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► **To cite this version:**

Alexandra Zak, Sara Violeta Merino-Cortés, Anaïs Sadoun, Farah Mustapha, Avin Babataheri, et al.. Rapid viscoelastic changes are a hallmark of early leukocyte activation. *Biophysical Journal*, 2021, 120 (9), pp.1692-1704. 10.1016/j.bpj.2021.02.042 . hal-03375058

**HAL Id: hal-03375058**

**<https://amu.hal.science/hal-03375058>**

Submitted on 16 Oct 2023

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# 1 **Rapid viscoelastic changes are a hallmark of early leukocyte activation.**

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## 20 **Abstract**

21 To accomplish their critical task of removing infected cells and fighting pathogens, leukocytes activate  
22 by forming specialized interfaces with other cells. The physics of this key immunological process are  
23 poorly understood, but it is important to understand them as leukocytes have been shown to react to  
24 their mechanical environment. Using an innovative micropipette rheometer, we show in three differ-  
25 ent types of leukocytes that, when stimulated by microbeads mimicking target cells, leukocytes be-  
26 come up to ten times stiffer and more viscous. These mechanical changes start within seconds after  
27 contact and evolve rapidly over minutes. Remarkably, leukocyte elastic and viscous properties evolve  
28 in parallel, preserving a well-defined ratio that constitutes a mechanical signature specific to each cell  
29 type. Our results indicate that simultaneously tracking both elastic and viscous properties during an  
30 active cell process provides a new way to investigate cell mechanical processes. Our findings also sug-  
31 gest that dynamic immuno-mechanical measurements can help discriminate between leukocyte sub-  
32 types during activation.

## 33 **Significance**

34 The mammalian immune response is largely based on direct interactions between white blood cells  
35 (leukocytes) and pathogens, inert particles or other host cells. Mechanical properties of leukocytes  
36 regulate their migration in the microcirculation to reach their targets, and their interaction with these  
37 targets. Yet, these mechanical properties and their evolution upon leukocyte activation remain poorly  
38 studied. We developed a micropipette-based rheometer to track cell viscous and elastic properties. We  
39 show that leukocytes become up to ten times stiffer and more viscous during their activation. Elastic  
40 and viscous properties evolve in parallel, preserving a ratio characteristic of the leukocyte subtype.  
41 These mechanical measurements set up a complete picture of the mechanics of leukocyte activation  
42 and provide a signature of cell function.

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44

## 45 **Introduction**

46 The understanding of cell mechanics has progressed with the development of micromanipulation  
47 techniques such as micropipette aspiration (1), indentation techniques (2), and others (3). Microfluidic-  
48 ics-based approaches such as real-time deformability cytometry now allow high-throughput mechan-  
49 ical measurements (4, 5). Yet, these techniques make it difficult to track mechanical changes in cells  
50 stimulated by soluble molecules (6, 7), and even more difficult when cells are stimulated by activating  
51 surfaces or partner cells (8).

52 Tracking these changes can bring a wealth of information on cell function in healthy conditions  
53 and diseases. Here we illustrate this by focusing on white blood cells (leukocytes). These cells, among  
54 other functions, fight infected cells or pathogens, remove dead cells, or identify antigens at the surface  
55 of other cells and eventually build an immune response against a detected threat. To do so, leukocytes  
56 activate by forming with other cells specialized interfaces called immunological synapses. Different  
57 leukocytes form different types of synapses, which share several molecular (9) but also mechanical  
58 features. One of these mechanical features is force generation, which is now well documented in dif-  
59 ferent types of leukocytes. For instance, we have shown that a T lymphocyte generates forces when  
60 forming a synapse (10, 11). Others recently showed that B cells generate traction forces during activa-  
61 tion (12, 13), and Evans *et al.* quantified decades ago contractile forces generated during phagocytosis  
62 (14). The role of these forces is still an open question. Their suggested functions, such as allowing a  
63 tight cell-cell contact or probing the mechanical properties of the opposing surface, place these forces  
64 at the core of leukocyte activation (15, 16). In this study, we focus on another mechanical feature:  
65 changes in leukocyte viscoelasticity during their activation. These changes have been shown to exist in  
66 T cells, phagocytes, and B cells. We have shown that during activation, T cells stiffen (17). Pioneering  
67 observations showed that during phagocytosis, the cortical tension of a phagocyte increases dramati-  
68 cally while this cell engulfs its prey (14, 18), and more recent observations confirmed mechanical stiff-  
69 ening of leukocytes during phagocytosis (19). We also recently quantified elastic changes in B cells  
70 during activation (20).

71 As for force generation, the function of the mechanical properties of leukocytes during activa-  
72 tion is not yet understood and open to speculation (see Discussion). Yet, it is important to quantify and  
73 describe them for two reasons. First, we need a complete picture of the mechanics of leukocyte activa-  
74 tion to properly incorporate mechanics into currently proposed models of the activation of the differ-  
75 ent types of leukocytes. Second, beside the fundamental interest of understanding these mechanical  
76 changes, they can have very important consequences in the context of disease. Indeed, mechanical  
77 properties of leukocyte dictate how easily leukocytes migrate in or out of tissue or flow in small blood  
78 vessels (21), so changes of these properties can perturb physiological processes. For instance, circulat-  
79 ing neutrophils stimulated by proinflammatory molecules exhibit changes in their mechanical proper-  
80 ties (22, 23), which contributes to their trapping in pulmonary vasculature in lung diseases such as  
81 acute respiratory distress syndrome (23, 24). Being trapped or slowed down while traveling in a capil-  
82 lary can be due to an increased cell stiffness, but also to an increased cell viscosity. Understanding cell  
83 viscous properties is thus also important, although viscous properties are much less explored than  
84 elastic ones due to inherent difficulties in quantifying them.

85 Most measurements of leukocyte mechanics, including the ones mentioned above, were fo-  
86 cused on elastic and not viscous properties. However, it was recognized decades ago that leukocytes  
87 are viscoelastic and not only elastic (25–27), so in many situations we miss an important aspect of  
88 leukocyte mechanics. Fabry *et al.* (28) and others have shown that one can expect the viscous proper-  
89 ties of leukocytes to be linked to their elastic properties. It was recognized that resting leukocytes such  
90 as neutrophils (27, 29) and macrophages (30) conserve a ratio of cortical tension to cell viscosity of  
91 about 0.2-0.3  $\mu\text{m/s}$ , even though macrophages can be ten times more tensed than neutrophils. The  
92 ratio of viscous to elastic properties seems to be constrained within a rather narrow range of values,  
93 and many studies showed this peculiar aspect of biological matter in various cell types (28, 31, 32).  
94 Here, we asked if for a given cell type, elastic and viscous properties of leukocytes respected a rela-  
95 tionship while they were both evolving over time. To do so, we quantified the evolution of both elastic

96 and viscous properties during the activation of three types of leukocytes. We used a micropipette rheo-  
97 ometer to activate leukocytes with standardized activating antibody-covered microbeads (17, 33-35),  
98 and we further probed early mechanical changes occurring in leukocyte-target cell contacts by using  
99 Atomic Force Microscopy in Single Cell Force Spectroscopy mode (36).

## 100 **Materials and Methods**

### 101 **Micropipette rheometer**

102 The rheometer was build based on the evolution of our profile microindentation setup(37) and Mi-  
103 cropipette Force Probe(17, 33, 38). Micropipettes were prepared as described previously(17, 33, 37,  
104 38) by pulling borosilicate glass capillaries (Harvard Apparatus, Holliston, MA, USA) with a P-97 mi-  
105 cropipette puller (Sutter Instruments, Novato, CA, USA), cutting them with an MF-200 microforge  
106 (World Precision Instruments, Sarasota, FL, USA) and bending them at a 45° angle with an MF-900  
107 microforge (Narishige, Tokyo, Japan). Micropipettes were held by micropipette holders (IM-H1,  
108 Narishige) placed at a 45° angle relative to a horizontal plane, so that their tips were in the focal plane  
109 of an inverted microscope under brightfield or DIC illumination (T cells: TiE ; PLB cells: Ti2, Nikon  
110 Instruments, Tokyo, Japan) equipped with a 100× oil immersion, 1.3 NA objective (Nikon Instruments)  
111 and placed on an air suspension table (Newport). The flexible micropipette was linked to a non-  
112 motorized micropositioner (Thorlabs, Newton, NJ, USA) placed on top of a single-axis stage controlled  
113 with a closed loop piezo actuator (TPZ001; Thorlabs). The bending stiffness  $k$  of the flexible micropi-  
114 pette (about 0.2 nN/μm) was measured against a standard microindenter previously calibrated with a  
115 commercial force probe (model 406A; Aurora Scientific, Aurora, ON, Canada). Once the activating mi-  
116 crobead was aspirated by the flexible micropipette, the cell held by the stiff micropipette was brought  
117 in adequate position using a motorized micromanipulator (MP-285; Sutter Instruments). Experiments  
118 were performed in glass-bottom Petri dishes (Fluorodish, WPI, Sarasota, FL, USA). Images were ac-  
119 quired using a Flash 4.0 CMOS camera (for T cells), or a SPARK camera (PLB cells), both from Hama-  
120 matsu Photonics, Hamamatsu City, Japan. In order to perform rheological experiments, the setup au-  
121 tomatically detects the position of the bead at the tip of the force probe at a rate of 400-500 Hz and  
122 imposes the position of the base of the flexible micropipette by controlling the position of the piezo  
123 stage. The deflection of the force probe is the difference between the position of the bead and the posi-  
124 tion of the piezo stage. Thus, the force applied to the cell is the product of this deflection by the bend-  
125 ing stiffness  $k$ . A retroaction implemented in Matlab (Mathworks) controlling both the camera via the  
126 Micromanager software (39) and the piezo stage moves the latter in reaction to the measurement of  
127 the bead position in order to keep a desired deflection of the cantilever: a controlled force is applied to  
128 the cell at any given time. Experiments were performed at room temperature to avoid thermal drift.  
129 Qualitative behavior of T cell and PLB cells is unchanged at 37°C (not shown). We quantified the fluid  
130 drag due to micropipette translation (Suppl. Mat. 1) and the potential effect of fast cell deformation on  
131 our rheological measurements (Suppl. Mat. 2). The accuracy when measuring the phase shift depends  
132 on the acquisition frequency of both force and cell deformation ( $x_{tip}$ ), which is 400-500 Hz depending  
133 on the size of the region of interest chosen for the acquisition. This leads to a time resolution of 2-2.5  
134 ms. The measured loss tangent had a typical value of  $\eta = \frac{K''}{K'} = \tan(\varphi) \approx 0.3 - 0.5$ , i.e.  $\varphi \approx 0.3 - 0.5$ .  
135 When expressed in seconds instead of radians, this phase lag represents a delay  $\Delta t = \frac{\varphi}{2\pi}T$ , where  $T =$   
136 1s is the period of the oscillatory force modulation, which leads to  $\Delta t = 46 - 74$  ms. This is reasonably  
137 long when compared to our time resolution so as to be accurately measured. Note that the uncertainty  
138 on the phase lag influenced more  $K''$  than  $K'$ , because  $K''$  is proportional to  $\sin(\varphi)$  while  $K'$  is propor-  
139 tional to  $\cos(\varphi)$ . *Data analysis.* Rheological measurements were analyzed by post-processing using a  
140 custom-made Python code. During force modulation sinusoidal  $x_{tip}$  signal was fitted every 0.5-1 period  
141 interval over a window that was 1.5-2.5 periods long (i.e. every 0.5-1 second with a window of 1.5-2.5  
142 seconds for a frequency of force modulation  $f=1$  Hz) using a function corresponding to a linear trend  
143 with an added sinusoidal signal (two free parameters for the linear trend, two for the sinusoidal signal

144 when imposing the frequency  $f=1$  Hz). Best fit was determined using classical squared error minimiza-  
145 tion algorithm in Python.

## 146 **T cell experiments**

147 **Cells and reagents.** Mononuclear cells were isolated from peripheral blood of healthy donors on a Ficoll  
148 density gradient. Buffy coats from healthy donors (both male and female donors) were obtained from  
149 Etablissement Français du Sang (EFS, Paris, France) in accordance with INSERM ethical guidelines.  
150 Human total CD4+ isolation kit (Miltenyi Biotech, 130-096-533) was used for the purification of T cells.  
151 Isolated T-cells were suspended in FBS: DMSO (90%:10% vol/vol) and kept frozen in liquid nitrogen.  
152 One to seven days before the experiment the cells were thawed. Experiments were performed in RPMI  
153 1640 1x with GlutaMax, supplemented with 10% heat-inactivated fetal bovine serum (FBS) and 1%  
154 penicillin-streptomycin (all from Life Technologies ThermoFisher Scientific, Waltham, MA), and filtered  
155 with (0.22  $\mu\text{m}$  diameter-pores, Merck Millipore).

156 **Beads.** Dynabeads® Human T-Activator CD3/CD28 for T Cell Expansion and Activation from Gibco,  
157 were purchased from Thermo Fisher Scientific (ref. 11131D, Carlsbad, CA). These 4.5- $\mu\text{m}$ , superpara-  
158 magnetic polymer beads, coated with an optimized mixture of mouse anti-human monoclonal IgG an-  
159 tibodies against the CD3 and CD28 cell surface molecules of human T cells, mimic stimulation by an  
160 antigen presenting cell (APC). The CD3 antibody is specific for the epsilon chain of human CD3. The  
161 CD28 antibody is specific for the human CD28 co-stimulatory molecule, which is the receptor for CD80  
162 (B7-1) and CD86 (B7-2). These beads induce the phosphorylation of several signaling proteins (17, 40).  
163 Control beads were non-coated 4.5- $\mu\text{m}$  polystyrene beads.

## 164 **B cell experiments**

165 **B cells.** Primary B lymphocytes were isolated from spleens of adult C57BL/6J mice (10- to 20-week-  
166 old), purified by negative selection using the MACS kit (130-090-862) in the total splenocytes. Animal  
167 procedures were approved by the CNB-CSIC Bioethics Committee and conform to institutional, nation-  
168 al and EU regulations. Cells were cultured in complete RPMI 1640-GlutaMax-I supplemented with 10%  
169 FCS, 1% penicillin-streptomycin, 0.1%  $\beta$ -mercaptoethanol and 2% sodium pyruvate (denominated  
170 hereafter as complete RPMI).

171 **Beads.** Silica beads ( $5 \times 10^6$ ; 5  $\mu\text{m}$  diameter; Bangs Laboratories) were washed in distilled water  
172 (5,000 rpm, 1 min, RT), and incubated with 20  $\mu\text{l}$  1,2-dioleoyl-PC (DOPC) liposomes containing GPI-  
173 linked mouse ICAM-1 (200 molecules/ $\mu\text{m}^2$ ) and biotinylated lipids (1000 molecules/ $\mu\text{m}^2$ ) (10 min,  
174 RT), and washed twice with beads-buffer (PBS supplemented with 0.5% FCS, 2 mM  $\text{MgCl}_2$ , 0.5 mM  
175  $\text{CaCl}_2$ , and 0.5 g/L D-Glucose). Then, lipid-coated beads were incubated with AF647-streptavidin (Mo-  
176 lecular Probes) (20 min, RT) followed by biotinylated rat anti- $\kappa$  light chain antibody (clone 187.1, BD  
177 Biosciences) (20 min, RT), used as surrogate antigen (su-Ag) to stimulate the B cell receptor. Control  
178 beads were coated with ICAM-1-containing lipids only (no su-Ag was added). The number of mole-  
179 cules/ $\mu\text{m}^2$  of ICAM-1 and su-Ag/biotin-lipids was estimated by immunofluorometric assay using anti-  
180 ICAM-1 or anti-rat-IgG antibodies, respectively; the standard values were obtained from microbeads  
181 with different calibrated IgG-binding capacities (Bang Laboratories). The lipid-coated silica beads  
182 were finally re-suspended in complete RPMI before use. Such a use of lipid-coated silica beads as  
183 pseudo-APCs to study immune synapse formation, cell activation, proliferation and antigen extraction  
184 by B cells has been previously set up and reported by us (20, 41). They provide the adhesion ligand  
185 ICAM-1 for the integrin LFA-1, expressed at the B cell surface, and tethered BCR-stimulatory signal  
186 (su-Ag) in a fluid phospholipid environment.

## 187 **PLB cell experiments**

188 **Cells and reagents.** The human acute myeloid leukemia cell line PLB-985 is a subline of HL-60 cells  
189 based on STR fingerprinting (42, 43). Gene expression in the two lines is similar but not identical (44).  
190 PLB-985 cells were cultured in RPMI 1640 1X GlutaMax (Gibco) supplemented with 10% sterile heat-  
191 inactivated FBS and 1% sterile penicillin/streptomycin. Cells were passaged twice a week and differ-  
192 entiated into a neutrophil-like phenotype by adding 1.25 % (v/v) of dimethyl sulfoxide (DMSO, Sigma-

193 Aldrich) to the cell suspension the first day after passage and, in a second time, three days after while  
194 changing the culture media (45). Twenty four hours before experiments, 2000 U/ml IFN- $\gamma$  (Immuno  
195 Tools) were added into the cell culture flask, cells were centrifuged for 3 min at 300g and resuspended  
196 in 0.22- $\mu$ m filtered HEPES medium (140 mM NaCl, 5 mM KCl, 1 mM MgCl<sub>2</sub>, 2 mM CaCl<sub>2</sub>, 10 mM HEPES  
197 pH 7.4, 1.8 mg/mL glucose, and 1% heat-inactivated FBS ; filters from Merck Millipore). Differentiated  
198 PLB-985 cells stay in suspension and have neutrophil-like properties (Suppl. Mat. 14).

199 **Beads.** 20- $\mu$ m and 8- $\mu$ m diameter polystyrene microbeads at 10<sup>6</sup> beads/mL (Sigma-Aldrich) were  
200 washed three times by centrifugation at 16000 g for 3min and re-suspended in Dulbecco's Phosphate  
201 Buffered Saline (DPBS, Gibco) filtered with 0.22  $\mu$ m diameter-pores (Merck Millipore). Beads were  
202 then incubated overnight at room temperature with 5% (w/v) Bovine Serum Albumin (BSA, Sigma-  
203 Aldrich) in DPBS. Beads were washed again three times by centrifugation at 16000 g during 3min, re-  
204 suspended in DPBS, and incubated with 1:500 anti-BSA rabbit antibody (Sigma-Aldrich, ref. B1520) in  
205 DPBS for an hour at room temperature. These IgG-coated beads were washed three times by centrifuga-  
206 tion at 16000g for 3 min with DPBS and resuspended in DPBS at 10<sup>6</sup> beads/mL before use. Control  
207 beads were uncoated polystyrene beads, inducing very rare cases of adhesion with the cell but no  
208 phagocytosis. PLB cells tried almost systematically to internalize 20- $\mu$ m activating beads by perform-  
209 ing frustrated phagocytosis, and phagocytosed 8- $\mu$ m activating beads.

## 210 **Atomic Force Microscope and Single Cell Force Spectroscopy**

211 **Cells and reagents.** 3A9m T cells were obtained from D. Vignali (46) and cultured in RPMI completed  
212 with 5 % FBS, 10 mM Hepes, 10mM Sodium Pyruvate in 5 % CO<sub>2</sub> atmosphere. COS-7 APCs were gen-  
213 erated as previously described (47) by stably co-expressing the  $\alpha$  and the  $\beta$  chains of the mouse MHC  
214 class II I-Ak , cultured in DMEM (5 % FBS, 1mM Sodium Pyruvate, 10 mM Hepes, and geneticin  
215 10 $\mu$ g/ml). Cells were passaged up to three times a week by treating them with either Trypsin/EDTA or  
216 PBS 1X (w/o Ca<sup>2+</sup>/Mg<sup>2+</sup>), 0.53mM EDTA at 37°C for up to 5 min.

217 The anti CD45 antibodies used for this study were produced from the hybridoma collection of CIML,  
218 Marseille, France (namely H193.16.3)(48). Briefly, cells were routinely grown in complete culture me-  
219 dium (DMEM, 10% FBS, 1mM sodium pyruvate) prior to switching to the expansion and production  
220 phase. Hybridoma were then cultured in DMEM with decreasing concentrations of low immunoglobu-  
221 lin fetal bovine serum down to 0.5%. Cells were then maintained in culture for 5 additional days ena-  
222 bling immunoglobulin secretion prior to supernatant collection and antibody purification according to  
223 standard procedures.

224 The expression of TCR and CD45 molecules on T cells and MHC II molecules on COS-7 APC was as-  
225 sessed once a week by flow cytometry (anti-TCR PE clone H57.597, anti-CD45 Alexa Fluor 647 clone  
226 30F11 were purchased from BD Pharmingen). The count and the viability, with Trypan blue, were as-  
227 sessed automatically twice by using Luna automated cell counter (Biozym Scientific). Mycoplasma test  
228 was assessed once a month. Culture media and PBS were purchased from Gibco (Life Technologies).  
229 PP2 (Lck inhibitor) was purchased from Calbiochem.

230 **Substrates.** Culture treated, sterile, glass bottom Petri dishes (World Precision Instruments  
231 Fluorodish FD35-100) were incubated with 50 $\mu$ g/ml anti-CD45 antibody for 1 hour at room tempera-  
232 ture. The surfaces were extensively washed with PBS 1X prior to a last wash with HBSS 1X 10 mM  
233 Hepes. The surfaces were kept wet with HBSS 1X 10 mM Hepes before seeding T cells.

234 **T cell preparation.** T cells were counted, centrifuged and resuspended in HBSS 1X 10 mM Hepes and  
235 were kept for one hour at 37 °C 5% CO<sub>2</sub> for recovery prior to seeding. The cells were seeded at room  
236 temperature and after 30 minutes a gentle wash was done by using two 1mL micropipettes in order to  
237 maintain, as much as possible, a constant volume in the Petri dish and hence avoid perturbations by  
238 important flows. After one hour incubation the Petri dish was mounted in a Petri dish Heater system  
239 (JPK Instruments) to maintain the temperature of the sample at ~ 37°C. 10 minutes before experi-  
240 ments, 10 $\mu$ M (final concentration) of Lck kinase inhibitor, PP2 drug was added and homogenized in  
241 the sample. We kept this drug present all along the experiments.

242 **COS-7 APC preparation.** The day before the experiment, COS-7 expressing MHCII cells were incubat-  
243 ed with the peptide of interest with a final concentration of 10 $\mu$ M allowing 100% occupation of MHC II

244 molecules. Before the experiment the cells were detached from the cell culture plates by removing the  
245 cell medium, washed once with PBS 1X and then by a 0.53 mM EDTA treatment for 5 minutes at 37°C  
246 10% CO<sub>2</sub>. The cells were resuspended in HBSS 1X 10mM Hepes and were allowed to recover before to  
247 be seeded into the sample. The peptide p46.61 (which is CD4 dependent) was purchased from Geno-  
248 sphere with a purity >95%.

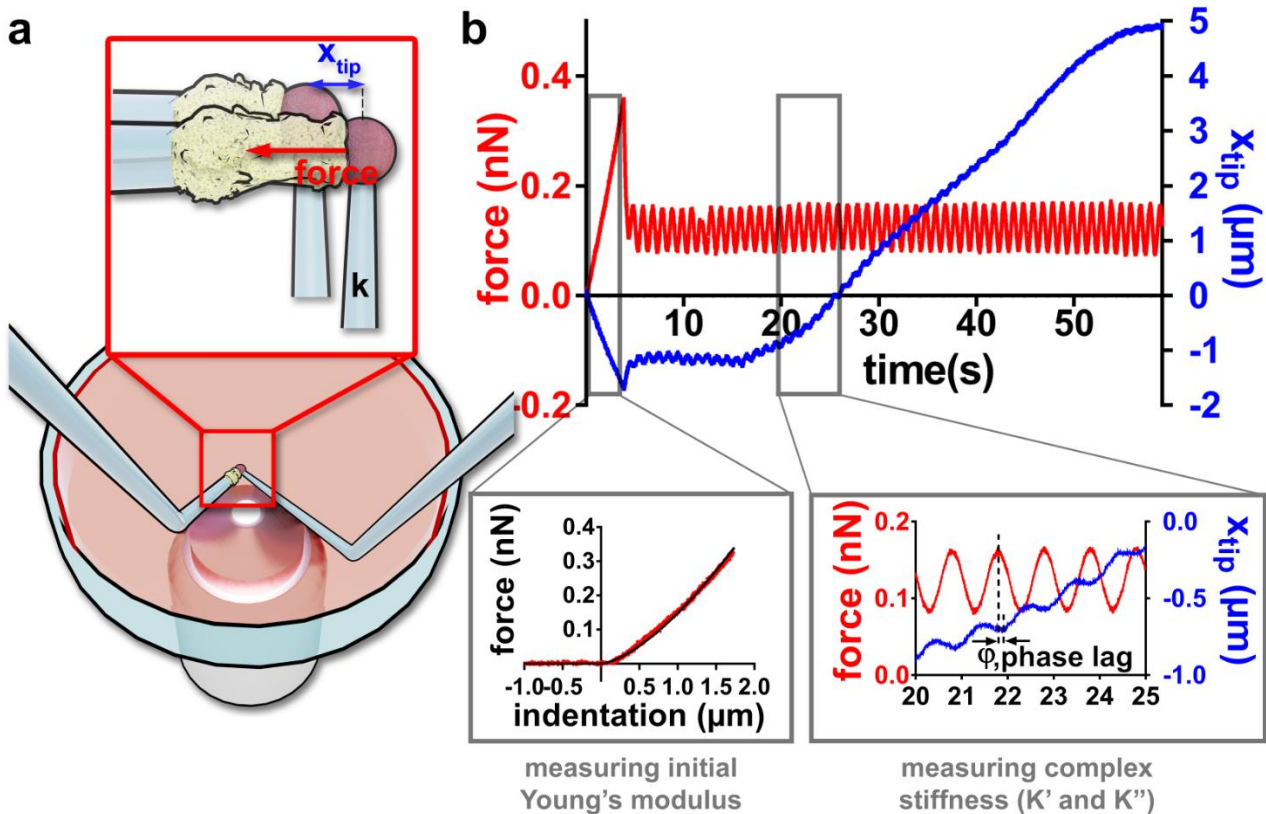
249 **AFM setup.** The setup has been described in great detail elsewhere (49). Measurements were con-  
250 ducted with an AFM (Nanowizard I, JPK Instruments, Berlin) mounted on an inverted microscope  
251 (Zeiss Axiovert 200). The AFM head is equipped with a 15 μm z-range linearized piezoelectric scanner  
252 and an infrared laser. The set-up was used in closed loop, constant height feedback mode (36). MLCT  
253 gold-less cantilevers (MLCT-UC or MLCT-Bio DC, Bruker) were used in this study. The sensitivity of the  
254 optical lever system was calibrated on the glass substrate and the cantilever spring constant by using  
255 the thermal noise method. Spring constant were determined using JPK SPM software routines *in situ* at  
256 the start of each experiment. The calibration procedure for each cantilever was repeated three times  
257 to rule out possible errors. Spring constants were found to be consistently close to the manufacturer's  
258 nominal values and the calibration was stable over the experiment duration. The inverted microscope  
259 was equipped with 10x, 20xNA0.8 and 40xNA0.75 lenses and a CoolSnap HQ2 camera (Photometrics).  
260 Bright field images were used to select cells and monitor their morphology during force measure-  
261 ments through either Zen software (Zeiss) or Micro-Manager (39).

262 **APC/T cell force measurements.** *Lever decoration.* To make the cantilevers strongly adhesive, we  
263 used a modified version of our previous protocols (50). Briefly, cantilevers were activated using a 10  
264 min residual air plasma exposure then dipped in a solution of 0.25mg/mL of Wheat Germ Agglutinin  
265 (WGA) or 0.5 mg/mL of Concanavalin A (ConA) in PBS 1x for at least one hour. They were extensively  
266 rinsed by shaking in 0.2μm filtered PBS 1x, stored in PBS 1x at 4°C. Before being mounted on the glass  
267 block lever holder, they were briefly dipped in MilliQ-H<sub>2</sub>O in order to avoid the formation of salt crys-  
268 tals in case of drying and hence alteration of the reflection of the laser signal. *APC capture.* In separate  
269 experiments, we used side view and micropipette techniques, and we observed that (i) this allows the  
270 presented cell to be larger than the lever tip, excluding any unwanted contact of T cell with lectins and  
271 (ii) the binding is resistant enough to ensure that any rupture event recorded is coming from the  
272 cell/cell interface (Suppl. Mat. 3, Suppl. Fig. S2). After calibration, a lectin-decorated lever is pressed on  
273 a given COS7 cell for 20 to 60 seconds with a moderate force (typically 1-2nN), under continuous  
274 transmission observation. Then, the lever is retracted far from the surface and the cell is let to recover  
275 and adhere/spread for at least 5 minutes before starting the experiments (Suppl. Fig. S2). *SCFS exper-*  
276 *iment.* The surrogate APC is then positioned over a desired T cell. Because the lever is not coated with  
277 gold, hence almost transparent, one can finely position the probe over the target. Force curves are then  
278 recorded using the following parameters: max contact force 1nN, speeds 2μm/sec, acquisition fre-  
279 quency 2048 Hz, curve length 10μm and contact time 60 sec. At the end of the retraction, lateral mo-  
280 tion of the cells is made by hand by moving the AFM head in the (x,y) plane, relative to the Petri dish  
281 substrate, until no more force is recorded by the lever, signaling that full separation was achieved  
282 (Suppl. Fig. S2). One APC cell was used to obtain one force curve on at least three different T cells, and  
283 3 APCs at least were used for each condition. No apparent bias was detected in the data when carefully  
284 observing the succession of the measures for a given APC, for a given lever and for a set of T cells. *Data*  
285 *processing & Statistics.* Force curves were analyzed using JPK Data Processing software on a Linux 64  
286 bits machine. Each curve was evaluated manually, except for calculation and plotting of mean±SD for  
287 relaxation curves where *ad-hoc* python scripts were used. Data were processed using Graphpad Prism  
288 (v7) on a Windows 7 machine. On graphs, data are presented as scatter dot plots with median and in-  
289 terquartile range. A data point corresponds to a force curve obtained for a given couple T cell/APC,  
290 except for the small detachment events where one data point corresponds to one such event (all  
291 events were pooled). Significance was assessed using Mann-Whitney tests in GraphPad Prism v7 with  
292 p<0.05 \*, <0.01 \*\*, <0.001 \*\*\*, and \*\*\*\* below; not significant otherwise.

293 **Results**

294 **Micropipette rheometer for monitoring rapid morphological and mechanical changes in non-adherent cells**  
 295

296 We implemented a real-time feedback loop in our Micropipette Force Probe setup (17, 33)(Fig. 1a)  
 297 allowing us to impose a controlled small oscillatory force modulation  $\Delta F \cos(\omega t)$  (angular frequency  
 298  $\omega = 2\pi f$ , frequency  $f = 1$  Hz) superimposed onto a constant force  $\langle F \rangle$ . A total force  $F(t) = \langle F \rangle +$   
 299  $\Delta F \cos(\omega t)$  is thus applied to the leukocyte during its activation following the contact with an activating  
 300 microbead coated with cell-specific antibodies. The contact is ensured by pressing the bead against  
 301 the cell with a force  $F_{comp}$  (0.12-0.36 nN). This initial compression, from which we extract the cell's  
 302 effective Young's modulus  $E_{young}$  (51, 52) (Fig. 1b, inset, bottom right, and Suppl. Mat. 4), is followed by  
 303 an imposed force modulation regime, where we measure oscillations in the position of the tip of the  
 304 flexible micropipette  $x_{tip}(t) = \langle x_{tip} \rangle + \Delta x_{tip} \cos(\omega t - \varphi)$ , of average value  $\langle x_{tip} \rangle$ , amplitude  $\Delta x_{tip}$ ,  
 305 (typically 100 nm, with a few-nanometer accuracy) and phase lag  $\varphi$ . Changes in  $x_{tip}(t)$  reflect changes  
 306 in cell length (Fig. 1b). As the cell is shorter when the force is higher,  $x_{tip}(t)$  decreases when  $F(t)$  in-  
 307 creases, and corresponds to the lag between a maximum of the force and the following minimum of  
 308  $x_{tip}(t)$  (Fig. 1b, inset, bottom right). From  $x_{tip}(t)$  and  $F(t)$ , we deduce the complex cell stiffness  $K^* =$   
 309  $K' + iK''$ , of elastic part  $K' = \frac{\Delta F}{\Delta x_{tip}} \cos \varphi$  and viscous part  $K'' = \frac{\Delta F}{\Delta x_{tip}} \sin \varphi$  (Suppl. Mat. 5, Suppl. Movie  
 310 S1). We validated our setup by quantifying  $K^*$  of red blood cells (suppl. movie S2). We obtained values  
 311 of  $K'$  very consistent with values predicted by existing models, and also obtained, as expected, very low  
 312 values of  $K''$  (Suppl. Mat. 6).  
 313



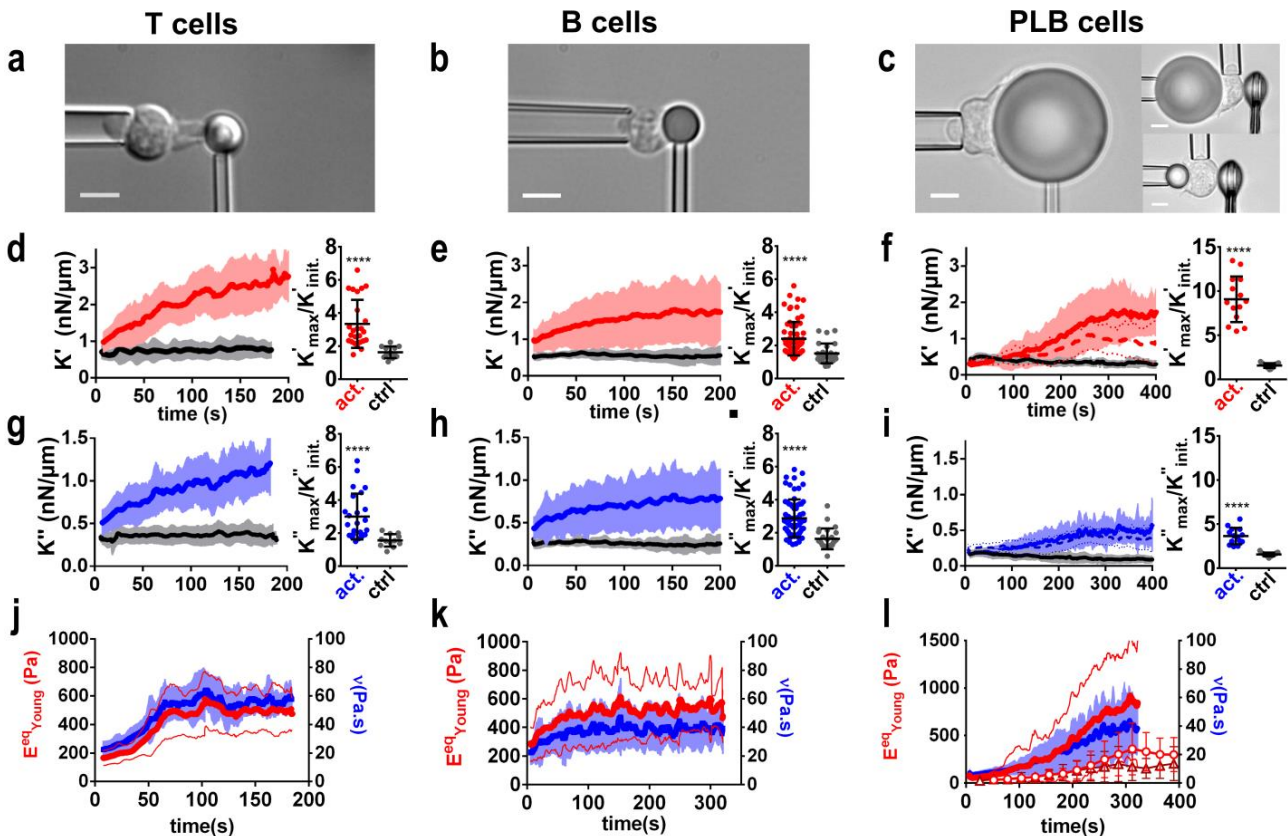
314  
 315 **FIGURE 1.** Rheological measurements during leukocyte activation. (a). Setup. Two micropipettes are plunged in a  
 316 Petri dish. A flexible pipette (right, bending stiffness  $k \approx 0.2$  nN/ $\mu$ m) holds an activating microbead firmly. A rigid  
 317 micropipette (left) gently holds a leukocyte (aspiration pressure of 10-100 Pa). The base of the flexible micropi-  
 318 pipette is displaced to impose a desired force on the cell. (b) Force applied on the cell (in red) and the position  $x_{tip}$   
 319 of the tip of the flexible micropipette (in blue). The cell is first compressed with a force  $F_{comp}$  (here 0.36 nN) to  
 320 measure the initial Young's modulus of the cell (inset, bottom left). Then an oscillatory force is applied to the cell,



321 leading to an oscillatory  $x_{tip}$  signal (inset, bottom right) superimposed to a slower variation of the average value  
 322  $\langle x_{tip} \rangle$  due to the growth of a protrusion produced by the leukocyte.

### 323 Leukocytes become stiffer and more viscous during activation

324 We probed three different types of leukocytes during their specific activation and observed qualitatively similar behavior: both  $K'$  and  $K''$  increase and reach a maximum that is 3- to 8-fold larger than  
 325 their initial value within few minutes ( $\sim 2$  minutes for T and B cells,  $\sim 5$  minutes for neutrophils-like  
 326 PLB985 cells) following the contact between the leukocyte and the relevant activating microbead (Fig.  
 327 2, Suppl. Movie S1). The increase of  $K'$  and  $K''$  is in clear contrast with the constant values obtained  
 328 using control non-activating beads (black lines on Fig. 2). Yet it is worth mentioning that the initial  
 329 value of  $K'$  and  $K''$  is higher for activating beads than for control beads for T and B cells. We explain this  
 330 by the fact that the initial time point for  $K'$  and  $K''$  is actually already  $\sim 10$  seconds after initial cell-bead  
 331 contact, leaving some time for early mechanical changes to occur (see later regarding mechanical  
 332 changes). We evaluated the effect of temperature in the case of T cells by performing experiments at  
 333  $37^\circ\text{C}$ . T cells behaved qualitatively the same at  $37^\circ\text{C}$ , but reacted faster, and reached higher levels in  $K'$   
 334 and  $K''$  (Suppl. Mat. 7).  
 335  
 336



337  
 338 FIGURE 2. Both  $K'$  and  $K''$  increase during leukocyte activation. Each column corresponds to a different cell type.  
 339 (a-c) Cell morphology. Scale bars are  $5\ \mu\text{m}$ . (d-f) Left:  $K'$  for activating (red lines) and non-activating control  
 340 beads (gray lines). In f, solid red line is for  $20\text{-}\mu\text{m}$  beads, dashed red line for  $8\text{-}\mu\text{m}$  beads with indentation in the  
 341 back (see Figure 3). Right: max/min ratio of  $K'$  values, for activating and non-activating control beads. (g-i) Left:  
 342  $K''$  values corresponding to  $K'$  values in d-f. In i, solid blue line is for  $20\text{-}\mu\text{m}$  beads, dashed blue line for  $8\text{-}\mu\text{m}$   
 343 beads with indentation in the back (see Figure 3). (j-l) Equivalent Young's modulus (red, left axis) and cell vis-  
 344 cosity (blue, right axis). In l, dots are the Young's modulus measured in the back of the cell for  $20\text{-}\mu\text{m}$  beads, tri-  
 345 angles are for  $8\text{-}\mu\text{m}$  beads. In all panels, leukocyte-bead contact time ( $t=0$ ) is detected as a force increase, thick  
 346 lines are mean  $\pm$  SD over at least 3 pooled experiments (T cells: 21 cells, B cells 71 cells, PLB cells: 14 cells). Mann-  
 347 Whitney test, \*\*\*\*:  $p < 0.0001$ .

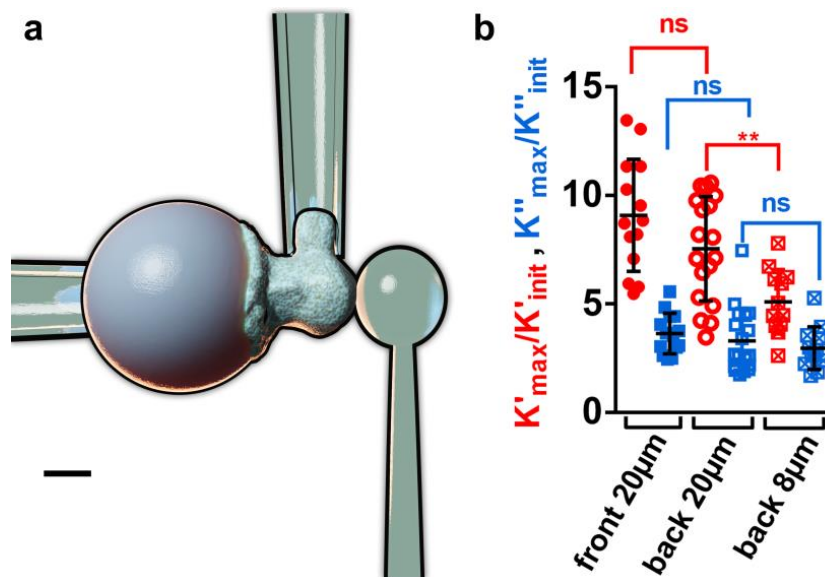
348 **Changes in  $K'$  and  $K''$  correspond to changes in intrinsic leukocyte mechanical**  
 349 **properties**

350 Both  $K'$  and  $K''$  depend on cell geometry, so to exclude the possibility that changes in  $K'$  and  $K''$  only  
 351 reflect geometrical changes, cell intrinsic mechanical properties such as Young's modulus and viscosity  
 352 have to be extracted from  $K'$  and  $K''$ . To do so, we first convert  $K'$  and  $K''$  into a storage modulus  $E'$  and  
 353 a loss modulus  $E''$ , respectively. In the case of T cells and B cells, we do so by modeling cell geometry  
 354 (Suppl. Mat. 8).

355 For PLB cells, we use a modified setup where a non-adherent and non-activating glass bead indents  
 356 the cell on its "back" (37) while the cell phagocytoses an activating bead at its "front" (Fig. 3a, Suppl.  
 357 Movie S3) (37). This alternative setup leads to the same increase in  $K'$  and  $K''$  as measured using the  
 358 original setup with front indentation with an activating bead (Fig. 3b). Having a sphere-against-sphere  
 359 geometry, we can use a linearized Hertz model to extract  $E'$  and  $E''$  moduli (Suppl. Mat. 8). Lastly, we  
 360 performed cyclic indentation experiments to measure directly the Young's modulus over time (with a  
 361 limited time resolution of  $\sim 30$  seconds, dotted curves in Fig. 2l): indentation occurs only for a short  
 362 time at the beginning of each cycle, after which the indenter retracts from the cell, marking the end of a  
 363 cycle, immediately followed by a new cycle (Suppl. Movie S4). In the case of PLB cells we tested the  
 364 influence of bead size by performing indentation in the "back" of cells phagocytosing either 20- or 8-  
 365  $\mu\text{m}$  beads. For both bead sizes the cells' Young's modulus increased significantly during phagocytosis  
 366 when comparing early ( $t=52$  s) vs. late ( $t=286$  s) times (Figure 2l,  $E_{\text{Young}}(t=286\text{ s}) > E_{\text{Young}}(t=52\text{ s})$ , at  
 367 least 14 cells from at least 3 independent experiments for each condition,  $p < 0.0001$  for 20- $\mu\text{m}$  and 8-  
 368  $\mu\text{m}$  beads, Mann Whitney test).

369 To allow a simple interpretation of  $E'$  and  $E''$ , which were obtained in a regime of oscillatory forces, we  
 370 convert them respectively into an effective Young's modulus  $E_{\text{Young}}^{eq}$  and an effective viscosity  $\nu$ . To  
 371 calculate  $E_{\text{Young}}^{eq}$ , we compare the Young's modulus measured by initial compression and  $E'$  measured  
 372 right after, when force modulation begins (Suppl. Mat. 9; Suppl. Fig. S6). For PLB cells, we used again  
 373 cyclic back-indentation as described above to measure both Young's modulus and  $E'$  not only at initial  
 374 time but during the whole activation. This showed that  $E'$  and  $E_{\text{Young}}^{eq}$  are proportional, with a phe-  
 375 nomenological coefficient  $C$  such that  $E' = CE_{\text{Young}}^{eq}$ .  $C$  is measured to be constant over time for PLB  
 376 cells, and so for T and B cells we assume that  $C$  is also constant over time (Suppl. Mat. 9).

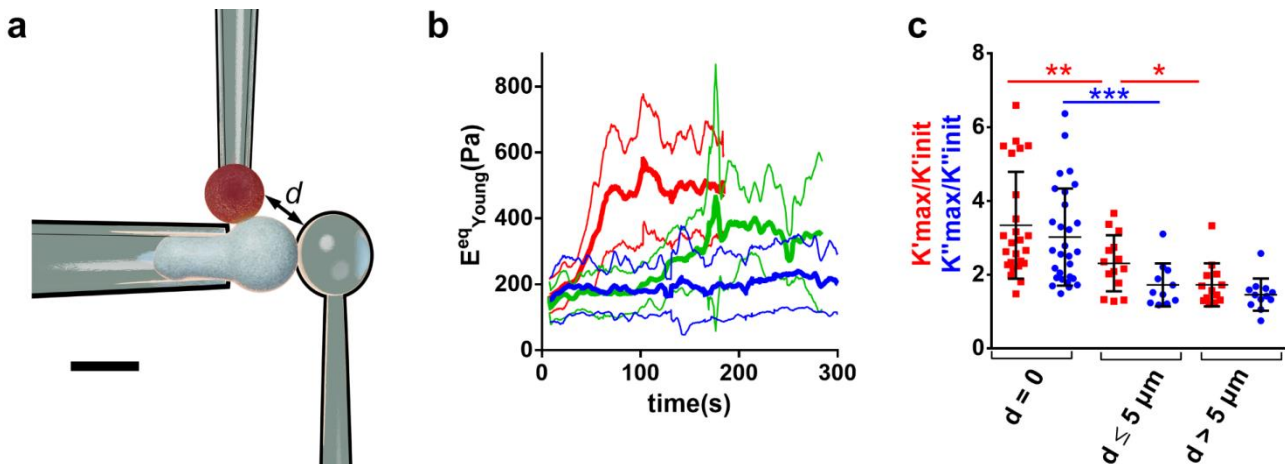
377 Using this approach, we measured the effective Young's modulus of the three cell types over time (Fig.  
 378 2j-l). To calculate the effective cell viscosity, we consider a simple model consistent with a Newtonian  
 379 liquid and cortical tension model of a cell (2), where a Newtonian viscosity is introduced as  $\nu = E''/\omega$ .  
 380 The resulting Young's modulus and viscosity increase until reaching a maximal value that is 2-3 fold  
 381 higher than the baseline value for T and B cells, and about 11 fold higher for PLB cells (Fig. 2j-l).



383 FIGURE 3. Indenting the “back” of a phagocyte during activation. (a) Modified setup. A stiff pipette holds the activating microbead (left), an auxiliary (stiff) pipette holds the cell by its “side” (top) and a flexible pipette whose tip consists of a non-adherent glass bead indents the cell on its “back” (right). Scale bar is 5  $\mu\text{m}$ . (b) Max/min ratios for  $K'$  and  $K''$  obtained with both versions of the setup during PLB cell activation (ns: non-significant, two-tailed unpaired t test). Using back indentation, in addition to the 20- $\mu\text{m}$  beads used with the non-modified setup, 8 $\mu\text{m}$  beads were tested.

### 389 Mechanical changes are localized close to the cell-bead interface

390 We asked if mechanical changes were localized close to the activation area in the leukocyte. In the case of T cells, we used a modified setup similar to the one allowing to indent PLB cells on their back (Fig. 3a). In that case an auxiliary pipette brought the activating microbead in contact with the T cell on its “side”: at the equatorial plane (distance between the activating and indenting beads smaller than 5  $\mu\text{m}$ ), or close to the tip of the holding pipette, at the farthest possible location from the activating bead (distance between the activating and indenting beads larger than 5 $\mu\text{m}$ , Fig. 4, Suppl. Movie S5). This allowed us to quantify mechanical changes during activation depending on the distance to the cell-bead contact area. We computed an equivalent Young’s modulus  $E^{eq}_{Young}$  as described in supplementary material 9, which led to a good agreement with  $E_{Young}$  measured at the initial time. T cells exhibited a different behavior depending on the location of the activating microbead: the closer the indenter was to the activating bead, the larger the maximum value of  $E^{eq}_{Young}$  measured during activation. Cell stiffening thus occurs with a different amplitude depending on the location on the cell relative to the contact zone with an activating surface.



403 FIGURE 4. (a) Modified setup where an auxiliary (stiff) pipette holds the activating microbead (top, red) to bring it in contact with the T cell at a chosen distance from a microindenter (right), which indents the cell on its “side” while the T cell gets activated. Scale bar is 5  $\mu\text{m}$ . (b) Equivalent Young’s modulus for three different ranges of distance  $d$  between the activating bead and the microindenter (red:  $d=0$ ; green:  $0 < d \leq 5 \mu\text{m}$ ; blue:  $d > 5 \mu\text{m}$ ). (c)  $K'_{max}/K'_{init}$  (red) and  $K''_{max}/K''_{init}$  (blue) ratios for the same ranges of distance  $d$  as in panel b.

### 409 Mechanical changes in T cells start within seconds

410 In T cells, mechanical changes precede morphological ones:  $K'$  increases slowly within seconds after cell-bead contact, before cell morphology starts changing (i.e. before the T cell produces a large protrusion (17), Suppl. Movie S1). The onset of this growth is followed by a short period during which  $K'$  stays relatively constant. This phase ends by a faster increase of  $K'$  during protrusion growth (Suppl. Mat. 10, Suppl. Fig. S7). The start of the faster increase in  $K'$  starts when the tail of the cell in the holding pipette starts retracting (a sign that  $K'$  and cell tension are two equivalent ways to describe cell stiffness, Suppl. Mat. 11). Of note, T cells have large membrane surface stores (38), so it is unlikely that the acceleration in  $K'$  increase could be explained by reaching the limits of membrane stores (Suppl. Mat. 12).

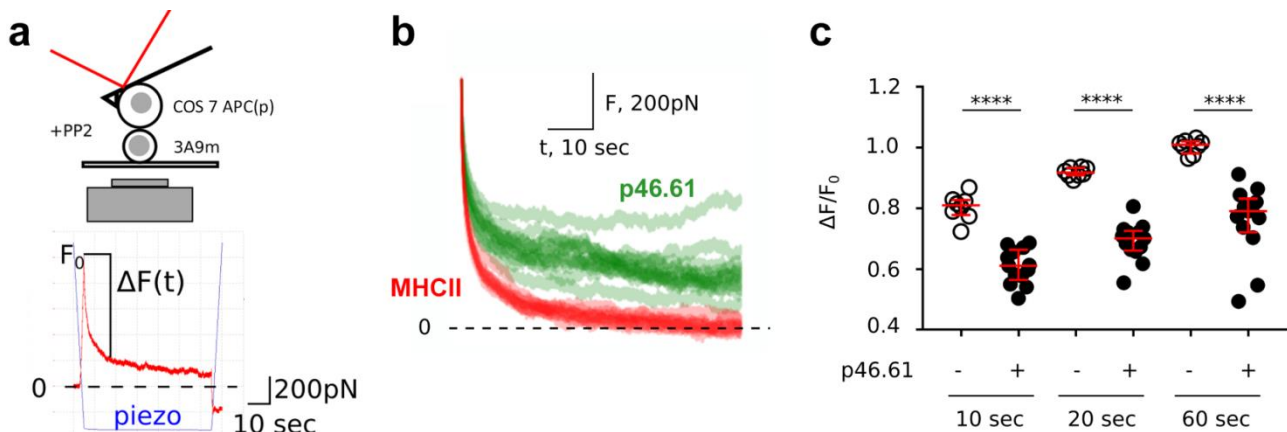
## 419 AFM measurements of early mechanical changes following T cell/APC contact

420 In order to confirm with an independent technique that T cell mechanics may be modulated within  
 421 seconds when it encounters an antigen presenting cell (APC) before any large morphological change  
 422 occurs, we used AFM and performed Single Cell Force Spectroscopy (SCFS) experiments (Fig. 5a, Suppl.  
 423 Mat. 3)(36). As a model APC, we used COS-7 cells expressing MHCII molecules that can be loaded with  
 424 peptides of desired affinity as previously shown (47, 48). We used murine 3A9m T cells and immobi-  
 425 lized them on a coverslip by adhering them using anti-CD45 antibodies (53). To measure rapid me-  
 426 chanical changes using this technique, we prevented potential large active cell deformations by using  
 427 the pan-Src kinase inhibitor PP2 (methods). Although PP2 inhibits TCR-mediated Lck- or Fyn- dependent  
 428 intracellular phosphorylation events and downstream signaling cascades (54), it does not inhibit all molecu-  
 429 lar events such as migration arrest, kinapse or microcluster formation upon T cell activation (55–57). Wash-  
 430 out experiment showed that PP2 in fact enables to pause signalization without abrogating the antigen de-  
 431 pendence of early T cell activation (58, 59).

432 The SCFS experiments provide for each cell doublet the force-tip sample separation curve,  
 433 which is the AFM equivalent of the force-indentation curve shown in Fig. 1b (inset, bottom left), with  
 434 three parts : (i) pushing the cells together, (ii) maintaining the contact and (iii) detaching the cells by  
 435 pulling them apart. The contact mechanics at initial time was quantified by measuring the slope of the  
 436 force-tip sample separation curve (called contact slope herein), which reflects the stiffness of the cell  
 437 doublet. This contact slope was not modified by the presence or absence on the APC of the HEL (Hen  
 438 Egg Lysozyme, peptide p46.61)-derived peptide recognized by the murine 3A9m T cells (Supp. Fig.  
 439 S2d). However, in the separation part of the force curves, the presence of the peptide induced an in-  
 440 crease in the force needed to fully detach the two cells, in parallel of an increase of the number of dis-  
 441 crete separation events, indicating that the interaction was indeed peptide specific (Suppl. Fig. S2e-f).

442 The measurement of the contact slope corresponds to the very first instants of cell-cell contact  
 443 (Fig. 5a, bottom). To measure cell mechanical changes following this contact, we quantified the force  
 444 relaxation when the AFM lever motion was stopped and piezo position was kept constant after reach-  
 445 ing a contact force of 1nN (Fig. 5b-c). We observed a striking difference in this force relaxation depend-  
 446 ing on the presence vs. absence of a saturating concentration of the antigenic peptide (Fig. 5b): the  
 447 force relaxation was much slower in presence of the antigenic peptide, as quantified by the lower val-  
 448 ue of the ratio  $\Delta F/F_0$  at different times following the start of the relaxation (Fig. 5c). This measurement  
 449 reflects global viscoelastic properties of the cell-cell doublet, but does not disentangle elastic and vis-  
 450 cous properties. Furthermore, having the two cells pressed against each other in series does not allow  
 451 simply excluding a contribution of a mechanical change in the APC. However, mechanical properties of  
 452 APC COS-7 cells were not modified by the presence of the peptide (Suppl. Fig. S3). As a consequence,  
 453 we expect that the major contribution to the difference in force relaxation is indeed due to the T cell.  
 454 Our SCFS experiments are thus consistent with our Micropipette Force Probe experiments, and show  
 455 that specific recognition during T cell/APC contact may induce mechanical changes, within few sec-  
 456 onds, and without any TCR signaling.

457



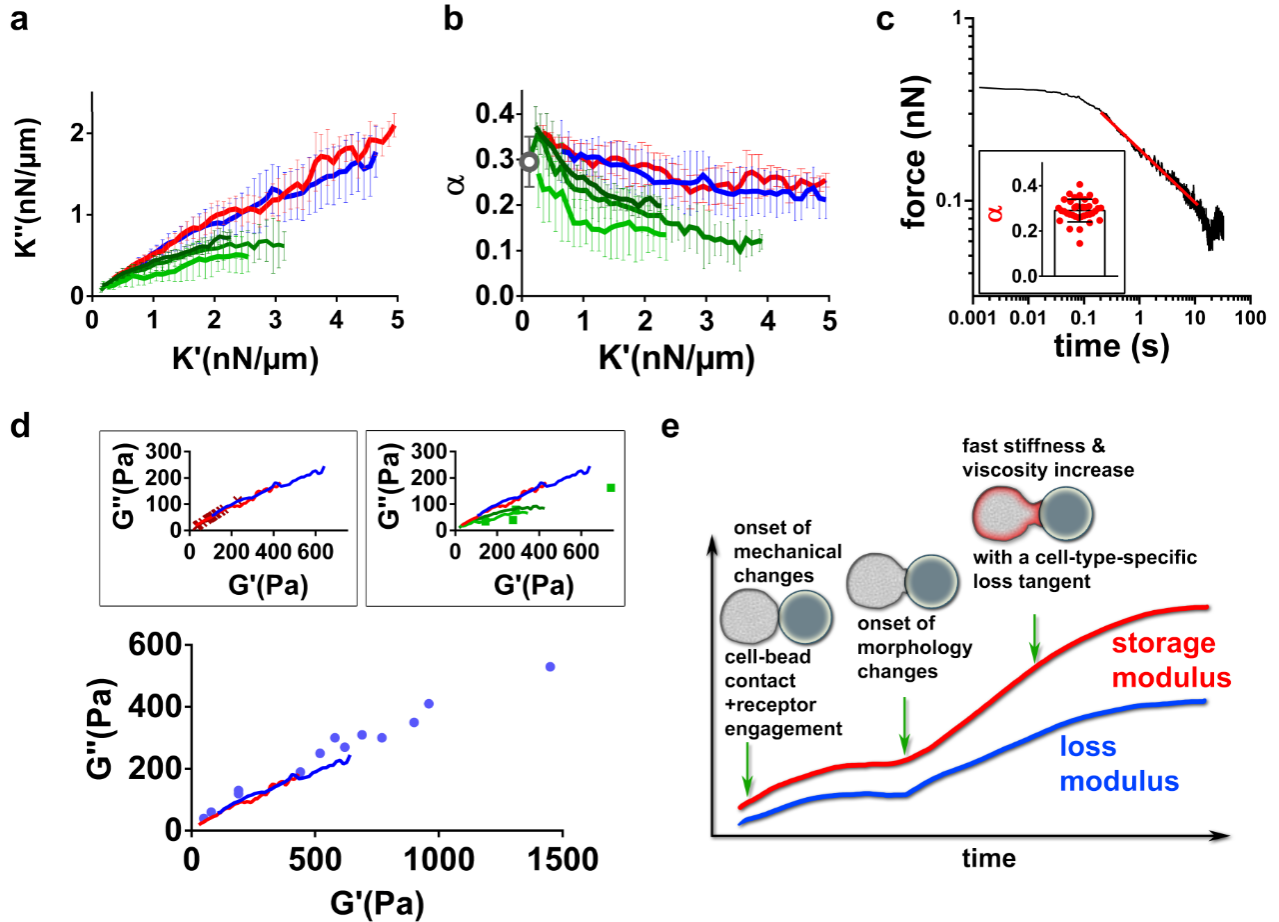


459 FIGURE 5. Cell-cell presentation induces early changes in force relaxation. (a) AFM SCFS Setup (top) and example  
 460 of individual force (in red) vs. time curve (bottom). The piezo signal (in blue) reports the position of the base of  
 461 the cantilever. (b) Relaxation part of force curves, for peptide (p46.61) vs. no-peptide (MHCII) cases. One curve  
 462 corresponds to one cycle, ie. 1 APC/T cell couple. See Suppl. Fig. S2h for plot of mean±SD curves. (c) Quantifica-  
 463 tion of the relaxation for three different time points, for MHCII (-) and MHCII/p46.61 (+);  $\Delta F/F_0$  is the ratio of the  
 464 drop in force  $\Delta F$  and the force level  $F_0$  imposed at initial time 0 (see a). This ratio is shown at three different time  
 465 following cell-cell contact: 10, 20, and 60s, and shows a consistent lower ratio (hence a slower relaxation) in  
 466 presence of the peptide (\*\*\*\* for  $p < 0.001$ ).

## 467 Relationship between elastic and viscous properties during leukocyte activation

468 We then asked if variations in elastic and viscous properties were related to each other. Prior works  
 469 show that in several cell types the loss modulus  $E''$  and storage modulus  $E'$  of a cell are related to each  
 470 other. In fact their ratio  $\eta = \frac{E''}{E'}$ , the loss tangent, lies within a narrow range of 0.1-0.3(6, 28, 31, 60-62).  
 471 For a material such as a purely viscous liquid, the storage modulus vanishes so that  $\eta$  is arbitrarily  
 472 large, while for a purely elastic material, the loss modulus vanishes and so does  $\eta$ . A finite and constant  
 473 value of  $\eta$  for a cell implies that the stiffer the cell, the more viscous it is. This narrow range of loss  
 474 tangent is consistent with the conserved ratio of cortical tension to cell viscosity in leukocytes (see  
 475 introduction). In biological matter, similar to soft glassy materials (63, 64), stress relaxation over time  
 476 often follows a power-law (with an exponent that we shall call  $\alpha$ )(31, 32, 60, 65, 66). The loss tangent  
 477  $\eta$  is frequency-dependent, and is expected to be approximately equal to  $\tan\left(\frac{\pi}{2}\alpha\right)$  at low frequencies  
 478 (Suppl. Mat. 13). Therefore, by measuring the power-law exponent  $\alpha$  independently, we should recover  
 479 the value of  $\eta = \frac{E''}{E'}$  (which should also be equal to  $\frac{K''}{K'}$  as the contribution from geometry is elimi-  
 480 nated in this ratio(7)). To test this prediction, we performed stress relaxation experiment on resting  
 481 PLB cells (Fig. 6c) and obtained values for  $\alpha$  that were very consistent with the predicted value of  
 482  $\frac{2}{\pi} \arctan\left(\frac{K''}{K'}\right)$  (which is equivalent to comparing  $\tan\left(\frac{\pi}{2}\alpha\right)$  and  $\frac{K''}{K'}$ , gray circle in Fig. 6b). We then  
 483 compared  $K''$  vs.  $K'$  curves obtained during activation of T cells, B cells, and PLB cells (Fig. 6a). In Figure  
 484 6b we plot the corresponding value of  $\alpha = \frac{2}{\pi} \arctan \eta = \frac{2}{\pi} \arctan \frac{K''}{K'}$ : these curves are remarkably con-  
 485 sistent for T and B cells; for PLB cells,  $K''$ - $K'$  curves obtained by indentation in the front and in the back  
 486 of the cell are very similar, and differ from the T and B cell common curve. Interestingly, Bafi *et al.* (67)  
 487 measured storage and loss moduli of various leukocytes at resting state or after priming by different  
 488 inflammatory signals. Their data lead to a loss tangent  $\eta$  consistent with our measurements (Fig. 6d)  
 489 but their measurements were made at a single time point. Maksym *et al.* (6) tracked over time both the  
 490 loss and storage moduli of human airway smooth muscle cells contracting due to administration of  
 491 histamine (Fig. 6d, inset, top left). Their data are again very consistent with our measurements. Roca-  
 492 Cusachs *et al.* (2) used AFM on neutrophils, and found a loss tangent very consistent with our meas-  
 493 urements on PLB cells obtained with 20- $\mu\text{m}$  and 8- $\mu\text{m}$  beads (Fig. 6d, inset, top right).  
 494 Based on our results, we propose a simple model of leukocyte mechanical changes during activation  
 495 (Fig. 6e): mechanical changes begin seconds after receptor engagement at the leukocyte surface. Cell  
 496 morphological changes start a few tens of seconds later, concomitant with stiffening and increase in  
 497 viscosity. Viscous and elastic properties of the leukocyte evolve following a characteristic relationship  
 498 defined by the loss tangent.

499



501

502 FIGURE 6. Relationship between elastic and viscous cell properties (a)  $K''$  vs.  $K'$  during activation of T cells (red),  
 503 B cells (blue) and PLB cells (front indentation: green (20- $\mu\text{m}$  beads), back indentation: light green (20- $\mu\text{m}$  beads)  
 504 and dark green (8- $\mu\text{m}$  beads)). Error bar are SDs. (b)  $\alpha = \frac{2}{\pi} \arctan\left(\frac{K''}{K'}\right)$  corresponding to the same data as in a.  
 505 The gray circle is the value of the power-law exponent in force relaxation experiments on resting PLB cells. (c)  
 506 Example of force relaxation curve of a PLB cell. In the log-log plot, a power-law relaxation is identified by the  
 507 straight line (red), whose slope is the exponent  $\alpha$ . Inset: individual  $\alpha$  values (1 dot per cell). (d) Loss modulus  $G''$   
 508 vs. storage modulus  $G'$  obtained by Bui *et al.* (67) (bottom, blue dots), Maksym *et al.* (6) (inset, top left, red cross-  
 509 es), and Roca-Cusachs *et al.* (2) (inset, top right, green dots). Our data from a (red and blue solid lines) are con-  
 510 sistent with the above-mentioned published data. (e) Proposed model of mechanical changes during leukocyte  
 511 activation.

## 512 Discussion

513 We showed that during activation, various types of leukocytes see their stiffness and viscosity increase  
 514 significantly within few minutes. The initiation of mechanical changes upon contact with an activating  
 515 surface starts within seconds and these changes precede morphological changes. By using the same  
 516 technique – avoiding potential differences due to different techniques (3) – we show to our knowledge  
 517 for the first time with  $\sim 1$ -s time resolution how both viscous and elastic properties evolve during im-  
 518 mune cell activation. We further show that elastic and viscous properties follow a relationship that is  
 519 cell-type dependent: T and B cells keep the same ratio between viscous and elastic properties, while  
 520 neutrophils follow a different ratio, independently of the size of their target.

521 Most published measurements of cell viscosity were performed on leukocytes in a resting state.  
 522 Some micropipette aspiration studies led to leukocyte viscosity in the range of 100-200 Pa.s (68–71),  
 523 and are consistent with values obtained in fibroblasts (72). These values are higher than the viscosi-  
 524 ties of  $\sim 5$ -20 Pa.s that we measured in resting leukocytes. This discrepancy might be explained by  
 525 model-dependence and by differences in the shear rate applied to cells. Our measurements are much

526 closer to results by other groups: Sung *et al.* (73) obtained a cytotoxic T cell viscosity of  $\sim 30$  Pa.s,  
527 Schmid-Schönbein *et al.* (25) measured a leukocyte viscosity of  $\sim 13$  Pa.s, Hochmuth *et al.* (29) ex-  
528 trapolated a value for neutrophil viscosity of  $\sim 60$  Pa.s. Interestingly, Lipowsky *et al.* (74) did not re-  
529 strict to baseline viscosity and studied neutrophils stimulated by the chemoattractant FMLP. The au-  
530 thors reported a neutrophil viscosity increasing from  $\sim 5$  Pa.s at resting state to  $\sim 70$  Pa.s upon stimula-  
531 tion.

532 Mechanical changes might not have a function *per se*, but might be concomitant to force gener-  
533 ation by cells. Indeed, cell contractility and cell stiffness are correlated in muscle cells (75–79), and  
534 single cells mirror mechanical and energetic features of muscle contraction: single myoblasts follow a  
535 single-cell equivalent of a Hill relationship between force generation and rate of contraction (80, 81).  
536 Furthermore, it also appears that single muscle cells become stiffer when they contract: Shroff *et al.*  
537 (79) observed a roughly 2-fold increase in stiffness of a cardiomyocyte during its contraction. Alt-  
538 hough leukocytes have a different organization of their actomyosin cytoskeleton, it is tempting to  
539 speculate that all leukocytes share generic cytoskeletal-based properties requiring that cells stiffen to  
540 generate force. This could explain the apparent contradiction that in leukocytes, the stiffening that we  
541 have quantified occurs in situations requiring an efficient spreading. Indeed, phagocytes need to in-  
542 crease their surface by spreading on their prey to engulf it, while T and B cells need to spread widely  
543 on the cell they encounter to quickly scan a surface that is as large as possible on the cell they interact  
544 with (18, 82). Alternatively, forming a stiff synapse might allow its stabilization (83, 84).

545 Although the molecular details leading to changes in viscoelastic properties are yet to be un-  
546 derstood, cell-scale physical considerations allowed Evans *et al.* (71, 85) to conclude that viscous  
547 properties of cells are conferred by bulk components of the cell, as opposed to surface viscosity due to  
548 the actin cortex. As a result, we expect that the large increase in viscous properties of leukocytes that  
549 we observed are largely due to intracellular reorganization, not only to cortical reorganizations. Inter-  
550 estingly, Tsopoulidis *et al.* recently showed that T cell receptor engagement triggers the formation of a  
551 nuclear actin network in T cells (86), which could contribute to these bulk mechanical changes.  
552 Fritzsche *et al.* observed submembrane actin pattern reorganization that influenced the membrane  
553 architecture of Hela cells, but not their mechanical properties (87). These observations may lift an  
554 apparent contradiction in T cells, in which the formation of an immunological synapse requires actin to  
555 depolymerize at the center of the synapse, including in cytotoxic T cells where actin gets actually de-  
556 pleted across the synapse within 1 min (88). Other contributors must then explain the stiffening that  
557 we observe, and in addition to the nuclear actin network (86), another possible contributor being a  
558 secondary actin network. Indeed Fritzsche *et al.* (84) found in T cells that beside the dense cortical  
559 network forming a rosette-shaped structure in the basal lamellipodium and devoid of actin at its cen-  
560 ter up to 3 min after initial contact, another actin network assembles 150 to 300 nm above the cell-  
561 substrate contact.

562 We expect inner reorganizations causing mechanical changes to impact inner cell dynamics,  
563 such as organelle trafficking. This includes mitochondrial relocation that is known to happen in several  
564 types of leukocytes upon activation (89, 90). Regarding cell surface, although we dismissed surface  
565 effects as key contributors to cell viscosity, surface receptors are transmembraneous so that the for-  
566 mation of diffusional barriers with slowed diffusion at the cell surface as observed during leukocyte  
567 activation could be one manifestation of the increase in cell viscosity (91, 92). As an alternative to the  
568 above considerations that mechanical changes might not have a specific function, these changes in  
569 viscosity might help confining molecules of interest in the vicinity of the synapse. By sequestering  
570 these molecules due to higher stiffness and viscosity (implicating an increased energetical cost to  
571 move away from this zone), early mechanical changes might help integrating at the cellular scale the  
572 information input incoming from the single-receptor scale at the synapse. Considering this efficient  
573 processing from nano- to micro-scale is also worth integrating in mechanistic description of early im-  
574 munological signaling, including models considering a frictional coupling of the TCR to the cytoskele-  
575 ton (93, 94).

576 Our study highlights that variations of both elastic and viscous properties are relevant to un-  
577 derstand immune cell mechanics during activation, as well as their relationship in the form of the loss

578 tangent  $\eta$ , and how the latter evolves over time. All the leukocytes that we tested show very large  
579 changes in both elastic and viscous properties, but monitoring the time evolution of the loss tangent  
580 exhibits a mechanical fingerprint specific to cell types. It is tempting to speculate that the evolution of  
581 the loss tangent during activation can become a valuable tool to discriminate between healthy and  
582 pathological leukocytes as already suggested by considering static values of  $\eta$  (95).

583 Finally, our observations suggest a possible generalization to cells other than leukocytes. In  
584 line with what was observed by Thoumine et al. (96) in fibroblasts or Abidine et al. in epithelial cancer  
585 cells (97), we propose that getting stiffer and more viscous while spreading is a common behavior  
586 across cell types. The time evolution of the loss tangent during cell spreading remains to be explored.

## 587 **Author contributions**

588 P.-H.P., Y.H. and J.H. designed experiments; A.Z., S.V.M.C., A.S., F.M., Y.H., P.-H.P. and J.H. performed ex-  
589 periments; A.Z., A.S., Y.H., P.-H.P. and J.H. analyzed the data; A.B., S.D., E.H., S.D.-C., H.-T.H., A.I.B., Y.R.C.,  
590 Y.H., C.H. and O.N. contributed material; P.-H.P. and J.H. wrote the manuscript with helpful critical  
591 reading by the other authors.

## 592 **Competing financial interests**

593 The authors declare no competing financial interests.

## 594 **Acknowledgements**

595 This work has benefited from the financial support of the Labex LaSIPS (ANR-10-LABX-0040-LaSIPS)  
596 managed by the French National Research Agency under the "Investissements d'avenir" program  
597 (n°ANR-11-IDEX-0003-02), from a CNRS PEPS grant, from Ecole Polytechnique, and from an endow-  
598 ment in cardiovascular bioengineering from the AXA Research Fund. This work was supported by  
599 funding from (a) Prise de Risques CNRS and ANR JCJC « DissecTion » (to PHP) ; (b) the Labex IN-FORM  
600 (ANR-11-LABX-0054) and A\*MIDEX project (n° ANR-11-IDEX-0001-02), sponsored by the Investisse-  
601 ments d'Avenir project funded by the French Government, managed by the French National Research  
602 Agency (ANR) (to LAI & CIML, and as PhD grant to AS) ; (c) the European Union's Horizon 2020 re-  
603 search and innovation programme under the Marie Skłodowska-Curie grant agreement No713750,  
604 with the financial support of the Regional Council of Provence- Alpes-Côte d'Azur and with of the  
605 A\*MIDEX (n° ANR- 11-IDEX-0001-02), funded by the Investissements d'Avenir project funded by the  
606 French Government, managed by the French National Research Agency (ANR) (as PhD grant to FM).  
607 Part of this work was also supported by institutional grants from INSERM, CNRS and Aix-Marseille  
608 University to the LAI and CIML. Material or technical help: R. Balland (Sensome) for RBCs; M. Pélicot-  
609 Biarnes (LAI); L. Borge (PCC cell culture facility); S. Mailfert / M. Fallet / PICSL imaging facility of the  
610 CIML (ImagImm), member of the national infrastructure France-BioImaging supported by the French  
611 National Research Agency (ANR-10-INBS-04); A. Formisano (He/Marguet lab). Discussion and support:  
612 D. Gonzalez-Rodriguez (LCP-A2MC, Univ. De Lorraine), R Roncagalli (CIML); P. Recouvreux (IBDML); L.  
613 Limozin, F. Rico (LAI). Companies: F. Eghiaian, T. Plake & JPK Instruments (Berlin, Germany), now part  
614 of Bruker, for continuous support and generous help; Zeiss France for support. SVMC and YRC have  
615 benefited from the financial support of an FPI contract (BES-2014-068006) and the grant BFU2013-  
616 48828-P from the Spanish Ministry of Economy (MINECO).

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