneity, and degree of aggregation of an MF sample with the optimum resolution of approximately 250 nm, which is defined by the Abbe’s diffraction limit. In the imaging of MFs, the essential challenge is that these samples are almost transparent, which makes them hard to image with the conventional optical imaging techniques, such as bright-filed microscopy, because those techniques can detect only the intensity and color of the light of field-sample interaction. However, compilation of bright-field microscopy with some techniques can provide sufficient contrast and supply more significant information about the structure and dynamics of a sample. First, as it is common in many other biomaterials investigation, fluorescence microscopy has been applied to study the MFs. However, staining of the MF samples with labels or probes can potentially affect the properties of biomaterial samples. Confocal microscopy seems to be a proper methodology to visualize the internal structure of the MFs due to its superior image clarity and provision of a composite three-dimensional (3D) image. However, this method is also based on scanning the sample, therefore, it is not a proper imaging modality for dynamic samples; sample movement in all directions leads to missing fast dynamics, reduction in the accuracy, and difficulty in focusing. Zernike’s phase contrast microscopy and differential interference contrast microscopy seem to fit well with MF samples, since they can detect the optical phase changes applied by transparent samples. However, the main drawback of these techniques is that they are inherently qualitative. Several attempts have been presented to resolve this problem with more quantitative approaches. Amongst, digital holographic microscopy (DHM) has been shown to be an effective technique for quantitative phase-contrast imaging.
Digital holograms are formed by the use of a coherent light and through interfering of a reference wave and a wave carrying full information of the object under investigation. The reconstruction is carried out numerically with a computer, and the process includes simulating the reference wave illumination and a free space propagation. There are various methods for numerical reconstruction. Amongst, the angular spectrum propagation approach, in which most of the process is carried out in Fourier space, has been used more often [48]. The reconstruction leads to the whole-field information about the object. However, a real and virtual image along with the undiffracted reference beam appear upon numerical illumination of the recorded hologram, due to the interferometric terms. In the off-axis DHM geometry, a slight angle between the object and the reference waves separates out the aforementioned terms in the Fourier domain by spectral filtering and it simplifies the numerical processing. Nevertheless, due to the interferometric nature, DHM is highly sensitive to environmental and mechanical vibrations. An elegant solution to overcome the problem has been considering the self-referencing DHM schemes. In these schemes in order to form the interference, a portion of the light beam interacting with the sample is redirected to overlay with another portion of it, preferably free of object. As a result the environmental uncorrelated noises are greatly reduced, and a robust and stable system is achieved. In recent years, extensive research have been dedicated to develop different self-referencing DHM setups [49,50]. The key point of this scheme is to apply a slight phase difference between the interfering portions of the object wave, which can be achieved by the use of a Lloyd’s mirror [49], a glass plate for applying a lateral shearing [51–54], etc. In shearing self-referencing arrangements, the shearing between the waves can be applied by incorporating of a wedge plate or a grating. The amount of the shearing, i.e., the holographic fringe period, depends on the thickness of the plate or the grating period. Therefore, it is a constant. On the other hand, for several applications, e.g., the ones dealing with the complex geometry of microscopic samples implementation of the fringe characteristics is a required task. A minimal possibility to change the fringe periods may be achieved by a slight rotation of the shearing module, which is not sufficient for most cases. We provide a tunable shearing DHM scheme that may overcome the problem. Accomplishment of a tunable self-referencing arrangement is the most appropriate approach to proceed the MFs dynamics, according to their varying and complex morphology.

In this paper, using this adjustable self-referencing DHM, we measure the morphometric changes of MFs affected by various kinds of acidic or alkaline aqueous solutions with different pH degrees. The details of the scheme are explained in the Materials and methods Section. In Section 3, the hologram processing methodology is described, and the experimental results are presented and discussed. The paper is concluded in Section 4.

2. Materials and methods

2.1. Sample preparation

There are several methods to produce MFs, such as contact method, drying drop method, and immersion-and-puncture method [28,55–58]. The contact method is the most common one in which, at relatively high temperatures, immediately after the connection of dehydrated plaque of amphiphiles with water and transformation of it to fluidic bilayers, several MFs grow out of the
plaque-water interface. In the drying drop method, evaporation of a drop of a dilute suspension of lipid vesicles on a glass slice results in the creation of pancake-like multilamellar disks at the surrounding of the drop. During this process, structure transformations happen from edges of some of these disks to form MFs. In the immersion-and-puncture method, initially a well-ordered lamellar structure is prepared, and then the surfactant plaques anneal to form planar structures with millimeter-sized domains. After cooling and dehydrating them, the container of the plaques is carefully flooded with water. Unlike the contact method, MFs are formed from the well-ordered surfactant plaques. In this method, MFs can also be produced by puncturing the well-ordered bilayer stack with a sharp needle [28].

We consider the contact method. 1-palmitoyl-2-oleoyl-sn-glycero-3-phosphocholine (DOPC) are purchased from Avanti Polar Lipids. A little amount of DOPC is dissolved in chloroform, and one microliter droplet of the solution is cast on a clean glass slide. The drop is air-dried at room condition for several hours so that the lipid plaque is completely pinned to the slide. Lipid plaque is finally sandwiched between two glass slides separated by a spacer with 1 mm thickness. The solution should be stored at -20°C in a glass container in which air is not allowed to penetrate. Separately, we prepare acidic and alkaline aqueous solutions with the pH range of 2 to 12. Deionized water is used for the neutral solution. To prepare the acidic solution, first 50 cc of 0.1 M HCL acid solution with a density of 1.18 is poured inside a beaker under fume hood. To combine water and acid, it is important to note that water is added to the acid, and the water-acid mixture is placed in a sonicator for 10 min until the solution is completely uniform. We put distilled water with a pH of 7 in a calibrated pH meter and slowly add the acidic solution to it to reach the desired acidic pH. Since the solution is 0.1 M, according to the relation $[H^+] = 10^{-pH} = 10^{-1} = 0.1$, we have a pH of 1 without adding distilled water. By adding distilled water, the desired acidic solutions of pH2 and pH4 are prepared. To prepare the alkaline solution, first we pour 50 mg of solid NaCl in distilled water and place it in the sonicator for 10 min to dissolve completely. Then we put distilled water in the pH meter and add NaCl to reach the desired pH. The desired alkaline solutions are pH10 and pH12. The deionized water, an acidic or alkaline solution at different pH ranges is injected into the chamber through a syringe connected to an injection pump (NE-300, Just Infusion™) at a controlled flow rate of 30 ml/hrs. The solution covers the entire dry film area. Upon contact, MFs grow rapidly from the edge of lipid plaques.

2.2. Experimental setup

Figure 1 shows the schematic of our tunable self-referencing setup. A laser beam (He-Ne, 632.8 nm, 2mW) after expansion by the beam expander is redirected by the mirror and is condensed onto the sample by the use of the condenser (Thorlabs, CSC200, NA=0.78). The microscope objective (Olympus, 10×, NA=0.3) collects the scattered light from the lipid sample. Light diverging from the sample enters the self-referencing module (SRM), which is the key element in our setup. Figure 1(a) shows a magnified view of SRM. SRM consists of a conventional shearing plane-parallel glass plate (3 mm thickness) and a couple of moveable right-angle prisms, all made of similar material (BK7). Using only the glass plate is the conventional approach to implement self-referencing geometry, in which two laterally sheared replicas of the object wavefront are created, and propagate collinear to form interference patterns on the recording device. In the presented SRM module, the existence of three conditions enables adjusting the shearing between the two replicas of the object beams: (1) divergence of the beam entering to SRM, (2) adjustment of the angle of the incident beam to the first prism, and (3) freely positioning the second prism so that some of the successive (total internally) reflected beams come out of the module; the longer the distance of two prisms, the more the number of the reflections. Then, according to the divergence of the beam, part of two adjacent reflected beams from the bottom surface overlap and propagate together to hit the camera. For example, in the magnified scheme of the setup,
part of the third reflection overlaps with the fifth reflection due to their divergence, and in the overlap area holographic fringes are formed. Therefore, adjusting the position of the second prism, i.e., the number of reflections, allows tuning the shearing value hence the fringe density. This, additionally, is useful in separating out the images in the reconstruction process. The digital holograms are recorded by a digital camera (DCC1545M, Thorlabs, 8 bit dynamic range, 5.2 µm pixel pitch) and are subjected to the numerical reconstruction process. Data acquisition starts before the injection of water or other solutions start to contact with the lipid plaque and monitors live the samples at 25 fps speed. Figure 1(b) shows a bright-field microscopy image of the MFs formed upon contacting with excess water. Our setup is based on an up-right geometry microscope, however, this module can similarly be used with inverted microscopy arrangements. We remark that by employing a proper pinhole, it is also possible to convert one of the interfering object beams into a separate reference beam and achieve a wider field of view [50]. Moreover, the setup has the possibility to be combined with microsphere assisted microscopy toward a super-resolution variable shearing DHM [59].

![Fig. 1. Schematic self-referencing DHM setup; (a) Detailed sketch of the hologram formation in the self-referencing module (SRM). (b) Bright-field image of MFs produced upon the contact of lipid plaque to excess water.](image)

3. Results and discussions

We conducted experiments on several lipid samples. For each pH level, more than 20 samples were prepared and examined. The recorded digital holograms immediately after contacting the aqueous solution with the lipid plaques are subjected to numerical reconstruction. At the beginning of each experiment, in order to remove the background contaminations from the sample container and the optical train, a reference hologram in which no lipid cake is present is recorded. We utilized the angular spectrum propagation approach in scalar Fresnel-Kirchhoff diffraction
theory for numerical reconstruction of the recorded holograms [21,41]. In the reconstruction process, the recorded holograms are numerically propagated to the reconstruction plane, from which the phase and intensity of the reconstructed wavefront can be computed. The reconstructed phase pattern is unwrapped by Goldstein’s branch-cut method to convert it into continuous-phase distribution leading to quantitative phase contrast image of the sample [60]. The reconstruction results of a typical MF formation and growth process is shown in Fig. 2. The growth rate of an MF may be obtained through measuring its length. The length can be obtained simply by pixel counting in conventional bright-field microscopy or (for higher contrast) in phase-contrast microscopy image. More precisely, the growth rate maybe obtained through similar processing on the area covered by an MF during its growth. To this aim, the successive conventional microscopic images, by defining a threshold intensity value, are converted into their binary versions and by summation over the bright pixels, covered area in each image is achieved. Then, the growth rate can be obtained by tracking the area or alternatively the length (calculated through area measurement) variations. However, these methods can hardly provide growth rates accurately, because the volumetric variations are lost. This is a more pronounced problem when frequently the formed MFs after a short time experience one or more coiling. The demonstrated results show that DHM can overcome the problem. In Fig. 2(a-c) the reconstructed 3D and 2D images of a MF at the beginning of its formation and after 6 s and 12 s afterwards are shown. Figure 2(d) is the 3D image of a coiled MF and Fig. 2(e) is its 2D map. These images can be used, similar to bright-field image processing, to measure the covered area by each MF during its growth. However, DHM reconstruction provides also the thickness value throughout the images, assuming negligible changes in MF refractive index during its growth. Therefore, measurement of volume variations will be possible, and from that true length variations can be achieved. Figure 2(f) shows the cross-sectional thickness profile along the lines crossing two different parts of a coiled MF. The difference between the thickness values in the two parts is evident, which, otherwise could not be revealed in conventional image processing.

The whole procedure is applied for several MFs and the variations in their relative length and volume during the dynamical changes are obtained. Figures 3(a) and 3(b) show the time evolution of the average relative length ($\Delta L/L_0$) and the average volume changes ($\Delta V$) of MFs for each pH level, respectively. The error bars correspond to the averaging over five measurement
associated with each pH. The results clearly show that the acidity or alkalinity of the surrounding matters; the acidity of the solution facilitates the growth substantially and the alkalinity hinders it. The time evolution of the MFs volume (Fig. 3(a)) is similar to that of length (Fig. 3(b)). Moreover, we observe that the average thickness of the produced MFs in the acidic environment is greater than that of the alkaline one. The insets of Fig. 3(b) show the recorded holograms of two typical MFs in pH=2 and pH=12 environments, 20 s after their formation. To quantify the effect of pH, we further find the best fitted functions to data and seek for the characterization of the fitted curves. As predicted by considering the diffusive model for the growth of MFs [26,28], our results indicate a power-law behavior for time evolution of them in all cases, \( t^\alpha \), \( \alpha \sim \frac{1}{2} \). However, the growth and dynamics of MFs are affected significantly by the variation of the pH degree. We proceed this influence by obtaining the exponent (\( \alpha \)) and the crossover time (\( t_c \)) of volume changes (Fig. 4) as characterization parameters to the volumetric dynamics of MFs. \( \alpha \) is obtained through linear fitting to volume changes in a log-log plot (inset of Fig. 4(a)), and \( t_c \) is the intersection of the lines fitted to the initial and final data points of relative volume changes, as shown for pH=4 in the inset of Fig. 4(b). \( t_c \) is a time from which the growth of MFs reaches to the saturation stage until it stops. Figure 4(a) shows that MFs grow much faster in acidic environment (\( \alpha > 0.8 \)), and Fig. 4(b) shows that the lower pH degree, the shorter the crossover time. We attribute the influence of pH to its effects on the interfacial tension of the multilayers in MF structure. Generally, the interfacial tension in phospholipid, as a model membrane, can be affected by various structural components [1,2]. In electrically neutral state, lipid molecules are in equilibrium with H as well as with OH ions. However, changing the pH degree leads to the presence of excess ions that oversets the equilibrium. This, in turn, affects the interfacial tension in MF multilammelar structure and its growth behavior.

**Fig. 3.** (a) Dependence of growth power (\( \alpha \)) of MFs on pH degree of their surrounding medium. Inset: volume changes in log-log plot to obtain \( \alpha \). (b) Dependence of crossover time (\( t_c \)) of MFs on pH degree of their surrounding medium. Inset: relative volume change for pH=4 and definition of \( t_c \).

4. Conclusion

In conclusion, we provide one achievement to the field of digital holography and one to the field of biomaterial characterization: (1) we introduce a novel, compact and robust DHM setup that may be used for several applications in biomedical applications, and (2) we apply the presented technique to study the effect of pH degree of the surrounding medium, as an important factor, on the dynamics of MFs. The setup is based on a self-referencing scheme, and hence, the
Uncorrelated noises that exist in conventional off-axis DHMs are substantially reduced. Moreover, by the use of a module consisting of a glass plate and a couple of right-angle prisms we provide easy control on the fringe density, which is a problem that most of the self-referencing schemes suffer from. The processing of the recorded digital holograms taken by this setup during the dynamical changes of MFs shows that the time evolution of relative length and volume changes of MFs follows a power-law. However, the acidity facilitates the growth rate, and causes the formation of thicker MFs.

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Disclosures
The authors declare no conflicts of interest.

References
1. J. Peretó, P. López-García, and D. Moreira, “Ancestral lipid biosynthesis and early membrane evolution,” Trends Biochem. Sci. 29(9), 469–477 (2004).
2. D. Bach and E. Wachtel, “Phospholipid/cholesterol model membranes: formation of cholesterol crystallites,” Biochim. Biophys. Acta, Biomembr 1610(2), 187–197 (2003).
3. C. M. Rosetti, A. Mangiarotti, and N. Wilke, “Sizes of lipid domains: What do we know from artificial lipid membranes? what are the possible shared features with membrane rafts in cells?” Biochim. Biophys. Acta, Biomembr 1859(5), 789–802 (2017).
4. G. Paredes-Quitiaja, H. Aranda-Espinoza, and A. Maldonado, “Shapes of mixed phospholipid vesicles,” J. Biol. Phys. 32(2), 177–181 (2006).
5. A. D. Peteliska and Z. A. Figaszewski, “Effect of pH on the interfacial tension of lipid bilayer membrane,” Biophys. J. 78(2), 812–817 (2000).
6. K. S. Vetrel and G. Thinakaran, “Membrane rafts in Alzheimer’s disease beta-amyloid production,” Biochim. Biophys. Acta, Biomembr 1801(8), 860–867 (2010).
7. L. Kalvodova, N. Kahya, P. Schwille, R. Ehehalt, P. Verkade, D. Drechsel, and K. Simons, “Lipids as modulators of proteolytic activity of bace involvement of cholesterol, glycosphingolipids, and anionic phospholipids in vitro,” J. Biol. Chem. 280(44), 36815–36823 (2005).
8. F. Mesa-Herrera, L. Tiaro-González, C. Valdés-Baizabal, M. Diaz, and R. Marín, “Lipid and lipid raft alteration in aging and neurodegenerative diseases: A window for the development of new biomarkers,” Int. J. Mol. Sci. 20(15), 3810 (2019).
9. Y. S. Tarahovsky, A. L. Arsenault, R. C. MacDonald, T. J. McIntosh, and R. M. Espan, “Electrostatic control of phospholipid polymorphism,” Biophys. J. 79(6), 3193–3200 (2000).
10. K. Larsson, Lipids: Molecular Organization, Physical Functions and Technical Applications, vol. 5 (The Oily Press, 1994).
11. A. Sharma and U. S. Sharma, “Liposomes in drug delivery: progress and limitations,” Int. J. Pharm. 154(2), 123–140 (1997).
12. Y. Sakurai, T. Suzuki, and S. Sakurai, “Cross-sectional view of myelin figures,” Biochim. Biophys. Acta, Biomembr 985(1), 101–105 (1989).
13. L. N. Zou and S. R. Nagel, “Stability and growth of single myelin figures,” Phys. Rev. Lett. 96(13), 138301 (2006).
14. Y. Honda, K. Tsunematsu, A. Suzuki, and T. Akino, “Changes in phospholipids in bronchoalveolar lavage fluid of patients with interstitial lung diseases,” Lung 166(1), 293–301 (1988).
15. A. Jeux, E. Kahn, F. Ménétrier, T. Montange, J. Lherminier, J.-M. Riedinger, and G. Lizard, “Cytotoxic oxysterols induce caspase-independent myelin figure formation and caspase-dependent polar lipid accumulation,” Histochem. Cell Biol. 127(6), 609–624 (2007).
16. M. Buchanan, J. Arrault, and M. Cates, “Swelling and dissolution of lamellar phases: role of bilayer organization,” Langmuir 14(26), 7371–7377 (1998).
17. A. P. Kennedy, J. Sutcliffe, and J.-X. Cheng, “Molecular composition and orientation in myelin figures characterized by coherent anti-stokes Raman scattering microscopy,” Langmuir 21(14), 6478–6486 (2005).
18. E. Evans, H. Bowman, A. Leung, D. Needham, and D. Tirrell, “Biomembrane templates for nanoscale conduits and networks,” Science 273(5277), 933–935 (1996).
19. W. Stoeckenius, “An electron microscope study of myelin figures,” The J. Cell Biol. 5(3), 491–500 (1959).
20. L. Tayebi, M. Mozafari, D. Vashaee, and A. N. Parikh, “Structural configuration of myelin figures using fluorescence microscopy,” Int. J. Photoenergy 2012, 1–7 (2012).
21. N. Fatih, A.-R. Moradi, M. Habibi, D. Vashaei, and L. Tayebi, “Digital holographic microscopy of the myelin figure structural dynamics and the effect of thermal gradient,” Biomed. Opt. Express 4(6), 950–957 (2013).
22. C. Santangelo and P. Pincus, “Coiling instabilities of multilamellar tubes,” Phys. Rev. E 66(6), 061501 (2002).
23. R. Mosaviani, A.-R. Moradi, and L. Tayebi, “Effect of humidity on liquid-crystalline myelin figure growth using digital holographic microscopy,” Mater. Lett. 173, 162–166 (2016).
24. M. Kummrow and W. Helfrich, “Deformation of giant lipid vesicles by electric fields,” Phys. Rev. A 44(12), 8356–8360 (1991).
25. K. Mishima, K. Satoh, and T. Oghara, “The effects of pH and ions on myelin figure formation in phospholipid-water system,” Chem. Phys. Lett. 106(6), 513–516 (1984).
26. K. Mishima, T. Oghara, M. Tomita, and K. Satoh, “Growth rate of myelin figures for phosphatidylcholine and phosphatidylethanolamine,” Chem. Phys. Lipids 62(2), 87–91 (1992).
27. I. Sakurai and Y. Kawamura, “Growth mechanism of myelin figures of phosphatidylcholine,” Biochim. Biophys. Acta, Biomembr. 777(2), 347–351 (1984).
28. L.-N. Zou, “Myelin figures: the buckling and flow of wet soap,” Phys. Rev. E 79(6), 061502 (2009).
29. B. Ruozi, D. Belletti, A. Tombesi, G. Tosi, L. A. Bagotoli, J. H. Ipsen, and P. Méléard, “Impact of membrane-anchored fluorescent probes on the mechanical properties of lipid bilayers,” Biochim. Biophys. Acta, Biomembr. 1798(7), 1333–1337 (2010).
30. S. Bibi, R. Kaur, M. Henriksen-Lacey, S. E. McNeil, J. Wilkhu, E. Lattmann, D. Christensen, A. R. Mohammed, and Y. Perrie, “Microscopy imaging of liposomes: from coverslips to environmental sem,” Int. J. Pharm. 417(1–2), 138–150 (2011).
31. R. Nallamothu, G. C. Wood, C. B. Pattillo, R. C. Scott, M. F. Kiani, B. M. Moore, and L. A. Thoma, “A tumor vasculature targeted liposomal delivery system for combretastatin a-4: design, characterization, and in vitro evaluation,” AAPS PharmSciTech 7(2), E7–E16 (2006).
32. H. Bouvais, T. Pott, L. A. Bagotoli, J. H. Ipsen, and P. Méléard, “Impact of membrane-anchored fluorescent probes on the mechanical properties of lipid bilayers,” Biochim. Biophys. Acta, Biomembr. 1798(7), 1333–1337 (2010).
33. D. Murphy and M. Davidson, “Confocal laser scanning microscopy,” Fundamentals of Light Microscopy and Electronic Imaging pp. 265–305 (2012).
34. L. Reissig, D. J. Fairhurst, J. Leng, M. E. Cates, A. R. Mount, and S. U. Egelhaaf, “Three-dimensional structure and growth of myelins,” Langmuir 26(19), 15192–15199 (2010).
35. O. Mertins and R. Dimova, “Insights on the interactions of chitosan with phospholipid vesicles. part II: Membrane stiffening and pore formation,” Langmuir 29(47), 14552–14559 (2013).
36. D. B. Murphy, Fundamentals of Light Microscopy and Electronic Imaging (John Wiley & Sons, 2002).
37. P. Marquet, B. Rappaz, P. J. Magistretti, E. Cuche, Y. Emery, T. Colomb, and C. Depeursinge, “Digital holographic microscopy: a noninvasive contrast imaging technique allowing quantitative visualization of living cells with subwavelength axial accuracy,” Opt. Lett. 30(5), 468–470 (2005).
38. F. Yi, I. Moon, B. Javidi, D. Boss, and P. P. Marquet, “Automated segmentation of multiple red blood cells with digital holographic microscopy,” J. Biomed. Opt. 18(2), 026006 (2013).
39. V. Farzamrad, A.-R. Moradi, A. Darudi, and L. Tayebi, “Digital holographic microscopy of phase separation in multicomponent lipid membranes,” J. Biomed. Opt. 21(12), 126016 (2016).
40. E. Cuche, P. Marquet, and C. Depeursinge, “Simultaneous amplitude-contrast and quantitative phase-contrast microscopy by numerical reconstruction of fresnel off-axis holograms,” Appl. Opt. 38(34), 6994–7001 (1999).
41. A. Anand, V. K. Chhanialw, and B. Javidi, “Real-time digital holographic microscopy for phase contrast 3D imaging of dynamic phenomena,” J. Disp. Technol. 6(10), 500–505 (2010).
42. O. Matoba, X. Quan, P. Xia, Y. Awatsuji, and T. Nomura, “Multimodal imaging based on digital holography,” Proc. IEEE 105(5), 906–923 (2017).
43. V. Farzam Rad, R. Tavakkoli, A.-R. Moradi, A. Anand, and B. Javidi, “Calcium effect on membrane of an optically trapped erythrocyte studied by digital holographic microscopy,” Appl. Phys. Lett. 111(8), 083701 (2017).
44. Y. Jo, H. Cho, S. Y. Lee, G. Choi, G. Kim, H.-S. Min, and Y. Park, “Quantitative phase imaging and artificial intelligence: a review,” IEEE J. Sel. Top. Quantum Electron. 25(1), 1–14 (2019).
45. V. Micó, J. Zheng, J. García, Z. Zalevsky, and P. Gao, “Resolution enhancement in quantitative phase microscopy,” Adv. Opt. Photonics 11(1), 135–214 (2019).
46. V. Abbasian, Y. Ganjkhani, E. A. Akhlaghi, A. Anand, B. Javidi, and A.-R. Moradi, “Super-resolved microsphere-assisted mirau digital holography by oblique illumination,” J. Opt. 20(6), 065301 (2018).
47. B. P. Thiesing, C. J. Mann, and S. Dryepondt, “High temperature measurements of martensitic transformations using digital holography,” Appl. Opt. 52(19), 4426–4432 (2013).
48. M. K. Kim, “Digital holographic microscopy,” in Digital Holographic Microscopy, (Springer, 2011), pp. 149–190.
49. V. Chhanialw, A. S. Singh, R. A. Leitgeb, B. Javidi, and A. Anand, “Quantitative phase-contrast imaging with compact digital holographic microscope employing lloyds mirror,” Opt. Lett. 37(24), 5127–5129 (2012).
50. P. Yora, V. Trvedi, S. Mahajan, N. R. Patel, M. Joglekar, V. Chhanialw, A.-R. Moradi, B. Javidi, and A. Anand, “Wide field of view common-path lateral-shearing digital holographic interference microscope,” J. Biomed. Opt. 22(12), 126001 (2017).
51. F. Ferraro, A. Wax, and Z. Zalevsky, Coherent Light Microscopy: Imaging and Quantitative Phase Analysis, vol. 46 (Springer Science & Business Media, 2011).
52. A. S. Singh, A. Anand, R. A. Leitgeb, and B. Javidi, “Lateral shearing digital holographic imaging of small biological specimens,” Opt. Express 20(21), 23617–23622 (2012).
53. T. O'Connor, A. Anand, B. Andemariam, and B. Javidi, “Deep learning-based cell identification and disease diagnosis using spatio-temporal cellular dynamics in compact digital holographic microscopy,” Biomed. Opt. Express 11(8), 4491–4508 (2020).
54. A. Anand, I. Moon, and B. Javidi, “Automated disease identification with 3-d optical imaging: a medical diagnostic tool,” Proc. IEEE 105(5), 924–946 (2017).
55. I. Sakurai, “Concentration gradient along the long axis of myelin figures of phosphatidylcholine,” Biochim. Biophys. Acta, Biomembr. 815(1), 149–152 (1985).
56. M. Buchanan, S. U. Egelhaaf, and M. E. Cates, “Dynamics of interface instabilities in nonionic lamellar phases,” Langmuir 16(8), 3718–3726 (2000).
57. R. Taribagil, M. Arunagirinathan, C. Manohar, and J. R. Bellare, “Extended time range modeling of myelin growth,” J. Colloid Interface Sci. 289(1), 242–248 (2005).
58. J.-R. Huang, L.-N. Zou, and T. A. Witten, “Confined multilamellae prefer cylindrical morphology,” Eur. Phys. J. E 18(3), 279–285 (2005).
59. V. Abbasian, S. Rasouli, and A.-R. Moradi, “Microsphere-assisted self-referencing digital holographic microscopy in transmission mode,” J. Opt. 21(4), 045301 (2019).
60. B. Gutmann and H. Weber, “Phase unwrapping with the branch-cut method: role of phase-field direction,” Appl. Opt. 39(26), 4802–4816 (2000).