IL-1α induces thrombopoiesis through megakaryocyte rupture in response to acute platelet needs

Satoshi Nishimura,1,2,5,6 Mika Nagasaki,1,3 Shinji Kunishima,7 Akira Sawaguchi,8 Asuka Sakata,5
Hiroyasu Sakaguchi,9 Tsukasa Ohmori,5 Ichiro Manabe,1 Joseph E. Italiano Jr.,10 Tomiko Ryu,11 Naoya Takayama,12
Issei Komuro,1,2 Takashi Kadowaki,2,4 Koji Eto,12 and Ryozo Nagai5

1Department of Cardiovascular Medicine, 2Translational Systems Biology and Medicine Initiative, 3Computational Diagnostic Radiology and Preventive Medicine,
4Department of Diabetes and Metabolic Diseases, The University of Tokyo, Tokyo 113-8654, Japan
5Center for Molecular Medicine, Jichi Medical University, Tochigi 329-0498, Japan
6Japan Science and Technology Agency (JST), Precursory Research for Embryonic Science and Technology (PRESTO), Saitama 332-0012, Japan
7Department of Advanced Diagnosis, Clinical Research Center, National Hospital Organization Nagoya Medical Center, Nagoya 460-001, Japan
8Department of Anatomy, Ultrastructural Cell Biology, Faculty of Medicine, University of Miyazaki, Miyazaki 889-1692, Japan
9Web Solution Group, IMAGICA Imageworks, Tokyo 141-0022, Japan
10Division of Hematology, Department of Medicine, Brigham and Women’s Hospital, Vascular Biology Program at Boston Children’s Hospital, Harvard Medical School, Boston, MA 02215
11Internal medicine, Social Insurance Central General Hospital, Tokyo 105-8330, Japan
12Center for iPS Cell Research and Application, Kyoto University, Kyoto 606-8507, Japan

Introduction

Circulating platelet counts and thrombopoietic processes in BM megakaryocytes (MKs) are both tightly regulated. In vitro, thrombopoiesis occurs via proplatelet formation (PPF) in the presence of thrombopoietin (TPO), which entails microtubule-dependent extension of elongated pseudopodal structures that exhibit platelet-sized swellings arranged in tandem and containing platelet organelles (Patel et al., 2005; Thon et al., 2010; Machlus et al., 2014). In vivo studies using two-photon microscopy also confirmed the presence of PPF in mouse BM (Junt et al., 2007; Zhang et al., 2012). However, the estimated platelet number released from each MK cannot explain rapid platelet turnover, especially when the need is acute, such as during inflammatory reactions. We therefore suspected that there is another rapid thrombopoietic mode, in addition to PPF.

Although TPO has been identified as the most important regulator of platelet production (de Sauvage et al., 1994; Kuter, 2007), it was recently reported that MK maturation and platelet biogenesis can occur independently of TPO (Ng et al., 2014). Additional studies using two-photon microscopy also confirmed the presence of PPF in mouse BM (Junt et al., 2007; Zhang et al., 2012). However, the estimated platelet number released from each MK cannot explain rapid platelet turnover, especially when the need is acute, such as during inflammatory reactions. We therefore suspected that there is another rapid thrombopoietic mode, in addition to PPF.
Figure 1. Proplatelet formation is the dominant mode of thrombopoiesis, but there is an alternative megakaryocyte rupture mode, which produces much larger numbers of platelet-like particles. (A–G) Time-lapse images of thrombopoiesis in living BM from 6-wk-old CAG-eGFP (green) mice under steady-state conditions (A, B, F, and G) or after treatment with TPO (C; 10 µg for 5 d; A, Video 1; B, Video 2; C, Video 3; F, Videos 4 and 5; and G, Video 6). Injected fluorescent dextran (red) shows the blood flow, and Hoechst (blue) labeled the nucleus. Slice views (top in A, C, and G), voxel views (bottom in B and F), and surface views (bottom row in A, B, C, and G) show MK surfaces and particle release at the single-platelet (triangle) level. (D and E) Numbers of particles released from MKs with proplatelet formation and MK rupture thrombopoiesis, which were calculated from visuals by automatic software. n = 50 cells from 5 animals in each group. Note that MK rupture thrombopoiesis is rapid and associated with much greater numbers of released particles. (H) Automatic software analysis of thrombopoiesis mode. Calculated changes in MK perimeters and cytoplasmic GFP signal intensities are shown. The long arm projections (>50% of the length of the mean MK diameter) were identified as proplatelets and divided into short (<100 µm) and long (>100 µm) proplatelet formation. Increases in the perimeter (deformity) and decreases in GFP intensity (during rupture) were identified as the MK rupture pattern.
However, no suggestions as to the detailed mechanism by which platelets are generated from MKs in the absence of TPO were provided. In addition, recent studies indicate that hematopoietic stem cells (HSCs) and MKs are in very close proximity within the hematopoiesis hierarchy, and that MKs and platelets can emerge directly from HSCs under stress conditions, e.g., after BM suppression by irradiation (Sanjuan-Pla et al., 2013; Yamamoto et al., 2013; Nakamura-Ishizu et al., 2014). Thus, the actual pathways of platelet biogenesis are not clear, and elucidation of unidentified thrombopoietic mechanisms, particularly under stressful conditions such as inflammation or acute thrombocytopenia, require direct visualization of the BM. We therefore endeavored to improve the capability of the two-photon microscopy technique such that we would be able to visualize platelet biogenesis from MKs at the single-platelet level and to trace the translocation of platelets into the blood circulation of the BM in living mice. Ultimately, we identified an alternative pathway entailing MK rupture–enhanced platelet release, which responded to acute platelet needs under regulation by IL-1α. Our results shed light on what appears to be a novel mode of platelet release from BM MKs.

Results

Proplatelet type thrombopoiesis continuously regulates the platelet supply but provides limited numbers of platelets from mature MKs

To address the mechanism by which rapid platelet turnover is regulated, especially under stress conditions, we visualized megakaryopoiesis and dynamic thrombopoiesis in three dimensions (3D) using an improved intravital visualization technique and focusing on BM MKs. The combined technologies of multicolor high-sensitivity GaAs detectors, resonance mirror high-speed scanners, and a piezo-drive electronically controlled stage were applied to CAG-eGFP and CD41-tandem(td)Tomato mice, enabling us to monitor the behavior of single platelets shed from BM MKs (Fig. 1, A–C; Fig. S1; and Videos 1–3). MKs identified based on their large size, multinucleation, CD41 positivity, and strong GFP signals in CAG-eGFP mice were mainly located in the border area between BM vessel lumens and the stroma.

With no intervention under steady-state conditions, filamentous (elongated) proplatelets released small platelet-like particles from the tips of the protrusions within vessels (Fig. 1, A and B). The entire time course of this process was usually longer than the observation periods (1 h), during which release was intermittent, and the number of released particles was only 1.4 ± 0.3 per minute from a single MK. TPO administration increased PPF with development of longer (>100 µm) arm projections, but the numbers of released platelets (2.6 ± 0.5 per minute) were still small (Fig. 1, C and D), which prompted us to search for alternative modes of thrombopoiesis.

MK rupture, the alternative mode of thrombopoiesis, can release large numbers of platelets over a short period

Rigorous 3D and high-speed examinations and surface visualization revealed that there is another, minor, but distinct, mode of thrombopoiesis, which we named “MK rupture” thrombopoiesis (Fig. 1, D–G). During the rupture phenomenon, MKs exhibited ruffling and then irregular changes in cell shape, and GFP+ platelet-like particles were released from the cytoplasm primarily into BM vessel lumens (Fig. 1, F and G; and Videos 4–6). Thereafter, MKs showed a marked loss of the GFP signal from the cytoplasm along with an increase in the dextran signal. When we profiled the visuals of the thrombopoiesis process in MKs using automatic, software-based algorithms, we found that short PPF was the dominant mode of platelet biogenesis in the steady state (Fig. 1, H and I), but that MK rupture thrombopoiesis also occurred with increased particle number per a single MK (Fig. 1 D). Moreover, the MK rupture thrombopoiesis was distinct from typical FasL-induced apoptosis, which had a much longer time course and was associated with blebbing, large protrusions, and gradual GFP loss (Fig. 1, J and K).

IL-1α contributes to MK rupture thrombopoiesis in response to acute platelet need

To determine whether TPO is involved in MK rupture thrombopoiesis, or whether other intrinsic factors are involved, we next sought mediators in BM cell culture medium that positively influenced MK production in the presence of TPO. Using this screening assay, we identified seven candidate factors and determined that IL-1α increases platelet production from MKs to an even greater degree than TPO (Fig. 3, A and B). Moreover, serum IL-1α levels were acutely increased during platelet recovery after depletion using an anti-CD42b antibody, whereas increases in TPO levels were not seen until day 7 (Fig. 3 C). Increases in serum IL-1α levels induced in response to acute peritoneal inflammation evoked by thioglycollate injection were accompanied by transient increases in platelets (Fig. 3 D). In addition, neutralizing anti–IL-1 receptor (IL-1R) antibody...
IL-1α–dependent platelets were larger in size than those in TPO-treated mice (Fig. 4 J and K), and anti-CD42b antibody increased the large platelet populations in control mice but not in IL-1R1−/− mice (Fig. S2). IL-1α–dependent platelets also had shorter lifetimes, which was not affected by clodronate-mediated macrophage suppression (Fig. 4 L), confirming the intrinsic nature of the lifetime. The particles released in vivo stained positively with mitochondrial membrane potential dyes, indicating intact mitochondrial function (Fig. S3 A). Platelets isolated from IL-1α–treated mice showed less JONA binding, but overall platelet aggregation in vitro and thrombus formation in vivo were comparable to the TPO-mediated responses. Thus, IL-1α stimulation leads to production of larger platelets with minor impairment but intact thrombotic function (Fig. S3).

IL-1α–induced MK differentiation, maturation, and MK rupture–dependent platelet biogenesis with PPF inhibition in vitro

IL-1α–induced MK rupture thrombopoiesis was also observed in fetal liver MKs, which enabled us to examine its molecular mechanism and to clarify differences from FasL-induced apoptosis (Fig. 5, A–D). As mentioned, the particles released from IL-1α–treated MKs were larger than those induced by TPO (Fig. 5 E). IL-1α increased the ploidy and number of MKs (Fig. 5, F and G), as well as the release of platelet-sized CD41+CD42b+ particles (Fig. 5 H), indicating positive effects of IL-1α on MK differentiation, maturation, and platelet biogenesis, i.e., IL-1α increased MK rupture thrombopoiesis.
IL-1α stimulation induces caspase-3 activation without evident apoptosis

RT-PCR analysis of cultured fetal liver MKs showed that IL-1α increased expression of *Gata1*, *Bak1*, and *Bax*, which are known to be involved in thrombopoiesis and preapoptotic gene activation (Fig. 6 A; Kaushansky, 2003). Although there has been controversy regarding the caspase cascade in thrombopoiesis (De Botton et al., 2002; Clarke et al., 2003; Josefsson et al., 2014), IL-1α induced caspase-3 as well as p53 activation and AKT/ERK phosphorylation via IL-1R1 (Fig. 6, B–D; and Fig. S4). siRNA-mediated knockdown of IL-1R1 efficiently suppressed the pAKT–pERK signal (Fig. 6 C and Fig. S4), but *Thpo* and *Il1r1* expression in fetal liver MKs and TPO-R expression in isolated platelets were unaltered by IL-1α (Fig. 6, E and F). Immunofluorescence experiments also revealed the activation of caspase-3 and release of von Willebrand factor–positive granules, but TUNEL staining was negative, which makes this process different from typical FasL-induced apoptosis (Fig. 6, G–J). Caspase inhibition using Z-VAD (OMe)-FMK suppressed the effect of IL-1α on platelet counts in vivo (Fig. 6 K) and platelet-like particle releases in vitro (Fig. 5 H). Indeed, IL-1α–induced MK rupture thrombopoiesis was blocked in *Casp-3−/−* mice but not *Thpo−/−* mice (Fig. 6 L). These results indicate the rupture machinery is associated with caspase-3 activation via IL-1α/IL-1R1 but is distinct from TPO-dependent signaling and typical apoptosis.

IL-1α-treated MKs show impairment of proportional microtubule assembly

We next asked how IL-1α–IL-1R1 signaling with preapoptotic gene activation changes the mode of platelet generation from PPF to rupture (Fig. 7). It was previously shown that PPF-mediated platelet generation is microtubule dependent (Patel et al., 2005; Thon and Italiano, 2010; Kunishima et al., 2014; Machlus et al., 2014). To determine whether IL-1α influences microtubule dependency, two microtubule inhibitors (colchicine or paclitaxel), were administered to mice or fetal liver MKs with or without subsequent IL-1α treatment, after stimulation induced caspase-3 activation without evident apoptosis. RT-PCR analysis of cultured fetal liver MKs showed that IL-1α increased expression of *Gata1*, *Bak1*, and *Bax*, which are known to be involved in thrombopoiesis and preapoptotic gene activation (Fig. 6 A; Kaushansky, 2003). Although there has been controversy regarding the caspase cascade in thrombopoiesis (De Botton et al., 2002; Clarke et al., 2003; Josefsson et al., 2014), IL-1α induced caspase-3 as well as p53 activation and AKT/ERK phosphorylation via IL-1R1 (Fig. 6, B–D; and Fig. S4). siRNA-mediated knockdown of IL-1R1 efficiently suppressed the pAKT–pERK signal (Fig. 6 C and Fig. S4), but *Thpo* and *Il1r1* expression in fetal liver MKs and TPO-R expression in isolated platelets were unaltered by IL-1α (Fig. 6, E and F). Immunofluorescence experiments also revealed the activation of caspase-3 and release of von Willebrand factor–positive granules, but TUNEL staining was negative, which makes this process different from typical FasL-induced apoptosis (Fig. 6, G–J). Caspase inhibition using Z-VAD (OMe)-FMK suppressed the effect of IL-1α on platelet counts in vivo (Fig. 6 K) and platelet-like particle releases in vitro (Fig. 5 H). Indeed, IL-1α–induced MK rupture thrombopoiesis was blocked in *Casp-3−/−* mice but not *Thpo−/−* mice (Fig. 6 L). These results indicate the rupture machinery is associated with caspase-3 activation via IL-1α/IL-1R1 but is distinct from TPO-dependent signaling and typical apoptosis.

IL-1α–treated MKs show impairment of proportional microtubule assembly.

We next asked how IL-1α–IL-1R1 signaling with preapoptotic gene activation changes the mode of platelet generation from PPF to rupture (Fig. 7). It was previously shown that PPF-mediated platelet generation is microtubule dependent (Patel et al., 2005; Thon and Italiano, 2010; Kunishima et al., 2014; Machlus et al., 2014). To determine whether IL-1α influences microtubule dependency, two microtubule inhibitors (colchicine or paclitaxel), were administered to mice or fetal liver MKs with or without subsequent IL-1α treatment, after
Figure 4. IL-1α-induced MK rupture yields larger platelets. (A–C) Time-lapse images of thrombopoiesis in living BM from 6-wk-old CAG-eGFP mice treated with IL-1α (10 µg/mouse s.c. daily for 5 d). (A, Video 8; B, Video 9; and C, Video 10.) (D) Quantification of MK dynamics and numbers and platelet counts in 6-wk-old CAG-eGFP mice treated with TPO (10 µg/mouse s.c. daily for 5 d [TPO10]), or with 70 µg/mouse daily for 3 d [TPO70]) or IL-1α (10 µg/mouse s.c. daily for 5 d). The BM was visualized and platelet counts were analyzed 7 d after the first administration. n = 50 high-power fields from 5 animals for each group. *, P < 0.05 versus vehicle treated mice. (E and F) Platelet counts in isolated blood (E) and the CD41+CD42b+ MK fraction among Lin- BM cells (F) treated with vehicle (CTRL), low- or high-dose TPO, IL-1α, anti-IL-1α neutralizing antibody (IL-1Ab), anti-IL-1R neutralizing antibody (IL-1RAb), or isotype-matched control antibody (IgG1 for IL-1Ab, IgG2 for IL-1RAb). All antibodies were used at 100 µg/mouse administered i.p. daily for 3 d. n = 8 animals in each group. (G) Identification of newly produced MKs using MX-Cre-GFP mice. GFP-labeled cells were analyzed among Lin- BM cells 2 d after PIPC injection. The data shown are from a single representative experiment from among three repeats. (H) Quantification of thrombopoiesis under physiological conditions in 6-wk-old IL-1α+/+, IL-1α−/−, IL-R1+/+, and IL-1R1−/− mice. (I) MK dynamics in chimeric mice. n = 50 high-power fields from
which we examined whether the dynamics of platelet-like particle formation and release were suppressed by either drug in the presence of IL-1α (Fig. 7, A and B). Examination of tubulin immunofluorescence in fetal liver–derived MKs showed that IL-1α promoted formation of small vesicles in the periphery of MKs that all stained positive for β1-tubulin, but some were negative for α-tubulin (Fig. 7, C and D). However, TPO accelerated PPF, with the well-coordinated distribution of α- and β1-tubulin within MKs. TPO also similarly increased expression of both Tubal1a and Tubb1. In contrast, IL-1α induced a 10-fold increase in Tubb1 expression, and a 50-fold increase when combined with TPO, while having no effect on Tubal1a, Tubalc, or Tuba4a expression (Fig. 7 E and not depicted). We found the formation of a demarcation membrane system and development of platelet territories in IL-1α–treated MKs before membrane rupture (Fig. 7 F and Fig. S5). Platelets from Thpo−/− mice treated with IL-1α showed an absence of proper tubulin distribution, suggesting β1-tubulin oversupply, and those from IL-1α–treated WT mice are mostly spheroid shape rather than elongated shape by WT or TPO-treated platelets (Fig. 7, G–I; and Fig. S5). We concluded that IL-1α–induced disorganization of microtubule assembly accounts for the dysregulated α- and β1-tubulin synthesis, which should contribute to the PPF inhibition and MK rupture as a result.

5 animals in each group. *, P < 0.05 versus control mice. (J) Fractions of thiazole orangehigh platelets in isolated blood from WT mice treated with vehicle, TPO, IL-1α, or/and clodronate. n = 5 mice. (K) Flow cytometric size analysis of thiazole orangehigh and thiazole orangelow platelets in WT mice treated with TPO or IL-1α. (L) Release of CD41+CD42b+ particles in culture medium. n = 5 experiments. *, P < 0.05.
Figure 6. IL-1α-induced atypical apoptosis with caspase-3 activation in fetal liver MKs. (A and B) Liver cells were obtained from fetal WT mice on embryonic day 13, cultured with TPO or IL-1α, and analyzed on day 7. (A) RT-PCR analysis of gene expression in the harvested cells. The values were normalized to a vehicle-treated control. n = 5 experiments. (B) Flow cytometric analysis of caspase-3 activation in Lin−CD41+CD42b+ MKs. (C) Fetal liver cells were cultured with TPO. siRNA-mediated knockdown was performed on day 4. On day 7, cells were washed, incubated with TPO, IL-1α, or IL-1β for an additional 1 d, and pAKT-pERK signaling in Lin−CD41+CD42b+ MKs was analyzed on day 8. The data shown are from a single representative experiment from among three repeats (B and C). (D) Western blotting of p53 and phospho-p53 in fetal liver cells cultured with TPO and IL-1α from day 0 to 7. MKs were enriched with discontinuous albumin density gradient centrifugation. (E) Fetal liver cells were differentiated using TPO for 7 d, and then stimulated with vehicle, TPO, IL-1α, or IL-1β for an additional 1 d, after which gene expression was analyzed by RT-PCR. (F) Flow cytometric analysis of T. orangehigh and T. orangelow platelets isolated from WT mice treated with TPO or IL-1α. NC denotes a negative control. The data shown are from a single representative experiment from among five repeats. (G–J) Immunofluorescence analysis of fetal liver MKs, which were cultured and differentiated from day 0 to 7 with TPO or IL-1α. Some cells were treated with Fas-ligand from day 6 to 7 after differentiation with TPO (FasL). Note that IL-1α–treated MKs were caspase-3–positive with release of von Willebrand factor–positive granules, but TUNEL staining was negative, which was different than typical FasL-induced apoptosis with blebbing. (K) Blood cell counts in WT mice treated with TPO, IL-1α, and/or Z-VAD (OMe)-FMK. (L) MK dynamics and platelet counts in WT, Casp3−/−, and Thpo−/− mice treated with vehicle or IL-1α. n = 3–8 mice. Bars, (red) 20 µm. *, P < 0.05 versus CTRL group.
IL-1α inhibited regulated tubulin assembly and proplatelet formation. (A) Quantification of MK numbers and dynamics and platelet counts in 6-wk-old CAG-eGFP mice treated with IL-1α (10 µg for 5 d), colchicine (5 mg/kg i.v. once 6 h before experiments), and/or paclitaxel (10 mg/kg i.v. once 6 h before experiments). The BM was visualized and platelet counts were analyzed 7 d after the first administration. n = 24 high-power fields from 8 animals in each group. * P < 0.05 versus control (WT). (B–E) Immunofluorescence analysis of fetal liver MKs cultured and differentiated from day 0 to 7 with TPO or IL-1α. In addition, the cells were treated with colchicine (2.5 µM) or paclitaxel (2.5 µM) from day 6 to day 7 (B). Some cells were also treated with Fas-ligand with TPO from day 6 to 7 (FasL). On day 7, the cells were fixed and stained (C and D). (E) RT-PCR analysis of gene expression in harvested cells. The values were normalized to vehicle-treated control. n = 8 experiments. (F) Electron microscopy of isolated BM MKs from TPO or IL-1α mice. Note that demarcation membrane system was similarly developed in two mice (1 and 2), before rupture, and fragments indicated several platelets (3 and 4). Bars, 2 µm. (G) Immunofluorescence study of tubulin distribution in platelets from WT, Thpo−/− mice, or Thpo−/− mice treated with IL-1α. (H) Electron microscopy of isolated platelets from WT mice treated with vehicle (WT), TPO, or IL-1α. (I) The short and long axis length was measured in randomly selected individual platelets, and ratio (short/long) was evaluated in 40 cells for each groups. Bars, 2 µm. * P < 0.05.
et al., 2007; Takayama et al., 2010). However, although recent in vivo imaging-based analyses confirmed the presence of PPF (Junt et al., 2007; Zhang et al., 2012), we found that the number of platelets produced from each MK was both consistent with previously reported numbers (Thon et al., 2012; Avanzi and Mitchell, 2014) and too low for an adequate response to acute platelet needs or inflammatory stimuli.

Herein we demonstrated an alternative MK rupture–type thrombopoietic process, which can provide 20 times more platelets than PPF, released primarily into BM vessels within 1 h (Fig. 1 D), and which we visualized for the first time using high-speed two-photon microscopy with multicolor high-sensitive GaAs detectors (Fig. 1). The novel MK rupture–dependent pathway is primarily regulated by the inflammatory cytokine IL-1α, and is morphologically and mechanically distinct from PPF and typical FasL-induced apoptosis, as well as from the recently reported cytoplasmic elongation of large platelet progenitor structures (Thon and Italiano, 2010; Kowata et al., 2014), as summarized in Table S1 and Fig. 9.

Ng et al. (2014) recently reported that MK maturation and final platelet shedding can both be independent of TPO signaling, but they did not address alternatives to TPO. In this study, we found that IL-1α–IL1R1 signaling positively influences the MK differentiation phase, as well as platelet shedding through rupture, which compensated for the deficiency of TPO signaling in MKs (Ng et al., 2014). In addition, a shear-dependent

**Discussion**

**Novel MK rupture thrombopoiesis**

In vitro platelet biogenesis from fetal liver MKs, human CD34+ cells, embryonic stem cells, or induced pluripotent stem cells all consistently show the presence of PPF and release of fragments from the tips of elongated pseudopodal structures under steadystate and TPO-stimulated conditions (Italiano et al., 1999; Eto et al., 2007; Takayama et al., 2010). However, although recent in vivo imaging-based analyses confirmed the presence of PPF (Junt et al., 2007; Zhang et al., 2012), we found that the number of platelets produced from each MK was both consistent with previously reported numbers (Thon et al., 2012; Avanzi and Mitchell, 2014) and too low for an adequate response to acute platelet needs or inflammatory stimuli.

Herein we demonstrated an alternative MK rupture–type thrombopoietic process, which can provide 20 times more platelets than PPF, released primarily into BM vessels within 1 h (Fig. 1 D), and which we visualized for the first time using high-speed two-photon microscopy with multicolor high-sensitive GaAs detectors (Fig. 1). The novel MK rupture–dependent pathway is primarily regulated by the inflammatory cytokine IL-1α, and is morphologically and mechanically distinct from PPF and typical FasL-induced apoptosis, as well as from the recently reported cytoplasmic elongation of large platelet progenitor structures (Thon and Italiano, 2010; Kowata et al., 2014), as summarized in Table S1 and Fig. 9.

Ng et al. (2014) recently reported that MK maturation and final platelet shedding can both be independent of TPO signaling, but they did not address alternatives to TPO. In this study, we found that IL-1α–IL1R1 signaling positively influences the MK differentiation phase, as well as platelet shedding through rupture, which compensated for the deficiency of TPO signaling in MKs (Ng et al., 2014). In addition, a shear-dependent

**IL-1α induces mechanically and functionally weaker plasma membrane structure**

The multiple functional changes seen in IL-1α–treated MKs before membrane rupture during thrombopoiesis were also examined. Using atomic force microscopy to evaluate MKs’ ability to push a bead-attached cantilever or exert contractile force to pull up a cantilever, it was found that IL-1α–treated MKs were less stiff and thus exerted less force (Fig. 8, A and B). Fluorescent recovery after photobleaching (FRAP) analysis further confirmed that membrane stability was reduced in IL-1α–treated mature polyploid MKs, but not in mononuclear CD41+ cells (immature MKs), and that it was restored by caspase inhibition (Z-VAD [OMe]-FMK; Fig. 8, C and D; Ahn et al., 2002). These results strongly indicate that IL-1α reduces plasma membrane potential, which leads to a mechanically and functionally weaker membrane structure and drives MKs toward MK rupture thrombopoiesis.

**Figure 8.** IL-1α drives MK rupture thrombopoiesis by reducing functional and mechanical membrane stability in MKs. (A and B) Fetal liver cells from WT mice were cultured for 7 d with IL-1α or TPO, after which MKs were evaluated using atomic force microscopy by pushing a bead-headed cantilever to measure stiffness (A) and pulling up on a membrane-attached cantilever to measure contractile force (B). Representative force-measurement curve, stiffness, and contractile force against cantilevers were shown. n = 20 measurements. (C and D) Fluorescence recovery after photobleaching (FRAP) analysis. Fetal liver MKs were cultured for 7 d with IL-1α or TPO. Some cells cultured with TPO were also treated with Z-VAD [OMe]-FMK (100 µM) 1 d before the experiments. MKs were stained with Di8-ANEPPS and then photobleached in a region of interest (ROI; red box). Restoration of Di8-ANEPPS intensity reflected membrane fluidity and instability. The left panels show representative snapshots during photobleaching and the corresponding Di8-ANEPPS signals. MKs were divided into mature highly segmented (more than two nuclear segmentations) and not segmented (one or two nuclear segmentations) groups. n = 20 examinations from 5 specimens. *, P < 0.05 versus TPO.
mechanism may also promote TPO-independent platelet generation (Jiang et al., 2014).

MKs respond to acute platelet demand via IL-1α

Under acute inflammatory or cytopenic conditions, rapid elevation of IL-1α promoted a change in cell progranling to MK rupture thrombopoiesis (Fig. 2 and 3). Reductions in platelet counts during acute inflammation induced by neutralizing anti–IL-1α antibody indicate that an urgent requirement for platelet recruitment or recovery as a host defense may be associated with inflammation-mediated elevation of IL-1α levels, as reported previously (Rider et al., 2011). However, serum levels of multiple inflammatory cytokines, including IL-1β, IL-2, IL-6, and IL-11, were also elevated and may contribute to the increase in platelet production (Fig. 3). Although IL-1β, produced from platelets during acute inflammation, reportedly stimulates MK polyploidization (Yang et al., 2000; Denis et al., 2005; Beaulieu et al., 2014), the levels of IL-1β were not associated with elevation of platelets after administration of anti-CD42b antibody (Fig. 3 C). This result is consistent with the fact that the phenotypes of IL-1α and IL-1β knockout mice differ from one another (Nambu et al., 2006).

IL-1α–treated MKs showed clear changes to their cell membrane before MK rupture (Fig. 8). The plasma membrane was functionally and mechanically less stable in IL-1α–treated MKs. Instability of membrane structure after IL-1α treatment may also be associated with platelet shape in generation. Platelets by IL-1α appeared to display more spheroid but PPF-dependent platelets were longitudinal (Fig. 7, G–I; and Fig. S5), as demonstrated previously (Thon and Italiano, 2010). Interestingly, this membrane instability was dependent on caspase activity in only mature MKs (Fig. 8 D). That the action of IL-1α in MK rupture thrombopoiesis was caspase-3 dependent (Fig. 6, K and L) suggests caspase activation may occur downstream of the IL-1α–ILR1 axis in association with alteration of the cell membrane structure. The caspase-dependent thrombopoiesis theory has been discussed for a long time (De Botton et al., 2002; Clarke et al., 2003; Josefsson et al., 2014). Our data provide evidence of caspase dependency in platelet biogenesis; however, the caspase involvement is independent of TPO-mediated PPF, as recently demonstrated (Josefsson et al., 2014). But the detailed mechanism of caspase activation or p53 up-regulation in MK rupture thrombopoiesis must be elucidated further.

In conclusion, our findings provide direct evidence that IL-1α is an acute platelet releasing factor that induces enhanced platelet release through rupture of the mature MK membrane. This novel mechanism may enable rapid restoration in platelet numbers, but with insufficient microtubule organization. It appears the balance between TPO and IL-1α determines the cellular programming of MKs for thrombopoiesis in response to acute and chronic platelet needs.
Materials and methods

Mice

WT C57BL/6, B6 and B6.129S7-Il1r1 tm1Nwa / Il1r1 tm1Nwa mice were obtained from Japan Charles River Laboratories. A null mutation in Il1r1 was generated by homologous recombination using a replacement vector in which a 2.4kb EcoRI–Pst1 fragment encompassing two exons was replaced with a PGKneo cassette. B6.Cg-Tg(CAG-floxed Neo-EGFP)REP08Osb mice carrying the transgene for the CAG promoter and EGFP cDNA separated by a floxed neo cassette were a gift from Riken [Matsumura et al., 2004]. B6.Cg-Tg(CAG-Tet-OFF-tdTomato)50G39Osb mice were provided by B. Heissig (The University of Tokyo, Tokyo, Japan; de Sauvage et al., 1996).

Animal models

To assess the contributions made by humoral factors, 5-week-old CAG-eGFP mice were treated with TPO (Sigma-Aldrich) or IL-1r1 (R&D Systems or Biolegend) at dosages of 10 µg/mouse subcutaneously (SC) daily for 5 or 70 µg/mouse SC daily for 3 days. The mice were visualized and examined at 6-week-old, 7 days after the first treatment. Some mice were also treated with neutralizing antibodies against IL-1r1 (R&D Systems or Biolegend), IL-1R (Biolegend), isotype-matched control antibody (Biolegend), or Fas Ligand (5 µg/mouse i.v.; R&D Systems). All antibodies were administered at 100 µg/mouse i.p. daily for 3 days.

To assess the effect of acute inflammation, thiglycolate (Sigma-Aldrich) was administered i.p. (3 ml of 3% solution/mouse once). To examine the effect of caspase inhibition, mice were treated with the pan-caspase inhibitor Z-VAD (OMe)-FMK (3 mg/kg IP daily for 5 d; Merck Millipore). To assess the effect of caspase inhibition, mice were treated with the pan-caspase inhibitor Z-VAD (OMe)-FMK (3 mg/kg IP daily for 5 d; Merck Millipore).

Immunofluorescence study

For immunofluorescence analyses, the cells were fixed in 4% paraformaldehyde for 45 min and permeabilized with 1% Triton X-100 (EMD Millipore). Differentiated MKs, identified as multinucleate and staining positively for CD41, were visualized at 37°C in DMEM using a confocal microscope (Nikon) with bead-attached or sharp-head cantilevers.

Immunofluorescence study

To evaluate plasma membrane stability, we performed FRAP analysis with cultured fetal liver MKs. The MKs were stained with DII-ANEPSS (5 µM), then incubated with a primary antibody. After 12 h with a primary antibody (rabbit anti–β1-tubulin [Patel-Hett et al., 2008], rat anti-CD41 [MWReg30], goat anti-willebrand factor (Emfret), or rabbit anti-active caspase-3 (Millipore). The cells were then incubated for 1 h with an RPE-conjugated rat anti-CD41 (MWReg30), Alexa Fluor-conjugated mouse anti–α-tubulin (Invitrogen) and/or goat secondary antibody (Invitrogen). Finally, the cells were counterstained for 1 h with Hoechst 33342, and images were captured using a confocal microscope (Nikon A1R). The cells were excited using three laser lines (405 nm, 488 nm, and 561 nm), and the emission was collected using appropriate narrow band-pass filters and GaAs detectors.

Electron microscopy

For electron microscopy, MKs were fixed in 2% glutaraldehyde for 45 min and permeabilized with 1% Triton X-100 (EMD Millipore) for 10 minutes. The specimens were blocked and incubated first for 12 h with a primary antibody (rabbit anti–β1-tubulin [Patel-Hett et al., 2008], rat anti-CD41 [MWReg30]; Biolegend), rabbit anti-α-tubulin (Millipore), or rabbit anti-active caspase-3 (Millipore). The MKs were evaluated using atomic force microscopy (Nanowizard JPK Instruments AG) with bead-attached or sharp-head cantilevers.

Preparation of cells and flow cytometry

As previously described (Nishimura et al., 2009), we isolated BM cells from the scalp and femurs by flushing them with PBS, and collected blood samples from tail veins or by cardiac puncture under anesthesia. The cells were washed twice with PBS, incubated for 8.5 min in erythrocyte-lyzing buffer, and finally suspended in PBS supplemented with 3% fetal bovine serum. The isolated cells were then incubated with FcBlock antibody (BD) for 15 min on ice, labeled with dye-conjugated antibodies, and analyzed by flow cytometry using a Canto II flow cytometer (BD), spectrum analyzer SP6800 (Sony), and FlowJo 7.2.2. software (Tomy Digital Biology). Propidium iodide (Invitrogen) or a Zombie NIR kit (Biolegend) at dosages of 10 µg/mouse subcutaneously (SC) daily for 5 d was used to exclude dead cells. Conventional approaches, including Western blotting,
cannot be applied to assess intracellular signals in MKs isolated from mice due to the small sample size. We therefore used flow cytometry for signal analysis.

Flow cytometry-based platelet aggregation assays were also performed using previously reported methods with some modification (De Cuyper et al., 2013). Platelet-rich plasma and whole blood were obtained and incubated with RPE-conjugated anti-CD41 (eBioscience; BioLegend) or FITC-conjugated anti-CD41 (Luc-H11; Emfret) antibody, after which the two populations of labeled cells were mixed, 1:1, and incubated at 37°C. The cells were then activated with thrombin (Sigma-Aldrich; 5 U/mL), fixed in 0.5% formaldehyde at the indicated times thereafter, and analyzed using a Canto II flow cytometer (BD). The appearance of two-color events was considered to reflect aggregated platelets.

Western blotting
Western blotting was conducted with fetal liver cells enriched through discontinuous albumin density gradient centrifugation using rabbit anti-p53 (Santa Cruz Biotechnology, Inc.), or rabbit anti-phospho-p53 (Ser15; Cell Signaling Technology) antibodies.

Real-time quantitative PCR
For real-time PCR, total RNA was isolated using TRIzol (Invitrogen) and relative mRNA levels were determined using a Superarray kit (Superarray). Real-time quantitative PCR

Statistics
Statistical analysis was performed using JMP (SAS) and Excel 2013 (Microsoft) software. Results were expressed as means ± SEM. The statistical significance of differences between two groups was evaluated using Student’s t tests. Differences between more than two groups were evaluated using ANOVA followed by post hoc Bonferroni tests. Correlations were evaluated using the Pearson correlation coefficient test. Values of P < 0.05 were considered significant.

Online supplemental material
Fig. S1 shows intravital visualization and validation study of BM MKs. Fig. S2 shows MK phenotype of IL-1β-induced platelets. (Santa Cruz Biotechnology, Inc.), or rabbit anti-phospho-p53 (Ser15; Cell Signaling Technology) antibodies.

We are grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.

We are very grateful to M. Taniguchi, C. Yoshinaga, M. Tajima, X. Yingda, and T. Hiranabashi for their excellent technical help. We appreciate the gifts of mice from Drs. Heissig and Iwakura.


Figure S1. Intravital visualization of MK dynamics. (A) Flow cytometric analysis of CAG-eGFP signals in Lin−CD41+CD42b+ MKs (red), F4/80+CD11b+ macrophages (Mac, Blue), CD3+ T cells (green), and CD45R+CD19+ B cells (purple) in 6-wk-old CAG-eGFP transgenic mice under steady-state conditions. Note that GFP expression was higher in MKs than other cell types. The black line denotes the negative control (NC) in age-matched WT mice. The data shown are from a single representative experiment from among three repeats. (B) eFluo450-conjugated anti-CD41 antibody (50 µg/mouse; blue) and Texas-red dextran were injected into 6-wk-old CAG-eGFP mice also under steady-state conditions. Intravital visualization of scalp BM revealed CD41 staining of MKs, which were identified based on their large size and stronger GFP signal. Bar, 20 µm. (C) Time-lapse images of thrombopoiesis in living BM in 6-wk-old CD41-tdTomato knock-in mice (red). Injected fluorescent dextran (green) shows the blood flow, and nuclei are labeled with Hoechst 33342 (blue). There are two thrombopoietic patterns. White arrows indicate the blood flow direction, and triangles point to platelet production. Bars, 20 µm. (D–F) The effect of laser power on MK dynamics in CAG-eGFP mice. (D) Representative MK image after 30 min of XYZ-T observation using three different laser (λ = 920 nm) powers: 100% (high), 50% (mid), and 25% (low) of the maximum power of the Vision II (Coherent). (E) We continued this laser irradiation, and MK dynamics were quantified as in Fig. 1. At high power, the laser irradiation slightly increased blebbing and apoptotic cell death, but not MK rupture. Note that laser power usually did not exceed 15% of maximum for observation, due to the usage of highly sensitive GaAs detectors. n = 50 high-power fields from 5 animals in each group. (G) To reduce the effect of reactive oxygen species photochemically produced during observation, CAG-eGFP mice were treated with ascorbic acid (50 mg/kg) or catalase (5 mg/kg) before the experiments, and MK dynamics were also quantified. There are no significant differences among the three groups. n = 50 high-power fields from 5 animals in each group. Bars, 20 µm.
**Figure S2. IL-1α and platelet biogenesis.** (A) Serum TPO levels in WT mice after vehicle or TPO treatment (10 µg/mouse s.c. daily for 5 d), n = 5 animals in each group. (B) Quantification of thrombopoiesis and platelet counts in 6-wk-old IL-1α+/+, IL-1 α−/−, IL-R1+/+ and IL-R1−/− mice treated with TPO (10 µg/mouse, s.c., daily for 5 d) or IL-1α (10 µg/mouse, s.c., daily for 5 d). n = 50 high-power fields from 5 animals in each group. *, P < 0.05 vs. each control untreated mice. (C) Flow cytometric analysis of platelets isolated from IL-1α+/+, IL-1 α−/−, IL-R1+/+, and IL-R1−/− mice treated with anti-CD42b antibody (R300; 100 µg/mouse). Platelets were separated into thiazole orange-high and low groups. T. Orange high FSC/SSC populations were smaller in IL-1 α−/− and IL-R1−/− mice than control mice.
Figure S3. Functional evaluation of IL-1α–induced platelets. [A] MK rupture in CAG-eGFP injected with IL-1α, tetramethylrhodamine ethyl ester (TMRE) to evaluate mitochondrial membrane potentials, and Hoechst 33342 to visualize nuclei. Note that particles released after MK rupture showed TMRE-positive staining. Bar, 20 µm. [B] CAG-eGFP mice were injected with Texas red dextran (red), Hoechst 33342 (blue), and hematoporphyrin, after which thrombosis was induced using laser irradiation, which causes production of ROS (reactive oxygen species) within the vessel, as previously reported (Nishimura et al., 2012). One-shot visualization of thrombus formation within testicular veins (left) and quantification of thrombus area after 60 s of laser irradiation (right). n = 20 vessels from 4 animals. Bars, 100 µm. Arrows: direction of blood flow. (C) JONA binding to washed platelets and MFI analysis of JONA binding to platelets isolated from WT mice treated with vehicle control (CTRL), IL-1α, or low dose TPO. n = 8 mice. (D) Platelet aggregation activity. Platelet and whole blood fractions isolated from WT mice treated with IL-1α or low dose TPO were labeled with anti-CD41 or -CD61 antibody, mixed, and then stimulated with thrombin (5 U/ml). Shown are representative double plots after stimulation for 15 min (left) and the quantification of two-color particles (right).
Figure S4. **IL-1α secretion, IL1R expression, and pERK–AKT signaling in MKs.** (A) Flow cytometric analysis of IL-1R in F4/80+CD11b+ macrophages (Mac), CD3+ T cells, and CD41+CD42b−Lin− MKs in BM cells from 6-wk-old WT mice. Shown are representative plots from five experiments. (B) ELISA analysis of IL-1α in culture medium from sorted F4/80+CD11b+ macrophages (Mac), CD3+ T cells, CD19+ B cells, and CD41+CD42b− MKs in BM. n = 5 experiments. (C) Fetal liver cells were collected from WT mice and differentiated into MKs under stimulation with TPO (50 ng/ml). On day 4 of culture, siRNA-mediated knockdown was performed. On day 7, the cells were stimulated with IL-1α (50 ng/ml) for an additional 1 d. Shown are RT-PCR and flow cytometric analyses of IL-1R1 and IL-1R2. n = 5 experiments. (D) Fetal liver cells were collected from WT mice and differentiated into MKs under the stimulation with TPO (50 ng/ml). The cells were then cultured for an additional 1 d without or with TPO (50 ng/ml), IL-1α (50 ng/ml) or IL-1β (50 ng/ml). After harvesting the cells, pAKT and pERK signals in the CD41+CD42b−Lin− MKs were analyzed using flow cytometry. Shown are % pAKT/pERK-positive cells among MKs. n = 5 experiments with 20 animals. Gating is shown in Fig. 6. *, P < 0.05. (E) Fetal liver cells were collected from WT mice and differentiated into MK under stimulation with TPO (50 ng/ml). On day 4 of culture, siRNA-mediated knockdown was performed. On day 7, the cells were stimulated with IL-1α (50 ng/ml) for an additional 1 d. Shown are percentage of pAKT/pERK-positive cells among MKs. n = 5 experiments with 20 animals. Gating is shown in Fig. 6. *, P < 0.05. MKs were enriched using a BSA gradient before culture.
Figure S5. Electron microscopy. (A–C) Electron microscopy of isolated BM MKs from TPO or IL-1α treated mice. The distances between platelet territory (shown by red lines in A and quantification in B), and number of granule contents per area [C] were evaluated. n = 100 measurements (B) and 20 ROI areas [C] for each group. (D and E) Representative images and axis length ratio of platelets isolated from WT, Thpo, IL-1α treated Thpo−/−, IL-1α−/−, and IL1R1−/− mice. The short and long axis lengths were measured in randomly selected individual platelets, and length ratio (short/long) was evaluated in 40 cells for each groups. Note more spheroid shape of platelets from IL-1α treated Thpo−/− mice. Bars, 2 µm. *, P < 0.05 versus control (WT).
Video 1. **Thrombopoiesis via short-type proplatelet formation under steady-state conditions.** BM MKs from 6-wk-old CAG-eGFP (green) mice were visualized under steady-state conditions using a high-speed, two-photon microscope (Nikon A1R MP). Injected fluorescent dextran (red) shows the blood flow, and Hoechst 33342 (blue) labeled the nucleus. XYZT images were taken every minute, and shown using maximum intensity projection. Captured images are from Fig. 1 A.

Video 2. **Thrombopoiesis via short-type proplatelet formation under steady-state conditions.** BM MKs from 6-wk-old CAG-eGFP (green) mice were visualized under steady-state conditions using a high-speed, two-photon microscope (Nikon A1R MP). Injected fluorescent dextran (red) shows the blood flow, and Hoechst 33342 (blue) labeled the nucleus. XYZT images were taken every minute, and shown using voxel views. Captured images are from Fig. 1 B.

Video 3. **Thrombopoiesis via long-type proplatelet formation after TPO treatment.** BM MKs from 6-wk-old CAG-eGFP (green) mice were visualized after treatment with TPO (10 µg for 5 d) using a high-speed, two-photon microscope (Nikon A1R MP). Injected fluorescent dextran (red) shows the blood flow, and Hoechst 33342 (blue) labeled the nucleus. XYZT images were taken every minute, and shown using slice views. Captured images are from Fig. 1 C.

Video 4. **MK rupture under steady-state conditions.** BM MKs from 6-wk-old CAG-eGFP (green) mice were visualized under steady-state conditions using a high-speed, two-photon microscope (Nikon A1R MP). Injected fluorescent dextran (red) shows the blood flow, and Hoechst 33342 (blue) labeled the nucleus. XYZT images were taken every minute, and shown using slice views. Captured images are from Fig. 1 F.

Video 5. **MK rupture under steady-state conditions.** BM MKs from 6-wk-old CAG-eGFP (green) mice were visualized under steady-state conditions using a high-speed, two-photon microscope (Nikon A1R MP). Injected fluorescent dextran (red) shows the blood flow, and Hoechst 33342 (blue) labeled the nucleus. XYZT images were taken every minute, and shown using voxel views. Captured images are from Fig. 1 F.

Video 6. **MK rupture under steady-state conditions.** BM MKs from 6-wk-old CAG-eGFP (green) mice were visualized under steady-state conditions using a high-speed, two-photon microscope (Nikon A1R MP). Injected fluorescent dextran (red) shows the blood flow, and Hoechst 33342 (blue) labeled the nucleus. XYZT images were taken every minute, and shown using slice views. Captured images are from Fig. 1 G.
**Video 7.** MK rupture after anti-CD42b antibody administration. BM MKs from 6-wk-old CAG-eGFP (green) mice were visualized after administration of a neutralizing anti-CD42b antibody (100 µg/mouse i.p. daily for 3 d) using a high-speed, two-photon microscope (Nikon A1R MP). Injected fluorescent dextran (red) shows the blood flow, and Hoechst 33342 (blue) labeled the nucleus. XYZT images were taken every minute, and shown using slice views. Captured images are from Fig. 2 A.

**Video 8.** MK rupture after IL-1α treatment. BM MKs from 6-wk-old CAG-eGFP (green) mice were visualized after treatment with IL-1α (10 µg/mouse s.c. daily for 5 d) using a high-speed, two-photon microscope (Nikon A1R MP). Injected fluorescent dextran (red) shows the blood flow, and Hoechst 33342 (blue) labeled the nucleus. XYZT images were taken every minute, and shown using slice views. Captured images are from Fig. 4 A.

**Video 9.** MK rupture after IL-1α treatment. BM MKs from 6-wk-old CAG-eGFP (green) mice were visualized after treatment with IL-1α (10 µg/mouse s.c. daily for 5 d) using a high-speed, two-photon microscope (Nikon A1R MP). Injected fluorescent dextran (red) shows the blood flow, and Hoechst 33342 (blue) labeled the nucleus. XYZT images were taken every minute, and shown using slice views. Captured images are from Fig. 4 B.

**Video 10.** MK rupture after IL-1α treatment. BM MKs from 6-wk-old CAG-eGFP (green) mice were visualized after treatment with IL-1α (10 µg/mouse s.c. daily for 5 d) using a high-speed, two-photon microscope (Nikon A1R MP). Injected fluorescent dextran (red) shows the blood flow, and Hoechst 33342 (blue) labeled the nucleus. XYZT images were taken every minute, and shown using slice views. Captured images are from Fig. 4 C.

**Table S1.** Characteristics of proplatelet formation, rupture type thrombopoiesis, and apoptosis.

<table>
<thead>
<tr>
<th></th>
<th>Proplatelet</th>
<th>Rupture</th>
<th>Typical apoptosis</th>
</tr>
</thead>
<tbody>
<tr>
<td><strong>Particle release</strong></td>
<td>Yes</td>
<td>Yes</td>
<td>No</td>
</tr>
<tr>
<td><strong>From tip</strong></td>
<td>All directions but preferentially into vessel lumens</td>
<td></td>
<td></td>
</tr>
<tr>
<td><strong>No. of released particles</strong></td>
<td>Small</td>
<td>Large</td>
<td></td>
</tr>
<tr>
<td><strong>Time course</strong></td>
<td>Slow, &gt;2–3 h</td>
<td>Rapid, usually within 1 h</td>
<td>Slow, &gt;80 min</td>
</tr>
<tr>
<td><strong>Cell death</strong></td>
<td>No</td>
<td>Yes</td>
<td>Yes</td>
</tr>
<tr>
<td><strong>Active Caspase-3</strong></td>
<td>~++</td>
<td>++</td>
<td>+++</td>
</tr>
<tr>
<td><strong>Annexin V staining</strong></td>
<td>-</td>
<td>-</td>
<td>+++</td>
</tr>
<tr>
<td><strong>vWF+ contents</strong></td>
<td>+++</td>
<td>+++</td>
<td>--+</td>
</tr>
<tr>
<td><strong>α-tubulin</strong></td>
<td>+</td>
<td>+</td>
<td>+</td>
</tr>
<tr>
<td><strong>β1-tubulin</strong></td>
<td>+</td>
<td>Accumulated or overloaded</td>
<td>decreased</td>
</tr>
<tr>
<td><strong>Frequency under steady state</strong></td>
<td>Major</td>
<td>Rare</td>
<td>Usually none</td>
</tr>
<tr>
<td><strong>Signals</strong></td>
<td>TPO dependent</td>
<td>IL-1α dependent</td>
<td></td>
</tr>
</tbody>
</table>

MK rupture thrombopoiesis is morphologically distinct from proplatelet formation or typical apoptosis.